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2005

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EDITORIAL STAFF

COVER: Aggregation of copperheads (*Agkistrodon contortrix*) captured near Yellville (Marion County, AR). Photo by Stan Trauth.

Arkansas Academy of Science 2005

April 8-9, 2005
89th Annual Meeting

Hendrix College
Conway, Arkansas

JOURNAL
ARKANSAS ACADEMY OF SCIENCE

ANNUAL MEETING 8-9 APRIL 2005
HENDRIX COLLEGE

Secretary's Report

MINUTES OF THE 89TH MEETING

ARKANSAS ACADEMY OF SCIENCE
SUMMARY OF 1st and 2nd BUSINESS MEETINGS
HENDRIX COLLEGE
APRIL 8-9, 2005

1. Call to Order: Betty Crump, President of the AAS, called the meetings to order. Betty wanted to call attention to two Academy officers who had been recognized this year. Joyce Hardin was named as an outstanding alumnus by the University of Arkansas in their alumni publication. David Saugey was named as the Educator of the Year by the U.S. Forest Service.

2. Local Arrangements Committee: Local Arrangements Chair and Arkansas Academy of Science Treasurer Joyce Hardin expressed her thanks to the many people who have helped to host the meeting who will also be recognized in the resolutions. There were 203 attendees that registered for the meeting. There were 92 oral presentations, 23 poster presentations as well as 35 students entered in the competitions. She then announced the various scholarly awards presented to students for their research presented at the meeting symposia. (They are listed elsewhere in this volume.)

3. Treasurer/Auditor: The financial report was presented by Joyce Hardin. Details on the income and expenses for the year highlighted in addition to journal cost issues. The expense of the journal was reduced last year and helped the Academy post a substantial gain compared to last year. The cost of the journal for this year was dramatically reduced due in part to new technologies at the printer and should help our finances even more. Stan Trauth and Malcolm McCallum were acting auditors and reviewed the financial statements. Everything was deemed "OK." (OK translating into the excellent integrity and good financial records kept by the Academy showing no inconsistencies or irregularities.)

4. Secretary: The minutes from 2004 Executive Committee business meeting in November 2004 were distributed and approved. Prior to this meeting, the current membership list included approximately 132 members (56 which are life members) of the Academy. A request for $200 was made to offset mailing charges incurred for the AAS mailings, the Newsletter, and Journals that are not picked up at the annual meeting was approved.

5. Historian: Henry Robison reported that this is the 89th annual meeting of the Arkansas Academy of Science. This is the 4th time the annual meeting was held at the Hendrix College Campus, the other meetings being in the years 1939, 1963, 1979.

6. *Journal* Editor-in-Chief: Stan Trauth reported that Vol. 58 of the journal is 144 pages. A request for the Academy to continue to support the *Journal* Editor-in-Chief with an allotment of $200 and $600 for assistant editor duties to cover incurred costs was accepted.

7. *Journal* Managing Editor: Chris McAllister sounded his appreciation for authors in following the formatting and submission instructions. He felt the quality of paper submissions has continued a steady rise. A request for $500 to cover incurred costs associated with managing editor duties was approved.

8. Arkansas Science Fair Association: A request and approval was obtained for $400 dollars to support the Arkansas Science Fair Association from Michael Rapp. The state science fair is in its 51st year and the annual winners go to an international fair that has 1000 students from 40 countries competing. Arkansas has had 4 top two finishers at that event.

ARKANSAS ACADEMY OF SCIENCE
2004 FINANCIAL STATEMENT

BALANCE – December 31, 2004	**$ 28,831.56**
BALANCE – January 1, 2004	**$ 26,164.94**
NET GAIN	**$ 2,666.62**

DISTRIBUTION OF FUNDS

Checking Account Bank of Ozarks, Conway, AR	**$ 2,236.42**
Certificates of Deposit Life Membership Endowment Bank of Ozarks, Conway, AR	**$ 14,269.90**
Dwight Moore Endowment Bank of Ozarks, Conway, AR	**$ 5,153.40**
Phoebe and George Harp Endowment Bank of Ozarks, Conway AR	**$ 5,671.84**
Unrestricted Bank of Ozarks, Conway, AR	**$ 1,500.00**
TOTAL	**$ 28,831.56**

INCOME:

1. ANNUAL MEETING		**$ 500.00**
2. ENDOWMENT DONATIONS		
a. AAS Unrestricted	$ 25.00	
b. Moore	25.00	
	50.00	**$ 50.00**
3. INTEREST		
a. Endowment	$ 397.19	
b. Checking	7.18	
	404.37	**$ 404.37**
4. JOURNAL		
a. Miscellaneous Sales	$ 2,550.00	
b. Page Charges	10,640.00	
c. Subscriptions	750.00	
	$ 13,940.00	**$ 13,940.00**
5. MEMBERSHIP		
a. Associate/Student	$ 495.00	
b. Regular	2,220.00	
c. Sustaining	105.00	
d. Life700.00		
e. Institutional	1,400.00	
	4,920.00	**$ 4,920.00**
6. MISCELLANEOUS Reimbursed bank service charges (12/03 and 1/04)		**$ 16.00**
TOTAL INCOME		**$ 19,830.37**

EXPENSES:

1. ANNUAL MEETING Betty Crump (1026)		**$ 43.20**
2. AWARDS		
a. Arkansas Science Fair(1028)	$ 400.00	
b. Arkansas Junior Academy(1029)	250.00	
c. Conway Trophy (1032)	97.43	
d. Student Awards (1033-1040)	650.00	
	$ 1,397.43	**$ 1,397.43**
3. MISCELLANEOUS Checking account service charge (1/04)		**$ 8.00**
4. NEWSLETTER		**$ 0.00**
5. OFFICE EXPENSES		**$ 0.00**
6. JOURNAL		
a. Stan Trauth – Editorial Consultation and Travel Volume 57 (1024)	$ 200.00	
b. Pinpoint Color Volume 57 (1031)	14,415.12	
c. Joy Trauth – Editorial Consultant Volume 58 (1042)	600.00	
d. Chris McAlister Volume 58 (1027)	500.00	
	$ 15,715.12	**$ 15,715.12**
TOTAL EXPENSES		**$ 17,163.75**

ARKANSAS ACADEMY OF SCIENCE
COST OF JOURNAL

VOLUME	COPIES	PAGES	PRINTER CHARGE	TOT. VOL. COST	COST/COPY	COST/PAGE
35 (1981)	450	96	$ 3,694.68	$ 4,620.99	$ 10.27	$ 48.14
36 (1982)	450	110	$ 5,233.28	$ 5,291.69	$ 11.76	$ 48.11
37 (1983)	450	103	$ 5,326.91	$ 5,944.44	$ 13.21	$ 57.71
38 (1984)	450	97	$ 5,562.97	$ 6,167.72	$ 13.71	$ 63.58
39 (1985)	450	150	$ 7,856.20	$ 8,463.51	$ 18.81	$ 56.42
40 (1986)	450	98	$ 6,175.20	$ 6,675.20	$ 14.23	$ 68.11
41 (1987)	450	116	$ 7,122.79	$ 7,811.25	$ 17.36	$ 67.34
42 (1988)	450*	116	$ 7,210.79	$ 7,710.15	$ 17.13	$ 66.47
43 (1989)	450*	119	$ 8,057.24	$ 8,557.24	$ 19.02	$ 71.91
44 (1990)	450*	136	$ 9,298.64	$ 9,798.64	$ 21.77	$ 72.05
45 (1991)	450*	136	$ 9,397.07	$ 9,929.32	$ 22.06	$ 73.01
46 (1992)	450*	116	$ 9,478.56	$ 10,000.56	$ 22.22	$ 86.21
47 (1993)	400	160	$ 12,161.26	$ 12,861.26	$ 32.15	$ 80.38
48 (1994)	450	270	$ 17,562.46	$ 18,262.46	$ 40.58	$ 67.63
49 (1995)	390	199	$ 14,725.40	$ 15,425.40	$ 39.55	$ 77.51
50 (1996)	345	158	$ 11,950.00	$ 12,640.75	$ 36.64	$ 80.00
51 (1997)	350	214	$ 14,308.01	$ 15,008.01	$ 42.88	$ 70.13
52 (1998)	350	144	$ 12,490.59	$ 13,190.59	$ 37.69	$ 91.60
53 (1999)	350	160	$ 13,686.39	$ 14,386.39	$ 41.10	$ 89.91
54 (2000)	350	160	$ 14,149.07	$ 14,849.07	$ 42.43	$ 92.81
55 (2001)	360	195	$ 16,677.22	$ 17,498.22	$ 48.61	$ 89.73
56 (2002)	350	257	$ 18,201.93	$ 19,001.93	$ 54.29	$ 73.94
57 (2003)	230	229	$ 14,415.12	$ 15,715.12	$ 68.33	$ 68.62

The Total Volume Cost equals the printer's charge plus the editor, editorial assistant, and other miscellaneous charges.

- On Volume 42 the Academy received 560 copies, but the printer did not charge us for the extra 110 copies. For comparison purposes the calculated cost/copy is based on 450 copies.
- On Volume 43 the Academy received 523 copies, but the printer did not charge us for the extra 73 copies. For comparison purposes the calculated cost/copy is based on 450 copies.
- On Volume 44 the Academy received 535 copies, but the printer did not charge us for the extra 85 copies. For comparison purposes the calculated cost/copy is based on 450 copies.
- On Volume 45 the Academy received 594 copies, but the printer did not charge us for the extra 144 copies. For comparison purposes the calculated cost/copy is based on 450 copies.
- On Volume 46 the cost was greater than usual due to the high cost of a second reprinting of 54 copies by a different printer.

9. Junior Academy of Science: The Junior Academy is in need of new directorship. Jerry Manion, the interim director, requested continued support through an allotment of $250 which was approved.

10. Intel Talent Search: A request of $64.95 from Jim Murray for student awards was approved. Jim Murray is also stepping down as director.

11. Junior Science and Humanities Symposium has a new director. Linda Kondrick has taken over duties as director for the late Tom Palko, life member of the Academy (see AAS memorial in this journal). A request and approval for continued support of $100 toward their student awards was given.

12. Committee Reports:

    a. Biota Committee: Doug James relays that online access to the Biota lists are moving forward, slowly but surely.
    b. Science Education Committee: Mostafa Hemmati reported that a large effort will be made at the 2006 meeting to advertise this committee meeting time and location so that many members interested can attend.
    c. Development Committee: Betty Crump related her experiences in the wonderful world of grant applications in order to secure funding and sponsorship of AAS programs. The Academy now has an NSF fastlane account and a Dunn's non-profit number to help facilitate our endeavors. Betty is looking at other Academies and their resources, sponsorship, budgets, etc. to provide some ideas for the AAS.
    d. AAAS: Mostafa Hemmati reported on his involvement in the American Association for the Advancement of Science Meeting in Washington, D.C. He will continue to serve as our representative to the AAAS.
    e. Arkansas Science Teachers Association: Tillman Kennon presented issues related to science education and teacher professional development to the Executive Committee.

13. New Business:

    a. Anthony Grafton from Lyon College announced the dates of the 90th Annual meeting as April 7-8, 2006 on the Lyon College campus in Batesville, Arkansas. They have already designed a website and part of their LOC were in attendance at this meeting scouting things out.
    b. It was suggested that two deadlines be followed for the 2006 meeting. Deadline #1 an intent deadline with author and title only, which could be as early as January, followed by the more traditional March abstract and registration deadline. This would allow more people to evaluate their interest in that year's meeting. Lyon College agreed.
    c. An invitation from Arkansas Tech University in Russellville to host the 2007 AAS meeting was also accepted. The 91st meeting will be held April 13-14, 2007 in order to avoid the Easter weekend during the second weekend of that month in 2007. Locations for 2008 and beyond are being solicited.
    d. Walt Godwin announced that the official AAS website is robust as ever. Any errors are to be reported to him immediately. Links to our various affiliations are going to be placed on the site as well, demonstrating our support and symbiosis with other science groups at the state and national level. The website is located at http://cotton.uamont.edu/~aas.
    e. A motion to "allow a member who makes a presentation (oral or poster) at the annual AAS meeting, to submit a manuscript for the peer review process of the *Journal* and that the author instructions in the *Journal* reflect this new policy," was approved.
    f. Wayne Gray suggested that the Executive Committee look into hosting the overall winner of the Junior Academy to come to the AAS annual meeting to make a short presentation. This will be considered.
    g. Resolutions for the annual meeting were read to the membership by David Saugey (see Resolutions).

14. Nominations Committee: Mostafa Hemmati announced nominations for Vice-President of the Academy by the nominations committee (Mostafa Hemmati, Scott Kirkconnell, and Robert Engelken) All Academy members voted between two candidates, Collis Geren, Dean of the Graduate School at the University of Arkansas, and Andrew Sustich, Dean of the Graduate School at Arkansas State University. After voting of the membership, Collis Geren was elected as Vice-President, joining Stan Trauth (President-Elect) and David Saugey (current Vice President) as the new leadership for the upcoming year.

15. Closing: New president Stan Trauth accepted the ceremonial gavel from outgoing president Betty Crump after sending her out with a fishy-looking plaque honoring her service to the Academy.

Meeting adjourned.

Jeff Robertson, AAS Secretary

APPENDIX A

2005 AAS Award Winners

ORAL PRESENTATIONS

GRADUATE STUDENT AWARDS

Life Science:

1st Place Toby M. Ward/UAMS
Construction of Recombinant Varicella Vaccines Espressing Respiratory Syncytial Virus Antigens.

2nd Place Nicholas J.C. Brown/ATU
Breeding Response of Indigo Buntings (*Passerina cyanea*) to Oak-Woodland Restoration in the Ozard National Forest.

3rd Place Jacy L. Wagnon/UAMS
ADE6-M26 mRNA Activates Meiotic Recombination at the *ADE6* GENE OF *S. pombe.*

Environmental Science Awards

1st Place J.L. McCallum/LSU–Shreveport
Use of an Urban Wetland by Waterbirds: a Baseline Study for a long-term ecological monitoring site.

2nd Place Justin M. Homan/ATU
Quantificaiton of Stream Dryness in Interior Highland Streams.

3rd Place Katherine Winsett/UofA–Fayetteville
Myxomycetes of Mississippi.

Physical Science Awards

1st Place C. Graves/ASU
A Survery on the Vertex Cover Problem.

UNDERGRADUATE STUDENT AWARDS

Life Science:

1st Place Trixie Lee/Harding University
Posthatching Yolk Reserves: The Effect of Starvation on Early Growth of *Apalone mutica* Haltchlings.

2nd Place Elizabeth C. Compton/ASU
Do Snake Skins Deter Predation of Great Crested Flycather Nests: An Artificial Nest Experiment.

3rd Place Jonathan Treece/ASU
Nicotine and Developing Autonomic Neurons of Mammalian Nervous System.

Environmental Science Awards

1st Place Michelle R. Dare/HSU
The Effect of Indigenous Villages on Coral Reef Community Structure in Kuna Yala, Panama

2nd Place Thomas P. Saul/HSU
Freshwater Sponge Community Composition and Characteristics of Occurrence of Egeria Densa in Degray Lake, Arkansas and a Report of Sponge Occurrence in Lake Ouachita, Arkansas.

Physical Science Awards

1st Place Christopher Fisher/ATU
Theory and Practicality of a Solar/Electric Car.

2nd Place Patrick McLaurin/Lyon College
Calculated Differences in the Solvation of Chiral Solutes in Chiral Solvents.

3rd Place Hunter Broadaway/ASU
Astrophysical Applications of the Nuclear Equation of State.

HONORABLE MENTIONS

Physical Science Awards

Matthew LeMay/ASU
Deposition and Characterization of Multifunctional Ferromagnetic/Optoelctronic Composite Films.

Robby Davis/ATU
Density Funtional Studies of the Structure and Bonding of Nitrosyl Metalloporphyrin Complexes..

APPENDIX B

RESOLUTIONS

BE IT RESOLVED that we, the membership of the Arkansas Academy of Science, offer our sincere appreciation to Hendrix College for hosting the 89th Annual Meeting of the Academy, held 8-9 April, 2005.

We thank the Local Arrangements Committee: Chair, Joyce Hardin, Pradip Bandyopadhyay, Mike Bell, Laura Conley, Jennifer Dearolf, Andrea Duina, Linda Gatti-Clark, Jennifer Gilley, Darby Grace, Bruce Haggard, Joe Lombardi, Matt Moran, Rick Murray, Rachel Rein, Jennifer Roller, Mark Sutherland, and all of the student workers and staff who collectively contributed to such a successful meeting. Special appreciation is extended to Becky Neis for her work on the design of the program cover.

Appreciation is expressed for inviting us to this gorgeous campus and for use of these excellent facilities and the hospitality shown to us by Hendrix personnel. We especially thank our keynote speaker Dr. Gary W. Barrett for his thought provoking presentation entitled "Challenges of Integrative Science."

We thank Hendrix College for their contributions to the Social and Banquet, which were both excellent and thoroughly enjoyed by all. And we thank Provost Dr. Bob Entzminger for his warm welcome. We sincerely appreciated and enjoyed the fine music provided by the Student Jazz Band.

The Academy recognizes the important roles assumed by session chairs and expresses sincere appreciation to Scott Austin, Leo Carson Davis, Jennifer Dearolf, James Engman, Gabe Ferrer, Anthony Grafton, Phoebe Harp, Mostafa Hemmati, Robert Kissell, Mike Rapp, Henry Robison, Brett Serviss, Bill Shepherd, Wayne Wahls, Richard Walker, and T. Yamashita.

A special appreciation is owed to those individuals who devoted considerable time and energy to judging student papers. They are Scott Austin, Betty Crump, Paul Doruska, Gabe Ferrer, Barry Gehm, Joe Guenter, Mostafa Hemmati, Malcolm McCallum, Mike Rapp, Blake Sasse, Malathi Srivatsan and Deborly Wade.

We gratefully acknowledge the various directors of the science and youth activities which are supported or supervised by the Academy: Mostafa Hemmati, Science Education Committee; Jim Murry, Intel Talent Search; Jerry Manion, Junior Academy of Science; and Linda Kondrick, Junior Science and Humanities Association.

We wish to thank all those who served as directors at Regional Science Fairs and Junior Academy meetings including Bryan DeBusk, Jim Edson, Lynne Hehr, Tillman Kennon, Brian Monson, Mike Rapp, Kathyrn Shinn and Gus Williamson.

We congratulate all who presented papers and posters at this meeting. Student participants are especially recognized since their efforts contribute directly to the future success of the Academy and the improvement and advancement of science in Arkansas.

We very much appreciate Walt Godwin for maintaining the Academy website.

The continued success of the Academy is due to its strong leadership. We offer sincere thanks to our officers for another excellent year: Betty Crump (President), Stan Trauth (President-Elect), David Saugey (Vice-President), Wayne Gray (Past-President), Jeff Robertson, (Secretary and Newsletter Editor), Joyce Hardin (Treasurer), Stan Trauth (*Journal* Editor-in-Chief), Chris McAllister (*Journal* Managing Editor), and Henry Robison (Historian).

Finally, the membership wishes to posthumously recognize Tom Palko for his many years of service and contributions to his students, the Academy, and to the science and biology profession.

Respectfully submitted this 9thday of April, 2005
Resolution Committee
David Saugey, Chair
Joyce Hardin
Mostafa Hemmati

MEMBERS 2006

LIFE MEMBERS

FIRST MI	LASTNAME	INSTITUTIONS
Robbin C.	Anderson	University of Arkansas-Fayetteville
Edmond J.	Bacon	University of Arkansas-Monticello
Vernon	Bates	Ouachita Mtns. Biological Station
Floyd	Beckford	Lyon College
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Linda	Gatti-Clark	Hendrix College
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Stephen	Sewell	University of Mississippi
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Michael	Rapp	University of Central Arkansas
Henry	Robison	Southern Arkansas University
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Betty	Speairs	Ouachita Mtns. Biological Station
Richard	Speairs	Ouachita Mtns. Biological Station
George	Templeton	University of Arkansas-Fayetteville
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Renn	Tumlison	Henderson State University
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REGULAR MEMBERS

FIRST MI	LASTNAME	INSTITUTIONS
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William R.	Bowen	Jacksonville State Univ. (AL-retired
Morris	Bramlett	UAM
Tom	Buchanan	University of Arkansas-Ft. Smith
Alan	Christian	Arkansas State University
Amanda	Crnkovic	Ouachita Mountains biological Station
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Jennifer	Dearolf	Hendrix College
Paul	Doruska	Arkansas Forest resources Center-UAM
Andrea	Duina	Hendrix College
Jane	Duina	Henderson State University
Rudolph	Eichenberger	Southern Arkansas University
Roberts	Ficklin	UAM
Thomas	Foti	Arkansas Natural Heritage Commission
Jonathan	Fuller	TAMU-Texarkana
Barry	Gehm	Assistant Professor
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Anthony	Grafton	Lyon College
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Laurence	Hardy	Ouachita Mountains biological Station
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Philip	Hyatt	US Forest Service
Abul	Kazi	UAPB
Tillman	Kennon	ASU
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Robert E.	Kissell Jr.	Uninversity of Arkansas-Monticello
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Scott	McConnell	UAM
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Blake	Sasse	Arkansas Game and Fish Com.
Bill	Shepherd	
David	Simons	Ouachita Mtns. Biological Station
Malathi	Srivatsan	Arkansas State University
Richard	Standage	USDA Forest Service
Philip A.	Tappe	University of Arkansas- Monticello
Bruce	Tedford	Arkansas Tech University
Jonathan	Treece	ASU
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Richard	Walker	UAPB
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Benjamin	Wheeler	Arkansas State University
William	Willingham	UAPB
Tsunemi	Yamashita	Arkansas Tech University
Douglas	Zollner	The Nature Conservancy

SUSTAINING MEMBERS

FIRST MI	LASTNAME	INSTITUTIONS
David L.	Davies	University of Ark./Medical Sciences

STUDENTS MEMBERS

Eileen	Banach	OMBS
Velbona	Boshan	Lyon College
Hunter	Broadaway	ASU-Jonesboro
Jeff	Brooks	University of Louisianna-Monroe
Nicholas	Brown	Arkansas Tech University
Christopher	Bryant	UA-Monticello
Stephanie	Bunch	UA-Monticello
Misty	Caple	TAMU-Texarkana
Elizabeth	Compton	
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Selma	Dagtas	UA-Monticello
Michelle	Dare	Henderson State University
Pupali	Das	UA-Monticello
Robby	Davis	Arkansas Tech University
Mark	DeWall	UA-Monticello
Ryan	Ewing	UT-Austin
Christopher	Fisher	Arkansas Tech University
Johnathan	Fuell	Henderson State University
Jonathan	Fuller	TAMU-Texarkana
Tobin	Fulmer	Henderson State University
Jun	Gao	UA-MonticelloS
Nicholas	Girondo	Arkansas Tech University
Adam	Green	UA-Monticello
Joe	Guenter	UA-Monticello
David	Harlan	ASU-Jonesboro
Jonathan	Hartley	UA-Monticello
Carrie	Hawkins	UA-Fayetteville
Waylon	Hiler	ASU-Jonesboro
Justin	Homan	Arkansas Tech University
Matthew	Hurd	UA-Monticello
Arvind	Jamakhandi	UA-Monticello
Erin	Knoll	Arkansas Tech University
Trixie	Lee	Harding
Matthew	LeMay	ASU-Jonesboro
Jamie	McCallum	LSU-Shreveport
Matthew	McConnell	UA-Monticello
Sam	McCord	ASU-Jonesboro
Patrick	McLaurin	Lyon College
Laura	McWilliams	Lyon College
Christy	Melhart	UA-Fayetteville
Joe	Milanovich	ASU-Jonesboro
Willie	Nelson	Arkansas Tech University
Yang	Ou	UA-Monticello
Tess	Perez	SAU
Elizabeth	Pope	Henderson State University
Jennifer	Roller	Hendrix College
Omar	Sabbarini	UA-Fayetteville
Thomas	Saul	Henderson State University
Christy	Schuchardt	Lyon College
Jason	Self	ASU-Jonesboro
Brian	Sepko	UA-Fayetteville
Harish	Shandilya	UA-Monticello
Alex	Skorcz	ASU-Jonesboro
C.J.	Spurlock	Lyon College
Nathan	Stephens	ASU-Jonesboro
Brandon	Thurow	Hendrix College
Jonathan	Treece	ASU-Jonesboro
Jacy	Wagnon	UA-Monticello
Toby	Ward	UA-Monticello
Bradley	Williams	UCA
Katherine	Winsett	UA-Fayetteville
Billy	Yancey	ASU-Jonesboro
Ashley	Young	UA-Monticello
Lauren	Young	UA-Monticello

HENDRIX

Arkansas Academy of Science
89th Annual Meeting
April 8-9, 2005

Welcome

On behalf of Hendrix College, welcome to the 89th meeting of the Arkansas Academy of Science. We are delighted to have you here and hope that you will take time to enjoy our campus. Hendrix has a tradition of excellence in the sciences from outstanding staff and students to modern facilities. This meeting brings the intellectual efforts of scientists across Arkansas to our campus so our community can participate in the engaging and active process of sharing scientific knowledge. We wish you a successful meeting.

J. Timothy Cloyd, Ph.D.
President, Hendrix College

Welcome to the Arkansas Academy of Science's 89th Annual Meeting. Thank you all for your attendance, participation and involvement in the AAS as well as the Arkansas Junior Academy of Science. We are proud to have renewed our association with and support of the AJAS. We are looking forward to the diverse presentations over the next two days, as well as the chance to visit with friends and colleagues that we seldom get to see otherwise. On behalf of the AAS membership, I want to extend our appreciation to Dr. Joyce Hardin and Hendrix College for hosting this year's annual meeting.

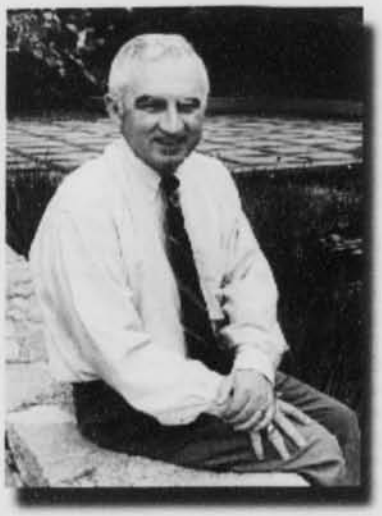
Betty G. Crump
President, Arkansas Academy of Science

2005 AAS Local Arrangements Committee

Dr. Pradip Bandyopadhyay, Mr. Mike Bell, Ms. Laura Conley, Dr. Jennifer Dearolf, Dr. Andrea Duina, Dr. Linda Gatti-Clark, Ms. Jennifer Gilley, Ms. Darby Grace, Dr. Bruce Haggard, Dr. Joyce Hardin, Dr. Joe Lombardi, Dr. Matt Moran, Dr. Rick Murray, Ms. Rachel Rein, Ms. Jennifer Roller, and Dr. Mark Sutherland.

The committee would like to thank the members of Hendrix Biological Society and
Dr. Bob Entzminger, Provost of Hendrix College, for their support.

Keynote Address



Gary W. Barrett was born and raised on a farm in Princeton, Indiana, attended a one-room school house, and spent most of his early days in this rural setting. He received a B.S. in biology from Oakland City University in 1961, an M.S. in biology from Marquette University in 1963, and a Ph.D. in zoology/ecology from the University of Georgia in 1967 under the guidance of the legendary ecologist Eugene Odum. Dr. Barrett spent one year teaching at Drake University before moving to the University of Miame, Ohio in 1968. There he worked for 26 years, eventually rising to the level of distinguished professor. In 1994 he moved to the University of Georgia where today he holds the Odum Professorship of Ecology. His research interests include landscape ecology, restoration ecology, agroecosystems, and ecological education. He served as the ecology program director for the National Science Foundation from 1981-83, president of the United States section of the International Association for Landscape Ecology from 1998-99, president of the American Institute of Biological Sciences in 1998, and president of the Association for Ecosystem Research Centers from 1995-1996. This year, he is receiving the annual award from the Center for Undergraduate Research Opportunities for his outstanding work with undergraduate students at Georgia. Dr. Barrett has made numerous contributions to the field of ecology and mentored many undergraduate and graduate students. He has authored five books and over 160 publications. In 2005, he coauthored with the late Eugene Odum, the 5th edition of *Fundmentals of Ecology*, one of the premier textbooks in the ecological field.

PROGRAM

Arkansas Academy of Science

89th Annual Meeting

April 8-9, 2005

Hendrix College

SCHEDULE OF EVENTS

Friday morning
- 10:00 a.m.- 4:00 p.m. Registration. West Lobby of DW Reynolds.
- 10:00 a.m.-11:30 a.m. Academy executive meeting. DW Reynolds 130.
- 10:00 a.m. Poster setup. DW Reynolds Rooms 011 and 012.
- 11:30 a.m.-12:30 p.m. Annual meeting of the advisors and directions of the Ouachita Mountains Biological Station. DW Reynolds 137.

Friday afternoon
- 12:15 p.m. Judges Meeting. DW Reynolds Room 206/208.
- 1:00 p.m. - 2:45 p.m. Oral presentations. See program for locations.
- 2:45 p.m.-3:15 p.m. Coffee break/Posters. DW Reynolds Rooms 011 and 012.
- 3:15 p.m.-5:00 p.m. Oral presentations resume. See program for locations.
- 5:00 p.m.-6:00 p.m. First business meeting. DW Reynolds Room 010.
- 6:00 p.m.-7:00 p.m. Keynote speech. Trieschman Theatre.
- 7:00 p.m.-9:00 p.m. Mixer and banquet. Cottage on Washington Avenue.

Saturday morning
- 7:30 a.m.-8:00 a.m. Coffee and donuts. DW Reynolds Rooms 011 and 012.
- 8:00 a.m.-9:00 a.m. Registration. West Lobby of DW Reynolds.
- 8:00 a.m.-9:15 a.m. Oral presentations. See program for locations.
- 9:15 a.m.-9:45 a.m. Coffee break/Posters. DW Reynolds 011 and 012.
- 9:45 a.m.-11:00 a.m. Oral presentations resume. See program for locations.
- 11:00 a.m.-12:00 p.m. Poster breakdown.
- 12:00 p.m. Adjourn.

SECTION PROGRAMS

*** Undergraduate **Graduate**

SCHEDULE OF ORAL PRESENTATION SESSIONS

		DW Rey. 008	DW Rey. 010	DW Rey. 013	DW Rey. 137
Friday	Session I 1:00 – 2:45 pm	Biochemistry/ Cell Biology	Vertebrate Biology I	Invertebrate Biology I	Engineering/ Computer Sci. I
	Session II 3:15 – 5:00 pm	Molecular Biology	Vertebrate Biology II	Invertebrate Biology II	Environmental Sciences I
Saturday	Session III 8:00 – 9:15 am	Physics/ Astronomy	Zoology	Botany I	Engineering/ Computer Sci.II
	Session IV 9:45 – 11:00 am	Chemistry	Myxomycetes/ Education	Botany II	Environmental Sciences II

ORAL PRESENTATIONS

(Speakers' Underlined)

Session I: Friday April 8, 2005, 1:00 pm – 2:45 pm

Biochemistry/Cell Biology DW Reynolds Room 008

1:00 pm **SYNTHESIS AND CHARACTERIZATION OF RUTHENIUM COMPLEXES FOR BIOLOGICAL ELECTRON TRANSFER STUDIES.** Anwar A. Bhuiyan[1], Ryan W. Dossey[2], Bill Durham[2] and Francis S. Millett[2], [1]Arkansas Tech University, Russellville, Arkansas 72801, [2]University of Arkansas, Fayetteville, Arkansas 72701

1:15 pm **REACTIVITY OF THIAZOLIDINES TOWARDS PEROXY-NITRITE.** Richard B. Walker, Department of Chemistry and Physics, University of Arkansas at Pine Bluff, Pine Bluff, Arkansas 71611 Stephen C. Grace, Department of Biology, University of Arkansas at Little Rock, Little Rock, Arkansas 72204

1:30 pm **THE SUPEROXIDE DISMUTASE-LIKE ACTIVITY OF COPPER (II) 3,5-DIBROMOSALICYLATE.** Johnmesha Sanders[1], Kendra Christian[2], Grant Wangila[1], and William Willingham[1], [1]Walker Center for Multi-Purpose Research and Sponsored Programs, University of Arkansas at Pine Bluff, Pine Bluff, Arkansas 71601 and [2]Pine Bluff High School, Pine Bluff, Arkansas 71601

1:45 pm **TRANSPOSON MUTAGENESIS OF DETERGENT DEGRADING BACTERIA.** Rachel Trana, Holly Strickland and Russell Nordeen, School of Mathematical and Natural Sciences P.O. Box 3480 Monticello, Arkansas 71656

2:00 pm **DECLINE IN PROTEASOMAL CATALYTIC ACTIVITY ACCOUNTS FOR AGE- RELATED DECREASE IN IMMUNE FUNCTION, WHICH IS REVERSIBLE BY PHENOLIC ANTI-OXIDANT 3H[1], 2-DITHIOLE 3-THIONE.** Rupali Das*, Subramaniam Ponnappan ‡ and Usha Ponnappan *‡. *Department of Microbiology and Immunology, ‡Department of Geriatrics, University of Arkansas for Medical Sciences (UAMS), CAVHS, 4300, West 7th Street, Little Rock, Arkansas 72205.

2:15 pm **CD4$^+$ T CELL ANERGY IS ASSOCIATED WITH INCREASED LEVELS OF p21^{Cip1} AND DECREASED ACTIVITY OF AP-1 AND NF- B.** A. Selma Dagtas, Kathleen Gilbert, Department of Microbiology and Immunology, University of Arkansas for Medical Sciences, Little Rock, Arkansas 72205

2:30 pm **NICOTINE AND DEVELOPING AUTONOMIC NEURONS OF MAMMALIAN NERVOUS SYSTEM.** Jonathan Treece, B.M. Prabhu and Malathi Srivatsan, Department of Biological Sciences, Arkansas State University, Jonesboro, Arkansas 72401

Vertebrate Biology I DW Reynolds Room 010

1:00 pm **SECOND REPORT OF THE SOUTHERN PAINTED TURTLE, *CHRYSEMYS DORSALIS* (TESTUDINES: EMYDIDAE), FROM TEXAS, WITH COMMENTS ON ITS GENETICS.** Jonathan P. Fuller[1], Chris T. McAllister[1], and Michael R. J. Forstner[2]. [1]Department of Biology, Texas A&M University-Texarkana, Texarkana, Texas 75505; [2]Department of Biology, Texas State University, San Marcos, Texas 78666.

1:15 pm **CAPTURE PATTERNS AND DISTRIBUTION RECORDS FOR BOTTOMLAND BAT SPECIES IN ARKANSAS' DELTA REGION.** Stephen C. Brandebura, Bobby H. Fokidis, and Thomas S. Risch. Arkansas State University, Department of Biological Sciences, P. O. Box 599, State University, Arkansas 72467

1:30 pm **TROPHIC INTERACTIONS BETWEEN A SPECIES IN DECLINE, THE OZARK HELLBENDER (*CRYPTOBRANCHUS ALLEGANIENSIS BISHOPI*), AND A PREY BASE COMPRISED OF MULTIPLE CRAYFISH SPECIES USING STABLE ISOTOPE ANALYSIS.** Waylon R. Hiler[1], Benjamin A. Wheeler[2], Stanley E.Trauth[1], and Alan D. Christian[1]; [1]Department of Biological Sciences, Arkansas State University, P.O. Box 599, State University, Arkansas 72467; [2]Environmental Sciences Program, Arkansas State University, P.O. Box 847, State University, Arkansas 72467

1:45 pm **COMPARISON OF ABNORMALITY RATES IN THE OZARK HELLBENDER (*CRYPTOBRANCHUS ALLEGANIENSIS BISHOPI*) FROM THE SPRING RIVER, FULTON COUNTY, ARKANSAS.** Waylon R. Hiler[1], Benjamin A. Wheeler[2], and Stanley E. Trauth[1]; Department of Biological Sciences, Arkansas State University, P.O. Box 0599, State University, Arkansas 72467-0599; [2]Environmental Sciences Program, Arkansas State University, P.O. Box 847, State University, Arkansas 72467

2:00 pm **USE OF THE BIOMARK® TAGGING SYSTEM ON THE OZARK HELLBENDER, *CRYPTOBRANCHUS ALLEGANIENSIS BISHOPI* (AMPHIBIA: CAUDATA), IN NORTHERN ARKANSAS.** Benjamin A. Wheeler[1], Stanley E. Trauth[2], Waylon R. Hiler[2], and Chris T. McAllister[3]. [1]Environmental Sciences Program, Arkansas State University, State University, Arkansas 72467; [2]Department of Biological Sciences, Arkansas State University, State University, Arkansas 72467; [3]Department of Biology, Texas A&M University-Texarkana, Texarkana, Texas 75505.

2:15 pm **COMPARISON OF THE REACH SCALE HABITAT CHARACTERISTICS OF HISTORIC AND CURRENT OZARK HELLBENDER (*CRYPTOBRANCHUS ALLEGANIENSIS BISHOPI*) LOCALITIES USING STANDARDIZED ASSESSMENT PROTO – COOLS.** Benjamin A. Wheeler[1], Waylon R. Hiler[2], Stan E. Trauth[2], and Alan D. Christian. [1]Environmental Sciences Program, Arkansas State University. [2]Department of Biological Sciences, Arkansas State University

2:30 pm **SEASONAL INCIDENCE OF SPERM WITHIN SPERMATHECAE OF THE OUACHITA DUSKY SALAMANDER (*DESMOGNATHUS BRIMLEYORUM*) IN ARKANSAS.** Stanley E. Trauth and Michelle N. Mary, Department of Biological Sciences, Arkansas State University, P.O. Box 599, State University, Arkansas 72467-0599

Invertebrate Biology I DW Reynolds Room 013

1:00 pm **A PRELIMINARY REPORT ON INSECTS INHABITING POCKET GOPHER BURROW IN ARKANSAS.** Peter W. Kovarik. 239 Crestview Road, Columbus, Ohio 43202. Stephen W. Chordas III. The Ohio State University, 1063 West 2nd Avenue, Columbus Ohio 43212. Eric G. Chapman. Department of Biological Sciences, Kent State University, Kent, Ohio 44242. Gary A. Heidt. Department of Biology University of Arkansas at Little Rock 2801 S. University Little Rock, Arkansas 72204. Henry W. Robison. Department of Biology, Southern Arkansas University, P. O. Box 9354, Magnolia, Arkansas 71754-9354.

1:15 pm **FIFTY-FIVE NEW ADDITIONS TO THE TRUE BUG (HEMIPTERA) FAUNA OF ARKANSAS.** Stephen W. Chordas III. The Ohio State University, 1063 West 2nd Avenue, Columbus Ohio 43212. Henry W. Robison. Department of Biology, Southern Arkansas University, P. O. Box 9354, Magnolia, Arkansas 71754-9354. Eric G. Chapman. Department of Biological Sciences, Kent State University, Kent, Ohio 44242. Betty G. Crump. Forest Service, P.O. Box 1270, Hot Springs, Arkansas 71902. Peter W. Kovarik. 239 Crestview Road, Columbus, Ohio 43202.

1:30 pm **CHECKLIST OF SUBTERRANEAN AMPHIPODA OF ARKANSAS.** G.O. Graening, Michael E. Slay, and John R. Holsinger. Arkansas Field Office, The Nature Conservancy, 601 North University Avenue, Little Rock, Arkansas 72205 and Department of Biological Sciences, Old Dominion University, Norfolk, Virginia 23529.

1:45 pm **HEPATOZOON INFECTIONS IN ARKANSAS SNAKES.** James J. Daly Sr., Charles H. Calhoun, Robert C. McDaniel and James W. Townsend, Departments of Microbiology and Immunology, and Pathology, University of Arkansas for Medial Sciences, Little Rock, Arkansas 72205.

2:00 pm **AQUATIC MACROINVERTEBRATES ASSOCIATED WITH *OPHIOGOMPHUS WESTFALLI* (ODONATA: GOMPHIDAE) IN MISSOURI OZARK STREAMS.** George L. Harp, Phoebe A. Harp and Sam McCord. Dept. of Biological Sciences, Arkansas State University, State University, Arkansas 72467.

2:15 pm **INCREASED HABITAT HETEROGENITY: EFFECTS ON MACROINVERTEBRATE BIOMASS AND DISTRIBUTION PATTERNS IN A SHALLOW EUTROPHIC RESERVOIR.** Joseph M. Shostell and Bradley S. Williams. Biology Department, Penn State University, Uniontown, Pennsylvannia 15401-0519, USA. Biology Department, University of Central Arkansas, Conway, Arkansas 72035-5003, USA.

Engineering/Computer Sci. I DW Reynolds Room 137

1:00 pm **PHOTOVOLTAIC AND PHOTOCONDUCTIVE PROPERTIES OF DOUBLE LAYER SEMICONDUCTOR / POLYMER COMPOSITE FILMS.** David Harlan, Robert Engelken, and Matthew LeMay. Optoelectronic Materials Research Laboratory, College of Engineering, Arkansas State University, P.O. Box 1740, State University, Arkansas 72467.

1:15 pm **DEPOSITION AND CHARACTERIZATION OF MULTI-FUNCTIONAL FERROMAGNETIC/OPTOELCTRONIC COMPOSITE FILMS**. Matthew LeMay, Robert Engelken, and David Harlan. Optoelectronic Materials Research Laboratory, College of Engineering, Arkansas State University, P.O. Box 1740, State University, Arkansas 72467.

1:30 pm **A NEW CITRATE-BASED SOLUTION FOR CHEMICAL BATH DEPOSITION OF NONHAZARDOUS BISMUTH (III) SULFIDE FILMS**. Demetrick Warren, Robert Engelken, David Harlan, and Matthew LeMay. Optoelectronic Materials Research Laboratory, College of Engineering, Arkansas State University, P.O. Box 1740, State University, Arkansas 72467.

1:45 pm **GROWTH RATE CALCULATIONS FOR ELECTRONIC DEVICE PRODUCTION**. Willie Nelson, Alvin Ong, and Daniel Bullock*, Department of Physical Science, Arkansas Tech University, Russellville, Arkansas 72801. *CONTACT PERSON

2:00 pm **A SURVEY ON THE VERTEX COVER PROBLEM.** A. G. Dhakne B. D. Freeman C. Graves. Department of Computer Science, Arkansas State University, P.O. Box 9, State University, Arkansas 72467.

2:15 pm **THEORY AND PRACTICALITY OF A SOLAR/ELECTRIC CAR.** Christopher Fisher, M. Hemmati, Dwayne Ahrens, and Don White. Department of Physical Sciences, Arkansas Tech University, Russellville, Arkansas 72801

Session II: Friday April 8, 2005, 3:15 pm – 5:00 pm

Molecular Biology DW Reynolds Room 008

3:15 pm **DEVELOPMENT OF A RECOMBINANT VARICELLA VACCINE THAT EXPRESSES SIMIAN IMMUNODEFICIENCY VIRUS ANTIGENS.** Yang Ou, and Wayne L. Gray. Dept. of Microbiology and Immunology, University of Arkansas for Medical Sciences, Little Rock, Arkansas 72205

3:30 pm **CONSTRUCTION OF RECOMBINANT VARICELLA VACCINES EXPRESSING RESPIRATORY SYNCYTIAL VIRUS ANTIGENS.** Toby M. Ward, Kara A. Davis, Wayne L. Gray.

3:45 pm **MULTIPLE FUNCTIONAL DOMAINS OF MEIOTIC REGULAR PROTEIN ATF1 IN FISSION YEAST S. POMBE.** Jun Gao, Mari K. Davidson, Gloria Glick, and Wayne P. Wahls. Department of Biochemistry and Molecular Biology, University of Arkansas for Medical Sciences, Little Rock, Arkansas 72205, U.S.A.

4:00 pm ***ADE6-M26* mRNA ACTIVATES MEIOTIC RECOMBINATION AT THE ADE6 GENE OF S. POMBE.** Jacy L. Wagnon, Mari K. Davidson, and Wayne P. Wahls. Department of Biochemistry and Molecular Biology, University of Arkansas for Medical Sciences, Little Rock, Arkansas 72205

4:15 pm **ZIP1 PROTEIN BINDS TO THE M26 DNA SITE TO MEDIATE HIGH COPY SUPPRESSION OF *ATF1* NULL MUTANT PHENOTYPES.** Harish K. Shandilya, Mari K. Davidson & Wayne P.Wahls. Department of Biochemistry & Molecular Biology, Slot#516 University of Arkansas for Medical sciences, Little Rock, Arkansas 72211.

4:30 pm **EVIDENCE THAT THE DISTRIBUTION OF THE ELONGATION FACTOR SPT16 OVER TRANSCRIBED GENES IS DEPENDENT UPON HISTONE H3 INTEGRITY IN YEAST.** Andrea A. Duina[1], and Fred Winston[2]. [1]Hendrix College, Biology Department, Conway, Arkansas 72032. [2]Harvard Medical School, Department of Genetics, Boston, Massachusetts 02115

4:45 pm **A TANDEM AFFINTY PURIFICATION APPROACH TO IDENTIFY RECOMBINATION PROTEIN COMPLEXES.** K. Mark DeWall and Wayne P. Wahls. Department of Biochemistry and Molecular Biology, University of Arkansas for Medical Sciences, Little Rock, Arkansas 72205

Vertebrate Biology II DW Reynolds Room 010

3:15 pm **SURVEY OF SMALL MAMMALS IN ARKANSAS VIA DISSECTION OF BARN OWL (*TYTO ALBA*) PELLETS.** Ewing, L. E., S. R. Thomas and G. A. Heidt. Department of Biology, University of Arkansas at Little Rock, Little Rock, Arkansas 72204

3:30 pm **DO SNAKE SKINS DETER PREDATION OF GREAT CRESTED FLYCATCHER NESTS: AN ARTIFICIAL NEST EXPERIMENT.** Elizabeth C. Compton, and Thomas S. Risch. Department of Biological Sciences, Arkansas State University, 117 S Caraway, State University, Arkansas 72467

3:45 pm **POSTHATCHING YOLK RESERVES: THE EFFECT OF STARVATION ON EARLY GROWTH OF *APALONE MUTICA* HATCHLINGS.** Trixie Lee, Michael V. Plummer, and Nathan E. Mills, Department of Biology, Box 12251, Harding University, Searcy, Arkansas 72149 USA

4:00 pm **BREEDING RESPONSE OF INDIGO BUNTINGS (*PASSERINA CYANEA*) TO OAK-WOODLAND RESTORATION IN THE OZARK NATIONAL FOREST.** Nicholas J.C. Brown and Christopher Kellner, Arkansas Tech University, Russellville, Arkansas 72801

4:15 pm **A SONGBIRD INVENTORY OF ARKANSAS POST NATIONAL MEMORIAL.** Philip A. Tappe, School of Forest Resources, Arkansas Forest Resources Center, University of Arkansas, Monticello, Arkansas 71656

4:30 pm **THE DECLINE OF NATURAL HISTORY STUDY IN HERPETOLOGY: AN ANALYSIS OF HISTORICAL PUBLICATION PATTERNS IN TWO JOURNALS.** McCallum, M.L. and J.L. McCallum. Department of Biology, Louisiana State University in Shreveport, Shreveport, Louisiana 71115.

4:45 pm **POPULATION RESPONSE TO HARVEST OF WHITE-TAILED DEER ON CHOCTAW ISLAND WILDLIFE MANAGEMENT AREA.** Robert E. Kissell, Jr., School of Forest Resources, Arkansas Forest Resources Center, University of Arkansas, Monticello, AR 71656. Philip A. Tappe, School of Forest Resources, Arkansas Forest Resources Center, University of Arkansas, Monticello, Arkansas 71656

Invertebrate Biology II DW Reynolds Room 013

3:15 pm **SCANNING ELECTRON MICROSCOPY OF THE GONOPODS OF THE MILLIPED, *THRINAXORIA LAMPRA* (CHAMBERLIN) (POLYDESMIDA: XYSTODESMIDAE).** Chris T. McAllister[1], Rowland M. Shelley[2], and Stanley E. Trauth[3]. [1]Department of Biology, Texas A&M University-Texarkana, Texarkana, TX 75505; [2]Research Lab, North Carolina State Museum of Natural Sciences, Raleigh, North Carolina 27607; and [3]Department of Biological Sciences, Arkansas State University, State University, Arkansas 72467.

3:30 pm **SECOND RECORD OF THE DIPLURAN, *OCCASJAPYX CARLTONI* ALLEN, 1988 (INSECTA: JAPYGIDAE), FROM ARKANSAS.** Chris T. McAllister[1] and Christopher Carlton[2]. [1]Department of Biology, Texas A&M University-Texarkana, Texarkana, TX 75505; and [2]Louisiana State Arthropod Museum, Louisiana State University, Baton Rouge, Louisiana 70803.

3:45 pm **DISTRIBUTION OF TARANTULAS (*APHONOPELMA*) IN ARKANSAS: RESULTS OF A CITIZEN-SCIENCE SURVEY.** Michael D. Warriner, Arkansas Natural Heritage Commission, 1500 Tower Building, 323 Center Street, Little Rock, Arkansas 72201

4:00 pm **PHYLOGEOGRAPHY OF THE STRIPED SCORPION, *CENTRUROIDES VITTATUS*, IN THE SOUTHWESTERN UNITED STATES.** Tsunemi Yamashita, Maria Longing, and Nick Pridgin, Department of Biological Sciences, Arkansas Tech University, Russellville, Arkansas 72801

Environmental Sciences I DW Reynolds Room 137

3:15 pm **ALTERNATE PARADIGMS FOR FOREST AND FOREST PRODUCT UTILIZATION.** Jason Self*, Dr. Robert Engelken*,**, David Harlan**, Matthew LeMay**. Environmental Science Ph.D. Program* and College of Engineering**, Arkansas State University, P.O. Box 1740, State University, Arkansas 72467

3:30 pm **THE EFFECT OF INDIGENOUS VILLAGES ON CORAL REEF COMMUNITY STRUCTURE IN KUNA YALA, PANAMA.** Michelle R. Dare, Elizabeth F. Pope and James A. Engman. Department of Biology, Henderson State University, Box 7520, Arkadelphia, Arkansas 71999-0001.

3:45 pm **FRESHWATER SPONGE COMMUNITY COMPOSITION AND CHARACTERISTICS OF OCCURRENCE ON *EGERIA DENSA* IN DEGRAY LAKE, ARKANSAS, AND A REPORT OF SPONGE OCCURRENCE IN LAKE OUACHITA, ARKANSAS.** Thomas P. Saul, Michelle R. Dare and James A. Engman. Department of Biology, Henderson State University, Box 7520, Arkadelphia, Arkansas 71999-0001.

4:00 pm **QUANTIFICATION OF STREAM DRYNESS IN INTERIOR HIGHLAND STREAMS.** Justin M. Homan, Nicholas M. Girondo, and Charles J. Gagen. Arkansas Tech University, Fisheries and Wildlife Biology Program, 1701 N. Boulder Ave., Russellville, Arkansas 72081

4:15 pm **EFFECTS OF HERBICIDE APPLICATION ON FOLIAR MORPHOLOGY AND NUTRIENT CONCENTRATIONS IN MID-ROTATION PINE PLANTATIONS.** Prabudhda Dahal and Hal O. Liechty, Program Technician and Associate Professor, respectively; School of Forest Resources, P.O. Box 3468, Monticello, Arkansas 71656.

4:30 pm **A CATALOG OF ALTERNATIVE CONCEPTIONS OF THE TSUNAMI OF 26 DECEMBER 2004.** Stephen K. Boss, Department of Geosciences, 113 Ozark Hall, University of Arkansas, Fayetteville, AR 72701 e-mail: sboss@uark.edu. Caroline Beller, School of Teaching & Curriculum Leadership, 227 Willard Hall, Oklahoma State University, Stillwater, Oklahoma 74078 e-mail: beller@okstate.edu.

4:45 pm **INTEGRATING SUPERVISED AND UNSUPERVISED CLASSIFICATION METHODS TO DEVELOP A MORE ACCURATE LAND COVER CLASSIFICATION.** Donald I. M. Enderle and Robert C. Weih, Jr., Spatial Analysis Laboratory (SAL), University of Arkansas at Monticello, Arkansas Forest Resources Center, School of Forest Resources, 110 University Court, Monticello, Arkansas 71656, Email: weih@uamont.edu, Phone: 870-460-1248, Fax: 870-460-1092

Session III: Saturday April 9, 2005, 8:00 am – 9:15 am

Physics/Astronomy DW Reynolds Room 010

8:00 am **A PURE DIPOLE MODEL FOR SPHERICAL RARE EARTH MAGNETS.** Alois J. Adams, Department of Physics and Astronomy, University of Arkansas at Little Rock, 2801 South University Avenue, Little Rock, Arkansas 72204-1099

8:15 am **SERENDIPITOUSLY DISCOVERED ECLIPSING-NEAR CONTACT BINARY HH95-79 IN AURIGA.** Scott Austin, University of Central Arkansas, Department of Physics and Astronomy, Conway, Arkansas 72035. Jeff Robertson, Arkansas Tech University, Department of Physical Sciences, Russellville, AR 72801. Tut Campbell, Whispering Pines Observatory, 7021 Whispering Pines Road, Harrison, Arkansas 76201

8:30 am **NEWLY DISCOVERED PULSATING VARIABLE IN ANDROMEDA.** Jeff W. Robertson. Arkansas Tech University, Department of Physical Sciences, 1701 North Boulder, Russellville, Arkansas 72801-2222

8:45 am **UU AQR ECLIPSES DURING 2003.** Jeff W. Robertson, Josh A. Higgins, R. Tut Campbell. Arkansas Tech University, Department of Physical Sciences, 1701 North Boulder, Russellville, Arkansas 72801-2222

9:00 am **ASTROPHYSICAL APPLICATIONS OF THE NUCLEAR EQUATION OF STATE.** Hunter Broadaway and Bao-An Li. Department of Chemistry and Physics, Arkansas State University, State University, Arkansas 72467

Zoology DW Reynolds Room 010

8:00 am **IDENTIFICATION OF CYSTACANTHS OF THE FAMILY OLIGACANTHORHYNCHIDAE (ACANTHOCEPHALA) BASEDON PROBOSCIS AND HOOK MORPHOMETRICS.** Dennis J. Richardson, Quinnipiac University, Box 71, 275 Mount Carmel Avenue, Hamden, Connecticut 06518

8:15 am **DISTRIBUTION AND STATUS OF *FALLICAMBARUS GILPINI* HOBBS AND ROBISON, AN ARKANSAS ENDEMIC CRAYFISH.** Henry W. Robison, Southern Arkansas University, P. O. Box 9354 SAU, Magnolia, Arkansas 71754-9354 and Brian Wagner, Arkansas Game and Fish Commission, 915 East Sevier St., Benton, Arkansas 72015

8:30 am **DISTRIBUTION AND STATUS OF THE KIAMICHI SHINER, *NOTROPIS ORTENBURGERI* HUBBS, IN ARKANSAS AND OKLAHOMA.** Henry W. Robison, Southern Arkansas University, P.O. Box 9354 SAU, Magnolia, Arkansas 71754-9354

8:45 am **A SURVEY OF THE FISHES OF THE PINE BLUFF ARSENAL, ARKANSAS.** Henry W. Robison, Southern Arkansas University, P.O. Box 9354 9354 SAU, Magnolia, Arkansas 71754-9354

9:00 am **SMALL FISH SPECIES OF ARKANSAS RESERVOIRS.** Thomas M. Buchanan, Department of Biology, University of Arkansas–Fort Smith, Fort Smith, Arkansas 72913.

Botany I DW Reynolds Room 013

8:00 am **SPECIFIC GRAVITY TRENDS IN THE LOWER PORTION OF LOBLOLLY PINE (*PINUS TAEDA* L.) PULPWOOD TREES IN SOUTHERN ARKANSAS.** Matthew B. Hurd, David W. Patterson, and Paul F. Doruska. University of Arkansas School of Forest Resources; Arkansas Forest Resources Center, PO Box 3468, Monticello, Arkansas 71656

8:15 am **GROWING SPACE AND FERTILIZATION IMPACTS ON STEM FORM OF JUVENILE LOBLOLLY PINE.** Jonathan I. Hartley, Paul F. Doruska, and Matthew B. Hurd. University of Arkansas-Monticello School of Forest Resources; Arkansas Forest Resources Center, PO Box 3468, Monticello, Arkansas 71656

8:30 am **LOBLOLLY PINE BIOMASS COROLLARIES OF SOIL ORGANIC CARBON CONTENT.** Robert L. Ficklin, Assistant Professor of Forest Soils and Ecophysiology, Arkansas Forest Resources Center, 203 Forest Resources Bldg.- UAM, Monticello, Arkansas 71656

8:45 am **INDIVIDUAL-TREE, GREEN STEM BIOMASS EQUATIONS FOR LOBLOLLY PINE PULPWOOD.** Paul F. Doruska and David W. Patterson. University of Arkansas School of Forest Resources; Arkansas Forest Resources Center, PO Box 3468, Monticello, Arkansas 71656

9:00 am **PRESETTLEMENT *PINUS TAEDA* IN THE MISSISSIPPI VALLEY ALLUVIAL PLAIN OF THE MONROE COUNTY, ARKANSAS AREA.** Don C. Bragg, USDA Forest Service, Southern Research Station, P.O. Box 3516 UAM, Monticello, Arkansas 71656

Engineering/Computer Sci. II DW Reynolds Room 137

8:00 am **MODELING OF A 4-PHASE 8/6 SWITCHED RELUCTANCE MACHINE OPERATING UNDER MULTI-PHASE EXCITATION BY UTILIZING ARTIFICIAL NEURAL NETWORKS.** Alex Skorcz, Student Researcher, 3609 Browning Cove, Jonesboro, Arkansas 72404. Chris S. Edrington, Mentor, LSW 239, College of Engineering, State University, Arkansas 72467

8:15 am **REDUCED PARTS CONVERTER FOR REALIZATION OF BIPOLAR EXCITATION FOR A 4-PHASE 8/6 SWITCHED RELUCTANCE MACHINE.** Billy Yancey III-Student Researcher, Dr. Chris S. Edrington-Mentor

8:30 am **EFFICIENCY OF TRANSMIT BEAMFORMING WITH CLOSED-LOOP BEAM CONTROL IN MOBILE WIRELESS SYSTEMS.** Brian Sepko and Wookwon Lee. Department of Electrical Engineering, University of Arkansas, 3217 Bell Engineering Center, Fayetteville, Arkansas 72701. Email: {bsepko, wookwon}@uark.edu

8:45 am **CHARACTERISTICS AND ESTIMATION OF DIRECTION-OF-ARRIVALS (DOAs) AND DIRECTION-OF-TRANSMISSION (DOTs) IN WIRELESS COMMUNICATION SYSTEMS.** Omar M. Sabbarini and Wookwon Lee. Department of Electrical Engineering, University of Arkansas, Fayetteville, Arkansas 72701. Email: {osabbar,wookwon}@uark.edu.

9:00 am **SYNCHRONIZATION AND EQUALIZATION IN MATCHED FILTER AND DFT BASED OFDM SYSTEMS.** Christopher S. Curry and Wookwon Lee. Department of Electrical Engineering, University of Arkansas, Fayetteville, Arkansas 72701. E-mail: {cscurry, wookwon}@uark.edu

Session IV: Saturday April 9, 2005, 9:45 am – 11:00 am

Chemistry DW Reynolds Room 008

9:45 am **DENSITY FUNCTIONAL STUDIES OF THE STRUCTURE AND BONDING OF NITROSYL METALLOPORPHYRIN COMPLEXES.** John P. Graham and Robby Davis, Department of Physical Science, Arkansas Tech University, Russellville, Arkansas 72801

10:00 am **CALCULATED DIFFERENCES IN THE SOLVATION OF CHIRAL SOLUTES IN CHIRAL SOLVENTS.** Patrick McLaurin, Anthony K. Grafton, and R. David Pace. Division of Science, Lyon College, PO Box 2317, Batesville, Arkansas 72501

10:15 am **GROUND STATE PREDICTION OF LINKAGE ISOMERISM OF TRANSITION METAL COMPLEXES USING QUANTUM MECHANICS.** Abul B. Kazi, Department of Chemistry and Physics, University of Arkansas at Pine Bluff, 1200 North University Drive, Pine Bluff, Arkanas 71601.

10:30 am **KINETIC STUDIES OF TRIS(2,2'-BIPYRIDINE)IRON(III) PERCHLORATE WITH COBALOXIME, $[Co(dmgBF_2)_2(H_2O)_2]$.** Grant W. Wangila[1] and Robert B. Jordan[2]. [1]Department of Chemistry and Physics, University of Arkansas at Pine Bluff, 1200 North University Drive, Mail Slot 4941, Pine Bluff, Arkansas, 71601 USA. [2]University of Alberta, Chemistry Department, Edmonton, Alberta, CANADA; T6G 2G2

10:45 am **SYNTHESIS, CHARACTERIZATION AND BIOLOGICAL ACTIVITY OF ORGANOMETALLIC RUTHENIUM COMPLEXES.** Floyd A. Beckford, Danielle Rinke and Valbona Bashari. Science Division, Lyon College, Batesville, Arkansas 72501

Myxomycetes/Education DW Reynolds Room 010

9:45 am **MYXOMYCETES OF MISSISSIPPI.** Katherine Winsett, Department of Biological Sciences, University of Arkansas, Fayetteville, Arkansas 72701

10:00 am **MOLECULAR STUDIES OF MYCETOZOANS.** Satyendra N. Rajguru, Steven L. Stephenson and Jeffrey Silberman, Department of Biological Sciences, University of Arkansas, Fayetteville, AR 72701, Jean-Marc Moncalvo and Simona Margaritescu, Center for Biodiversity and Conservation Biology, Department of Natural History, Royal Ontario Museum, Toronto, ON Canada M5S 2C6

10:15 am **MYXOMYCETES IN THE CLASSROOM.** Rodney K. Nelson, Department of Biology, University of Arkansas–Fort Smith, Fort Smith, Arkansas 72913 and Steven L. Stephenson, Department of Biological Sciences, University of Arkansas, Fayetteville, Arkansas 72703

10:30 am **BRIDGING THE GAP: BRINGING MOLECULAR DYNAMICS CALCULATIONS TO UNDERGRADUATES USING A POWERFUL, WEB-BASED INTERFACE.** Anthony K. Grafton. Division of Science, Lyon College, PO Box 2317, Batesville, Arkansas 72501

10:45 am **USING MOUSETRAP VEHICLES TO FOSTER STUDENT LEARNING IN PHYSICS.** Wilson J. Gonzalez-Espada, Assistant Professor of Physical Science, Arkansas Tech University, 1701 North Boulder Avenue, Russellville Arkansas 72801, (479) 968-0248, wilson.gonzalezespad@mail.atu.edu. Ed Roberts, Physics Teacher, Pottsville High School, 500 Apache Drive, Pottsville Arkansas 72858, (479) 968-6334, ed.roberts@pottsville.k12.ar.us.

Botany II DW Reynolds Room 013

9:45 am **PLANTS NEW TO THE ARKANSAS FLORA.** Johnathan Fuell and Brett E. Serviss. Department of Biology. Henderson State University. Arkadelphia, Arkansas 71999–0001.

10:00 am **NATURALIZATION AND EXTINCTION OF WATER HYACINTH (*EICHHORNIA CRASSIPES*) (PONTEDERIACEAE) IN SOUTH- WESTERN ARKANSAS.** Renn Tumlison and Brett Serviss, Department of Biology, Henderson State University, Arkadelphia, Arkansas 71999

10:15 am **CROWN RADIUS/DIAMETER AT BREAST HEIGHT RELATIONSHIPS FOR SIX BOTTOMLAND HARDWOOD TREE SPECIES.** Brian Roy Lockhart, U.S. Forest Service Southern Research Station, Center for Bottomland Hardwoods Research, P.O. Box 227, Stoneville, Mississippi; Robert C. Weih, Jr., School of Forest Resources, University of Arkansas, 110 University Court, Monticello, Arkansas 71656; and Keith Smith, 801 McHenry Street, Jacksonville, Arkansas 72076-6000

10:30 am **AN INVESTIGATION OF THE DIFFERENCES BETWEEN INDUSTRIAL AND NON-INDUSTRIAL PRIVATE TIMBER SALES IN ARKANSAS.** Sayeed R. Mehmood and Prabudhda Dahal, Assistant Professor and Research Technician, respectively; School of Forest Resources, P.O. Box 3468, Monticello, Arkansas 71656

Environmental Sciences II DW Reynolds Room 137

9:45 am **DEER-VEHICLE COLLISIONS IN ARKANSAS.** Philip A. Tappe, School of Forest Resources, Arkansas Forest Resources Center, University of Arkansas, Monticello, Arkansas 71656

10:00 am **PESTICIDE RESIDUES IN GUANO OF GRAY BATS IN ARKANSAS.** D. Blake Sasse, Arkansas Game and Fish Commission, #2 Natural Resources Drive, Little Rock, Arkansas 72205.

10:15 am **THE EFFECT OF PARAQUAT ON THE ANTIOXIDANT DEFENSE SYSTEM IN THE AMERICAN BULLFROG (*RANA CATESBEIANA*).** McCallum, M.L., L. Jones, D.R. Gossett, and S.W. Banks. Department of Biology, Louisiana State University in Shreveport, Shreveport, Louisiana 71115.

10:30 am **A COMPARISON OF HERPETOFAUNA COMMUNITIES IN THREE NORTHWESTERN LOUISIANA WILDLIFE MANAGEMENT AREAS.** Malcolm L. McCallum, Eric Walsh, Steven Gabry, Vic Bogosian, Jamie L. McCallum

10:45 am **USE OF AN URBAN WETLAND BY WATERBIRDS: A BASELINE STUDY FOR A LONG-TERM ECOLOGICAL MONITORING SITE.** J. L. McCallum and M. L. McCallum. Department of Biology, Louisiana State University in Shreveport, Shreveport, Louisiana 71115.

Poster Presentation Abstracts

Poster 1 **1-BUTYL-3-METHYL IMIDAZOLIUM PERBROMIDE BROMINATION OF KETONES**

Timothy Akin and R. David Pace. Lyon College, P.O. Box 2317, Batesville, Arkansas 72501

Poster 2 **SOIL EROSION AND SEDIMENT LOSS FROM COTTON FIELDS USING CONSERVATION AND CONVENTIONAL TILLAGE METHODS.**

C.K. Bryant[1], J.S. McConnell[1], and M. Mozaffari[2]. [1]University of Arkansas-Southeast Research and Extension Center, Box 3508, Monticello, Arkansas 71656. [2]University of Arkansas-Soil Testing Laboratory, Drawer 767, Marianna, Arkansas 72360

Poster 3 **SPATIAL DISTRIBUTIONS OF ARKANSAS' ACRES FOR WILDLIFE PARTICIPANTS AND ENROLLED LANDS**

Stephanie Bunch and Philip A. Tappe, School of Forest Resources and Arkansas Forest Resources Center, University of Arkansas, Monticello, Arkansas 71656

Poster 4 **DEVELOPMENT OF AN IMPROVED SMALLPOX VACCINE**

Kara Davis and Wayne L. Gray. Dept. of Microbiology and Immunology, University of Arkansas for Medical Sciences, Little Rock, Arkansas 722205.

Poster 5 **WHO'S YOUR DADDY? USING MICROSATELLITES FOR PATERNITY DETERMINATION IN FRESHWATER MUSSELS.**

M. Jason Gambill, Jeannette Loutsch, Raven Lawson, A. Grace Miller, and Alan D. Christian. Department of Biological Sciences, Arkansas State University, P.O. Box 599, State University, Arkansas, 72467.

Poster 6 **SYNTHESIS AND EVALUATION OF NEW CATHEPSIN D INHIBITORS**

Adam Green, Carol Trana, Susan E. Hatfield, Matthew McConnell, Ashley Young, Lauren Young, Walter E. Godwin, and Rose McConnell. School of Mathematical & Natural Sciences, University of Arkansas at Monticello, Monticello, Arkansas 71657

Poster 7 **GENETIC AND BIOCHEMICAL STRATEGIES FOR STUDYING SPT16-HISTONE INTERACTIONS IN YEAST**

Jeffrey Hall[1], Fred Winston[2], and Andrea A. Duina[1,2]. [1]Hendrix College, Biology Department, Conway, Arkansas 72032. [2]Harvard Medical School, Department of Genetics, Boston, Massachusetts 02115

Poster 8 **STUDY OF BEAM VIBRATIONS USING EMBEDDED SENSORS AND ITS APPLICATION TO STRUCTURAL HEALTH MONITORING**

Justin Cole, Shakhrukh Ismonov, and Dr. Shivan Haran, Ph.D., Assistant Professor, College of Engineering, Arkansas State University, State University, Arkansas 72467

Poster 9 **PROTEIN-PROTEIN INTERACTIONS THROUGH SHEET 2-HELIX C LOOP OF P450 REDUCTASE**

Arvind P. Jamakhandi, Sharon A. Ellazar, and Grover P. Miller. University of Arkansas for Medical Sciences, Biochemistry Dept, Little Rock, Arkansas 72205

Poster 10 **COMPARISON OF HISTORICAL AND CURRENT NESTING HABITAT FOR INTERIOR LEAST TERNS (*STERNA ANTILLARUM ANTHALASSOS*) ON THE ARKANSAS RIVER, ARKANSAS**

Knoll, Erin L. and Thomas Nupp, Biology Department, Arkansas Tech University, Russellville, Arkansas 72801

Poster 11 **FIRST-YEAR RESULTS: REPRODUCTIVE SUCCESS, PHILOPATRY AND PREDATION OF PRAIRIE WARBLERS, BLUE-WINGED WARBLERS, INDIGO BUNTINGS, AND FIELD SPARROWS IN SCRUB HABITAT IN WESTERN CONNECTICUT**

Christy A. Melhart and Kimberly G. Smith. Department of Biological Sciences, 601 Science Engineering, University of Arkansas, Fayetteville, Arkansas 72701

Poster 12 **THE REPRODUCTIVE ECOLOGY OF THE WESTERN SLIMY SALAMANDER (*PLETHODON ALBAGULA*) FROM A MINE SHAFT IN THE OUACHITA NATIONAL FOREST, ARKANSAS**

Joseph R. Milanovich, Stanley E. Trauth, and [1]David A. Saugey; Department of Biological Sciences, Arkansas State University, P.O. Box 599, State University, Arkansas 72467; [1]United States Forest Service, 8607 Highway 7 North, Jessieville, Arkansas, 71949

Poster 13 **WATER QUALITY AND ECOSYSTEM FUNCTION ON UPPER AND LOWER REACHES OF TRIBUTARIES OF THE L'ANGUILLE RIVER, ARKANSAS**

Mellissa Milligan and Richard S. Grippo. Department of Biological Sciences, Arkansas State University, State University, Arkansas 72467

Poster 14 **AN INVESTIGATION OF CARBOHYDRATE MIMIKING PEPTIDES' CLUSTERING AND SECONDARY STRUCTURE PROPERTIES**

Jennifer Roller[1], Anastas Pashov[2], Jason Plaxco[2], Rinku Saha[2], Stewart Macleod[2], Thomas Kieber-Emmons[2]. [1]Hendrix College, 1600 Washington Avenue, Conway, Arkansas 72032 and [2]Arkansas Cancer Research Center and Department of Pathology, University of Arkansas for Medical Sciences, 4301 West Markham, Little Rock, Arkansas 72205

Poster 15 **TESTING THE OXYGEN PARADOX WITH ANTIOXIDANT-DEFICIENT CYANOBACTERIA**

Christy L. Schuchardt, C. J. Spurlock, and David J. Thomas. Science Division, Lyon College, Batesville, Arkansas 72501.

Poster 16 **CELLULAR SLIME MOLDS IN OZARK CAVES**

John C. Landolt, Michael E. Slay, and Steven L. Stephenson. Department of Biology, Shepherd University, P.O. Box 3210, Shepherdstown, West Virginia 25443. Arkansas Field Office, The Nature Conservancy, 601 North University Avenue, Little Rock, Arkansas 72205. Department of Biological Sciences, 601 Science Engineering, University of Arkansas, Fayetteville, Arkansas 72701.

Poster 17 **NITRATE-REDUCING BACTERIA FROM CHILE'S ATACAMA DESERT (A POTENTIAL MARTIAN ANALOG).**

CaSandra J. Spurlock[1], Christy L. Schuchardt[1], Shawn M. Zimmerman[1], Christopher P. McKay[2], and David J. Thomas[1]. 1Science Division, Lyon College, Batesville, Arkansas 72501; [2]Space Science Division, NASA Ames Research Center, Moffett Field, California 94035.

Poster 18 **SODA BOTTLE WINOGRADSKY COLUMNS AS ASTROBIOLOGY EXPERIMENTS IN ELEMENTARY SCHOOL THROUGH COLLEGE.**

David J. Thomas[1] and Gayle Ross[2]. [1]Science Division, Lyon College, Batesville, Arkansas 72501; [2]Sulphur Rock Junior/Senior High School, Sulphur Rock, Arkansas 72579.

Poster 19 **CROSS-SECTIONAL VARIATION IN THE FIBER-TYPE PROFILE OF THE BOTTLENOSE DOLPHIN (*TURSIOPS TRUNCATUS*) SCALENUS MUSCLE**

Brandon Thurow and Jennifer L. Dearolf, Biology Department, Hendrix College, Conway, Arkansas 72032

Poster 20 **EVIDENCE FOR AN ALTERNATIVE SERINE RACEMASE TRANSCRIPT**

Deborly Wade, Central Baptist College, Conway, Arkansas 72034 and Steven W. Barger, Ph.D., University of Arkansas for Medical Sciences, 1501 W. Markham, Little Rock, Arkansas 72205

Poster 21 **SYNTHESIS AND ANT-APOPTOTIC EFFECT OF CU-DISMUTASE MIMETIC IN CULTURED KIDNEY EPITHELIAL CELLS**

Grant W. Wangila[1]; Kiran K. Nagothu[2]; Richard Steward III[1]; Renu Bhatt[2]; John R. J. Sorenson[2]; Sudhir V. Shah[2] and Didier Portilla[1]. [1]University of Arkansas at Pine Bluff, 1200 University Drive, Pine Bluff, 71601. [2]University of Arkansas for Medical Science Campus, 4301 West, Markham Street, Little Rock, Arkansas 72205.

Poster 22 **A FORGOTTEN CORPS OF ECOLOGICAL DISCOVERY: THE DUNBAR AND HUNTER EXPEDITION**

Robert C. Weih, Jr.[1], Don C. Bragg[2], James T. Hartshorn[1], David W. Rowton[1]. [1]Spatial Analysis Laboratory (SAL), University of Arkansas at Monticello, Arkansas Forest Resources Center, School of Forest Resources, 110 University Court, Monticello, Arkansas 71656, Email: weih@uamont.edu, Phone: 870-460-1248, Fax: 870-460-1092. [2]USDA Forest Service, Southern Research Station, P.O. Box 3516 UAM, Monticello, Arkansas 71656

Poster 23 **SPINAL CORD INJURIES STUDY–DESIGN OF A SMART IMPACTOR**

Ronald Kelly, Antonius Hasan, Logan Hardin, Jim Yancey, and Shivan Haran, Assistant Professor, College of Engineering, Malathi Srivatsan, Assistant Professor, Biological Sciences, Arkansas State University, State University, Arkansas 72467

Studies of Zeolite Entrapped Ruthenium Polypyridine Complexes

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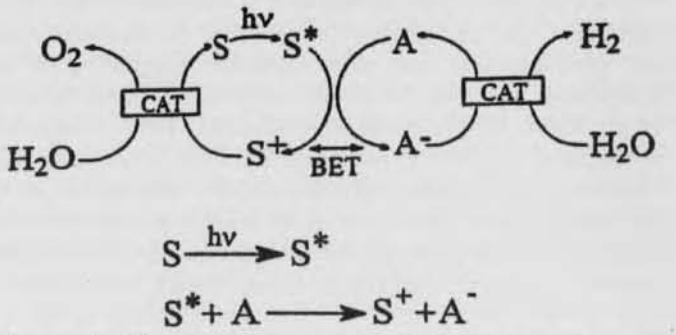
Abstract

There is an intense interest in designing molecular systems which will absorb visible sunlight, initiate an electron transfer process, and ultimately convert the solar energy to useful chemical energy of fuels such as hydrogen produced from water. The zeolite-entrapped polypyridine complexes of divalent ruthenium hold promise as efficient photocatalysts for net charge separation and such efficiencies are further enhanced by organized incorporation of donor and acceptor components. This paper deals with the synthesis and spectroscopic investigation of zeolite-entrapped ruthenium polypyridine complexes which may be useful in the development of solar energy conversion schemes. The sensitizer molecules, such as $Ru(bpy)_3^{2+}$ (bpy = 2,2'-bipyridine), are entrapped within the supercages of structurally well-defined zeolite Y by the so called "ship in a bottle" synthesis, which eliminates the undesirable diffusion of the complex and inhibits the wasteful back-electron transfer reaction. This complex has a dimension of ~12Å, which is too large to introduce through a 7.4 Å window opening. Once the complex is formed in the supercage, it cannot escape through the windows and is effectively entrapped within the supercage. The zeolite-entrapped ruthenium complexes are characterized by diffuse reflectance, electronic absorption, electronic emission, and resonance Raman (RR) spectroscopy, as well as excited state lifetime measurements. A brief summary of the synthetic and characterization procedure of the zeolite-entrapped ruthenium polypyridine complexes is presented here. Emphasis is given on the author's work, although a discussion of some of the important contributions made by other workers is also included. This study clearly demonstrates that entrapment of ruthenium complex within the supercage of Y-zeolite can alter inherent photophysical properties of the complex in an advantageous manner.

Introduction

Photosynthesis is the process that converts solar energy into chemical energy and maintains life on earth (Lawlor, 1993). Only a very small fraction (~0.05%) of the huge amount of solar energy available is converted by green plants in photosynthesis, and the rest of the energy is wasted. For the last two decades there has been an intense interest in designing molecular systems that mimic photosynthesis. The strategy has been to design a molecular assembly that will absorb visible light, that will initiate an electron transfer process, ultimately the solar energy is used to cleave water to produce hydrogen fuel (Kalyanasundaram, 1987; Parmon and Zamarev, 1989). The inexhaustible solar energy can be converted to environmentally clean fuels by assembling a molecular suprastructure.

There are three basic phenomena that control the use of light energy: energy capture, energy transfer, and photoinitiated electron transfer. A synthetic photocatalytic system that can produce hydrogen by the reduction of water is shown in Scheme 1. In this scheme the excited state molecule S* (S is the sensitizer molecule which absorbs the visible light) reacts with the acceptor molecule A forming S^+ and A^-. The reduced A^- intermediate can transfer the electron to water via an appropriate catalyst, leading to the production of hydrogen. The sensitizer, S, is then regenerated by the electrons provided by a donor (another ruthenium complex or a sacrificial electron donor such as EDTA), D, to form D^+, which is then available to convert H_2O to O_2.



Scheme 1. Photocatalytic system for splitting of water (S = sensitizer, A = acceptor, BET = back electron transfer, CAT = catalyst).

In order to make Scheme 1 practical, several issues need to be resolved. The issues of concern are: (1) proper choice of sensitizer (S) from the view point of absorption of sunlight; (2) production of a reasonably long excited state lifetime of S* so that it can react with A; (3) separation of S^+ and A^- from each other to minimize back electron transfer; (4) suitable ground and excited state potential of the redox species so that some useful chemistry can be carried out; and (5) regeneration of the photochemical cycle.

Many attempts have been made (Ramamurthy, 1991) to overcome these problems and to make Scheme 1 practical. Several classes of molecules have been shown to possess the necessary properties to serve as effective photosensitizers for such schemes. The most promising are

those based on polypyridine complexes of divalent ruthenium (i.e, $Ru(bpy)_3^{2+}$ and related complexes; Kalyanasundaram, 1982), various metal complexes of porphyrins (Persaud et al., 1987), and phthalocyanines (Darwent et al., 1982). Even though suitable photosensitizers are available in terms of excited state lifetime, other fundamental problems need to be solved. Simple homogeneous systems such as liquid solutions are not suitable for solar devices because of the random character of molecular thermal motion, uncontrolled diffusion, and the lack of a barrier to prevent wasteful back reaction.

There is an intense interest in designing effective organizational strategies to overcome the above-mentioned problems. These strategies include synthesis of covalently linked redox assemblies as well as the incorporation of the photoactive species into larger spatially well-organized media such as modified glass surfaces or microheterogeneous systems (Gafney, 1990) such as micelles, colloids, lipids, or polymers. One of the most interesting attempts reported so far is the use of highly ordered host materials such as zeolites (Turbeville et al., 1992; Dutta and Turbeville, 1992). The sensitizer molecules such as $Ru(bpy)_3^{2+}$ (bpy = 2,2'-bipyridine) are entrapped within the supercages of structurally well defined zeolite Y (Turbeville et al., 1992; Dutta and Turbeville, 1992), which eliminates the undesirable diffusion of the complex, and the photophysical and photochemical properties of the complexes can be favorably influenced (Maruszewski et al., 1993; Maruszewski and Kincaid, 1995). Electronic absorption, electronic emission, as well as excited state lifetime measurements provide valuable information about the photophysical properties of the complexes in solution as well as for zeolite entrapped complexes, and such studies clearly document modifications upon zeolite entrapment.

Materials and Methods

Structure of Zeolites.—Zeolites are crystalline aluminosilicates of sodium, potassium, or calcium. The general chemical composition can be expressed by the formula $M_{2/n} \cdot Al_2O_3 \cdot xSiO_2 \cdot yH_2O$ (Breck, 1974), where n is the charge of the cation and x is usually ≥2. The structural framework is assembled by sharing the corner of the SiO_4 and AlO_4 tetrahedra in a three dimensional network. The cations M^{n+} occupy extra framework positions and balance the negative charge of the AlO_4 unit of the framework. The internal structure of the zeolite is composed of interconnecting cages and channels. The channels and void spaces of the three dimensional zeolites are usually occupied by cations and water molecules.

Under typical conditions, water molecules fill the interior volume of zeolites. The water molecules can be removed by heating without any structural change, which is in contrast to other hydrated compounds. In the dehydrated zeolites, the empty channels can be filled with other molecules. Another useful feature of the zeolites is that the M^{n+} cations can be readily ion exchanged with other cations in aqueous media. The size of the supercages and size of the channels in zeolites vary from 2 to 13 Å and extend in a regular fashion throughout the structure. The internal architecture of zeolite makes it suitable for spatial arrangement of molecules. The steric and electrostatic constraints imposed by the supercages and channels may change the photophysical and photochemical properties of entrapped molecules, sometimes in an advantageous manner.

There are 34 known naturally occurring zeolite minerals and about 100 types of synthetic zeolites. Only a few of them are of practical use because the others are structurally unstable upon dehydration. The 6 most commonly examined zeolites as hosts for photocatalytic systems are zeolites X, Y, A, and L, mordenite, and ZSM-5. The synthetic analogue of the mineral faujasite is known as zeolite X or Y depending on the ratio of Si/Al in the framework. The unit cell of this zeolite is cubic and contains 192 $(Si, Al)O_4$ tetrahedra as shown in Fig. 1. The basic unit is formed by sodalite cages connected in a tetrahedral arrangement by double-6-ring (D6R) units, which produce the largest supercages of internal diameter ~ 13 Å and a window opening of ~ 7.4 Å. This large supercage makes it a very attractive host for immobilized photochemical reactants. The Na^+ cation present in this zeolite can easily be ion-exchanged with a wide variety of ions having dimensions less than the 7.4 Å window opening of the supercages.

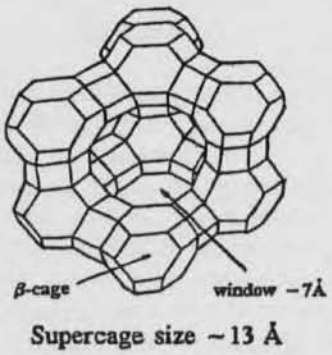


Fig. 1. The structure of a zeolite-Y supercage.

Among all the zeolites, zeolite Y has the largest void space, which is 50% of the dehydrated crystal. It is thermally stable up to 700°C. Zeolite X has a Si/Al ratio of 1:1.4 and zeolite Y has a ratio of 1.4:3. Zeolite A has no natural counterpart and can be synthesized in the laboratory. In the framework Si and Al atoms are repeating alternately with

a Si/Al ratio typically of 1. The framework is made up of sodalite cages which are connected via double four-membered rings to form a supercage of diameter 11.4 Å and window size of 4.1 Å. Zeolite L is a one-dimensional tunnel-like framework, and Mordenite is a one-dimensional 12-membered ring system. Among all the stable zeolites, zeolite Y is an attractive one for the entrapment of a sensitizer molecule for carrying out photochemical reactions. The one-dimensional zeolites are not suitable for the entrapment. The internal diameter and window sizes are not suitable for the entrapment of the sensitizer for some of the zeolites. The popularity of zeolite Y arises from its ready availability, rigid framework structure, the largest void space of any known zeolite, and it is used in this research work as a host.

Assembly within Zeolite.–There are several strategies used to introduce the molecules of interest into the zeolites, depending on the nature and size of the molecular species. Positively charged species can be ion exchanged into the zeolite if they are small enough to penetrate through the window openings. Neutral molecules can be transported in vapor phase or via a solvent into the dehydrated zeolites. Using both of these strategies, it is possible to assemble a molecule inside the zeolite supercages that is then too large to escape via the 7.4 Å window opening. This so-called "ship in a bottle" synthesis is a 2-step process. The first step is the ion exchange with the desired cation, and the second step is the addition of the ligand followed by a thermal treatment to form the complex. One of the most impressive examples is the synthesis of zeolite (Z)-entrapped $Ru(bpy)_3^{2+}$ complex reported in the pioneering work of DeWilde and coworkers (1980). This molecule has a dimension of ~12 Å, which is too large to introduce through the window opening. Once it is formed, it cannot escape through the 7.4 Å windows and is effectively entrapped within the supercage. The reaction scheme is shown below:

$$Z\,(aq) + Ru(NH_3)_6^{3+} \rightarrow Z\text{-}Ru(NH_3)_6^{3+}\,(aq) \quad \text{(Ion exchange)}$$

$$Z\text{-}Ru(NH_3)_6^{3+}\,(aq) \rightarrow Z\text{-}Ru(NH_3)_6^{3+}\,(dry) \quad \text{(Filtration \& drying)}$$

$$Z\text{-}Ru(NH_3)_6^{3+}\,(dry) + x\ bpy \rightarrow \text{Crude } Z\text{-}Ru(bpy)_3^{2+} + \text{bpy degradation products} \quad \text{(Synthesis)}$$

$$\text{Crude } Z\text{-}Ru(bpy)_3^{2+} \rightarrow \text{Pure } Z\ Ru(bpy)_3^{2+} \quad \text{(Purification)}$$

The calcinated (pre-cleaned from organic impurities by oxidation under flow of oxygen at 500°C for 5 hours) zeolite sample (Z) is loaded with $Ru(NH_3)_6^{3+}$ by ion exchange from aqueous solution and then filtered and dried under vacuum. The color of the solid changes from white to yellow when heated at high temperature (~200°C) with excess bipyridine (bpy) in a sealed tube. Surface adsorbed $Ru(bpy)_3^{2+}$ is removed by washing extensively with 10% NaCl solution, and excess ligand can be removed by extensive soxhlet extraction with ethanol. The zeolite entrapped complex is characterized by diffuse reflectance, electronic absorption, electronic emission, and resonance Raman (RR) spectroscopy.

Maruszewski and coworkers (1991, 1993, and 1995) and Bhuiyan and Kincaid (1999, and 2001) developed an extended approach to synthesize mixed-ligand complexes of ruthenium(II) within zeolite Y. In this procedure the bis complex, $Z\text{-}RuL_2^{2+}$ (L = polypyridine ligand), is first synthesized within zeolite by heating at a relatively low temperature (~90°C). Then the third ligand is inserted at higher temperature (~200°C). The integrity of the material is confirmed by spectroscopic methods. The ability to generate zeolite-entrapped, tris-ligated, heteroleptic complexes opens up many more possibilities for synthesis of a wide range of complexes. More importantly, this type of heteroleptic complex is very useful for the construction of zeolite-based organized molecular assemblies, which can effectively reduce the rate of wasteful back-electron transfer and increase the net charge separation efficiency.

Maruszewski and Kincaid (1995) demonstrated that upon the entrapment of $Ru(bpy)_2(daf)^{2+}$ (daf = diazafluorene) within the Y-zeolite supercages, an increase in the energy of the ^{3}dd state is observed. In solution this complex is essentially non-emitting because of its very low-lying ^{3}dd state (E_{dd}=2271 cm^{-1}). Upon entrapment within the zeolite supercage, the complex exhibits easily detectable emissions and a dramatically increased ^{3}MLCT(metal-to-ligand charge transfer-state) lifetime at room temperature. By conducting lifetime measurements at many temperatures, it was shown that the increased lifetime of the entrapped complex results from zeolite-induced destabilization (by ~1700 cm^{-1}) of the ^{3}dd state. It was concluded that both steric and electrostatic interactions of the entrapped species change the photophysical properties of the complexes.

One fundamental question which arises concerns the size limitation imposed by the 7Å window opening on the polypyridine ligands, which can be efficiently delivered to the intra-zeolitic ruthenium ions. In order to address this issue, efforts have been made to prepare the zeolite-entrapped ruthenium complex of terpyridine: i.e., $Z\text{-}Ru(tpy)_2^{2+}$ (Bhuiyan and Kincaid, 1998). Having established the feasibility of utilizing ligands the size of terpyridine, we (Bhuiyan and Kincaid, 1999) took another project to employ the commonly used bridging ligand 2,3-bis(2-pyridyl) pyrazine (dpp) to demonstrate the possibility that covalently-linked binuclear complexes may be formed within the three-dimensional intrazeolitic framework. The synthesis of $Z\text{-}Ru(bpy)_2dpp^{2+}$ was successfully accomplished, and the spectroscopic and photophysical properties of this material were throughly documented (Bhuiyan and Kincaid, 1999). Structures of some of the complexes synthesized inside zeolite Y are shown in Fig. 2.

There are examples of large, transition-metal

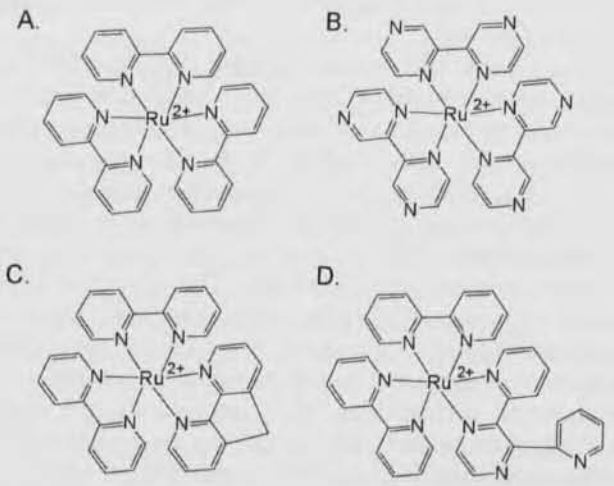


Fig. 2. Structure of metal complexes synthesized in zeolite Y supercages (A) $Ru(bpy)_3 2+$ (B) $Ru(bpz)_3 2+$ (C) $Ru(bpy)_2(daf) 2+$ (D) $Ru(bpy)_2(dpp) 2+$ where bpy = 2,2'-bipyridine, bpz = 2,2'-bipyrazine, daf = 4,5-diazafluorene, dpp = 2,3-bis(2-pyridyl)pyrazine

complexes entrapped in the supercages of zeolite Y, such as iron (II) phthalocyanine (Herron, 1988) and zinc porphyrazine (Szulbinski and Kincaid, 1998). In these substances, the large guest molecules are presumably distorted from their planar configuration since the dimensions of the ligands (i.d. 14-15 Å) exceed the diameter of the zeolite supercages. The selectivity and activity of a metal phthalocyanine catalyst is enhanced with its inclusion in the zeolite cavity.

Persaud and coworkers (1987) reported the construction of a zeolite-based, multicomponent, photocatalytic assembly, which provides spatial organization of the electron donor and acceptor. A one-dimensional tunnel-like zeolite L was used as a host in that work. Small platinum clusters were formed inside the zeolite channel, and then the channel was loaded with a large amount of methyl viologen acceptor (MV^{2+}). The zinc porphyrin photosensitizer, $(ZnTMPy)^{4+}$, was too large to enter the 7Å zeolite channels, but it was strongly adsorbed on the outer surface. In the presence of a sacrificial electron donor (EDTA), this system was capable of photocatalytic generation of hydrogen from water. Upon photolysis, the electron transfer from zinc porphyrin to the acceptor methyl viologen (MV^{2+}) forming the $MV^{+\cdot}$ radical and the photosensitizer was regenerated by the sacrificial electron donor (EDTA), which prevents back electron transfer. Electron transfer along the chain of included MV^{2+} cations ultimately leads to the reduction of water to hydrogen in the presence of the included platinum catalyst.

Dutta and Turbeville (1992) reported the photoinduced electron transfer between the zeolite-entrapped $Ru(bpy)_3^{2+}$ and an acceptor, methyl viologen (MV^{2+}), located in neighboring cages. In this photoredox study, $Z\text{-}Ru(bpy)_3^{2+}$ was synthesized, and then MV^{2+} was ion exchanged at high loadings such that each supercage contained two MV^{2+} ions. Upon photolysis under anaerobic conditions, the orange pellet turned blue in color, indicating the formation of the $MV^{+\cdot}$ radical. The presence of the methyl viologen radical was confirmed by diffuse reflectance and time resolved resonance Raman (TR^3) spectroscopy. This blue species was stable for several hours under anaerobic conditions. The results were interpreted to indicate that the back electron transfer was retarded, and the photogenerated $MV^{+\cdot}$ transfers its electron to more remote MV^{2+} acceptors.

Results and Discussion

Synthesis and Characterization of Zeolite-Entrapped Bis-Terpyridine Ruthenium(II).–The results of detailed studies of the photophysical properties of a range of zeolite entrapped complexes demonstrates that the most important effect of entrapment of such complexes within the Y-zeolite supercages is an increase in the energy of the so-called "ligand-field" (^{3}dd) state (Maruszewski and Kincaid, 1995). The referenced study, demonstrated the effectiveness of zeolite entrapment in eliminating the LF (ligand-field)-state destabilization pathway, and prompted us (Bhuiyan and Kincaid, 1998) to undertake the study of zeolite-entrapped complexes of terpyridine (tpy). In free solution the bis-tpy complex, $Ru(tpy)_2^{2+}$, is essentially non-luminescent with a very short lifetime (~250 ps) at room temperature (Winkler et al., 1987). The origin of this short lifetime has been debated. The results of our previous study (Bhuiyan and Kincaid, 1998) revealed that the increased LF-state destabilization in this complex is comparable to that observed upon zeolite-Y-entrapped $Ru(bpy)_2daf^{2+}$ and nicely demonstrated the concept that zeolite entrapment provides a useful strategy for advantageous manipulation of the photophysical properties of such systems.

The zeolite sample was purified by calcinations (Incavo and Dutta, 1990). The zeolite-entrapped complex, $Z\text{-}Ru(tpy)_2^{2+}$, was prepared by a modification of a method previously developed in Kincaid's laboratory (Bhuiyan and Kincaid, 1998; Maruszewski et al., 1991), which is based on the pioneering work of DeWilde and coworkers (1980) and Quayle and Lunsford (1982). Spectroscopic measurements indicated that $Z\text{-}Ru(tpy)_2^{2+}$ was formed. The integrity of the zeolite-entrapped sample was confirmed by RR, electronic absorption, and emission spectra. The zeolite-entrapped complex was extracted from the zeolite matrix by the hydrofluoric acid method (Maruszewski et al., 1991). The reference complex, $[Ru(tpy)_2](PF_6)_2$, was prepared following the procedure of Maestri and coworkers (1995).

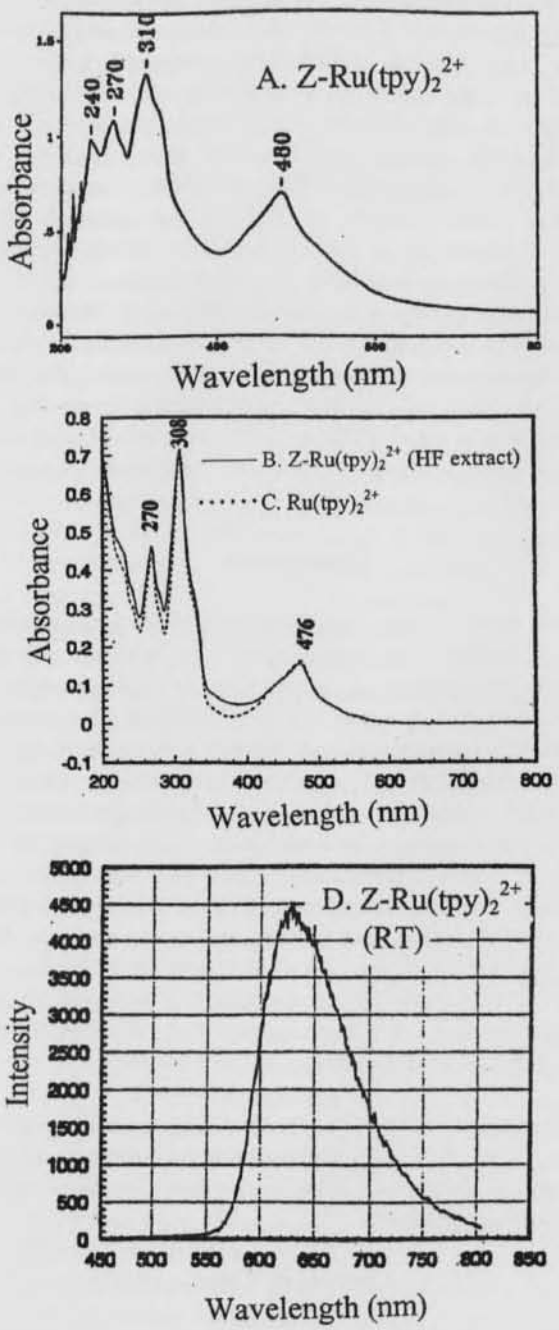


Fig. 3. Absorption and emission spectra of Ru(tpy)$_2$2+ complex.

The electronic absorption and emission spectra of the bis-terpyridine complex in various forms are given in Fig. 3. The absorption spectrum of the independently synthesized complex in acetonitrile solution (trace C) matches that reported in the literature (Maestri et al., 1995). The absorption spectrum of the zeolite-entrapped complex (trace A) as well as the liberated complex that is obtained following dissolution of the zeolite matrix (trace B) shows no significant differences in the positions of the absorption maxima compared to the spectrum of the independently prepared solution-phase complex (trace C). The very intense bands in the UV region can be assigned to ligand-centered $\pi \rightarrow \pi^*$ transitions. The relatively intense and broad absorption band in the visible region, which is responsible for the deep red color, is due to spin allowed $d \rightarrow \pi^*$ metal-to-ligand charge-transfer (MLCT) transitions (Stone and Crosby, 1981). At room temperature, Ru(tpy)$_2^{2+}$ is practically non-luminescent, but upon entrapment in zeolite, there is a dramatic increase in luminescence (trace d) at room temperature. The RR spectra of the complex in solution and that of the zeolite-entrapped species are in good agreement (Bhuiyan and Kincaid, 1998). The spectral pattern does not significantly change upon entrapment of the complex into the zeolite matrix. There are only slight shifts observed for the zeolite entrapped complex relative to the solution-phase complex.

Effects on Photophysical Properties.–The ^{3}MLCT-state lifetime of Ru(tpy)$_2^{2+}$ in solution is too short (250 ps at room temperature) in comparison with the other ruthenium-polypyridine complexes (Winkler et al., 1987). However, the luminescence of the zeolite-entrapped sample at room temperature has an associated lifetime of 140 ns in aqueous suspension (Bhuiyan and Kincaid, 1998). As expected, excited-state lifetimes obtained at low temperatures increase with decreasing temperature, reaching 844 ns at -50°C. As is

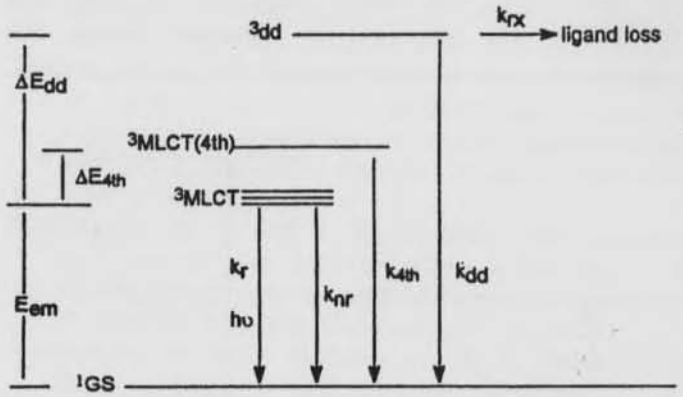


Fig. 4. Schematic representation of the excited-state deactivation pathways in ruthenium polypyridine complexes.

summarized in Fig. 4, the lowest energy ^{3}MLCT states of ruthenium(II)-polypyridine complexes may relax to the ground state via a number of pathways, including

population of two thermally accessible upper states whose participation can be documented by analysis of lifetime data acquired over a range of temperatures (Allen et al., 1984; Sykora and Kincaid, 1995). For most cases, a single thermal term (eq 2) is adequate to fit the experimental temperature-dependent lifetime data, but in some cases it is necessary to use two thermal terms (eq 1) in order to fit the experimental data (Maruszewski et al., 1993; Maruszewski and Kincaid, 1995; Sykora and Kincaid, 1995; Bhuiyan and Kincaid, 1998; Bhuiyan and Kincaid, 1999). The excited state lifetimes (T) are given by

$$1/T = k_{total} = k_r + k_{nr} + k_{dd} \exp(-\Delta E_{dd}/kT) + k_{dd} \exp(-\Delta E_{4th}/kT) \quad (1)$$

$$1/T = k_{total} = k_r + k_{nr} + k_{dd} \exp(-\Delta E_{dd}/kT) \quad (2)$$

In equations 1 and 2, k_r and k_{nr} are the rate constants for direct radiative and nonradiative decays. The deactivation rate constant of the thermally populated (^{3}dd) states is designated k_{dd} .ΔE_{dd} is the energy gap between the ^{3}dd states and the ^{3}MLCT-emitting states. The deactivation-rate constant, k_{4th}, is associated with an additional low lying ^{3}MLCT state (the so-called fourth MLCT state), which may be thermally populated as a consequence of the small magnitude of .ΔE_{4th} (typically 600-900 cm^{-1}; Maruszewski et al., 1993; Maruszewski and Kincaid, 1995; Sykora and Kincaid, 1995; Bhuiyan and Kincaid, 1998; Bhuiyan and Kincaid, 1999).

Both equations (equations 1 and 2) were tested (Bhuiyan and Kincaid, 1998) in an attempt to reproduce the observed data. Analysis of the curves reveals that the monoexponential model (single thermal term) does not satisfactorily reproduce the observed lifetime data. However, introduction of the second thermal term yields excellent agreement between the calculated and observed curves. The kinetic parameters obtained from both models are quite similar to those obtained for the majority of complexes (Maruszewski et al., 1993; Maruszewski and Kincaid, 1995; Sykora and Kincaid, 1995; Bhuiyan and Kincaid, 1998; Bhuiyan and Kincaid, 1999). Comparison of the ΔE_{dd} values (shown in Fig. 5) for solution phase $Ru(tpy)_2^{2+}$ (Clark et al., 1991) and the zeolite-entrapped complex (Bhuiyan and Kincaid, 1998) shows a substantial increase upon zeolite entrapment (1181 cm^{-1}). This increase in E_{dd} values accounts for the dramatic increase in lifetime and emission intensity upon zeolite entrapment. The steric constraint induced by the rigid zeolite cage on the electronically excited Z-$Ru(tpy)_2^{2+}$ results in destabilization of the LF state, leading to a decrease in thermal population of this state.

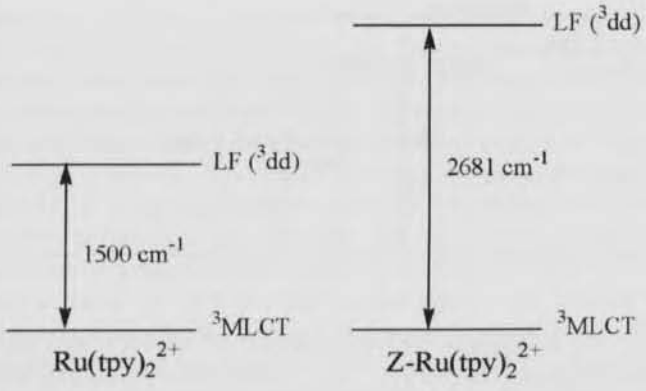


Fig. 5. Energy gap (E_{dd}) diagram for solution-phase $Ru(tpy)_2^{2+}$ complex and zeolite-entrapped Z-$Ru(tpy)_2^{2+}$ complex.

Conclusions

This study clearly demonstrates that entrapment of $Ru(tpy)_2^{2+}$ within the supercage of Y zeolite can alter inherent photophysical properties of the complex in an advantageous manner. In free solution this complex is practically nonluminescent, having a very short excited-state lifetime (250 ps) at room temperature. However, entrapment within the zeolite supercage results in dramatic increases in emission intensity and excited-state lifetime (140 ns) at room temperature. The observed temperature dependence of the excited-state lifetime has been modeled by a kinetic equation with two thermal terms corresponding to the population of the so-called fourth ^{3}MLCT state and the ligand field state (LF), respectively. It is shown that the increased lifetime of the entrapped complex results from zeolite-induced destabilization of the LF state.

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Small Fish Species of Arkansas Reservoirs

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Abstract

Sixty-six Arkansas reservoirs were sampled with rotenone from 1997 through 2004 to determine the distribution and species richness of small, nongame fish species in manmade lentic environments. Eighty-five small fish species distributed among 13 families were collected. Fish distribution and species richness varied by reservoir size, ecoregion, and reservoir type. Species richness was significantly correlated with reservoir size and the number of small species occurring in reservoir drainages. Some small species apparently maintained breeding populations in reservoirs, other species occurred in low numbers and may not have maintained breeding populations, and some species occurred sporadically, probably as stragglers from nearby tributary streams. This study should provide information for comparing and interpreting future successional changes in reservoir fish communities as the reservoirs age.

Introduction

The native Arkansas fish fauna is dominated by stream-adapted species because there are few natural lakes in the state. During the 20th century, man-made impoundments of various types and sizes were constructed throughout Arkansas creating an abundance of artificial lentic habitats. At least 56 reservoirs exceeding 200 ha in surface area were built, mainly during the last 60 years. Impoundments serve various purposes including flood control, irrigation, power production, water supply, navigation, and recreation.

When a natural stream is impounded, the resulting reservoir drastically alters the aquatic environment of the area. Some stream-adapted fishes are unable to survive under reservoir conditions and are extirpated from the affected area; other stream species are able to survive in the reservoir at low population levels or occur there occasionally as waifs from tributary drainages, and some stream species are able to thrive under reservoir conditions. Prediction of the reaction of small fish species that evolved in rivers (or even natural lakes) to new reservoir ecosystems has been based largely on experience or judgment rather than scientific evidence (Benson, 1973), although there have been many studies on the population dynamics and fisheries resources of gamefish and commercial species in reservoirs. Most studies of the effects of reservoirs on stream fish populations focused on the benefits derived from the creation and management of new sport fisheries opportunities or on negative aspects, such as the loss of populations of rare or endangered species and the overall drastic reduction in biodiversity resulting from a simplified ecosystem (reservoir). Information on the adverse impacts of reservoir construction on natural stream fish assemblages can be found in Bain et al. (1988), Baxter (1977), Crisp et al. (1984), Cross and Moss (1987), Etnier and Starnes (1993), Hubbs and Pigg (1976), Jenkins and Burkhead (1993), Luttrell et al. (1999), Mahon and Ferguson (1981), Martinez et al. (1994), and Neves and Angermeier (1990).

The presence of small, stream-adapted fish species in reservoirs has received little attention. Hall (1949, 1950) first noted the occurrence of stream-adapted fishes in new Oklahoma reservoirs and subsequently discussed the need for long-term study of post-impoundment stream fish succession (Hall, 1953). Although mostly anecdotal comments exist in the scientific literature about small, nongame stream fishes occurring in reservoirs, there is little published information documenting the distribution and abundance of those species in the reservoirs of a specific geographic region as large as Arkansas. A number of studies documented the pre- and post-impoundment fish populations of individual North American reservoirs, but most of the post-impoundment surveys were conducted within a year or two after the new reservoir filled, and no attempt was made to determine possible successional changes in small species composition as the reservoirs aged. There is some information comparing the fish species composition over time for a few reservoirs, such as Lake Texoma in Oklahoma (Riggs and Bonn, 1959; Echelle et al., 1971; Matthews, 1998), but there are few, if any, long-term studies of the small, stream-adapted species occurring in impoundments of different sizes and ages of an entire state. Fish sampling in 13 Arkansas reservoirs between 1970 and 1996 produced some surprising species records and numbers for small fishes generally associated with lotic environments (TMB, unpublished data). In that sampling, several nongame species in each of the 3 largest Arkansas fish families (Cyprinidae, Ictaluridae, and Percidae) were abundant in a few of the reservoirs sampled, including some species which are commonly considered most likely to be adversely affected by reservoir construction.

The objectives of this study were as follows:

(1) To determine which small, nongame fish species occur in Arkansas reservoirs;

(2) To determine the small species distributional patterns by ecoregion among the reservoirs sampled statewide; and

(3) To compare the small species richness of the reservoirs sampled.

Materials and Methods

Fishes were collected from 66 Arkansas reservoirs (Table 1) from 1997 through 2004 with the ichthyocide rotenone during Arkansas Game & Fish Commission fish population sampling conducted each year from June through September. One hundred ninety-two rotenone samples were taken in the 66 reservoirs, which ranged in age from 1 (Isabella) to 74 (Catherine) years and in surface area from 25 (Pineda) to 16,228 (Ouachita) ha. Each cove rotenone population sample required 2 days for a complete pickup of fishes in the sample area. The sample area of known surface area and depth was blocked off with a net to prevent fishes from entering or leaving the area sampled. All specimens of the small fish species collected in the sample area, with the exception of *Dorosoma petenense*, which was usually processed in the field due to the large numbers collected, were preserved in 10% formalin. The small species from the 1st day of the sample were preserved separately from those collected the 2nd day. Preserved specimens were later identified, enumerated by length category, and massed in grams (after the specimens were blotted with paper towels). Representative specimens were deposited in the University of Arkansas–Fort Smith Zoology Collection and in the collections of Arkansas Tech University, Southern Arkansas University, and the University of Louisiana Monroe. Excess specimens, particularly those collected on the 2nd day of sampling, were discarded.

Six reservoirs, Bois D'Arc, Coronado, DeSoto, Erling, Isabella, and Merrisach, were sampled by a nontraditional method. Three to 12 small rotenone samples were conducted in a variety of habitats in the upper, middle, and lower portions of those reservoirs. Each sample covered a small area (generally less than 0.1 ha) and required 1 to 2 hr.

Arkansas fishes considered to be small, nongame species in this study were species in which the adults do not normally exceed 26 cm total length (TL). All species of native minnows, madtom catfishes, and darters were considered to be small species. Arbitrary decisions were made about which species in the herring (Clupeidae), pike (*Esocidae*), and sunfish (*Centrarchidae*) families to include. The threadfin shad (*Dorosoma petenense*) was included, but its large, nongame relative, the gizzard shad (*D. cepedianum*), was not. The grass pickerel (*Esox americanus*) was included even though adults occasionally exceed 26 cm in length. The sunfish family was the most difficult to categorize, because individuals of most of its species rarely exceed 26 cm TL. The sunfishes that are generally considered gamefish species in Arkansas were not included as small species in this study. Only the 3 smallest Arkansas sunfish species, orangespotted sunfish (*Lepomis humilis*), dollar sunfish (*L. marginatus*), and bantam sunfish (*L. symmetricus*), were designated as small, nongame species. One hundred forty-five currently described native Arkansas fishes were designated as small, nongame species. Complete species lists and current systematic nomenclature for Arkansas fishes are presented in Robison and Buchanan (1988, 1993) and Nelson et al. (2004).

The term "reservoir" is used herein to include a variety of manmade impoundments. Four main types of impoundments were sampled during this study. They were categorized as follows:

(1) Flow-through impoundment on a large, navigable river (Type F).

These reservoirs were formed by constructing locks and dams on the Arkansas and Ouachita rivers to provide suitable pools for navigation and were designed to maintain downstream flows unlike the storage impoundments. These impoundments have both reservoir-like and river-like qualities. Eight Type F reservoirs were sampled.

(2) Leveed, pump-in impoundment (Type P).

These small reservoirs were created by building levees on 3 or 4 sides and pumping water into them, usually from a nearby river. The river water may or may not be filtered. Five Type P impoundments were sampled.

(3) Impoundment built by damming a flowing stream (Type S).

This type of impoundment creates a lentic environment and is the most common type of impoundment in Arkansas. Fifty-two Type S impoundments were sampled. These reservoirs can vary from small to large in size, but all of the largest reservoirs in the state are of this type.

(4) A low-water dam impoundment (Type L).

This type of impoundment is formed by building a low-water dam on a stream, creating impounded water only

during periods of low flow. Champagnolle Creek was the only Type L reservoir sampled during this study.

Results and Discussion

Eighty-five small fish species, representing approximately 59% of the small fish species native to Arkansas, were collected from 66 reservoirs statewide (Table 2). Appendix 1 lists the reservoirs in which each species occurred. Based on the number of individuals collected and the number of reservoirs in which a species was found (Table 2), the small fishes can be grouped into 3 categories:

(1) Species that maintained breeding populations in reservoirs.

Several of the small species occurred in numbers that indicated resident breeding populations. Presumably, the species which inhabit quiet-water areas of streams, such as brook silverside, *Labidesthes sicculus*, mosquitofish, *Gambusia affinis*, and blackspotted topminnow, *Fundulus olivaceus*, readily adapted to impoundments. Other stream species maintain sizeable populations in impoundments having extensive rocky substrates and shorelines. Some stream species can adapt to a variety of environmental conditions. For example, 2 of the logperch species, *Percina caprodes* and *P. fulvitaenia*, together occurred in more than one-half of the reservoirs sampled in a variety of environments but were most abundant in large impoundments having extensive areas of gravel bottoms and low turbidity.

(2) Species that occurred in low numbers in reservoirs and which may or may not have maintained breeding populations in the reservoirs.

Many of the small species found in this study can be placed in this category. For example, the slough darter, *Etheostoma gracile*, was one of the most widely distributed darters, occurring in 17 reservoirs in 4 ecoregions; however, it was found only in small numbers in those reservoirs.

(3) Species that occurred sporadically, probably as occasional stragglers from nearby tributaries.

Species represented by only 1 or a few specimens or those which occurred in only 1 out of several samples from a reservoir are included in this category Some reservoirs may provide temporary refuge for small species when tributary creeks go dry.

Sixteen Arkansas fish families contained species designated in this study as small, nongame fishes. Representatives of 13 of those families were found in Arkansas reservoirs. The three Arkansas fish families with none of their small species found in reservoirs were Umbridae (mudminnows), containing only a single species which has not been reported from Arkansas in more than 100 years; Amblyopsidae (cavefishes), containing 2 rare species restricted to subterranean environments; and Cottidae (sculpins), with two Arkansas species.

Seventy percent of the small species taken from Arkansas reservoirs were from the 2 largest Arkansas fish families, Cyprinidae (minnows, 35 species) and Percidae (darters, 25 species), and a number of those species were widely distributed and abundant (Table 2 and Appendix 1). The golden shiner (*Notemigonus crysoleucas*) was the most widely distributed cyprinid but was usually not found in large numbers. Its widespread distribution was undoubtedly due to its common use as a bait species. Lake Conway had the largest apparent breeding population of golden shiners. The bluntnose minnow, *Pimephales notatus*, was another widely distributed cyprinid and was usually taken in large numbers where it occurred. The bluntnose darter, *Etheostoma chlorosoma*, cypress darter, *E. proeliare*, logperch, *P. caprodes*, slough darter, *E. gracile*, and Ozark logperch, *P. fulvitaenia*, were the most widely distributed percids. The remaining 25 species were distributed among 11 other families as follows: Ictaluridae (6 species), Fundulidae (6 species), Centrarchidae (3 species), Catostomidae, (2 species), Atherinopsidae (2 species), and Petromyzontidae, Clupeidae, Esocidae, Aphredoderidae, Poeciliidae, and Elassomatidae with 1 species each.

A few species that occurred in large numbers were not widely distributed. The large numbers reported for the bullhead minnow, *Pimephales vigilax* (Table 2), were due mainly to large populations of that species in Type F impoundments on the Arkansas River. The checkered madtom, *Noturus flavater*, was 7th in abundance in 1999 samples but was collected from just 2 reservoirs (Bull Shoals and Norfork), both in the Ozark Highlands Ecoregion. In contrast, the tadpole madtom (*Noturus gyrinus*), which was also taken in large numbers, was much more widely distributed, occurring in 36 reservoirs in 4 of the 6 ecoregions of Arkansas. Large numbers of cypress minnows, *Hybognathus hayi*, were also collected in 1997-1999, but that species was found in only 1 impoundment, Felsenthal (Shallow Lake).

Three species, wedgespot shiner, *Notropis greenei*, Ozark shiner, *N. ozarcanus*, and fathead minnow, *Pimephales promelas*, were represented by a single specimen each. All of those species are uncommon in the natural waters of Arkansas and/or are restricted in their distributions in that state. The scarcity of *P. promelas* in reservoir population samples is surprising because that species is commonly reared in nursery ponds in Arkansas for release into reservoirs as forage fish.

Twelve small species were found in only a single reservoir (Table 2), and 5 of those species were taken in

more than 1 year of sampling. Ten species were taken from only 2 reservoirs. The johnny darter, *Etheostoma nigrum*, was collected from Lake Hinkle (Poteau River drainage) and 5 impoundments in the Saline River drainage. There are few previous records of *E. nigrum* from the eastern Saline River drainage, and all records from the Arkansas portion of the Poteau River drainage are pre-1960 records (Robison and Buchanan, 1988).

An unusual population of madtoms was found in Lake Ouachita in 1999. Three population samples on that reservoir produced 612 specimens reported as the brindled madtom, *Noturus miurus*. However, those specimens did not completely conform to all diagnostic morphological characters of *N. miurus*. Practically all of those specimens possessed the adipose fin pigmentation of the rare Caddo madtom, *N. taylori*, known from the upper part of the Ouachita River drainage above Lake Ouachita. Neil Douglas, who originally described *N. taylori* examined specimens of those madtoms and confirmed that they shared characters of both *N. miurus* and *N. taylori* and appeared to be intermediate between those 2 species. The Lake Ouachita population of madtoms may represent a case of introgressive hybridization, but there is insufficient evidence to completely support that hypothesis. Further study is needed to clarify this situation. The madtom specimens are herein tentatively reported as *N. miurus* because more of their characters conformed to that species than to *N. taylori*.

Another example of possible hybridization was found in Lake Millwood in 1998 with 2 logperch species. The bigscale logperch, *Percina macrolepida*, and logperch, *P. caprodes*, were both found in that reservoir, and some of the specimens taken from the middle and lower parts of that lake exhibited characters of both species. Buchanan et al. (1996) provided diagnostic features for separating Arkansas populations of *P. caprodes* and *P. macrolepida*. Further study of the Millwood specimens is required to confirm the occurrence (if any) and extent of hybridization.

The Ouachita madtom, *Noturus lachneri*, occurred in 6 Saline River drainage impoundments. This uncommon species is endemic to the Ouachita River drainage of Arkansas and is primarily restricted to clear, high-gradient streams of the Saline River headwaters (Robison and Buchanan, 1988). The United States Fish and Wildlife Service has periodically studied the status of known populations of *N. lachneri* but has not assigned that species a federally protected status. The ability of the Ouachita madtom to survive in reservoirs in small numbers has not been previously reported.

A Spearman rank correlation test showed a positive correlation ($p = 0.05$) between reservoir size and species richness. In general, large reservoirs from which multiple population samples were taken had more small fish species than small reservoirs (Table 1). Millwood, Bull Shoals, Greers Ferry, Dardanelle, DeGray, and Norfork were large impoundments that ranked near the top in species richness. Beaver Lake and Lake Ouachita were surprisingly low in species richness considering the large size and large number of small species historically known from the drainages of those reservoirs. Low species richness was found in Beaver Lake in 3 consecutive years of sampling (9 population samples from 1997 through 1999). Beaver Lake is the uppermost reservoir in a series of impoundments on the White River, and Lake Ouachita occupies the same position in a series of impoundments on the Ouachita River. Thornton et al. (1990) documented some of the differences in physicochemical and other parameters in reservoirs occurring in series on a river, but little attention has been focused on quantifying differences in fish populations in such circumstances. The low species richness of Beaver and Ouachita may be related to the uppermost position in a series that each occupies, although further study would be required to adequately assess that hypothesis. The reservoirs downstream from Beaver Lake on the White River, (Bull Shoals and Norfork) ranked high in species richness. Table Rock is the next reservoir downstream from Beaver Lake, but only a small portion of that reservoir is in Arkansas, and it is not routinely sampled by the Arkansas Game & Fish Commission. The reservoirs downstream from Lake Ouachita on the Ouachita River (Lake Hamilton and Lake Catherine, both sampled in 1997) ranked low in the number of small species. Hamilton and Catherine, however, are both much smaller than the White River reservoirs downstream from Beaver Lake and were not sampled as extensively as those reservoirs in this study. Other variables may also contribute to the low species richness of Beaver and Ouachita.

Another method of assessing the small species richness in a reservoir is to determine what percentage of the small species historically known from a reservoir's drainage area occur in that reservoir (Table 1). It was difficult to accurately determine the number of fish species historically known from the drainage area of most Arkansas impoundments because of the lack of preimpoundment fish surveys in the state. For most Arkansas reservoirs, information on historic species distribution must be inferred from scattered preimpoundment fish samples (taken mainly by seine) and from the more extensive sampling in recent decades (most of which was postimpoundment sampling) reported by Robison and Buchanan (1988). The number of small species in the drainage areas of Type S and Type L impoundments was defined as the number of species historically known to occur in the stream and its tributaries upstream from the dam site (determined largely from the distribution maps of Robison and Buchanan, 1988). For Type F reservoirs on the large, navigable rivers (Arkansas and Ouachita rivers), the number of small species in the drainage was considered to be the number of species historically known from the

main channel in the vicinity of the reservoir. For Type P impoundments the potential number of small species was defined as the number of species historically known from the drainage serving as the source of the pumped water. Table 1 lists the percentage of the small species known from the drainage area that was found in each reservoir. The number of small species found in reservoirs was positively correlated ($P = 0.05$) with the number of small species occurring in reservoir drainages.

The threadfin shad, *D. petenense*, was by far the most abundant small species due to the large numbers taken from a few of the largest reservoirs (Table 2). The number of *D. petenense* collected varied from year to year. For a 3-year period, the numbers of *D. petenense* collected were as follows: 1997 (226,362), 1998 (849,256), and 1999 (123,979). The most widely distributed species was *L. sicculus*, found in 86% of the reservoirs sampled. Other widely distributed species occurring in at least 30% of the reservoirs sampled were as follows: *Gambusia affinis* (74%), *F. olivaceus* (71%), *D. petenense* (62%), *N. crysoleucas* (61%), *N. gyrinus* (55%), *E. chlorosoma* (45%), *Aphredoderus sayanus* (45%), *P. notatus* (44%), *Opsopoeodus emiliae* (38%), *E. proeliare* (36%), *P. caprodes* (35%), and *Campostoma anomalum* (30%).

Five species, *D. petenense*, *N. crysoleucas*, *L. sicculus*, *F. olivaceus*, and *E. proeliare*, occurred in reservoirs in all 6 ecoregions (Table 2). Seven other species were taken in 5 ecoregions, 9 species occurred in 4 ecoregions, 15 species occurred in 3 ecoregions, 26 species occurred in 2 ecoregions, and 23 species occurred in only 1 ecoregion.

There were distinct differences among the 6 ecoregions of Arkansas in the number of small species found. The ecoregions with number of species in parenthesis were Gulf Coastal Plain (50), Arkansas River Valley (48), Ouachita Mountains (37), Delta (34), Ozark Highlands (28), and Boston Mountains (25). The number of small species found in the 6 ecoregions was correlated with the number of reservoirs sampled in those ecoregions (Spearman rank correlation test, $P = 0.05$). The low species richness of the Boston Mountains and Ozark Highlands ecoregions was due mainly to the small number of reservoirs sampled. Those ecoregions have fewer impoundments than the others, but 3 reservoirs in those regions (Bull Shoals, Greers Ferry, and Norfork) ranked high in small species richness. Other variables, such as size of reservoirs sampled, the number of small species occurring in reservoir drainages, and human impact on natural drainages, could also contribute to differences in small species richness among ecoregions.

Five of the 10 reservoirs (Mallard, Charles, Poinsett, Frierson, and Hogue) that ranked lowest in species richness were in the Delta Ecoregion. All 5 of those reservoirs were small (< 263 ha), and all had a low number of small species historically known from their drainage areas (Table 1). Those reservoirs were also low in the percentage of the species known from the drainage areas captured in those reservoirs.

Eighteen reservoirs were sampled in 3 or more years of this study. Most of those reservoirs varied only slightly in species richness from year-to-year. Those reservoirs that exhibited high species richness were high in all years, and those with low species richness were low in all years. The greatest variation in species composition occurred in Felsenthal (Shallow Lake), a flow-through impoundment of the Ouachita River with a strong riverine influence. Other Type F impoundments also had greater variation in species composition from year-to-year than the other types of impoundments.

Cove rotenone population sampling used in this study does not sample all possible habitats within a reservoir; however, rotenone sampling probably captures a large percentage of the small fish species in a reservoir because most of those species (with the exception of schooling cyprinids and threadfin shad) prefer shallow water environments. Most of the coves sampled in this study contained a variety of microhabitats, including open water areas with depths of 10 to 15 m in addition to the shallow areas with varying types of substrate and vegetation.

A single rotenone population sample from a reservoir is not likely to capture 100% of the small species in the reservoir. A 2-day sample in 1998 on Lake DeQueen, a small reservoir with little heterogeneity of environments, produced 11 small species from the sample cove. Three additional small rotenone samples (conducted on the 1st day of the cove sample) from other areas of Lake DeQueen yielded only 1 additional small species (orangebelly darter, *Etheostoma radiosum*) not found in the cove sampling. In contrast, a rotenone samples on 16-17 July 1997 in Lake Erling, a moderately large impoundment with great heterogeneity of shallow water environments, produced 10 small species. Small-scale rotenone samples on 19 July 1999 in the upper, middle, and lower parts of Lake Erling yielded 18 small species, including 1 species, the ironcolor shiner, *Notropis chalybaeus*, not collected from any other Arkansas reservoir.

Although a single cove rotenone sample probably does not capture representatives of all small fish species in the reservoir, multiple samples should capture a high percentage of the small species in most reservoirs. Multiple coves were sampled from the larger reservoirs included in this study. For example, 2 coves were sampled on Conway, Dardanelle, and Greeson, and 3 coves were sampled on Beaver, Bull Shoals, DeGray, Greers Ferry, and Ouachita in years when those reservoirs were sampled. It is unlikely that seining, shocking, gill netting, or trapping would produce many small species not taken by rotenone sampling in Type S, Type P, and Type L reservoirs. That may not be true for the flow-through impoundments (Type F). Sampling by methods other than rotenone in fall, winter, or spring could

produce some different species records than the summer sampling employed in this study.

Even though 59% of the small fish species native to Arkansas were found in reservoirs in this study, it is unlikely that all of those species maintain breeding populations in those reservoirs. Based on number of specimens collected and number of reservoirs in which a species was found, it is estimated that less than 50% of the small native species can maintain breeding populations in some reservoirs. This study should provide information for comparing and interpreting future successional changes in reservoir fish communities as the reservoirs age.

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Table 1. Arkansas reservoirs sampled by rotenone, 1997-2004. Reservoir types are flow-through (F), leveed impoundment where water is pumped in (P), impoundment built by damming a stream (S), and low-water dam impoundment (L). The ecoregions of Arkansas are A (Arkansas River Valley), B (Boston Mountains), D (Delta), G (Gulf Coastal Plain), Ou (Ouachita Mountains), and Oz (Ozark Highlands).

Reservoir Name	Type	Surface area (ha)	Ecoregion	Number of small species	% of small species known from drainage captured in reservoir
1. Ashbaugh	P	202	D	12	21
2. Atkins	S	304	A	7	54
3. Balboa	S	405	Ou	14	47
4. Barnett	S	106	A	8	31
5. Beaver	S	11,421	Oz	13	28
6. Blue Mountain	S	1,178	A	24	60
7. Bob Kidd	S	81	B	1	3
8. Bois D'Arc	S	263	G	6	33
9. Brewer	S	445	A	16	40
10. Bull Shoals	S	15,095	Oz	25	58
11. Calion	S	202	G	15	60
12. Cane Creek	S	688	G	16	64
13. Cargile	S	58	A	7	19
14. Catherine	S	785	Ou	13	27

Table 1. Continued.

Reservoir Name	Type	Surface area (ha)	Ecoregion	Number of small species	% of small species known from drainage captured in reservoir
15. Champagnolle Creek	L	86	G	20	125
16. Charles	S	261	D	6	35
17. Columbia	S	1,194	G	12	41
18. Coronado	S	155	Ou	11	37
19. Conway	S	2,711	A	14	70
20. Cortez	S	99	Ou	10	33
21. Dardanelle	F	13,881	A	29	66
22. DeGray	S	5,423	Ou	21	43
23. DeQueen	S	680	Ou	11	52
24. Des Arc	P	142	D	7	25
25. DeSoto	S	81	Ou	10	33
26. Dierks	S	550	Ou	13	42
27. Elmdale	S	51	Oz	1	3
28. Erling	S	2,873	G	18	86
29. Felsenthal (Shallow Lake)	F	5,868	G	28	58
30. Frierson	S	95	D	5	20
31. Georgia-Pacific	P	688	G	17	31
32. Gillham	S	554	Ou	15	45
33. Greers Ferry	S	12,748	B	25	61
34. Greeson	S	2,938	Ou	18	40
35. Hamilton	S	3,019	Ou	10	23
36. Harris Brake	S	526	A	7	54
37. Hinkle	S	389	A	16	47
38. Hogue	P	113	D	1	5
39. Isabella	S	11	Ou	2	7
40. Jack Nolan	S	84	A	6	27
41. Leatherwood	S	40	Oz	1	4
42. Lower White Oak	S	405	G	14	40
43. Mallard	P	121	D	6	23
44. Maumelle	S	3,602	A	15	52
45. Merrisach	S	809	D	14	52

Table 1. Continued.

Reservoir Name	Type	Surface area (ha)	Ecoregion	Number of small species	% of small species known from drainage captured in reservoir
46. Millwood	S	12,141	G	32	51
47. Monticello	S	607	G	5	25
48. Nimrod	S	1,457	A	20	54
49. Norfork	S	8,337	Oz	21	53
50. Ouachita	S	16,228	Ou	12	29
51. Overcup	S	415	A	8	35
52. Ozark Pool (Arkansas R.)	F	4,290	A	20	56
53. Peckerwood	S	1,619	D	11	50
54. Pineda	S	25	Ou	11	37
55. Poinsett	S	223	D	5	26
56. Pool 2 (Arkansas R.)	F	1,485	D	27	90
57. Pool 7 (Arkansas R.)	F	3,927	A	27	90
58. Pool 8 (Arkansas R.)	F	1,672	A	13	43
59. Pool 9 (Arkansas R.)	F	1,987	A	23	85
60. Pool 13 (Arkansas R.)	F	2,307	A	21	58
61. Sugarloaf	S	136	A	7	28
62. Swepco	S	214	Oz	2	12
63. Tri-County	S	127	G	9	64
64. Upper White Oak	S	324	G	12	34
65. Wilhelmina	S	81	Ou	5	28
66. Winona	S	289	Ou	13	52

Table 2. Small fish species found in Arkansas reservoirs, 1997-2004. The ecoregions are Arkansas River Valley (A), Boston Mountains (B), Delta (D), Gulf Coastal Plain (G), Ouachita Mountains (Ou), and Ozark Highlands (Oz).

Fish species	No. of reservoirs found in (n = 66)	No. of specimens collected	Ecoregions
Ichthyomyzon castaneus	4	5	A, B, Ou, Oz
Dorosoma petenense	41	1,198,076	A, B, D, G, Ou, Oz
Campostoma anomalum	20	720	A, B, Ou, Oz
Campostoma oligolepis	4	1,082	B, Oz

Table 2. Continued.

Fish species	No. of reservoirs found in (n = 66)	No. of specimens collected	Ecoregions
Cyprinella galactura	2	30	Oz
Cyprinella lutrensis	7	873	A, D
Cyprinella venusta	12	1,819	A, D, G
Cyprinella whipplei	6	782	A, B, Ou, Oz
Erimystax harryi	1	15	Oz
Hybognathus hayi	1	6,293	G
Hybognathus nuchalis	6	8,476	A, D, G
Hybopsis amblops	1	49	Oz
Hybopsis amnis	8	3,697	A, G, Ou
Luxilus cardinalis	1	5	A
Luxilus chrysocephalus	3	71	G, Oz
Luxilus pilsbryi	4	446	B, Oz
Lythrurus fumeus	1	28	G
Lythrurus snelsoni	1	8	Ou
Lythrurus umbratilis	5	90	G, Ou
Macrhybopsis storeriana	6	1,398	A, D
Notemigonus crysoleucas	40	3,494	A, B, D, G, Ou, Oz
Notropis atherinoides	12	2,882	A, D, G, Ou
Notropis blennius	4	122	A, D
Notropis boops	12	1,593	A, B, Ou
Notropis buchanani	5	1,215	A
Notropis chalybaeus	1	13	G
Notropis greenei	1	1	Oz
Notropis maculatus	8	1,664	D, G
Notropis ozarcanus	1	1	Oz
Notropis rubellus	2	21	Oz
Notropis texanus	5	313	A, D, G
Notropis volucellus	5	72	A, D
Opsopoeodus emiliae	25	11,888	A, B, D, G, Ou
Pimephales notatus	29	39,006	A, B, G, Ou, Oz
Pimephales promelas	1	1	Oz

Table 2. Continued.

Fish species	No. of reservoirs found in (n = 66)	No. of specimens collected	Ecoregions
Pteronotropis hubbsi	2	43	G
Erimyzon oblongus	2	11	A, G
Erimyzon sucetta	7	422	D, G
Noturus exilis	7	1,965	A, B, Ou
Noturus flavater	2	4,149	Oz
Noturus gyrinus	36	5,770	A, D, G, Ou
Noturus lachneri	6	329	Ou
Noturus miurus	7	1,977	B, G, Ou
Noturus nocturnus	2	112	A, G
Esox americanus	9	141	A, G
Aphredoderus sayanus	30	2,362	A, D, G, Ou
Labidesthes sicculus	57	20,123	A, B, D, G, Ou, Oz
Menidia beryllina	17	21,287	A, D, G, Ou
Fundulus blairae	1	50	G
Fundulus catenatus	2	9	Ou, Oz
Fundulus chrysotus	11	1,380	A, D, G
Fundulus dispar	3	111	G
Fundulus notatus	9	97	A, D, G, Ou
Fundulus olivaceus	47	4,354	A, B, D, G, Ou, Oz
Gambusia affinis	49	5,636	A, D, G, Ou, Oz
Elassoma zonatum	9	80	A, G
Lepomis humilis	16	13,500	A, D, G
Lepomis marginatus	9	661	G
Lepomis symmetricus	6	180	D, G, Ou
Ammocrypta vivax	2	14	G, Ou
Etheostoma artesiae	7	74	G, Ou
Etheostoma asprigene	5	177	A, D, G
Etheostoma blennioides	5	50	B, Ou, Oz
Etheostoma caeruleum	4	1,532	B, Oz
Etheostoma chlorosoma	30	2,478	A, B, D, G, Ou
Etheostoma collettei	9	704	G, Ou
Etheostoma fusiforme	3	78	D, G

Table 2. Continued.

Fish species	No. of reservoirs found in (n = 66)	No. of specimens collected	Ecoregions
Etheostoma gracile	17	135	A, D, G, Ou
Etheostoma nigrum	6	138	A, Ou
Etheostoma proeliare	24	382	A, B, D, G, Ou, Oz
Etheostoma punctulatum	4	716	B, Oz
Etheostoma radiosum	6	409	Ou
Etheostoma spectabile	4	312	A, Oz
Etheostoma stigmaeum	2	23	B, Oz
Etheostoma whipplei	8	228	A, B
Percina caprodes	23	15,230	B, D, G, Ou, Oz
Percina copelandi	5	136	A, Ou
Percina fulvitaenia	13	8,683	A
Percina macrolepida	8	389	A, D, G
Percina maculata	9	112	A, B, D, G, Ou
Percina nasuta	1	7	B
Percina phoxocephala	2	4	A
Percina sciera	9	41	A, D, G
Percina shumardi	8	1,703	A, D, G

Fig. 1–Locations of the 66 reservoirs sampled, 1997-2004, and the 6 ecoregions of Arkansas. Numbers correspond to reservoir numbers in Table 1.

Appendix 1. The Arkansas reservoirs in which each small fish species occurred, 1997-2004.

Species	Reservoirs
Ichthyomyzon castaneus	Bull Shoals, DeGray, Greers Ferry, Pool 13
Dorosoma petenense	Balboa, Barnett, Beaver, Blue Mountain, Brewer, Bull Shoals, Calion, Catherine, Champagnolle Creek, Columbia, Conway, Dardanelle, DeGray, DeQueen, Dierks, Erling, Felsenthal, Georgia-Pacific, Gillham, Greers Ferry, Greeson, Hamilton, Hinkle, Jack Nolan, Lower White Oak, Maumelle, Merrisach, Millwood, Monticello, Norfork, Ouachita, Ozark Pool, Pool 2, Pool 7, Pool 8, Pool 9, Pool 13, Sugarloaf, Swepco, Upper White Oak, Winona

Appendix 1. Continued.

Species	Reservoirs
Campostoma anomalum	Balboa, Blue Mountain, Brewer, Bull Shoals, Coronado, Cortez, Dardanelle, DeGray, DeQueen, DeSoto, Dierks, Gillham, Greers Ferry, Greeson, Hinkle, Isabella, Nimrod, Norfork, Pineda, Winona
Campostoma oligolepis	Beaver, Bull Shoals, Greers Ferry, Norfork
Cyprinella galactura	Bull Shoals, Norfork
Cyprinella lutrensis	Dardanelle, Ozark Pool, Pool 2, Pool 7, Pool 8, Pool 9, Pool 13
Cyprinella venusta	Blue Mountain, Dardanelle, Erling, Mallard, Millwood, Nimrod, Ozark Pool, Poinsett, Pool 2, Pool 7, Pool 8, Pool 9
Cyprinella whipplei	Beaver, Brewer, Bull Shoals, Dardanelle, DeGray, Greers Ferry
Erimystax harryi	Bull Shoals
Hybognathus hayi	Felsenthal
Hybognathus nuchalis	Champagnolle Creek, Felsenthal, Nimrod, Pool 2, Pool 7, Pool 9
Hybopsis amblops	Bull Shoals
Hybopsis amnis	DeGray, Dierks, Felsenthal, Georgia-Pacific, Gillham, Hinkle, Millwood, Pool 7
Luxilus cardinalis	Dardanelle
Luxilus chrysocephalus	Bull Shoals, Millwood, Norfork
Luxilus pilsbryi	Beaver, Bull Shoals, Greers Ferry, Norfork
Lythrurus fumeus	Felsenthal
Lythrurus snelsoni	Gillham
Lythrurus umbratilis	DeSoto, Dierks, Gillham, Millwood, Pineda
Macrhybopsis storeriana	Dardanelle, Ozark Pool, Pool 2, Pool 7, Pool 9, Pool 13
Notemigonus crysoleucas	Atkins, Beaver, Bob Kidd, Bois D'Arc, Brewer, Calion, Cane Creek, Catherine, Champagnolle Creek, Charles, Columbia, Conway, Dardanelle, DeQueen, Dierks, Elmdale, Erling, Frierson, Gillham, Greers Ferry, Harris Brake, Jack Nolan, Lower White Oak, Mallard, Merrisach, Millwood, Monticello, Nimrod, Norfork, Ouachita, Overcup, Ozark Pool, Peckerwood, Pineda, Poinsett, Pool 2, Sugarloaf, Swepco, Tri-County, Upper White Oak

Appendix 1. Continued.

Species	Reservoirs
Notropis atherinoides	Blue Mountain, Dardanelle, Felsenthal, Gillham, Millwood, Nimrod, Ozark Pool, Pool 2, Pool 7, Pool 8, Pool 9, Pool 13
Notropis blennius	Ozark Pool, Pool 2, Pool 7, Pool 13
Notropis boops	Balboa, Blue Mountain, Catherine, Cortez, DeGray, Desoto, Gillham, Greers Ferry, Greeson, Hinkle, Maumelle, Winona
Notropis buchanani	Dardanelle, Ozark Pool, Pool 7, Pool 9, Pool 13
Notropis chalybaeus	Erling
Notropis greenei	Bull Shoals
Notropis maculatus	Ashbaugh, Calion, Cane Creek, Champagnolle Creek, Felsenthal, Merrisach, Millwood, Peckerwood
Notropis ozarcanus	Bull Shoals
Notropis rubellus	Bull Shoals, Norfork
Notropis texanus	Ashbaugh, Felsenthal, Georgia-Pacific, Nimrod, Pool 7
Notropis volucellus	Blue Mountain, Dardanelle, Pool 2, Pool 7, Pool 13
Opsopoeodus emiliae	Ashbaugh, Blue Mountain, Calion, Cane Creek, Catherine, Champagnolle Creek, Columbia, Conway, Dardanelle, Dierks, Felsenthal, Georgia-Pacific, Greers Ferry, Harris Brake, Hinkle, Merrisach, Millwood, Nimrod, Ozark Pool, Peckerwood, Pool 2, Pool 7, Pool 9, Pool 13, Tri-County
Pimephales notatus	Balboa, Blue Mountain, Brewer, Bull Shoals, Catherine, Coronado, Cortez, Dardanelle, DeGray, DeQueen, DeSoto, Dierks, Georgia-Pacific, Gillham, Greers Ferry, Greeson, Hamilton, Hinkle, Lower White Oak, Maumelle, Millwood, Nimrod, Norfork, Ouachita, Ozark Pool, Pineda, Pool 13, Wilhelmina, Winona
Pimephales promelas	Bull Shoals
Pimephales vigilax	Ashbaugh, Blue Mountain, Calion, Catherine, Dardanelle, Felsenthal, Georgia-Pacific, Greers Ferry, Greeson, Mallard, Ozark Pool, Pool 2, Pool 7, Pool 8, Pool 9, Pool 13
Pteronotropis hubbsi	Champagnolle Creek, Millwood
Erimyzon oblongus	Blue Mountain, Champagnolle Creek
Erimyzon sucetta	Champagnolle Creek, Columbia, Erling, Felsenthal, Lower White Oak, Merrisach, Upper White Oak

Appendix 1. Continued.

Species	Reservoirs
Noturus exilis	Beaver, Brewer, Bull Shoals, Greers Ferry, Hinkle, Maumelle, Norfork
Noturus flavater	Bull Shoals, Norfork
Noturus gyrinus	Ashbaugh, Atkins, Barnett, Blue Mountain, Bois D'Arc, Brewer, Calion, Cane Creek, Cargile, Catherine, Champagnolle Creek, Charles, Conway, Dardanelle, DeGray, DeQueen, Des Arc, Erling, Felsenthal, Georgia-Pacific, Gillham, Greeson, Hamilton, Harris Brake, Lower White Oak, Mallard, Maumelle, Merrisach, Millwood, Nimrod, Overcup, Peckerwood, Poinsett, Pool 2, Pool 9, Tri-County
Noturus lachneri	Balboa, Coronado, Cortez, DeSoto, Pineda, Winona
Noturus miurus	Bois D'Arc, DeGray, Georgia-Pacific, Greers Ferry, Greeson, Hamilton, Ouachita
Noturus nocturnus	Felsenthal, Pool 9
Esox americanus	Columbia, Conway, Felsenthal, Lower White Oak, Millwood, Nimrod, Pool 7, Pool 9, Upper White Oak
Aphredoderus sayanus	Ashbaugh, Atkins, Balboa, Blue Mountain, Brewer, Calion, Cane Creek, Cargile, Champagnolle Creek, Columbia, Conway, Cortez, Erling, Felsenthal, Georgia-Pacific, Greeson, Harris Brake, Lower White Oak, Maumelle, Millwood, Nimrod, Ouachita, Overcup, Pineda, Pool 2, Pool 7, Pool 9, Sugarloaf, Upper White Oak, Winona
Labidesthes sicculus	Ashbaugh, Atkins, Balboa, Barnett, Beaver, Blue Mountain, Brewer, Bull Shoals, Calion, Cane Creek, Cargile, Catherine, Champagnolle Creek, Columbia, Conway, Coronado, Cortez, Dardanelle, DeGray, DeQueen, Des Arc, DeSoto, Dierks, Erling, Felsenthal, Frierson, Georgia-Pacific, Gillham, Greers Ferry, Greeson, Hamilton, Harris Brake, Hinkle, Jack Nolan, Lower White Oak, Mallard, Maumelle, Merrisach, Millwood, Monticello, Nimrod, Norfork, Ouachita, Overcup, Ozark Pool, Peckerwood, Pineda, Pool 2, Pool 7, Pool 8, Pool 9, Pool 13, Sugarloaf, Tri-County, Upper White Oak, Wilhelmina, Winona
Menidia beryllina	Bois D'Arc, Charles, Conway, Dardanelle, DeGray, Felsenthal, Hamilton, Merrisach, Millwood, Monticello, Ozark Pool, Peckerwood, Pool 2, Pool 7, Pool 8, Pool 9, Pool 13
Fundulus blairae	Millwood
Fundulus catenatus	Bull Shoals, DeGray
Fundulus chrysotus	Cane Creek, Champagnolle Creek, Conway, Dardanelle, Erling, Felsenthal, Merrisach, Millwood, Peckerwood, Tri-County, Upper White Oak

Appendix 1. Continued.

Species	Reservoirs
Fundulus dispar	Cane Creek, Champagnolle Creek, Felsenthal
Fundulus notatus	Catherine, Conway, Des Arc, Erling, Felsenthal, Merrisach, Millwood, Pool 2, Pool 7
Fundulus olivaceus	Atkins, Balboa, Barnett, Beaver, Blue Mountain, Brewer, Bull Shoals, Calion, Cane Creek, Cargile, Catherine, Champagnolle Creek, Charles, Conway, Coronado, Dardanelle, DeGray, Des Arc, Dierks, Erling, Frierson, Georgia-Pacific, Gillham, Greers Ferry, Greeson, Hamilton, Harris Brake, Hinkle, Lower White Oak, Maumelle, Monticello, Nimrod, Norfork, Ouachita, Overcup, Ozark Pool, Peckerwood, Pineda, Poinsett, Pool 2, Pool 7, Pool 8, Pool 9, Pool 13, Sugarloaf, Tri-County, Winona
Gambusia affinis	Atkins, Barnett, Beaver, Blue Mountain, Bois D'Arc, Brewer, Calion, Cane Creek, Cargile, Catherine, Champagnolle Creek, Charles, Columbia, Conway, Coronado, Cortez, Dardanelle, DeGray, DeQueen, Des Arc, Dierks, Erling, Felsenthal, Frierson, Georgia-Pacific, Gillham, Greeson, Hinkle, Jack Nolan, Leatherwood, Lower White Oak, Maumelle, Merrisach, Millwood, Nimrod, Norfork, Ouachita, Overcup, Peckerwood, Poinsett, Pool 2, Pool, 7, Pool 8, Pool 9, Pool 13, Sugarloaf, Tri-County, Upper White Oak, Wilhelmina
Elassoma zonatum	Brewer, Cane Creek, Cargile, Champagnolle Creek, Columbia, Georgia-Pacific, Lower White Oak, Maumelle, Millwood
Lepomis humilis	Blue Mountain, Calion, Cane Creek, Charles, Dardanelle, Felsenthal, Georgia-Pacific, Mallard, Nimrod, Ozark Pool, Peckerwood, Pool 2, Pool 7, Pool 9, Pool 13, Tri-County
Lepomis marginatus	Calion, Cane Creek, Champagnolle Creek, Columbia, Erling, Felsenthal, Lower White Oak, Millwood, Upper White Oak
Lepomis symmetricus	Champagnolle Creek, Dierks, Erling, Merrisach, Millwood, Pool 2
Ammocrypta vivax	Greeson, Millwood
Etheostoma artesiae	Balboa, Coronado, DeSoto, Erling, Isabella, Pineda, Winona
Etheostoma asprigene	Ashbaugh, Felsenthal, Pool 2, Pool 8, Pool 9
Etheostoma blennioides	Balboa, Bull Shoals, DeGray, Greers Ferry, Norfork
Etheostoma caeruleum	Beaver, Bull Shoals, Greers Ferry, Norfork

Appendix 1. Continued.

Species	Reservoirs
Etheostoma chlorosoma	Ashbaugh, Blue Mountain, Brewer, Calion, Cane Creek, Cargile, Catherine, Columbia, Conway, DeGray, Des Arc, Erling, Felsenthal, Georgia-Pacific, Greers Ferry, Greeson, Hamilton, Harris Brake, Hogue, Lower White Oak, Maumelle, Millwood, Nimrod, Ouachita, Overcup, Peckerwood, Pool 2, Pool 7, Tri-County, Upper White Oak
Etheostoma collettei	Balboa, Coronado, Cortez, DeQueen, DeSoto, Gillham, Greeson, Pineda, Winona
Etheostoma fusiforme	Champagnolle Creek, Merrisach, Millwood
Etheostoma gracile	Atkins, Barnett, Blue Mountain, Bois D'Arc, Calion, Cane Creek, Columbia, DeQueen, Erling, Frierson, Jack Nolan, Lower White Oak, Maumelle, Millwood, Overcup, Sugarloaf, Upper White Oak
Etheostoma nigrum	Balboa, Coronado, Cortez, DeSoto, Hinkle, Winona
Etheostoma proeliare	Ashbaugh, Balboa, Barnett, Blue Mountain, Brewer, Cane Creek, Champagnolle Creek, Coronado, Cortez, DeGray, DeQueen, DeSoto, Erling, Georgia-Pacific, Greers Ferry, Greeson, Hinkle, Jack Nolan, Norfork, Ouachita, Pineda, Pool 2, Upper White Oak, Winona
Etheostoma punctulatum	Beaver, Bull Shoals, Greers Ferry, Norfork
Etheostoma radiosum	DeGray, Dierks, Greeson, Hamilton, Ouachita, Wilhelmina
Etheostoma spectabile	Beaver, Bull Shoals, Hinkle, Norfork
Etheostoma stigmaeum	Greers Ferry, Norfork
Etheostoma whipplei	Blue Mountain, Brewer, Dardanelle, Greers Ferry, Hinkle, Maumelle, Nimrod, Pool 13
Percina caprodes	Ashbaugh, Balboa, Beaver, Bull Shoals, Calion, Catherine, Coronado, DeGray, DeQueen, Des Arc, Dierks, Felsenthal, Georgia-Pacific, Gillham, Greers Ferry, Greeson, Hamilton, Merrisach, Millwood, Norfork, Ouachita, Wilhelmina, Winona
Percina copelandi	Blue Mountain, Dardanelle, DeGray, Greeson, Nimrod
Percina fulvitaenia	Blue Mountain, Brewer, Conway, Dardanelle, Hinkle, Maumelle, Nimrod, Ozark Pool, Pool 2, Pool 7, Pool 8, Pool 9, Pool 13
Percina macrolepida	Dardanelle, Millwood, Ozark Pool, Pool 2, Pool 7, Pool 8, Pool 9, Pool 13
Percina maculata	Ashbaugh, Barnett, Blue Mountain, Dardanelle, DeGray, Felsenthal, Greers Ferry, Nimrod, Pool 7

Appendix 1. Continued.

Species	Reservoirs
Percina nasuta	Greers Ferry
Percina phoxocephala	Dardanelle, Ozark Pool
Percina sciera	Blue Mountain, Dardanelle, Felsenthal, Millwood, Ozark Pool, Pool 2, Pool 7, Pool 9, Pool 13
Percina shumardi	Dardanelle, Millwood, Ozark Pool, Pool 2, Pool 7, Pool 8, Pool 9, Pool 13

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Fifty-four State Records of True Bugs (Hemiptera: Heteroptera) from Arkansas

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Abstract:

The terrestrial true bug (Hemiptera : Heteroptera) fauna of Arkansas is poorly represented in the literature. Between 1998 and 2004, we retained Hemiptera specimens collected while conducting a few scattered entomological projects. Ninety-nine species of terrestrial Hemiptera, representing 15 families, were collected from various locations within 9 Arkansas counties. Of these 99 species, 54 are new state records for Arkansas. The majority of these 54 new state records are of common, widespread species that would be expected for Arkansas. Twenty-two of the 54 species have been reported for at least 4 states bordering Arkansas, whereas only 5 species (all Miridae) were not previously reported for any bordering state. Our specimens of *Pycnoderes convexicollis* (Blatchley, 1926) represent a fairly significant range extension for this species, previously known only from Indiana and Illinois.

Introduction

The aquatic and semi-aquatic true bug (Hemiptera: Heteroptera) fauna of Arkansas is fairly well known. There have been several state-wide studies (Chordas and Harp, 1991; Harp and Harp, 1990; Harp, 1985) as well as other regional aquatic investigations (e.g., Chordas et al, 1996; Cochran and Harp, 1990; Harp and Harp, 1980) that included Hemiptera. Conversely, the terrestrial Hemiptera of Arkansas are less well documented. There are 5 terrestrial hemipteran families that have been comprehensively investigated for Arkansas: Aradidae (flat bugs; Taylor and McPherson, 1989), Pentatomidae (stink bugs; Barton and Lee, 1981) and Cydnidae (burrower bugs), Scutelleridae (shield back bugs), Thyrecoridae (negro bugs; Lee and Barton, 1983). The remaining terrestrial Hemiptera are largely underreported for Arkansas. Based on distribution records in the *Catalog of True Bugs* (Henry and Froeschner, 1988), 20 families of terrestrial hemipterans should occur in Arkansas. However, only 17 have been recorded in the literature. Three small families, Enicocephalidae (unique-headed bugs), Largidae (largid bugs), Piesmatidae (piesmatid bugs), lack literature records for Arkansas (Henry and Froeschner, 1988). Further, many of the hemipteran families that are recorded for Arkansas have several common, widespread species, which, although expected for the state, are as of yet unreported in the literature.

We retained Hemiptera that were collected during a few of our ongoing localized insect projects. The purposes of this paper are to report 54 hemipteran species as new state records for Arkansas and to provide a list of hemipteran species that we collected during our isolated projects.

Materials and Methods

Hemipterans were collected during sampling for projects targeting other insect groups. Bugs were collected in sweepnets, pitfall traps, black light pan traps, and sheets. Bugs were also collected with beating sheets or aspirated/hand captured. Collections were made while investigating the Diptera fauna of the White River National Wildlife Refuge (Chordas et al., 2004), the insect fauna of springs in the Ouachita Highlands in the Ouachita National Forest (i.e., Beyers and Robison, 1997), the insect fauna inhabiting pocket gopher borrows (both *Geomys bursarius ozarkensis* and *Geomys breviceps*) in the White River basin, and aquatic Hemiptera (plus general) collecting in wildlife or park areas. Samples from 17 collection sites in 9 counties contained hemipteran specimens that were identifiable (Fig. 1, Table 1). Specimens were preserved in 70-80% ethanol. Voucher specimens of state records were deposited in the University of Arkansas arthropod museum (Fayetteville, Arkansas). Remaining specimens were deposited in the senior author's collection (SWAC Collection, Columbus Ohio).

Blatchley (1926), Kelton (1978), Knight (1941),

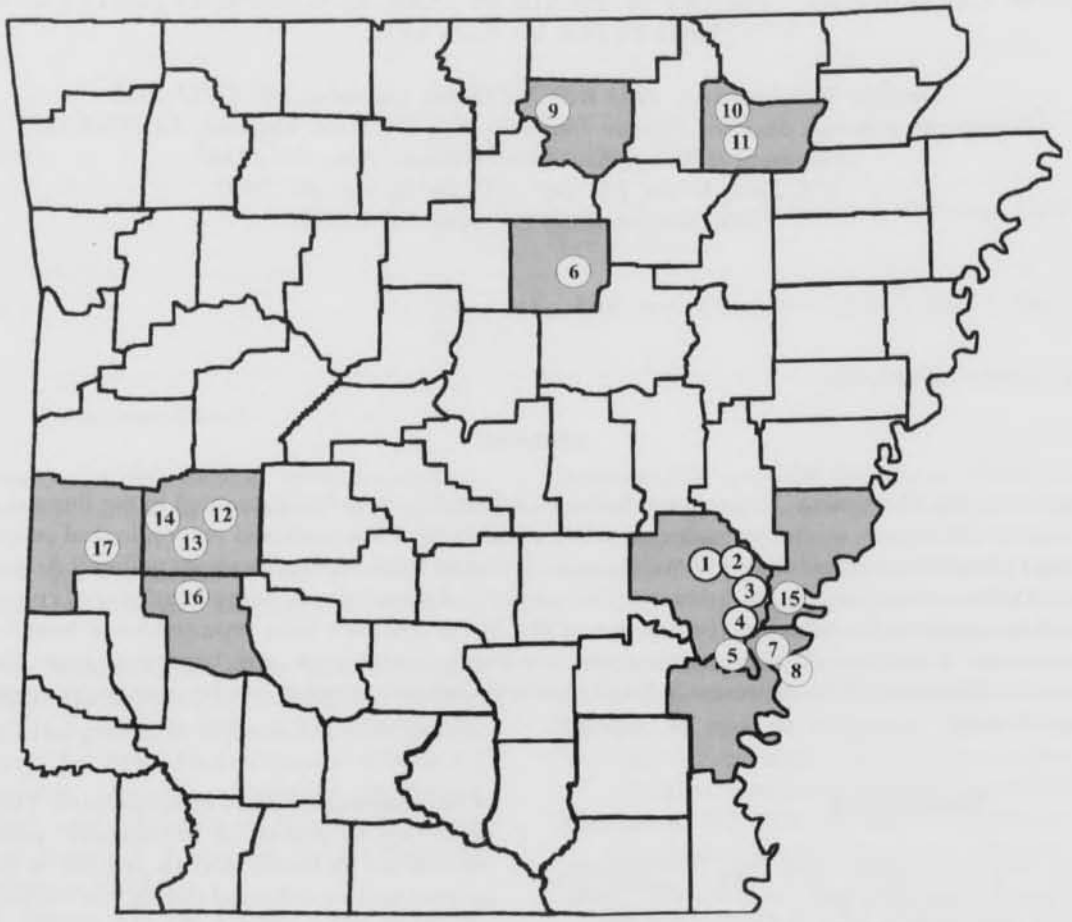

Fig. 1. Collection sites (see Table 1 for site specifics).

McPherson (1982), McPherson et al. (1990), and Moore (1955) were used as taxonomic references, and a few specimens were sent to recognized experts for verification (see Acknowledgements). Only specimens that were confidently determined to the species level are included herein. Barton and Lee (1981), Blatchley (1926), Lee and Barton (1983), Henry and Froeschner (1988), Lariviere and Larochelle (1991), McPherson (1982), McPherson et al. (1990), McPherson et al. (1991) and Taylor and McPherson (1989) were used as distributional references.

Results and Discussion

We collected 99 species of Hemiptera, representing 15 families, from 9 Arkansas counties (Table 2, Fig. 1). Of these, 54 are recorded for the first time from Arkansas (Table 2).

The majority of the species we are reporting as new for Arkansas are common, widespread species that, based on distributional data, would be expected for Arkansas. Of the 54 species new for Arkansas, 22 (40%), including all 9 of the Lygaeidae, have been reported for at least 4 of the 6 states bordering Arkansas (Table 2). Only 5 of the 54 species (9%) have not been reported for any bordering state. Interestingly, all five species are Midridae. This indicates that the Arkansas hemipteran fauna has been truly under-reported.

***Alydidae* (*Broad-Headed Bugs*).**–Prior to our addition of *Megalotomus quinquespinosus* as a new state record, only a single broad-headed bug species *Alydus eurinus* had been reported from Arkansas (Henry and Froeschner, 1988). *Alydus eurinus* is a common and widespread species. *Megalotomus quinquespinosus* was previously known only from Missouri of those states bordering Arkansas. Both species were collected in 3 separate regions of Arkansas (Tables 1 and 2), and we suspect that both species likely occur statewide. A few additional, less common, broad-headed bug species may also occur in Arkansas.

***Anthocoridae* (*Minute Pirate Bugs*).**–Three anthocorid species are now known for Arkansas. Prior to our 2 new state records (Table 2), only a single anthocorid species *Macrotracheliella nigra* (Parshley, 1917) was known for the state. Both species we encountered, represented by single specimens, were hand collected/aspirated while searching for Aradidae. Arkansas is within the known range, which nearly spans the southern United States, for both species (Henry and Froeschner, 1988). Some anthocorids are attracted to lights, which makes their collection easier. Several additional anthocorid species may be found by searching around outdoor lights.

***Aradidae* (*Flat Bugs*).**–All 4 aradid species we encountered were previously reported for Arkansas by Taylor and McPherson (1989). All specimens were hand collected by pulling bark off of fallen timber. These insects are cryptic and are one of the few insects we targeted during general collecting. For a full treatment of the 9 species known for Arkansas, see Taylor and McPherson (1989).

***Berytidae* (*Stilt Bugs*).**–Three species of stilt bugs are known from Arkansas. The 2 species of stilt bugs we encountered are common species in the United States: *Jalysus whickhami* occurs coast to coast, and *Jalysus spinosus* is commonly found east of about the 100th meridian. *Jalysus whickhami* is reported as a potential economically important pest species in North America (Wheeler and Henry, 1981). We often encountered both species in samples. These two species likely occur abundantly in Arkansas. In addition to these two *Jalysus* species, *Neides muticus* (Say, 1832) is known from Arkansas, and 1 other species *Metacanthus multispinus* (Ashmead, 1887) occurs in bordering states and may occur in Arkansas.

***Coreidae* (*Leaf-Footed Bugs*).**–Leaf-footed bugs are larger bodied bugs (specimens of 10–15 m are common) that are almost exclusively plant feeders. On appropriate host plants, they can occur in large numbers. Our collections were of either a single or a few specimens that we sporadically encountered. Four of the 6 species we collected (66%) are new state records for Arkansas. One of our new state records, *Leptoglossus fulvicornis*, is an eastern species that appears to be migrating west (McPherson et al., 1990). The other 3 new state records are widespread species and were expected for Arkansas. There are about 10 species of leaf-footed bugs now known for Arkansas with about twice that many known from bordering states.

***Cydnidae* (*Burrowing Bugs*).**–Lee and Barton (1983) provided an excellent treatment of this family for Arkansas. Our collection methods were not appropriate for attracting or collecting cydnids; our 3 specimens, therefore, were unexpected in our traps. We did not find any additional taxa for Arkansas.

***Lygaeidae* (*Chinch Bugs*).**–With about 3,000 species worldwide, this large, diverse family is second only to the Miridae in number of species. Over 320 species occur in North America (Henry and Froeschner, 1988). Nine of the 14 species we collected (64%) are new state records for Arkansas. All 9 species are common and widespread species that were anticipated for the state, as all are known from four or more states bordering Arkansas.

***Miridae* (*Plant Bugs*).**–We found the mirids to be difficult to identify and only utilized intact specimens that could be confidently identified; often only male specimens were able to be confidently determined. Miridae is the largest, most specious family of Hemiptera. In the genus *Phytocoris* alone, there are more than 200 species known for North America. All 6 of the Phytocoris species that we collected are new state records (Table 2). One mirid species is an Arkansas endemic (*Lopidea arkansae* Knight, 1965). We did not find this species, but the single species that we did find in this genus is a new state record (Table 2).

There are relatively few literature records of Arkansas Miridae. As a consequence, 79% of the mirid species we collected are new state records (Table 2). Although we encountered common and fairly widespread species that would be expected for Arkansas, there was at least one notable find. Our specimens of *Pycnoderes convexicollis* represent a significant range extension for this species. It was previously known only from Illinois and Indiana (Blatchley, 1926; Henry and Froeschner, 1988). An additional species of interest is *Fulvius slateri* which, although listed as occurring from California to Florida (Henry and Froeschner, 1988), had not previously been reported for Arkansas or any bordering state. Two additional species *Phytocoris puella* and *Phytocoris quercicola* lack records from states bordering Arkansas.

***Nabidae* (*Damsel Bugs*).**–Damsel bugs belong to a small predatory family of bugs that usually have a consistent morphology. Males are most reliable to identify. We report 3 nabid species as new for Arkansas (Table 2). All 3 have been reported for 2 or more states bordering Arkansas. There are now 6 damsel bug species reported for the state. In addition to the 4 species we found (Table 2), *Hoplistoscelis sericans* (Reuter, 1872) and *Nabis capsiformis* (Germar, 1838) are also recorded from Arkansas.

***Pentatomidae* (*Stink Bugs*).**–The Stink bug family is a large family; its members are some of the most commonly encountered and collected bugs. Barton and Lee (1981) provided an excellent treatment of this family for Arkansas. We did not find any additional taxa in our collections.

***Phymatidae* (*Ambush Bugs*).**–Ambush bugs are aptly named because they hide, often in flowers, waiting for unsuspecting prey to approach. They are voracious predators and many can capture prey larger than themselves. There have been 4 ambush bug species reported for Arkansas. Based on distribution, there are several more that may occur in the state (see Henry and Froeschner, 1988). The species that we found is the most common and widespread species, which likely

occurs statewide.

***Reduviidae* (*Assassin Bugs*).**–The members of this family are robust, predatory insects that will feed on almost anything that they can capture. Assassin bugs will inflict a very painful bite to humans, primarily as a defense mechanism. The senior author personally experienced the bite of a *Rasahus hamatus* specimen at a black light in the White River National Wildlife Refuge. The individual escaped, but left a severely painful reminder of its presence. A few (such as the *Triatoma*) feed on blood and will bite mammals. Many of these blood feeding species, however, have a painless bite.

Of the seventeen species we encountered, twelve (70%) are recorded for Arkansas for the first time. The ranges of all twelve species overlap Arkansas, and thus, were expected for the state. Ten of these twelve had been reported for at least 3 bordering states. Only 1 species *Rocconota annulicornis* was known from a single bordering state, Texas. Among these 12 is the "wheel bug" (*Arilus cristatus*). This easily recognizable and large species was known from Missouri, Oklahoma and Texas. Given its notoriety and distinctive characters for identification, the authors were surprised that this species had not been previously reported. Additionally, on a taxonomic note, we follow McPherson et al. (1991) in recognizing *Melanolestes abdominalis* (Herrich-Schaeffer, 1846) as a junior synonym of *Melanolestes picipes.*

***Rhopalidae* (*Scentless Plant Bugs*).**–Six rhopalid species have been reported for Arkansas (Henry and Froeschner, 1988). The 3 species that we encountered are common and widespread, and all had previously been reported for Arkansas. One rhopalid species *Boisea trivittata* (Say, 1825) (the box elder bug) is not listed for Arkansas by Henry and Froeschner (1988), and we could not find a record of it in the literature through 2004. This species has been reported for every state bordering Arkansas (except Louisiana) and undoubtedly occurs in the state.

***Thyreocoridae* (*Negro Bugs*).**–Lee and Barton (1983) provided an excellent treatment of this family (under the junior family name of Corimelaenidae) for Arkansas. We did not find any additional taxa in our collections.

***Tingidae* (*Lace Bugs*).**–These small (most species are under 5 millimeters) phytophagous insects are replete with anastomotic veins throughout their expanded membranous covering, which gives them their common name. Four of the 5 species that we collected are new for Arkansas (Table 2). These 4 species are common, as all have been reported for at least 25 states in the United States (Henry and Froeschner, 1988). Further, all 4 species were previously reported for Missouri and Texas, as well as at least 1 other bordering state and were expected for Arkansas. Several more widely distributed lace bug species likely occur in Arkansas.

Approximately 170 species of terrestrial Hemiptera have previously been reported from Arkansas. Almost half of those come from the 3 works of Barton and Lee (1981–50 species), Lee and Barton (1983–24 species) and Taylor and McPherson (1989–9 species). The addition of our 54 new records brings the known terrestrial hemipteran fauna of Arkansas to over 220 species. Based on distributional data (Henry and Froeschner, 1988), 220 species may be only a fraction (less than half) of the Arkansas terrestrial hemipteran fauna.

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Table 1. Collection sites (arranged by alphabetical order of county).

Site #	County	Location	Collection Method	Co-ordinates	Date
1	Arkansas	White River at State Route 1 bridge (White River National Wildlife Refuge)	Pitfall	N34.38 : W-91.12	22 June 2001
2	Arkansas	White River at State Route 1 bridge (White River National Wildlife Refuge)	Sweepnet	N34.38 : W-91.12	22 June 2001
3	Arkansas	White River & Arkansas Post Canal confluence (White River Refuge)	Sweepnet	N34.02 : W-91.18	21 June 2001
4	Arkansas	Lowland forest area of Panther Creek (White River National Wildlife Refuge)	Black-light sheet & pan	N34.29 : W-91.11	21 June 2001

Table 1. Continued.

Site #	County	Location	Collection Method	Co-ordinates	Date
5	Arkansas	Jack's Bay Landing, off Jack's Bay Road (White River National Wildlife Refuge)	Sweepnet	N34.10 : W-91.16	21 June 2001
6	Cleburne	Greers Ferry Lake, John F. Kennedy park	Sweepnet	N35.50 : W-91.97	2,5 September 1999 & 2000
7	Desha	Alligator Lake, (White River National Wildlife Refuge)	Black-light sheet	N34.05 : W-91.09	22 June 2001
8	Desha	Alligator Lake, (White River National Wildlife Refuge)	Sweepnet	N34.05 : W-91.09	22 June 2001
9	Izard	Roadside vegetation, (State Route 9 & CR 3 junction)	Sweepnet/hand coll.	N36.03 : W-91.91	25 May 2004
10	Lawrence	Open field, wetland area, off CR 316: (Shirey Bay-Rainey Brake Wildlife Area)	Black-light pan/hand coll.	N35.98 : W-91.11	22 May 2004
11	Lawrence	Open field, wetland areas, off CR 316: (Shirey Bay-Rainey Brake Wildlife Area)	Sweepnet/ beating sheet	N35.98 : W-91.11	23 May 2004
12	Montgomery	Boxx Spring, Forest service road 73 (Ouachita National Forest)	Black-light sheet	N34.44 : W-93.78	25 June 2001
13	Montgomery	Caddo ponds and Gardens, (Ouachita National Forest)	Black-light sheet	N34.38 : W-93.50	26 June 2001
14	Montgomery	Rattlesnake Creek Spring, (Ouachita National Forest)	Sweepnet	N34.43 : W-93.57	25 June 2001
15	Phillips	Hudson's Landing of the White River (White River National Wildlife Refuge)	Sweepnet	N34.22 : W-91.06	22 June 2001
16	Pike	Brier Creek/Little Missouri River (Ouachita National Forest)	Sweepnet	N34.38 : W-93.86	25 June 2001
17	Polk	McKinley Mountain (Ouachita National Forest)	Aspirated/Pitfalls/ hand collected	N34.43 : W-94.01	26 June 1998

Table 2. Species list of Hemiptera collected from Arkansas.

Family: Species	Collection Sites (from Table 1)
Alydidae: Broad-Headed Bugs (2 species; 1 new state record)	
Alydus eurinus (Say, 1825)	5, 7, 14
**Megalotomus quinquespinosus* (Say, 1825)	8, 9, 14
Anthocoridae: Minute Pirate Bugs (2 species; 2 new state records)	
**Lytocoris stalii* (Reuter, 1871)	10
**Xylocoris sordidus* (Reuter, 1871)	10
Aradidae: Flat bugs (4 species)	
Aradus robustus (Uhler, 1871)	10
Mezira granulata (Say, 1832)	10
Mezira sayi (Kormilev, 1982)	10
Neuroctenus simplex (Uhler, 1876)	9
Berytidae: Stilt Bugs (2 species)	
Jalysus spinosus (Say, 1824)	6, 8, 11, 16
Jalysus wickhami (Van Duzee, 1906)	6, 7, 11, 16
Coreidae: Leaf-Footed Bugs (6 species; 4 new state records)	
***Acanthocephala terminalis* (Dallas, 1852)	3, 11, 12
**Chariesterus antennator* (Fabricius, 1803)	11
**Euthochtha galeator* (Fabricius, 1803)	16
Leptoglossus corculus (Say, 1832)	16
**Leptoglossus fulvicornis* (Westwood, 1842)	16
Leptoglossus opposites (Say, 1832)	11
Cydnidae: Burrowing Bugs (2 species)	
Amnestus pusillus (Uhler, 1876)	10
Pangaeus bilineatus (Say, 1825)	17
Lygaeidae: Chinch Bugs (14 species; 9 new state records)	
Antillocoris pilosulus (Stal, 1874)	4, 12
Blissus leucopterus (Say, 1832)	16
***Cymus angustatus* (Stal, 1874)	5, 11
Geocoris punctipes (Say, 1832)	6, 16
Geocoris uliginosus (Say, 1832)	6
***Heraeus plebejus* (Stal, 1874)	13
***Kleidocerys resedae geminatus* (Say, 1832)	11
Myodocha serripes (Olivier, 1811)	6, 12, 13
***Neopamara albocincta* (Barber, 1953)	4, 7, 12
***Neopamara bilobata* (Say, 1832)	6
***Neortholomus scolopax* (Say, 1832)	16
***Oedancala dorsalis* (Say, 1832)	5, 8, 11, 13, 16
***Phlegyas abbreviatus* (Uhler, 1876)	5, 13, 16
***Pseudopachybrachius basalis* (Dallas, 1852)	6
Miridae: Plant Bugs (24 species; 19 new state records)	
***Agnocoris rossi* (Moore, 1955)	10
**Alepidia gracilis* (Uhler, 1895)	12
**Ceratocapsus quadrispiculus* (Knight, 1927)	12
**Ceratocapsus modestus* (Uhler, 1887)	11
Deraeocoris nebulosus (Uhler, 1872)	4, 5, 10
**Deraeocoris histrio* (Reuter, 1876)	10
**Fulvius slateri* (Wheeler, 1977)	1
**Lopidea confluenta* (Say, 1832)	16
Lygus lineolaris (Palisot, 1818)	6, 8, 10, 11, 13
Neurocolpus jessiae (Knight, 1934)	14, 16
Neurocolpus nubilus (Say, 1832)	2
**Phytocoris angustifrons* (Knight, 1926)	12
**Phytocoris canadensis* (Van Duzee, 1920)	10
**Phytocoris eximius* (Reuter, 1876)	10
**Phytocoris mundus* (Reuter, 1909)	12, 13
**Phytocoris puella* (Reuter, 1876)	12
**Phytocoris quercicola* (Knight, 1920)	10
**Plagiognathus obscurus* (Uhler, 1872)	9
**Plagiognathus politus* (Uhler, 1895)	8, 11
**Prepops fraternus fraternus* (Knight, 1923)	10
***Prepops rubrovittatus* (Stal, 1862)	3, 11
***Pseudoxenetus regalis* (Uhler, 1890)	10
**Pycnoderes convexicollis* (Blatchley, 1926)	13
Reuteroscopus ornatus (Reuter, 1876)	10, 16
Nabidae: Damsel Bugs (4 species; 3 new state records)	
***Hoplistoscelis sordidus* (Reuter, 1872)	2, 8
**Lasiomerus annulatus* (Reuter, 1872)	2
Nabis alternatus (Parshley, 1922)	16
***Nabis americoferus* (Carayon, 1961)	6
Pentatomidae: Stink Bugs (9 species)	
Acrosternum hilare (Say, 1832)	10, 13
Banasa dimiata (Say, 1832)	4
Brochymena cariosa (Stal, 1872)	16
Euschistus servus servus (Say, 1832)	2, 3, 5, 9, 16
Euschistus tristigmus tristigmus (Say, 1832)	8, 11, 13, 14
Mormidea lugans (Fabricius, 1775)	5, 16
Oebalus pugnax (Fabricius, 1775)	7
Podisus maculiventris (Say, 1832)	17
Thyanta accerra (McAtee, 1919)	10
Phymatidae: Ambush Bugs (1 species)	
Phymata americana americana (Melin, 1930)	6, 11

Table 2. Continued.

Family: Species	Collection Sites (from Table 1)
Reduviidae: Assassin Bugs (17 species; 12 new state records)	
**Arilus cristatus* (Linnaeus, 1763)	6
***Barce fraterna fraterna* (Say, 1832)	4, 7
**Emesaya brevipennis brevipennis* (Say, 1828)	15
Melanolestes picipes (Herrich-Schaeffer, 1846)	17
**Microtomus purcis* (Drury, 1782)	17
**Oncocephalus geniculatus* (Stal, 1872)	7
**Pnirontis modesta* (Banks, 1910)	10
***Pselliopus barberi* (Davis, 1912)	2, 6, 9
***Pygolampis pectoralis* (Say, 1832)	10
**Rasahus hamatus* (Fabricius, 1781)	4, 12
**Rocconota annulicornis* (Stal, 1872)	4
Sinea diadema (Fabricius, 1776)	6
Sinea spinipes (Herrich-schaeffer, 1846)	5
Triatoma sanguisuga (Leconte, 1856)	17
***Zelus cervicalis* (Stal, 1872)	6
Zelus luridus (Stal, 1862)	3, 11
Rhopalidae: Scentless Plant Bugs (3 species)	
Arhyssus lateralis (Say, 1825)	3, 6
Arhyssus nigristernum (Signoret, 1859)	6, 13
Harmostes reflexulus (Say, 1832)	6
Thyreocoridae: Negro Bugs (4 species)	
Corimelaena lateralis (Fabricius, 1803)	8, 9, 11
Corimelaena marginella (Dallas, 1851)	5, 6
Corimelaena pulicaria (Germar, 1839)	5, 16
Galgupha loboprostethia (Sailer, 1940)	5, 6, 8
Tingidae: Lace Bugs (5 species; 4 new state records)	
Corythucha aesculi (Osborn & Drake, 1916)	5
**Corythucha arcuata* (Say, 1832)	5
***Corythucha ciliata* (Say, 1832)	15
***Corythucha marmorata* (Uhler, 1878)	5, 13, 15, 16
**Leptoypha mutica* (Say, 1832)	4

*new state record for Arkansas.

**new state record + species previously recorded from 4 (or more) of the states bordering Arkansas (Louisiana, Mississippi, Missouri, Oklahoma, Tennessee, Texas)

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Effects of Herbicide Application on Foliar Morphology and Nutrient Concentrations in Mid-Rotation Pine Plantations

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Abstract

Application of herbicide to reduce competing brush and hardwood species is a common silvicultural activity in young loblolly pine (*Pinus taeda*) stands. A reduction in competition generally increases the amount of available resources to the loblolly pine crop trees thereby increasing foliage biomass, fascicle dimensions, and foliar nutrient concentrations. To what extent herbicide application and competition control alters these foliar characteristics in mid-rotation stands has rarely been reported. The purpose of this paper is to evaluate whether the application of herbicide alters the morphology, mass, and/or nitrogen concentration of mid-rotation loblolly pine foliage. We aerially applied an imazapyr herbicide to 6 study plots within each of four mid-rotation stands in Louisiana and Arkansas. Another 6 plots in each stand were untreated and served as a control Average fascicle length, fascicle mass, and foliage nitrogen concentrations in the herbicided-treated plots did not significantly differ from that in the control plots. However, foliage concentrations and fascicle size one year after herbicide application were greatest in plots with the greatest competing vegetation mortality.

Introduction

Silvicultural activities that increase the amount of leaf area and foliage biomass enhance photosynthetic activity and thus result in increased stem growth and timber production of crop trees (Will et al., 2002). Silvicultural practices such as thinning and fertilization alter foliage morphology as well as leaf area and mass (Velazques-Martinez et al., 1992; Yang, 1998) by increasing available resources such as light, water, and/or nutrients. Herbicides are extensively used to control brush and hardwood competition in loblolly pine (*Pinus taeda*) stands (Scultz, 1997). Competition control can increase site resources available for crop trees which would otherwise be utilized by competing vegetation. In young pine plantations, competition control increases soil water thereby reducing the water stress of crop trees (Perry et al., 1994), potentially increases nitrogen availability (Nusser and Wentworth, 1987), and increases the amount of light available to recently established crop trees (Morris et al., 1993). As a result, loblolly pine foliage morphology, nutrient concentrations, mass, and area can be altered (Zutter et al., 1999) by controlling competition in early stages of stand development. To what degree competition control in older, mid-rotation loblolly pine stands modifies these foliar attributes has not been documented. As part of a study investigating the effect of competition control and fertilization on productivity of mid-rotation pine stands, we monitored the response of pine foliage (physical attributes and nutrient concentrations) to the application of herbicide and the resulting reductions in hardwood and brush competition.

Materials and Methods

The study was established in 4 loblolly pine stands within the Gulf Coastal Plain of Arkansas and Louisiana, one to one and half years following an initial thinning operation. Two stands (Crossroads and Marion) are located in Union Parrish, Louisiana. The other stands (South Crossett and West Crossett) are located in Ashley County, Arkansas. Soils in all stands were either Alfisols or Ultisols. Table 1 contains soil and stand characteristics for each study site.

Loblolly pine is the most dominant species in the stands with loblolly and shortleaf pine (present only in one stand) accounting for approximately 91% of the total basal prior to treatment application. Sweetgum, red maple, blackgum, and water oak represented 69% of the basal area of the hardwood and brush stems that had diameters at breast heights (dbh) greater than or equal to 2.54 cm. These hardwood species accounted for 62% of the hardwood and brush stems prior to treatment application. The Louisiana stands had lower pine and higher hardwood densities than the stands in Arkansas. The total basal area ranged from a low of 15.2 m^2 ha^{-1} at Marion to 17.2 m^2 ha^{-1} at West Crossett study site (Table 1). The proportion of hardwood and brush to pine basal area ranged from approximately 6.5% to 20.1%. Thus the stands comprised a wide range of stand densities as well as diversity in stand composition.

Table 1. Soil and stand characteristics for each site at the time of study initiation

Site Yr.	Age m^2ha^{-1}	Pine BA[1] BAm^2ha^{-1}	Non-pine Index[2]	Site Height m	Mean Pine Family	Dominant Soil Family
Crossroads	17	16.0	3.2	20.1	13.1	Plinthic Paleudults and Typic Paleudults
Marion	17	12.9	2.3	9.2	13.6	Aquic Paleudults
South Crossett	22	16.5	0.7	18.3	13.1	Aquic Fraglossudalfs and Oxyaquic Fraglossudalfs
West Crossett	17	26.8	1.7	18.9	12.1	Oxyaquic Fraglossudalfs

[1]Trees >2.54 cm dbh
[2]Estimated with base age 25

A total of 12 plots between 0.036 and 0.097 ha in size was established in each stand during the fall of 2001 or 2002. The stands serve as blocks in the experimental design. In September or October of the year of plot establishment, imazapyr herbicide was operationally applied to 6 of the 12 plots. The application rate was 1.162 l of herbicide and 0.234 l of surfactant per hectare. The remaining 6 plots in a stand were not herbicided and were retained as a control.

Twenty five first-flush, current-year-fascicles were collected annually from 5 dominant or codominant loblolly pine trees in each plot during January. The initial collection occurred 15–16 months after herbicide application. The same 5 trees from each plot were sampled each year. We used a shotgun to collect a primary lateral branch from the upper one third portion of the live crown. After branch collection, 25 fascicles were removed from the lateral branch section using latex gloves. Only whole, healthy fascicles typical of the crown were collected. We then removed dirt and contaminants from the fascicles if necessary. The fascicles from each tree on a plot were composited to make a total of 125 fascicles from each plot. The fascicles were then stored at 4°C until foliage could be dried. Within one week of collection the foliage was dried at 65-70°C for 24–28 hr. All 125 fascicles from a plot were massed to the nearest 0.01 gram after drying. In addition we measured the lengths of 10 randomly selected fascicles from each plot composite.

All fascicles collected on a plot for a given sampling period were then ground to pass through a 0.5 mm screen for chemical analysis. Nitrogen concentration was determined by combustion using an Elementar CN analyzer. We used a generalized randomized block design ANOVA to analyze fascicle length, fascicle mass, and nitrogen concentration as a random effect with sites as blocks. We analyzed data obtained for two consecutive years following herbicide application. We also used Pearson correlations coefficients to investigate the relationship of fascicle length, fascicle mass and foliage nitrogen concentration with competitor mortality.

Results

Mortality.–At the end of the first growing season following herbicide application hardwood and brush mortality in the herbicide-treated plots was 49.9, 56.7, 23.8, and 32.7 % of the initial hardwood and brush basal area for the Crossroads, Marion, South Crossett, and West Crossett stands, respectively. The greatest mortality occurred in the Louisiana sites, which had the highest hardwood basal area and stem density. Prior to imazapyr application, hardwood and brush comprised a greater proportion of the total basal area at the Louisiana sites (15-17%) than the Arkansas sites (4-6%).

Fascicle Length.–Mean fascicle length in the first year following the herbicide application was higher in herbicide plots than the control plots at 3 of the 4 stands (Table 2). In the second year following herbicide application, mean fascicle length was higher in the herbicide plots than the control plots in only 2 stands. The ANOVA tests did not indicate that differences in fascicle length were significant at $P = 0.05$ for either year (first year $P = 0.10$; second year $P = 0.45$). The variation in fascicle length during the first year following herbicide application was consistently greater in the herbicide-treated plots than the control plots. The coefficient of variation for the control and herbicide-treated plots in the first year was 4.0% and 7.4%, respectively. Variances were not significantly different

Table 2. The mean and coefficient of variation (CV) for fascicle length, fascicle mass, and foliage nitrogen concentration for each site by herbicide treatment one year following herbicide application.

Site	Treatment	Fascicle length		Fascicle mass		Foliage N (%)	
		Mean(cm)	CV(%)	Mean(mg)	CV(%)	Mean	CV
Crossroad	Control	17.4	3.4	151	6.4	1.4	5.6
	Herbicide	18.2	6.7	177	6.7	1.4	7.2
Marion	Control	17.5	3.4	141	8.1	1.3	5.3
	Herbicide	17.4	7.7	144	11.8	1.4	3.3
South Crossett	Control	17.7	4.7	158	9.3	1.2	6.0
	Herbicide	18.2	6.6	159	8.2	1.2	3.2
West Crossett	Control	16.7	2.6	146	10.6	1.2	2.5
	Herbicide	17.1	8.8	135	10.3	1.2	6.5

between the treatments for foliage collected the second year following herbicide application.

Fascicle Mass.–Like fascicle length, fascicle mass in the first year following herbicide application was higher in the herbicide-treated plots than the control plots at three of the four stands (Table 2). The ANOVA indicated that there was no significant difference in the mean fascicle mass between the herbicide and control treatments in either year following herbicide application (first year $P = 0.57$ and second year $P = 0.92$). Similar to fascicle length, there was a consistent difference in the variance of fascicle mass between the herbicide-treated and control plots. The coefficients of variation of fascicle mass in the control and herbicide-treated plots were 9.2% and 13.5%, respectively.

Nitrogen Concentration.–Foliar nitrogen concentrations (Table 2) was lowest in the control plots at South Crossett (mean = 1.20 %) and highest in the herbicide plots at Crossroads site (mean = 1.41 %). Differences in foliage nitrogen concentration between treatments was neither significant the first year ($P = 0.28$) or the second year ($P = 0.32$) following herbicide application.

Correlation Analysis.–Mass, length, and nitrogen concentrations of fascicles in the herbicide-treated plots generally increased with increased hardwood mortality (Figs. 1a and 1b). Hardwood and brush mortality were positively and significantly correlated with fascicle length ($r = 0.28$, $P = 0.054$), fascicle mass ($r = 0.42$, $P = 0.003$), and foliage nitrogen concentration ($r = 0.48$, $P = 0.001$) the first year following herbicide application. Hardwood and brush mortality was not significantly correlated with any of these fascicle characteristics the second year following herbicide application.

Discussion

There was generally no consistent impact of the herbicide treatment on the foliage among the 4 sites. ANOVA tests indicated that differences in fascicle length, fascicle mass, and N concentrations between treatments were not significant for either the first or second year after herbicide application. The lack of response may be related to position of the pine and hardwood/brush competitors within the canopy. Loblolly pine occurs in the mid and upper portion of the canopy while the hardwood/brush occurs primarily in the lower portions of the canopy. Since the foliage samples were collected from the upper third of the loblolly pine crowns , removal of hardwoods and brush which occur in the lower portion of the canopy would have little impact on the light regimes where the foliage samples were collected. In young pine plantations, brush and hardwood competition occurs within the upper portion of the canopy and any reductions in competition increases the amount of light throughout the entire of canopy. This change in light increases fascicular length, fascicle mass as well as photosynthesis throughout the length of a pine's

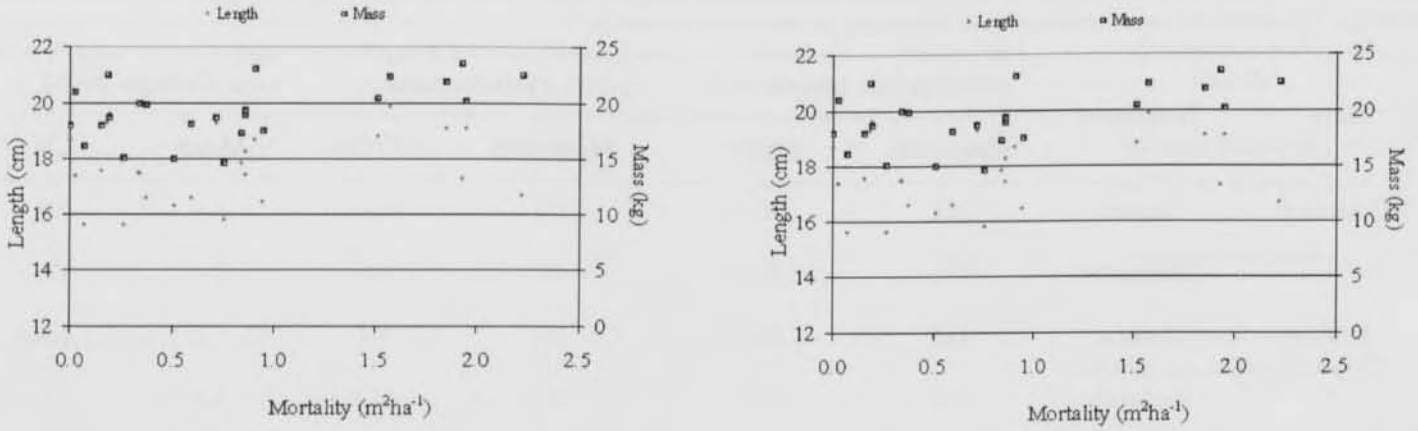

Fig. 1. Hardwood and brush mortality with length and mass (a) and nitrogen concentrations (b) of loblolly pine foliage one year after herbicide application.

crown. Foliar responses in older stands following thinning reflect the fact that tree removal results in crown removal throughout the entire canopy. Thus, removal of these trees increases light intensity to a greater degree than from the removal of midstory and understory competition in mid-rotation stands.

There was also no consistent response of foliar N concentrations to herbicide application. It seems likely that the hardwood and brush competitors did not represent a major sink for N at these sites and thus significantly reduce the amount of N available to the loblolly pine crop trees. In addition, N contained in trees killed by the herbicide needs to mineralize before the N would become available for uptake by the pine trees. This may take several years and would only result in an increase in foliar N concentration after a considerable period of time following herbicide application.

One consistent impact of the herbicide was an increase in the variation of both the fascicle length and mass during the first year following application. The coefficient of variation of these parameters was higher for the herbicide-treated plots than the control plots at each individual site. This may reflect a direct antagonistic impact of the herbicide on the foliage. The fascicles collected the first year following herbicide treatment were set in the bud at the time of the herbicide application. Potentially, variation in densities, sample tree locations, and other plot characteristics may have contributed to the variation in herbicide contact with the buds and thus the impact of herbicide on the emergent foliage.

The significant, positive correlations of first year foliar measurements with hardwood and brush mortality suggest that foliage response increases with the level of competition release. Intuitively we expect that the greater the reduction in competition, the greater the amount of resources available for the remaining crop trees and thus the alteration of foliar characteristics. The lack of significant differences in foliar characteristics between the herbicide-treated and control plots indicated by the ANOVA may reflect the wide range in hardwood densities and mortality among sites. The lack of any significant correlations of mortality and second year characteristics suggests that any changes in resources were either short-lived or that these resources were used by the crop trees to produce more fascicles rather than larger fascicles with higher N concentrations.

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Bedrock Geology of Rogers Quadrangle, Benton County, Arkansas

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Abstract

A digital geologic map of Rogers quadrangle was produced at 1:24,000 scale using the geographic information system (GIS) software MapInfo. Data regarding stratigraphic relations observed in the field were digitized onto the United States Geological Survey (USGS) digital raster graphic (DRG) of Rogers quadrangle. The geology of Rogers quadrangle consists of sedimentary rocks of the Ordovician, Devonian, and Mississippian systems. The Cotter, Powell, and Everton formations represent the Ordovician System. The Clifty and Chattanooga formations represent the Devonian System. The St. Joe and Boone formations represent the Mississippian System. This mapping effort represents the first time stratigraphy of Rogers quadrangle was mapped utilizing digital technologies. The prominent geologic structures in Rogers quadrangle are east–west and north–south trending normal faults, commonly inferred from stratigraphic relations across small drainages inundated by Beaver Lake; the most extensive faulting was located in the Blackburn Creek arm and the Prairie Creek sub-basin of Beaver Lake. Complex faulting in the Prairie Creek area appears to have a long geologic history; here the Devonian Chattanooga Shale lies directly on top of the Ordovician Cotter formation, suggesting that the Ordovician Powell and Everton formations and much of the Devonian Clifty formation were either never deposited or have eroded from this area. In either case, the Prairie Creek area appears to represent a structural high developed during the Middle to Late Ordovician that was eventually inundated by rising sea level to permit deposition of the Chattanooga Shale. Detailed mapping of Rogers and other northwest Arkansas quadrangles is providing new insights into the geologic evolution of the southern continental craton and Ozark Plateaus during the Paleozoic Era.

Introduction

Rogers quadrangle (Fig. 1) is located in Benton County, Arkansas, and is named for the city of Rogers located on the western boundary of the quadrangle. The quadrangle boundaries are 36°15.0'N 94°07.5'W (southwest), 36°22.5'N 94°07.5'W (northwest), 36°22.5'N 94°00.0'W (northeast), and 36°15.0'N 94°00.0'W (southeast).

Benton County is located on the south flank of the Ozark Dome (Croneis, 1930). The county occupies portions of two erosional plateaus formed along the southern portion of the Ozark Dome. The Springfield Plateau is defined by the top of the Boone formation, a sequence of Lower Mississippian limestone and chert, whereas the higher Boston Mountains Plateau south of Benton County is formed by Upper Mississippian and Lower to Middle Pennsylvanian strata capped by the Middle Pennsylvanian Atoka formation. Brown (2000) and Sullivan and Boss (2002) illustrated the lithostratigraphic succession observed in Rogers quadrangle (Fig. 2).

The landscape of Rogers quadrangle is a maturely dissected, dendritic drainage system dominated by the White River, which flows north through the eastern third of the quadrangle (Figs. 3, 4) and is impounded by Beaver Dam to form Beaver Lake. Whereas upland areas

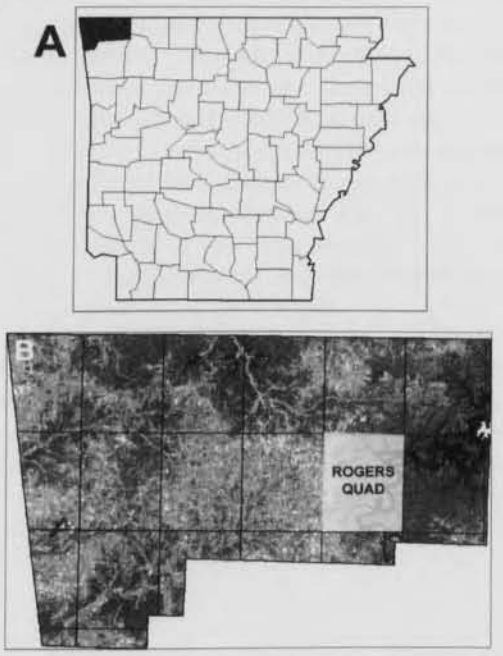

Fig. 1 A) Location map of Arkansas showing Benton County (shaded) and B) Rogers quadrangle in Benton County.

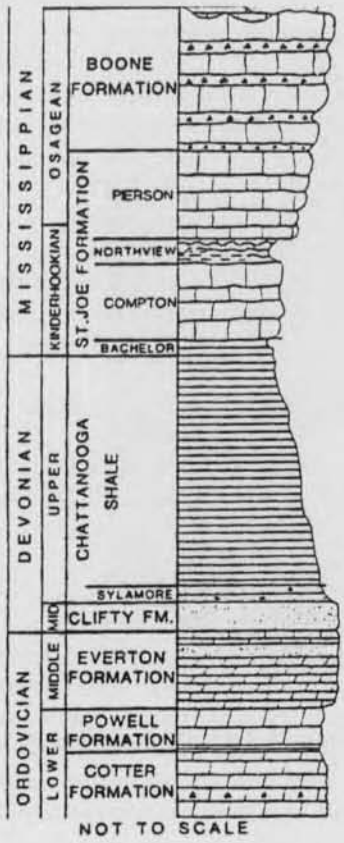

Fig. 2. Generalized stratigraphic column of Rogers quadrangle, Benton County, Arkansas (adapted from Brown, 2000; Sullivan, 1999.)

throughout the quadrangle are heavily forested, excellent exposures of all lithostratigraphic units can be observed along the shores of Beaver Lake, roadcuts along highways US 12 and 94, and along numerous farm-to-market and unimproved roads in the area.

The topography of the quadrangle is controlled principally by the St. Joe and Boone formations (Figs. 3, 4). The Boone formation is found at the tops of most hills in the quadrangle and tends to be a slope forming unit. The underlying limestone of the St. Joe formation tends to form bluffs generally 6 to 9 m height. The Chattanooga Shale is a slope forming formation below the St. Joe formation. Units of Ordovician and Devonian ages are principally exposed along the shores of Beaver Lake in the former bluffs of the White River Valley.

The geologic history and depositional dynamics of Paleozoic rocks in northwest Arkansas continue to attract the attention of the geologic community as a means of investigating the interplay of tectonics and eustasy in the development of continental margin and foreland basin sequences (Houseknecht, 1986; Viele, 1989; Ethington et al., 1989; Thomas, 1989; Viele and Thomas, 1989; Handford and Manger, 1990, 1993; Hudson, 2000). However, relatively little modern research on the geological history of northwest Arkansas exists in the academic literature. Most published reports are from early in the first half of the 20th century and represent early reconnaissance mapping of northwest Arkansas. During the 1950s and early 1960s, a number of quadrangles were mapped at 1:24,000 scale by graduate students at the University of Arkansas –Fayetteville, but most of these works were never published. As such, the present effort to re-map quadrangles in northwest Arkansas in the context of modern geologic theory and advanced digital technologies (Geographic Information Systems, GIS) is providing new insights into the geologic history of this area and the geologic evolution of the southern continental margin and craton during the Paleozoic Era.

Branner (1891) of the Arkansas Geological Survey described the topography, hydrogeology, and stratigraphy of Benton County, Arkansas. The 1891 report also included a geologic map at the scale of 1:126,720 (1 inch to 2 miles). The map showed the carboniferous Boone formation to be widespread at the surface in the vicinity of Rogers, Arkansas, with sandstones, magnesian limestones, and cherts of "Silurian" age (now known to be part of the Ordovician and Devonian Systems) in the White River valley to the east.

Croneis (1930) of the Arkansas Geological Commission described the stratigraphy and structure of the Springfield Plateau. Croneis identified two prominent geologic structures, the Price Mountain (Fayetteville) Fault and the Glade Fault, both of which trend southwest to northeast and are located to the southeast and northeast respectively of Rogers quadrangle. These are normal faults downthrown to the southeast of their respective fault traces Normal faults, the majority of which are downthrown on the southeast side of the fault trace, are typical in the Springfield Plateau region (Croneis, 1930). The report by Croneis (1930) includes a geologic map at a scale of 1:38,160 (1 inch to 6 miles).

Gibbons (1962) studied the fracture patterns in northwest and west central Arkansas looking for a correlation with the timing of the Ouachita Orogenic episode. Gibbons concluded that there are five distinct shear fracture sets of post-Mississippian through Permian age, two sets of folds as a result of compressional forces, and multiple linear zones of tension fractures that are parallel to other structures in the region that compose the regional pattern of northwest Arkansas. Based on the work of Gibbons, Quinn (1963) concluded that the folding and faulting in northwest Arkansas was due to compressional forces from the northwest and southeast directions. In later petrographic

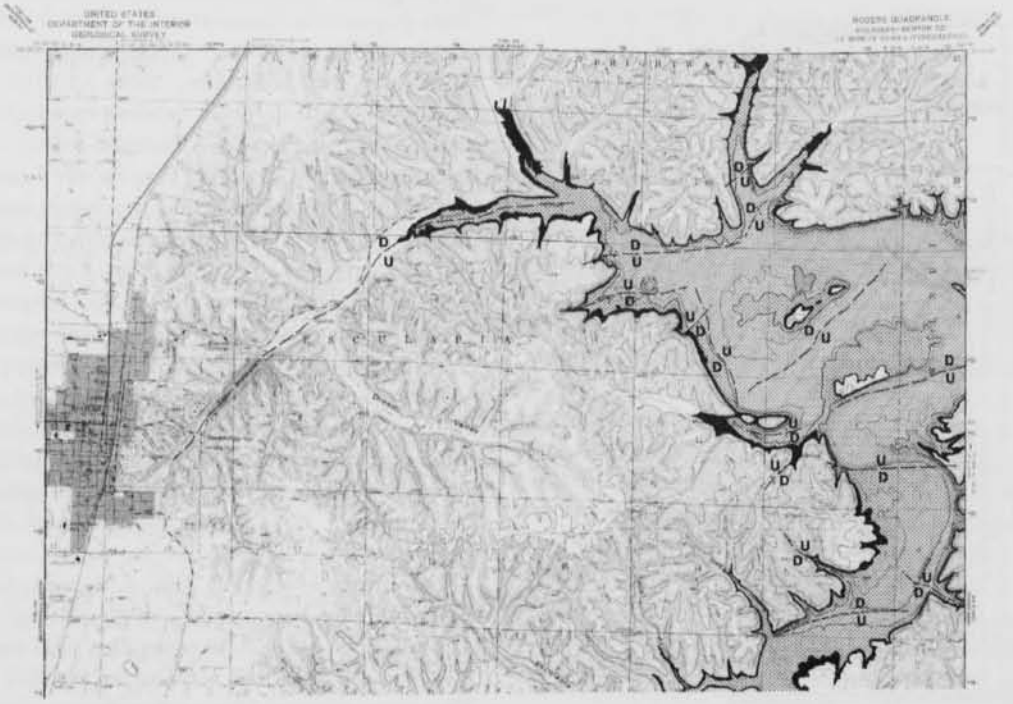

Fig. 3. Map showing bedrock quadrangle geology of northern half of Rogers quadrangle digitized onto Rogers quadrangle 7.5-minute digital raster graphic (DRG).

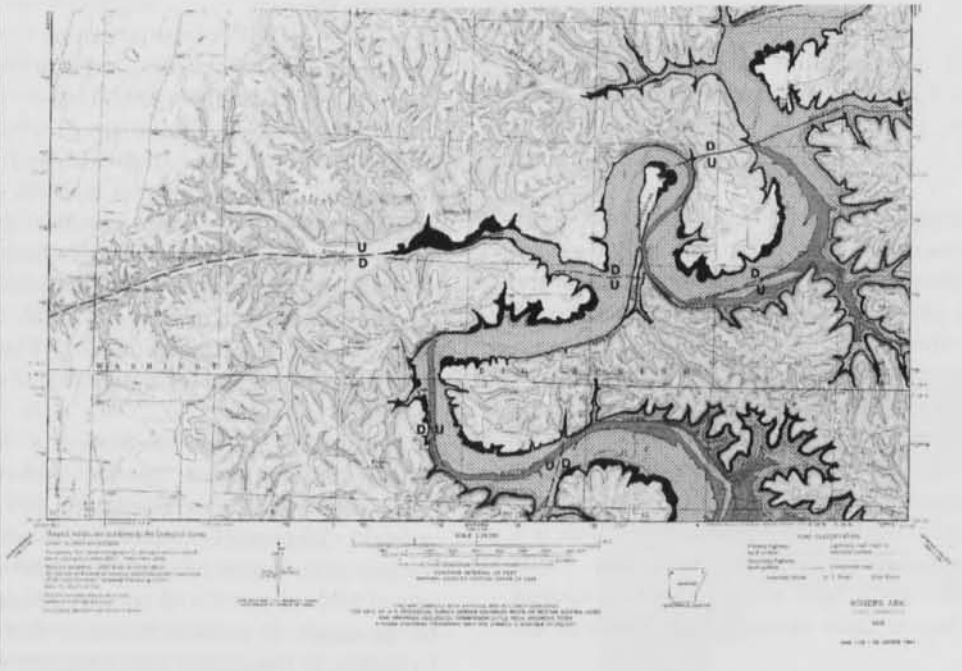

Fig. 4. Map showing bedrock quadrangle geology of southern half of Rogers quadrangle digitized onto Rogers quadrangle 7.5-minute digital raster graphic (DRG).

studies of calcite twin lamellae, Chinn and Konig (1973) showed evidence supporting deformation of northwest Arkansas from north–south compression, but timing of the compression was inconclusive.

Arrington's (1962) thesis and corresponding geologic map were the only 1:24,000-scale geologic map of the area. At the beginning of this study, only a single copy of Arrington's (1962) 1:24,000 map of Rogers quadrangle was preserved in the special collections of the University of Arkansas library. This map was of particular interest because it predated inundation of Beaver Lake. Thus, it provided the only modern description of strata in the main channel of the White River valley (Figs. 3, 4).

Materials and Methods

Field mapping of Rogers quadrangle was conducted in the summer of 2003 through the spring of 2004; various locations were accessed from a network of county and state roadways and by boat on Beaver Lake. Locations of outcrop sites for individual stratigraphic members and observed geologic structures were determined using global positioning system (GPS) receivers capable of receiving differential corrections (horizontal position accuracies of ca. 3 m). A Garmin Etrex hand-held GPS unit was used to determine elevation, latitude, and longitude for most outcrops. These elevations and coordinates were noted in the field notebook, and the location was indicated on the field map.

Information regarding field geologic relations was transferred from the field map to a digital raster graphic (DRG) of Rogers quadrangle using a "heads-up" digitizing method. Using this method, geologic contacts were drawn directly on the computer screen by moving the cursor over a digital raster graphic (DRG) of Rogers quadrangle and clicking the mouse button at short intervals to trace contacts onto the displayed topography (King et al., 2002; Sullivan and Boss, 2002; King et al, 2001a and b; Sullivan, 1999). Each stratigraphic unit was digitized as a separate layer within the geographic information system such that the display of each layer could be toggled on or off. Faults were digitized as lines onto a separate layer as well. Once all stratigraphic units and geologic structures were digitized, map layers representing those stratigraphic units and geologic structures could be displayed hierarchically to generate the geologic map of the study area (Figs. 3, 4). A legend for the map is presented also (Fig. 5). All data were archived on CD-ROM, and a large-format digital image of the final geologic map is available upon request from the corresponding author.

Results and Discussion

Strata in Rogers quadrangle range from Ordovician through Mississippian periods (Fig. 6A). Detailed lithostratigraphic descriptions of Paleozoic strata can be found in King et al. (2002), Sullivan and Boss (2002), King et al. (2001a and b), and McFarland (1998). The oldest strata exposed in Rogers quadrangle are those of the Ordovician Period (approximately 490-443 Ma BP; Palmer and Geissman, 1999); these strata are comprised of (in ascending order) the Cotter, Powell, and Everton formations (Hopkins, 1893; Adams and Ulrich, 1904; Purdue and Miser, 1916). The oldest Ordovician stratum is the Cotter formation (Fig. 6B). Arrington (1962) mapped the Cotter formation primarily in the main valley of the White River. However, extensive outcrops of Cotter formation were observed throughout the northwest quarter of Rogers quadrangle around the shoreline of the Prairie Creek sub-basin of Beaver Lake. While this and other Ordovician strata were not mapped at these elevations by Arrington (1962), it is interesting to note that Ordovician strata were mapped extensively in this area by Branner (1891). Though the stratigraphic nomenclature used by Branner (1891) was different, it is clear from the rock descriptions that this was the Cotter formation. The Powell formation is not well exposed at the surface throughout Rogers quadrangle (Fig. 6C). Southeast of the U.S. Highway 12 bridge over Beaver Lake, a thin exposure (<2 m) of the Powell formation was observed in unconformable contact with the underlying Cotter formation. The Powell is also exposed on the north shore of the Prairie Creek sub-basin of Beaver Lake on the east side of the mouth of Coose Hollow. Elsewhere around the Prairie Creek sub-basin, it appears that the Powell formation was either eroded or never deposited, suggesting the presence of a localized structural high, perhaps persisting from Late Ordovician through Early to Middle Devonian time. Likewise, the uppermost Ordovician stratum (the Kings River Sandstone Member of the Everton formation) was not observed to crop out throughout the entire northern half of the quadrangle, but was observed around the lake shore south of Blackburn Creek and particularly to the north of Hickory Creek.

Devonian (417–354 Ma BP; Palmer and Geissman, 1999) strata in Rogers quadrangle are the Clifty formation and the Chattanooga Shale. The Clifty formation was named by Purdue and Miser (1916) for friable quartz sandstone exposed on the east fork of Little Clifty Creek, Benton County, Arkansas. Conodonts collected from the Clifty formation exposures at Beaver Dam (Hall and Manger, 1978) are middle Devonian (391–370 Ma BP). In the northwest quarter of Rogers quadrangle, the Clifty formation occurs mainly as massive sandstone in discontinuous mounds or pods. Relatively continuous exposures of Clifty formation (up to 2-3 m thick; Fig. 6D) were observed along the shore of Beaver Lake in the extreme southern portion of the quadrangle. Here, it lies unconformably on the Everton formation (Kings

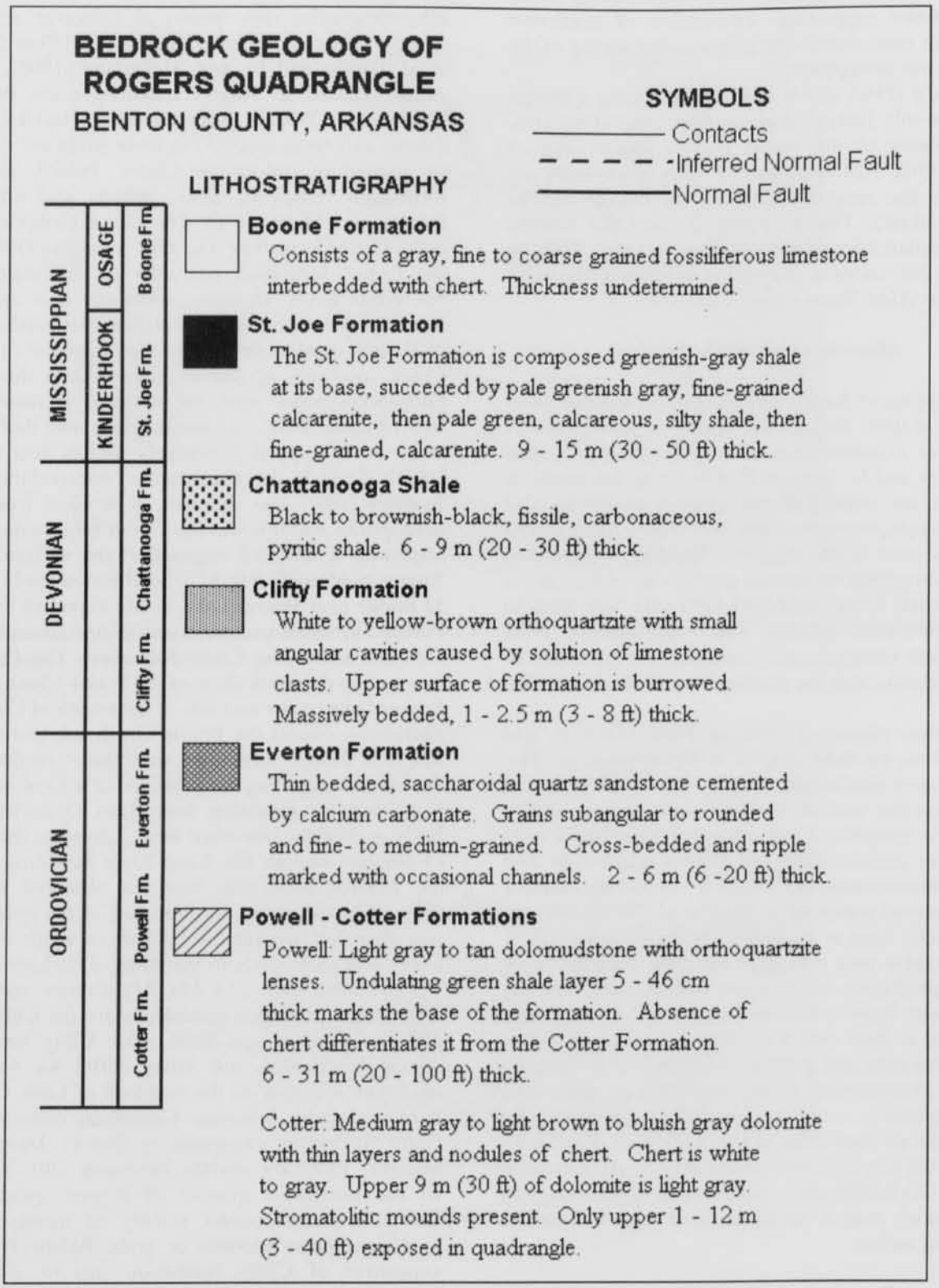

Fig. 5. Legend to accompany geologic map of Rogers quadrangle (Figs. 3, 4).

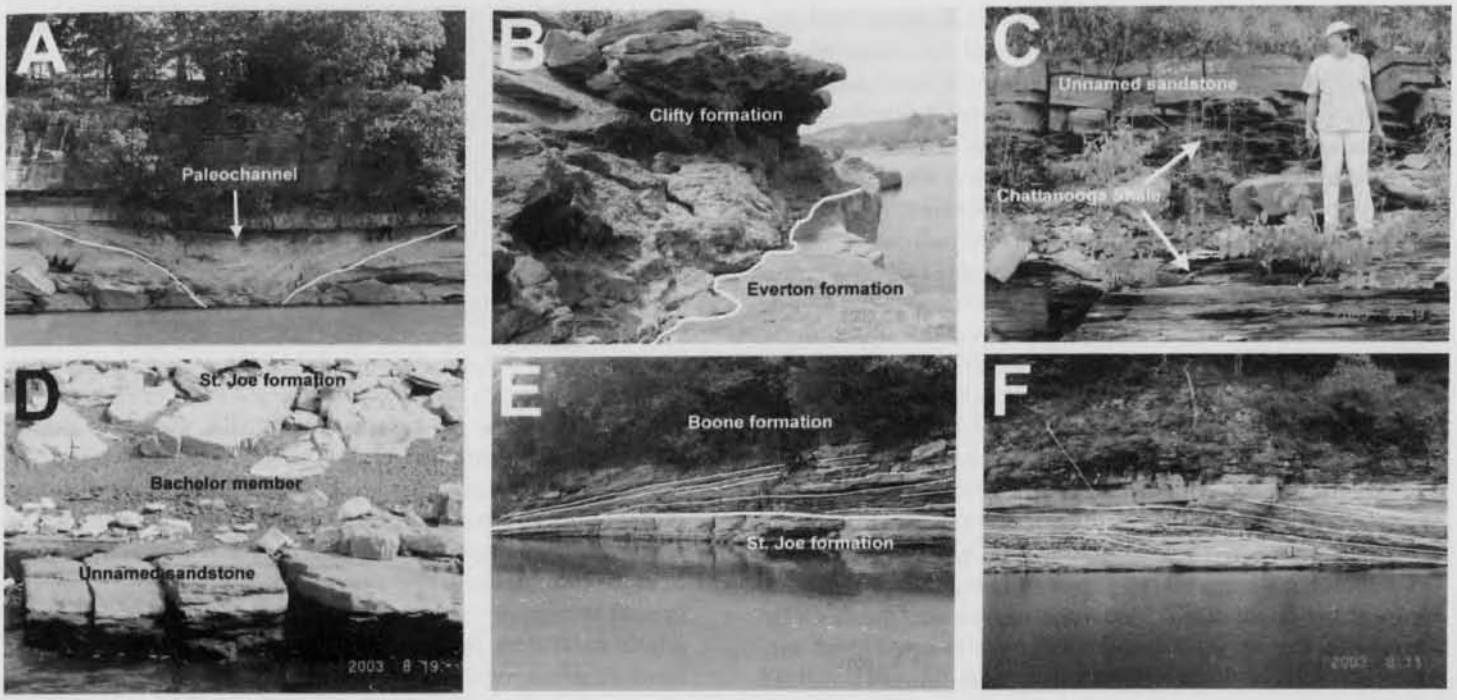

Fig. 6. Images of geologic formations exposed in Rogers quadrangle. A) Succession exposed in hillside along shore of Beaver Lake. Stratigraphic units labeled. B) Domal stromatolite preserved in Cotter formation dolomite. C) Small boulder of Clifty formation (Devonian) sandstone with reworked clasts of Ordovician Powell formation dolomite or limestone. D) Relatively thick (approximately 2–3 meters) outcrop of Clifty formation exposed along shore of Beaver Lake in southern portion of Rogers quadrangle. E) Unconformable contact of Chattanooga Shale (Devonian) on the Cotter formation (Ordovician). Large rock slabs are weathered Sylamore Sandstone, the basal unit of the Chattanooga Shale in Rogers quadrangle. Outcrop located southeast of U.S. Highway 12 bridge, northern portion of Rogers quadrangle. F) Isolated nodular chert embedded in St. Joe formation limestone.

River Sandstone Member). The Clifty formation appears to become thicker as one moves southward into Sonora quadrangle (Hutchinson et al., 2005).

The Chattanooga Shale was identified by Adams and Ulrich (1904). Throughout Rogers quadrangle, the Chattanooga Shale incorporates a thin (0.15–0.45 m), basal sandstone containing phosphatic pebbles called the Sylamore Sandstone (Fig. 6E; Penrose, 1891). Branner (1891) named the Sylamore Sandstone for exposures along Sylamore Creek, Stone County, Arkansas. The Sylamore Sandstone commonly displays a chert breccia at its base (Hall, 1978), indicating that it is unconformable on the underlying Devonian or Ordovician strata. The Sylamore Sandstone is correlative with the Misner Sandstone of Oklahoma and the Hardin Sandstone of Tennessee (Cooper et al., 1942). The Sylamore Sandstone appears to be conformable with the overlying Chattanooga Shale.

The Chattanooga Shale is a black to brownish-black, fissile, carbonaceous, pyritic shale that averages 6 to 9 m (20 to 30 feet) thick and ranges to 15 m (50 feet). The Chattanooga often occurs at or near lake level in Rogers quadrangle where it forms gentle slopes and broad valleys unless it is protected from weathering and erosion by overlying massive limestone of the St. Joe formation. The Chattanooga Shale correlates with the Chattanooga and Woodford Shales of Oklahoma (Frezon, 1962) and the type Chattanooga Shale of Tennessee (Cooper et al., 1942).

Mississippian (354–323 Ma BP; Palmer and Geissman, 1999) strata in Rogers quadrangle are the St. Joe formation and the Boone formation. These are the youngest rocks exposed in Rogers quadrangle. The stratigraphic status of the St. Joe Limestone has been the subject of debate for an extended time (Hopkins, 1893; Cline, 1934; Mehl, 1960; McFarland, 1975; Shanks, 1976; Manger and Shanks, 1977; Shelby, 1986). For this study and for mapping purposes, the St. Joe Limestone was considered a discrete formation. Additional evidence suggesting formation status for the St. Joe Limestone was observed in Sonora quadrangle (Hutchinson et al., this volume) where the St. Joe–Boone formation contact was clearly unconformable. The St. Joe formation is a cliff-forming unit, which helps to distinguish it from the overlying Boone formation where the

St. Joe formation occurs on slopes and wooded hillsides. The St. Joe formation is typically limestone, though locally it contains some nodular chert in southern Rogers quadrangle (Fig. 6F). In Rogers quadrangle, the basal layer of the St. Joe formation is the Bachelor Member, a greenish-gray shale approximately 0.5 m thick that is unconformable on the top of the Chattanooga Shale. The remaining Mississippian stratum is the Boone formation. Branner (1891) named the Boone formation for exposures in Boone County, Arkansas. The Boone formation is the most widespread rock exposed in Rogers quadrangle, occupying approximately 81% of the surface area of the quadrangle. The Boone formation is readily recognized by its abundant chert in a limestone matrix. Weathering and dissolution of the Boone formation results in development of a residuum composed of chert cobbles and red-to orange-colored clay.

Features of the structural geology of Rogers quadrangle were more complex than previously mapped (Branner, 1891; Arrington, 1962). A number of previously undocumented faults were mapped during this project, and faulting is particularly conspicuous along the axis of Blackburn Creek (Fig. 7) and within the Prairie Creek sub-basin of Beaver Lake (Figs 3, 4). Fault orientations are northeast-southwest and east-west and steeply dipping, creating a structurally complicated pattern of tilted fault blocks. Uncertainty exists as to whether these are normal or reverse faults. Normal faulting is generally assumed based on previous work in Rogers quadrangle and elsewhere across northwest Arkansas. Fault offsets are often small (1 to 10 m) and most visible on the cliffs surrounding Beaver Lake (Fig. 7). The timing of faulting cannot be determined precisely since faults offset all strata in the area. Though faulting was presumed to be related to the Ouachita Orogeny (Hudson, 2000), there is some stratigraphic evidence of active faulting and associated uplift predating the Devonian Period (e.g., the apparent structural high observed in the Prairie Creek area where the Chattanooga Shale rests unconformably on Ordovician Cotter formation).

Exposed rocks of Ordovician-Mississippian age in Rogers quadrangle provide insight into the geologic history and evolution of the Ozark Plateaus and southern craton margin during the Paleozoic Era. Revised, detailed, digital mapping of Rogers quadrangle provides several important, practical revisions to the previous geologic map of Rogers quadrangle by Arrington (1962). These include 1) mapping the St. Joe Limestone as a formation distinct from the Boone formation, 2) separation of the Bachelor Member shale from the Chattanooga Shale and assigning it as the basal member of the St. Joe formation, and 3) documentation of previously unknown faults throughout the quadrangle, particularly in the Prairie Creek area. These revisions provide new insights into the geologic evolution of the Ozark Plateaus and

Fig. 7. Exposure of prominent normal fault in Boone formation limestone and chert along lake shore west of the mouth of Blackburn Creek. Fault trends east-west through axis of Blackburn Creek and appears to extend into War Eagle quadrangle to the east (Sullivan and Boss, 2002). "U" is upthrown block, "D" is downthrown block.

southern craton margin during the Paleozoic Era in the context of modern geological thought and plate tectonic theory. In particular, newly identified exposures around the Prairie Creek sub-basin of the Cotter formation (Ordovician) overlain directly by the Chattanooga Shale demonstrate a pronounced unconformity (Ordovician-Devonian) and suggest this area was a localized structural high subjected to erosion or non-deposition prior to deposition of the Chattanooga Shale. Supplemental evidence that the area around Prairie Creek existed as a localized structural high can be found in the northward thinning of the Clifty formation (Middle Devonian) in Rogers quadrangle. In the southern portion of the quadrangle, the Clifty formation is 2–3 m thick, but in the Prairie Creek area, the Clifty formation occurs as isolated pods and lenses of sandstone that appear to be erosional remnants or deposits in localized depressions in the underlying Cotter formation dolomite. Northward thinning of the Clifty formation suggests it was deposited around the margins of a localized structural high with maximum relief in the northwest quarter of the quadrangle.

The outcrop belt of the Cotter formation in the Prairie Creek area is bound by several faults. It is possible that these faults were responsible for the observed uplift/erosion or non-deposition across this area before it was finally inundated to permit deposition of the Sylamore Sandstone and Chattanooga Shale. Thus, it appears possible that tectonic activity along the southern craton margin related to plate convergence far to the south may have initiated sometime between the Silurian and Middle Devonian periods and possibly ceased to permit deposition from Late Devonian through Mississippian periods. This is generally much older than what has previously been supposed, and

while it may represent a very localized tectonic episode, it does agree with a growing consensus that tectonism along the southern craton margin had a prolonged history throughout much of the Paleozoic Era (Hudson, 2000). In addition, stratigraphic and structural relations observed within Rogers quadrangle may reflect the interplay of global eustasy and tectonics during the Devonian Period.

In addition to the tentative evidence of relatively early tectonism, documentation of faults with different orientations suggests polyphase deformation episodes across the Ozark Plateaus. Previous workers (Quinn, 1963; Chinn and Konig, 1973; Hudson, 2000) have suggested multiple episodes of brittle deformation of the Ozark region related to progress of the Ouachita Orogeny to the south. There is increasing evidence from mapping across northwest Arkansas (Hudson, 2000) that deformation of the Ozark area was a very prolonged and polyphase process.

Continued detailed mapping in northwest Arkansas will ultimately provide the base from which greater understanding of the geologic evolution of the southern craton will emerge. Mapping around the margins of Beaver Lake (Hutchinson et al., 2005; Sullivan and Boss, 2002) shows particular promise in this regard, as the lake level provides a standard datum from which subtle faults can be recognized. Identifying and mapping these faults along with stratigraphic relations throughout a number of quadrangles will thus provide additional details of the intriguing geologic history of the Ozark Plateau.

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Integrating Supervised and Unsupervised Classification Methods to Develop a More Accurate Land Cover Classification

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Abstract

The classification and mapping of land cover provides fundamental information about the characteristics, activities, and status of specific areas on the earth's surface. The quality of the final classification is critical in providing accurate information for ecologists and resource managers in decision-making and for developing a landscape-level understanding of an ecosystem. A land cover classification was developed for 5 research watersheds in Garland and Saline counties in Arkansas using 2002 LANDSAT 7 Enhanced Thematic Mapper Plus (ETM+) satellite imagery. The supervised classification was based upon 146 training areas identified from reference data and then applied to the imagery using the maximum likelihood classification algorithm. The unsupervised classification used an Iterative Self-Organizing Data Analysis Techniques (ISODATA) algorithm to classify the imagery into 300 spectral classes which then were identified from reference data. Data from 171 field locations were used to assess the accuracy of the final classifications using an error matrix. The supervised classification had an overall accuracy of 74.85% compared to 40.94% for the unsupervised classification. However, the dense canopy pine plantation class, which comprises 10.69% of the total area of the watersheds (1,216.69 ha), was more accurately classified in the unsupervised classification (64.29%) than the supervised classification (43.86%). The unsupervised classification of dense canopy pine plantation was incorporated into the supervised classification to produce a final integrated classification with an improved overall accuracy of 76.61%. We found that, where greater accuracy is desired, both classification methods should be used and the results integrated to utilize each method's strengths.

Introduction

Land cover is a distinct concept applied to the classification of the earth's land surface (Estes et al., 1982). Estes et al. (1982) define land cover as æthe vegetational and artificial constructions covering the land surface". The classification of land cover is the assignment of geographic areas to certain classes based upon similar characteristics of land cover. There are numerous uses and purposes for the classification of land cover. Ustin et al. (1999) stated that land cover can provide insight into the underlying soils and geologic conditions of an area. Land use/land cover maps also have the potential for use in preserving prime agricultural farmland, in guiding land development decisions in metropolitan areas, or in developing large scale inventories of resources at the county, state, or federal level (Anderson, 1982). Land cover data, particularly when used in conjunction with other data such as terrain maps available from Digital Elevation Models (DEMs), can be useful in identifying areas more or less suited to specific land management practices and thereby aid in the assessment of appropriate practices for use in a specific area to attain certain goals (Bonner et al., 1982). Development of land cover maps can also be critical in monitoring the changes in land cover for a given area of study or management (Estes et al., 1982). Often an understanding of changes that have occur and the extent of such changes is critical for making appropriate land management decisions (Estes et al., 1982). Land cover classification of a region can help clarify the status of an ecosystem at a specific time. The accuracy of a land cover classification is therefore critical to its utility and value in providing accurate information for ecologists and resource managers.

Supervised and unsupervised are 2 primary methods of image classification, such as a land cover classification. Supervised classification involves the classification of pixels of unknown identity by means of a classification algorithm using the spectral characteristics of pixels of known informational class (referred to as training areas) identified by the analyst (Campbell, 2002). There are several advantages to using this approach to classification. First, the analyst has full control of the informational categories, or classes, to be assigned in the final classification. This allows for easier comparison with other classifications by using identical classes for both. Second, through the process of selecting training areas, the resulting classification is tied to

specific areas on the image of known identity. Third, the analyst does not face the problem of matching spectral classes to informational classes, because this is addressed during the selection of training areas. Finally, the training data can be compared with the final classification as one means of detecting serious errors or problems in the classification process (Campbell, 2002). There are also disadvantages and limitations to the use of supervised classification. First, the analyst is "imposing a classification structure upon the data" (Campbell, 2002) by the selection of training areas and of specific information classes, which may not necessarily be present in the data. Second, spectral properties are generally not the primary characteristics used in identifying training areas, which can lead to overlap and ambiguity during the classification process. Third, the selection of training areas requires of the analyst an extensive knowledge of the area and an investment of time and resources that is not required for unsupervised classification. Finally, unique classes present in the image may be overlooked by the analyst during the selection of classes and training areas.

Unsupervised classification involves the separation of image pixels into natural groupings based upon similar spectral characteristics by means of a classification algorithm and the resultant assignment of those groupings to informational classes by the analyst. There are three primary advantages to using this approach to classification. First, extensive knowledge of the area being classified is not required for the initial separation of image pixels. Second, there is less opportunity for human error as the analyst is not required to make as many decisions during the classification process. Third, unique classes in the data will be recognized by unsupervised classification, where as they may be overlooked in a supervised classification. There are also disadvantages and limitations to the use of unsupervised classification. First, the natural groupings identified by the classification process are spectrally homogeneous, which may not necessarily correspond with the informational classes of interest. Second, the analyst has limited control over the classes chosen by the classification process, and the relationships between the natural groupings of spectral classes and that of the desired informational classes are not always directly correlated.

When evaluating an image classification, there are two forms of accuracy that can be considered. The first is non-site-specific accuracy, which looks at the overall agreement between the classified image and the reference data without examination of the agreement between them at specific locations. For example, non-site-specific accuracy involves the examination of the percent Mature Pine Forest in the classified image and the comparison of it to the percent Mature Pine Forest in the reference data. Relying solely on non-site-specific accuracy to evaluate a classification can hide errors resulting from disagreement in the placement of classes between the classified image and the reference data.

The second form of accuracy is site-specific accuracy, which examines the agreement between classes at specific locations on the classified image and in the reference data. This examination is done by means of an error matrix (also known as a confusion matrix or contingency table) to compare, for specific locations, what an area is in the reference data versus how that area has been classified. The error matrix helps to identify instances of classification error for specific classes. There are 2 main types of these classification errors: errors of omission and errors of commission. Errors of omission are instances in which site has been excluded from a class to which it actually belongs. Errors of commission are instances in which a site is included in an incorrect class. Campbell (2002) noted that these errors tend to balance each other, as an error of omission for one class will also be tabulated in the error matrix as an error of commission in another class. Given the characteristics of these errors, it is best to examine them on a class-by-class basis before assuming the errors in one class reflect the errors found in all classes.

For site-specific accuracy assessment using the error matrix, there are three primary measures of classification accuracy: overall classification accuracy, producer's accuracy, and user's accuracy. Overall classification accuracy is the measure of how much area was correctly classified out of the entire area classified. From the error matrix, overall classification accuracy is the sum of the diagonals divided by the total. Producer's accuracy is calculated for each class and gives an indication of how well a particular class has been classified by the producer of that classification. This accuracy is most often used by the producer as a means to assess how well the classification was performed. From the error matrix, the producer's accuracy for each class is the result of dividing the correctly classified pixels by the number of reference data pixels in that class (as determined by the column total). User's accuracy is also calculated for each class and gives an indication of how often the areas assigned to a given class on the image classification actually belong to that class "on the ground". This accuracy is of greater importance to the users of the classification because this indicates how true the classified image is to the actual situation on the ground. From the error matrix, the user's accuracy for each class is the result of dividing correctly classified pixels in a given class by the total number of pixels in that class on the classified image (as determined by the row total).

This paper describes the development of a land cover classification using 2 separate methods (supervised and unsupervised) that were then compared and integrated to improve the overall accuracy of the final classification as determined by means of an accuracy assessment. The land cover classification was derived from LANDSAT 7 Enhanced Thematic Mapper Plus (ETM+) imagery for five

watersheds in the Ouachita Mountains in Garland and Saline counties north of Hot Springs, Arkansas.

Materials and Methods

Study Area.–A land cover classification was developed for five research watersheds included in the Ouachita Mountains Ecosystem Management Research Project (OMEMRP) shown in Fig. 1. These watersheds are located in the Ouachita Mountains in Garland and Saline counties north of Hot Springs, Arkansas. The watersheds are as follows: Little Glazypeau – 2,275 ha predominantly under Weyerhaeuser Company ownership; North Alum Creek – 3,961 ha with approximately equal mixtures of Weyerhaeuser Company and USDA Forest Service ownership; Bread Creek – 1,535 ha predominantly under USDA Forest Service ownership; South Alum Creek – 1,499 ha predominantly under USDA Forest Service ownership; and Validation Watershed – 2,110 ha with mixture of USDA Forest Service and Weyerhaeuser Company ownership.

Data Preparation.–The base images used in the classification were LANDSAT 7 Enhanced Thematic Mapper Plus (ETM+) satellite images taken January 15th, March 4th, and September 12, 2002. The raw LANDSAT 7 ETM+ satellite images were preprocessed prior to inclusion in classification. These images were orthorectified by the Spatial Analysis Laboratory (SAL) with ERDAS Imagine® software using National Elevation Dataset (NED) Digital Elevation Models (DEMs) for vertical ground control and Digital Orthophoto Quadrangles (DOQ) data for horizontal ground control. Orthorectification is the process of tying image coordinates to ground coordinates by means of ground control for the purpose of creating a planimetrically and geometrically correct image. This process removes or minimizes errors produced by scale variation, sensor attitude/orientation, and internal sensor errors and provides the image with a real coordinate system that can be tied to the ground. Once the satellite images were orthorectified, 3 bands, the 2 thermal bands and the panchromatic band, were removed from each image and not included in the classification. The images were then subset to a bounding rectangle where the outer edges of the watersheds were at least 1.6 km (1 mile) from the bounding rectangle. The remaining bands from all 3 images (January, March, and September) were then merged for use in classification.

Reference Data.–Three primary sources were utilized for reference during the classification process: a prior land cover classification of the area, color infrared (CIR) digital orthophoto quadrangle (DOQ) images, and field-collected data. The prior land cover classification was created from 1995 LANDSAT 5 Thematic Mapper (TM) satellite images for OMEMRP that included 4 of the research watersheds and was reported and used by Tappe et al. (2004). The color infrared (CIR) digital orthophoto quadrangle (DOQ) images used as reference during the classification process were acquired between April 2000 and March 2001, with most of the images acquired in late January and February 2001. The DOQ images had a pixel resolution of 1m. These images were obtained from the Natural State Digital Database (http://sal.uamont.edu) which is maintained by the Spatial Analysis Laboratory, University of Arkansas at Monticello.

The field-collected data were obtained during several trips between late January and early March in early 2004 to the study area with two objectives in mind. The first objective was to become more familiar with the area and to collect land cover data from selected locations throughout the watersheds to assist in performing the classification. This first objective was accomplished during the first trip of January 28-30 during which land cover data were recorded for 64 locations throughout the study area. Spatial locations were determined by a Trimble Global Positioning System (GPS) receiver and visual estimates and measurements were made for land cover, forest composition, canopy cover, tree height, forest status (natural vs. plantation), and age. These data were then incorporated into the classification process to assist in identifying spectral classes generated during unsupervised classification and in developing training areas for the supervised classification in order to improve the accuracy of the classification.

The second and final objective was to collect land cover data to be used in developing an accuracy assessment for the classification. This final objective was completed when data collected for use in the accuracy assessment were recorded for 171 additional locations during two trips in early 2004. Spatial location was determined using a Trimble GPS receiver for spatial location, a photograph was taken of the plots in each of the 4 cardinal directions, and measured and visual estimates were taken for land cover, forest composition, canopy cover, tree height, forest status (natural vs. plantation), and age.

Supervised Classification.–The combined satellite images were classified by means of supervised classification with ERDAS Imagine® software. Information from the field data, CIR DOQs, and a prior 1995 land cover classification were utilized to identify 146 training areas representing the land cover classes described in Table 1. The Signature Editor in ERDAS Imagine® is an important tool for creating a supervised classification from training areas. Once each training area is identified on the image, the spectral characteristics across all bands and all dates for each pixel in the training area are then input into the Signature Editor where the signature for that training area can be labeled, evaluated, edited, and then incorporated into the supervised

classification. The Signature Editor is a means of managing all of the spectral signatures from the training areas for the image(s) being classified. Using the Signature Editor, the spectral signature across all image bands for each training area was obtained and then labeled by land cover class for use in the classification process. The supervised classification, using the maximum likelihood classification method, utilized all 146 individual signatures from the training data. The classification was then passed through both a 3 by 3 pixel majority filter and a 3 by 3 pixel class variety filter using ArcGIS software to allow for possible location inaccuracies during the classification's accuracy assessment.

Unsupervised Classification.–The combined satellite images were classified by means of unsupervised classification using an Iterative Self-Organizing Data Analysis Techniques (ISODATA) algorithm with ERDAS Imagine® software. ISODATA is a clustering algorithm that uses an iterative process to separate image pixels into spectrally similar clusters based upon their position in nth dimensional spectral space. The algorithm begins with an initial clustering of the data and the calculation of cluster means in nth dimensional space. Each iteration compares the spectral distance of each pixel to the cluster means and assigns them to the cluster whose mean is closest. Once all pixels are assigned, the cluster means are recalculated, and the pixels are again compared and clustered based on spectral distance to cluster means in nth dimensional space. This process is repeated until specified criteria, such as a convergence threshold, are met or the maximum number of iterations is reached. This process is highly successful at finding inherent clusters in the data and is not biased by initial clustering because of the iterative nature of this algorithm. The parameters for the unsupervised classification were set to 300 initial classes with maximum iterations of 350 and a convergence threshold of 0.990. Information from the field data, CIR DOQs, and a prior 1995 land cover classification were utilized to assign the resulting 300 spectral classes to the land cover classes described in Table 1. The classification was then passed through both a 3 by 3 pixel majority filter and a 3 by 3 pixel class variety filter using ArcGIS software to allow for possible location inaccuracies during the classification's accuracy assessment.

Integrated Approach.–As previously discussed, the supervised and unsupervised classification methods each have advantages and disadvantages. An integrated approach that incorporates both methods was explored. The resulting classifications from both methods were compared visually and by using the results of the accuracy assessment to assess the strengths and weaknesses of each with the goal of combining the results for a more accurate and useful final classification. The preliminary results found that the supervised classification was most accurate overall (see Table 3). One land cover class, dense canopy pine, was more correctly classified by the unsupervised method than the supervised method. Using the Spatial Analyst extension in ArcGIS®, the dense canopy pine pixels in the unsupervised classification were incorporated into the supervised classification by means of a CON statement, (If–then–else statement), which determined if a given pixel was a dense canopy pine pixel in the unsupervised classification. If it was, it would be assigned that value in the final classification, but if not, then the value for that pixel was based upon its value in the supervised classification. The integrated classification was also passed through both a 3 by 3 pixel majority filter and a 3 by 3 pixel class variety filter using ArcGIS® software to allow for possible location inaccuracies during the classification's accuracy assessment.

Results and Discussion

Based upon the final classification, there are four primary land cover classes found within the five watersheds in the Ouachita Mountains Ecosystem Management Research Project: Mixed Forest at 18.88% (2,148.19 hectares); Sparse Pine at 16.73% (1,903.98 hectares); Hardwood/Pine Forest at 11.60% (1,319.82 hectares); and Dense Canopy Pine Plantation at 10.69% (1,216.69 hectares) (see Table 2). There are four other land cover classes with at least 5.00% coverage within the five watersheds: Thinned Pine Plantation at 7.97% (907.25 hectares), Mature Pine Forest at 7.90% (898.42 hectares), Mature Hardwood Forest at 7.00% (796.84 hectares), and Sparse Hardwood Forest at 5.60% (636.98 hectares). The remaining six land cover classes with less than 5.00% coverage within the five watersheds are: Young Pine Plantation at 4.69% (533.65 hectares), Pine/Hardwood Forest at 3.46% (394.12 hectares), Clear-cut at 3.21% (365.68 hectares), Urban/Roads/Bare Ground at 1.82% (206.91 hectares), Field/Grass at 0.43% (48.68 hectares), and Water at 0.02% (2.23 hectares).

Accuracy Assessment.–The unsupervised classifi-cation had an overall accuracy of 40.94% (see Table 3), which was the lowest of the three classifications considered. Furthermore, only four classes in the unsupervised classification had either the producer's or user's accuracy greater than 60%: Urban/Roads–user's accuracy 100.00%; Clear-cut–producer's accuracy 71.43%; Dense Canopy Pine Plantation–producer's accuracy 64.29%; and Pine/Hardwood Forest–user's accuracy 66.67%.

The supervised classification had an overall accuracy of 74.85% (see Table 3). Unlike the unsupervised classification, only four classes in the supervised classification had either producer's or user's accuracy below 60.00%, with most over 75.00%: Field–producer's accuracy 25.00%; Dense Canopy

Pine Plantation – producer's accuracy 42.86%; Thinned Pine Plantation – user's accuracy 48.15%; and Mature Pine Forest – producer's accuracy 57.14%.

As classification of Dense Canopy Pine Plantation was more accurate using the unsupervised classification (producer's accuracy of 64.29%) than the supervised classification (42.86%), it was decided to incorporate the unsupervised classification of Dense Canopy Pine Plantation into the supervised classification to improve its accuracy in a combined classification. The Field class was left as is due to the low incidence of this class in the watersheds. Also the Water class was not included in the accuracy assessments for 2 reasons: first, water is spectrally distinct from all other classes and therefore easy to separate from them during classification; second, water constituted only 0.02% of the total area of all watersheds and was not available for ground truthing (precluding involvement in the accuracy assessment).

The integrated classification, which incorporated the Dense Canopy Pine Plantation from the unsupervised classification into the supervised classification, had an overall accuracy of 76.61% (see Table 3). The result was accuracies for all but two classes being over 60.00% with most being 75.00% or greater. The Field class continued to have a producer's accuracy of 25.00%, and the Mature Pine Forest class had a producer's accuracy of 52.38%. Given the overall performance of the integrated classification in the accuracy assessment, the integrated classification was selected as the final classification for use in the Ouachita Mountains Ecosystem Management Research Project.

Given the performance of both supervised and unsupervised methods for the current classification of these 5 watersheds in the Ouachita Mountains, the question arises as to why the unsupervised method produced poorer results overall when compared to the supervised method. One answer appears to be that many of the classes shared similar spectral properties across the 3 image dates, leading to potential confusion in the natural groupings that were based solely on spectral properties by the unsupervised classification algorithm. During the assignment of these groupings to land cover classes, it was a fairly common experience to find a single grouping having several possible land cover classifications as judged from the reference data. This experience suggests that, although it would increase the amount of time required to complete the classification, setting the parameter for the number of initial class groupings higher than the 300 used in this research might have reduced the number of confused classes during the assignment process of unsupervised classification.

Another answer may lie in the use of a predetermined set of land cover classes for this classification. As the analyst has little control over the groupings determined in unsupervised classification, assigning those groupings to preset classes can be more difficult and complicated than assigning them to a more open set of land cover classes. This is 1 of the inherent disadvantages of the unsupervised method of classification. It should be noted, however, that in other situations where the final set of land cover classes is more open to adjustment this disadvantage may not be an issue in the classification.

Likewise, the supervised method produced better results for the current classification than the unsupervised method for similar reasons. The inherent disadvantages of the unsupervised method are advantages of the supervised method, and vice versa. Thus, the use of training areas that are determined by the analyst based on the predetermined set of land cover classes allowed for greater control and accuracy using the supervised method of classification.

The question then arises as to why not just use the supervised classification since it was more accurate than the unsupervised classification for most land cover classes. Comparison of the accuracy assessment results between the integrated classification and the supervised classification offers some reasons for using the integrated classification. First, even though it was small, there was an increase in the overall accuracy of the integrated classification (76.61%) versus the supervised classification (74.85%). Second, two of three accuracy results that were below 50% (Grass/Field Producer's – 25.00%; Dense Canopy Pine Producer's – 42.86%; Thinned Pine User's – 48.15%) for the supervised classification were improved to over 70% in the integrated classification (Dense Canopy Pine Producer's – 78.57% and Thinned Pine User's – 70.59%). Third, although a few accuracies were higher in the supervised classification (Dense Canopy Pine User's 100.00%; Thinned Pine Producer's – 81.25%; Mature Pine Producer's – 57.14%; Mature Pine User's – 85.71%; and Pine/Hardwood Forest User's – 80.00%) versus the integrated classification (Dense Canopy Pine User's 61.11%; Thinned Pine Producer's – 75.00%; Mature Pine Producer's – 52.38%; Mature Pine User's – 84.62%; and Pine/Hardwood Forest User's – 66.67%), only one of these was below 60% in the integrated classification (Mature Pine Producer's – 52.38%), and it should also be noted as below 60% in the supervised classification (Mature Pine Producer's – 57.14%). Thus, overall the integrated classification was an improvement over the supervised classification.

The final question is when the integrated approach should be used to produce a land cover classification. In circumstances where there are only enough resources to use one classification method, considerations should be made as to whether a particular method is best suited for the task when applied. For example, for the classification developed in this study and, by extension, classifications of a similar nature, the supervised method resulted in a more accurate classification than the unsupervised method for reasons already discussed. If the situation were reversed, it is likely that a classification developed using the unsupervised

method could result in a more accurate classification. The main consideration then is whether the classification itself will maximize the effect of a particular method's advantages while minimizing the impact of its disadvantages. For circumstances where resources allow the use of both methods, the findings of the current study suggest that using both classification methods followed by integrating the results can produce an improved and more accurate classification, making use of the advantages found in both supervised and unsupervised classification methods.

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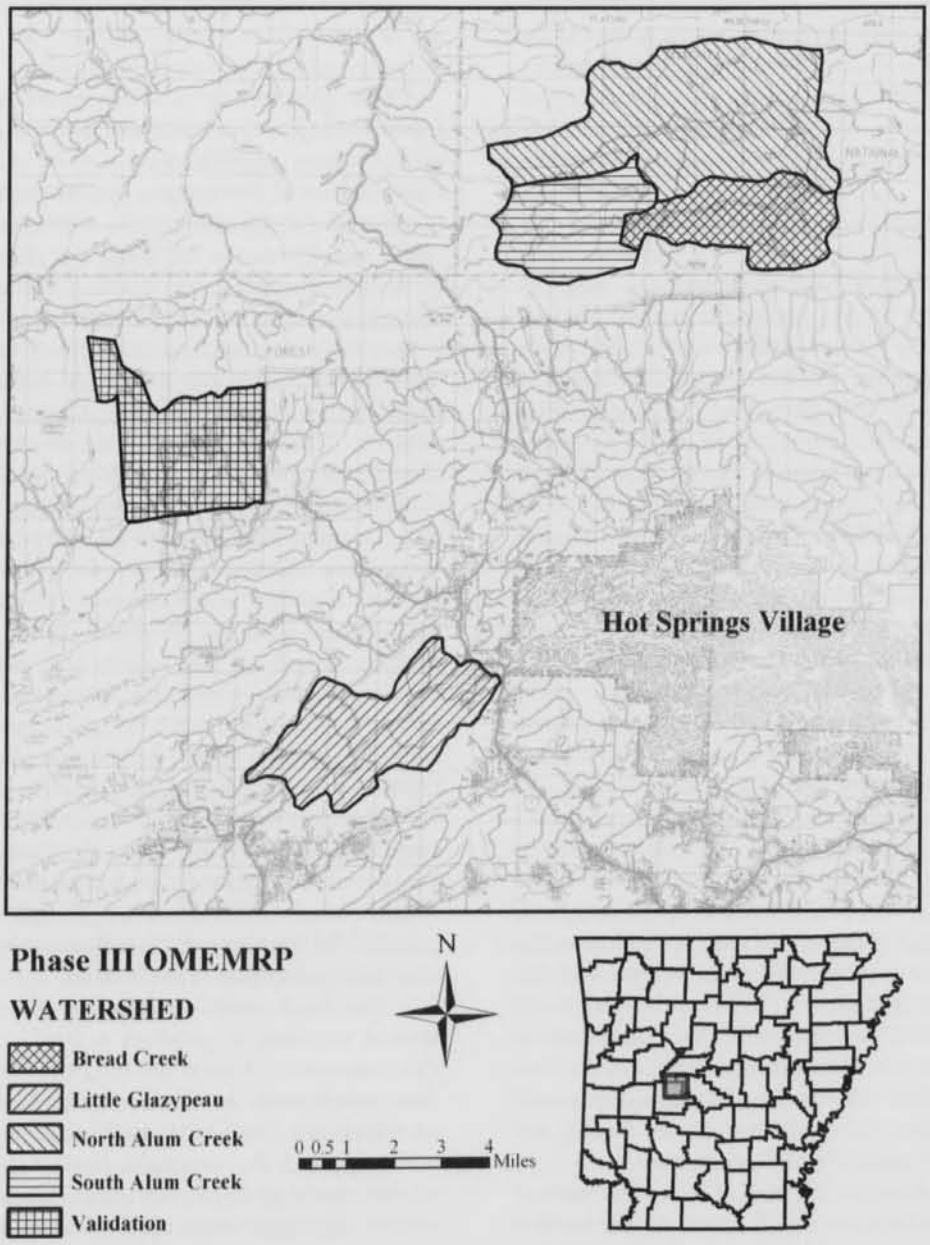

Fig. 1. The five watersheds involved in the land cover classification for the Ouachita Mountain Ecosystem Management Research Project (OMEMRP).

Table 1. Land cover classes and descriptions used in the classification of 5 watersheds in the Ouachita Mountains Ecosystem Management Research Project (OMEMRP).

Class Number	Land Cover Description (2002)
1	Water
2	Urban Area/Roads/Bare Ground/Rocks
3	Grass/Field
4	Clear-cut
5	Young Pine Plantation
6	Dense Canopy Pine Plantation
7	Thinned Pine Plantation
8	Mature, Pine Dominant (>75%) Forest
9	Sparse Pine
10	Mature Pine/Hardwood (60-75% Pine) Forest
11	Mature Mixed Forest
12	Mature Hardwood/Pine (60-75% Hardwood) Forest
13	Sparse Hardwood
14	Mature Hardwood Dominant (>75%) Forest

Table 2. Percent land cover and acreage for each land cover class for all 5 watersheds of the Ouachita Mountains Ecosystem Management Research Project (OMEMRP).

Class Number	Description	% Land Cover	Area	
			acres	hectares
1	Water	0.02%	5.52	2.23
2	Urban/Roads/Rocks/Ground	1.82%	510.88	206.91
3	Grass/Field	0.43%	120.21	48.68
4	Clear-cut	3.21%	902.91	365.68

Table 2. Continued.

Class Number	Description	% Land Cover	Acreage	
			acres	hectares
5	Young Pine Plantation	4.69%	1,317.66	533.65
6	Dense Canopy Pine Plantation	10.69%	3,004.17	1,216.69
7	Thinned Pine Plantation	7.97%	2,240.13	907.25
8	Mature Pine Forest	7.90%	2,218.34	898.42
9	Sparse Pine	16.73%	4,701.19	1,903.98
10	Pine/Hardwood Forest	3.46%	973.14	394.12
11	Mixed Forest	18.88%	5,304.18	2,148.19
12	Hardwood/Pine Forest	11.60%	3,258.82	1,319.82
13	Sparse Hardwood Forest	5.60%	1,572.79	636.98
14	Mature Hardwood Forest	7.00%	1,967.52	796.84
	Total	100.00%	28,097.46	11,379.45

Table 3. Comparison of accuracy assessment results for final integrated classification, supervised classification, and unsupervised classification of five watersheds in the Ouachita Mountains Ecosystem Management Research Project (OMEMRP). Class Number 1 (Water) is not included in the accuracy assessment results for 2 reasons: first, water is spectrally distinct from all other classes and therefore easy to separate from them during classification; second, water constituted only 0.02% of the total area of all watersheds and was not available for ground truthing (precluding involvement in the accuracy assessment).

Table 3. Continued.

Class Number	Description	Integrated Classification (Final) Producer's Accuracy	Integrated Classification (Final) User's Accuracy	Supervised Classification Producer's Accuracy	Supervised Classification User's Accuracy	Unsupervised Classification Producer's Accuracy	Unsupervised Classification User's Accuracy
1	Water						
2	Urban/Roads/Rocks/Ground	100.00%	71.43%	100.00%	71.43%	40.00%	100.00%
3	Grass/Field	25.00%	100.00%	25.00%	100.00%	0.00%	0.00%
4	Clear-cut	85.71%	85.71%	85.71%	85.71%	71.43%	55.56%
5	Young Pine Plantation	75.00%	85.71%	75.00%	75.00%	0.00%	0.00%
6	Dense Canopy Pine Plantation	78.57%	61.11%	42.86%	100.00%	64.29%	56.25%
7	Thinned Pine Plantation	75.00%	70.59%	81.25%	48.15%	50.00%	34.78%
8	Mature Pine Forest	52.38%	84.62%	57.14%	85.71%	52.38%	44.00%
9	Sparse Pine	76.19%	72.73%	76.19%	72.73%	52.38%	37.93%
10	Pine/Hardwood Forest	66.67%	66.67%	66.67%	80.00%	33.33%	66.67%
11	Mixed Forest	75.00%	62.50%	75.00%	60.00%	35.00%	33.33%
12	Hardwood/Pine Forest	90.91%	83.33%	90.91%	83.33%	31.82%	41.18%
13	Sparse Hardwood Forest	100.00%	92.31%	100.00%	92.31%	0.00%	0.00%
14	Mature Hardwood Forest	80.00%	100.00%	80.00%	100.00%	53.33%	42.11%
	Overall Accuracy	**76.61%**		**74.85%**		**40.94%**	

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Distributions of Bats in Bottomland Hardwood Forests of the Arkansas Delta Region

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Abstract

Bat distribution data is incomplete for the delta region of Arkansas. We extensively surveyed 16 counties within the Mississippi alluvial plain that comprises the delta from late spring to early fall 2004 using mist nets. We obtained 44 new county records for 9 species: *Myotis lucifigus*, *M. austroriparius*, *Pipistrellus subflavus*, *Eptesicus fuscus*, *Lasiurus seminolus*, *L. borealis*, *L. cinereus*, *Nycticeius humeralis*, and *Corynorhinus rafinesquii*. We generated updated distribution maps for these species and eastward Arkansas range expansions were documented for *L. seminolus*. Possible sampling concerns and research directions are discussed in relation to the needs of bats inhabiting bottomland forests of the delta, particularly *M. austroriparius* and *C. rafinesquii*.

Introduction

Published studies of bats within Arkansas have primarily focused on the Interior Highland region, which contains 3 endangered species (Sealander and Heidt, 1990), and where presence of caves and concentrations of foraging bats near limited water resources make sampling efficient. Much less research has occurred within the Mississippi Alluvial valley (MAV) of the Arkansas's Delta Region (although see Gardner and McDaniel, 1978). Historically, large tracts of bottomland hardwood forests dominated by white oak (*Quercus alba*), red oak (*Q. falcata*), sweet gum (Liquidambar styraciflua), sycamore (*Platanus occidentalis*), and bald cypress (*Taxodium distichum*) provided roosting opportunities for many bat species. However, agricultural practices composed primarily of monoculture farms of soybean, rice, winter wheat, and cotton have eliminated large forested areas, resulting in probable declines in bat populations associated with these habitats. Two species in particular, the southeastern myotis (*Myotis austroriparius*) and the Rafinesque big-eared bat (*Corynorhinus rafinesquii*), have gained recent attention due to their rare status and poorly known natural history (Horner, 1995, 1996; Mirowsky and Horner, 1997; Hoffman et al., 1999; Menzel and Menzel, 2001; Mirowsky et al., 2004). The distribution of these two species within Arkansas is largely incomplete with few records within the Delta region.

Additionally, other bat species, whose distributions have been well documented in other regions of the state, such as the highlands, have only anecdotally been reported within the Delta. Although Sealander and Heidt (1990) suggest the distribution of both *C. rafinesquii* and *M. austroriparius* encompass all bottomland forest regions of the state, few complete surveys of this region of Arkansas have been conducted for these bats (but see: Baker and Ward, 1967; Gardner and McDaniel, 1978). To better ascertain effects of local land-management practices on bat populations, complete data are required on distributions of sensitive species. Here we report on new county records of bat species captured during an extensive county by county survey of *C. rafinesquii* and *M. austroriparius* in the bottomland, hardwood forests of the MAV (hereafter, Delta) in eastern Arkansas. We also provide an updated distribution map for 9 bat species in the state of Arkansas, resulting from an extensive review of published accounts and this study.

Methods

This survey was conducted in 12 of the 16 counties (Arkansas, Clay, Craighead, Crittenden, Cross, Desha, Greene, Jackson, Lee, Mississippi, Monroe, and Poinsett) that encompass the Arkansas Delta region, three Central region counties (White, Prairie and Lonoke), and one Ozark region county (Lawrence). The Central and Ozark counties were sampled due to a lack of records for the two target species and the similarity of habitats to those in the Delta region. The regional divisions were based on Arkansas Game and Fish conventions. All sampling sites consisted of the following public lands: Arkansas Game and Fish Commision (AGFC) Wildlife Management Areas (WMAs), National Wildlife Refuges (NWRs), national forests, and state parks. Counties that already had pre-existing records for both species were not sampled. Bats were captured using mist nets placed in potential flight corridors (foot paths, all-terrain vehicle trails, unpaved roads), streams, ponds, and river edges. Nets were checked every 15 minutes and were left open for at least 5 hours beginning at dusk. Data obtained from captured bats include species, gender, age (juvenile or adult, as determined by the degree of ossification of the epiphyseal-diaphyseal fusion in the finger bones, Edythe, 1988), mass, and forearm length. Additionally, we determined the reproductive status of males (scrotal, non-reproductive) and females (pregnant,

lactating, non-reproductive), according to Racey (1988). All captured bats were fitted with a uniquely numbered plastic band and then released. As the study primarily focused on obtaining distribution data for *C. rafinesquii* and *M. austroriparius*, netting continued in each county until these two species were captured or a maximum number of 5 nights was reached. Distribution maps were generated from our own bat captures and from previously published county records for the state. The following sources were used for these records: Davis et al., (1955), Baker and Ward, (1967), Laval, (1970), Gardner, (1978), Gardner and McDaniel, (1978), Heath et al., (1983), Heath et al., (1986), Steward et al., (1986), Heidt et al., (1987), Saugey et al., (1988), Steward, (1988). Saugey et al., (1989), Sealander and Heidt, (1990), Tumilson et al., (1992), Saugey et al., (1993), McAllister et al., (1995), Saugey et al., (1998), Wilhide et al., (1998), Cochran, (1999), Caviness and James, (2001), Tumilson et al., (2002), and McAllister et al., (2005).

Results and Discussion

We captured 267 bats from 35 netting locations in 16 counties. The sampling period consisted of 41 nights beginning on 27 May 2004 and ending on 6 Sept 2004. Netting duration by county ranged from 1 to 6 nights with the total netting effort equal to 172 net nights. This study resulted in 44 new county records for 9 species of bats: the little brown bat (*M. lucifigus*, 3 records), the southeastern bat (*M. austroriparius*, 12 records), the eastern pipistrelle (*Pipistrellus subflavus*, 4 records), the big brown bat (*Eptesicus fuscus*, 1 record), the seminole bat (*Lasiurus seminolus*, 2 records), the red bat (*L. borealis*, 7 records), the hoary bat (L. cinereus, 2 records), the evening bat (*Nycticeius humeralis*, 6 records), and the Rafinesques' big eared bat (*C. rafinesquii*, 7 records). Dates and locations of county records organized by species are provided below.

Corynorhinus rafinesquii

1) Clay Co.–Dave Donaldson/Black River WMA 36°16' N 90°39' W, 2 June 2004. 5.1 km NW of Peach Orchard along shore of Little Black River. Two pregnant females, potentially indicating a nearby roost.

2) Desha Co.–Trusten Holder WMA 33°55' N 91°14' W, 20 Aug 2004. 3.7 km ENE of Pendleton. ATV trail leading into bottomland forest from the main road. Single non-reproductive (NR) female.

3) Lee Co.–Ozark-St. Francis National Forest 34°42' N 90°39'W, 20 Aug 2004. 0.5 km E of Jeffersonville. Cottonwood (Populus deltoidus) forest at confluence of L'anguille and St. Francis rivers. Single NR female.

4) Monroe Co.–Dagmar WMA 34°51' N 91°14' W, 22 July 2004. 10.1 km SW of Brinkley. Netted ATV trail and camping area along shoreline of bayou. Two males (one scrotal, one NR) and one NR female.

5) Poinsett Co.–Earl Buss/Bayou De View WMA 35°33' N 90°53' W, 3 June 2004. 2.9 km W of Weiner. Netted on ATV trails and river underneath Bayou DeView road bridge. Single scrotal male.

6) Prairie Co.–Wattensaw WMA 34°51' N 91°28' W, 13 July 2004. 4.2 km S of Gospoda. Campground area in upland forest along main road going to White River. Single NR female.

7) White Co.–Henry Gray/Hurricane Lake WMA 35°12' N 91°21' W, 10 July 2004. 6.4 km E of Mitchell Corner. Forest ATV trail in mature bottomland stand with trail leading from open water. Single scrotal male.

Eptesicus fuscus

8) Lonoke Co.–Holland Bottoms WMA 34°51' N 92°03' W, 15 July 2004. 0.5 km E of Jacksonville. Secondary-growth bottomland forest along shoreline of Jacks Bayou. Three NR females, one lactating female, and one NR male.

Lasiurus borealis

9) Clay Co.–Dave Donaldson/Black River WMA 36°16' N 90°39' W, 2 June 2004. Locality same as #1. Two females, one pregnant and one in estrus.

10) Crittenden Co.–Wapanocca NWR 35°20' N 90°11' W, 23 June 2004. 5.5 km SSE of Turrell. Ephemeral ponds located in open fields near large tracts of bottomland forests. Single lactating female.

11) Jackson Co.–Cache River NWR 35°29' N 91°07' W, 5 Aug 2004. 1.2 km W of Algoa. Captured along shore of Cache river adjacent to soybean field. Single NR female.

12) Lee Co.–Ozark-St. Francis National Forest 34°42' N 90°39'W, 20 Aug 2004. Locality same as # 3. One scrotal male.

13) Monroe Co.–Dagmar WMA 34°51' N 91°14' W, 22 July 2004. Locality same as # 4. Single NR female.

14) Poinsett Co.–Earl Buss/Bayou De View WMA

35°33' N 90o53' W, 3 June 2004. Locality same as #5. A single NR adult male.

15) Prairie Co.–Wattensaw WMA 34°51' N 91°28' W, 12 July 2004. 7.0 km NE of Center Point. Netted various points along river flowing towards main road in WMA. Four NR females.

Lasiurus cinereus

16) Crittenden Co.–Wapanocca NWR 35°20' N 90°11' W, 23 June 2004. Locality same as #10. Single unknown gendered individual landed in net but escaped before being further identified.

17) Jackson Co.–Cache River NWR 35°29' N 91°07' W, 5 Aug 2004. Locality same as # 11. Single inactive male.

Lasiurus seminolus

18) Crittenden Co.–Wapanocca NWR. 35°20' N 90°11' W, 3 Sept 2004. 3.2 km SSE of Turrell. On main dirt road in forested area at the easternmost end of levee. Single NR female.

19) Lonoke Co.–Holland Bottoms WMA 34°51' N 91°56' W, 30 July 2004. 3.5 km E of Jacksonville. Under bridge at Graham Road netted across creek. Single NR female.

Nycticeius humeralis

20) Arkansas Co.–Bayou Meto WMA 34°12' N 91°35' W, 10 Aug 2004. 13.4 km SE of Wabbaseka. Forest trail running along a large pond, netted both trail and pond. Two NR females, one NR male, and four scrotal males.

21) Crittenden Co.–Wapanocca NWR 35°20' N 90°11' W, 23 June 2004. Locality same as #10. Single NR male.

22) Lonoke Co.–Holland Bottoms WMA 34°51' N 92°03' W, 21 July 2004. Locality same as #8. Single scrotal male.

23) Mississippi Co.–Big Lake WMA 35°54' N 90°04' W, 13 June 2004. 13.0 km NE of Manila. Dense secondary bottomland forest intersection of ATV trail and ditch near MO border. Single pregnant female.

24) Prairie Co.–Wattensaw WMA 34°51' N 91°28' W, 12 July 2004. Locality same as # 15. Two scrotal males, one NR female, one pregnant female and one post-lactating female.

25) White Co.–Henry Gray/Hurricane Lake WMA 35°08N 91°21' W, 9 July 2004. 5.1 km SE of Mitchell Corner. Ditch running out of bottomland forest toward a small lake. Single pregnant female.

Pipistrellus subflavus

26) Crittenden Co.–Wapanocca NWR. 35°20' N 90°11' W, 23 June 2004. Locality same as #10. Two pregnant females and one lactating female.

27) Lonoke Co.–Holland Bottoms WMA 34°51' N 92°03' W, 21 July 2004. Locality same as #8. Single NR male.

28) Poinsett Co.–Earl Buss/Bayou De View WMA 35°33' N 90°53' W, 3 June 2004. Locality same as #5. Single pregnant, adult female.

29) White Co.–Henry Gray/Hurricane Lake WMA 35°08N 91°21' W, 9 July 2004. Locality same as #25. Three NR males and three NR females.

Myotis austroriparius

30) Arkansas Co.–Bayou Meto WMA 34°12' N 91°35' W, 10 Aug 2004. Locality same as #20. Three females and two males, all NR.

31) Clay Co.–Dave Donaldson/Black River WMA 36°16' N 90°39' W, 2 June 2004. Locality same as #1 Single scrotal adult male, one post-lactating and one NR female.

32) Craighead Co.–St. Francis Sunken Lands WMA, 35°46' N 90°18' W, 6 June 2004. 2.7 km SE of Lake City. Interior of cypress swamp netted in areas clear of obstructions on the water surface. Single pregnant female.

33) Crittenden Co.–Wapanocca NWR 35°20' N 90°11' W, 2 Sept 2004. 4.3 km SSE of Turrell. Forest trail leading to Wapanocca Lake. Single scrotal male and an accidental release of an unknown gender.

34) Jackson Co.–Cache River NWR 35°29' N 91°07' W, 15 Aug 2004. 0.7 km W of Algoa. Flooded forest and shoreline along the Cache river. Single NR male.

35) Lawrence Co.–Shirey Bay/Rainey Brake WMA 35°59' N 91°07' W, 11 June 2004. 5 km SW of Lynn. Netted at confluence of creek with CR 316. Single scrotal male.

36) Lee Co.–Ozark-St. Francis National Forest 34°42' N 90°39'W, 20 Aug 2004. Locality same as # 3. Single

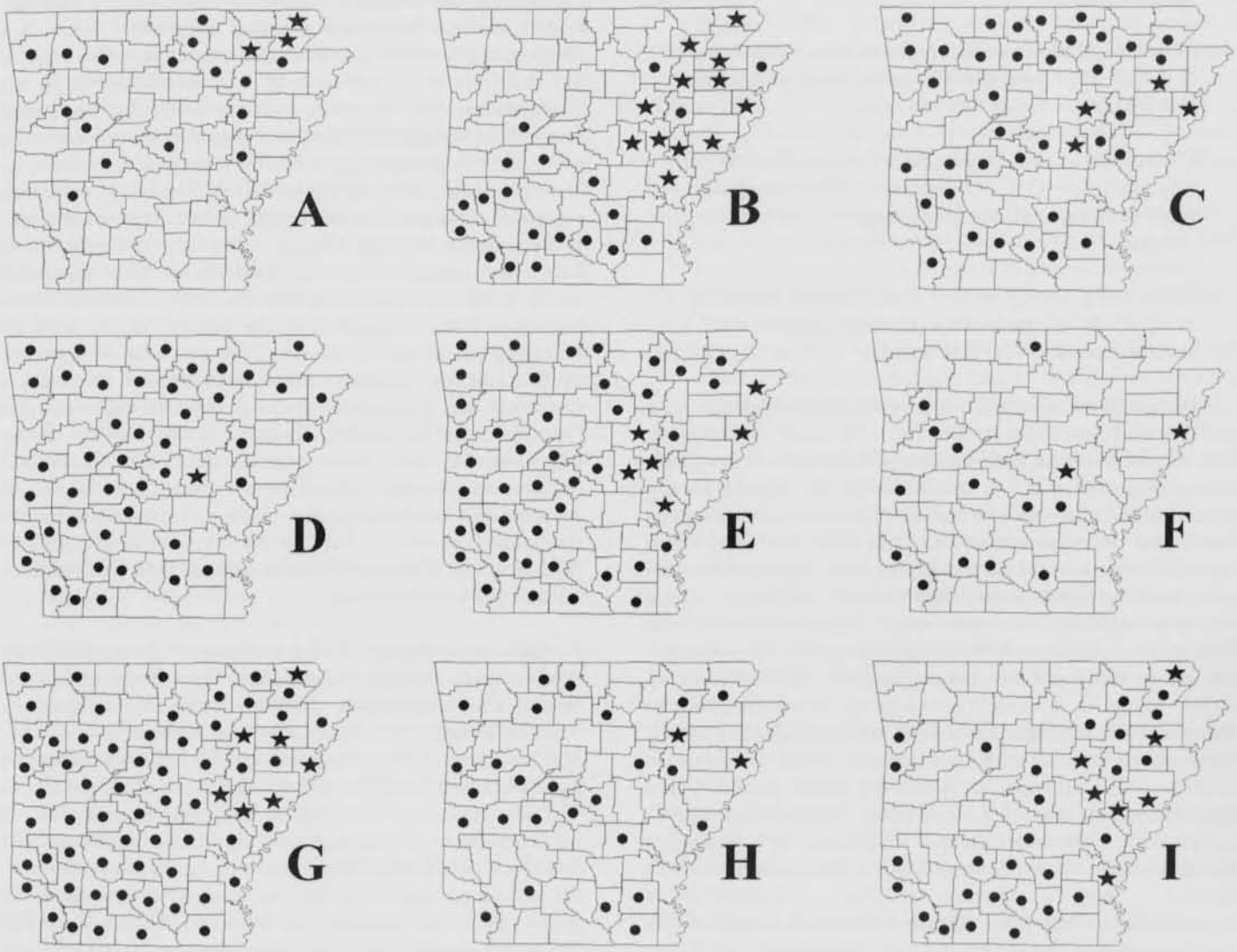

Fig. 1. Distributions for 9 species of bats encountered during a 2004 county by county survey of the Arkansas delta region. A. Little brown bat, *Myotis lucifugus*, B. Southeastern bat, *Myotis austroriparius*, C. Eastern pipistrelle, *Pipistrellus subflavus*, D. Big brown bat, *Eptesicus fuscus*, E. Evening bat, *Nycticeius humeralis*, F. Seminole Bat, *Lasiurus seminolus*, G. Red bat, *Lasiurus borealis*, H. Hoary bat, *Lasiurus cinereus*, I. Rafinesque's big eared bat, *Corynorhinus rafinesquii*. "Stars" indicate county records from this study and "solid circles" indicate previously published county records.

NR female that was likely attracted by the distress calls of a Rafinesque big-eared bat that was being removed at the time resulting in two county records at once.

37) Lonoke Co.–Holland Bottoms WMA 34°51' N 91°56' W, 30 July 2004. Locality same as # 19. One NR and one scrotal male.

38) Monroe Co.–Dagmar WMA 34°51' N 91°14' W, 22 July 2004. Locality same as # 4. Three NR females and one post-lactating female.

39) Poinsett Co.–Earl Buss/Bayou De View WMA 35°33' N 90°53' W, 3 June 2004. Locality same as # 5. Four females, three pregnant adults and one juvenile.

40) Prairie Co.–Wattensaw WMA 34o51' N 91o28' W, 12 July 2004. Locality same as # 15. Single post-lactating female.

41) White Co.–Henry Gray/Hurricane Lake WMA 35°08N 91°21' W, 9 July 2004. Locality same as #25. Two scrotal males, one NR male, and one post-lactating female.

Myotis lucifigus

42) Clay Co.–Dave Donaldson/Black River WMA 36°16' N 90°39' W, 2 June 2004 Locality same as #1. Three NR females.

43) Greene Co.–Lake Ashbaugh 36°11' N 90°46' W, 25 July 2004. 5.9 NW of Deleplaine. Netted along shore of ditch running from the lake. Single NR unknown gender.

44) Lawrence Co.–Shirey Bay/Rainey Brake WMA 35°59' N 91°07' W, 11 June 2004. Locality same as #35. One scrotal male and one NR female.

Distributions of bats from previous published accounts and this study are illustrated in Fig. 1. Both *M. austroriparius* and *C. rafinesquii* appear to occur throughout the Delta region (Fig. 1-B and 1-I, respectively) but appear locally abundant seeming to favor tracts of late-successional forests dominated by cypress-tupelo and oak trees. Although often captured together, *M. austroriparius* was captured in nets over standing water more often than *C. rafinesquii*, which was only captured once over water. This is consistent with Menzel and Menzel (2001) suggestion that *C. rafinesquii* forages in more upland areas. Similarly, Mirowsky et al. (2004) found *C. rafinesquii* roosts to be more prevalent in American beech (*Fagus grandifolia*) and oak (*Quercus*), which were more characteristic of upland sites, whereas in contrast, *M. austroriparius* preferred water tupelo (*Nyssa aquatica*) and sweetgum (*L. styraciflua*). The association of *C. rafinesquii* with bottomland habitats is somewhat paradoxical, as their roosts are more often associated with upland tree species. The capture of *L. seminolus* in Crittenden County expands the range of this species in Arkansas, eastward toward the Mississippi River, (Fig. 1-F). Captures of *M. lucifigus* in the northeastern corner of the state (Fig. 1-A) provide evidence for this species on the southern limit of its more northerly range, as it has rarely been reported in southern parts of the state (Sealander and Heidt, 1990).

In this study we targeted *M. austroriparius* and *C. rafinesquii*, and county sampling was completed when these species were captured. As a result of this, non-target bat species may have been overlooked when sampling in the area only consisted of a night or two. Repeated sampling of these areas may have revealed more uncommon species that were overlooked in this study. More intensive sampling in these areas would definitely be worthwhile, since distribution records for non-target species, such as *L. seminolus* and *L. cinereus* are similarly incomplete for the Delta region.

The range of both *M. austroriparius* and *C. rafinesquii* encompass most of the southeastern US, and Arkansas is situated on the westerly portion of their range, although both species extend into portions of eastern Texas (see Horner, 1995; 1996). The westerly range of these bats mimic the distribution of bottomland hardwood forests in the Southeastern US. However, most research on the natural history and biology of these two bat species have focused on more easterly populations, with less research on the margins of their range. As late-successional forests in the Delta region of Arkansas are becoming increasingly fragmented and separated by large areas of agriculture, the impacts on bat communities could be substantial, thus increased research on these species in this area may aid conservation initiatives. For instance, knowledge of roosting behavior for *M. austroriparius* and *C. rafinesquii* has not been well studied in Arkansas (but see Reed, 2004), and research on roosts of the latter have primarily focused on artificial structures (Tumilson et al., 1992; Saugey et al., 1993). Forest fragmentation and water management may potentially impact specialized bottomland species, such as *M. austroriparius* and thus more research is required on the specialized needs of bats in the bottomland forests of Arkansas, so that suitable management and conservation initiatives can be devised.

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Annotated Checklist of the Amphipoda of Arkansas with Emphasis upon Groundwater Habitats

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Abstract

Based on recent collections and review of the literature, 20 species of freshwater amphipod crustaceans are listed from the state of Arkansas. Included are species from the families Allocrangonyctidae, Crangonyctidae, Gammaridae and Hyalellidae and the genera *Allocrangonyx*, *Bactrurus*, *Crangonyx*, *Stygobromus*, *Gammarus*, and *Hyalella*. Ten of the species are restricted to subterranean groundwaters, 2 are closely associated with groundwater but also occur in surface waters, and 8 are known primarily from surface waters. Two of the species are in the process of being described in the literature, whereas 2 remain only provisionally recognized to date. On the basis of this new list, some revisions to the current rarity rankings are recommended.

Introduction

All records of Amphipoda in Arkansas are summarized, including new state, county, and site records. More than one-half of all species recorded from the state are closely associated with groundwater habitats and the majority of them are stygobites. These species are typically troglomorphic (i.e., eyeless, unpigmented) and obligatory to subterranean groundwaters. The principal groundwater habitats investigated in Arkansas include streams and pools in caves, water wells and the outflows of springs and seeps. Collections were made by hand using pipettes, dip nets, aspirators, and occasionally bait traps consisting of mesh bags filled with leaves. Specimens collected during this study were preserved in 70-90% ethanol, and most are in the research collection of Holsinger. All of this material will eventually be deposited in the Smithsonian Institution's National Museum of Natural History (USNM). Taxonomic identifications were performed by Holsinger, with assistance from Slay and S. Longing (University of Arkansas). Taxonomic keys used included those in Holsinger (1967, 1972) and in unpublished manuscripts. Records of amphipoda from all available literature sources were also reviewed, summarized, and cited, as well as those from unpublished sources including the Natural Heritage Database maintained by the Arkansas Natural Heritage Commission (ANHC, C. Osborne, data manager) and the Subterranean Amphipod database (in progress) at Old Dominion University, Norfolk, Virginia (searchable on the Internet at the following address, (URL=http://web.odu.edu/sci/biology/amphipod/). Amphipod records published by others are cited after each occurrence; all other records are unpublished data of the authors and colleagues.

List of All Amphipod Taxa Recorded at Present from the State of Arkansas

Family Allocrangonyctidae Holsinger, 1989

Allocrangonyx hubrichti Holsinger, 1971

White County: M. Longley's well in the town of Romance, 6 Nov. 1996, 1 male (Robison and Holsinger, 2000). *Allocrangonyx hubrichti* is also reported from caves and the hyporheic habitat (subterranean underflow) of surface streams in 14 counties in Missouri (Holsinger, 1989; Sarver and Lister, 2004).

Family Crangonyctidae Bousfield, 1973

Bactrurus pseudomucronatus Koenemann and Holsinger, 2001

Lawrence County: "deep cistern, 5 miles south of Imboden" 16 Sept. 1940, 1 male collected by B. Marshall in USNM (Koenemann and Holsinger, 2001). **Randolph County:** Mansell Cave (Koenemann and Holsinger, 2001). *Bactrurus pseudomucronatus* is also reported from Missouri but

it is restricted to the Salem Plateau subecoregion of both states (Koenemann and Holsinger, 2001). Dunivan et al. (1982) mistakenly referred to the Mansell Cave population of this species as *Bactrurus mucronatus* (Forbes, 1876) but this record was for the closely similar *B. pseudomucronatus*. *Bactrurus mucronatus* is recorded from subterranean groundwaters, primarily from drain tile outlets, in glacial drift areas in Illinois, Indiana, Iowa, Michigan, and Ohio (Koenemann and Holsinger, 2001).

Bactrurus speleopolis Holsinger et al., 2006

Marion County: Marble Falls Cave, 7 Sep. 2001, 3 counted in subterranean stream and 1 collected by Graening and Slay. **Sharp County:** Cave City Cave, 13 Dec. 2001, 20 counted and 6 collected by Graening, D. Fenolio, and J. Stark; 23 Nov. 2002, 25 counted by Graening and D. Fenolio; 11 Dec. 2004, 8 counted by S. Wallace.

Bactrurus sp. (unidentified)

Independence County: Cave Spring Cave, 5 Oct. 2002, 2 collected by Graening, S. McGinnis, H. Bryant, and C. Blevins.

Crangonyx aka Zhang and Holsinger, 2003

Crangonyx aka is known only from central Arkansas and from only 4 collections: 1 stream in Pope County; 2 streams in Van Buren County; and 1 seep in Saline County–"seep 0.8 km S of Hector on state rd. 27" (Zhang and Holsinger, 2003).

Crangonyx forbesi (Hubricht and Mackin, 1940)

Fulton County: Mammoth Spring; "small spring near Mammoth Spring" (Zhang and Holsinger, 2003). **Independence County:** Cave Spring Cave, 5 Oct. 2002, 32 counted and 2 collected by Graening, S. McGinnis, H. Bryant, and C. Blevins. **Lawrence County:** "spring 3.7 miles south of Imboden" (Hubricht, 1943). **Sharp County:** Eckel Cave, 22 Nov. 2002, 1 collected by Graening and D. Fenolio. Although not a stygobite, *C. forbesi* is commonly found in cave streams and springs in Kansas, Kentucky, Illinois, Indiana, Missouri, Ohio, and Oklahoma. It is also reported from a number of surface streams and occasionally ponds. Many of the cave populations show some degree of morphological modification for a subterranean existence (Hubricht, 1943; Zhang and Holsinger, 2003).

Crangonyx minor Bousfield, 1958

Greene County: "seep 8.0 km N of Brookland" (Zhang and Holsinger, 2003). *Crangonyx minor* is also reported from Illinois, Iowa, Kentucky, Ohio, Oklahoma, Tennessee, and southern Ontario and inhabits a variety of aquatic habitats including small streams, sloughs, ditches, drains, springs, and ponds (Bousfield, 1958; Zhang and Holsinger, 2003).

Crangonyx obliquus (Hubricht and Mackin, 1940)

Crangonyx obliquus is recorded from surface waters in the following Arkansas counties: Faulkner, Jefferson, Johnson, Monroe, Perry, Phillips, and Yell (Hubricht and Mackin, 1940; Hubricht, 1943; Zhang and Holsinger, 2003). This species is largely restricted to the Coastal Plain of the south-central United States (Zhang and Holsinger, 2003). It was incorrectly listed as a troglophile [stygophile] in the cave fauna of Arkansas by McDaniel and Smith (1976).

Crangonyx pseudogracilis Bousfield, 1958

Boone County: "large spring near Willcockson", 8 April 1939 (Zhang and Holsinger, 2003). *Crangonyx pseudogracilis* is recorded from surface waters in the following Arkansas counties: Arkansas, Ashley, Calhoun, Conway, Cross, Dallas, Faulkner, Garland, Grant, Jackson, Jefferson, Johnson, Lawrence, Monroe, Nevada, Ouachita, Perry, Phillips, Pulaski, Union, and Yell (Zhang and Holsinger, 2003). *Crangonyx pseudogracilis* is widely distributed in southern Canada and east-central United States (Bousfield, 1958; Zhang and Holsinger, 2003). Earlier Arkansas records for *Eucrangonyx gracilis* by Hubricht and Mackin (1940) and *C. gracilis gracilis* by Hubricht (1943) refer to *C. pseudogracilis* as presently understood.

Stygobromus alabamensis sensu latu (Stout, 1911)

Baxter County: Norfork Bat Cave, 13 Sep. 2000,20 counted by Graening and B. Wagner (Graening et al., 2004). **Benton County:** Cold Cave, 10 April 2000, 50 counted by Graening and Slay; "seep near Big Spring, Bella Vista" (Holsinger, 1967). **Boone County:** "seep 9 miles southwest of Harrison" (Holsinger, 1967). **Carroll County:** cave on North Boundary Trail, 12 Aug. 2000, 11 counted in drip pool by Graening; Huckleberry Point Cave, 18 Sep. 2002, 1 collected by B. Wagner; sampling site on Kings River, 6 March 2002, several collected by Slay and A. Brown. **Crawford County:** US Forest Service cave # 230109, 9 April 2000, 4 collected in drip pool by Slay and J. Briggler; US Forest Service cave #23040, 9 April 2000, 2 collected by Slay and J. Briggler. **Independence County:** (Holsinger, 1967). **Izard County:** Bergren Cave, 16 Aug. 2002, 1 collected by Graening and R. Schroeder; Donovan Cave, 1976, reported as *Stygobromus* sp. in McDaniel and Smith (1976); Needles Cave, 7 June 1975 (Smith, 1977), and 1 Feb. 2003, 10 counted and 5 collected by Graening, Slay, and E. Corfey. **Jackson County:** Mason's Cave (McDaniel et al.,

1979; this study); "spring 1.5 miles southwest of Olyphant" (Holsinger, 1967). **Logan County:** "seep 0.6 miles east of Magazine Mt. Lodge" (Holsinger, 1967). **Madison County:** Simpson's Cave, 9 July 2000, 100 counted by Graening and S. McGinnis; Wounded Knee Cave, 27 May 2001, 2 collected by Graening and C. Brickey. **Marion County:** Coon Cave, 14 Sep. 1979 (Welbourn and Lindsley, 1979); Elm Cave, 16 Nov. 2001, 1 collected by Graening and B. Sasse; Middle Creek Spring Cave, 15 July 1977 (Lindsley and Welbourn, 1977). **Montgomery County:** Brier Springs and Rattlesnake Springs, collected by H. Robison. **Newton County:** Cave Mountain Cave, 29 June 2001, 4 collected by C. Bitting; Chilly Bowl Cave, 4 Aug. 2001, 1 collected by Slay, C. Brickey, and M. Covington; Copperhead Cave, 14 Nov. 1999, 1 collected by Slay; Corkscrew Cave (Youngsteadt and Youngsteadt, 1978); Friday the 13th Cave, 15 April 2000, 10 counted by Slay and S. Allen; Lewis Spring Cave, 1976 (Youngsteadt and Youngsteadt, 1978); Mr. Clean Cave, 6 July 2001, 2 counted in drip pools, 1 collected by Slay and C. Bitting; Saltpeter Cave, 17 March 2002, 50 counted and 2 collected by Slay and M. Covington; Stillhouse Hollow Cave, 23 June 2001, 10 counted and 2 collected by Graening, Slay, and C. Bitting; Tom Watson's Bear Cave, 26 Jan. 2002, 4 collected by Slay, C. Brickey, and M. Ross; "seep 9.6 miles south of Boxley" (Holsinger, 1967); "seeps 4 miles south of Boxley" (Holsinger, 1967); "seeps below Lookout Point, 7 miles south of Jasper" (Holsinger, 1967); Wolf Creek Cave, 14 Jan. 2000, 1 collected by Graening and R. Redman. **Searcy County:** Big Creek Cave, 16 March 2002, 13 counted by Graening and C. Brickey (Graening et al., 2004); "seeps 3.0 miles east of Harriet" (Holsinger, 1967); "small seep 4.1 miles west of Marshall" (Holsinger, 1967); Wood's Hollow Cave #1, 16 March 2002, 10 counted and 1 collected by Graening and C. Brickey (Graening et al., 2004). **Stone County:** Bald Scrappy Cave (McDaniel and Smith, 1976); Biology Cave, 23 May 1981 (Welbourn, 1983), and 17 Sep. 2000, 2 counted by D. Fenolio, C. Brickey, and S. Longing (Graening et al., 2004); Blanchard Springs Caverns, 1976 (McDaniel and Smith, 1976); Breakdown Cave, 17 May 1980, R. Schroeder (Welbourn, 1980); Gunner Cave, 17 May 1980 (Welbourn, 1980); Hammer Springs Cave, 26 April 1980, Jagnow, Welbourn, and Blore (Welbourn, 1980); Martin Hollow Cave, 14 Oct. 2000, 3 collected by Graening, Slay, M. Covington, C. Brickey, and J. Gunter; Saltpeter Cave, 31 March 2002, 1 collected by Graening, D. Fenolio and C. Brickey (Graening et al., 2004); "seep near Blanchard Falls" (Holsinger, 1967). **Van Buren County:** "seep 5.5 miles north of Winslow" (Holsinger, 1967). **Washington County:** seep on M. Evan's property, 1 March 2002, 3 collected by Graening and Slay; spring at Bradley Shelter, 2 April 2000, 30 counted and 2 collected by Graening and Slay; storm sewer under University of Arkansas Physics Building, 17 Feb. 2003, 2 collected by Graening and D. Fenolio. *Stygobromus alabamensis* is also reported from numerous groundwater habitats in Alabama, Kansas, Louisiana, Mississippi, Missouri, Oklahoma, Tennessee, and Texas, and it is the most widely distributed stygobitic species in North America (Holsinger, 1967).

Stygobromus elatus (Holsinger, 1967)

Stygobromus elatus is known only from a single site in Logan County: "seep 0.2 miles east of Magazine Mt. Lodge," 4 May 1940, 4 deposited in USNM by L. Hubricht (Holsinger 1967), and 1 April 1980, K. Smith (ANHC 2001). There is a strong possibility that this species is synonymous with *Stygobromus alabamensis* (see above) (Holsinger, in manuscript).

Stygobromus montanus (Holsinger, 1967)

Stygobromus montanus is known only from Polk County in 2 springs at Queen Wilhelmina State Park on Rich Mountain, 26 April 1936, 20 collected by L. Hubricht (Holsinger, 1967), and 22 April 1981, 9 collected by K. Smith and J. Rettig (ANHC, 2001).

Stygobromus onondagaensis (Hubricht and Mackin, 1940)

Benton County: Arkansas Archaeological Survey Site #3BE532, 9 Nov. 1999, 1 collected by Graening and M. Evans; Big Spring, 7 July 2000, 1 collected by Graening and Slay; Cave Springs Cave, 1968, T. Poulson, M. Cooper, and R. Norton; Tanyard Creek Nature Trail Cave, 5 Jan. 2003, 5 counted and 1 collected by Graening and S. McGinnis. *Stygobromus onondagaensis* is relatively common in caves in Missouri and is also recorded from caves in the adjacent states of Kansas and Oklahoma (Hubricht, 1943; Holsinger, in manuscript).

Stygobromus ozarkensis (Holsinger, 1967)

Benton County: Bear Hollow Cave, 7 Dec. 2000, 8 counted and 1 collected by Slay and Graening; Blowing Springs Cave, 27 Sep. 2001, 1 collected by Slay, L. Moritz, and M. Covington; Cave Springs Cave, 30 Oct. 1972, J. Holsinger (Holsinger, 1972), and 30 Nov. 2000, 1 counted by Graening; Civil War Cave, 23 Nov. 1999, 200 counted and 2 collected by Graening, A. Brown and Slay, and 29 Oct. 2000, 14 counted by Slay, Graening, and A. Brown; Dickerson Cave, 19 April 1980, A. Brown and M. Schram (Schram, 1980), and 8 Oct. 1999, 1 counted by Slay; Logan Cave, K. Herbert (Herbert, 1994), and 15 Dec. 1999, 1 counted by Graening , and 21 Nov. 2000, 2 counted by Graening and Slay; Old Pendergrass Cave, 10 Dec. 1999, 2 collected by Graening and Slay, and 24 April 2000, 1 counted by Graening and B. Wagner; Spavinaw Creek

Cave, 1 Sept. 1999, 2 collected by Slay; Tom Danforth Cave, 14 Oct. 1963, 1 collected by D. Martin (Holsinger, 1967); War Eagle Cavern, 11 Feb. 2000, 1 collected by Graening and S. McGinnis , and 11 May 2001, 2 counted by A. and C. Brown. **Carroll County:** cave above Black Bass Lake, 11 Oct. 2002, 1 collected by Graening and D. Renko; "White River below Beaver Dam" (Schram, 1982). **Izard County:** Clay Cave (McDaniel et al. 1979); Needles Cave, 1 Feb. 2003, 1 collected by Graening, Slay, and E. Corfey. **Madison County:** Hunter's Cave, 28 April 2001, 1 collected by Graening and J. Gunter; War Eagle Cave, 6 Aug. 1978, M. Schram (Schram, 1983); Withrow Springs Cave, 2 collected by M. Schram (Schram, 1983). **Marion County:** Boat Creek Mine, 5 Aug. 2002, 2 collected by Slay, C. Bitting and M. Taylor; Reed Cave, 9 March 2002, 1 collected by Graening and S. McGinnis. **Newton County:** Fitton Cave, 1982, L. Willis, and 15 Jan. 2000, 1 collected by Graening and R. Redman, and 13 May 2001, 3 counted by Graening and C. and C. Bitting; Fitton Spring Cave, 5 Oct. 2000, 6 counted and 3 collected by Slay and C. and C. Bitting; John Eddings Cave, 21 Sep. 2000, 1 counted by Graening, Slay, and C. Bitting; Pretty Clean Cave, 7 July 2001, 1 collected by Slay and C. Bitting; Sherfield Cave, 10 June 2000, 2 collected by Graening; Walker Mountain Overflow Cave, 19 March 1983, 1 collected by A. Grubbs. **Stone County:** Flitterin' Pit, 24 Nov. 2002, 1 collected by Graening, D. Fenolio, and C. Brickey. **Washington County:** Copperhead Spring, 28 Nov. 2000, 4 counted by Slay and J. Gunter. *Stygobromus ozarkensis* is also reported from Missouri and Oklahoma but it is restricted to the Ozark Plateaus ecoregion of all three states (Holsinger, 1967). Earlier Arkansas records for *S. clantoni* from Clay Cave by McDaniel et al. (1979), from Fitton Spring Cave by Lindsley (1977), and from John Eddings Cave by Welbourn and Lindsley (1979) are erroneous and refer to *Stygobromus ozarkensis* as presently understood (Holsinger, in manuscript). *Stygobromus clantoni* (Creaser, 1934) was previously reported in Arkansas by Mackin and Hubricht (1940) and Hubricht (1943), but all of these records have since been attributed to other species of *Stygobromus.* However, *S. clantoni* is authentically recorded from caves and water wells in nearby Kansas and Missouri (Holsinger, 1967; in manuscript).

Stygobromus sp. nov. Holsinger, in manuscript

Carroll County: Blowing Springs Cave, 28 April 2001, 20 counted by Graening, J. Gunter, R. Honebrink, and B. Wagner (Graening et al., 2004). **Independence County:** Cave Spring Cave, 5 Oct. 2002, 1 collected by Graening, S. McGinnis, H. Bryant, and C. Blevins; Chinn Springs Cave, 10 Nov. 2000, 5 counted and 1 collected by Graening, E. Corfey, and B. Wagner; Blowing (Dozen's Den) Cave, 12 Dec. 2000, 6 counted by Graening, Slay, and B. Wagner. **Marion County:** Reed Cave, 15 Nov. 2001, 3 counted and 1 collected by Graening, T. Snell, and P. Shurgar. **Sharp County:** Cave City Cave, 23 Nov. 2002, Graening and D. Fenolio, 1 collected; Eckel Cave, 22 Nov. 2002, 1 collected by Graening and D. Fenolio. **Stone County:** Nesbitt Spring Cave, 30 March 2002, 1 collected by Graening, Slay, B. Wagner, and C. Brickey; Rowland Cave, 5 Oct. 2001, 2 collected by Graening, Slay, D. Taylor, and W. Meurer (Graening et al., 2004). This new species of *Stygobromus* is also recorded from many caves in Missouri but is restricted to the Ozark Plateaus ecoregion in both states (Holsinger, in manuscript).

Stygobromus sp. nov.

Montgomery County: Boxx Springs, 19 June 1996, 6 specimens collected by H. Robison. This is a provisionally recognized undescribed new stygobitic species distinguished by a sexually dimorphic male gnathopod 2 and the absence of a ramus from uropod 3 (Holsinger, unpublished data).

Stygobromus sp. (unidentified)

Benton County: Congo Crawl, 1 May 2001, 1 counted by Slay and A. Brown. **Madison County:** Pine Creek Cave, 11 Feb. 2000, 1 counted by Graening and Slay; Womack Spring Cave, 6 Dec. 2000, 1 collected by Graening and C. Brickey. **Marion County:** Rush Landing Spring Cave, 26 March 1977 (Lindsley and Welbourn, 1977). **Newton County:** Stockman Cave, 11 Dec. 2004, 3 collected from drip pools by Graening and D. Fenolio; Walnut Cave, 13 July 1977 (Lindsley and Welbourn, 1977). **Searcy County:** Back o' Beyond Cave, 31 March 2001, 1 counted by Slay and C. Bitting. **Stone County:** Herald Hollow Cave, 23 March 2001, 3 counted by Graening and Slay (Graening et al., 2004). Most of these specimens could not be positively determined because they were sexually immature or damaged.

Synurella bifurca (Hay, 1882)

Jackson County: "spring 1.5 miles southwest of Olyphant" (Hubricht and Mackin, 1940). *Synurella bifurca* is also reported from surface water habitats in the following Arkansas counties: Calhoun, Craighead, Cross, Dallas, Jefferson, Lawrence, Monroe, Phillips, and Pulaski. *Synurella bifurca* is a widespread epigean species in the southern United States and commonly occurs throughout much of Louisiana and Mississippi (Hubricht and Mackin, 1940; Hubricht, 1943; Holsinger, 1972).

Family Gammaridae Latreille, 1802

Gammarus minus sensu latu Say, 1818

Benton County: Big Spring, Bella Vista (Hubricht, 1943); Cave Springs Cave and spring run, 1 Dec. 1996 and 4 Nov. 1999, 1 to 100 individuals per square meter in cave stream resurgence counted by Graening; Logan Cave and spring run, 7 Nov. 1982, 407 counted by L. Willis (Brussock et al., 1988), and 24 May 2002, 4 counted by Graening; "spring, 2 miles south of Gentry" (Hubricht, 1943); "rocky creek and spring 1 mile south of Missouri-Arkansas state line on U.S. Hwy. 59" (Reimer, 1969). **Boone County:** "large spring near Willcockson" (Hubricht and Mackin, 1940). **Fulton County:** Mammoth Spring (Hubricht and Mackin, 1940). **Independence County:** Cushman Cave, 26 Jan. 2001, 1,000 counted by Graening, C. Brickey, and E. Corfey. **Izard County:** cave on Mr. Griffin's property, 25 June 2002, 8 collected by B. Wagner; Needles Cave, 7 June 1975 (Smith, 1977), and 1 Feb. 2003, 500 counted and 6 collected by Graening, Slay, and E. Corfey. **Marion County:** Cold Spring, 1 Oct. 1979 (Welbourn and Lindsley, 1979); Rush Spring (Welbourn and Lindsley, 1979); Wishbone Spring, 23 March 1977 (Lindsley and Welbourn, 1977). **Newton County:** Flowstone Facade Cave, 5 Oct. 2000, 50 counted and 1 collected by Slay and C. Bitting; John Eddings Cave, 31 Oct. 1979 (Welbourn and Lindsley, 1979); Sprite Cave, 16 March 2002, 30 counted and 18 collected by Slay and M. Covington. **Searcy County:** Blowing Spring Cave, 12 Dec. 2001, 100 counted and 7 collected by Graening and D. Fenolio (Graening et al., 2004); resurgence of Hurricane River Cave (Hubricht, 1943). **Stone County:** Martin Hollow Cave, 14 Oct. 2000, 1,000 counted and 2 collected by Graening, Slay, M. Covington, C. Brickey, and J. Gunter. **Washington County:** Cave Spring, 31 March 2000, 100 counted by Graening and J. Gunter. *Gammarus minus* is probably a species complex and is reported from springs and cave streams throughout the Appalachian Mountains, Interior Low Plateaus, and Ozark Plateaus ecoregions (Hubricht, 1943; Holsinger, 1972). Populations of *G. minus* occurring in Arkansas, Missouri, and Oklahoma have been defined as a geographical type (Ozarkian) based on morphological variation (Cole, 1970). Previous records for *Gammarus propinquus* from "a large spring near Willcockson" in Boone County and from Mammoth Spring in Fulton County by Hubricht and Mackin (1940), and *Gammarus elki* from a "rocky creek and spring 1 mile south of the Missouri-Arkansas state line" in Benton County by Reimer (1969) refer to *G. minus* as presently understood and listed above. Both *G. propinquus* Hay and *G. elki* Reimer are now considered synonyms of *G. minus* (see Shoemaker, 1940; Holsinger, 1972).

Gammarus pseudolimnaeus Bousfield, 1958

Lawrence County: "Wautuga Springs, 2.9 miles southeast of Ravenden" (Hubricht, 1943). This species is recorded from streams and cave springs in northern Arkansas, where it may occur syntopically in springs with *G. minus* (Holsinger, 1972). *Gammarus pseudolimnaeus* is widespread and reported from a number of states, including Illinois, Missouri, Oklahoma, Kentucky, Michigan, Wisconsin, and Quebec and Ontario in Canada (Holsinger, 1972). The record for *G. limnaeus* from Wautuga Springs in Lawrence County by Hubricht (1943) is referable to *G. pseudolimnaeus* as presently understood. Many of the earlier records for *Gammarus limnaeus* Smith became *G. pseudolimnaeus* when Bousfield (1958) described the latter as a new species and made *G. limnaeus* a subspecies of *Gammarus lacustris* (see Bousfield, 1958; Holsinger, 1972).
Gammarus sp. nov. (awaiting description)

Stone County: Martin Hollow Cave, 14 Oct. 2000, 20 counted and 7 collected by Graening, Slay, M. Covington, C. Brickey, and J. Gunter.

Gammarus sp.

Benton County: War Eagle Cavern, 4 Nov. 1978, M. Schram (Schram, 1980). **Carroll County:** White River below Beaver Dam, 1 July 1978, M. Schram (Schram, 1980). **Stone County:** Cave River Cave, 24 Nov. 2002, 10,000 counted by Graening, D. Fenolio, and C. Brickey. Hargis (1995) reported *Gammarus* from Crawford, Franklin, and Johnson counties.

It should be noted that *Gammarus fasciatus* (Say, 1818), was reported from Arkansas by Cather and Harp (1975) and listed in Johnson (1979). However, the established range of this species suggests that the Arkansas records are in error. As presently understood, *Gammarus fasciatus* is known authentically from the upper Mississippi River drainage eastward throughout the Great Lakes area and south along the Atlantic Coastal plain to southern North Carolina (Holsinger, 1972).

Family Hyalellidae Bulycheva, 1957

Hyalella azteca (Saussure, 1858)

Benton County: Big Spring, 7 July 2000, 100 counted and 5 collected by Graening and Slay. **Craighead County:** Big Creek, 1969 (Cather and Harp, 1975). **Garland County:** Meyers Springs, collected by H. Robison. **Montgomery County:** Boxx Springs, Rattlesnake Springs, Singing Springs, and Wehunt Springs, collected by H. Robison. **Randolph County:** Janes Creek, 1969 (Cather and Harp, 1975). Despite its widespread distribution throughout much of North America, (Hubricht and Mackin, 1940; Hubricht, 1943; Bousfield, 1958), *Hyalella azteca* apparently represents a complex of morphologically closely similar cryptic species

(Witt et al., 2000). It is probably more common in Arkansas than current records indicate.

Results and Discussion

The first state checklist of the Amphipoda of Arkansas was by Johnson (1979), who reported 13 taxa, 11 of which remain valid. Twenty species of amphipods are known at present, 18 of which have been found in groundwater habitats. The species are distributed among 4 families as follows: Allocrangonyctidae (1 species of *Allocrangonyx*); Crangonyctidae (2 species of *Bactrurus*, 5 species of *Crangonyx*, 7 species of *Stygobromus*, and 1 species of *Synurella*); Gammaridae (3 species of *Gammarus*); Hyalellidae (1 species – *Hyalella azteca*). Four of the 20 are provisionally recognized new species that belong to *Gammarus*, *Bactrurus*, and *Stygobromus* as indicated in the preceeding list. A description of the new species of *Bactrurus* is in press and a description of one of the new species of *Stygobromus* is in manuscript. Previous studies suggest that more than one-half of North American freshwater amphipod species occur exclusively in subterranean waters (Holsinger, 1967), and this observation applies generally to Arkansas, where 10 of the 20 species recognized in this report are stygobites and 2 others are stygophiles that are closely associated with cave waters.

Two of the principal goals of this checklist are to update the range and conservation status of the species of freshwater amphipods reported from Arkansas. Contained in the checklist are the first state records for *S. onondagaensis*, and new county records for *S. alabamensis*, *S. ozarkensis*, *C. forbesi*, and *G. minus*. However, *S. montanus*, *S. elatus*, and *C. aka* remain single-site endemics. Therefore, based on the revised distribution of amphipods in Arkansas, new biodiversity rankings are recommended for the Natural Heritage Program and its scientific advisory group NatureServe. Of special concern are the locally-rare species *A. hubrichti*, *B. pseudomucronatus*, *C. aka*, *C. forbesi*, *S. elatus*, *S. montanus*, and *S. onondagaensis*. Conversely, *S. ozarkensis* and *S. alabamensis* are now known from enough sites to warrant their removal from the list of rare and imperiled fauna. Suggested revisions of rarity rankings for Arkansas amphipods are enumerated in Table 1.

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Table 1. Current rarity rankings and suggested revisions at the Global (G-rank) and Subnational /State (S-Rank) levels, where a rank of 1 indicates that the species is critically imperiled and a rank of 5 indicates that the species is demonstrably widespread and secure. The reader is referred to NatureServe (2005) for a complete explanation of the ranking system and access to the national database.

Species	Current Global Rank	Suggested Global Rank	Current State Rank	Suggested State Rank
Allocrangonyx hubrichti	G2G3	G2	not ranked	S1
Bactrurus pseudomucronatus	G2G3	G2	not ranked	S1
Crangonyx aka	not ranked	G1	not ranked	S1
Crangonyx forbesi	not ranked	G3	not ranked	S1
Crangonyx minor	not ranked	G5	not ranked	S4
Crangonyx obliquus	not ranked	G4	not ranked	S3
Crangonyx pseudogracilis	not ranked	G5	not ranked	S4
Gammarus minus	not ranked	G4	not ranked	S4
Gammarus pseudolimnaeus	G5	G4	not ranked	S3
Hyalella azteca	G5	no change	not ranked	S4
Stygobromus alabamensis	G5	no change	not ranked	S4
Stygobromus elatus	G1G2	G1	S1	no change
Stygobromus montanus	G1G2	G1	S1	no change
Stygobromus onondagaensis	G5	G4	not ranked	S1
Stygobromus ozarkensis	G4	no change	S1	S3
Synurella bifurca	not ranked	G4	not ranked	S3

Allen, C. and C. Bitting, C. Blevins,C. Brickey, J. Briggler, A. and C. Brown, H. Bryant, E. Corfey, M. Covington, M. Evans, D. Fenolio, R. Ginsburg, J. Gunter, S. Hensley, R. Honebrink, S. Longing, S. McGinnis, W. Meurer, L. Moritz, R. Nilius, M. Oliver, R. Redman, D. Renko, H. Robison, M. Ross, M. Schram, R. Schroeder, K. Shirley, T. Snell, D. Taylor, M. Taylor, K. Tinkle, S. Todd, B. and D. Wagner, S. Wallace, L. Willis, N. and J. Youngsteadt, and T. Zawislak Furthermore, two caving societies–Boston Mountain Grotto of the National Speleological Society and the Arkansas Association for Cave Studies–provided assistance in locating caves and conducting safe caving trips.

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Abnormalities in the Ozark Hellbender (*Cryptobranchus alleganiensis bishopi*) in Arkansas: A Comparison between Two Rivers with a Historical Perspective

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Abstract

We documented abnormalities of Ozark hellbender (*Cryptobranchus alleganiensis bishopi*) populations in the Eleven Point River (Randolph County, Arkansas) and the Spring River (Fulton County, Arkansas) as part of ongoing monitoring efforts in this species. We found abnormalities in 90% (9 of 10) and 40% (36 of 97) of hellbenders in the Spring River and Eleven Point River, respectively, during the 2003-2004 field seasons. Most abnormalities found in Eleven Point hellbenders were generally less invasive and seemed to be more intrinsic to the species' natural history (i.e., vicissitudes of living), whereas those found in Spring River hellbenders were gross morphological aberrations. We compared the type and rate of observed abnormalities with those found in museum vouchers collected from the Spring River between 1970 and 1975. Abnormalities were found in 12.5% of the museum specimens from our Spring River localities. This rate is much higher than previously reported for hellbenders. The increase in the abnormality rate appears to be concurrent with the documented population decline observed in the Spring River. Our study illustrates an increasing trend of hellbenders exhibiting unusual morphological problems (e.g., epidermal papillomas, extreme abrasions/lacerations, fungal infections, etc.) and also stresses the need for inclusion of abnormalities observed in field data. The causes of hellbender abnormalities remain speculative; however, plausible explanations may be related to intraspecific interactions, anthropogenic interactions with the microhabitat, viral infections, non-point/point source pollution, and the preponderance of older individuals. These findings emphasize the need for a proactive conservation effort within this species.

Introduction

The Ozark hellbender (*Cryptobranchus alleganiensis bishopi*) is a large, permanently aquatic salamander that is endemic to five south flowing rivers in Missouri and northern Arkansas (Firschein, 1951). Two rivers in Arkansas known to have populations of hellbenders are the Spring River (Nickerson and Mays, 1973; Peterson, 1985) and Eleven Point River (Trauth et al., 1993). Since the early 1980's, Ozark hellbender populations have undergone decline throughout the entire range (Trauth et al., 1992; Wheeler et al., 2003). In Arkansas, the Spring River population has had the most drastic decline witnessed over the past 20 years (Trauth et al., 1992; W. Hiler, unpubl. data). Currently, the U.S. Fish and Wildlife Service lists the Ozark hellbender as an Endangered Species Candidate (Federal Register, 2001), and the species is protected from collection, at the state level, in both Arkansas and Missouri.

Population declines in this species are characterized by fewer small individuals (an indication of reduced recruitment–Wheeler et al., 2003), a lower capture rate (Trauth et al., 1992; Wheeler et al., 2003), loss of historic habitats (Trauth et al., 1993), and high abnormality rates (Wheeler et al., 2002) compared to historical data. Potential causes of declines include interactions with non-native (stocked) species, poor land management, reduction in the riparian zone, urban development, heavy human traffic on and through riverine habitat, and over/illegal collection (Bartlett, 1988; Trauth et al., 1992, 1993; Federal Registry, 2001; Wheeler et al., 2003). These causes remain speculative and are especially difficult to quantify for the Spring River due to the drastic depression in current hellbender numbers. We feel it is realistic to examine the differences between a river which harbors a relatively stable population of hellbenders (the Eleven Point) and Spring River whose hellbender population is essentially extirpated. One condition that has yet to be compared between rivers is occurrence of abnormalities.

The objectives of this study were to describe the types of abnormalities and quantify their rates within the Eleven Point and Spring rivers and museum specimens from the Spring River which may provide a historical perspective on abnormality rates.

Materials and Methods

Detailed documentation of abnormalities began in August 2003 and continued through December 2004. Hellbenders (n = 10) examined in this study were collected from three locations in the Spring River (Fulton County),

Arkansas. We found hellbenders ($n = 96$) at 20 locations in the Eleven Point River from the Arkansas/Missouri state line to just north of the Arkansas State Highway 90 bridge. We also examined 47 hellbenders from the Milwaukee Public Museum collected from the Spring River during the early-to-mid 1970s.

Hellbenders were collected using standard rock-flipping techniques, while either scuba or skin diving. The total length (TL), snout-vent length (SVL) to the anterior end of the cloacal opening, mass, and sex were recorded for each individual. An encrypted (AVID® Identification Systems, Inc., Norco, CA) passive integrated transponder (PIT) tag was implanted in the dorsal musculature of the tail immediately posterior to the hind limbs for unique identification. Abnormalities were documented in our field notes, described morphologically based on their gross appearance, and were photographed with a Sony® CD Mavica 5.0 megapixel camera. Abnormalities were then grouped into the following categories: 1) tumors, 2) open wounds, 3) fungal infections, 4) necrotic limbs, 5) missing limbs, 6) digital abnormalities, 7) eye abnormalities, 8) cloacal wounds, and 9) bite marks.

Results

Our examinations revealed 24 abnormalities in 9 of 10 (90%) of the Spring River animals during 2003–2004 sampling periods. Six of 9 exhibited multiple abnormalities. We found 59 abnormalities in 38 of 96 (40%) animals encountered in the Eleven Point River throughout the 2003–2004 field seasons. Only 23% of the hellbenders in Eleven Point River sample exhibited multiple abnormalities. Of the 47 Spring River museum specimens, 6 (12.5%) exhibited abnormalities (one per individual). In the following, we characterize the types of abnormalities observed.

Tumors.– Neoplasms included epidermal papillomas and tumor-like, small white nodules with diameters of ca. 2-3 mm (see Trauth et al., 2002; Fig. 1A). Tumors were documented in 3 Spring River hellbenders (12.5% of total abnormalities [= TA]), 1 Eleven Point animal (1.5% of TA), and 1 Spring River museum specimen (16.5% of TA). A Spring River female captured below the Arkansas State Highway 63 bridge contained multiple epidermal papillomas and subsequently died during transport to the laboratory for tumor biopsy.

Open Wounds.– These abnormalities included minor-to-severe gashes, abrasions, and lacerations. Six hellbenders with (25.0% of TA) sores, lesions, and lacerations were documented in the Spring River, one (1.5% of TA) in the Eleven Point River, and 2 (33.3% of TA) identified in the museum specimens. One individual captured below Dam 3 exhibited an enlarged ovoid ulcer on the lower jaw ca. 20 mm in diameter (Fig. 1B). Another animal had multiple, severe gashes which began just above the right shoulder and extended in an anterior to posterior direction on the dorsum (Fig. 1C). These wounds appeared to be the result of a gigging or snagging incident and showed signs of a fungal resistance. The Spring River female with epidermal papillomas also displayed extensive abrasions along the lower jaw which showed no signs of healing (Fig. 1D).

Fungal Infections.– Fungal infections were only found on 1 Spring River hellbender. In addition to its dorsal fungal infection, this individual exhibited an oral infection on the tongue (ca. 20 mm in diameter; Fig. 1E) and another on the palm of the left rear foot (Fig. 1F). This hellbender also had a fingernail clam (Sphaerium sp.) as well as a leach (Fig. 1F) attached to the left rear limb. Fungal infections comprised 12.5% of the TA observed in the Spring River.

Necrotic Limb(s).– Necrotic limbs were characterized by "worn palms" exhibiting exposed musculature and, in some cases, exposed bone (see Wheeler et al., 2002). Necrotic limbs comprised 20.8% of TA documented in the Spring River and 13.5% of those in the Eleven Point River. No museum specimens exhibited this abnormality. Typically, the epidermis surrounding the open flesh appeared to be dead and peeling away from the wound. Protruding bone from flesh was less common than simple bone exposure at the center of the necrotic limb.

Missing Limb(s).– Missing limbs (see Wheeler et al., 2002) were distinguished by the absence of all tarsal and/or carpal regions. Missing limbs represented 20.8% of all abnormalities in the Spring River and 17.0% in the Eleven Point River. No cases museum specimens had missing limbs. In some cases, limbs had healed and epidermal tissue covered the entire appendage. In several instances remnants of digits remained attached to the limb. Individuals missing multiple limbs were not as common as those missing 1 limb.

Digital Abnormalities.– Missing, fused, or super-numerary digits (see Wheeler et al., 2002) were the most common abnormalities found in Eleven Point hellbenders. Digital abnormalities represented 12.5% of TA in the Spring River, 51.0% in the Eleven Point River, and 33.3% in the museum specimens. Typically, there was no sign of amputation of the digit, only the physical absence. Fused digits were 2 digits fused by epidermal tissue and lack any of open wound.

Eye Abnormalities.– Eye abnormalities were fairly uncommon and only documented in 2 (3.4% of TA) Eleven Point River hellbenders. Both occurrences were characterized by the presence of a small opaque piece of tissue protruding from an eye.

Cloacal Wounds.– One Eleven Point River male hellbender was missing a cloacal lip (1.5% of TA), and a moderate amount of scarring overlay the afflicted area. The animal was captured on 11 September, 2004, just before the breeding season and the intact lip was swollen, typical of reproductively active males. This male and other males

were not leaking milt. This type of wound has been observed in other individuals (B. A. Wheeler, unpubl. data).

Bite Marks.—Bite marks were characterized by open wounds or scarring patterns matching the dentition of another hellbender (Fig. 2). These wounds appeared on various parts of the body. We captured several individuals with wounds on appendages, apparently caused by a slicing action from the teeth of another hellbender (Fig. 2E and 2F). Bite marks were not documented in any Spring River hellbenders; however, they comprised 10% of TA in the Eleven Point River and 16.5% in museum specimens.

Discussion

Physical abnormalities in hellbender populations are known throughout their range (Nickerson and Mays, 1973; Pfingsten, 1990), but have received little attention in the literature. Pfingsten (1990) was the first to quantify abnormality rates within hellbender populations and reported abnormality rates which exceeded those expected to occur naturally (Johnson et al., 1999; Kaiser, 1999). Wheeler et al. (2002) documented an 8% abnormality rate throughout 12 years of sampling Ozark hellbenders in three rivers. They noted that no consistent effort was made to record every abnormality observed in the field, and they also did not compare different river systems. Our findings indicate the relatively low rate reported by Wheeler et al. (2002) is not an accurate assessment of the actual condition within individual populations. This illustrates the necessity of acquiring detailed field data including observations of injuries and abnormalities.

Most abnormalities found in Eleven Point River hellbenders were generally less conspicuous and appeared to be more related to the species' natural history, whereas those found in Spring River hellbenders were gross morphological aberrations. Over one half (51.0%) of the abnormalities in the Eleven Point River were digital abnormalities, whereas only 12.5% with this type of abnormality were observed in the Spring River. The proportion of hellbenders missing limbs was similar, 17.0% versus 20.8%, in the Eleven Point River and the Spring River, respectively. Numerous aspects of a hellbender's life history could make them susceptible to these types of injuries. Through time, older animals in the Spring River might have a greater chance to accumulate digital injuries which can be masked by missing limbs.

When comparing the number of abnormalities per individual, Spring River rates are much greater than those in the Eleven Point River. In the Eleven Point River, the multiple abnormalities primarily consisted of digital injuries and missing limbs, whereas the Spring River abnormalities were a mixture of all types. The chance of acquiring an abnormality probably increases through time. As age increases there may be decreases in immune efficiency which in turn may leave individuals more susceptible to viral (i.e., epidermal papillomas) and fungal infections.

All fresh bite marks were observed during the fall (reproductive season) on individuals from Eleven Point River animals. Pfingsten (1990) also documented fresh bite marks only during August and September (the reproductive season in Ohio). We concur with Wheeler et al. (2002) and Pfingsten (1990) in suggesting that intraspecific aggression may be the cause of limb injuries. This behavior implies territoriality and can be attributed to the establishment of nesting cavity by males (Nickerson and Mays, 1973). We, therefore, suggest that the presence of bite marks on animals in the Eleven Point River is indicative of reproductive behavior.

The bite marks present in the Eleven Point hellbenders were not exhibited on present day Spring River hellbenders, and this may indicate a decrease in antagonistic behavior among individuals. Only 1 hellbender examined from Spring River museum animals had scar tissue from bites. Habitat loss or fragmentation, a situation now present in the Spring River, could isolate individuals and reduce interactions among hellbenders. Another possibility could be low population numbers which should reduce the number of territoriality interactions and perhaps their intensity.

To further understand the implications of the current abnormality rates, we gathered historic data from museum records as well as raw field data not presented in Nickerson and Mays (1973). Nickerson (pers. comm.) found a 2.3% abnormality rate in hellbenders (n = 479) from the North Fork of the White River, Missouri, while making an effort to record each abnormality. If we assume that abnormalities occur equally throughout and across all populations, then we can further assume that the 2.3% found by Nickerson would be the expected rate found in a healthy population. Museum specimens collected from the Spring River prior to 1975 (presumably before the current population decline) showed a 12.5% abnormality rate. Today, the rate for the Spring River is 90%. This observed increase appears to be concurrent with the documented population decline observed in the Spring River. We can, therefore, postulate that the elevated rate in museum specimens is indicative of the beginning stage of the Spring River population decline as mentioned by Trauth et al. (1992). At present, the Eleven Point River has a 40% abnormality rate and may already be in jeopardy of undergoing a similar decline.

The severe nature of the abnormalities observed in the Spring River may be cause for concern. For example, the open wounds shown in Fig. 1B and D have no known causes and appear to be life threatening. The tumorous animal reported by Trauth et al. (2002) was only the second reported occurrence found within Cryptobranchus. We observed an additional 5 animals with tumors in our study.

Historically, the Spring River has received the most

publicity and has been recognized as a unique river system in Arkansas inhabited by hellbenders (Nickerson and Mays, 1973; Peterson, 1985; Peterson et al., 1988, 1989a, 1989b; Trauth et al., 1992; Wheeler et al., 2002). The river itself, and its tributaries, have been designated as Extraordinary Resource Waters by the Arkansas Pollution Control and Ecology Commission (APCEC, 2004), indicating that the drainage is an invaluable resource for recreational activities as well as science. The Spring River has also been designated as an ecologically sensitive water body by APCEC, indicating that the river harbors rare, threatened, endangered, or endemic species. These features as well as numerous other unique characters make this river an important ecological asset for Arkansas.

Conclusions

The specific causes of hellbender abnormalities remain speculative; still, we feel that there are several factors which may influence these abnormality rates. First, we contend that human interactions may disrupt hellbender microhabit and indirectly lead to some abnormalities or even mortality. These activities include gigging, snagging, and wade fishing. Secondly, intraspecific interactions such as antagonistic behavior, including biting, undoubtedly lead to appendage aberrations. It is also plausible that the most severe abnormalities would be most conspicuous in older individuals and would be more frequently observed or expressed in a greater percentage in senescent populations similar to those in the Spring River. We realize that there are other factors that may cause abnormalities and assume that most abnormalities are not directly influencing the observed population declines. However, increasing abnormalities are occurring concurrently with these declines. Hellbenders live in many streams which have substantial human activity. The very characteristics that make these river systems important recreationally are the same features that have been neglected ecologically. This disregard in the Spring River's ecological health is evident in the current status of the Ozark hellbender population.

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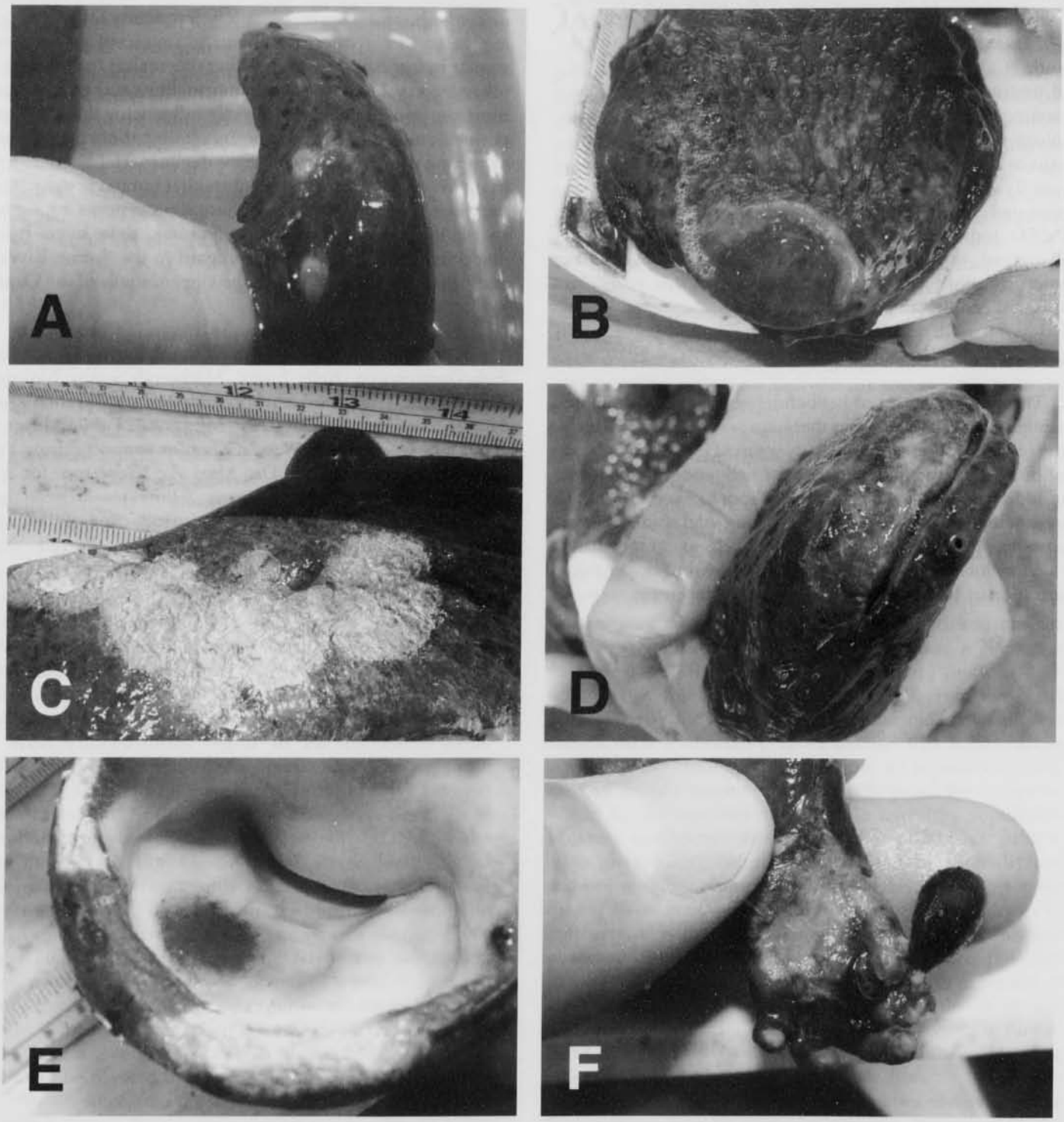

Fig. 1. Abnormalities observed in the Ozark hellbender from the Spring River (Fulton County), Arkansas, during 2003-2004 sampling period. A. Epidermal nodules on lateral surface of tail. B. Highly vascularized, circular cyst on the lower jaw. C. Large dorsal wounds posterior to head with massive fungal infection. D. Extensive abrasion around mental symphysis. E. Oral fungal infection. F. Left hind limb with fungal infection, leech attached to palm, and fingernail clam (*Sphaerium* sp.) attached to digit.

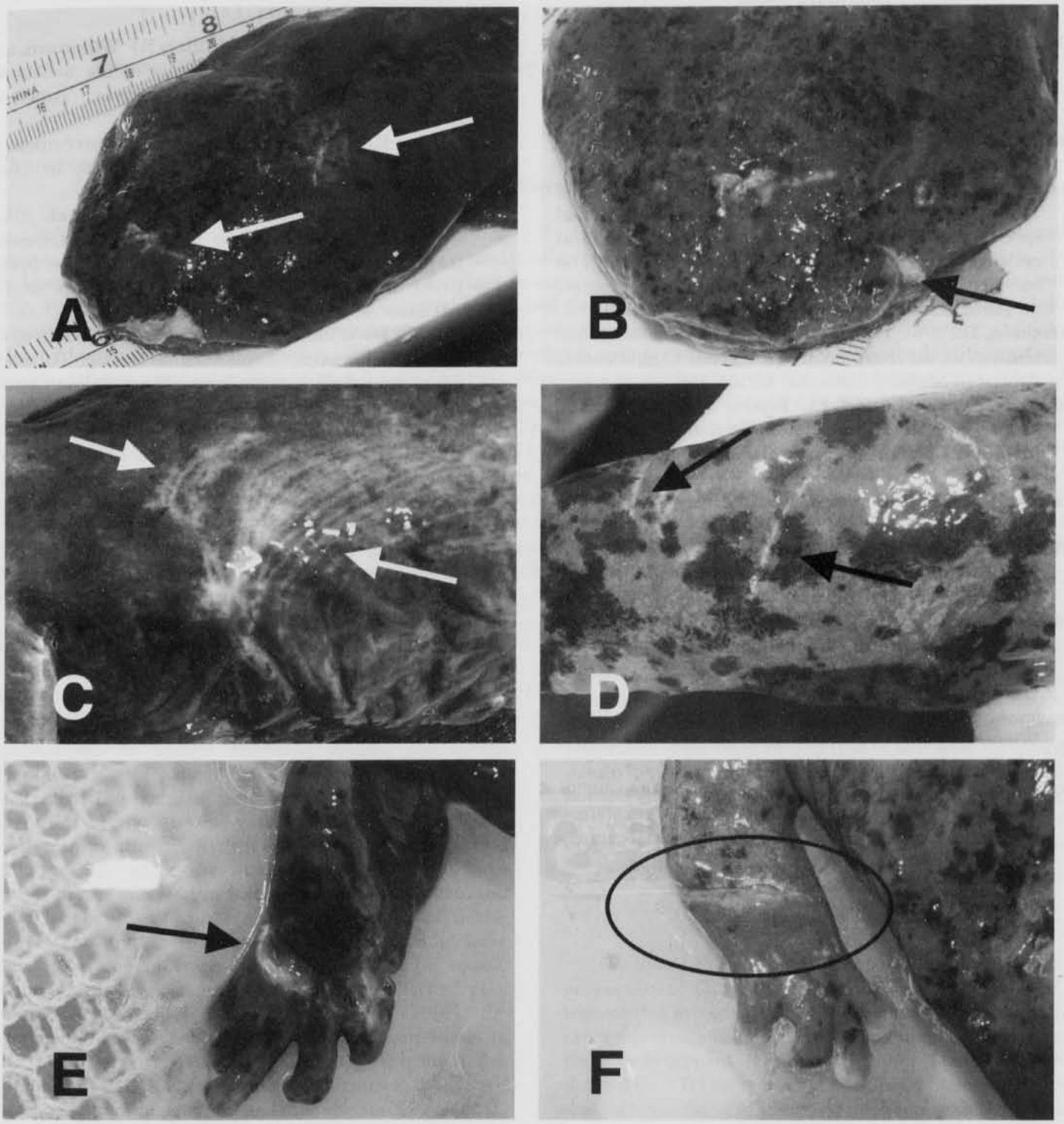

Fig. 2. Bite marks resulting from intraspecific aggression in the Ozark hellbender (Eleven Point River, Randolph County, Arkansas) during the 2003-2004 sampling period. A and B. Arrows point to wounds on head and snout. C and D. Arrows point to a series of semicircular scars on separate individuals, each caused by a single encounter with another hellbender (C, on abdomen; D, on tail). E and F. Arrow points to wound encircling entire limb; circled area in F highlights linear wound across limb.

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Quantification and Prediction of Stream Dryness in the Interior Highlands

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Abstract

Although ecological studies have noted streams drying in the Interior Highlands, published measurements of streambed dryness are lacking. Clearly, stream drying has the potential to affect benthic macroinvertebrate and fish communities. In 2003, we initiated an assessment of streambed dryness for three streams in the Ouachita Mountains representative of the Central Hills, Ridges, and Valleys. In the following summer, we applied the approach to 15 similar size watersheds in three distinct ecoregions of the Interior Highlands: Ouachita Mountains–Athens Plateau, Ozark Highlands–Springfield Plateau, and Lower Boston Mountains. Repeated dryness measurements were recorded in each stream and correlated to nearby USGS stream gage records. Dryness reached as high as 86% for the Ouachita Mountains in 2003; whereas, flow was continuous in 2004. One stream in the Ozark Highlands dried completely in 2004, and dryness reached 84% in the Boston Mountains. Percent dry streambed was negatively correlated (Spearman rank) to discharge for the Ouachita Mountains in 2003 and the Boston Mountains in 2004 ($r_s = -0.94$ and -0.60, respectively; $p \leq 0.01$). Lowest monthly mean daily discharge, corrected for watershed area, differed among ecoregions for May through October in 2004 (highest discharge in the Ouachita Mountains, $p \leq 0.05$, Tukey-Kramer). Maximum dryness during these months was significantly lower for the Ouachita Mountains than the Boston Mountains and Ozark Highlands. Thus, discernable patterns of stream dryness exist among the different ecoregions of the Interior Highlands. Aquatic ecologists and resource managers in these ecoregions could employ such measures to further understand habitat limitations associated with these stream systems.

Introduction

Stream drying is a potentially important ecological phenomenon in Interior Highland streams due to the temporary loss of habitat that occurs as streams become desiccated. The extent of continuous surface flow reflects an interaction between several variables, such as precipitation, evapotranspiration, hydrogeologic pathways, and anthropogenic water use within a watershed. Extensive stream drying can lead to loss or isolation of habitats and possible mortality of aquatic macroinvertebrates and fishes.

Recent studies in the Interior Highlands have investigated the consequences of stream drying on aquatic communities. Investigations considered the effect of pool connectivity on fish assemblages (Taylor, 1997), the interaction between fish and aquatic macroinvertebrates in intermittent streams (Williams et al., 2003), variation in fish assemblages in drying stream pools (Magoulick, 2000), and metapopulation dynamics of endemic species (Gagen et al., 1998). While these investigators provided detailed assessments of community characteristics, few physical measurements were provided to characterize the extent of dryness in the context of the stream network.

Understanding key ecosystem processes that cause and maintain the association of habitat patches is important to management and understanding of stream fish populations (Schlosser and Angermeier, 1995). Thus, systematically assessing the extent of stream drying should contribute to understanding ecological processes within the Interior Highlands. The objectives of this study were to quantify stream drying within the Interior Highlands, to characterize possible drying patterns relative to major ecoregions within the Interior Highlands, and to predict the degree of stream drying from discharge data available from nearby United States Geological Survey (USGS) stream gages.

Materials and Methods

We focused dryness measurements in the major upland ecoregions of the state. Ecoregions have been defined as regions that are relatively similar with respect to ecological processes involving interrelationships among organisms and their environment (Omernick, 1995). Omernick (1987) defined level III ecoregions of the United States on the basis that ecosystems and their components exhibit regional patterns that are reflected in spatially variable associations of underlying factors including mineral availability, climate, vegetation, and physiography. Woods et al. (2004) also defined subecoregions (level IV) in Arkansas based on landuse, wildlife, fish, and hydrology.

We piloted methods for quantification of streambed dryness in the summer of 2003 and further applied the

methods at a larger scale in the summer of 2004. We measured stream dryness in 2003 in three streams within the Ouachita Mountains (level III)–Central Hills, Ridges, and Valleys (level IV) ecoregion (Woods et al., 2004). These three streams drained relatively small watersheds (220 to 800 ha) within the Ouachita River drainage upstream of Lake Ouachita. In 2004, we measured stream dryness in 15 streams of similar size watersheds in three different ecoregions of the Interior Highlands. A study area in each ecoregion consisted of three streams at the 2,800 ha watershed size and two streams at the 5,600 ha watershed size. The five streams in the Ouachita Mountains (level III)–Athens Plateau (level IV) were tributaries of the Cossatot River, and were expected to remain perennial throughout the year (Hines, 1975). We selected five streams in the Ozark Highlands (level III)–Salem Plateau (level IV); three were tributaries of North Sylamore Creek (two 2,800 ha and one 5,600 ha), whereas the other two were within adjacent Livingston Creek drainage (one 2,800 ha and one 5,600 ha). We expected streams in this ecoregion to have an intermediate level of baseflow in the summer (Hines, 1975). We also measured five streams in the Boston Mountains (level III)–Lower Boston Mountains (level IV) ecoregion. These were tributaries to the Illinois Bayou, which was known to have little baseflow in the summer (Hines, 1975).

The method of measurement of stream dryness was the same for both years of the study. For a 2-km study reach beginning at the watershed boundary, one person walked upstream with a hip-chain recording the length of each wet or dry section. We considered a section of streambed dry where no surface water was visible across the width of the streambed. In 2003, dryness was measured once in June and twice in July in all three creeks. A fourth measurement was recorded for one creek (Rocky Creek) in September. In 2004, all creeks were sampled from June through October with a minimum of four samples per stream.

All study sites in 2003 and 2004 had nearby USGS stream gages on larger tributaries. Distance from beginning of watershed boundary to gages ranged from 5 to 41 km. We used regression analysis to search for relationships between our dryness measurements and published discharge data from these stream gages. Discharge data from the USGS gages were available online for 2002 to 2004 at (http://water.usgs.gov/pubs/wdr/#AR) and for previous years at (http://nwis.waterdata.usgs.gov/usa/nwis/discharge). We expressed stream discharge from these reference gages as $L/s{\cdot}ha^{-1}$ to facilitate comparison among USGS watersheds of different sizes.

We used the Kruskal-Wallis test to evaluate differences in percent streambed dry and discharge at reference gages between ecoregions in 2004. We made multiple comparisons of ecoregions by applying the Tukey-Kramer test to rank-transformed data (Conover and Iman, 1981). We selected the lowest mean daily discharge ($L/s{\cdot}ha^{-1}$) for each month from June through October as an index of low discharge. We compared the low discharge index among ecoregions in the same manner as for the dryness index.

We used correlation and regression analyses to examine relationships between percent dry streambed and low discharges at reference gages. To determine if there was a relationship between percent dry streambed and reference discharge, we used Spearman rank correlation for each stream in each ecoregion. We also used linear regression analysis to predict percent stream dryness from reference discharge. We transformed discharge to its reciprocal ($1/L{\cdot}s{\cdot}ha^{-1}$) to produce a more linear relationship. These empirical regression equations were used to estimate percent dry streambed during past years. For ecoregion comparisons we also arbitrarily selected the number of days each creek was at least 25 percent dry as a criterion likely to have ecological relevance (Q @ $\geq$ 25%). To determine the Q @ $\geq$ 25% dry, we used the linear equation from the regression analyses. All statistical analyses were performed using Number Cruncher Statistical Software (Hintze, 1995).

Results and Discussion

Stream dryness and discharge at reference gages varied among ecoregions. Streamflow was continuous along the lengths of Ouachita Mountains-Central Hills, Ridges, and Valleys streams when the study began in June 2003; however, dryness reached as high as 86% by September (Fig. 1). In 2004, streams of the Ouachita Mountains-Athens Plateau flowed continuously whereas, dry reaches appeared in streams of the Lower Boston Mountains and the Ozark Highlands-Springfield Plateau. All but one stream in the Lower Boston Mountains had continuous surface flow at the start of the 2004 study, and each stream dried gradually throughout the summer becoming up to 84% dry (Fig. 1). Dryness ranged from 0 to 100% for streams draining the karst watersheds of the Ozark Highlands–Springfield Plateau; however, the pattern of wet and dry reaches was well established at the beginning of the study and fluctuated little during the remainder of the study (Fig. 1). For similar size watersheds, the maximum percentage of dry streambed was significantly different among ecoregions with the Lower Boston Mountains (driest), Ouachita Mountains-Athens Plateau (wettest), and the Ozark Highlands-Springfield Plateau (intermediate; $p < 0.05$). The minimum mean daily discharge at reference gages also varied significantly across ecoregions and followed the pattern indicated by dryness percentages ($p < 0.05$, using a single value per month calculated as $L/s{\cdot}ha^{-1}$).

Stream dryness was related to discharge at reference gages ($L/s{\cdot}ha^{-1}$) in 2003 and 2004. Percent dry streambed for 2003 in the Ouachita Mountains–Central Hills, Ridges, and Valleys ecoregion was negatively correlated to minimum mean daily discharge at the

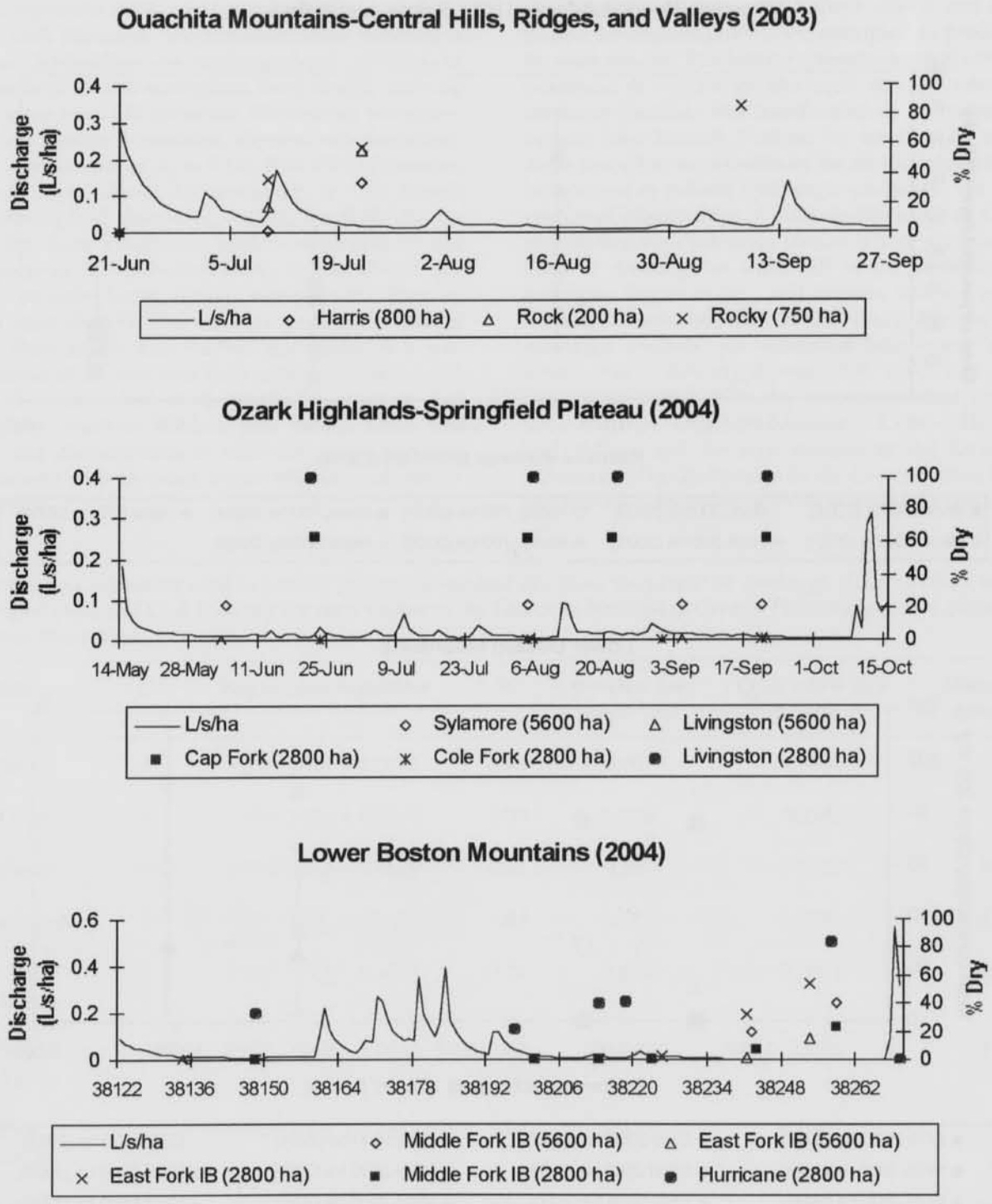
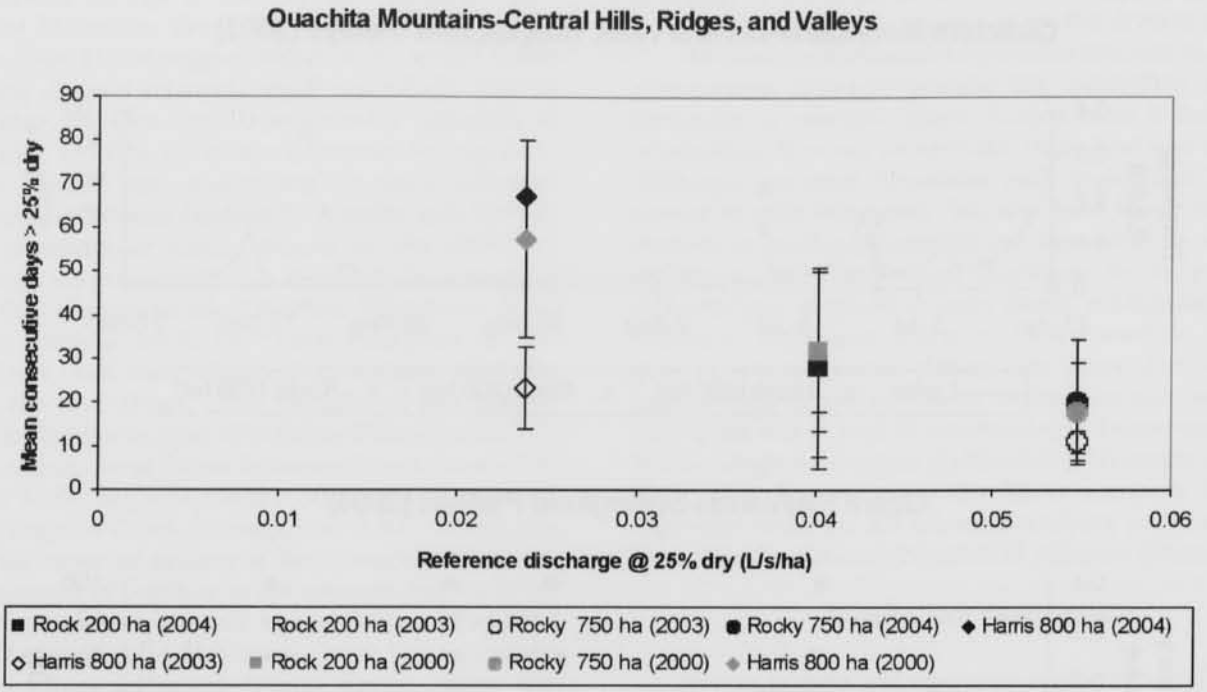


Fig. 1. Percent dry (symbols) and discharge (solid line) over time in ecoregions where dryness was measured. Dryness was not observed in the Ouachita Mountians-Athens Plateau in 2004.



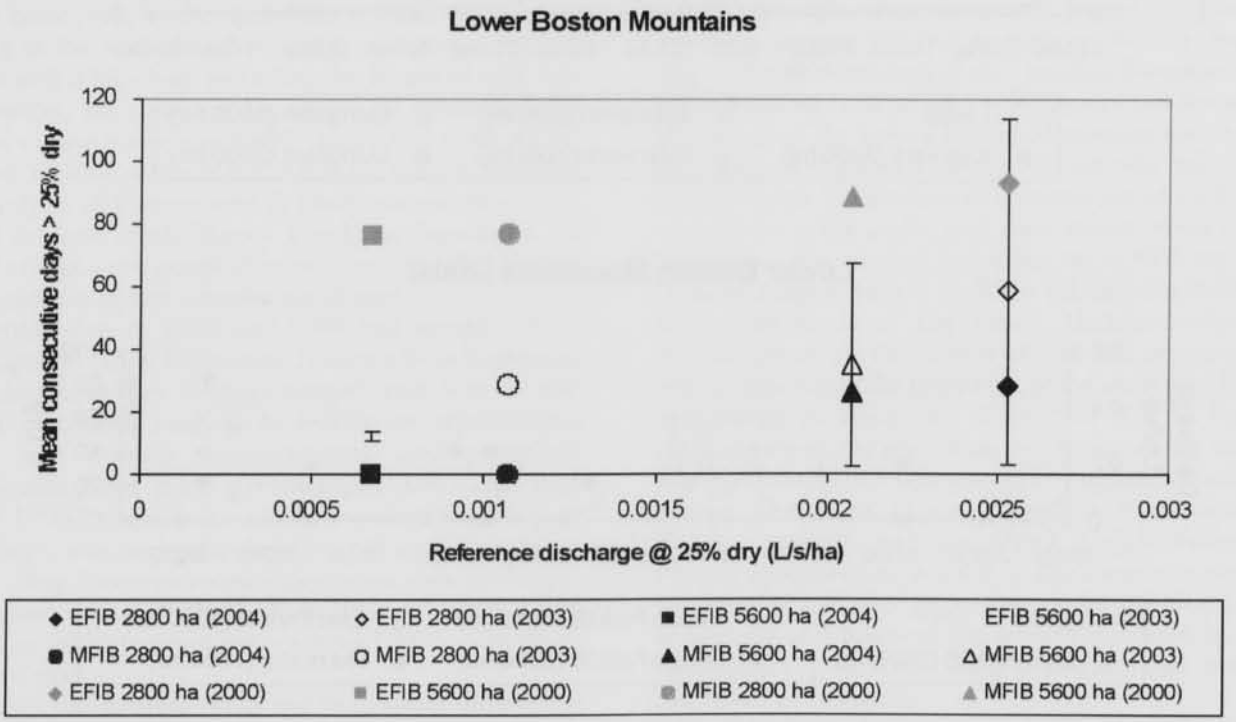


Fig. 2. Mean consecutive days 25 % dry in 2000 (gray), 2003 (hollow), and 2004 (black). Error bars are ±1SE. (Hurricane Creek (Lower Boston Mountains) is not pictured, but Q @ 25% dry is given in Table 1).

reference gage (r_s = -0.94, $P < 0.01$). In contrast the Athens Plateau portion of the Ouachita Mountains ecoregion showed no dryness in 2004, which precluded any search for correlation with discharge. We attributed these differences in summer streamflow to hydrogeologic differences between the level IV sub-ecoregions, even though both are within the same level III (Ouachita Mountains) ecoregion. In the Lower Boston Mountains, dryness was negatively correlated with discharge (r_s = -0.60, $P < 0.01$). However, dryness was not related to discharge in the Ozark Highlands-Springfield Plateau (r_s = -0.01, P = 0.98). In this ecoregion the karst conditions likely contributed to the observed patterns of headwaters being almost always dry and larger streams being almost always wet. That is, portions of these streams flow underground except during stormflow. Thus, of the four studied ecoregions, flow was relatively constant for two (one had continuous (perennial) flow and the other had both perennial reaches and completely dry reaches). Surface flow in the other two ecoregions was discontinuous in time and space and was highly correlated with an index of low discharge at nearby reference gages.

Based on the correlation between % dry streambed and low discharge at reference gages in the Ouachita Mountains-Central Hills, Ridges, and Valleys (2003) and the Lower Boston Mountains (2004), we attempted to predict dryness for each stream. The linear regression analyses resulted in a minimum $R^2 = 0.74$ for the eight streams when the low discharge variable was transformed to its reciprocal. Low sample sizes limited P-values for significance of slope in some cases, but we considered the empirical relationships to be relevant to natural hydrologic processes. We used these empirical relationships to estimate discharge at 25% dry (Q @ 25% dry) for each study stream. There was not a specific level of discharge at which all of the streams within an ecoregion began to dry, and dryness was not consistently related to watershed size in this study. By searching past discharge records, we estimated how many days each stream was ≥ 25% dry during 2000, 2003, and 2004. The amount of time ≥ 25% dry was determined for all three streams in the Ouachita Mountains-Central Hills, Ridges, and Valleys and for five streams in the Lower Boston Mountains (Fig. 2). Streams in the Lower Boston Mountains reached 25% dry at lower levels of discharge than streams in

Table 1. Regression equations used to predict percent streambed dry from reciprocal of discharge (1/Q, L/s·ha^{-1}) at reference gage, watershed area, and Q @ 25% dry for each stream in the Ouachita Mountains-Central Hills, Ridges, and Valleys and the Lower Boston Mountains.

Stream	N	Regression equation	R^2	p-value for slope test	Q @ ≥25% dry (L/s·ha^{-1})	Watershed Area (ha)
Rock Creek	3	1.176 (1/Q) – 4.2679	0.99	0.007	0.0401	220
Rocky Creek	4	1.393 (1/Q) + 0.4536	0.90	0.052	0.0547	760
Harris Creek	3	0.759 (1/Q) – 7.0145	0.91	0.197	0.0237	800
Hurricane Creek	6	0.111 (1/Q) + 19.051	0.83	0.011	0.0187	2770
Middle Fork Illinois Bayou	6	0.029 (1/Q) – 2.4712	0.74	0.028	0.0011	2920
East Fork Illinois Bayou	6	0.077 (1/Q) – 5.2036	0.99	0.001	0.0025	3150
East Fork Illinois Bayou	4	0.018 (1/Q) – 2.2424	0.78	0.123	0.0007	5540
Middle Fork Illinois Bayou	4	0.061 (1/Q) – 4.317	0.91	0.047	0.0021	5630

the Ouachita Mountains-Central Hills, Ridges, and Valleys (Table 1).

Stream drying could be impacted by land management practices that alter the hydrologic regime. Miller et al. (1988) reported higher, more frequent, and extended stormflows in the first two years after clearcutting and selection cutting of forested watersheds in the Ouachita Mountains. Where annual water yield remains similar, land management practices that lead to higher stormflow should also contribute to decreased baseflow and consequently increased extent of dryness in summer. Conversely, increased water yield associated with baseflow could decrease stream dryness.

Measurement of dryness seems to be a valid, but often overlooked, aspect of habitat quality associated with streams of the Interior Highlands and perhaps elsewhere. The approach described in this study is simple to implement, and may be relevant to any stream with seasonally discontinuous surface flow, especially when nearby historical discharge data are readily available. However, these methods may not be applicable in karst ecoregions as they may not produce linear relationships between stream dryness and discharge. Prediction of stream dryness showed promise as an approach to determine if dryness in a previous year(s) might be associated with biological variation, such as year class strength or past estimates of abundance or diversity. More extensive studies of seasonally discontinuous surface flow may increase confidence in measures of stream dryness as predictive tools. Aquatic ecologists and resource managers in these ecoregions may benefit from considering such measures to further understand habitat limitations.

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Bedrock Geology of Sonora Quadrangle, Washington and Benton Counties, Arkansas

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Abstract

A digital geologic map of Sonora quadrangle was produced at 1:24,000 scale using the geographic information system (GIS) software MapInfo. The geology of Sonora quadrangle consists of sedimentary rocks from the Ordovician, Devonian, Mississippian, and Pennsylvanian Systems. The Cotter, Powell, and Everton formations represent the Ordovician System. The Clifty and Chattanooga formations represent the Devonian System. The St. Joe Limestone, Boone, Batesville, and Fayetteville formations represent the Mississippian System. The Hale formation represents the Pennsylvanian System. The St. Joe Limestone crops out extensively in Sonora quadrangle and is unconformably overlain by the Boone formation in the southern portion of the quadrangle. This unconformity adds credence to the suggestion that the St. Joe Limestone should be elevated to formation status rather than remain as a member of the Boone formation. The Fayetteville formation consists of the informally named lower Fayetteville Shale, Wedington Sandstone, and informally named upper Fayetteville Shale. The only member of the Hale formation observed in Sonora quadrangle was the Cane Hill member. The two prominent geologic structures in Sonora quadrangle are the White River fault running generally east-west and the Fayetteville fault running generally southwest-northeast. Other subsidiary faults are associated with these primary faults, creating fault zones within the quadrangle. Detailed mapping of stratigraphy and structure in Sonora quadrangle provides new insights into the geologic evolution and sea-level history of the Ozark Plateaus and the southern craton margin during the Paleozoic Era.

Introduction

The Paleozoic geology of the southern Ozark region has attracted worldwide interest because of exposures of the Morrowan Series at the base of the Pennsylvanian System and for the excellent outcrops of fossiliferous strata in proximity to the Mississippian-Pennsylvanian boundary (Frezon and Glick, 1959; Manger and Sutherland, 1984; McFarland, 1998). The geologic history and depositional dynamics of this Paleozoic interval continues to attract the attention of the geologic community as a means of investigating the interplay of global tectonics and global eustasy in the development of continental margin and foreland basin sequences (Houseknecht, 1986; Viele, 1989; Ethington et al., 1989; Thomas, 1989; Viele and Thomas, 1989; Handford and Manger, 1990, 1993; Valek, 1999; Hudson, 2000; Anderson, 2001; Combs, 2001; Cooper, 2001). However, despite continued interest in the Paleozoic stratigraphy of northern Arkansas, no detailed mapping of the geology of Sonora quadrangle has occurred since thesis work undertaken in the early 1960's (Metts, 1961; Cate, 1962; Carr, 1963; Clardy, 1964) at the University of Arkansas and since preparation of the revised Geologic Map of Arkansas by Haley et al. (1976 and 1993).

With the advent of satellite positioning services, advanced digital technologies, and geographic information systems during the last decade, it is now possible to develop highly detailed geologic maps from field data with locations determined using the global positioning system (GPS) and transferred to digital mapping programs. Development of geologic maps in digital formats permits relatively easy manipulation of these data and their export to a variety of software platforms where they can be modified or adapted for many projects. This project represents the first effort to map individual stratigraphic members in Sonora quadrangle using digital technologies.

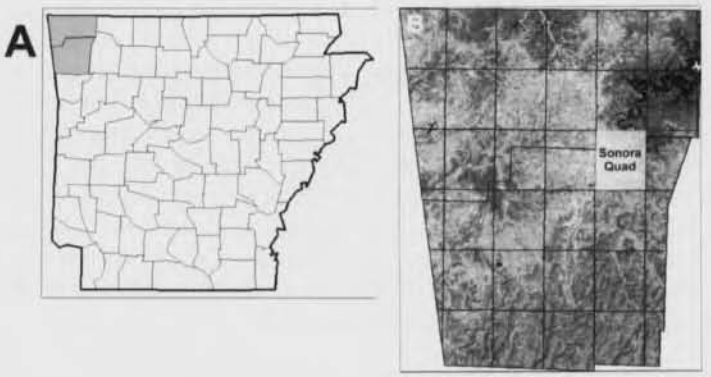

Fig. 1. A) Location map of Arkansas showing Washington and Benton Counties (shaded) and B) Sonora quadrangle in Washington and Benton Counties.

Sonora quadrangle (Fig.1) is located in northeast Washington County and southeast Benton County, Arkansas, and is named for the community of Sonora, which occupies the central portion of the quadrangle (Fig. 1). The quadrangle boundaries are 36°15.0'N 94°07.5'W (northwest), 36°07.5'N 94°07.5.0'W (southwest), 36°15.0'N 94°00.0'W (northeast), and 36°07.5'N 94°00.0'W (southeast). The landscape is a maturely dissected, dendritic drainage system dominated by the White River, which flows north through the quadrangle and into Beaver Lake (Figs. 3, 4). Whereas upland areas throughout the quadrangle are heavily forested, excellent exposures of all lithostratigraphic units through the Hale formation can be observed in ravines associated with Beaver Lake, the White River and its tributaries, roadcuts along highways U.S. 412 and AR 265, and in on-going excavations produced by construction activities in the region.

Washington and Benton counties occupy the boundary of two erosional plateaus formed along the southern portion of the Ozark Dome (Croneis, 1930). The Springfield Plateau is composed of strata deposited during the Ordovician (490-443 Ma BP; Palmer and Geissman, 1999) through Mississippian (354-323 Ma BP; Palmer and Geissman, 1999) periods. The higher Boston Mountains Plateau in the extreme southern portion of Sonora quadrangle is formed of late Mississippian through middle Pennsylvanian strata and is capped by the Pennsylvanian-aged Cane Hill member of the Hale formation (Fig. 2; Stanton, 1993).

The topography of the quadrangle is controlled by a number of stratigraphic units. The Wedington member of the Fayetteville formation is often expressed as an elevated bench on hillsides and caps some hills in the western portion of the quadrangle. Prominent bluffs around the lake and river (ranging to 30 meters high) in Sonora quadrangle are also associated with outcrops of the Everton formation, St. Joe Limestone, and the Boone formation. Finally, sandstone units of the Hale formation form bluffs and cap some hill tops in the southern portion of Sonora quadrangle.

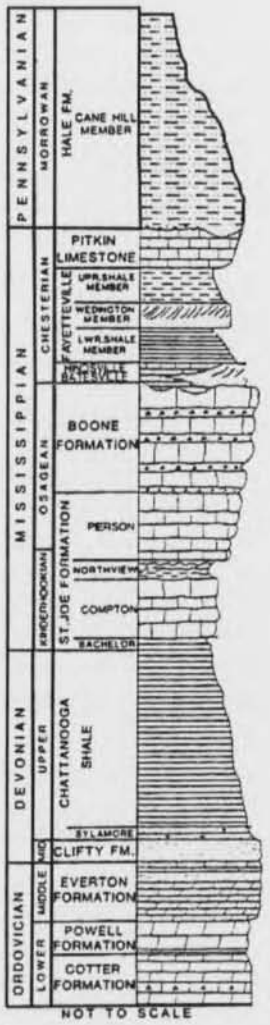

Fig. 2. Generalized stratigraphic column of Sonora quadrangle, Washington and Benton Counties, Arkansas (adapted from Stanton, 1993).

Materials and Methods

Field mapping of Sonora quadrangle was conducted throughout the summer of 2003 accessing various locations from a network of county and state roadways or on foot and from boat around the shoreline of Beaver Lake. Commonly, rock fragments and soil type could be used to determine the bedrock stratigraphy in areas of low relief where outcrops were not discernable.

Locations of outcrop sites for individual stratigraphic members and observed geologic structures were determined using global positioning system (GPS) receivers capable of receiving differential corrections. These receivers typically have horizontal accuracy of approximately 3 meters. For each outcrop or sample location, GPS coordinates were noted in the field notebook, and the location was indicated on the field map. A Garmin Etrex was used to determine elevations. The locations gathered were recorded onto a 1:24,000 topographic map in the field and logged into the field book for later transfer to the MapInfo digital mapping program.

A geographic information system (GIS) is a computer system that records, stores, and analyzes geospatial information. Information regarding field geologic relations was transferred from the field map to a digital raster graphic (DRG) of Sonora quadrangle using a "heads-up" digitizing method that was described in detail in J.T. King et al. (2002), Sullivan and Boss (2002), M.E. King et al. (2001a and b). Using this method, stratigraphic units and geologic structures (e.g. faults) were drawn directly on the computer screen by

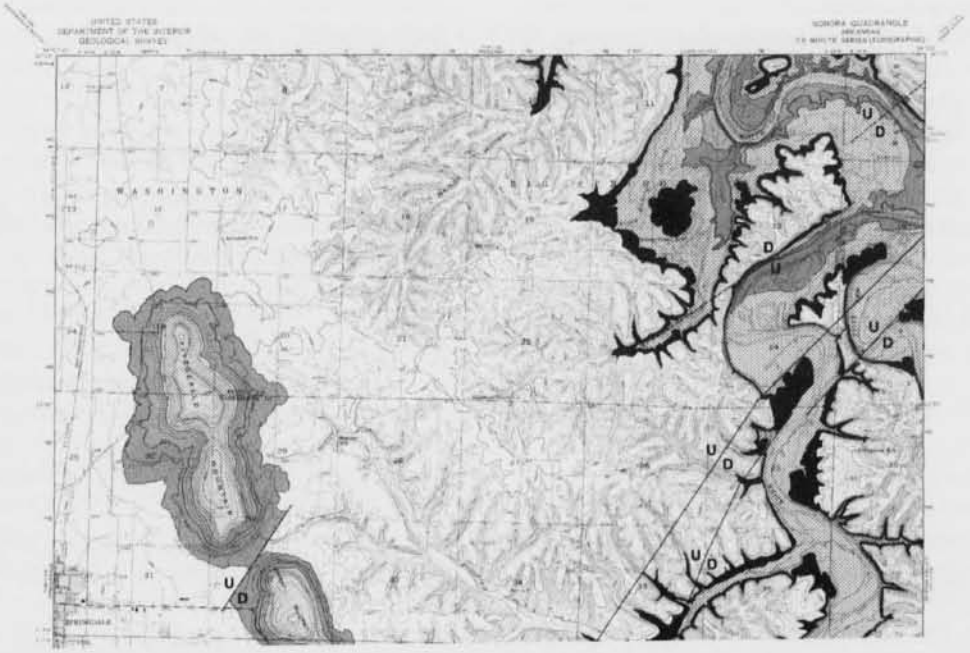

Fig. 3. Map showing bedrock geology of the northern half of Sonora quadrangle digitized onto Sonora quadrangle 7.5-minute digital raster graphic (DRG).

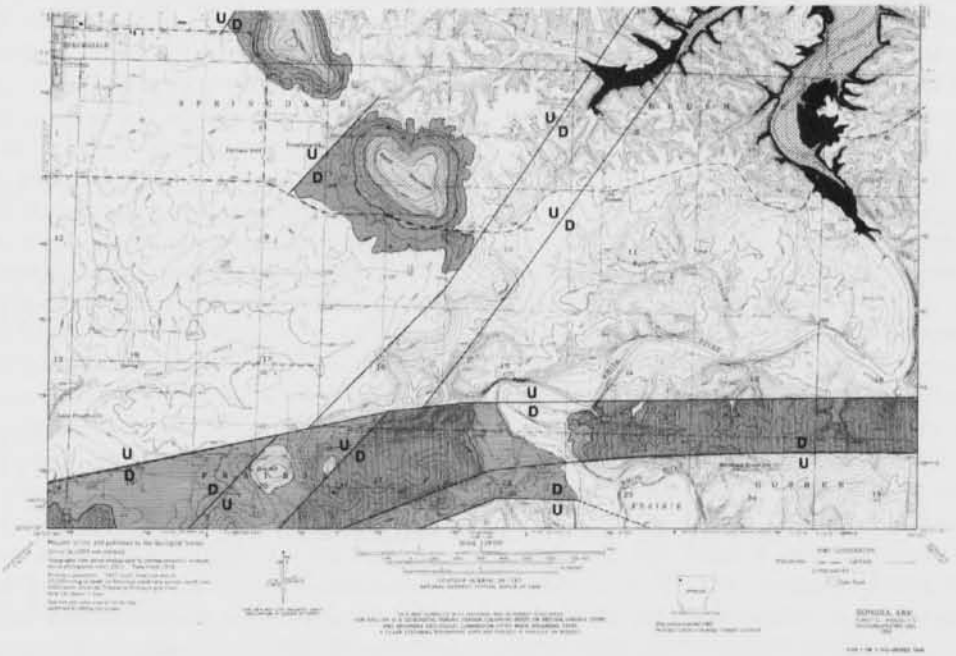

Fig. 4. Map showing bedrock geology of the southern half of Sonora quadrangle digitized onto Sonora quadrangle 7.5-minute digital raster graphic (DRG).

moving the cursor over a DRG of Sonora quadrangle and clicking the mouse button at short intervals to trace contacts onto the displayed topography. Each stratigraphic unit or structural feature was digitized as a separate layer within the GIS such that the display of each layer could be toggled on or off. Once all stratigraphic units and geologic structures were digitized, map layers could be displayed hierarchically to generate the geologic map of the study area (Figs. 3, 4, 5). The final step in preparing the digital geologic map was to convert all data layers to several digital formats to ensure compatibility with popular GIS applications. All data were archived on CD-ROM and are available from the corresponding author upon request.

Results

Sedimentary rocks of the Ordovician Period (490-443 Ma BP; Palmer and Geissman, 1999) through Pennsylvanian Period (323-290 Ma BP; Palmer and Geissman, 1999) are present throughout Sonora quadrangle. Rocks of the Ordovician Period (in ascending order) are the Cotter formation, the Powell formation, and the Kings River member of the Everton formation. Ordovician strata are generally present only along the shoreline of Beaver Lake in the northern portion of the quadrangle (north of the War Eagle Marina and Recreation Area) and on the bottom of the lake along the main axis of the former channel of the White River (Figs. 3, 4).

The Cotter formation in Sonora quadrangle is mostly inundated by Beaver Lake, though exposures of the top of the Cotter formation can be observed when lake level is low east of the Hickory Creek Recreation Area (Fig. 3). When exposed, the Cotter weathers to cobbles of dark gray, blocky chert.

The Powell formation is also poorly exposed in Sonora quadrangle. Outcrops of the Powell formation were observed lying unconformably on the Cotter formation along the northern lakeshore east of the Hickory Creek Recreation Area (Fig. 3). The Powell formation is generally a fine-grained, light-gray to greenish-gray, limy, argillaceous dolostone with thin beds of light green shale (Purdue and Miser, 1916). The Powell formation reaches 65 m (215 feet) thick in its type area. However, within Sonora quadrangle, observed exposures of the Powell formation are less than 3 meters. The top of the Powell formation is unconformable with the overlying Everton formation.

The Everton formation was named for Everton, Arkansas, in Boone County (Purdue, 1907). The Everton formation shows considerable differences in lithologic character across the Ozark region (Suhm, 1970; 1974). It is composed of various mixtures of dolostone, sandstone, and limestone. The formation also has some conglomeratic facies, shale, and chert in limited areas. The limestones are light gray to brownish gray and are generally more or le s dolomitic and sandy. The dolostones are light- to dark-gra / and generally more or less limy and sandy. The Kings Riv r Sandstone is the only representative of the Everto formation within Sonora quadrangle. This member s composed of massive to thinly parallel layers of friabl quartz sandstone. In Sonora quadrangle, the Everto formation varies in thickness from 2 to nearly 12 m. It is be t exposed at the Hickory Creek Recreation Area, along th lakeshore east of Hickory Creek Recreation Area, an around the Pleasant Heights development (Figs. 6A, B). I these areas, the Everton formation forms resistant bluffs and was observed to be filling sinkholes in the underlying Powell and Cotter formations. Exposed cross-sections of Everton formation bluffs also reveals paleochannels (Fig. 6A). Devonian strata present in Sonora quadrangle are (in ascending order) the Clifty formation and Chattanooga Shale (Figs. 3, 4, 5). The Clifty formation rests unconformably on the Everton formation, and excellent exposures of this contact were documented in the vicinity of the Hickory Creek Recreation Area (Fig. 6B). The Clifty formation is often confused with the Sylamore Sandstone member of the Chattanooga Shale (Metts, 1961). However, it is distinguishable from the Sylamore Sandstone in that fresh surfaces are white, saccharoidal quartz sand, whereas fresh surfaces of the Sylamore Sandstone are typically yellow to yellowish brown containing phosphatic pebbles and limonitic concretions (Hall, 1978). McFarland (1998) suggested that the maximum thickness of the Clifty formation was approximately 1 m. However, in Sonora quadrangle, the Clifty formation appears to reach maximum thickness in the Hickory Creek area of 3–4 m.

The Chattanooga formation is a black, fissile, clay shale that weathers into thin flakes. The beds are usually cut by prominent joints creating polygonal blocks upon weathering. In Sonora quadrangle, the basal sandstone member (Sylamore Sandstone; Branner, 1891) of the Chattanooga Shale was not observed. The thickness of the Chattanooga Shale ranged from 3 to approximately 9 m.

An important discovery of this mapping project was documentation of a very fine-to fine-grained, silty sandstone capping the Chattanooga Shale north and northwest of Friendship Creek (Figs. 6C, D). This unnamed sandstone occurred as several layers, each of which was approximately 0.3–0.6 m, and separated the Chattanooga Shale from the overlying Bachelor member of the St. Joe formation (Fig. 6D). In addition, a channel incised into the Chattanooga Shale and backfilled with this sandstone was observed at one location. Elsewhere throughout the quadrangle, this sandstone was not observed, and the Bachelor member lies directly on the Chattanooga Shale. It is not known if this sandstone represents a basal sand unit of

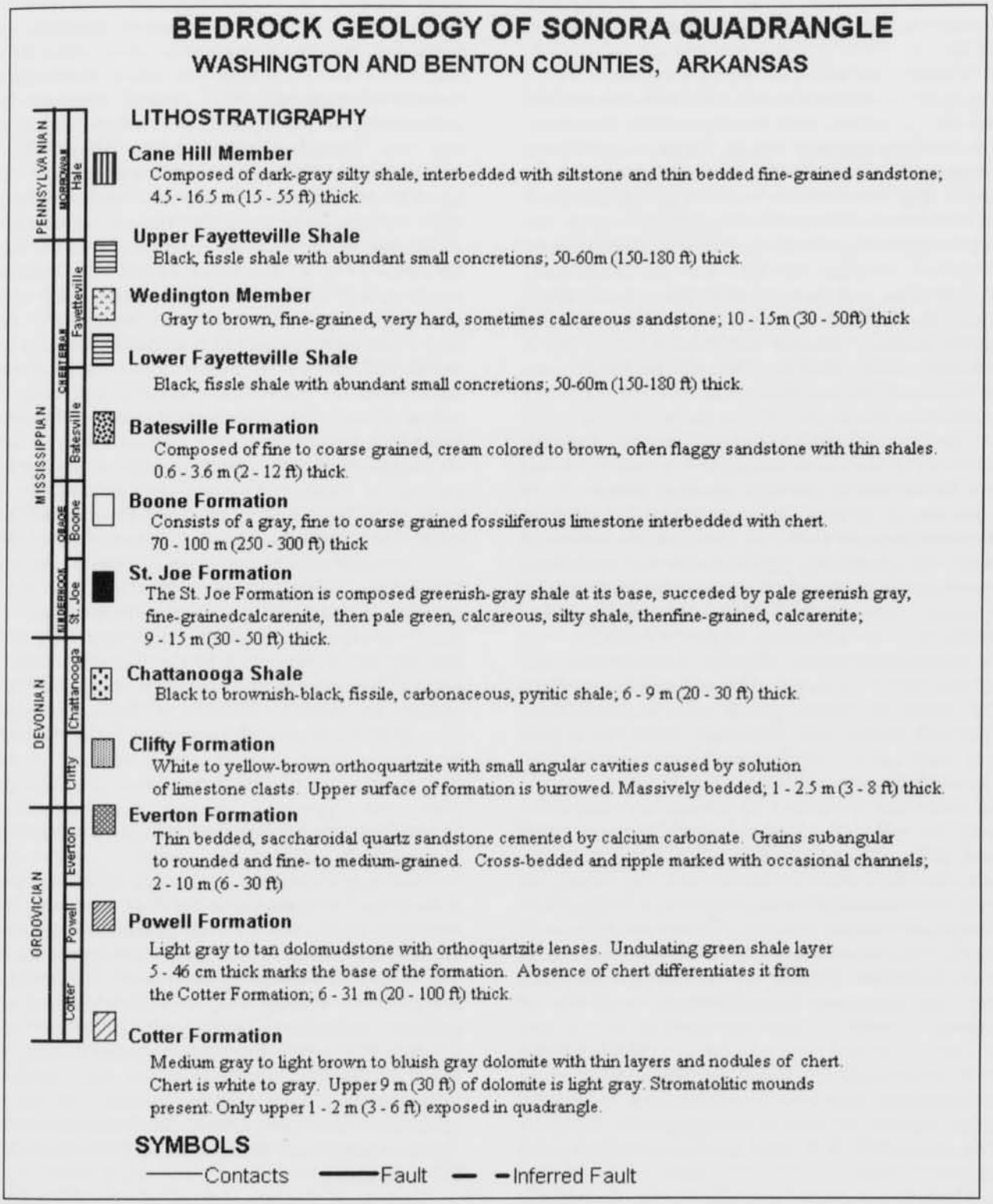

Fig. 5. Legend to accompany geologic map of Sonora quadrangle (Figs. 3, 4).

the Bachelor member. However, its association as the sediment filling a channel incised into the Chattanooga Shale demonstrates the unconformable nature of its contact on the shale.

The Mississippian strata in Sonora quadrangle are (in ascending order) the St. Joe formation, the Boone formation, the Batesville formation, and the Fayetteville formation. Rocks of the Mississippian Period comprise the largest surface exposures throughout the quadrangle (Figs. 3, 4, 5).

The St. Joe formation is the oldest Mississippian stratum. The formation is named for exposures near St. Joe, Arkansas in Searcy County (Hopkins, 1893). The formation is a fine-grained, crinoidal limestone that may occasionally contain some dark gray nodular chert (McFarland, 1998). The base of the St. Joe formation is a greenish-gray shale, the Bachelor member (Manger and Shanks, 1977). The St. Joe formation occurs throughout Sonora Quadrangle with the best exposures around Beaver Lake. Along the White River in the southern portion of the quadrangle, the top of the St. Joe formation is exposed and is overlain unconformably by the Boone formation (Figs. 6E, F). Boone formation layers were observed to downlap onto the top of the St. Joe formation in some areas (Fig. 6E). In others, broad shallow channels in the top of the St. Joe formation were filled with prograding clinoforms at the base of the Boone formation (Fig. 6F), and lowermost Boone formation layers contained limestone and chert clasts that appeared to be reworked from the underlying St. Joe formation.

The Boone formation is named for the unit's extensive development in Boone County, Arkansas (Branner, 1891; Simonds, 1891). The Boone formation is as gray, fine-to coarse-grained fossiliferous limestone interbedded with abundant chert and is the most widespread stratrigraphic unit exposed in Sonora quadrangle (Figs. 3, 4).

The Batesville formation is named for Batesville, Arkansas, in Independence County (Branner, 1891; Simonds, 1891). The formation is divided into two members, the Batesville Sandstone and the Hindsville Limestone. The Batesville Sandstone is often a flaggy, fine-to coarse-grained, cream-colored to brown sandstone with thin shales. The Hindsville member, found mostly in outcrops in northwest Arkansas, is a crystalline, fossiliferous limestone that, when present, usually occurs at the base of the Batesville formation and can have a chert-pebble conglomerate developed from reworking of chert fragments eroded from the underlying Boone formation. No outcrops of the Hindsville member were observed in Sonora quadrangle. Indeed, in Sonora quadrangle the Batesville formation was only 1–2 m thick and was poorly exposed (Figs. 3, 4).

The Fayetteville Shale was named for Fayetteville, Arkansas. Its type locality is in the valley of the West Fork of the White River in Washington County south of the city of Fayetteville (Simonds, 1891). The Fayetteville Shale is black to dark gray, organic-rich, and calcareous in places (McFarland, 1998). It locally contains abundant septarian concretions ranging from a few cm to almost a m in diameter, some of which contain hydrocarbons and siderite cement (Hutchinson, 2001). The Fayetteville Shale is subdivided into two informally named stratigraphic units and one formal member: lower Fayetteville Shale (informal), the Wedington Sandstone (formal), and the upper Fayetteville Shale (informal). The lower Fayetteville Shale is black, fissile shale. The base is exposed in Sonora quadrangle at the base of Fitzgerald, Webber, and Price Mountains (Fig. 3). The lower Fayetteville Shale outcrops occur widely throughout the southern half of Sonora quadrangle (Fig. 4). The shale often weathers to expansive clay, resulting in damage to foundations of structures built on this shale (King et al., 2001b). The Wedington Sandstone member of the Fayetteville Shale is tan to gray, well-indurated, very fine-to medium-grained sandstone with an average thickness of 2 m. The thickest observed outcrop of Wedington Sandstone (approximately 10 m) is located on the top of Webber Mountain, southeast of the town of Springdale (Figs. 3, 4). The upper Fayetteville Shale is a black, fissile shale that contains abundant iron concretions (<0.2 m diameter). This informally named member of the Fayetteville Shale is much thinner than the lower Fayetteville Shale. The upper Fayetteville Shale weathers quickly to expansive clay and is rarely observed in outcrop. The upper Fayetteville Shale can be seen in the southwestern area of the quadrangle along Zion Road east of Arkansas Highway 265 (Fig. 4). In Sonora quadrangle, the top of the Fayetteville formation is an erosion surface with minor relief overlain unconformably by the Cane Hill member of the Hale formation (McFarland, 1998). This unconformable contact also represents the Mississippian–Pennsylvanian boundary (Handford and Manger, 1990, 1993).

The only Pennsylvanian stratum in Sonora quadrangle is the Cane Hill member of the Hale formation. The Hale formation was named for Hale Mountain in the vicinity of Washington County, Arkansas (Adams and Ulrich, 1905). The Cane Hill member is comprised of several lithologic components: a basal tan, very thin-bedded, medium-grained, siliceous/calcareous sandstone or calcareous conglomerate, alternating with very thin-bedded (<0.15 m thick) siltstone and sandstone layers, often ripple-marked, and thick, tan, ripple-marked, medium grained, siliceous sandstone (Adams and Ulrich, 1905; Henbest, 1953; Cate, 1962; Handford and Manger, 1990, 1993; M.E. King et al., 2001a and b).

Structural Geology.–Sonora quadrangle is situated on the southern flank of the Ozark Dome that is centered in southeast Missouri (Croneis, 1930). The regional dip of

exposed strata is generally less than 5° to the south. Fractures were observed in outcrops of Ordovician through Pennsylvanian strata, and these fractures were believed to result from brittle deformation related to flexure of the Ozark Plateaus during the Ouachita orogeny (Viele, 1989; Viele and Thomas, 1989; Hudson, 2000). Fractures observed on outcrops of the Chattanooga Shale (Devonian) have strikes of N90°E and N20°W with vertical dips (Fig. 6C).

Several faults were observed in Sonora quadrangle. The dominant structures in the quadrangle are the Fayetteville fault, which crosses the central portion of the quadrangle from southwest to northeast, and the White River fault, which crosses the southern half of the quadrangle from west to east (Figs. 3, 4). The Fayetteville fault is a normal fault downthrown to the southeast. In Sonora quadrangle, the fault is exposed along the shores of Beaver Lake where it is observed to offset the St. Joe and Boone formations. It is poorly expressed in the remainder of the quadrangle because it occurs in the Boone formation. However, it was inferred from a dominant lineament observed on both aerial photographs and digital elevation models of Sonora quadrangle.

The White River fault in Sonora quadrangle is a prominent fault in the southern portion of the quadrangle. It is oriented east-west (Fig. 4) and is downthrown to the south. Several other smaller faults run parallel to the White River fault creating a series of small horsts and grabens. Along the primary trace of the White River fault (Fig. 4), the Boone formation (on the north side) is juxtaposed against the Cane Hill member of the Hale formation (on the south side). Thus, offset along the White River fault is substantial. The White River fault also offsets the Fayetteville fault.

Discussion

The stratigraphy of Sonora quadrangle is composed of alternating layers of shale, limestone, and sandstone in genetically related packages bound by prominent regional unconformities. These sedimentary rocks of Sonora quadrangle represent the response of Earth surface systems to global processes affecting global tectonics and globally fluctuating relative sea level throughout the Paleozoic Era. Understanding the geology of northwest Arkansas is the first step in understanding the interplay of processes controlling the long-term geologic evolution of continental margins in general and the southern cratonic margin of North America in particular. As such, geologic mapping in northwest Arkansas during the last few years has helped develop new insights and ideas into the rate and magnitude of Earth processes, and is leading to renewed interest in the stratigraphy of the Ozark Plateaus.

The present study contributed to this geologic renaissance with several important discoveries. First, Metts (1961) did not recognize outcrops of the Cotter and Powell formations east of Hickory Creek and misidentified the Kings River member of the Everton formation as the Sylamore Sandstone member of the Chattanooga Shale. Ordovician deposits are quite extensive in the Hickory Creek area, and the Sylamore Sandstone member of the Chattanooga Shale was not observed anywhere in Sonora quadrangle. This is interesting because the Sylamore Sandstone is present in the northern portion of Rogers quadrangle immediately north of Sonora quadrangle. A second contribution of revised geologic mapping in Sonora quadrangle is the description of sandstone capping the Chattanooga Shale along with documentation of the occurrence of an incised channel at the top of the formation. The presence of the sandstone and the erosional channel between the Chattanooga Shale and the Bachelor member of the St. Joe formation demonstrates the unconformable relationship of the Chattanooga Shale and Bachelor member elsewhere in northwest Arkansas. In addition, it provides some insight into the timing of tectonism, global sea-level variability, and associated relative sea-level changes related to incipient orogenic activity far to the south in the Ouachita area. Similarly, documentation of the apparent unconformable contact of the St. Joe and Boone formations throughout the southern portion of Sonora quadrangle provides additional insight into the timing and magnitude of relative sea-level changes along the southern craton margin during the Mississippian Period. Finally, offset of the Fayetteville fault by the White River fault (Fig. 4) may help bracket the timing of faulting episodes in northwest Arkansas, and this may ultimately improve understanding of the geologic evolution of the Ouachita Orogen and associated uplift and brittle deformation of the Ozark Dome (Croneis, 1930; Viele, 1989; Viele and Thomas, 1989; Hudson, 2000). For example, since the White River fault offsets the Cane Hill member of the Hale formation, it is reasonable to conclude that movement on the White River fault post-dates deposition of the Cane Hill interval (earliest Pennsylvanian). However, displacement on the Fayetteville fault must pre-date movement on the White River fault because the Fayetteville fault is offset by the White River fault. This relation also indicates that the brittle deformation history of the Ozark region must have multiple episodicity and is, perhaps, more complex and long-lasting than had been previously assumed (Chinn and Konig, 1973; Hudson, 2000).

It is clear from the foregoing discussion that revised mapping of Sonora quadrangle and other quadrangles in northwest Arkansas over the past few years (J.T. King et al., 2002; Sullivan and Boss, 2002; M.E. King et al., 2001a) is providing new insights into the geologic evolution of the southern cratonic margin in the context of modern plate tectonic and sequence stratigraphic paradigms.

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Crown Radius and Diameter at Breast Height Relationships for Six Bottomland Hardwood Species

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Abstract

The relationship between a tree's crown radius and diameter at breast height (DBH) has a variety of uses including forest competition studies, tree crown densities, spacing and stocking relationships, wildlife habitat suitability models, and tree volume estimations. Estimating DBH from mean crown radius (MCR) is of interest to natural resource managers because MCR can be estimated from high resolution digital imagery using remote sensing techniques. DBH is a common tree dimensional characteristic that is used to quantify tree and stand structure. This research presents MCR/DBH and DBH/MCR relationships for boxelder (*Acer negundo* L.), sweet pecan *(Carya illinoensis* (Wang) K. Koch), sugarberry (*Celtis laevigata* Willd.), green ash (*Fraxinus pennsylvanica* Marsh.), Nuttall oak (*Quercus nuttallii* Palmer), and American elm (*Ulmus americana* L.). The linear model, $y = a + b * x$, provided the best model fit with adjusted r^2 values of 0.567 to 0.855 for the 6 species. Crown radius can be determined from digital imagery and then used to predict DBH.

Introduction

A tree's crown is defined as that part of a tree bearing live branches and foliage (Helms, 1998). Photosynthesis occurs in leaves and the products from photosynthesis, that is photosynthates, are translocated through the crown's branches from the leaves to the remainder of the tree. Concurrently, water and mineral nutrients absorbed by the roots are translocated through the trunk to branches and leaves. A tree's crown therefore represents the above-ground spatial requirements needed for a tree to survive, grow, and reproduce (Kramer and Kozlowski, 1979).

The shape of a tree's crown is influenced by 2 broad factors: genetics and physical environment (Zimmerman and Brown, 1971; Daniel et al., 1979). Specific tree species tend to have characteristic crown shapes, especially when growing in an open environment. These shapes are then modified by the physical environment including competition between tree crowns through physical abrasion from wind events. Crown shape therefore represents the physical space a tree utilizes for growth as modified by the physical environment.

While a tree's crown represents its potential for growth and development, crown measurements are difficult to obtain (Bechtold et al., 2002). A more easily measured tree variable, such as diameter at breast height (DBH, 1.4 m above the ground), is often used as a surrogate for a tree's crown dimensions. Tree crown dimensions, especially the horizontal dimension, radius or diameter, are well correlated with a tree's DBH. The mean crown radius MCR (or crown diameter)/DBH relationship is particularly useful for determining crown competition factors (Krajicek et al., 1961; Vezina, 1962; 1963; Strub et al., 1975), stand density and stocking relationships (Dawkins, 1963; Roberts and Ross, 1965; Minckler and Gingrich, 1970; Goelz, 1996), and tree growth (Zeide, 1986; Cole and Lorimer, 1994). Likewise, the DBH/tree crown radius (or diameter) relationship is useful for determining tree and stand volumes from aerial photographs (Minor, 1951; Bonnor, 1968; Gering and May, 1995). Volume determination is especially important with recent advances in remote-sensing technology that allow for rapid crown radius or crown diameter measurement, conversion to DBH, then determination of tree volume. Inventory costs are greatly reduced compared to conventional tree DBH measurements in the forest. The objectives of this study were to develop MCR/DBH and DBH/MCR relationships for selected bottomland hardwood species.

Materials and Methods

Location.–The study site was located on Pittman Island in Issaquena County, MS (32°55′ N latitude, 91°09′W longitude) within the unprotected lands along the Mississippi River (batture lands). The site is characterized by ridge and swale topography due to channel migration of the Mississippi River (Mitsch and Gosselink, 1986). Soils vary but are primarily composed of Commerce silt loam (fine-silty, mixed, superactive, nonacid, thermic Fluvaquentic Endoaquepts), Sharkey clay (very fine, smectitic, thermic Chromic Epiaquerts), Bowdre silty clay (clayey over loamy, smectitic, thermic Fluvaquentic

Hapludolls), and Robinsonville very fine sandy loam (coarse-loamy, mixed, superactive, nonacid, thermic Typic Udifluvents). The climate is characterized as humid and warm. The monthly average high temperature is 28°C in July and the monthly average low temperature is 6°C in January. Precipitation averages 142 cm per year with the greatest monthly average in March (15.7 cm) and the lowest monthly average in August (6.8 cm) (Rolling Fork, MS weather station located about 25 km north of Pittman Island; source http://www.msstate.edu/dept/GeoSciences/climate). Periodic summer droughts occur in the region. Past management activities in the forest included a partial harvest in 1979-1980 and infrequent light harvests before 1969.

Measurements.–Six bottomland hardwood species were selected based on their commonality in forests of the Mississippi River batture. These species are boxelder (*Acer negundo* L.), sweet pecan (*Carya illinoensis* (Wang) K. Koch), sugarberry (*Celtis laevigata* Willd.), green ash (*Fraxinus pennsylvanica* Marsh.), Nuttall oak (*Quercus nuttallii* Palmer), and American elm (*Ulmus americana* L.). Trees from each species were selected from control plots of a larger study of the effects of reproduction methods on flora and fauna common in the batture (Lockhart et al., 1996). Trees were selected from a variety of DBH classes to represent a range of diameters and crown widths (see Table 1 for descriptive statistics for each species). Each tree was measured for DBH (cm) and ocularly assessed for crown radius (m) in eight directions from the main bole–every 45° beginning with magnetic north–to the vertically projected edge of the crown. Crown classes, a reflection of a tree's relative competitive status (Smith et al., 1997), were assessed for each tree. Each tree was assigned a crown class of dominant, codominant, intermediate, or overtopped.

Analyses.–Crown radii for each tree were summed and MCR determined. MCR/DBH ratios (MCR divided by DBH) were calculated and compared to an independent data set collected from the Delta Experimental Forest located near Stoneville, MS (about 60 km north of Pittman Island). A description of data collection methods for this independent data set is found in Francis (1986). MCR/DBH ratios were compared within species between the two data sets using t-tests in PC-SAS (SAS, 1986). Comparisons between species were done with a one-way analysis-of-variance. Significant differences were noted at $p \leq 0.05$. Regression models using DBH as the independent variable and MCR as the dependent variable were evaluated using Table Curve version 5.01. Likewise, models were also evaluated using MCR as the independent variable and DBH as the dependent variable. Table Curve evaluates more than 8,000 model forms ranging from simple linear to complex non-linear models.

Results and Discussion

DBH/MCR Ratios.–The DBH/MCR ratio is a measure of the efficiency of a tree to accumulate DBH per unit of crown area. The higher the ratio, the more efficient a tree (or species) is at accumulating DBH. Comparing all trees in the study, green ash was found to be the most efficient species at accumulating DBH and American elm the least efficient (Table 2). For example, for each meter of crown radius in green ash, 13.9 cm of DBH was accumulated, whereas only 7.9 cm of DBH was accumulated in American elm. No differences occurred among the remaining species. When overtopped trees were removed from the data set leaving only trees that received a minimum of direct sunlight at the top of the crown, green ash was still the most efficient species at accumulating DBH and American elm was still the least efficient (Table 2). Boxelder was also more efficient than sweet pecan, sugarberry, and Nuttall oak. Note though that a higher percentage of trees from shade-tolerant species (boxelder, sugarberry, and American elm) were crown classed as overtopped compared to moderately shade-intolerant species (sweet pecan, green ash, and Nuttall oak).

The DBH/MCR ratios for green ash and Nuttall oak in the present study were significantly greater than those found by Francis (1986) for the same species, $P=0.0001$ and 0.0001, respectively. Sugarberry and American elm had similar ratios between the two studies ($P=0.0640$ and 0.1432, respectively). Apparently, site conditions or stand history may influence the DBH/MCR ratio for a given species. The site studied by Francis (1986) was fairly homogeneous consisting of Sharkey clay. Further, Francis collected data only for trees in the dominant, codominant, and intermediate crown classes while the ratios in the present study included overtopped trees in addition to the other three crown classes. Data in the present study represent a wider range of diameter classes than Francis (1986; see Table 1). Furthermore, the forest in the present study was subject to periodic harvesting, favoring moderately shade-intolerant species such as green ash and Nuttall oak, whereas the stand in Francis (1986) was relatively undisturbed, which may have influenced the difference in the DBH/MCR ratios between the two studies.

MCR/DBH Regression Models.–Theoretically, the MCR/DBH relationship would be sigmoid for forest grown trees (Dawkins, 1963). Crown expansion would be slow relative to early DBH growth as trees are crowded in dense young stands. As trees begin to express dominance, DBH growth increases almost linearly as crown expansion increases. When the tree reaches maturity, crown expansion essentially ceases while DBH continues to increase as photosynthates acquired through photosynthesis are increasingly used for tree maintenance and support.

Dawkins (1963) further identified 6 general crown-diameter to bole-diameter (e.g., DBH) relationships in trees based on work from previous investigators. Three relationships were linear and differed in whether the y-intercept was zero, positive, or negative. The 3 other relationships were non-linear. One was sigmoid as described above while the other two were power functions with a positive or negative slope.

Results from the present study indicated that high coefficients of determination ($r^2 > 0.90$) were attainable for each species. Non-linear equations with multiple polynomials, up to 14 to 16 order polynomials, accurately described MCR/DBH relationships within species. But these equations are not robust since they are specific to this particular data set and probably not applicable to other MCR/DBH data sets of the same species. The linear equation (1) was selected as the best general relationship to describe the MCR/DBH relationship, indicating that crown radius increases with increasing DBH within the limits of our data set.

MCR = a + b * DBH where:

MCR = mean crown radius (m),

DBH = diameter at breast height (cm), and

a, b = coefficients determined from regression.

Linear equation 1

Results from linear regression for the 6 bottomland hardwood species are shown in Table 3. Coefficients of determination (r^2) ranged from 0.87 for sweet pecan to 0.56 for boxelder. These coefficients of determination are similar for 3 of the 4 species common between this study and Francis (1986). Coefficients of determination were 0.61, 0.82, and 0.86, for sugarberry, green ash, and Nuttall oak, respectively from Francis (1986). The one species with a considerable difference in the coefficient of determination is American elm–0.65 in the present study and 0.81 in Francis (1986). A likely explanation for this difference is the spreading, umbrella-like crown of overstory American elm. Francis (1986) measured only trees with a portion of their crown in the overstory (dominant, codominant, and intermediate crown classes) while our data also included trees in the overtopped crown class. Shade-tolerant species, such as American elm, that are in the overtopped crown class tend to have a large crown radius per unit DBH in an effort to capture more sunlight in the understory. The species with the lowest coefficients of determination (boxelder, American elm, and sugarberry) are all shade-tolerant species.

The linear MCR/DBH relationships with positive y-intercepts shown in Table 3 follow the Type 2 behavior described by Dawkins (1963). Dawkins (1963) stated that a possible depression in the relationship could occur at the upper end of the MCR/DBH relationship due to tree senility. Such a depression is possible with bottomland hardwood species, but none was found in the present study. The forest in which the crown radii and DBH data were collected was under management. As trees approach a large size, about 70 to 80 cm, they are harvested, thereby preventing them from reaching even larger sizes and making testing for a depression in the MCR/DBH relationship impossible.

DBH/MCR Regression Models.–Much interest exists in DBH/MCR relationships due to their utility in forest inventory using remote sensing techniques. Measurements of crown radius (or crown diameter) from aerial photographs or digital imagery can be converted to DBH at the individual tree level. Diameter at breast height can then be readily converted to volume. Use of remote sensing techniques for stand volume determination reduces inventory costs because the expense and difficulty of establishing and measuring sample plots on the ground is reduced or eliminated (Gering and May, 1995).

Results from linear regression using MCR as the independent variable and DBH as the dependent variable for the 6 bottomland hardwood species are in Table 4. Coefficients of determination (r^2) were the same as in Table 3 because the independent and dependent variables were only switched. The coefficients for sweet pecan, Nuttall oak, and green ash ($r^2 \geq 0.74$) were similar to Gering and May (1995) for upland hardwoods ($r^2=0.80$), oaks/hickories ($r^2 = 0.85$), and gum/yellow-poplar ($r^2=0.94$) species groups. Gering and May (1995) noted that caution should be exercised when using DBH/MCR relationships from remote sensing measurements. Crown diameter (or crown radius) measurements obtained from remote sensing measurements will generally be less than measurements of the same trees using ground-based measurements (Spurr, 1948). They stated that only that portion of the crown visible from directly above will be measured on imagery while branches obscured by others trees will not be seen. Gering and May (1995) further stated that it may be inappropriate to use DBH/MCR relationships developed from ground-measured crown variables with data obtained from remote sensing. Further work is needed to compare ground-measured and remote-sensing measured crowns in bottomland hardwood species.

In summary, linear equations were developed to predict MCR from DBH and DBH from MCR for 6 bottomland hardwood species in the batture along the Mississippi River. These equations can be useful in predicting crown radius

rom forest inventories for stocking guideline development, growth models, remote sensing/stand volume estimation, and wildlife suitability index models that use crown characteristics. Results were in general agreement with a previous study of crown radius and DBH relationships in bottomland hardwoods; although enough differences existed to warrant further study. Future research is needed with a greater variety of site and stand conditions in addition to a greater variety of tree sizes among the various bottomland hardwood species.

ACKNOWLEDGMENTS.– We thank the Arkansas Forest Resources Center and Anderson-Tully Company for their financial support in the development of the Pittman Island Project. We also thank Tom Dell, Steve Meadows, Ted Leininger, 2 anonymous reviewers, and the Assistant Editor for their constructive reviews of this manuscript.

Table 1. Basic statistics for tree dimensions for bottomland hardwood species for Pittman Island and the Delta Experimental Forest from Francis (1986).

Species	n	DBH (cm)			MCR(m)		
		average	range	stand. dev.	average	range	stand. dev.
Pittman Island							
boxelder	85	28.1	9.6–55.9	11.0	2.87	0.74–4.76	0.92
sweet pecan	106	35.5	11.0–85.6	19.2	3.60	0.85–8.87	1.95
sugarberry	111	26.7	9.7-61.5	11.8	2.82	0.97-6.08	1.04
green ash	85	44.5	10.3-89.5	20.4	3.38	0.29-7.50	1.48
Nuttall oak	92	50.9	10.3-97.0	20.3	4.87	1.56-9.67	1.79
American elm	89	29.6	8.9-61.0	13.9	3.67	1.22-6.49	1.16
Delta Experimental							
Forestsugarberry	75	38.2	19.1-69.3	11.3	4.24	2.45-5.89	0.84
green ash	75	39.0	14.2-72.4	14.9	4.55	1.98-8.78	1.52
Nuttall oak	75	47.6	17.3-89.7	18.8	6.33	2.55-11.12	2.26
American elm	75	39.0	19.1-69.3	12.9	4.66	2.19-8.19	1.33

Table 2. DBH/MCR ratios for 6 bottomland hardwood species on Pittman Island, Issaquena County, Mississippi.

Species	All trees			Dominant, Codominant, and Intermediate Crown Classes only		
	n	ratio	std. dev.	n	ratio	std. dev.
boxelder	85	10.2b[1]	4.1	47	12.3b	4.2
sweet pecan	106	10.1b	2.6	77	10.3c	2.2
sugarberry	111	9.5b	2.5	53	10.7c	2.1

Table 2. Continued

Species	All trees			Dominant, Codominant, and Intermediate Crown Classes only		
	n	ratio	std. dev.	n	ratio	std. dev.
green ash	85	13.9a	6.8	67	14.0a	3.5
Nuttall oak	92	10.4b	2.1	81	10.9c	1.6
American elm	89	7.9c	2.5	45	9.2d	1.8

[1]Ratios within a column followed by different letters are significantly different at $p \leq 0.05$.

Table 3. Linear regression coefficients[1], coefficient of determination, and mean square error for regression to predict MCR (m) from DBH (cm) for 6 bottomland hardwood species on Pittman Island, Issaquena County, MS.

Species	a	b	adjusted r^2	mean square error
boxelder	1.03881	0.06024	0.56	0.57914
sweet pecan	0.22824	0.09022	0.87	0.65957
sugarberry	0.83957	0.06901	0.67	0.54968
green ash	0.55786	0.05782	0.76	0.65359
Nuttall oak	0.71736	0.07655	0.84	0.68130
American elm	1.61597	0.06395	0.65	0.64182

[1] Regression coefficients are for the linear equation MCR = a + b * (DBH).

Table 4. Linear regression coefficients[1], coefficient of determination, and mean square error for regression to predict DBH (cm) from MCR (m) for 6 bottomland hardwood species on Pittman Island, Issaquena County, MS.

Species	a	b	adjusted r^2	mean square error
boxelder	2.32630	9.44191	0.56	7.25046
sweet pecan	2.22484	9.69720	0.87	6.83823
sugarberry	-0.15586	10.02871	0.69	6.62635
green ash	4.59360	12.62015	0.74	10.40775
Nuttall oak	0.25439	10.97732	0.84	8.15868
American elm	-6.47036	10.29775	0.65	8.14452

[1]Regression coefficients are for the linear equation DBH = a + b * (MCR).

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Sediment Loading and Water Quality of Field Run-off Water

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Abstract

Intensive tillage is commonly employed in many agronomic production systems in the United States. Tillage operations may include disking the field, re-smoothing the soil, seedbed formation, reducing the seedbeds, and shallow cultivation for weed control. Tillage practices in conjunction with rainfall have been linked to soil erosion, which may adversely affect the environment. The soil erosion dynamics of two large-scale production cotton fields that utilized both modern-conventional and conservation-tillage technology were examined. Studies were conducted in the cotton-producing region of southeast Arkansas in the Bayou Bartholomew watershed. Bayou Bartholomew is currently listed by the United States Environmental Protection Agency as an impacted stream. The soils at these sites were related, coarse-textured alfisols. One field was cropped to conventionally tilled cotton and intensively tilled. The second field was cropped to cotton using modern conservation tillage technology. Both fields were furrow-flow irrigated using piped water. Intense rainfall usually occurs in the Mississippi River Delta Region, particularly in the winter and spring months. Conservation tillage proved to be immediately beneficial in controlling soil erosion and sediment loss due to field run-off water from rainfall. Sediment content of run-off water induced by rainfall from the conventionally tilled cotton field was significantly greater than the sediments found in run-off water from the conservation tilled cotton field. The amount of sediment found in rainfall run-off water decreased more rapidly with time under conservation tillage than under conventional tillage. The tillage system made little difference in sediment content of run-off water from irrigation. The water flow from furrow irrigation is typically slow and steady. There is no droplet impact on the ground from furrow-flow irrigation as there would be from rainfall. Apparently, the gentle flow of the water down the furrows was insufficient to dislodge large numbers of soil particles.

Introduction

The Soil Erosion Process.–Soil erosion by water is a two-step process (Brady and Weil, 2002). First, water droplets from rainfall strike the soil surface and tear away primary soil particles–this process is the detachment phase. Second, as the water collects and recedes, the soil particles are carried away from their native site with run-off water–this is called the transportation phase. Some soil erosion occurs on all soils. Normal rates of soil erosion range from 228 to 456 kg ha-1 (0.1 to 0.2 ton acre-1) every year. Accelerated erosion occurs when the normal rates of soil erosion are exceeded (Wild, 1993). Accelerated erosion in the Arkansas Delta region is typically caused by water run-off from intense winter and spring rainfall.

Farmers and agricultural producers in the Mississippi River Delta region typically prepare seed beds for crops in early spring. Conventional tillage operations used for seedbed formation are primarily disking and raised crown seedbed formation (Bonner, 1993; Waddle, 1984). The finished beds allow the soil to warm rapidly and promote drainage of excess surface water. The weather conditions in the Delta region vary widely from season to season, and early spring rains frequently occur as heavy down pours. Heavy, frequent rainfall events on loose, unconsolidated soil surfaces promote accelerated soil erosion (Dendy, 1981). Losses of soil from freshly tilled fields may reach tonnes per hectare depending on field slope and weather. Sediments generated from tillage may result in surface water contamination.

Further, additional nitrogen and phosphorus carried in eroded sediments or as soluble species from fields may ultimately increase the potential for eutrophication of surface waters (Boesch et al., 2000; Goolsby and Battaglin, 2000). Sediments and nutrients that find their way from Arkansas Delta region soils into surface waters could also find their way to tributary rivers and streams and then the Mississippi River. The final and ultimate fate of these sediments and nutrients may be to contribute to the growing hypoxic zone in the Gulf of Mexico.

Conservation Tillage.–Cotton (*Gossypium hirsutum* L.) production in the United States is typically a tillage intensive culture. Tillage operations employed in most cotton production include disking to disrupt the soil surface, re-smoothing the field, bedding, knocking down the beds, and shallow cultivation for weed control during the growing season. These tillage practices have been linked to soil compaction (McConnell et al., 1989) and soil erosion (Mutchler et al., 1985), which may reduce crop yields and adversely affect the environment.

Utilizing conservation tillage systems in cotton production has been shown to substantially reduce soil erosion (Mutchler et al., 1985). However, residue cover of the soil surface from cotton is usually less than from high residue crops such as corn or grain sorghum. Production systems that include winter cover crops further reduce soil loss by reducing raindrop impact, slowing run-off, and holding soil in place when winter rainfall becomes intense (Stallings, 1957).

Experimental Objectives.–These studies were designed to determine the impact of conventional tillage and conservation tillage on sediment loss from cotton fields near a stream (Bayou Bartholomew) that is currently listed as "impacted" by the United State Environmental Protection Agency (EPA). The rate of sediment loss as a function of tillage system, time of year, and within each rainfall event was also investigated.

Experimental Methods

Field and laboratory studies of run-off water quality from agricultural fields employing modern, conservation-tillage technology and conventional-tillage technology were conducted. The site for the demonstrations was within the cotton-producing region of Southeast Arkansas on producer fields in the Bayou Bartholomew watershed. The Bayou Bartholomew has been classed as impacted by the EPA, primarily for sediment content of the water. Two large, producer fields cropped to cotton were utilized in these studies. The fields were approximately 16 hectares in area and rectangular. The soils at these sites were related, coarse-textured alfisols. One field was in conventional cotton production and intensively tilled. The second field utilized conservation tillage production technology. Both fields were furrow-flow irrigated. Furrow-flow irrigation requires that water be pumped to the field through pipe, either plastic or metal, and released upslope in the furrows of a field. The water then moves slowly down slope by gravity and replenishes the crop.

Run-off water from rainfall and from irrigation events was sampled from low points at the drainage ends of the fields. The water samples were collected with an ISCO 6700 automated sampler at various times during the growing season. All samples were collected from the sites within 24 hours and analyzed at the Arkansas Water Quality Lab using EPA approved analysis and QNQC procedures within 48 hours. The water samples were analyzed for sediments, N, P, K, electrical conductivity, and soluble pesticides. Only the sediment content of the run-off water is reported here.

Field-wide sediment loss was calculated using sediment concentration of the run-off water, average water infiltration rates, and historical precipitation data. The soils at the test sites infiltrate an average of 1.1 cm of water hr^{-1} (Soil Cons. Service, 1972). Historical precipitation data was found from the National Oceanic and Atmospheric Administration (NOAA, 2001). We estimate that 50% of the total rainfall infiltrated the soil, while 50% of the rainfall left the fields as run-off water. The average sediment content of the run-off water was multiplied by the estimated volume of run-off water to calculate estimated sediment losses.

Results and Discussion

Soil Erosion and Sediment Loss–Prior to Planting. Total average rainfall during the months of March and April in southeastern Arkansas is 18.5 cm (NOAA, 2001). Rainfall infiltration is assumed to average 50%, while the other half of the water is assumed to leave the field as run-off. Runoff water samples were collected in conjunction with precipitation on 4 April 2000. Run-off water, particularly under conventional tillage, contained large amounts of sediments that slowly declined with time (Fig. 1). The conventionally tilled field had run-off water containing an average of 491 mg of sediment L^{-1} (Table 1). This translates into 4,302 kg of sediment ha-1 lost from the field. This is calculated to be 68.8 tonnes of sediments lost from the 16-ha field during the early growing season. The run-off water from the field utilizing conservation tillage technology contained an average of 491 mg of sediment L^{-1}. Calculations show the field loss to be 454 kg of sediment ha-1 or 7.3 tonnes of sediment for the entire 16-ha field. Conservation tillage reduced sediment content of the March and April run-off water by 3,848 kg ha^{-1} or 89%.

Winter weeds and debris from the previous year's crop protected the soil surface of the conservation-tilled field in the spring. Prior to planting in the spring, the soil on the conventionally tilled field was loose and bare, with no plant life to block direct impact of rain droplets onto the soil surface. Loose, bare soil produced by conventional tillage was especially vulnerable to soil erosion from intense rainfall compared to conservation tillage prior to planting. Sediment loading of run-off water prior to planting was excessive under conventional tillage, and moderate under conservation tillage.

Soil Erosion and Sediment Loss–Early Season.–Total average rainfall during May in southeastern Arkansas when the soil would generally be tilled and bare is 12.1 cm (NOAA, 2001). Runoff water samples were collected in conjunction with precipitation on 4 May 2000. With less than 5% of the soil surface covered, accelerated erosion still occurred in the conventionally tilled field. Run-off water from rainfall averaged 3,200 mg of sediment L^{-1} or 1,936 kg of sediment ha^{-1} from the conventional tillage field (Table 1). The net field loss was calculated to be 31.0 tonnes of sediment from the 16-ha conventionally tilled field.

Table 1. Calculated yearly sediment losses from cotton fields employing conservation and conventional tillage in the Bayou Bartholomew watershed.

		Run-off Sediment Content		Sediments Lost Through Erosion	
Type of Event	Average Rainfall (cm)	Conservation Tillage (mg L^{-1})	Conventional Tillage (mg L^{-1})	Conservation Tillage (kg ha^{-1})	Conventional Tillage (kg ha^{-1})
Early Spring					
Rainfall[1]	18.5	491	4,651	454	4,302
Mid-Spring					
Rainfall[2]	12.1	580	3,200	351	1,936
Early Summer					
Rainfall[3]	8.9	597	951	266	424
Irrigation[4]	10.2–25.45	≤ 10	≤ 10	0	0
Total Calculated Yearly Sediment Loss				**1,071**	**6,662**

[1]March 15 through April.

[2]May.

[3]June.

[4]Non-rainfall water. Typically irrigations are required in late June, July, and August.

[5]Between 2 and 5 irrigations of approximately 5.1 cm of water applied per irrigation.

Run-off water under conventional tillage contained larger amounts of sediment that slowly declined with time, while conservation tillage run-off water contained less sediment and declined faster with time (Fig. 2). Run-off water from the conservation tillage field on 4 May contained an average of 580 mg of sediment L-1. Using the total rainfall and estimated total run-off, the conservation-tilled field was determined to have lost 351 kg of sediment ha-1. The calculated net field loss was 5.6 tonnes of sediment from the 16-ha conservation tilled field. Conservation tillage reduced sediment content of run-off water and soil erosion by 1,585 kg ha-1 or 82% during May.

After planting on 4 May 2000, the soil on the conventionally tilled field was still bare, with only small cotton seedlings, which did little to block the direct impact of rain droplets onto the soil surface. Dead winter weeds and previous crop residues protected the soil of the conservation tillage field. Cotton seedlings alone did little to impede droplet impact, hold the soil together, or slow run-off water.

Soil Erosion and Sediment Loss–Mid-Season.–Total average rainfall during June in southeastern Arkansas when there would generally be actively growing cotton plants is 8.9 cm (NOAA, 2001). Runoff water samples were collected in conjunction with precipitation on 5 June 2000. Run-off water from the conventionally tilled field averaged 951 mg of sediment L-1 or 424 kg of sediment ha-1 (Table 1). The net field loss from the conventionally tilled field was calculated to be 6.8 tonnes of sediment from the 16-ha field. Run-off water from the conservation tillage field on 5 June contained an average of 597 mg of sediment L -1 or 226 kg of sediment ha-1. The calculated net field loss was 3.6 tonnes of sediment from the 16-ha field. Sediment tended to erratically decline with time under both tillage systems (Fig. 3). Conservation tillage reduced sediment content of the run-off water and soil erosion by 189 kg ha-1 or 47%.

As the cotton plants continued to grow they provided better protection of the soil surface by intercepting more droplets of rain. Additionally, easily eroded soil had already been removed by prior rainfall events. Run-off water under conventional tillage contained larger amounts of sediments

han run-off water from conservation tillage. The soil in the onventionally tilled field on 5 June was only partially rotected by the cotton plants. Conservation tillage better rotected the soil with dead residue of the previous crop, nd older, larger cotton plants than on 4 May.

Soil Erosion and Sediment Loss–Irrigation.– Weather patterns in the Delta region of southeast Arkansas during the mid-summer typically result in less rainfall than in the spring and early summer. During this period, the water requirements of the developing cotton crop are usually met with in-furrow irrigation. Runoff water samples were collected in conjunction with irrigation on 22 July 1999. Run-off water from irrigation of both the conventional-tillage and conservation-tillage field averaged less than 10 mg of sediment L^{-1}, less than 1.0 kg of sediment ha^{-1} (Table 1). The net field loss of sediments due to irrigation was negligible.

In-furrow irrigation is the most common method of providing supplemental water. The water flow from furrow irrigation is slow and steady. There is no droplet impact on the ground as there would be from rainfall. Run-off water from irrigation of both the conservation and conventionally tilled fields contained almost no sediments (Fig. 4). The gentle flow of the water down the furrows was not found to be sufficient to dislodge soil particles. Without droplet impact, the sediment load of the run-off water was greatly reduced.

Total Calculated Sediment Loss.–Calculated soil erosion and sediment loss was less for the conservation-tillage field than the conventional-tillage fields (Table 1). Although estimates of yearly soil erosion for both conservation tillage and conventional tillage exceeded established limits for accelerated erosion, these studies found that conservation tillage was very effective in reducing soil erosion and sediment content of run-off water. The estimated yearly reduction of sediment loss due to soil erosion made possible by employing conservation-tillage practices was found to be 84%.

Conclusions

Conservation tillage was of immediate benefit in controlling soil erosion and sediment loading in run-off water under the intense rainfall conditions that may occur in the Delta Region. Sediment loading was significantly greater in run-off water from conventionally tilled cotton as compared to conservation-tilled cotton. Sediment content of run-off water was found to decline with time. Sediment loading of the run-off water during early months of the growing season was greater than in later, summer months. Generally, sediment content of the run-off water began at its highest level and declined with time within each rainfall event.

The water flow from furrow-flow irrigation was typically slow and steady. There was no droplet impact on the ground from this method of irrigation. The gentle flow of the water down the furrows was not sufficient to dislodge soil particles. Without droplet impact, the run-off water sediment load due to irrigation was greatly reduced.

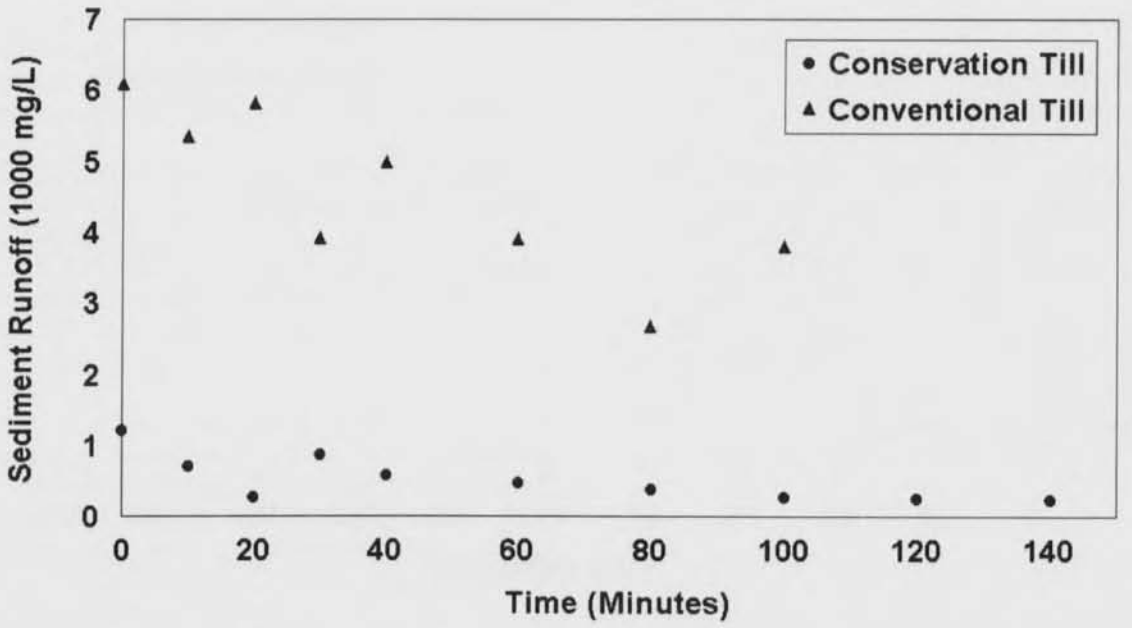

Fig. 1. Sediment losses found in rainfall run-off water on 4 April 2000 (prior to planting) under conservation and conventional tillage.

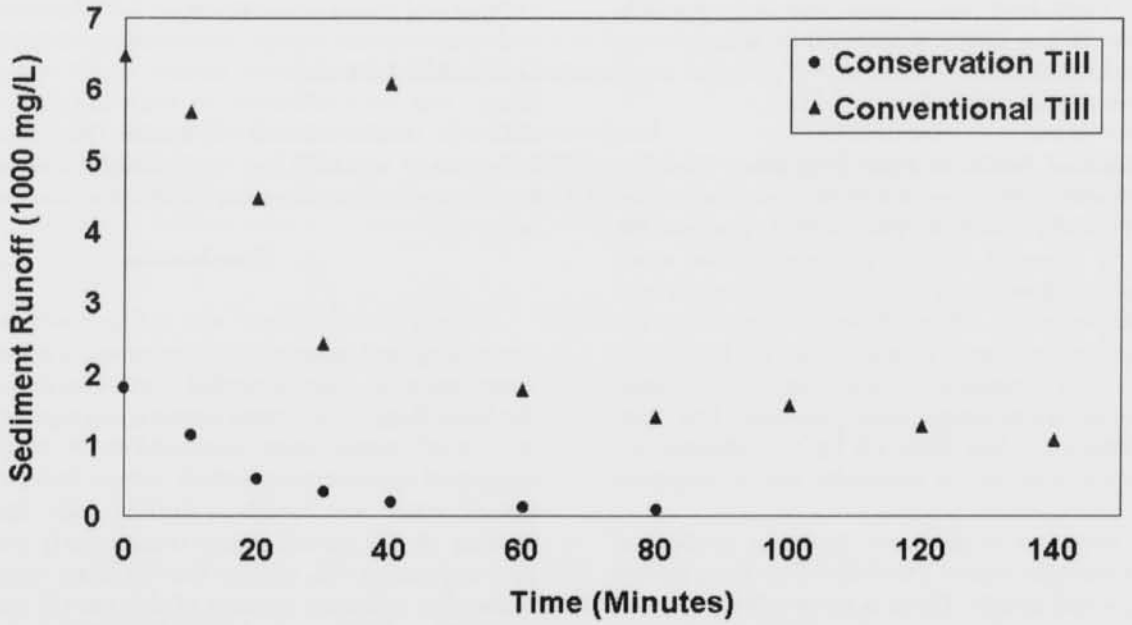

Fig. 2. Sediment losses found in rainfall run-off water and soil erosion on 4 May 2000 (shortly after cotton was emerged) under conservation and conventional tillage.

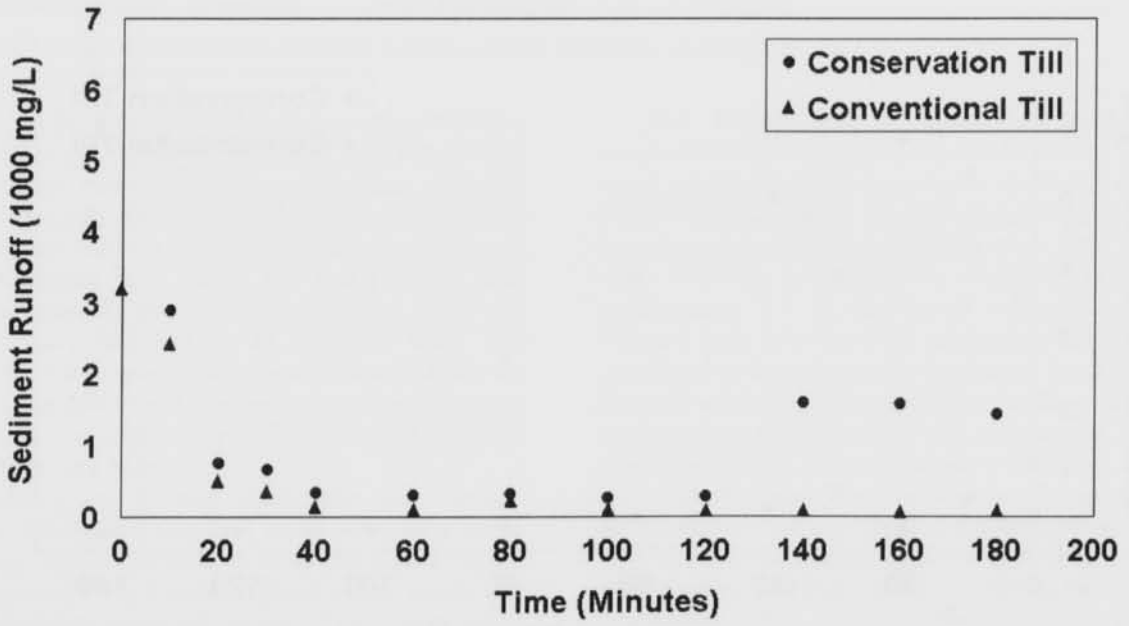

Fig. 3. Sediment losses found in rainfall run-off water on 5 June 2000 under conservation and conventional tillage. The cotton crop was approaching the first square growth stage.

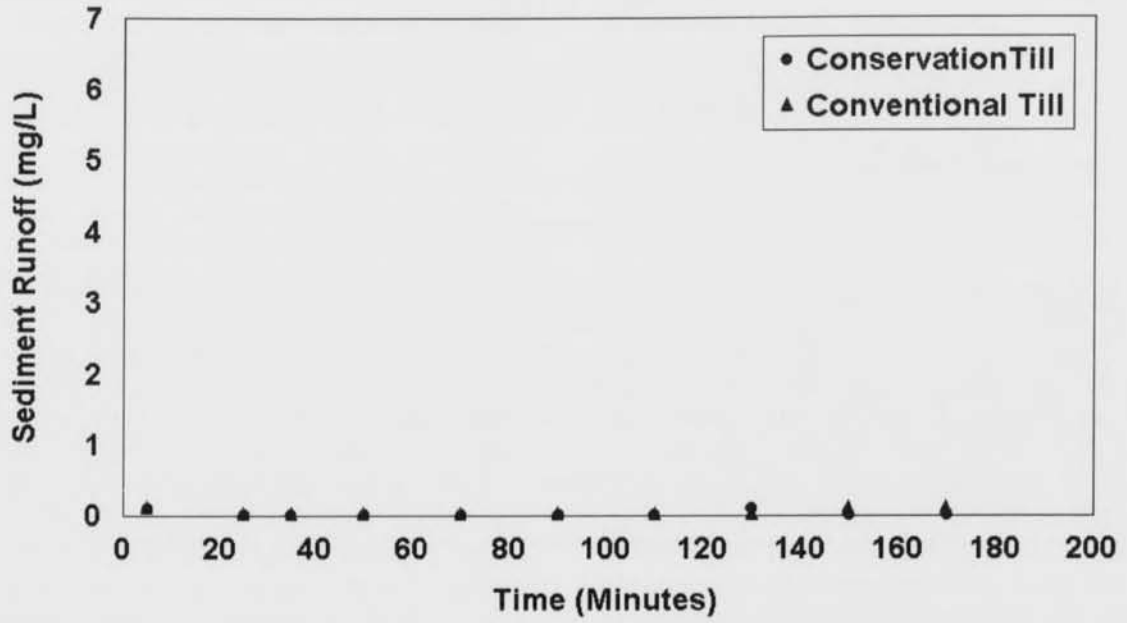

Fig. 4. Sediment losses found in irrigation run-off water found on 22 July 1999 under conservation and conventional tillage. The cotton crop was near mid-bloom stage.

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Synthesis and Evaluation of New Cathepsin D Inhibitors

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Abstract

Cathepsin D, a lysosomal aspartic protease, has been suggested to play a role in the metastatic potential of several types of cancer A high activated cathepsin D level in breast tumor tissue has been associated with an increased incidence of relapse and metastasis. High levels of active cathepsin D have also been found in colon cancer, prostate cancer, uterine cancer, and ovarian cancer. Hydroxyethyl isosteres with cyclic tertiary amine have proven to be clinically useful as inhibitors of aspartyl proteases, such as cathepsin D and the HIV-1 aspartyl protease. Also cathepsin D has recently been associated with the development of Alzheimer's disease. Specific proteinase inhibitors, useful in investigations of the mechanisms and pathways of intracellular protein degradation, could lead to the development of therapeutic agents for treatment of many types of carcinomas as well as Alzheimer's disease. The design and the synthesis of (hydroxyethyl)amine isostere inhibitors with the cyclic tertiary amines is described. The IC_{50} and apparent Ki values for several cathepsin D inhibitors are reported.

Introduction

A number of drugs currently in clinical use exert their action by inhibiting a specific enzyme, the target enzyme, present either in the tissues of the individual under treatment, or those in the invading organism. Proteolytic enzymes (proteases) are involved in many biological functions and deteriorative diseases (Handsley and Edwards, 2005; Zamorano et al., 2005). Among the most biologically important proteases are aspartyl proteases. Many serious medical problems, such as cardiac disease (Soylu et al., 2004), acquired immunodeficiency syndrome (AIDS); (Bonhoeffer et al., 2004), Alzheimer's disease (Bornebroek and Kumar-Singh, 2004), malaria (Bjelic and Aqvist, 2004), as well as colorectal and breast cancer (Wiedswang et al., 2004), and pancreatic cancer (Shen et al., 2004), either result directly from, or are characterized by, uncontrolled aspartyl protease activity. For example, the HIV-1 aspartyl protease, which is responsible for the maturation of HIV into the infectious viral particles (Darke and Huff, 1994), has become an important therapeutic target for treatment of AIDS (Johnson et al., 2004; Harrigan et al., 2005).

Cathepsin D is an aspartyl protease that is very similar to the HIV-1 aspartyl protease in substrate specificity. Cathepsin D is normally restricted to the lysosomes where it is involved in normal protein degradation. However, high levels of active serum cathepsin D are often found in many cancer patients (Vetvicka, 2004), and also in patients with advanced Alzheimer's disease (Li et al., 2004). Cathepsin D is clearly involved in the process of tumor invasion and metastasis (Wang and Lin, 2004). In fact, blood tests are given to many cancer patients where cathepsin D is measured as a prognostic indicator in several cancers, including breast cancer (Fan et al., 2004), bladder cancer (Gontero et al., 2004), and lung cancer (Vetvicka, 2004).

The first step in the development of a new drug is the discovery or synthesis of a library of compounds with a desirable biological activity (Berwowitz and Katzung, 2001). A large number of these compounds are selected for cell studies. Many are then eliminated at this point due to poor cell permeability. Factors such as shape, size, polarity, solubility, lipophilicity, and pKa of the compounds effect cell permeability. Only those compounds that prove most effective in cell studies are carried on to animal studies. In animal studies poor bioavailability, pharmacokinetics, or pharmacodynamics eliminate many other compounds as potential drug candidates. Therefore, the larger the initial library of compounds with the desirable biological activity, the more likely a viable drug candidate will be found. Also, occasionally, compounds reported in the literature to be good inhibitors of one enzyme constitute important lead compounds for the development of inhibitors of a different but similar enzyme. So, the structure of reported cathepsin D inhibitors can be lead to the design of anti-HIV-1 or anti-malarial agents. We have, therefore, undertaken the development of a library of cathepsin D inhibitors with varying physical properties (solubility, pKa, etc.). The compounds reported in this article are important additions to our earlier work (McConnell, et. al., 2003).

The use of hydroxyethyl isosteres with cyclic tertiary amines has lead to compounds with enhanced oral

bsorption (Smith et al., 1997). The (R)-hydroxyethylamine ısert is incorporated as a key component of many clinically sed, highly potent, HIV-1 protease inhibitors. Initially everal compounds that contain hydroxyethyl amine sosteres with flexible alkyl amines were developed Beaulieu et al., 1997), but they suffered limited in vivo half-ives and were not therapeutically useful. Molecular modeling (HYPERCHEM) has shown that a six-member ring forming the tertiary amine is able to orient the backbone of the inhibitor toward a bioactive conformation. This also provides more of a non-peptide functionality which may greatly improve the half-life of the inhibitor in vivo. We have shown by molecular modeling that the phenyl group of a phenyalanine-type hydroxyethylene or hydroxyethyl amine is easily positioned in the S1 site of the HIV-1 aspartyl protease (McConnell et al., 1991, 2003). Other studies show that a bulky amine or amide might fit reasonably well into the S_2 and S_3 sites (Paul et al., 1995). Therefore, we decided to synthesize compounds that contain a peptide portion to accommodate the S_1, S_2, and S_3 subsites and a non-peptide hydroxyethyl isostere portion with a cyclic tertiary amine to accommodate the S_1, S_2, and S_3 enzyme subsites. The general structure of our synthetic target is shown in Fig. 1.

Materials and Methods

All reagents were purchased from either Aldrich, Sigma, or Bachem Chemical Company. Anhydrous solvents were "anhydrous grade" from Aldrich Chemical Company. Dry solvents were distilled from sodium just prior to use. All other solvents were HPLC grade. Thin layer chromatography (TLC) was run on Whatman PE SIL G/UV 250µm silica gel plates. Column chromatography was run on either Aldrich TLC grade silica gel 2-25µm particle size with average pore diameter 60D or Sigma Sephadex LH-20, lipophilic, bead size 20-100µm. The ^{1}H NMR spectra were collected either on a Bruker 200 MHz AC 200 superconducting spectrometer or on a Hitachi 60 MHz R1200 RS NMR spectrometer. ^{1}H NMR of final compounds and major intermediates were collected on the 200 MHz spectrometer, while the spectra of minor intermediates were collected on the 60 MHz NMR spectrometer. The spectral data were processed by NTNMR software produced by TeleMag.

3-(*S*)-BOCamino-4-phenyl-1-*N*-piperazine-2(*S*)-butanol (1a). A solution of 3-(*S*)-*t*-butoxycarbonyl (BOC) amino-4-phenyl-2-(*R*)-oxirane (McConnell et al., 2003); (1.0 g, 3.1 mmol) in 100 mL dry THF was treated with 0.861 g (10 mmol) piperazine. The solution was refluxed for 48 hrs. The mixture was then cooled to room temperature, concentrated under reduced pressure to about one half its volume, and partitioned between ethyl acetate (200 mL) and 5% aqueous sodium potassium tartarate (200 mL) containing 1.0 g NaCl. The organic layer was washed with distilled water (100 mL) and dried over anhydrous magnesium sulfate. The solvent was evaporated under reduced pressure to give a white solid (1.127 g). The crude product was purified by silica gel column chromatography (2.5 cm x 60 cm length) using 60% ethyl acetate/hexanes as the mobile phase to give 0.784 g. TLC (60% ethyl acetate/hexanes) R_f=0.59. ^{1}H NMR (CDCl3/TMS, 200 MHz) ß 0.9895 (9H, s), 1.653 (2H, t), 2.255 (8H, t), 2.445 (2H, d), 3.845 (1H, d of t), 4.3821 (1H, m), 5.015 (1H, m, exchangable), 7.115 (5H,s).

3-(*S*)-BOC-amino-4-phenyl-1-*N*-phenylpiperazine-2-(*S*)-butanol (1b). A solution of 3-(*S*)-*t*-butoxycarbonyl (BOC) amino-4-phenyl-2-(*R*)-oxirane (McConnell et al., 2003); (1.0 g, 3.1 mmol) in 100 mL dry THF was treated with 3.25 g (20 mmol) N-phenylpiperazine. The solution was refluxed for 48 hrs. The mixture was then cooled to room temperature, concentrated under reduced pressure to about one half its volume, and partitioned between ethyl acetate (200 mL) and 5% aqueous sodium potassium tartarate (200 mL) containing 1.0 g NaCl. The organic layer was washed with distilled water (100 mL) and dried over anhydrous magnesium sulfate. The solvent was evaporated under reduced pressure to give a white solid (0.837 g). The crude product was purified by silica gel column chromatography (2.5 cm x 60 cm length) using 60% ethyl acetate/hexanes as the mobile phase to give 0.663 g. TLC (50% ethyl acetate/hexanes) R_f = 0.45. ^{1}H NMR (CDCl3/TMS, 200 MHz) ß 1.003 (9H, s), 1.685 (2H, t), 2.338 (4H, t), 2.498 (2H, d), 2.691 (4 H, t), 3.892 (1H, d of t), 4.491 (1H, m), 4.925 (1H, m, exchangable), 7.135 (5H,s), 7.367 (5 H, s).

3-(*S*)-BOC-amino-4-phenyl-1-*N*-(p-nitrophenyl) piperazine-2-(*S*)-butanol (1c). A solution of 3-(*S*)-*t*-butoxycarbonyl(BOC) amino-4-phenyl-2-(*R*)-oxirane (McConnell et al., 2003); (1.0 g, 3.1 mmol) in 125mL dry THF was treated with 5.18 g (25 mmol) 1-(4-nitro-phenyl)piperazine. The solution was refluxed for 48 hrs. The mixture was then cooled to room temperature, concentrated under reduced pressure to about one half its volume, and partitioned between ethyl acetate (200 mL) and 5% aqueous sodium potassium tartarate (200 mL) containing 1.0 g NaCl. The organic layer was washed with distilled water (100 mL) and dried over anhydrous magnesium sulfate. The solvent was evaporated under reduced pressure to give a white solid (0.717 g). The crude product was purified by silica gel column chromatography (2.5 cm x 60 cm length) using 60% ethyl acetate/hexanes as the mobile phase to give 0.588 g. TLC (60% ethyl acetate/hexanes) R_f = 0.53. ^{1}H NMR (CDCl3/TMS, 200MHz)ß1.002 (9H, s), 1.639 (2H, t), 2.445 (4H, t), 2.501 (2H, d), 3.081 (4 H, t), 3.892 (1H, m), 4.531 (1H, m), 5.035 (1H, m, exchangable), 7.105 (5H,s), 7.557 (2 H, d), 8.015 (2H, d).

3-(*S*)-amino-4-phenyl-1-*N*-piperazine-2-(*S*)-butanol

dihydrochloride (2a). 0.50 g of 1a was dissolved in 100 mL cold (0°C) 4 M HCl in chloroform. The mixture was stirred at 0°C for 1 hour. Cold diethyl ether (300 mL) was added to induce precipitation of the product. The liquid was decanted, and the precipitant was washed twice with cold ether (150 mL). The crude solid was dissolved in 25 mL methanol and then recrystallized by the addition of 300 mL cold ether. The white solid was again washed twice with cold ether (150 mL) and dried under reduced pressure (0.44 g). TLC (10% ethanol/ethyl acetate) $R_f = 0.38$. ^{1}H NMR (D2O, 60MHz) ß 1.9 (2H, t), 2.9 (8H, m), 3.2 (2H, d), 3.7 (1H, m), 4.4 (1H, m), 7.1 (5H,s).

3-(*S*)-amino-4-phenyl-1-*N*-phenylpiperazine-2-(*S*)-butanol dihydrochloride (2b). 0.55 g of 1b was dissolved in 100 mL cold (0°C) 4 M HCl in chloroform. The mixture was stirred at 0°C for 1 hour. Cold diethyl ether (300 mL) was added to induce precipitation of the product. The liquid was decanted, and the precipitant was washed twice with cold ether (150 mL). The crude solid was dissolved in 20 mL methanol and then recrystallized by the addition of 300 mL cold ether. The white solid was again washed twice with cold ether (150 mL) and dried under reduced pressure (0.43 g). TLC (10% ethanol/ethyl acetate) $R_f = 0.43$. ^{1}H NMR (D_2O, 60MHz) ß 2.0 (4H, t), 2.7 (8H, m), 3.0 (2H, d), 3.8 (1H, m), 4.4 (1H, m), 7.1 (5H,s).

3-(*S*)-amino-4-phenyl-1-*N*-(*p*-nitrophenyl) piperazine-2-(*S*)-butanol dihydrochloride (2c). 0.53 g of 1c was dissolved in 100 mL cold (0°C) 4 M HCl in chloroform. The mixture was stirred at 0°C for 1 hour. Cold diethyl ether (300 mL) was added to induce precipitation of the product. The liquid was decanted, and the precipitant was washed twice with cold ether (150 mL). The crude solid was dissolved in 20 mL methanol and then recrystallized by the addition of 300 mL cold ether. The white solid was again washed twice with cold ether (150 mL) and dried under reduced pressure (0.45 g). TLC (10% ethanol/ethyl acetate) $R_f = 0.26$. ^{1}H NMR (D_2O, 60MHz) ß 2.0 (4H, t), 2.8 (8H, m), 3.1 (2H, d), 3.8 (1H, m), 4.4 (1H, m), 7.1 (5H,s), 7.6 (2H, d), 8.1 (2H, d).

General Procedure for Coupling Cbz-dipeptide to 3-(*S*)-amino-4-phenyl-1-*N*-piperazine (*N*-phenyl piperazine or *N*-*p*-nitrophenylpiperazine)-2-(*S*)-butanol (3ak). A precooled solution (-150°C) of the appropriate carbobenzoxy-dipeptide (Sigma) (0.35 mmol) in 10 mL anhydrous DMF was treated with 56 µL (0.40 mmol) triethyl amine. The mixture was allowed to react at -150°C for 30 minutes and was then treated with 34 µL (0.35 mmol) ethyl chloroformate. The mixture was stirred under N_2 atmosphere for 1 hour at -150°C. A precooled (0°C) solution containing 0.32 mmole of either 2a, 2b, or 2c in 25 mL anhydrous DMF and 125µL (1.0 mmole) triethyl amine was then added to the mixed anhydride of the Cbz-dipeptide. The combined mixture was stirred under N_2 at 0°C for 4 hours, allowed to warm to room temperature, and stirred overnight at room temperature. The mixture was partitioned between the layers of ethyl acetate (250 mL) and 0.01 M aqueous NaOH. The organic layer was removed and saved. The aqueous layer was extracted again with another 250 mL ethyl acetate. The organic layers were pooled, washed with distilled water (100 mL), dried over anhydrous magnesium sulfate, and evaporated under reduced pressure.

3-(*S*)-[Cbz-L-alanyl-L-phenylalanylamino]-4-phenyl-1-*N*-piperazine-2-(*S*)-butanol hydrochloride (3a). 0.162 g. TLC (15% ethanol/ethyl acetate) $R_f = 0.19$. ^{1}H NMR (DMSO-d_6 60 MHz) ß 1.9 (4H, m), 2.5 (2H, d), 2.9 (8 H, m), 3.1 (2H, d), 3.7 (1H, m), 4.6 (3H, m), 5.1 (1H, m), 5.4 (2H, s), 6.8 (5H, s), 7.1 (5H,s), 7.4 (5H, s).

3-(*S*)-[Cbz-L-valyl-L-phenylalanylamino]-4-phenyl-1-*N*-piperazine-2-(*S*)-butanol hydrochloride (3b). 0.125 g. TLC (15% ethanol/ethyl acetate) $R_f = 0.27$. ^{1}H NMR (DMSO-d_6 60 MHz) ß 1.3 (6H, d), 1.9 (4H, t), 2.5 (3, m), 2.7 (2H, d), 2.9 (8H, m), 3.2 (2H, d), 3.7 (1H, m), 4.6 (3H, m), 5.0 (1H, m), 5.4 (2H, s), 6.8 (5H, s), 7.1 (5H,s), 7.4 (5H, s).

3-(*S*)-[Cbz-L-leucyl-L-phenylalanylamino]-4-phenyl-1-*N*-piperazine-2-(*S*)-butanol hydrochloride (3c). 0.131 g. TLC (15% ethanol/ethyl acetate) $R_f = 0.34$. ^{1}H NMR (DMSO-d_6 60 MHz) ß 1.3 (6H, d), 1.8 (4H, m), 1.9 (3H, m), 2.7 (2H, d), 3.0 (8H, m), 3.2 (2H, d), 3.7 (1H, m), 4.6 (3H, m), 5.0 (1H, m), 5.4 (2H, s), 6.9 (5H, s), 7.3 (5H,s), 7.5 (5H, s).

3-(*S*)-[Cbz-L-leucyl-L-leucyl]-4-phenyl-1-*N*-piperazine-2-(*S*)-butanol hydrochloride (3d). 0.145 g. TLC (15% ethanol/ethyl acetate) $R_f = 0.43$. ^{1}H NMR (DMSO-d_6 60 MHz)ß 1.4 (12H, d), 1.7 (6H, m), 1.9 (2H, d), 2.9 (8H, m), 3.1 (2H, d), 3.7 (1H, m), 4.5 (3H, m), 4.8 (1H, m), 5.3 (2H, s), 7.0 (5H, s), 7.3 (5H, s).

3-(*S*)-[Cbz-L-alanyl-L-phenylalanylamino]-4-phenyl-1-*N*-phenylpiperazine-2-(*S*)-butanol hydrochloride (3e). 0.122 g. TLC (18% ethanol/ethyl acetate) $R_f = 0.21$. ^{1}H NMR (DMSO-d_6 60 MHz)ß 1.9 (4H, m), 2.3 (3H, d), 3.0 (10H, d), 3.7 (1H, m), 4.5 (3H, m), 4.8 (1H, m), 5.4 (2H, s), 6.8 (5H, s), 7.1 (5H,s), 7.5 (5H, s), 7.9 (5H, s).

3-(*S*)-[Cbz-L-valyl-L-phenylalanylamino]-4-phenyl-1-*N*-phenylpiperazine-2-(*S*)-butanol hydrochloride (3f). 0.115 g. TLC (18% ethanol/ethyl acetate) $R_f = 0.25$. ^{1}H NMR (DMSO-d_6 60 MHz)ß 1.3 (6H, d), 2.0 (5H,mt), 3.1 (10H, m), 3.8 (1H, m), 4.7 (3H, m), 5.0 (1H, m), 5.3 (2H, s), 6.9 (5H, s), 7.2 (5H,s), 7.6 (5H, s).

3-(*S*)-[Cbz-L-leucyl-L-phenylalanylamino]-4-phenyl-1-*N*-phenylpiperazine-2-(*S*)-butanol hydrochloride (3g). 0.112 g. TLC (18% ethanol/ethyl acetate) $R_f = 0.31$. ^{1}H NMR (DMSO-d_6 60 MHz)ß 1.1 (6H, d), 1.5 (3H, m), 2.0 (4H, d), 3.1 (6H, m), 3.3 (4H, t), 3.8 (1H, m), 4.7 (3H, m), 5.0 (1H, m), 5.3 (2H, s), 6.8 (5H, s), 7.2 (5H,s), 7.6 (5H, s).

3-(*S*)-[Cbz-L-leucyl-L-leucyl]-4-phenyl-1-*N*-phenylpiperazine-2-(*S*)-butanol hydrochloride (3h). 0.112 g. TLC (18% ethanol/ethyl acetate) $R_f = 0.31$. ^{1}H NMR (DMSO-d_6 60 MHz) ß 1.3 (12H, d), 1.6 (6H, m), 2.0 (2H, d),

0 (6H, m), 3.3 (4H, t), 3.7 (1H, m), 4.7 (3H, m), 5.0 (1H, ı), 5.3 (2H, s), 6.9 (5H, s), 7.3 (5H,s), 7.6 (5H, s).

3-(*S*)-[Cbz-L-alanyl-L-phenylalanylamino]-4-phenyl--*N*-(*p*-nitrophenyl)piperazine-2-(*S*)-butanol ydrochloride (3i). 0.102 g. TLC (20% ethanol/ethyl cetate) $R_f = 0.26$. ^{1}H NMR (DMSO-d6 60 MHz)ß 2.0 (4H, n), 2.2 (3H, d), 3.0 (6H, d), 3.5 (4H, t), 3.7 (1H, m), 4.5 (3H, n), 4.8 (1H, m), 5.4 (2H, s), 6.8 (5H, s), 7.1 (5H,s), 7.5 (5H, s), 8.0 (2H, d), 8.4 (2H, d).

3-(*S*)-[Cbz-L-valyl-L-phenylalanylamino]-4-phenyl-1-*N*-(*p*-nitrophenyl)piperazine-2-(*S*)-butanol hydrochloride (3j). 0.122 g. TLC (20% ethanol/ethyl acetate) $R_f = 0.29$. ^{1}H NMR (DMSO-d6 60 MHz) ß 1.3 (6H, d), 2.0 (5H, m), 3.0 (6H, m), 3.5 (4H, t), 3.8 (1H, m), 4.7 (3H, m), 5.0 (1H, m), 5.3 (2H, s), 6.9 (5H, s), 7.2 (5H,s), 7.6 (5H, s), 8.0 (2H, d), 8.4 (2H, d).

3-(*S*)-[Cbz-L-leucyl-L-phenylalanylamino]-4-phenyl-1-*N*-(*p*-nitrophenyl)piperazine-2-(*S*)-butanol hydrochloride (3k). 0.108 g. TLC (20% ethanol/ethyl acetate) $R_f = 0.33$. ^{1}H NMR (DMSO-d6 60 MHz) ß 1.1 (6H, d), 1.5 (3H, m), 2.0 (4H, d), 3.1 (6H, m), 3.4 (4H, t), 3.8 (1H, m), 4.7 (3H, m), 5.0 (1H, m), 5.3 (2H, s), 6.8 (5H, s), 7.2 (5H,s), 7.5 (5H, s), 8.1 (2H, d), 8.5 (2H, d).

3-(*S*)-[Cbz-L-leucyl-L-leucyl]-4-phenyl-1-*N*-(*p*-nitrophenyl)piperazine-2-(*S*)-butanol hydrochloride (3l). 0.912 g. TLC (20% ethanol/ethyl acetate) $R_f = 0.35$. ^{1}H NMR (DMSO-d6 60 MHz) ß 1.3 (12H, d), 1.6 (6H, m), 2.0 (2H, d), 3.0 (6H, m), 3.5 (4H, t), 3.7 (1H, m), 4.7 (3H, m), 5.0 (1H, m), 5.3 (2H, s), 6.9 (5H, s), 7.3 (5H,s), 7.6 (5H, s), 8.1 (2H, d), 8.5 (2H, d).

General Procedure for Preparation of 3-(*S*)-[Acetyldipeptide-amino]-4-phenyl-1-*N*-piperazine (or phenylpiper azine or *p*-nitrophenylpiperazine)-2-(*S*)-butanol hydro chloride (4a-l). A solution of the carbobenzy protected compound (3a-l); (0.20 mmol) in 250 mL methanol and 1 mL 0.01 M aqueous HCl was treated with 0.050 g pre-moistened 10% Pd-C to form a slurry in a 3 neck flask. H_2 gas was bubbled (1 atm) through the rapidly stirring mixture at room temperature for 3 hours. The mixture was then filtered to remove the catalyst, and the solvent was evaporated under reduced pressure. The crude amine hydrochloride was dissolved in 15 mL dimethyl sulfoxide (DMSO) and treated with 125 µL (0.10 mole) triethyl amine. The mixture was stirred at room temperature for 30 minutes. Acetic anhydride (95 µL, 1.0 mmol) was added, and the mixture was stirred overnight at room temperature. Cold diethyl ether (250 mL) was added to precipitate the product. The liquid was decanted, and the white solid was washed three times with cold ether (200 mL). The crude product was purified by Sephadex LH-20 column chromatography (column size 5 cm dia. x 80 cm) using methanol as the mobile phase.

3-(*S*)-[Acetyl-L-alanyl-L-phenylalanylamino]-4-phenyl-1-*N*-piperazine-2-(*S*)-butanol hydrochloride (4a). 0.56 g. TLC (1-butanol/H_2O/acetic acid, 15/2/1) R_f=0.46. ^{1}H NMR (methanol-d4 200 MHz)ß 2.071 (4H, d), 2.165 (3H, d), 2.279 (3H, s), 2.462 (4H, m), 2.992 (4H, t), 3.189 (2H, d), 3.465 (4H, t), 3.716 (1H, m), 4.610 (3H, m), 5.081 (1H, m), 6.949 (5H, s), 7.210 (5H,s).

3-(*S*)-[Acetyl-L-valyl-L-phenylalanylamino]-4-phenyl-1-N-piperazine-2-(*S*)-butanol hydrochloride (4b). 0.49 g. TLC (1-butanol/H_2O/acetic acid, 15/2/1) R_f= 0.51. ^{1}H NMR (methanol-d_4 200 MHz)ß 1.689 (6H, d), 1.989 (1H, m), 2.087 (4H, d), 2.266 (3H, s), 2.458 (4H, m), 3.099 (2H, d), 3.477 (4H, t), 3.720 (1H, m), 4.613 (3H, m), 4.999 (1H, m), 6.959 (5H, s), 7.203 (5H,s).

3-(*S*)-[Acetyl-L-leucyl-L-phenylalanylamino]-4-phenyl-1-N-piperazine-2-(*S*)-butanol hydrochloride (4c). 0.46 g. TLC (1-butanol/H_2O/acetic acid, 15/2/1) R_f = 0.66. ^{1}H NMR (methanol-d_4 200 MHz)ß 1.298 (6H, d), 1.690 (2H, m), 1.990 (1H, m), 2.077 (4H, t), 2.199 (3H, s), 2.459 (4H, m), 3.102 (2H, d), 3.479 (4H, t), 3.721 (1H, m), 4.614 (3H, m), 4.993 (3H, m), 6.960 (5H, s), 7.213 (5H,s).

3-(*S*)-[Acetyl-L-leucyl-L-leucyl]-4-phenyl-1-N-piperazine-2-(*S*)-butanol hydrochloride (4d). 0.49 g. TLC (1-butanol/H_2O/acetic acid, 15/2/1) R_f = 0.71. ^{1}H NMR (methanol-d_4 200 MHz)ß 1.308 (12H, d), 1.694 (4H, m), 1.990 (2H, m), 2.077 (2H, t), 2.201 (3H, s), 2.461 (4H, m), 3.109 (2H, d), 3.519 (4H, t), 3.724 (1H, m), 4.634 (3H, m), 5.013 (3H, m), 6.977 (5H, s), 7.243 (5H,s).

3-(*S*)-[Acetyl-L-alanyl-L-phenylalanylamino]-4-phenyl-1-N-phenylpiperazine-2-(*S*)-butanol hydrochloride(4e). 0.61 g. TLC (1-butanol/H_2O/acetic acid, 15/2/1) R_f = 0.58. ^{1}H NMR (methanol-d_4 200 MHz)ß 1.999 (4H, d), 2.155 (3H, d), 2.315 (3H, s), 3.156 (2H, d), 3.257 (4H, d), 3.431 (4H, t), 3.715 (1H, m), 4.615 (3H, m), 4.998 (3H, m), 6.999 (5H, s), 7.242 (5H,s), 8.045 (5H, s).

3-(*S*)-[Acetyl-L-valyl-L-phenylalanylamino]-4-phenyl-1-N-phenylpiperazine-2-(*S*)-butanol hydrochloride (4f). 0.74 g. TLC (1-butanol/H_2O/acetic acid, 15/2/1) R_f = 0.63. ^{1}H NMR (methanol-d_4 200 MHz)ß 1.652 (6H, d), 2.009 (4H, d), 2.160 (1H, m), 2.396 (3H, s), 3.180 (2H, d), 3.264 (4H, d), 3.401 (4H, t), 3.722 (1H, m), 4.625 (3H, m), 4.999 (3H, m), 6.989 (5H, s), 7.301 (5H,s), 8.066 (5H, s).

3-(*S*)-[Acetyl-L-leucyl-L-phenylalanylamino]-4-phenyl-1-N-phenylpiperazine-2-(*S*)-butanol hydrochloride (4g). 0.78 g. TLC (1-butanol/H_2O/acetic acid, 15/2/1) R_f = 0.69. ^{1}H NMR (methanol-d_4 200 MHz)ß 1.409 (12H, d), 1.650 (6H, m), 2.020 (4H, d), 2.297 (3H, s), 3.162 (2H, d), 3.255 (4H, d), 3.411 (4H, t), 3.725 (1H, m), 4.622 (3H, m), 4.997 (3H, m), 6.996 (5H, s), 7.300 (5H,s), 8.088 (5H, s).

3-(*S*)-[Acetyl-L-leucyl-L-leucyl]-4-phenyl-1-N-phenylpiperazine-2-(*S*)-butanol hydrochloride (4h). 0.69 g. TLC (1-butanol/H_2O/acetic acid, 15/2/1) R_f = 0.72. ^{1}H NMR (methanol-d_4 200 MHz)ß 1.308 (12H, d), 1.694 (4H, m), 1.990 (2H, m), 2.077 (2H, t), 2.300 (3H, s), 3.261 (2H, d),

3.375 (4H, d), 3.509 (4H, t), 3.724 (1H, m), 4.634 (3H, m), 5.013 (3H, m), 6.977 (5H, s), 7.243 (5H,s), 8.097 (5H, s).

3-(*S*)-[Acetyl-L-alanyl-L-phenylalanylamino] -4-phenyl-1-*N*-(p-nitrophenyl)piperazine-2-(*S*)-butanol hydrochloride (4i). 0.45 g. TLC (1-butanol/H_2O/acetic acid, 15/2/1) $R_f = 0.42$. ^1H NMR (methanol-d_4 200 MHz)ß 1.999 (4H, d), 2.155 (3H, d), 2.315 (3H, s), 3.156 (2H, d), 3.257 (4H, d), 3.491 (4H, t), 3.715 (1H, m), 4.615 (3H, m), 4.998 (3H, m), 6.999 (5H, s), 7.242 (5H,s), 8.055 (2H, d), 8.444 (2H, d).

3-(*S*)-[Acetyl-L-valyl-L-phenylalanylamino]-4-phenyl-1-*N*-(*p*-nitrophenyl)piperazine-2-(*S*)-butanol hydrochloride (4j). 0.55 g. TLC (1-butanol/H_2O/acetic acid, 15/2/1) $R_f = 0.43$. ^1H NMR (methanol-d_4 200 MHz)ß 1.652 (6H, d), 2.009 (4H, d), 2.160 (1H, m), 2.396 (3H, s), 3.180 (2H, d), 3.264 (4H, d), 3.500 (4H, t), 3.722 (1H, m), 4.625 (3H, m), 4.999 (3H, m), 6.989 (5H, s), 7.301 (5H,s), 8.077 (2H, d), 8.467 (2H, d) .

3-(*S*)-[Acetyl-L-leucyl-L-phenylalanylamino]-4-phenyl-1-*N*-(*p*-nitrophenyl)piperazine-2-(*S*)-butanol hydrochloride (4k). 0.56 g. TLC (1-butanol/H_2O/acetic acid, 15/2/1) $R_f = 0.45$. ^1H NMR (methanol-d_4 200 MHz)ß 1.409 (12H, d), 1.650 (6H, m), 2.020 (4H, d), 2.297 (3H, s), 3.162 (2H, d), 3.255 (4H, d), 3.521 (4H, t), 3.725 (1H, m), 4.622 (3H, m), 4.997 (3H, m), 6.996 (5H, s), 7.300 (5H,s), 8.091 (2H, d), 8.444 (2H, d).

3-(*S*)-[Acetyl-L-leucyl-L-leucyl]-4-phenyl-1-*N*-(*p*-nitrophenyl)piperazine-2-(*S*)-butanol hydrochloride (4l). 0.39 g. TLC (1-butanol/H_2O/acetic acid, 15/2/1) $R_f = 0.54$. ^1H NMR (methanol-d_4 200 MHz) ß 1.309 (12H, d), 1.695 (4H, m), 1.991 (2H, m), 2.078 (2H, t), 2.302 (3H, s), 3.261 (2H, d), 3.344 (4H, d), 3.619 (4H, t), 3.725 (1H, m), 4.635 (3H, m), 5.011 (3H, m), 6.987 (5H, s), 7.244 (5H,s), 8.099 (2H, d), 8.455 (2H, d).

Cathepsin D Assay. The potency of compounds 4a-l was measured as inhibitors of the cathepsin D hydrolysis of human hemoglobin (Sigma), and the results are presented in Table 1. Inhibition of cathepsin D was measured (Yasuda et al., 1999) by the following method: 225 µL of the inhibitor of appropriate concentration in sodium formate-formic acid buffer (0.50 M, pH 3.2) and 250 µL of a 0.5% hemoglobin solution were mixed and incubated at 450°C for 20 minutes. Human cathepsin D (Sigma), 25 µL of a 1.0 µg/mL solution, was added and mixed for a total enzyme concentration of 1.1×10^{-9} M. The mixture was incubated at 450°C for 1.5 hours. The reaction was quenched by the addition of 1.0 mL cold 0.3 M trichloroacetic acid. The solution was mixed thoroughly and then chilled in ice for 30 min to allow separation of precipitated protein. The mixture was centrifuged and warmed to 250°C. The liquid was decanted into a quartz cuvette and the absorbance measured at 280 nm. The absorbance of a blank containing no enzyme was subtracted from the reading. The inhibition of the enzyme activity was measured 4 times at 5 or more inhibitor concentrations. The average absorbance of each inhibitor concentration was utilized in the calculations of the IC_{50} values. All absorbances were within #0.002 standard deviations from the mean for a given inhibitor concentration. The standard error for the linear regression plots was in each case less than 3%. A plot of percent inhibition versus the log of the inhibitor concentration provided a value for the 50% inhibition concentration (IC_{50}). All plots were linear through the 50% inhibition value and have slopes ranging from 22 to 40. The apparent inhibition constants, $Ki_{(app)}$, were calculated (Evans et al., 1985; McConnell et al., 1991) as $Ki_{(app)} = IC_{50} - 0.5[E_0]$, where $[E_0]$ is the enzyme concentration.

Results and Discussion

Our synthetic plan of the potential cathepsin D inhibitors involved two phases: (a) preparation of the protected hydroxyethyl amine isostere portion and (b) condensation and deblocking of the peptide and non-peptide portions. The hydroxyethyl amine isosteres were prepared from a tert-butoxycarbonyl-(BOC) chiral amino aminoalkyl epoxide reported earlier (McConnell et al., 2003). Similar chiral aminoalkyl epoxides (with opposite stereochemistry) have been used successfully in the preparation of several HIV-1 aspartyl protease inhibitors with hydroxyethyl amine isosteres (Fassler et al., 1996; Barrish et al., 1994). The 2S,3S epoxide is utilized to prepare HIV-1 protease inhibitors with the desired *R*-hydroxyethyl amine isostere (Fassler et al., 1996; Barrish et al., 1994). However, since the *S*-hydroxyethyl amine isostere is reported to be the more active isomer for cathepsin D inhibition (Kick et al., 1997), we utilized the 2*R*,3*S* protected amino epoxide in our synthesis (Scheme 1). Either piperazine, *N*-phenyl piperazine, or *N*-(*p*-nitrophenyl) piperazine was used as a nucleophile in the preparation of the cyclized tertiary amines. The BOC protecting group was removed from the primary amine with non-aqueous acid (4 M HCl in chloroform).

In the second phase of our synthesis, the Cbz-protected dipeptide was condensed with ethyl chloro formate and then reacted with the basified primary amine of the hydroxyethyl amine isostere portion (Scheme 2). The Cbz protecting group of the resulting compound was then removed and replaced with an acetyl group. The final product was purified by sephadex HP chromatography and characterized by TLC and ^1H NMR.

The twelve synthetic compounds were screened for their cathepsin D inhibition by a spectrophotometric assay (Yasuda et al., 1999) of hemoglobin hydrolysis (Table 1). Modifications in the ring of the hydroxy ethyl tertiary amine appears to have affected the potency of the inhibitors. Those compounds with a phenyl piperazine or *N*-(*p*-

nitrophenyl) piperazine ring (4e-l) show a slightly better cathepsin D inhibition than the compounds without a phenyl group attached to the piperazine (4a-d). Also, the variation in the *N*-terminal amino acid side appears to affect the cathepsin D inhibition. Compounds with an alanine in the P_3 position, were somewhat less effective inhibitors than those compounds with a valine or leucine in the P_3 position. These are general trends observed in the initial screening.

Conclusions

Our synthetic route shows a great deal of promise for the future synthesis of similar hydroxyethyl amine isosteres. The initial screening shows our synthetic compounds to be potent inhibitors of cathepsin D activity. Since a major method for developing new drug candidates is through random screening of large libraries of compounds previously shown to have desirable biological activity (Berwowitz and Katzung, 2001), the inhibitors described in this paper, along with our earlier work, are important contributions to the development of a library of cathepsin D inhibitors. Although many of our cathepsin D inhibitors will be limited in their therapeutic usefulness, due to potential limitations in bioavailability, with a large enough pool of active compounds it is possible that a few of these inhibitors may someday prove to be promising drug candidates for the treatment of cancer. Detailed kinetic data of the synthetic inhibitors will be determined by more sensitive fluorometric techniques (Pimenta et al., 2001) to determine the inhibition mechanism.

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Table 1. Synthetic Inhibitors

No.	Compound Name	IC_{50}, M	$Ki_{(app)}$, M
4a	3(*S*)-[Acetyl-L-alanyl-L-phenylalanylamino]-4-phenyl-1-*N*-piperazine-2(*S*)-butanol	5.0×10^{-7}	4.28×10^{-7}
4b	3(*S*)-[Acetyl-L-valyl-L-phenylalanylamino]-4-phenyl-1-*N*-piperazine-2(*S*)-butanol	3.0×10^{-7}	3.38×10^{-7}
4c	3(*S*)-[Acetyl-L-leucyl-L-phenylalanylamino]-4-phenyl-1-*N*-piperazine-2(*S*)-butanol	1.8×10^{-7}	1.68×10^{-7}
4d	3(*S*)-[Acetyl-L-leucyl-L-leucylamino]-4-phenyl-1-*N*-piperazine)-2(*S*)-butanol	9.5×10^{-7}	8.28×10^{-7}
4e	3(*S*)-[Acetyl-L-alanyl-L-phenylalanylamino]-4-phenyl-1-*N*-(*N*-phenylpiperazine)-2(*S*)-butanol	1.3×10^{-7}	1.28×10^{-7}
4f	3(*S*)-[Acetyl-L-valyl-L-phenylalanylamino]-4-phenyl-1-*N*-(*N*-phenylpiperazine)-2(*S*)-butanol	3.5×10^{-7}	3.38×10^{-7}
4g	3(*S*)-[Acetyl-L-leucyl-L-phenylalanylamino]-4-phenyl-1-*N*-(*N*-phenylpiperazine)-2(*S*)-butanol	1.8×10^{-7}	1.68×10^{-7}
4h	3(*S*)-[Acetyl-L-leucyl-L-leucylamino]-4-phenyl-1-*N*-(*N*-phenylpiperazine)-2(*S*)-butanol	3.5×10^{-7}	1.28×10^{-7}
4i	3(*S*)-[Acetyl-L-alanyl-L-phenylalanylamino]-4-phenyl-1-*N*-(*N*-*p*-NO_2-phenylpiperazine)-2(*S*)-butanol	5.5×10^{-7}	5.49×10^{-7}
4j	3(*S*)-[Acetyl-L-valyl-L-phenylalanylamino]-4-phenyl-1-*N*-(*N*-*p*-NO_2-phenylpiperazine)-2(*S*)-butanol	4.5×10^{-8}	4.38×10^{-8}
4k	3(*S*)-[Acetyl-L-leucyl-L-phenylalanylamino]-4-phenyl-1-*N*-(*N*-*p*-NO_2-phenylpiperazine)-2(*S*)-butanol	4.6×10^{-8}	4.48×10^{-8}
4l	3(*S*)-[Acetyl-L-leucyl-L-leucylamino]-4-phenyl-1-*N*-(*N*-p-NO_2-phenylpiperazine)-2(*S*)-butanol	7.7×10^{-7}	7.69×10^{-7}

Spectrophotometric assays (A_{280}) at pH 3.2 of cathepsin D hydrolysis of hemoglobin. Apparent inhibition constants, $K_{i\ (app)}$, were calculated [29] as $K_{i\ (app)} = IC_{50} - 0.5[E_0]$, where $[E_0]$ is the initial enzyme concentration.

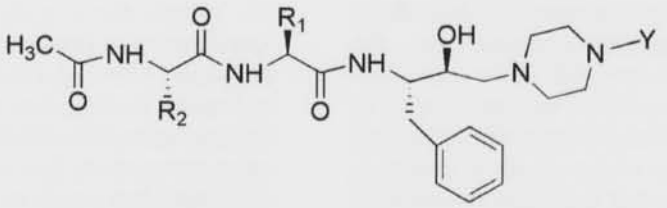
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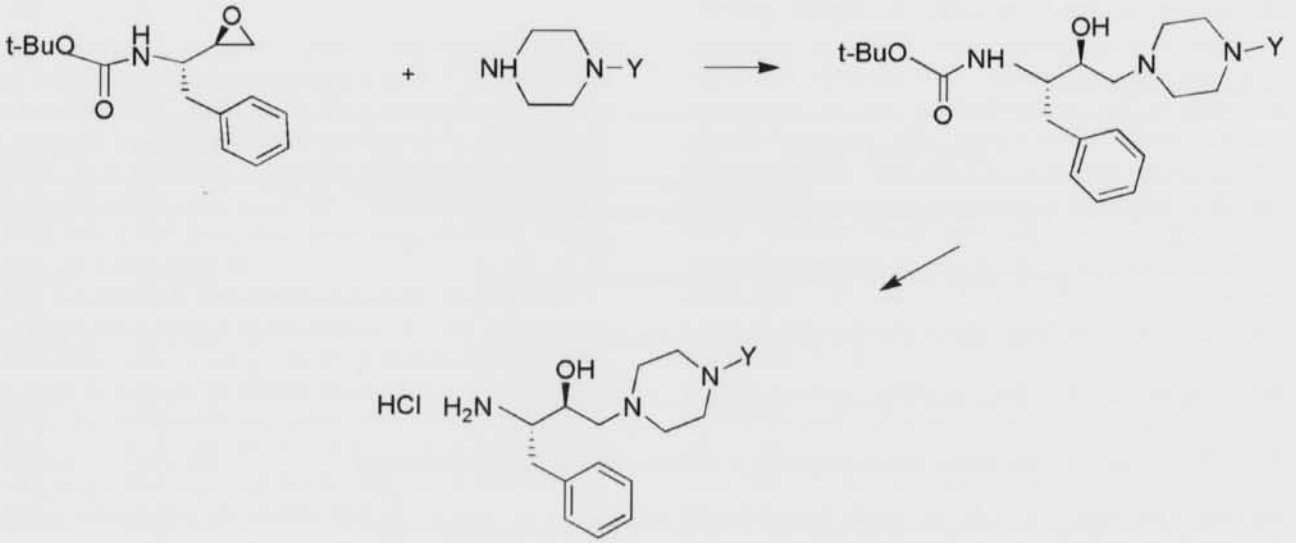
Fig. 1.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Y = H,
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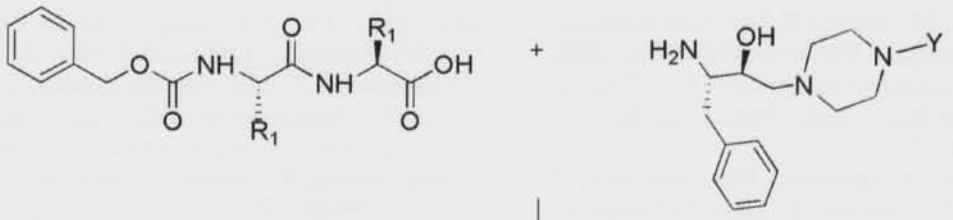
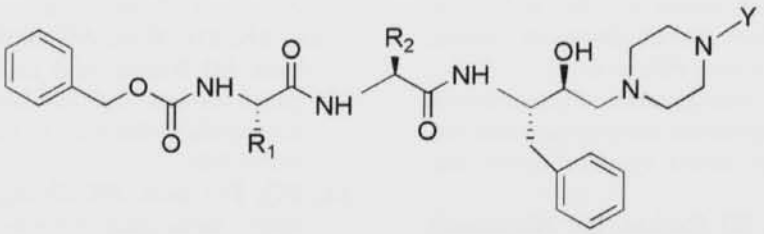
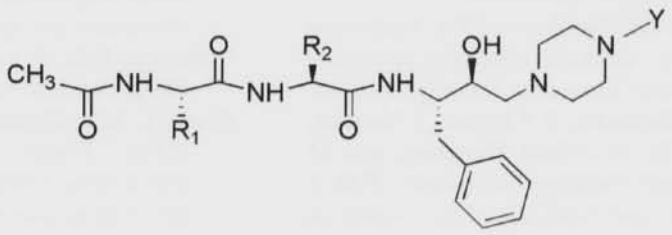
Sch. 1.



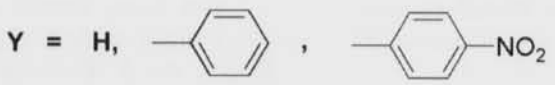
2 a-c



3 a-l



4 a-l



Sch. 1.

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Identifying the Factors Distinguishing Timber Sales on Industrial and Non-industrial Private Forest Lands in Arkansas

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Abstract

Although forests provide a wide variety of products and services, timber still continues to be the most valued forest product in the marketplace. More than two-third of the nation's forests are under private control, some are owned by industries (about 10%) while a much larger portion (about 59%) is owned by individuals. This study investigates the differences between timber sales offered by industrial and non-industrial ownerships. A test of means revealed that there is a significant difference between per hectare bid for these 2 types of sales. A logistic regression model was then estimated to identify important factors characterizing this difference. Results indicated that industrial forests were more likely to obtain higher bids. They were also more likely to have shorter contract lengths. Industrial ownerships were found to be more likely to have clearcuts. However, they had a higher likelihood of restricting harvesting during wet-weather conditions. Forest industries were also found to be less likely to have pulpwood for sale than non-industrial private owners.

Introduction

Appropriately called the "Natural State," Arkansas has approximately 7.6 million hectares of forests within her borders. These resources, along with the associated forest sector industries, provide significant contributions to the state's economy. In 2002, these forests produced 21 million cubic meters of industrial roundwood (U.S. Forest Service, 2004). In addition to these tangible economic benefits, Arkansas' forests also provide various recreational and environmental services. Some of these services, such as camping, fishing, and hunting, also provide economic benefits to the state's economy (Williams and Kluender, 1997).

The study of economics is primarily concerned with efficient allocation of goods and services to competing demands within the society. In a market-based economy, this allocation is achieved through the operation of the market. The market operates through numerous transactions that reflect the exchange of goods and services available within the marketplace. This transfer of goods and services is also a transfer of property rights. The "bundle of rights" to a good is relinquished through its sale in the marketplace, resulting in a market transaction. Market transactions, however, often involve costs known as transaction costs, which include items such as the costs of contracting.

An estimated 159 million ha of forests in the U.S. are privately owned. About 59% of all non-government forest owners are non-industrial private forest (NIPF) landowners (Birch, 1996). NIPF lands currently supply about half of the country's demand for wood fiber. This number is expected to rise to about 60% by the year 2030 (Haines, 1995; Harrell, 1989). This increase is primarily due to a sharp decline in supply from public forests caused by a general shift in public forest management due to environmental concerns and production of amenity goods (Smith et al., 2001; Mehmood and Zhang, 2001; Mehmood and Zhang, 2002). In Arkansas, the proportion of NIPF landowners within total number of forest landowners has traditionally been very similar to the national average. Additionally, about a quarter of the state's forests are owned by the forest products industry (Birch, 1996).

These two types of private forest ownerships, namely NIPF and industrial, have significantly different characteristics. Industrial forests are usually owned by public companies and are therefore intensively managed for wood products in order to maximize profit for the shareholders. This focus on profit maximization leads to management efficiency and minimization of waste. Non-industrial forests, however, are owned by numerous individuals and occur in a wide variety of parcel sizes ranging from a few ha to thousands of hectares. These owners also have a wide variety of management objectives including timber, recreation, and aesthetics. Due to this diversity of objectives and a widespread lack of knowledge regarding forest management, the level of efficiency in the management of NIPF forests is low. This low level of efficiency leads to waste of scarce resources. Existing literature in NIPF management is a testament to this fact.

Our objective was to investigate the differences between timber sales on private industry-owned and non-industry-owned forest lands in Arkansas. In order to achieve this objective, we identified the significant factors distinguishing timber sales from these two types of private ownerships. Knowledge of such factors may provide some insights on possible efforts toward gaining economic efficiency in NIPF forest management.

Materials and Methods

For this study, sealed-bid, timber-sale data from the state of Arkansas were used. The data were collected from a variety of sources across the state including private consultants, the Arkansas Forestry Commission, and the U.S. Forest Service. The materials collected from these sources included bid abstracts, prospectus, and timber-sale contracts. Once these materials were collected, the necessary information contained within these documents was identified and compiled in a spreadsheet. The final data set contained information on 625 timber sales that occurred in 38 counties around the state (Dahal and Mehmood, 2005). For this study, however, only the data from industrial and NIPF lands were used, resulting in a sample size of 436 observations.

A two-sample test of means (t-test) was first performed on the data to determine if there is a significant difference in per hectare bid price for timber sold on these two types of ownerships. The data were then used in a binomial logistic regression model in order to identify the important distinguishing factors of timber sales on industrial and non-industrial private forest ownerships.

The specific model estimated through logistic regression was as follows:

$$OWN = f(BIDPERHA, CLENGTH, NOOFBIDS, HVSTTYPE, WETWEATH, PSTPERHA, PPWPERHA, HSTPERHA, HPWPERHA)$$

The dependent variable, OWN, represents the type of ownership. This is a binary variable that takes the value of one when the sale is on industry-owned land, and is zero otherwise. The first independent variable, BIDPERHA, represents bid price per hectare of forest from the winning bid. Since industrial forests are relatively more intensively and efficiently managed, we expect this variable to have a positive sign. CLENGTH is the length of the contract for harvesting timber, expressed in number of days. Again, since the level of efficiency on industrial ownerships is high, the length of the contract in those cases is expected to be short. Therefore, we expect this variable to have a negative sign.

The next explanatory variable, NOOFBIDS, represent the number of bids received for the sale. It is difficult to form an *a priori* expectation for this variable since number of bids could be a function of any number of other factors. However, conventional wisdom would suggest that since timber from industrial forests may be perceived as high quality due to intensive management, these sales are likely to receive a higher number of bids. HVSTTYPE represents the method of harvesting employed in each timber sale. This is a dummy variable that takes the value of one if any type of selection harvest is employed and zero when the stands are clearcut. Since selection harvest imposes a cost on the timber buyer due to the extra time and effort required in harvesting and because of the relative difficulty of moving logging equipment around the tract, industrial owners are more likely to opt for clearcuts. Therefore, we expect this variable to have a negative sign.

The variable WETWEATH represents whether or not the timber sale has a restriction on logging during wet weather conditions. Logging during wet conditions has a higher likelihood of causing soil erosion and impairment of water quality. If timber is not harvested during wet weather, it imposes a cost on the buyer. This would imply that strictly on the basis of economic efficiency, industrial owners would be more likely to allow wet-weather logging. However, there are voluntary policies in place to prevent such damage, known as the Best Management Practices (BMPs). Due to their commitment to the Sustainable Forestry Initiative (SFI), forest products industries adhere to strict guidelines regarding logging in wet weather conditions and implement BMPs to prevent soil and water damage. Therefore, we expect industrial timber sales to forbid wet weather logging. Consequently, we expect a negative sign for this variable.

The following four variables, PSTPERHA, PPWPERHA, HSTPERHA, and HPWPERHA represent the amount of pine sawtimber, pulpwood, hardwood sawtimber, and pulpwood in the sale, respectively. These variables are included to determine if the types of primary wood products available for sale are different by ownership. These variables also have quality implications for forest products from different types of ownerships. In general, because of aforementioned management efficiency reasons, products from industrial ownerships are more likely to be of higher quality. This implies that industrial ownerships are more likely to have a larger amount of sawtimber for sale rather than pulpwood since sawtimber is the higher valued product. Additionally, the market for pulpwood thinnings in Arkansas is not as well developed as some states in the Southeast such as Georgia and Alabama. Therefore, we expect the two sawtimber variables to have positive signs and the two pulpwood variables to have negative signs.

Since the dependent variable is binary, a logistic regression procedure is used to estimate the model. In binomial logit models, probabilities are assigned for each of

the two possible outcomes for the dependent variable (i.e. industrial and non-industrial ownerships).

In this case, these probabilities are:

$$P(Y_i = 1) = P_i = \frac{e^{X_i\beta}}{1 + e^{X_i\beta}} \quad \text{and}$$

$$P(Y_i = 0) = 1 - P_i = \frac{1}{1 + e^{X_i\beta}}.$$

Where P_i represents the probability that a timber sale took place on industrial land, and X_i_ is a standard regression notation representing the right side of a regression model in matrix terms. Unlike ordinary least squares (OLS) regression, the logistic procedure involves estimating the regression parameters by maximizing a likelihood function. The likelihood function that is maximized can be expressed as

$$L = \prod_{i=1}^{n} P_i^{y_i} (1 - P_i)^{(1-y_i)}.$$

The coefficient estimates in logistic regression do not have the same implication of per unit impact by each individual independent variable on the dependent variable as in the OLS case. In order to draw such implications parallel to the OLS case, marginal effects for each independent variable are calculated as follows,

$$\frac{\delta P_i}{\delta X_i} = P_i(1 - P_i)\beta.$$

Results and Discussion

The t-test of mean bid price per ha was performed using SAS version 8.2. Results of this test along with some descriptive statistics are presented in Table 1. T-test results revealed that there indeed is significant difference between winning bid prices per hectare for timber sales on industrial and non-industrial private forest ownerships. The t-test was performed assuming that the variances of these two samples were not equal.

Table 2, on the other hand, presents the estimates of the logistic regression model. The log-likelihood test (analogous to F-test in the OLS case) was significant at the 99% confidence level. There were no large correlations among the variables in the model. Standard tests for specification errors did not reveal the presence of such errors. Most of the variables had expected signs. The only exceptions were the two sawtimber volume variables. Contrary to our *a priori* expectations, these variables had negative signs. However, they were not significant; therefore no statistical implications could be drawn for these two variables. Additionally, the variable representing the number of bids received in each sale was also not significant.

The variable representing winning bid price per hectare was positive and significant at the 99% confidence level. This implied that industrial landowners have a higher probability of receiving a higher bid price for their timber. As mentioned earlier, forests under industrial ownership are likely to be comparatively more efficiently managed with the help of the best available knowledge on forest management techniques. Management of these forests is optimized for timber production. Therefore, these forests are more likely to have better quality products. Additionally, industrial landowners have up-to-date market information and have better access to the market. Because of these advantages over non-industrial ownerships, it is not a surprise that industrial owners would be able to obtain higher revenues from timber sales.

The variable contract length was negative and significant at 99% confidence level implying that industrial ownerships are more likely to have a shorter contract length for timber removal. This result is also a function of the higher efficiency on industrial lands. Industrial owners, due to their expertise and current information on the forest products market, are better able to negotiate with timber buyers so that the timber can be harvested in the shortest possible length of time.

The method-of-harvest variable was significant at 99% and had a negative sign. Since this variable took a value of one if some type of selection harvest was employed, the sign indicated that industrial landowners were more likely to employ clearcutting rather than selection harvests. Selection harvesting has significant cost implications associated with it. For instance, it is more time consuming to selectively harvest a site. It is also difficult to move harvesting equipment in such a site. Therefore, selection harvest requires additional time in planning and execution and a considerable amount of additional resources, thereby contributing to the cost of harvesting. Therefore, it is expected that a profit maximizing firm would minimize cost by employing clearcuts where possible. It should be noted here, however, that by the inclusion of this choice of harvest method variable we do not intend to make any implication regarding the health and soundness of forest management. Rather, the only intended implication is purely economic in

nature. Our basic argument is that since clearcutting is the least-cost method of harvest, profit-maximizing firms are more likely to choose clearcutting over other methods. The nature of management (even or uneven aged), however, could have some impact on the choice of harvesting method.

As expected, industrial landowners were more likely to minimize logging during wet weather conditions. The variable was significant at the 99% confidence level. This is indicative of the forest products industry's (at least those that are members of the American Forest and Paper Association, which include almost all of the large forest products firms) commitment to the Sustainable Forestry Initiative that they would strictly adhere to state BMP prescriptions. We weather logging also increases the cost of road maintenance. Therefore, profit-maximizing firms are likely to avoid such costs. Both pine and hardwood pulpwood volumes were negative and were significant at the 95% confidence level. These variables represent the amount of pulpwood that was being offered for sale. The results indicated that industrial landowners were less likely to have a large amount of pulpwood for sale. Pulpwood is a significantly lower-valued product compared to sawtimber. Profit maximizing firms are therefore expected to opt for the higher valued product. Additionally, pulpwood prices have been very low in the recent years. This result, therefore, may also be indicative of forest landowners' response to market conditions.

Conclusions

Results from the logistic regression estimates identified the important factors distinguishing timber sales on industrial forest lands from non-industrial private forest lands. The results indicated that efficiency, both in forest management and business decisions, was the key factor. Due to this efficient management, industrial landowners are more likely to minimize costs and make better economic decisions regarding product and timing. Information is an important component in this regard. Providing better information on forest management techniques and market conditions to non-industrial private forest landowners would be an important step in making these lands more productive and efficiently managed. While NIPF landowners would still have to overcome a host of other obstacles such as differences in financial investment, and access to markets; this could still help in reducing the waste of natural resources and in opening more land to non-timber uses and services from forests.

Table 1. Descriptive statistics and t-test results.

Group	N	Mean bid/ha	St. Dev.	St. Error
Non-indus.	211	2918.18	1978.80	136.23
Indus	225	6881.61	2411.50	160.77

Hypotheses

Null: $\mu_{Non\text{-}indus} - \mu_{Indus} = 0$

Alternative: $\mu_{Non\text{-}indus} - \mu_{Indus} \neq 0$

t-statistic	DF	*P*-value
-18.81	426	< 0.0001

Table 2. Estimates of the binomial logistic regression.

Variable	Coefficient (*t*-statistic)	Marginal effect (st. error)	Mean of variable
Constant	-1.20***		
	(-5.04)		
BIDPERHA	0.0004***	0.00009	4963.52
	(2.98)	(0.00003)	
CLENGTH	-0.004***	-0.008	533.71
	(-6.03)	(0.001)	
NOOFBIDS	-0.02	-0.004	6.82
	(-0.19)	(0.02)	
HVSTTYPE	-3.06***	-0.73	0.35
	(-3.66)	(0.19)	
WETWEATH	-2.48***	-0.59	0.38
	(-3.89)	(0.15)	
PSTPERHA	-0.000008	-0.000002	13438.65
	(-0.32)	(0.000006)	
PPWPERHA	-0.0006**	-0.0002	420.05
	(-2.56)	(0.00006)	
HSTPERHA	-0.00009	-0.00002	797.44
	(-0.51)	(0.00004)	
HPWPERHA	-0.0006**	-0.0001	805.83
	(-2.46)	(0.00006)	
Log-likelihood		-50.15	
Restrict. log-likelihood		-301.99	
Chi-square		503.67***	
No. of Observations		436	

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Distribution and Status of the Kiamichi Shiner, *Notropis ortenburgeri* Hubbs (Cyprinidae)

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Abstract

The Kiamichi shiner, *Notropis ortenburgeri*, a diminuitive, silvery, upland stream fish in southwestern Arkansas and eastern Oklahoma was studied from 1999-2001 to determine its distribution, habitat, and conservation status in Arkansas and Oklahoma. Eighty-five collections were made during the two-year study. The present distribution in Arkansas and Oklahoma is described as well as the conservation status of the Kiamichi shiner in both states.

Introduction

The Kiamichi shiner, *Notropis ortenburgeri* Hubbs, is a small, slim, silvery shiner, which occupies small to moderate-sized, clear upland streams of moderate gradient in the Ouachita Highlands of Arkansas and Oklahoma (Robison, 1980). This shiner was originally described by Carl L. Hubbs in Ortenburger and Hubbs (1927) from a single specimen collected in the Mountain Fork River (Little River system) in southeastern Oklahoma. Later, Hubbs and Ortenburger (1929) amplified the original description based on additional specimens primarily from the Red River drainage. Since that time, little has been published concerning this diminutive shiner other than notations regarding locality records or cursory descriptions of ecological requirements (Miller and Robison, 1973; Finnell et. al., 1956; Pigg and Hill, 1974; Robison, 1980; Robison and Buchanan, 1988).

This one-year survey was to determine the present distribution and conservation status of the Kiamichi shiner in Arkansas and Oklahoma.

Materials and Methods

Fieldwork was conducted from September 1999 through September 2000. Seventy-nine collections of fishes were made to document the presence of the Kiamichi shiner in Arkansas and Oklahoma. Fishes were collected using standard minnow seines varying in length from 4.6-6 m and 1.8 m in height with a bar mesh of either 0.3 or 0.6 cm. Fishes were preserved in 10% formalin in the field and later transferred to 50% isopropyl alcohol for permanent storage. Representative specimens of the Kiamichi shiner were preserved from certain sites where the Kiamichi shiner was deemed common. Associated fishes collected with Kiamichi shiners were also collected and enumerated.

In addition, all known contemporary and historical literature regarding the Kiamichi shiner was reviewed and relevant findings summarized or referenced herein. Museums known to house Kiamichi shiners collected in Arkansas and Oklahoma were canvassed. Coverage includes the University of Michigan Museum of Zoology (UMMZ), Tulane University (TU), University of Louisiana - Monroe (NLU), Arkansas State University Museum of Zoology (ASUMZ), Oklahoma State University (OSU), Cornell University (CU), and the Sam Noble Oklahoma Museum of Natural History at the University of Oklahoma (OU), and the University of Tulsa (UTULSAC).

Historical Review

The Kiamichi shiner was originally described by Carl L. Hubbs in 1927 from a single specimen collected in the Mountain Fork River (Little River system) in southeastern Oklahoma. Later, Hubbs and Ortenburger (1929) and Cross and Moore (1952) expanded the range of this species. Miller and Robison (1973) provided information about the distribution and habitat of the Kiamichi shiner in their *Fishes of Oklahoma*. Robison (1980) summarized information on the cyprinid for the *Distributional Atlas*. He reported its distribution as upland streams draining the Ouachita Mountains of west central and southwestern Arkansas and eastern Oklahoma, including portions of the Arkansas and Ouachita drainages and the Kiamichi and Little River systems of the Red River drainage.

Relatively little attention has been focused on this small shiner other than notations as to its occurrence and/or abundance in various stream surveys. Even today, little is known about the biology of the Kiamichi shiner.

Taxonomic Comments.–The Kiamichi shiner has not been assigned to a subgenus and no systematic review of the species has been published to date. The closest relative of the Kiamichi shiner is *Notropis melanostomus* from Florida and southern Mississippi (Bortone, 1989), which is considered to be a sister species of *Notropis ortenburgeri* (Suttkus and Bailey, 1990). Hubbs and Raney (1951) suggested a possible relationship with *Notropis cummingsae*, but this has not been investigated.

Type Locality.–The type locality of the Kiamichi shiner is the Mountain Fork River, 16 km southeast of Broken Bow, McCurtain County, Oklahoma (Ortenburger and Hubbs, 1927; Moore, 1973).

Habitat

Except for cursory statements regarding the general nature of the habitat, little has been written on the ecological requirements of *Notropis ortenburgeri.* Hubbs and Ortenburger (1929) described the habitat as "more or less quiet water, of acid reaction (pH 6.8 or less), in the upland streams of southeastern Oklahoma and western, especially southwestern Arkansas." Black (1940) in a dissertation on the "Fishes of Arkansas" reported the Kiamichi shiner to be similar to the wedgespot shiner (*Notropis greenei*) in its ecology. It is usually found in pools of creeks and rivers and favors the ends of pools near the beginning or end of riffles, where food is no doubt easily secured. In the Little River system, Reeves (1953) reported the Kiamichi shiner to be an inhabitant of small, rocky tributaries of medium size rivers. Miller and Robison (1973) described the habitat as small to moderately sized, clear upland streams in Oklahoma, particularly in quiet pools over large boulder substrates. Pigg and Hill (1974) found *N. ortenburgeri* associated with small to moderate-sized upland streams in the Kiamichi River, Oklahoma. Harris and Douglas (1978) reported this species from clear, deep pools with rocky bottoms in the main Ouachita River. Johnson (1978) described the habitat of the Kiamichi shiner in the Saline River (Red River Drainage) as clear pools over rocky substrate. Herrock (1986) commonly collected the Kiamichi shiner from deep, rock-bottom pools of the headwater tributaries of the Ouachita River. Robison (1980) and Robison and Buchanan (1988) described the habitat of the Kiamichi shiner as pools over gravel, rubble, or boulder-strewn substrates in small to moderate-sized clear upland streams of moderate gradient.

During this study, field collecting was carried out in all major drainages where *Notropis ortenburgeri* was known to occur in an effort to document its habitat more precisely. *Notropis ortenburgeri* was found most frequently in clear, small to moderate-sized upland streams and rivers characterized by moderate to slow gradient. In these upland areas, the Kiamichi shiner tends to inhabit clear, quiet, pools over substrates variously composed of sand, gravel, cobble, and boulders. Such pools typically had a slight flow through them. Rooted aquatic vegetation was generally absent although beds of *Justicia americana* predominated at the edges of many of the pools. *Notropis ortenburgeri* avoided swifter stream sections and riffles. This species seems intolerant of the turbidity and ecological perturbations caused by ditching in agricultural areas and modification of the watershed by clearcutting. Visits to streams previously occupied by *Notropis ortenburgeri* yielded no specimens, following such environmental alteration.

Species Associates.–Species most closely associated environmentally with *Notropis ortenburgeri* are the striped shiner (*Luxilus chrysocephalus*); brook silverside (*Labidesthes sicculus*), blackspotted topminnow (*Fundulus olivaceus*), longear sunfish (*Lepomis megalotis*), smallmouth bass (*Micropterus dolomieu*), and greenside darter (*Etheostoma blennioides*).

Distribution

Notropis ortenburgeri was initially discovered in the Mountain Fork River (Little River system) about 16 km (10 mi.) southeast of Broken Bow, McCurtain County, in southeastern Oklahoma. This small range was later expanded into the Arkansas River drainage of both eastern Oklahoma and western Arkansas (Hubbs and Ortenburger, 1929; Cross and Moore, 1952). Hubbs et al. (1954) erroneously extended the distribution of the Kiamichi shiner southward to include eastern Texas within the range. This error was later repeated by Moore (1968) and Miller and Robison (1973). Reexamination of the Texas specimens by HWR revealed them to be *Notropis hubbsi* described by Bailey and Robison (1978). Texas is thus deleted from the known range of *N. ortenburgeri.*

Presently the Kiamichi shiner is known only from streams draining the Ouachita Mountains of eastern Oklahoma and west-central and southwestern Arkansas, Kiamichi River, and Little River system (Red River drainage) and Ouachita River drainages. In addition, there are several problematic localities previously reported from north of the Arkansas River in Osage, Delaware, and Tulsa counties, Oklahoma. The presence of small populations of *Notropis ortenburgeri* north of the Arkansas River in Oklahoma is puzzling when viewing the geographic range and abundance of this species in the Ouachita Mountains in southeastern Oklahoma and southwestern Arkansas.

On 12 August 1936, W. F. Blair (biology professor at Tulsa University) reported *Notropis ortenburgeri* from a collection of 20 species below the dam on Spavinaw Creek in Delaware County, Oklahoma (Moore, 1973). As the specimens were identified by Carl L. Hubbs, the original describer, the identities seem valid (G. A. Moore, pers. comm.); however, no additional specimens have ever been taken from this Ozarkian stream making this collection suspect. Several other records have been reported from northeastern Oklahoma including Osage County, Oklahoma at Sand Creek collected by G. A. Moore and F. M. Baumgartner on 13 April 1940 and Lost Creek (Sec. 36, T26N, R10E) by W. F. Blair on 1 August 1936. HWR was unable to locate these specimens to verify their

continued existence and identification. Recent extensive collecting of Sand Creek by W. J. Matthews failed to find any specimens of the Kiamichi shiner (W. J. Matthews, University of Oklahoma, pers. comm.). Warren Adams (a former student at Tulsa University, pers. comm.) collected small numbers of *N. ortenburgeri* from Turkey Creek in Osage County in the 1970s. Two specimens (UTULSAC 2511) from Bird Creek, Tulsa County, Oklahoma, represent the westernmost known locality of *N. ortenburgeri.* Unfortunately, these collections also can not be located presently. Intensive collecting in northeastern Oklahoma over the years has substantiated the rarity of this species north of the Arkansas River in Oklahoma as no additional collections are known. These previous collections in Osage, Delaware and Tulsa Counties, may represent the remnants of a once more widely distributed population occupying the western Ozark foothills. Interestingly, no collections of *Notropis ortenburgeri* have been made north of the Arkansas River in Arkansas (Robison and Buchanan, 1988).

Notropis ortenburgeri seems to rather common only in the upper Ouachita tributaries (particularly Wingfield and Hollywood creeks of the Little Missouri River system) in Arkansas. Northward into the Arkansas River drainage (e.g. the Poteau River system), the Kiamichi shiner becomes more rare. Southward, in the Little River system of southwestern Arkansas and Oklahoma and in the Kiamichi River of Oklahoma, the species also becomes less common. The western range limit of this species in the Ouachitas is McGee Creek, a tributary of Muddy Boggy River, 0.8 km (0.5 mile) west of the divide between the Kiamichi and Muddy Boggy River drainages (Pigg, 1977).

The following is a presentation of the distribution of the Kiamichi shiner by river system or main river area. Comments are made concerning this shiner's historical presence, plus the findings of this survey are given.

Arkansas

Fourche La Fave River System (Arkansas River Drainage).–Black (1940) reported the Kiamichi shiner from the upper Fourche la Fave River for the first time. Both Robison (1980) and Robison and Buchanan (1988) mapped the distribution of this species in the Fourche la Fave River system. A total of 8 specimens was taken in 5 collections from the Fourche la Fave River from three localities during this study. The localities in Scott County, Arkansas are as follows: (1) Fourche la Fave at gravel road 0.8 km (0.5 mile) southeast of Boles (5 specimens); (2) Brush Creek at AR St. Hwy. 28 bridge (2 specimens); (3) Black Fork Creek, 14.4 km (9 mi.) south of Winfield on gravel road (1 specimen). This shiner appears to be rare in the various upper tributaries of the Fourche la Fave River system.

Upper Poteau River System (Arkansas River Drainage).–Black (1940) mapped a single occurrence of the Kiamichi shiner from the upper Poteau River in Arkansas. In this study, no specimens of the Kiamichi shiner were collected although 5 collections were made in the upper Poteau River system (Table 1).

Upper Ouachita River and Smaller Tributaries.–Harris (1977) collected 57 specimens of the Kiamichi shiner from a single locality in the upper Ouachita River at U.S. Hwy. 71 bridge (Sec. 21/28, T1S, R30W), Polk Co. This single collection of 57 Kiamichi shiners was the only time Harris (1977) took this shiner, although he made 76 collections from 26 different localities in the upper Ouachita River system and collected over 40,000 individual fishes. J. E. Herrock (1986) subsequently surveyed the upper Ouachita River system almost 10 years later and collected 330 specimens of the Kiamichi shiner in only 2 collections, although he made 74 collections from 31 different localities and captured a total of 28,412 specimens distributed among 61 different species. The localities in Polk County were (1) Ouachita River at bridge on gravel road approximately 45 m north of U.S. Hwy. 270 and 7.2 km (4.5 mi.) west of Acorn (Sec. 14, T1S, R31W) (1 specimen) and (2) Ouachita River at U.S. Hwy. 71 bridge at Acorn (Sec. 21/28, T1S, R30W) (329 specimens).

A total of 10 collections of fishes was taken from the upper Ouachita River in Polk County. These yielded 109 specimens of the Kiamichi shiner at 3 localities: (1) Ouachita River at gravel road 7.2 km (4.5 mi.) west of Acorn (Sec. 14, T1S, R31W) (79 specimens); (2) Ouachita River at U.S. Hwy. 71 at Acorn (Sec. 21/28, T1S, R30W) (21 specimens); (3) Ouachita River at gravel road (Sec. 27, T2S, R30W) (9 specimens).

Lower Ouachita River.–Raymond (1975) surveyed the fishes of the lower Ouachita River from the Remmel Dam to the AR/LA state line. He did not record any Kiamichi shiners from the lower Ouachita River. No collections were made in the lower Ouachita River during this study.

Little Missouri River System (Ouachita River Drainage).–Three localities for the Kiamichi shiner were shown by Black (1940) (Map 4; p. 247). Interestingly, Myers (1977) did not collect a single specimen of the Kiamichi shiner in his survey of the fishes of the Little Missouri River, although he took 91 species in 58 collections from 20 localities and a total of 23,852 specimens. In a subsequent survey of the same river system, Loe (1983) collected 98 species in 57 collections from 35 localities and a total of 25,039 specimens, but he also failed to collect the Kiamichi shiner. Ponder (1983) surveyed Terre Noire Creek, the largest lower tributary of the Little Missouri River, and took 392 individuals of the Kiamichi shiner from 6 localities out

of 44 collections from 28 different localities and 20,010 specimens distributed among 78 fish species. The 6 localities in Clark County where the Kiamichi shiner was collected are (1) Terre Noire Creek at AR St. Hwy. 8, 6.4 km east of Alpine (Sec. 26, T6S, R22W) (172 specimens); (2) Hollywood Creek at TAR (Timber Access Road), 11.2 km east of Alpine (Sec. 28, T6S, R21W) (185 specimens); (3) Terre Noire Creek at TAR, 12.8 miles NW of Hollywood (Sec. 3, T7S, R22W) (1 specimen); (4) Terre Noire Creek at TAR, 3.7 km south of junction of AR St. Hwy. 8 (Sec. 34, T6S, R22W) (4 specimens); (5) Hollywood Creek at TAR, 4 km east of AR St. Hwy 53 (Sec. 10, T7S, R21W) (29 specimens); and (6) Terre Noire Creek at AR St. Hwy 26, 4.0 km west of Hollywood (Sec. 31, T7S, R21W).

In this study, a total of 12 collections of fishes was made from the Little Missouri River system, and 210 specimens of the Kiamichi shiner were captured from the 10 localities. The 10 collection sites in Clark County are (1) Hollywood Creek at AR St. Hwy. 8 (Sec. 28, T6S, R21W) (123 specimens); (2) Hollywood Creek at TAR (Sec. 4, T7S, R21W) (3 specimens); (3) Hollywood Creek at TAR (Sec. 10, T7S, R21W) (5 specimens); (4) Hollywood Creek at AR St. Hwy. 8 (Sec. 28, T6S, R21W) (10 specimens); (5) Hollywood Creek at Hollywood, AR (Sec. 28, T7S, R21W) (2 specimens); (6) Bell Creek at TAR (Sec. 18, T7S, R21W) (48 specimens); (7) Bell Creek at AR St. Hwy. 26 (Sec. 35/36, T7S, R22W)(1 specimen); (8) Terre Noire Creek at AR St. Hwy 26 (Sec. 31, T7S, R22W) (3 specimens); (9) Terre Noire Creek at AR St. Hwy 8 (Sec. 26, T6S, R22W) (9 specimens); (10) Terre Noire Creek at TAR (Sec. 14, T6S, R22W) (6 specimens). The Kiamichi shiner seems to favor the upland tributaries of this system, especially in the upper areas of Hollywood Creek, Bell Creek, and Terre Noire Creek where it is common and locally abundant.

Saline River System (Ouachita River Drainage).– William J. Matthews (pers. comm.) has collected the Kiamichi shiner from the upper Saline River system (Ouachita River drainage), and several years ago, HWR was asked to identify some of these fish specimens. These specimens were identified as the Kiamichi shiner, *Notropis ortenburgeri.* The discovery of the Kiamichi shiner in the upper Saline River is indeed interesting, given the fact that two intensive surveys by graduate students from Northeast Louisiana University (Reynolds, 1971; Stackhouse, 1982) failed to find a single specimen of this shiner despite the combined effort of 177 collections from 82 localities and a total of 64,555 individual fishes taken from the Saline River system. The discovery of this diminutive shiner in a small upland tributary in the Saline River system by Matthews after such a massive effort by others reinforces the idea that we still have much to learn about the distributions of a number of our smaller stream fishes.

To date, 321 specimens of the Kiamichi shiner have been taken from six localities in Saline County in the upper region of the South Fork of Alum Creek of the Saline River in Saline County, Arkansas (W. J. Matthews, pers. comm.). The 6 localities in Saline County are (1) Station 1 – South Fork of Alum Creek, (Sec. 27, T2N, R19W) (2 specimens); (2) Station 2– South Fork of Alum Creek (Sec. 27, T2N, R19W) (217 specimens); (3) Station 3– South Fork of Alum Creek (Sec. 27, T2N, R19W) (3 specimens); (4) Station 4 – South Fork of Alum Creek (Sec. 27, T2N, R19W) (94 specimens); (5) Station 5 – South Fork of Alum Creek (Sec. 29, T2N, R19W) (3 specimens); (6) Station 6 – South Fork of Alum Creek (Sec. 32, T2N, R19W) (2 specimens). Four collections made by HWR and crew failed to include any additional specimens of the Kiamichi shiner in the upper Saline River system (Table 1).

Caddo River System (Ouachita River Drainage).– Neither Fruge (1971) nor L. W. Herrock (1986) found any Kiamichi shiners in their independent fish surveys of the Caddo River, thus the Kiamichi shiner is not believed to inhabit this river system.

In the present study no specimens of the Kiamichi shiner were taken, although 10 collections were made in the Caddo River system (Table 1).

Mountain Fork River System (Red River Drainage).– Black (1940) reported a single locality for the Kiamichi shiner in the upper Mountain Fork River of Arkansas. In this study, no specimens of the Kiamichi shiner were collected, although 5 collections were made in this system (Table 1).

Rolling Fork River System (Red River Drainage).– Black (1940) did not show any collection localities for the Kiamichi shiner. Corkern (1979) surveyed the fishes of the Rolling Fork River and found 13 specimens of the Kiamichi shiner at two different locations. The 2 locations are (1) Rolling Fork River at FAS road, 7 km west of Gillham (sec. 29, T7S, R32W) (12 specimens) and (2) Rolling Fork River at County Rd. 132 bridge, west of DeQueen (Sec. 14, T8S, R32W) (1 specimen). The 13 specimens of the Kiamichi shiner were collected out a total of 17,264 specimens distributed within 67 species from 38 collections from 13 localities. Five collections were made in this study from the Rolling Fork River system without finding a single specimen of the Kiamichi shiner (Table 1).

Cossatot River System (Red River Drainage).–A single location in the upper Cossatot River was shown by Black (1940) for the Kiamichi shiner. Although 5 collections were made in this river system, no additional specimens of the Kiamichi shiner were taken (Table 1).

Saline River System (Red River Drainage).–Black (1940) showed a single occurrence of the Kiamichi shiner in a tributary of the Saline River system, a tributary of the Little River (Red River drainage). Johnson (1978) surveyed the fishes of the Saline River system in western Arkansas. He made 55 collections from 21 localities throughout the Saline River system and took 22,468 specimens of fishes, only 11 of which were Kiamichi shiner. The 11 individuals of the Kiamichi shiner were found at only 2 locations in the system. The 2 localities were (1) Saline River below Dierks Dam, about 4 miles west of Dierks, AR (Sec. 21, T7S, R29W)(4 specimens) and (2) Saline River at bridge on gravel road about 3.5 miles west of Dierks, AR (Sec. 27/28, T7S, R29W) (7 specimens). Five collections of fishes from this river system failed to find the Kiamichi shiner during this study (Table 1).

Oklahoma

Arkansas River Drainage.–The presence of *N. ortenburgeri* north of the Arkansas River in Oklahoma is puzzling when viewing the geographic range and abundance of this species in the Ouachita Mountains in southeastern Oklahoma and southwestern Arkansas. On 12 August 1936, W. F. Blair reported *Notropis ortenburgeri* from a collection of 20 species below the dam on Spavinaw Creek in Delaware County, Oklahoma. As the specimens were identified by Carl L. Hubbs, the original describer, the identities are probably valid. Several other record sites for *N. ortenburgeri* are available from northeastern Oklahoma including Osage County, Oklahoma at Sand Creek, a tributary of the Caney River. Kiamichi shiners were collected by G. A. Moore and F. M. Baumgartner on 13 April 1940 as mentioned by Cross and Moore (1952). Kiamichi shiners were also collected from Lost Creek (Sec. 36, T26N, R10E) by W. F. Blair on 1 August 1936. In the 1970's Warren Adams (pers. comm.) collected small numbers of *N. ortenburgeri* from Turkey Creek in Osage County, Oklahoma. Two specimens (UTULSAC 2511) were taken from Bird Creek, Tulsa County, representing the westernmost known locality of *N. ortenburgeri*; however, these specimens can not now be located. Intensive collecting of northeastern Oklahoma over the years by W. J. Matthews and others has substantiated the rarity of this species north of the Arkansas River in Oklahoma. Not a single collection has been made north of the Arkansas River in over three decades, despite substantial collections being made from these target areas. These relict populations in Osage, Delaware, and Tulsa counties, Oklahoma may represent the remnants of a once more widely distributed population occupying the western Ozark foothills, but probably no longer exist. Interestingly, no collections of *Notropis ortenburgeri* have been reported north of the Arkansas River in Arkansas (Robison and Buchanan, 1988).

Poteau River System (Arkansas River Drainage).–Cross and Moore (1952) surveyed the fishes of the Poteau River system and reported two locations for the Kiamichi shiner in the Oklahoma portion of their survey. Black (1940) had earlier figured 1 locality for the Kiamichi shiner in the upper Poteau River in Arkansas near Waldron, Arkansas (Hubbs and Ortenburger, 1929). No collections were made in Oklahoma from the Poteau River system in this survey.

Kiamichi River System (Red River Drainage).–Pigg and Hill (1974) surveyed the fishes of the Kiamichi River system from 1972-1973. In their study they also included collections from a number of ichthyologists and museums. They found the Kiamichi shiner to be very common in all areas of the river except near the mouth and in lowland tributaries. The Kiamichi shiner was associated with small to moderate-sized upland streams. Echelle and Schnell (1976) performed a factor analysis of species associations among fishes of the Kiamichi River. The Kiamichi shiner was mentioned as a member of the "brook silverside" group of fishes, a group that seems to prefer the more sluggish sections of small to large relatively clear streams in the upper section of the Kiamichi River system. However, in the 1980s W. J. Matthews and R. C. Cashner surveyed the Kiamichi River system and found the species to be scarce (W. J. Matthews, pers. comm.).

Ten collections from 9 localities were made in the upper Kiamichi River system during this study (Table 1). The Kiamichi shiner was found at 4 localities in the upper Kiamichi River system and 68 specimens were collected. The four localities in LeFlore, OK were (1) Kiamichi River at U.S. Hwy. 259 south of Big Cedar (Sec. 14, T2N, R25E) (39 specimens); (2) Kiamichi River at OK St. Hwy 63 east of Big Cedar (Sec. 18, T2N, R26E) (17 specimens); (3) Little Cedar Creek at OK St. Hwy 63 (Sec.7, T2N, R25E) (11 specimens); (4) Billy Creek at Billy Creek Recreation Area (Sec. 36, T3N, R24E) (1 specimen). The Kiamichi shiner appears to be a fairly widespread, but relatively uncommon inhabitant of the upper Kiamichi River system.

Little River System (Red River Drainage).–Reeves (1953) surveyed the fishes of the Little River system in Oklahoma in a doctoral dissertation. He found the Kiamichi shiner at six stations and commented that it was "nowhere abundant." Finnell et al. (1956) collected 33 specimens of the Kiamichi shiner from the Little River. No collections were made in this study in the mainstem Little River in Oklahoma.

Mountain Fork River (Little River Tributary, Red River Drainage).–Finnell et al. (1956) collected 10 specimens of the Kiamichi shiner from Lick Creek, a tributary of the Mountain Fork River. Additional specimens were available from the OU museum. No collections were made in the

Mountain Fork River system in the Oklahoma portion of its drainage during this study.

Glover River (Little River Tributary, Red River Drainage).–Taylor and Wade (1972) provided an inventory of the biological resources of the Glover River watershed. They did not collect any Kiamichi shiners in their survey, although they made 50 collections and collected 11,038 individual fishes in 50 species. Five collections were made in the Glover River in Oklahoma during the present study; however, no specimens of the Kiamichi shiner were taken. However, there are 159 specimens of the Kiamichi shiner housed at the OU museum, which document its presence in the Glover River.

In summary, the Kiamichi shiner inhabits upland streams of the Kiamichi River, Little River system (Red River drainage), and Ouachita River drainages flowing out of the Ouachita Mountains of eastern Oklahoma and west-central and southwestern Arkansas, respectively. In addition, this shiner formerly occurred in several disjunct localities north of the Arkansas River in Osage, Delaware, and Tulsa counties, Oklahoma (Fig. 1); however, it has not been found in over 30 years in this region despite an intensive search.

Conservation Status

Historical Conservation Status.–Both Robison (1974) and Buchanan (1974) independently concluded that the Kiamichi shiner was rare in Arkansas. Later in their *Fishes of Arkansas,* Robison and Buchanan (1988) listed the Kiamichi shiner as "threatened" within the state in their discussion of the rare and endangered fishes of Arkansas.

In Oklahoma, Robison et al. (1974) listed the Kiamichi shiner as "rare" stating that "disjunct populations of this species make interpretation of its distribution and status difficult." The Rare and Endangered Species of Oklahoma Committee (1975) also concurred in assigning the Kiamichi shiner a "rare-2" status, which meant the species may be quite abundant where it occurs, but it is known in only a few localities or in a restricted habitat within Oklahoma.

Warren et al. (2000) recently reviewed the status of 662 native freshwater fishes of the southern United States in which Arkansas and eastern Oklahoma were included. Their findings listed the Kiamichi shiner as "vulnerable" which meant a species or subspecies that may become endangered or threatened by relatively minor disturbances to its habitat or that deserve careful monitoring of its distribution and abundance in the continental waters of the United States.

Fishes and the other aquatic fauna are disproportionately imperiled when compared to terrestrial fauna (Warren and Burr, 1994). Interestingly, Warren et al. (2000) found that 6% of the southern fishes were "endangered" while 7% were considered "threatened" and 15% were "vulnerable." Williams et al. (1989) listed habitat loss as one of the greatest causes of the declines in populations of native fishes in North America. Widespread reservoir construction and declines in water quality have severely altered most of North America's clean, free-flowing riverine habitats (Benke, 1990). Sadly, Warren et al. (2000) concluded that the trend for southern fishes in the United States is clear; jeopardized fishes are successively moving from a vulnerable category to that of imminent threat of extinction.

Present Conservation Status.–The state of Arkansas presently has no official state list of threatened or endangered wildlife or plants. Instead, protection is afforded by the Arkansas Game and Fish Commission primarily to federally threatened species.

A total of 79 collections of fishes was made during this study within the historical distribution of the Kiamichi shiner. From these 79 collections, 392 specimens of Kiamichi shiners were captured in Arkansas and Oklahoma (Table 1)). After careful review of all of the major museum holdings available of the Kiamichi shiner, a year of intensive field work collecting Kiamichi shiners, review of all pertinent literature, and discussions with virtually all of the major collectors of Kiamichi shiners in Arkansas and Oklahoma, it seems that the Kiamichi shiner is a widespread, locally-abundant shiner that lives in upland habitats and probably undergoes population fluctuations through time. It seems apparent that it has not precipitously declined in abundance throughout its historical range in Arkansas and Oklahoma (Table 2), although it has been collected in smaller numbers in recent years, primarily because of the tremendous ichthyological field collecting that occurred in the 1970's, which yielded so many specimens. This "golden period" occurred when fish populations of numerous streams were examined by master's thesis projects. Many of these projects surveyed Arkansas streams south of the Arkansas River within the geographic range of the Kiamichi shiner.

Table 2 provides a quick view of the abundance of the Kiamichi shiner in major Arkansas and Oklahoma drainages by decade. While certainly not definitive, Table 2 shows the Kiamichi shiner apparently declining in the decade of the 1980s but thriving in the decade of the 1990s. Earlier years show little in the way of trends, other than a gradual increase in numbers after its initial discovery in the 1920s.

On the basis of all known collections of this shiner, good populations of *Notropis ortenburgeri* occur in the upper Kiamichi River system (49.4% of all known collected specimens), Little Missouri River (Terre Noir Creek specifically) (17.1%), and upper Ouachita River system (14 %) (Table 3), but several other historical areas did not

produce specimens of the Kiamichi shiner in this study.

While destruction and modification of habitat from impoundments with concomitant cold water release have harmed numerous small, non-game stream fishes, the Kiamichi shiner has escaped the fate of many other stream fishes because of its upland habitat requirements. These upland habitats are usually located above many of the environmental perturbations that have occurred within the various watersheds where this species resides. Some reduction in population numbers may have occurred due to poor land practices such as road building, farming, clearing of land for pasture, clearcutting, destruction of riparian buffer strips, and other human perturbations that continue in these watersheds. Gravel removal operations in many Arkansas streams (Filipek and Oliver, 1994), nutrient enrichment from the enormous increase in poultry and swine operations, and human population increases probably all threaten populations of this shiner.

During this study the continued presence of the Kiamichi shiner was documented in several of the river systems in Arkansas and Oklahoma from which it was collected historically including the Kiamichi, upper Ouachita, Little Missouri, and Fourche la Fave river systems. No specimens were collected from the Caddo, Mountain Fork, Poteau, Rolling Fork, or Saline river systems, where historically the Kiamichi shiner had been taken. No new populations of the Kiamichi shiner were discovered in river systems where they were previously unknown.

Thus, after reviewing the collection records of the Kiamichi shiner from the UMMZ, NLU, OSU, OU, TU, ASUMZ, UTULSAC, and CU and after a year of field work, the Kiamichi shiner is not herein recommended for official federally threatened status at this time. Rather, this small silvery cyprinid species should be accorded a status of "vulnerable," and a program should be initiated to monitor its continued existence in southeastern Oklahoma and southwestern Arkansas. The apparent disappearance from several historical localities makes it imperative that a careful watch on this species be maintained in the future.

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Table 1. Number of collections and Kiamichi shiners obtained in Arkansas and Oklahoma from 1999-2001.

Locality (River System)	No. of Collections	No. Kiamichi Shiners
1. Ouachita River (mainstem)	10	106
2. Caddo River	10	0
3. Little Missouri River	10	210
4. Saline River (Ouachita)	4	0
5. Kiamichi River	10	68
6. Glover River	5	0
7. Mountain Fork River	5	0
8. Rolling Fork River	5	0
9. Cossatot River	5	0
10. Saline River (Red)	5	0
11. Poteau River (AR)	5	0
12. Fourche la Fave River (AR)	5	8
TOTAL	**79**	**392**

Table 2. Number of Kiamichi shiners collected during various years, 1927-2001.

Years	No. Kiamichi Shiners
1927–1939	23
1940–1949	2
1950–1959	43
1960–1969	137
1970–1979	2,951
1980–1989	563
1990–1999	1,505
2000–2001	395
Totals	**5,619**

Table 3. Number of the Kiamichi shiners by river system.

River System	No. Kiamichi Shiners	Percentage (%)
Ouachita River	497	14.0
Caddo River	0	0.0
Little Missouri River	605	17.1
Saline River (Ouachita)	321	9.1
Little River	77	2.2
Glover River	159	4.5
Mountain Fork River	50	1.4
Rolling Fork River	13	0.4
Saline River (Red)	11	0.3
Kiamichi River	1,755	49.4
Poteau River	33	0.9
Fourche la Fave River	26	0.7
Total	**3,547**	**100.0**

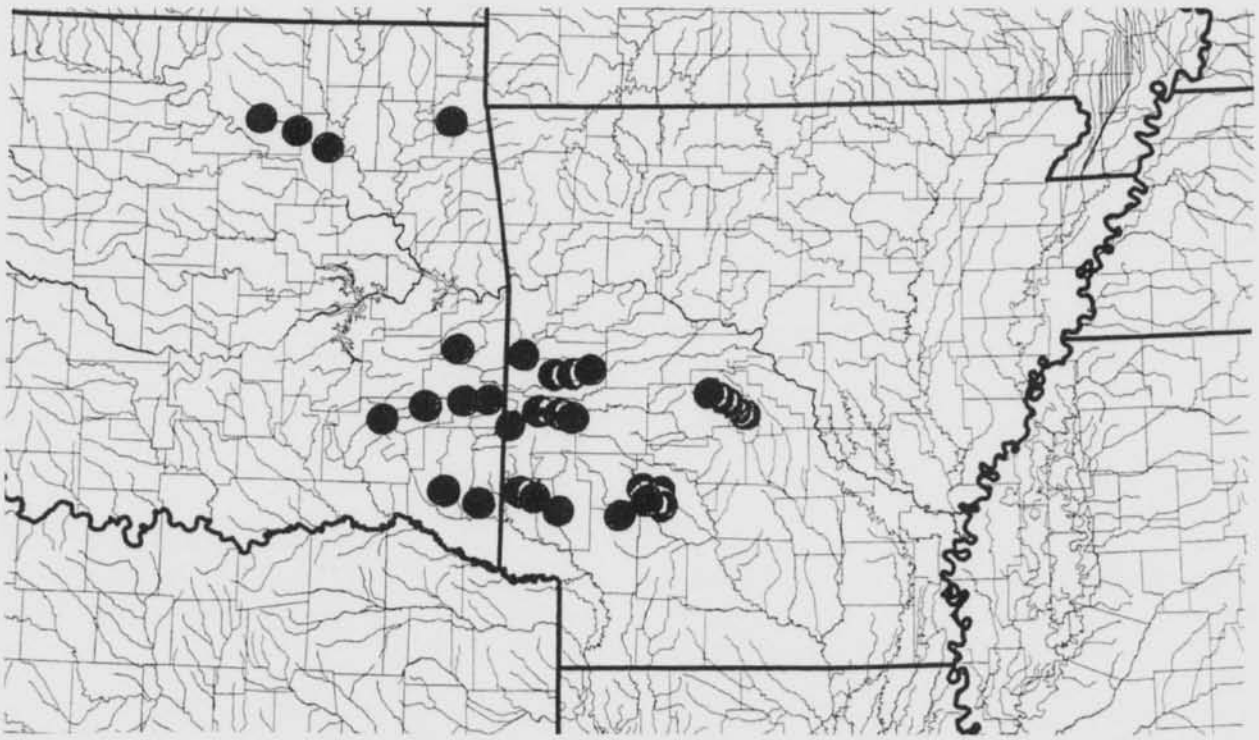

Fig. 1. Distribution of the Kiamichi Shiner, *Notropis ortenburgeri*, in Arkansas and Oklahoma.

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Fishes of the Pine Bluff Arsenal, Jefferson County, Arkansas

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Abstract

A survey of the fishes of the Pine Bluff Arsenal (PBA) located in Jefferson County, Arkansas was initiated in February 1999 and continued until October 1999 with several supplemental collections made in 2000. A total of 3,396 fishes was taken in 81 collections on the PBA and revealed 59 species distributed in 17 families and 36 genera. The most abundant fishes collected were *Dorosoma petenense*, *Gambusia affinis*, *Labidesthes sicculus*, *Notemigonus crysoleucas*, and *Lepomis marginatus*.

Introduction

Arkansas has a diverse ichthyofauna of over 215 species of fishes distributed in 63 genera and 27 families that occupy a myriad of different aquatic habitats within its political boundaries (Robison and Buchanan, 1988). Ecologically, the fish fauna of Arkansas is primarily dominated by fluviatile forms because of the absence of natural lakes other than oxbow lakes situated along the larger rivers (Robison and Buchanan, 1988). While the overall distributions of state fish species are well known, detailed data regarding the abundance and diversity for specific smaller areas within Arkansas are lacking. This study represents the first attempt to inventory the fishes of the Pine Bluff Arsenal, Jefferson County, Arkansas and to obtain baseline data on the ichthyofauna of the arsenal necessary for future monitoring and possible replication of this study. Specific purposes of this study were (1) to provide an inventory of the fishes inhabiting the five aquatic habitat types located on the Pine Bluff Arsenal (PBA) (2) to search for any taxa of fishes known from the PBA or adjoining counties that are tracked by the Arkansas Natural Heritage Commission (ANHC) and (3) to report any findings for populations of threatened, endangered, or otherwise significant taxa occurring on the PBA.

Materials and Methods

Field work for this project was conducted from February 1999 through October 1999. Collecting trips to the Pine Bluff Arsenal were made on February 26, March 13-14, April 9-10, June 28-29, July 1-2, August 13-14, September 10-11, October 8-9, and October 16, 1999. Nine trips were taken and 16 field days were spent during which 81 collections of fishes were made on the arsenal. All 81 collections are listed in Robison (1999).

Collecting was concentrated in streams, ponds, and lakes with some collections made in the mainstream Arkansas River adjacent to the arsenal. A variety of collecting methods was used including the use of seines (3.65 m X 1.21 m., 6.1 m X 1.83 m, and 9.15 m X 2.44 m.), aquatic dip nets, and gill nets.

Representative specimens were preserved in 10% formalin in the field and later washed and transferred to 45 percent isopropyl alcohol. Preserved specimens were deposited in the Southern Arkansas University Fish Collection. In addition, all pertinent literature was searched for records of fishes previously collected from Jefferson County, Arkansas.

Description of the Area

The U. S. Pine Bluff Arsenal (PBA) is a 19 km by 6.4 km government military installation located in Jefferson County, Arkansas on the west bank of the Arkansas River approximately 51 km southeast of Little Rock, AR and 4.8 km northwest of Pine Bluff, AR. The arsenal covers 6,052 ha of which 4,313 ha are in forest (Charles Becker, PBA Biologist, pers. comm.). The remaining 1,739 ha of open land consist of lawns, buildings, roads, railroads, lakes and streams, wildlife plots, and open fields (Becker, 1992).

Topographically, the arsenal is generally flat with poor drainage. The eastern portion of the arsenal is about 12.2 m lower in elevation due to an abrupt drop to the river floodplain (Becker, 1992). The northernmost portion of the arsenal is characterized by rolling hills and numerous streams.

The arsenal is located within the Arkansas River drainage with the Arkansas River flowing along most of the eastern boundary (Campbell et al., 1997). The arsenal is drained by perennial, intermittent, and ephemeral drainage systems that flow east-southeast to the Arkansas River. The primary streams of the arsenal are Jackson Creek, Eastwood Bayou, Phillips Creek, Tulley Creek, Caney Creek, and White Creek. Numerous artificial impondments, beaver dams, and one modified natural lake, Yellow Lake, occur on the arsenal. Yellow Lake is the largest lake on the arsenal with a surface area of 105.3 ha and a maximum depth of 2.74 m. It is flooded several times a year by the Arkansas River. Tulley Lake is next in size with a surface area of 12.15 ha and a maximum depth of 5.2 m.

Historical Review

Black (1940) completed a doctoral dissertation on the fishes of Arkansas, but he did little collecting near Pine Bluff. To the south of the arsenal, Thomas (1976) finished a master's thesis on the fishes of Bayou Bartholomew which drains southeast Arkansas and northeast Louisiana. Thomas (1976) made collections in Jefferson County, but none were on the arsenal. Buchanan (1976) studied the fishes of the Arkansas River navigation sysytem and made collections on the Arkansas River slightly above and slightly below the arsenal boundaries.

The Arkansas Game and Fish Commission has not sampled the lakes of the arsenal according to Allen Carter (AGFC Fishery Biologist, pers. comm..). No other ichthyological studies have been documented from within the arsenal boundaries.

Results and Discussion

Fishes of Pine Bluff Arsenal.–Arkansas has 197 native fish species inhabiting the state (Robison and Buchanan, 1988). Fifty-nine native species of fishes were collected on the arsenal, representing about 29 percent of the total documented ichthyofauna of Arkansas. The 59 species of fishes were distributed in 17 families and 36 genera.

Abundance of Fishes.–A total of 3,396 fishes was collected during the study. Table 1 indicates the abundance of the fishes collected from the PBA by providing actual number of each species collected, plus the relative abundance of each species.

The most abundant species on the arsenal was the threadfin shad (*Dorosoma petenense*, 1,234 individuals), which comprised 36.34 percent of the total fishes collected. The second most abundant species collected was the western mosquitofish (*Gambusia affinis*) comprising 24.47 percent (831 individuals)of the total while the third most abundant species was the brook silverside (*Labidesthes sicculus*, 167 individuals) comprising 4.92 percent. Other abundant species included the golden shiner (*Notemigonus crysoleucas*, 129 individuals) with 3.80 percent and dollar sunfish (*Lepomis marginatus*, 106) with 3.12 percent of the total.

Table 2 designates each fish species collected on the arsenal according to a scheme of four categories: abundant, common, uncommon, and rare. These terms are defined as follows: rare = 0-2 individuals; uncommon = 3-10 individuals; common = 11-50 individuals, and adundant = over 50 individuals.

Using the above definitions, 10 species were categorized as being abundant on the arsenal: threadfin shad (*D. petenense*), red shiner (*Cyprinella lutrensis*), golden shiner (*N. crysoleucas*), emerald shiner (*Notropis atherinoides*), western mosquitofish (*G. affinis*), brook silverside (*L. sicculus*), inland silverside (*Menidia beryllina*), green sunfish (*Lepomis cyanellus*), dollar sunfish (*L. marginatus*), and largemouth bass (*Micropterus salmoides*).

Common species (16 species) were the gizzard shad (*Dorosoma cepedianum*), blacktail shiner (*Cyprinella venusta*), redfin shiner (*Lythrurus umbratilus*), fathead minnow (*Pimephales promelas*), bullhead minnow (*P. vigilax*), yellow bullhead (*Ameiurus natalis*), blue catfish (*Ictalurus furcatus*), channel catfish (*I. punctatus*), tadpole madtom (*Noturus gyrinus*), pirate perch (*Aphredoderus sayanus*), blackspotted topminnow (*Fundulus olivaceus*), orangespotted sunfish (*Lepomis humilis*), bluegill (*L. macrochirus*), redear sunfish (*L. microlophus*), white crappie (*Pomoxis annularis*), and cypress darter (*Etheostoma proeliare*).

The 16 uncommon fish species collected were the common carp (*Cyprinus carpio*), river shiner (*Notropis blennius*), creek chubsucker (*Erimyzon oblongus*), smallmouth buffalo (*Ictiobus bubalus*), black bullhead (*Ameiurus melas*), grass pickerel (*Esox americanus*), golden topminnow (*Fundulus chrysotus*), blackstripe topminnow (*F. notatus*), white bass (*Morone chrysops*), flier (*Centrachus macropterus*), warmouth (*Lepomis gulosus*), longear sunfish (*L. megalotis*), redspotted sunfish (*L. miniatus*), black crappie (*Pomoxis nigromaculatus*), swamp darter (*Etheostoma fusiforme*), and sauger (*Stizostedion canadense*).

Rare species with only one or two individuals collected were represented by 17 species. These rare species were the shovelnose sturgeon (*Scaphirhynchus platorynchus*), spotted gar (*Lepisosteus oculatus*), longnose gar (*L. osseus*), bowfin (*Amia calva*), goldeye (*Hiodon alosoides*), goldfish (*Carassius auratus*), creek chub (*Semotilus atromaculatus*), river carpsucker (*Carpiodes carpio*), bigmouth buffalo (*Ictiobus cyprinellus*), spotted sucker (*Minytrema melanops*), flathead catfish (*Pylodictis olivaris*), yellow bass (*Morone mississippiensis*), bluntnose darter (*Etheostoma chlorosoma*), slough darter (*E. gracile*), redfin darter (*E. whipplei*), logperch (*Percina caprodes*), and freshwater drum (*Aplodinotus grunniens*).

Distribution by Habitat.–Five distinct aquatic habitat types were identified on the PBA. They are (1) small woodland streams, (2) sluggish bayou sections, (3) big river (Arkansas River mainstem), (4) ponds, and (5) lakes. Table 3 lists the fishes collected on the arsenal by habitat type.

Thirty-four species of fishes were collected from the lake habitat while 24 species were found in the big river habitat (Arkansas River), and 23 fish species were taken in the sluggish bayou sections of the arsenal. Ponds, as expected, yielded the fewest number of species (eight) because their fish faunas consist of "stocked" species like *I. punctatus* and *P. promelas* for the most part.

Only two species, *G. affinis* and *M. salmoides*, were collected from each of the five habitat types. *N. crysoleucas*, *A. natalis*, *I. punctatus*, *L. cyanellus*, *L. macrochirus*, and

L. marginatus were each found in four of the five habitat types.

Small woodland streams are fairly abundant on the arsenal. Such aquatic systems are relatively clear, tannin stained, shallow bodies of water with mud and sand substrates.Little aquatic vegetation occurs at the stream margins. Nineteen species of fishes were taken from the small woodland stream habitat (Table 3)

In several areas of the arsenal larger sluggish bayou sections of Eastwood and Caney bayous served as habitat for 23 species of fishes (Table 3). These areas were typically deeper, devoid of vegetation, and more turbid than the smaller streams of the arsenal. Substrates were generally mud and sand.

The Arkansas River forms the northeastern border of the Pine Bluff Arsenal. This "big river" habitat created by the Arkansas River adds an additional component of the fish fauna not generally found when surveying the fishes of other lowland delta regions. A total of 24 species (Table 3) was taken from the big river habitat

There are a number of artificial ponds created by arsenal personnel as recreational areas for both base and other personnel. Such ponds are depauperate in species composition because they are stocked for fishing recreation. The pond habitat type yielded only eight fish species (Table 3).

Arsenal lakes include Yellow Lake, Tulley Lake, Upper Duck Pond and Lower Duck Pond. Yellow Lake is by far the largest lake on the arsenal and supports the largest and most diverse fish population. Thirty-four species of fishes were documented from Yellow Lake and other lakes (Table 3).

Conservation Status.–A single specimen of the goldeye (*Hiodon alosoides*) was collected on the arsenal from the Arkansas River (Station 47). The goldeye is considered a species of "Special Concern" by the ANHC with relatively few records in state collections; however its perceived scarcity probably results more from a lack of collecting the big river habitats in Arkansas than from actual scarcity. Ongoing and future collecting in the big river habitat in Arkansas will no doubt reveal additional collecting sites for this species. Robison and Buchanan (1988) did not consider the goldeye as having any conservation status in Arkansas and did not include it among the 15 fish species they listed as being of "Special Concern" for Arkansas. In fact, it is fairly common in the Arkansas River near Fort Smith and was documented from 15 locations in the Arkansas River by Buchanan (1976) and Robison and Buchanan (1988).

The ANHC lists the swamp darter (*Etheostoma fusiforme*) from nearby Prairie and Grant counties as of "Special Concern." Seven specimens of the swamp darter were taken from Yellow Lake during this study. This species is probably more abundant than our collecting indicated.

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Table 1. Number and relative abundance (percent of total number) of fish species collected from the Pine Bluff Arsenal, Jefferson County, Arkansas from February–October, 1999.

Species	Pine Bluff Drainages N = 3396	
	Number	Percentage
Family Acipenseridae – Sturgeons		
Scaphirhynchus platorynchus – shovelnose sturgeon	1	0.03

ıble 1. Continued.

Species	Pine Bluff Drainages N = 3396	
	Number	Percentage
Family Lepisosteidae – Gars		
Lepisosteus oculatus – spotted gar	2	0.06
Lepisosteus osseus – longnose gar	1	0.03
Family Amiidae – Bowfins		
Amia calva – bowfin	2	0.06
Family Hiodontidae – Mooneyes		
Hiodon alosoides – goldeye	1	0.03
Family Clupeidae – Herrings		
Dorosoma cepedianum – gizzard shad	38	1.12
Dorosoma petenense – threadfin shad	1,234	36.34
Family Cyprinidae – Carps and Minnows		
Carassius auratus – goldfish	2	0.06
Cyprinella lutrensis – red shiner	83	2.44
Cyprinella venusta – blacktail shiner	29	0.85
Cyprinus carpio – common carp	8	0.24
Lythrurus umbratilis – redfin shiner	46	1.35
Notemigonus crysoleucas – golden shiner	129	3.80
Notropis atherinoides – emerald shiner	94	2.77
Notropis blennius – river shiner	8	0.24
Pimephales promelas – fathead minnow	35	1.03
Pimephales vigilax – bullhead minnow	17	0.50
Semotilus atromaculatus – creek chub	2	0.06
Family Catostomidae – Suckers		
Carpiodes carpio – river carpsucker	2	0.06
Erimyzon oblongus – creek chubsucker	5	0.15
Ictiobus bubalus – smallmouth buffalo	3	0.09
Ictiobus cyprinellus – bigmouth buffalo	1	0.03
Minytrema melanops – spotted sucker	2	0.06
Family Ictaluridae – Bullhead Catfishes		
Ameiurus melas – black bullhead	9	0.27
Ameiurus natalis – yellow bullhead	16	0.47
Ictalurus furcatus – blue catfish	11	0.32
Ictalurus punctatus – channel catfish	22	0.65
Noturus gyrinus – tadpole madtom	13	0.38
Pylodictis olivaris – flathead catfish	2	0.06
Family Esocidae – Pikes		
Esox americanus – grass pickerel	6	0.18
Family Aphredoderidae – Pirate Perches		
Aphredoderus sayanus – pirate perch	14	0.41

Table 1. Continued.

Species	Pine Bluff Drainages N = 3396	
	Number	Percentage
Family Fundulidae – Killifishes		
Fundulus chrysotus – golden topminnow	7	0.21
Fundulus notatus – blackstripe topminnow	5	0.15
Fundulus olivaceus – blackspotted topminnow	48	1.41
Family Poeciliidae – Livebearers		
Gambusia affinis – mosquitofish	831	24.47
Family Atherinidae – Silversides		
Labidesthes sicculus – brook silverside	167	4.92
Menidia beryllina – inland silverside	63	1.86
Family Moronidae – Temperate Basses		
Morone chrysops – white bass	3	0.09
Morone mississippiensis – yellow bass	2	0.06
Centrarchus macropterus – flier	8	0.24
Lepomis cyanellus – green sunfish	57	1.68
Lepomis gulosus – warmouth	10	0.29
Lepomis humilis – orangespotted sunfish	21	0.62
Lepomis macrochirus – bluegill	39	1.15
Lepomis marginatus – dollar sunfish	106	3.12
Lepomis megalotis – longear sunfish	8	0.24
Lepomis microlophus – redear sunfish	14	0.41
Lepomis miniatus – redspotted sunfish	3	0.09
Micropterus salmoides – largemouth bass	62	1.83
Pomoxis annularis – white crappie	41	1.21
Pomoxis nigromaculatus – black crappie	3	0.09
Family Percidae – Perches		
Etheostoma chlorosoma – bluntnose darter	2	0.06
Etheostoma fusiforme – swamp darter	7	0.21
Etheostoma gracile – slough darter	2	0.06
Etheostoma proeliare – cypress darter	42	1.24
Etheostoma whipplei – redfin darter	1	0.03
Percina caprodes – logperch	2	0.06
Stizostedion canadense – sauger	3	0.09
Family Sciaenidae – Drums		
Aplodinotus grunniens – freshwater drum	1	0.03
Totals	3,396	100.00

Table 2. Abundance of the fishes collected from Pine Bluff Arsenal, Jefferson County, Arkansas from February–October, 1999.

Species	Abundant	Common	Uncommon	Rare
Family Acipenseridae – Sturgeons				
Scaphirhynchus platorynchus – shovelnose sturgeon	–	–	–	X
Family Lepisosteidae – Gars				
Lepisosteus oculatus – spotted gar	–	–	X	–
Lepisosteus osseus – longnose gar	–	–	–	X
Family Amiidae – Bowfins				
Amia calva – bowfin	–	–	–	X
Family Hiodontidae – Mooneyes				
Hiodon alosoides – goldeye	–	–	–	X
Clupeidae – Herrings				
Dorosoma cepedianum – gizzard shad	–	X	–	–
Dorosoma petenense – threadfin shad	X	–	–	–
Family Cyprinidae – Carps and Minnows				
Carassius auratus – goldfish	–	–	–	X
Cyprinella lutrensis – red shiner	X	–	–	–
Cyprinella venusta – blacktail shiner	–	X	–	–
Cyprinus carpio – common carp	–	–	X	–
Lythrurus umbratilis – redfin shiner	–	X	–	–
Notemigonus crysoleucas – golden shiner	X	–	–	–
Notropis atherinoides – emerald shiner	X	–	–	–
Notropis blennius – river shiner	–	–	X	–
Pimephales promelas – fathead minnow	–	X	–	–
Pimephales vigilax – bullhead minnow	–	X	–	–
Semotilus atromaculatus – creek chub	–	–	–	X
Family Catostomidae – Suckers				
Carpiodes carpio – river carpsucker	–	–	–	X
Erimyzon oblongus – creek chubsucker	–	–	X	–
Ictiobus bubalus – smallmouth buffalo	–	–	X	–
Ictiobus cyprinellus – bigmouth buffalo	–	–	X	–
Minytrema melanops – spotted sucker	–	–	–	X
Family Ictaluridae – Bullhead Catfishes				
Ameiurus melas – black bullhead	–	–	X	–
Ameiurus natalis – yellow bullhead	–	X	–	–
Ictalurus furcatus – blue catfish	–	X	–	–
Ictalurus punctatus – channel catfish	–	X	–	–
Noturus gyrinus – tadpole madtom	–	X	–	–
Pylodictis olivaris – flathead catfish	–	–	–	X
Family Esocidae – Pikes				
Esox americanus – grass pickerel	–	–	X	–

Table 2. Continued.

Species	Abundant	Common	Uncommon	Rare
Family Aphredoderidae – Pirate Perches				
Aphredoderus sayanus – pirate perch	–	X	–	–
Family Fundulidae – Killifishes				
Fundulus chrysotus – golden topminnow	–	–	X	–
Fundulus notatus – blackstripe topminnow	–	–	X	–
Fundulus olivaceus – blackspotted topminnow	–	X	–	–
Family Poeciliidae – Livebearers				
Gambusia affinis – mosquitofish	X	–	–	–
Family Atherinidae – Silversides				
Labidesthes sicculus – brook silverside	X	–	–	–
Menidia beryllina – inland silverside	X	–	–	–
Family Moronidae – Temperate Basses				
Morone chrysops – white bass	–	–	X	–
Morone mississippiensis – yellow bass	–	–	–	X
Family Centrarchidae – Sunfishes				
Centrarchus macropterus – flier	–	–	X	–
Lepomis cyanellus – green sunfish	X	–	–	–
Lepomis gulosus – warmouth	–	–	X	–
Lepomis humilis – orangespotted sunfish	–	X	–	–
Lepomis macrochirus – bluegill	–	X	–	–
Lepomis marginatus – dollar sunfish	X	–	–	–
Lepomis megalotis – longear sunfish	–	–	X	–
Lepomis microlophus – redear sunfish	–	X	–	–
Lepomis miniatus – redspotted sunfish	–	–	X	–
Micropterus salmoides – largemouth bass	X	–	–	–
Pomoxis annularis – white crappie	–	X	–	–
Pomoxis nigromaculatus – black crappie	–	–	X	–
Family Percidae – Perches				
Etheostoma chlorosoma – bluntnose darter	–	–	–	X
Etheostoma fusiforme – swamp darter	–	–	X	–
Etheostoma gracile – slough darter	–	–	–	X
Etheostoma proeliare – cypress darter	–	X	–	–
Etheostoma whipplei – redfin darter	–	–	–	X
Percina caprodes – logperch	–	–	–	X
Stizostedion canadense – sauger	–	–	X	–
Stizostedion vitreum – walleye				
Family Sciaenidae – Drums				
Aplodinotus grunniens – freshwater drum	–	–	–	X
TOTALS	**10**	**16**	**17**	**16**

Table 3. Habitats of fishes collected from the Pine Bluff Arsenal, Jefferson County, Arkansas from February–October, 1999.*

Species	1	2	3	4	5
Family Acipenseridae – Sturgeons					
Scaphirhynchus platorynchus – shovelnose sturgeon	–	–	X	–	–
Family Lepisosteidae – Gars					
Lepisosteus oculatus – spotted gar	–	X	–	–	X
Lepisosteus osseus – longnose gar	–	–	–	–	X
Family Amiidae – Bowfins					
Amia calva – bowfin	–	–	–	–	X
Family Hiodontidae – Mooneyes					
Hiodon alosoides – goldeye	–	–	X	–	–
Family Clupeidae – Herrings					
Dorosoma cepedianum – gizzard shad	–	X	X	–	X
Dorosoma petenense – threadfin shad	–	X	X	–	X
Family Cyprinidae – Minnows					
Carassius auratus – goldfish	–	–	–	–	X
Cyprinella lutrensis – red shiner	–	–	X	–	–
Cyprinella venusta – blacktail shiner	–	X	X	–	–
Cyprinus carpio – common carp	–	X	–	–	–
Lythrurus umbratilis – redfin shiner	X	X	–	–	–
Notemigonus crysoleucas – golden shiner	X	X	–	X	X
Notropis atherinoides – emerald shiner	–	–	X	–	–
Notropis blennius – river shiner	–	–	X	–	–
Pimephales promelas – fathead minnow	–	–	X	X	X
Pimephales vigilax – bullhead minnow	–	–	X	–	X
Semotilus atromaculatus – creek chub	X	–	–	–	–
Family Catostomidae – Suckers					
Carpiodes carpio – river carpsucker	–	–	X	–	–
Erimyzon oblongus – creek chubsucker	X	X	–	–	–
Ictiobus bubalus – smallmouth buffalo	–	X	X	–	–
Ictiobus cyprinellus – bigmouth buffalo	–	–	X	–	–
Minytrema melanops – spotted sucker	–	X	–	–	–
Family Ictaluridae – Bullhead Catfishes					
Ameiurus melas – black bullhead	X	X	–	–	X
Ameiurus natalis – yellow bullhead	X	X	–	X	X
Ictalurus furcatus – blue catfish	–	–	X	–	–
Ictalurus punctatus – channel catfish	–	X	X	X	X
Noturus gyrinus – tadpole madtom	–	–	–	–	X
Pylodictis olivaris – flathead catfish	–	–	X	–	–
Family Esocidae – Pikes					
Esox americanus – grass pickerel	X	–	–	–	X

Table 3. Continued.

Species	1	2	3	4	5
Family Aphredoderidae – Pirate Perches					
Aphredoderus sayanus– pirate perch	X	X	–	–	–
Family Fundulidae – Killifishes					
Fundulus chrysotus– golden topminnow	–	–	–	–	X
Fundulus notatus– blackstripe topminnow	–	X	–	–	–
Fundulus olivaceus– blackspotted topminnow	X	X	–	–	X
Family Poeciliidae – Livebearers					
Gambusia affinis– mosquitofish	X	X	X	X	X
Family Atherinidae – Silversides					
Labidesthes sicculus– brook silverside	X	X	–	–	X
Menidia beryllina– inland silverside	–	–	X	–	–
Family Moronidae – Temperate Basses					
Morone chrysops– white bass	–	–	–	–	X
Morone mississippiensis– yellow bass	–	–	–	–	X
Famly Centrarchidae – Sunfishes					
Centrarchus macropterus– flier	–	–	–	–	X
Lepomis cyanellus– green sunfish	X	X	–	X	X
Lepomis gulosus– warmouth	–	–	–	–	X
Lepomis humilis– orangespotted sunfish	–	–	X	–	X
Lepomis macrochirus– bluegill	X	X	–	X	X
Lepomis marginatus– dollar sunfish	X	X	X	–	X
Lepomis megalotis– longear sunfish	–	–	X	–	–
Lepomis microlophus– redear unfish	–	–	–	–	X
Lepomis miniatus– redspotted sunfish	–	–	–	–	X
Micropterus salmoides– largemouth bass	X	X	X	X	X
Pomoxis annularis– white crappie	–	–	–	–	X
Pomoxis nigromaculatus– black crappie	–	–	–	–	X
Family Percidae – Perches					
Etheostoma chlorosoma– bluntnose darte	X	–	–	–	–
Etheostoma fusiforme– swamp darter	–	–	–	–	X
Etheostoma gracile– slough darter	X	–	–	–	–
Etheostoma proeliare– cypress darter	X	X	–	–	X
Etheostoma whipplei– redfin darter	X	–	–	–	–
Percina caprodes– logperch	–	–	–	–	X
Stizostedion canadense– sauger	–	–	X	–	–
Family Sciaenidae – Drums					
Aplodinotus grunniens– freshwater drum	–	–	X	–	–
TOTALS	**19**	**23**	**24**	**8**	**34**

*1 = small woodland streams
2 = sluggish bayou section
3 = big river
4 = ponds
5 = lakes

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Status Survey of the Arkansas Endemic Crayfish, *Fallicambarus gilpini* Hobbs and Robison

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Abstract

Fieldwork was conducted during 2002-2003 on the rare, Arkansas endemic crayfish, *Fallicambarus gilpini* Hobbs and Robison. Collections at 87 localities revealed this crayfish at 8 sites, all located in southeastern Arkansas in Jefferson and Cleveland counties which significantly expands its known range. *Fallicambarus gilpini* was generally found inhabiting roadside ditches and areas of standing water where it was always taken from upslope areas away from the static water. A sex ratio of 1:1.3 males to females was determined for this species. Ovigerous females were collected from burrows on 20 March 2003. A conservation status of "threatened" is recommended for this crayfish species.

Introduction

Crayfish represent 1 of the largest aquatic faunal groups in North America north of Mexico with approximately 353 known species or nearly two thirds of the world's crayfish fauna (Butler, et al. 2003). To illustrate how poorly crayfishes are known, Williams et al. (1997) reported common names for less than 28% of the species they listed.

Hobbs (1969) proposed the taxon *Fallicambarus* to receive 8 species of crayfishes that had been formerly assigned to the more commonly known crayfish genus, *Cambarus*. Hobbs (1973) revised the genus *Fallicambarus* and divided this assemblage of the then known 11 species into 2 subgroups or subgenera. Six were placed in the nominate subgenus *Fallicambarus*, and 5 species were placed in the subgenus *Creaserinus*, which presently includes *F. gilpini*. Currently, there are 16 species included in the genus *Fallicambarus*, 7 in the subgenus *Fallicambarus* and 9 in the subgenus *Creaserinus*.

The genus *Fallicambarus* is thought to have originated in southwestern Arkansas on the West Gulf Coastal Plain (Bouchard and Robison, 1980). Of the 16 known species of *Fallicambarus* in North America, 8 occur in Arkansas. Six of the 8 crayfish species, *Fallicambarus strawni*, *F. caesius*, *F. jeanae*, *F. gilpini*, *F. harpi* and *F. petilicarpus*, are endemic to Arkansas (Robison and Allen, 1995). Distribution, biology, and conservation status of most of these state endemics are poorly known. One of the endemic species is *F. gilpini* which was originally described by Hobbs and Robison (1985) from 3 localities in the vicinity of Pine Bluff, Jefferson County, Arkansas, and is the subject of this investigation.

General Habitat Description.—*Fallicambarus* species are rarely found in permanent bodies of water and as adults frequent temporary pools or runoff only after rains or during floods (Hobbs and Robison, 1989). As primary burrowers, they inhabit burrows where the water table does not drop more than a meter or so beneath the surface for most of the year. Hydrophilic sedges characterize such areas and many occur near highways in roadside ditches or low-lying areas near the roadbed.

Characteristically, burrows of *Fallicambarus* crayfish are occasionally topped with slender chimneys, although more often the burrows are marked by irregular mounds of earthen pellets of a size proportional to that of the crayfish occupant. In rare cases, large colonies of these crayfishes occupy an entire field.

Taxonomic Status.—*Fallicambarus gilpini* was originally described by Hobbs and Robison (1989) from Jefferson County, Arkansas. *F. gilpini* has its closest affinities with *F. caesius* (Hobbs and Robison; 1989). The 2 species share many features, including being the only typically blue members of the genus and the only ones that lack a ventrolateral row of tubercles on the merus of the first cheliped. The most readily observed features that distinguish the 2 species are the absence of tubercles on the mesial surface of the dactyl of the chela and the presence of a distolateral spine on the mesial ramus of the uropod in *F. gilpini*. While the close relationship of *F. caesius* and *F. gilpini* is acknowledged, discovery of an undescribed species in Bastrop, Louisiana recently may alter this view. These specimens appear to have a number of characteristics in common with *F. gilpini* (Joseph Fitzpatrick, pers. comm.). Twenty specimens of this new related form were collected by George Patton and Martha Ann Messenger and sent to Keith Crandall, Brigham Young University, for DNA analysis. HWR and Crandall are currently studying this form.

The objectives of this study were to determine the relative abundance and distributional limits of *F. gilpini*; to gather data on life history aspects of *F. gilpini* including information on habitat, description of burrows, and reproductive period; to gather data on ecological requirements of *F. gilpini*; and to assess the current conservation status (as to rarity) of *F. gilpini*.

Methods and Materials

Fieldwork was conducted from September 2002 hrough the spring and early summer of June 2003. Most ollecting occurred in March, April, May, and into early une 2003, when conditions were optimal. *Fallicambarus gilpini* is a primary burrower, i.e. it burrows all year long in 1 place and rarely exits, therefore to collect specimens, it is necessary to physically dig individuals out once the burrow is discovered. In addition to digging specimens from burrows, baited strings and crayfish traps were used; however, excavation proved to be the superior method of collecting specimens of *F. gilpini.*

While most specimens were released unharmed, a few specimens were preserved in 95% ethyl alcohol and deposited in the Brigham Young University Crayfish Collection after identification to species.

Prior to this study, *F. gilpini* was known from only three localities in Jefferson County. Based on this localized distribution, a search pattern for additional populations was centered on the type locality and radiated outward from this area and Jefferson County. Six counties in that circle were searched, as well as Jefferson County itself. Collection sites were searched for by driving area highways and looking for chimneys in the roadside ditches. This method has previously produced good results for members of the genus *Fallicambarus.* Crayfishes were collected from 87 sites where burrows were seen in the 7 county search area in an effort to locate additional populations of *F. gilpini.*

Results and Discussion

Habitat.–Inspection of the type locality began in September 2002 and continued monthly until May 2003, revealing no burrowing activity prior to March. The first burrows of *F. gilpini* were seen on 20 March 2003 at the type locality. The height of burrowing activity was 25 April 2003; 27 burrows were seen at the type locality; and burrowing activity was greatest at other locations in Jefferson County.

Fallicambarus gilpini has been taken only in complex burrows consisting of branching galleries, several of which, except in dry seasons, reach the surface, some of their openings marked by rather crudely constructed turrets (Hobbs and Robison, 1989). In this study 12 burrows of *F. gilpini* were completely excavated, and all were complex burrows with branching galleries. Crudely constructed turrets topped ten of these burrows. Of the 12 burrows excavated completely, burrow depth ranged from 37.5 cm to 77.5 cm and chimney height ranged from 2.5 cm to 10 cm. In all cases excavated, burrows of *F. gilpini* were complex burrows in sandy clay soil situated in wet grassy areas, often with abundant sedges nearby. No burrows were found in or directly adjacent to standing water.

In this study it was noted that burrows of F. gilpini were always situated high up on the seepage slope and never down near the standing water areas, just as reported previously by Hobbs and Robison (1989). Hobbs and Robison (1989) hypothesized that *F. gilpini* might prefer areas in which the groundwater is moving rather than static. In areas where *F. gilpini* was collected syntopically with *F. fodiens,* the latter was always collected from burrows situated in areas in which the water was more static while the burrows of *F. gilpini* were away from the static water more upslope.

Distribution.–*Fallicambarus gilpini* was known from only three locations prior to this study (Hobbs and Robison, 1989). These 3 sites are all located within Jefferson County, Arkansas (Fig. 1). The sites are: (1) Type Locality: Roadside seepage, 4.96 km south of southern junction of State Route 54 and U.S. Highway 79 at junction of latter with Pepperridge Road (T7S, R10W, Sec. 19), approximately 17.6 km south of Pine Bluff and about 4.8 km north of the Cleveland County line; (2) Roadside ditch, 0.32 km south of Pine Bluff on U. S. Highway 79; and (3) Roadside seepage, 5.76 km north of Cleveland County line on U.S. Highway 79.

Searches for additional populations of *F. gilpini* were made in 6 counties contiguous with Jefferson County including Lonoke, Arkansas, Lincoln, Cleveland, Grant and Pulaski, plus Jefferson County itself. Only 1 new population was discovered in Cleveland County, and 4 additional populations were discovered in Jefferson County (See below). Interestingly, no populations were discovered north of the Arkansas River, thus all known populations of *F. gilpini* occur south of the Arkansas River.

New populations discovered during this study are as follows: Jefferson County: (1) Roadside seepage, ca. 6.4 km south of Pine Bluff on U. S. Hwy. 79 (Sec. 17, T7S, R10W). 20 March 2003. H. W. Robison.; (2) Roadside ditch ca. 11.2 km south of Pine Bluff on U. S. Hwy. 79 (Sec. 20, T7S, R10W). 18 April 2003. H. W. Robison; (3) Roadside seepage, ca. 1.6 km south of Pine Bluff on U. S. Hwy. 79 (Sec. 3, T7S, R10W). 25 April 2003. H. W. Robison.; and (4) Roadside seepage along U. S. Hwy. 79, ca. 3.2 km south of Pine Bluff (Sec. 9, T7S, R10W). 26 April 2003. H. W. Robison. Cleveland County: (1) Roadside seepage ca. 5.6 km south of the Cleveland-Jefferson County line on U. S. Hwy. 79 (Sec. 23, T8S, R11W). 25 April 2003. H. W. Robison.

In summary, the distribution of *F. gilpini* now includes eight localities in two Arkansas counties, Jefferson and Cleveland (Fig. 1). A new population was discovered in Cleveland County, as well as 4 new sites in Jefferson County. At each of these locations, *F. gilpini* was found to be a highly localized and uncommon crayfish. It was never abundant at any site collected during the study. *Fallicambarus fodiens* was present and always numerically superior at each site where *F. gilpini* was collected.

Biological Aspects.–Nineteen collections of *F. gilpini* were made during this 1-year study (Table 1). Form I males were first collected on 20 March 2003 from the type-locality, and were only collected in March and April. Seventeen males were collected, of which 5 were Form I, 9 were Form II, and 3 were juveniles.

Twenty-two females were taken in the study, of which 17 were adults and 5 were juveniles (Table 1). Two ovigerous females were collected from burrows on 20 March 2003. Hobbs and Robison (1989) reported three ovigerous females taken from burrows on 11 March 1988. One of these had a carapace length of 22.3 mm and 18 eggs, a second had a carapace length of 24.9 mm and 20 eggs and the third had a carapace length 0f 25.5 mm and 35 eggs (Hobbs and Robison, 1989). Of the 2 ovigerous females collected in this study, 1 had a carapace length of 24.2 mm and carried 26 eggs while the other specimen had a carapace length of 23.7 mm and carried 17 eggs.

During this study 437 individual crayfishes were collected, including six additional species taken while searching for *F. gilpini*. These species included *F. fodiens*, *Procambarus clarkii*, *Procambarus acutus*, *Orconectes lancifer*, *Cambarus ludovicianus*, and *Faxonella clypeata*.

Sex Ratio.–During this study collections of *F. gilpini* included 17 males (5 Form I males, 9 Form II males, and 3 juvenile males) versus 22 females (17 adult females and 5 juvenile females). This provides a sex ratio of 1:1.3 (male to female) for *F. gilpini*.

Conservation Status.–Because of the long-term degradation of freshwaters in North America, it should not come as a surprise that some freshwater crustacean species are having difficulty surviving (Schuster, 1997). In particular, a number of crayfishes in the United States are in trouble and their continued survival is in question. The degree of crayfish imperilment may exceed that of fishes and is second only to the most imperiled group in North America, freshwater mussels (Master et al., 2000; Butler et al., 2003).

Taylor et al. (1996) published a paper entitled "Conservation Status of Crayfishes of the United States and Canada" which provides the most current conservation estimates dealing with crayfishes. They found 19.2% of the crayfish fauna in the United States and Canada to be endangered, 13.3% threatened and 14.8% of special concern. While 52.0% or 176 of the 338 native crayfishes were considered "stable," a whopping 48.0% or 162 species were in need of some conservation status! Only 2 species of crayfish inhabiting Arkansas are currently listed as endangered under the Endangered Species Act, *Cambarus aculabrum* and *C. zophonastes*, both of which are cave forms with very limited distribution.

In their report, Taylor et al., (1996) listed *Fallicambarus gilpini* as "endangered" based on the best information available at the time. In this survey it appears that *F. gilpini* is slightly more common than previously believed, having been found at 5 additional localities in 2 counties. The known range now stands at 8 localities in 2 counties in southeast Arkansas where it is quite localized and never abundant. It is therefore recommended to move *F. gilpini* from its "endangered" status to a status of "threatened." Future monitoring is needed to document trends in the population.

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Table 1. Frequency of occurrence of form I males, form II males, females, and juveniles in collections of *Fallicambarus gilpini*.*

Month	Number of Collections	Number of Individuals				
		Form I Males	Form II Males	Females	Juveniles	Total
March	3	2	0	1	0	3
April	10	3	5	8	3	19
May	4	0	3	6	5	14
June	2	0	1	2	0	3
Totals	**19**	**5**	**9**	**17**	**8**	**39**

* No specimens of *F. gilpini* were collected in January, February, or July through December of either 2002 or 2003.

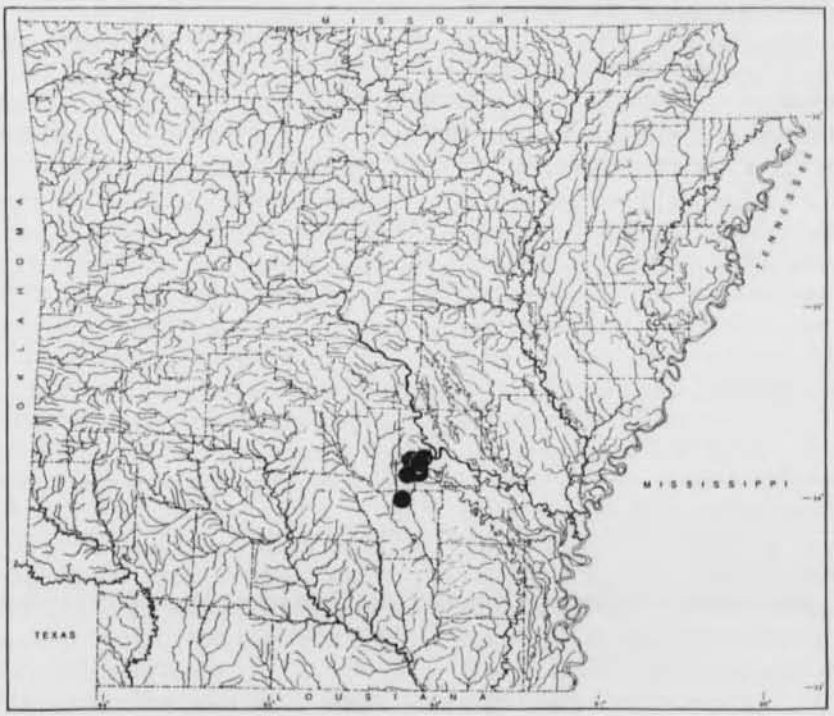

Fig. 1. Known localities of *Fallicambarus gilpini* following 2002-2003 survey.

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A Songbird Inventory for Arkansas Post National Memorial

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Abstract

Two geographically separate units of Arkansas Post National Memorial were surveyed via fixed-radius plots to document songbird species composition, richness, and diversity by migratory status and nesting guild. At the Memorial Unit, 60 species were recorded with the Brown-headed Cowbird, Red-winged Blackbird, and Northern Cardinal being most common. Individuals of these three species comprised 30% of the total number of birds recorded despite representing only 5% of the species encountered. About 2½ times more resident birds were recorded than migratory birds. However, species richness and diversity of resident and migratory species were similar. The number of individuals, species richness, and diversity of canopy nesting species were greater than other nesting guilds. At the Osotouy Unit, 42 species were recorded with the most common species encountered being the Indigo Bunting, Carolina Wren, and Yellow-billed Cuckoo. Individuals of these 3 species comprised 30% of the total number of birds recorded despite representing only 7% of the species encountered. About 50% fewer resident birds were recorded than migratory birds. Migratory birds represented approximately 40% more species than resident birds. Likewise, diversity was greater for migratory species than for resident species. As in the Memorial Unit, the number of individuals, species richness, and diversity of canopy-nesting species were greater than other nesting guilds. No federal or state threatened or endangered species were documented, but 8 species currently tracked by the Arkansas Natural Heritage Commission were documented. These results have implications for future park management activities, particularly in respect to potential development plans at the Osotouy Unit.

Introduction

Congress passed the National Parks Omnibus Management Act in 1998 in response to concerns about the condition of natural resources within the national parks. The act requires each park to gather baseline inventory data on pertinent natural resources, data that will provide a pivotal step toward establishing an effective monitoring program furthering the ability to effectively manage and protect park resources and abide by the National Park Service (NPS) mission statement. The NPS responded with the Natural Resource Challenge program, including the establishment of biome-based inventory and monitoring networks (NPS, 1999). The Heartland Network, as part of the NPS Inventory and Monitoring program, has undertaken inventories of vascular plants and vertebrates within 15 parks in 8 midwestern states. Stemming from this challenge and a concern regarding the status of songbird populations at Arkansas Post National Memorial, an inventory was deemed necessary to establish baseline data of songbirds within the park.

Arkansas Post National Memorial, including the Osotouy Unit, provides refuge to numerous species of songbirds. Songbirds are an ecologically important faunal group that can be influenced by structural and floristic habitat alterations that may result from a variety of naturally occurring ecosystem processes and/or management activities (Wiens, 1989). Songbirds help facilitate seed and fungi dispersal, help control insect numbers, play essential roles in food web dynamics, and can create habitat for other wildlife species through excavation of cavities (Hunter, 1999). In addition to their ecological values, nongame birds are important as a recreational resource to millions of people who watch and feed birds (U.S. Dept. of the Interior, 2002). Neotropical migratory birds are of particular research interest due to recent evidence of long-term population declines in many species (Finch, 1991; Robbins et al., 1989).

An inventory of bird species is a necessary first step toward understanding how songbird populations relate to natural and cultural resources and associated management activities at the park, and will also help the park better manage resources and predict the possible impacts of management decisions on avian species (an important component of the National Environmental Policy Act). It will also provide managers with information about future research, such as fecundity surveys on species of concern. Additionally, an inventory of bird species establishes a baseline for future monitoring efforts aimed at detecting population/species composition trends. Thus, the objective for this inventory was the assessment of species composition, richness, evenness, and diversity of migrant and resident species.

Study Area

Arkansas Post National Memorial is made up of 2 units, the Memorial and Osotouy Units. Both units are located in the southeastern portion of Arkansas County, Arkansas. The units are not contiguous and are separated by 8.0 km. The Memorial Unit is located 11.2 km south of Gillett, and the Osotouy Unit is located approximately 12.8 km from the community of Tichnor. The Memorial Unit (157.6 ha) is a

peninsula surrounded by Moore and Post bayous along the north/northwest border and Post Lake, a backwater of the Arkansas River, on the north and northeastern border. The Osotouy Unit (145.8 ha) is bordered on the southwest by an old oxbow of the Arkansas River, Lake Dumond, and on the south by the White River National Wildlife Refuge. Remaining boundaries are adjacent to private land.

Both the Memorial and Osotouy Units are characterized by a terrace landscape, flat terrain, and various stands of upland and lowland hardwoods, interspersed with bayous and swamps. The Memorial Unit consists of a mosaic of successional seres within forested vegetation types that roughly follow a gradient from bottomland forest types that occupy mesic sites to upland types that occupy more xeric sites. This mosaic combined with maintained lawns, trails, and roads creates a diverse and fragmented environment. Forest composition at the Osotouy Unit is similar to that at the Memorial Unit. However, fewer successional seres are present, though some portions of the Osotouy Unit have been logged or are under cultivation. Land immediately adjacent to both units is either under agricultural cultivation or has been logged.

Methods

Arkansas Post National Memorial was surveyed to determine current songbird species composition from 9 June – 7 August 2003 via fixed radius census plots. Fourteen 50-m fixed-radius bird census plots were established at the Memorial Unit, whereas 8 were established at the Osotouy Unit due to its more homogenous landcover. Plots were located to provide an adequate sample of bird species that occur in the various vegetation types. However, the size of vegetation areas at both units precluded replication within those areas. Plots were situated to provide easy access for future monitoring purposes (i.e., along roads and trails). At the Memorial Unit, the interspersion and juxtaposition of a variety of vegetation types along with maintained lawns, trails, and roads provided a landscape with numerous edges and little continuity. Thus, placement of census points along roads and trails was reasonable for this particular landscape. Each point was recorded (Lat/Lon) using a eTrex Vista Global Positioning System (GPS) portable hand-held unit with WAAS enabled accuracy less than 3m.

Each plot was sampled using a 5-minute count of all songbirds heard or seen. All counts were conducted within 3.5 hours of sunrise on days with little or no rain and with winds < 6 kph. Plots were sampled 3 times each by 2 observers on different days; thus, each plot was sampled a total of 6 times. Species that do not breed in the area, species for which point sampling is an inappropriate sampling methodology, and flyovers were recorded but not used in the analyses. Species nomenclature follows the American Ornithologist Union Checklist for North American Birds (2004).

Mean numbers of individuals, species richness (number of species), diversity (Shannon diversity index), and evenness (Pielou's J) were computed for all breeding birds combined and for each of the following subsets: residents, migrants (short- and long-distance combined), canopy nesters, cavity nesters, ground nesters, and shrub nesters. Species associated with multiple nesting preferences were included in each of the appropriate nesting guilds for analysis. Resident and migratory means were compared using an independent t-test. Nesting guild means were compared using a one-way ANOVA and tukey's mean separation test. All analyses were conducted using SPSS 13.0 (SPSS, Inc., 2004).

Results

Memorial Unit.–A total of 1,153 individual birds ($\bar{x}$ = 164/day) representing 60 species ($\bar{x}$ = 32/day) was recorded (Tables 1 and 2). The most common species encountered was the Brown-headed Cowbird (*Molothrus ater*), followed by the Red-winged Blackbird (*Agelaius phoeniceus*) and the Northern Cardinal (*Cardinalis cardinalis*). Individuals of these 3 species comprised 30% of the total number of birds recorded despite representing only 5% of the species encountered.

About 2½ times more resident birds ($\bar{x}$ = 116 individuals/day) were recorded than migratory birds ($\bar{x}$ = 48 individuals/day) (Table 2). A similar number of resident ($\bar{x}$ = 17) and migratory ($\bar{x}$ = 16) species were encountered (Table 2). Likewise, diversity was similar for resident ($\bar{x}$ = 2.4/day) and migratory ($\bar{x}$ = 2.4/day) species (Table 2). However, evenness was greater for migratory species ($\bar{x}$ = 0.88/day) than for resident species ($\bar{x}$ = 0.85/day) (Table 2).

An average of 78 canopy-nesting birds was recorded per day, compared to an average of 32 cavity nesters, 56 shrub nesters, and 12 ground nesters (Table 3). Additionally, an average of 16 canopy-nesting species was encountered per day (Table 3). This was approximately twice as many species as that recorded for cavity nesters ($\bar{x}$ = 9) and shrub nesters ($\bar{x}$ = 8) and 8 times greater than the number of recorded ground nesting species ($\bar{x}$ = 2) (Table 3). Diversity ($\bar{x}$ = 2.4/day) of canopy nesting species was also greater than diversity of other nesting guilds (Table 3). Cavity nesters were the second most diverse group ($\bar{x}$ = 1.8/day), followed by shrub ($\bar{x}$ = 1.4/day) and ground ($\bar{x}$ = 0.5/day) nesters (Table 3). Evenness ($\bar{x}$ = 0.88/day) of canopy nesting species was similar to that of cavity nesters ($\bar{x}$ = 0.84/day) and greater than shrub ($\bar{x}$ = 0.70/day) or ground ($\bar{x}$ = 0.59/day) nesters (Table 3).

Osotouy Unit.–A total of 472 individual birds ($\bar{x}$ = 74/day) representing 42 species ($\bar{x}$ = 19/day) was recorded (Tables 4 and 5). The most common species encountered

as the Indigo Bunting (*Passerina cyanea*), followed by the arolina Wren (*Thryothorus ludovicianus*) and the Yellow-illed Cuckoo (*Coccyzus americanus*). Individuals of these iree species comprised 30% of the total number of irds recorded despite representing only 7% of the ›ecies encountered.

About 50% fewer resident birds ($\bar{x}$ = 28 individuals/day) ere recorded than migratory birds ($\bar{x}$ = 46 individuals/day) Table 5). Migratory birds represented approximately 40% nore species ($\bar{x}$ = 11/day) than resident birds ($\bar{x}$ = 8/day) (Table 5). Likewise, diversity was greater for migratory species ($\bar{x}$ = 2.2/day) than for resident species ($\bar{x}$ = 1.8/day) (Table 5). However, evenness values for migratory ($\bar{x}$ = 0.90/day) and resident ($\bar{x}$ = 0.89/day) species were similar (Table 5).

An average of 40 canopy-nesting birds was recorded per day, compared to 13 cavity nesters, 21 shrub nesters, and 8 ground nesters (Table 6). Additionally, an average of 10 canopy-nesting species was encountered per day (Table 6). This was approximately twice as many species as that recorded for cavity nesters ($\bar{x}$ = 4) and shrub nesters ($\bar{x}$ = 5) and 5 times greater than the number of recorded ground nesting species ($\bar{x}$ = 2)(Table 6). Diversity of canopy nesting species ($\bar{x}$ = 2.1/day) was also greater than diversity of other nesting guilds (Table 6). Shrub nesters were the second most diverse group ($\bar{x}$ = 1.3/day), followed by cavity ($\bar{x}$ = 1.1/day) and ground ($\bar{x}$ = 0.2/day) nesters (Table 6). Evenness was least for ground nesting species ($\bar{x}$ = 0.58/day) but was similar among canopy ($\bar{x}$ = 0.91/day), cavity ($\bar{x}$ = 0.79/day), and shrub ($\bar{x}$ = 0.81/day) nesters (Table 6).

Eight species currently tracked by the Arkansas Natural Heritage Commission (2002) were documented. These include 2 that are currently being inventoried: Common Moorhen (*Gallinula chloropus*) and Purple Gallinule (*Porphyrio martinica*); 2 that are being monitored: Great Blue Heron (*Aredea herodias*) and Double-crested Cormorant (Phalacrocorax auritus); and 4 that are on the watch list: Yellow Warbler (*Dendorica petechia*), Red-headed Woodpecker (*Melanerpes erythrocephalus*), Hairy Woodpecker (*Picoides villosus*), and Blue-winged Warbler (*Vermivora pinus*).

Discussion

The composition and structure of the bird communities found at both units are dissimilar. Numerically, bird species richness and diversity was greater at the Memorial Unit compared to the Osotouy Unit for both resident and migratory birds as well as for all nesting guilds. The 2 most common species at the Memorial Unit were the Brown-headed Cowbird and the Red-winged Blackbird. In contrast, the 2 most common species at the Osotouy Unit were the Indigo Bunting and the Carolina Wren. These differences in bird communities are likely the result of differences in the composition, structure, and patterns of vegetation. However, because the bird surveys were conducted relatively late in the breeding season (9 June – 7 August), it is possible that a few uncommon species were not detected or were under-represented.

At the Memorial Unit, the interspersion and juxtaposition of a variety of vegetation types along with maintained lawns, trails, and roads characterize a diverse and fragmented landscape with numerous edges. This variety of habitats provides for a diverse bird community. However, combined with a close proximity to agricultural fields, the diverse, fragmented habitat also creates an ideal environment for the Brown-headed Cowbird (Temple and Cary, 1988; Wilcove, 1985; Yahner and Scott, 1988). The prevalence of the Brown-headed Cowbird raises a concern about the level of nest parasitism occurring at the unit. If nest parasitism is high nest success rates could be low and thus this unit could potentially represent a population sink for some bird species (Robbins et al., 1989).

Though the Osotouy Unit is in relatively close proximity to agricultural fields and bodies of water, it represents a less diverse and less fragmented environment. Thus, the number and diversity of bird species is less than those found at the Memorial Unit. However, the Brown-headed Cowbird represented only 0.4% of the birds encountered at Osotouy. In the future, any development at Osotouy should consider possible ramifications of changes to habitat, particularly in respect to fragmentation that could result in an increase in nest parasitism by Brown-headed Cowbirds.

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Table 1. List of birds recorded at the Memorial Unit of Arkansas Post National Memorial, June – August 2003.

Common Name	Scientific Name	# of Individuals	% Total
Brown-headed Cowbird	*Molothrus ater*	163	14.1
Red-winged Blackbird	*Agelaius phoeniceus*	108	9.4
Northern Cardinal	*Cardinalis cardinalis*	86	7.5
Red-bellied Woodpecker	*Melanerpes carolinus*	59	5.1
Carolina Wren	*Thryothorus ludovicianus*	59	5.1
Eastern Wood Peewee[4]	*Contopus virens*	55	4.8
Yellow-billed Cuckoo[4]	*Coccyzus americanus*	51	4.4
Mourning Dove	*Zenaida macroura*	46	4.0
Tufted Titmouse	*Baeolophus bicolor*	38	3.3
Common Grackle	*Quiscalus quiscula*	36	3.1
Carolina Chickadee	*Poecile carolinensis*	36	3.1
Northern Rough-winged Swallow[1]	*Stelgidopteryx serripennis*	35	3.0
Acadian Flycatcher[4]	*Empidonax virescens*	31	2.7
Great Egret[1]	*Ardea alba*	30	2.6
Northern Mockingbird	*Mimus polyglottos*	28	2.4
Summer Tanager[4]	*Piranga rubra*	25	2.2
Fish Crow	*Corvus ossifragus*	21	1.8
Blue-gray Gnatcatcher[4]	*Polioptila caerulea*	21	1.8
Blue Jay[4]	*Cyanocitta cristata*	19	1.6
Downy Woodpecker	*Picoides pubescens*	18	1.6
Baltimore Oriole[4]	*Icterus galbula*	15	1.3
American Crow[4]	*Corvus brachyrhynchos*	15	1.3
Cattle Egret[3]	*Bubulcus ibis*	14	1.2
Barn Swallow[1]	*Hirundo rustica*	10	0.9
Hairy Woodpecker	*Picoides villosus*	9	0.8

ble 1. Continued.

Common Name	Scientific Name	# of Individuals	% Total
Wood Thrush[4]	*Hylocichla mustelina*	9	0.8
Eastern Towhee	*Pipilo erythrophthalmus*	8	0.7
Eastern Bluebird[4]	*Sialia sialis*	8	0.7
Northern Parula[4]	*Parula americana*	8	0.7
Great-crested Flycatcher[4]	*Myiarchus crinitus*	6	0.5
Indigo Bunting[4]	*Passerina cyanea*	6	0.5
Brown Thrasher	*Toxostoma rufum*	6	0.5
White-eyed Vireo[4]	*Vireo griseus*	6	0.5
Orchard Oriole[4]	*Icterus spurius*	5	0.4
Prothonotary Warbler[4]	*Protonotaria citrea*	5	0.4
Northern Flicker	*Colaptes auratus*	5	0.4
Common Moorhen[1]	*Gallinula chloropus*	4	0.3
Eastern Kingbird[4]	*Tyrannus tyrannus*	4	0.3
Mallard[3]	*Anas platyrhynchos*	4	0.3
American Robin	*Turdus migratorius*	4	0.3
Yellow Warbler[4]	*Dendroica petechia*	4	0.3
Double-crested Comorant[2]	*Phalacrocorax auritus*	3	0.3
Pileated Woodpecker	*Dryocopus pileatus*	3	0.3
Purple Gallinule[1]	*Porphyrio martinica*	3	0.3
Red-headed Woodpecker	*Melanerpes erythrocephalus*	3	0.3
Blue Grosbeak[4]	*Guiraca caerulea*	2	0.2
Belted Kingfisher[1]	*Ceryle alcyon*	2	0.2
European Starling	*Sturnus vulgaris*	2	0.2
Great Blue Heron[1]	*Ardea herodias*	2	0.2
Ovenbird[2]	*Seiurus aurocapillus*	2	0.2

Table 1. Continued.

Common Name	Scientific Name	# of Individuals	% Total
Red-eyed Vireo[4]	*Vireo olivacaus*	2	0.2
Chimney Swift[3]	*Chaetura pelagica*	1	0.1
Blue-winged Warbler[2]	*Vermivora pinus*	1	0.1
Horned Lark	*Eremophila alpestris*	1	0.1
Kentucky Warbler[4]	*Oporornis formosus*	1	0.1
Barred Owl[1]	*Strix varia*	1	0.1
Pine Warbler	*Dendroica pinus*	1	0.1
American Redstart[4]	*Setophaga ruticilla*	1	0.1
Red-shouldered Hawk[3]	*Buteo lineatus*	1	0.1
Gray Catbird[4]	*Dumetella carolinensis*	1	0.1
Total		**1,153**	**100%**

[1] Inappropriate sampling technique.

[2] Non-breeding migrant.

[3] Recorded only as a flyover.

[4] Breeding, migratory species (short- or long-distance).

Table 2. Mean number per day (SD) of individuals and species, and mean diversity and evenness per day (SD) by migratory status for birds recorded at the Memorial Unit of Arkansas Post National Memorial, June – August 2003.

		Migratory Status		
Variable	**All Species**	**Resident**	**Migrant**	***p*[1]**
Individuals	164.2 (13.64)	116.0 (13.34)	48.2 (4.92)	<0.001
Species	32.3 (3.62)	16.8 (2.32)	15.5 (1.76)	0.288
Diversity	2.990 (0.0587)	2.377 (0.0801)	2.410 (0.1004)	0.560
Evenness	0.862 (0.0240)	0.845 (0.0303)	0.881 (0.0195)	0.033

[1]Probability associated with independent t-test of H_o: $\bar{x}$ resident = $\bar{x}$ migrant and H_A: $\bar{x}$ resident ≠ $\bar{x}$ migrant.

ıble 3. Mean number per day (SD) of individuals and species, and mean diversity and evenness per day (SD) by nesting guild r birds recorded at the Memorial Unit of Arkansas Post National Memorial, June – August 2003.

	Nesting Guild			
Variable	**Canopy**	**Cavity**	**Shrub**	**Ground**
Individuals	78.2 A[1] (10.87)	31.7 B (3.72)	56.2 C (9.79)	11.7 D (2.16)
Species	16.0 A (2.10)	8.8 B (0.98)	8.0 B (1.27)	2.3 C (1.03)
Diversity	2.434 A (0.0610)	1.821 B (0.0927)	1.436 C (0.0977)	0.456 D (0.3189)
Evenness	0.881 A (0.0323)	0.838 A (0.0162)	0.695 B (0.0339)	0.585 B (0.1373)

[1]Means in the same row followed by the same letter are not significantly different ($P < 0.05$).

Table 4. List of of birds recorded at the Osotouy Unit of Arkansas Post National Memorial, June – August 2003

Common Name	**Scientific Name**	**# of Individuals**	**% Total**
Indigo Bunting[4]	*Passerina cyanea*	61	12.9
Carolina Wren	*Thryothorus ludovicianus*	43	9.1
Yellow-billed Cuckoo[4]	*Coccyzus americanus*	42	8.9
Northern Cardinal	*Cardinalis cardinalis*	42	8.9
Eastern Wood Peewee[4]	*Contopus virens*	33	7.0
Wood Thrush[4]	*Hylocichla mustelina*	30	6.4
Acadian Flycatcher[4]	*Empidonax virescens*	29	6.1
Mourning Dove	*Zenaida macroura*	24	5.1
Red-bellied Woodpecker	*Melanerpes carolinus*	21	4.4
Summer Tanager[4]	*Piranga rubra*	19	4.0
White-eyed Vireo[4]	*Vireo griseus*	14	3.0
Blue Jay[4]	*Cyanocitta cristata*	13	2.8
Blue Grosbeak[4]	*Guiraca caerulea*	12	2.5
Tufted Titmouse	*Baeolophus bicolor*	12	2.5

Table 4. Continued.

Common Name	Scientific Name	# of Individuals	% Total
Red-eyed Vireo[4]	*Vireo olivaceus*	8	1.7
Great Egret[1]	*Ardea alba*	8	1.7
Carolina Chickadee	*Poecile carolinensis*	7	1.5
Northern Flicker	*Colaptes auratus*	5	1.1
Prothonotary Warbler[4]	*Protonotaria citrea*	5	1.1
Downy Woodpecker	*Picoides pubescens*	4	0.8
Fish Crow	*Corvus ossifragus*	3	0.6
Red-shouldered Hawk[3]	*Buteo lineatus*	3	0.6
Pileated Woodpecker	*Dryocopus pileatus*	3	0.6
Common Yellow-throat[4]	*Geothlypis trichas*	2	0.4
Cattle Egret[3]	*Bubulcus ibis*	2	0.4
Brown-headed Cowbird	*Molothrus ater*	2	0.4
Northern Mockingbird	*Mimus polyglottos*	2	0.4
Wild Turkey[1]	*Meleagris gallopavo*	2	0.4
Ruby-throated Hummingbird[4]	*Archilochus colubris*	2	0.4
Red-winged Blackbird	*Agelaius phoeniceus*	2	0.4
Ovenbird[2]	*Seiurus aurocapillus*	2	0.4
Great Blue Heron[1]	*Ardea herodias*	2	0.4
Northern Bobwhite[1]	*Colinus virginianus*	2	0.4
Blue-gray Gnatcatcher[4]	*Polioptila caerulea*	2	0.4
Kentucky Warbler[4]	*Oporornis formosus*	2	0.4
Chipping Sparrow[4]	*Spizella passerina*	1	0.2
Double-crested Comorant[2]	*Phalacrocorax auritus*	1	0.2
Hairy Woodpecker	*Picoides villosus*	1	0.2
Red-tailed Hawk[1]	*Buteo jamaicensis*	1	0.2

ble 4. Continued.

Common Name	Scientific Name	# of Individuals	% Total
Brown Thrasher	*Toxostoma rufum*	1	0.2
Eastern Phoebe	*Sayornis phoebe*	1	0.2
American Crow	*Corvus brachyrhynchos*	1	0.2
Total		472	100%

[1] Inappropriate sampling technique.
[2] Non-breeding migrant.
[3] Recorded only as a flyover.
[4] Breeding, migratory species (short- or long-distance).

Table 5. Mean number per day (SD) of individuals and species, and mean diversity and evenness per day (SD) by migratory status for birds recorded at the Osotouy Unit of Arkansas Post National Memorial, June – August 2003.

		Migratory Status		
Variable	All Species	Resident	Migrant	*P*[1]
Individuals	73.5 (5.24)	28.0 (4.82)	45.5 (6.35)	<0.001
Species	19.3 (1.86)	8.2 (1.84)	11.2 (1.33)	0.009
Diversity	2.695 (0.0851)	1.841 (0.2145)	2.163 (0.0963)	0.012
Evenness	0.911 (0.0105)	0.885 (0.0300)	0.899 (0.0138)	0.187

[1] Probability associated with independent t-test of H_o: $\bar{x}$ resident = $\bar{x}$ migrant and H_A: $\bar{x}$ resident ≠ $\bar{x}$ migrant.

Table 6. Mean number per day (SD) of individuals and species, and mean diversity and evenness per day (SD) by nesting guil for birds recorded at the Osotouy Unit of Arkansas Post National Memorial, June – August 2003.

	Nesting Guild			
Variable	**Canopy**	**Cavity**	**Shrub**	**Ground**
Individuals	39.8 A[1] (3.06)	12.7 B (2.73)	21.0 C (2.76)	7.7 D (1.63)
Species	9.7 A (0.82)	4.3 B (1.63)	5.2 B (0.75)	1.5 C (0.55)
Diversity	2.069 A (0.1025)	1.119 B (0.4170)	1.328 B (0.1689)	0.200 C (0.2268)
Evenness	0.913 A (0.0194)	0.793 A (0.0803)	0.811 A (0.0477)	0.578 B (0.1300)

[1]Means in the same row followed by the same letter are not significantly different ($P < 0.05$).

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Seasonal Incidence of Sperm within the Spermathecae of Ouachita Dusky Salamanders (*Desmognathus brimleyorum*) in Arkansas

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Abstract

We examined 91 adult female Ouachita dusky salamanders (*Desmognathus brimleyorum*) to determine the seasonal incidence of sperm within spermathecae. The spermatheca (sperm storage gland) along with supporting tissue was removed from the dorsal cloacal wall of each female and prepared for light microscopy. We recorded the reproductive condition of females (diameter of enlarged ovarian follicles = EOF) and found large aggregates of sperm within the spermathecae during all months, except February (no specimens available). The highest incidence of sperm in spermathecae occurred in July specimens (53%; n = 17). Although the known nesting season runs from July into August in this species, the mating season does not appear to be restricted to spring and summer months. Moreover, females in any month with EOF may or may not possess sperm.

Introduction

The Ouachita dusky salamander, *Desmognathus brimleyorum*, is a large, semi-aquatic plethodontid salamander which ranges throughout the Ouachita Mountains of Arkansas and Oklahoma (Conant and Collins, 1998). Available information on the reproductive biology of this species has been reviewed by several authors (Means, 1999; Petranka, 1998; Trauth et al., 2004), although Petranka (1998) indicated a lack of reproductive information. Both sexes breed annually, but no study has clearly delineated the exact timing and duration of the breeding season (Petranka, 1998). In fact, a combination of several seasonal data sets of information on females as well as on larval size and growth is necessary to clarify this species' breeding phenology. Trauth et al. (1990) reported on the annual oogenic cycle through seasonal sampling of females by examining the total number of ovarian follicles produced, the maximum ovum size, and ovarian clutch size. This kind of data was used in another study (Taylor et al., 1990) to determine a gonosomatic index (GSI) for the species. The GSI peaked during the height of vitellogenesis or growth of ovarian mass in July and, as expected, occurred just prior to ovulation and presumably the onset of oviposition (Taylor et al., 1990; Trauth, 1988). Observations on the presence of nesting females (or egg clutches) and possibly the size of developing embryos have provided an indication of the overall nesting period (Taylor et al., 1990; Trauth, 1988). Herein, we report on the seasonal incidence of sperm within the spermathecae of *D. brimleyorum*. In addition, we include reproductive information on the ovarian cycle of all adult females. These data add critical life-history information that is currently lacking within the reproductive database of this desmognathine salamander.

Materials and Methods

Most of the 91adult female *D. brimleyorum* ($n = 77$) used in this study were collected over a five-year period (1980-1984), and 57% (n = 44) were sampled from May to December, 1980. All specimens were taken from Polk and Montgomery counties and are currently deposited as voucher specimens in the Arkansas State University herpetological collection (ASUMZ). Additional specimens were obtained from the ASUMZ. Salamanders were sacrificed in a dilute chloretone solution within 24-48 hr following capture, fixed in 10% formalin, and preserved in 70% ethanol. The diameter of enlarged (vitellogenic) ovarian follicles (EOF) in each female was also measured to the nearest 0.1 mm with a set of vernier calipers or with the aid of an ocular micrometer. Mean values, when provided, are accompanied by ± 1 standard deviation.

Following preservation, the snout-vent length of each specimen was measured from the tip of the snout to the anterior margin of the vent (range, 62-83 mm; mean = 72.3 ± 5.5). The spermatheca along with supporting tissue was then removed from the cloacal region with a razorblade, and tissue slabs were placed into vials of 70% ethanol. Tissues were dehydrated in a graded series of ethanol, cleared in xylene, embedded in paraffin, sectioned with a rotary microtome into ribbons 8 µm in thickness, stained with Harris hematoxylin, and counterstained with eosin. The descriptive histology of the spermathecal gland of *D.*

brimleyorum has been described elsewhere (Sever and Trauth, 1990) and, thus, is not given herein. All histological slides are deposited in the Arkansas State University Center for Microscopy.

Results

We found large aggregates of sperm (Fig. 1A) within spermathecal sacs of 26 specimens collected from December through August (no February specimens available). Nearly all of these specimens (n = 25) possessed EOF which averaged ≥ 2 mm in diameter (Fig. 2). During the same time period, nearly an equal number of females with EOF ≥ 2 mm in diameter (n = 29) lacked any evidence of sperm within their spermathecae (Fig. 2). We also found 14 females during that time period that exhibited a very small amount (trace) of sperm within their spermathecae (Fig. 1B; 2). Most of these females (n = 8), however, exhibited EOF ≤ 2.0 mm in diameter (Fig. 2). In addition, five females collected from September through November showed a trace of sperm. Number of females with sperm is shown in Fig. 3. For instance, from April through July, the number of females possessing an abundance of sperm generally increased to a peak in July (9/17, 53%), whereas during the same time period the number of females lacking sperm remained greater than the other two categories (except for July values). The percent of total females showing as absence of sperm was also highest from April through July. The number of females possessing sperm decreased sharply following July, but, at the same time, the number of females exhibiting a trace of sperm increased.

Discussion

Long-term sperm storage remains a poorly-studied aspect of the biology of many species of salamanders that exhibit internal fertilization. In a review of urodele courtship and mating glands, Sever (2003) pointed out that very few studies provide a critical analysis of the mating season by noting the presence of sperm within the spermatheca or sperm storage gland(s) found within the roof of the female's cloaca. The timing of mating in plethodontid salamanders can be inferred from a histological examination of the spermathecae (Sever, 2000); the duration of sperm storage can also be derived from an adequate seasonal sample of adults (e.g., Meshaka and Trauth, 1995; Trauth, 1983, 1984). As a general rule, any salamander reproductive researcher attempting to determine the length of sperm retention in females should not only document the duration of the seasonal ovarian cycle, but should also concurrently, examine spermathecal sperm storage (Sever, 2000, 2003).

Trauth (1988) examined the spermathecae of three nesting female *Desmognathus brimleyorum* collected in late July and in mid August and found only traces of residual sperm and the presence of small ovarian follicles (≤ 1.3 mm in mean diameter). We also found females (Fig. 2) exhibitin similar conditions following a probable ovipositional perio (late June – mid July); however, these same morphologica features existed in other females collected during th spring months. Sever (2003) reviewed spermiophagy, a phe nomenon that occurs within the spermathecal epitheliun and the lumina of spermathecal tubules in some species o salamanders. Spermiophagy provides the spermatheca means of removing degenerating sperm prior to the nex mating season (Sever et al., 2001). Our findings suggest however, that viable sperm may be retained within the spermatheca for a prolonged length of time. This time frame may occur from immediately following oviposition to, and possibly including, the time of sperm transfer from spermatophores during the next mating season (i.e., a time span of approximately one year extending from one ovi positional period to the next). Tilley and Hausman (1976), using genotypic comparisons of females and their offspring, determined that multiple inseminations occurred at least 7% of the time in a population of a congeneric desmognathine *D. ochrophaeus.* Moreover, Houck and Schwenk (1984) examined the spermathecae of pre- and post-ovipositional *D. ochrophaeus* and, because of the abundance of sperm in their spermathecae, indicated the strong possibility of sperm competition in this species. The presence of residual sperm throughout the year in *D. brimleyorum* (Fig. 2), therefore, suggests long-term sperm storage and the possibility of sperm competition and multiple paternities in this species. Sever and Hamlett (1998) suggested that residual sperm of desmognathine salamanders may become embedded in spermathecal epithelial cells and are eventually degraded. Whether residual sperm in *D. brimleyorum* are actually capable of fertilization must await future investigations.

Taylor et al. (1990) found that the GSI of male *D. brimleyorum* peaks in August, which means that testicular size had reached maximum size and that sperm can begin evacuating the testes to be stored in the vasa deferentia for mating. We found large aggregates of sperm within the spermathecae of several winter specimens (Fig. 2; n = 5). These findings provide credible evidence that post ovipositional insemination does occur and occurs much earlier than a previously-assumed, spring/early summer mating season.

In conclusion, the present study found support for post-ovipositional, long-term sperm retention in *D. brimleyorum.* Insemination may also occur in some females during the winter months. Females that undergo their oogenic cycle may do so with or without the presence of stored sperm in their spermathecae. Future studies can add greatly to the growing reproductive database for this species by elucidating the span of the nesting season and by determining larval growth increments during all seasons of the year.

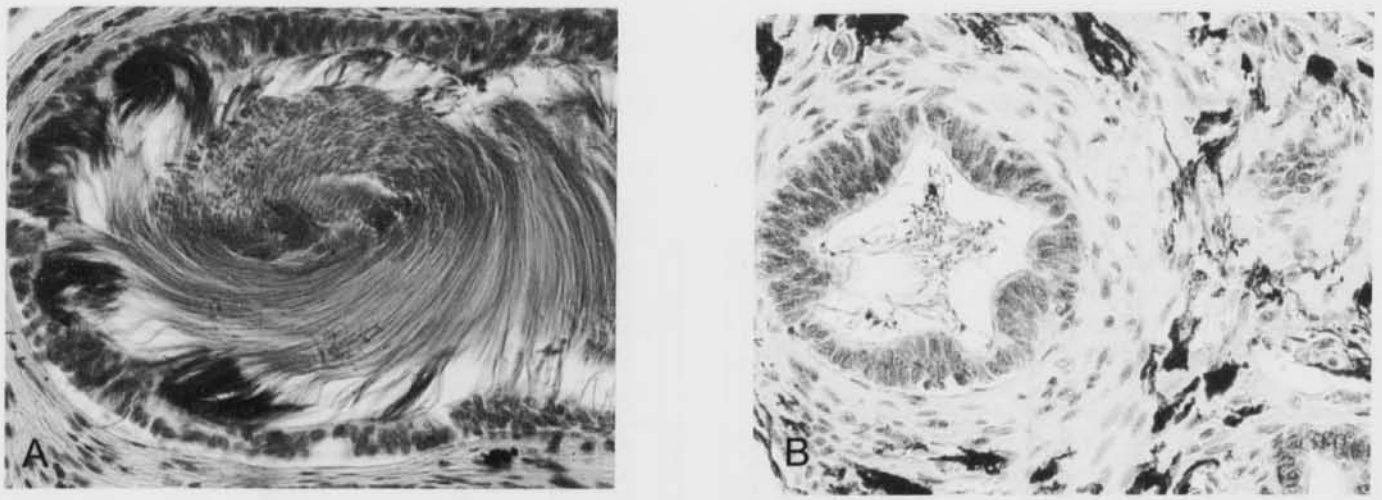

Fig. 1. Spermathecal sacs illustrating an abundance of sperm in a June specimen (A) compared to a trace of sperm in a March specimen (B).

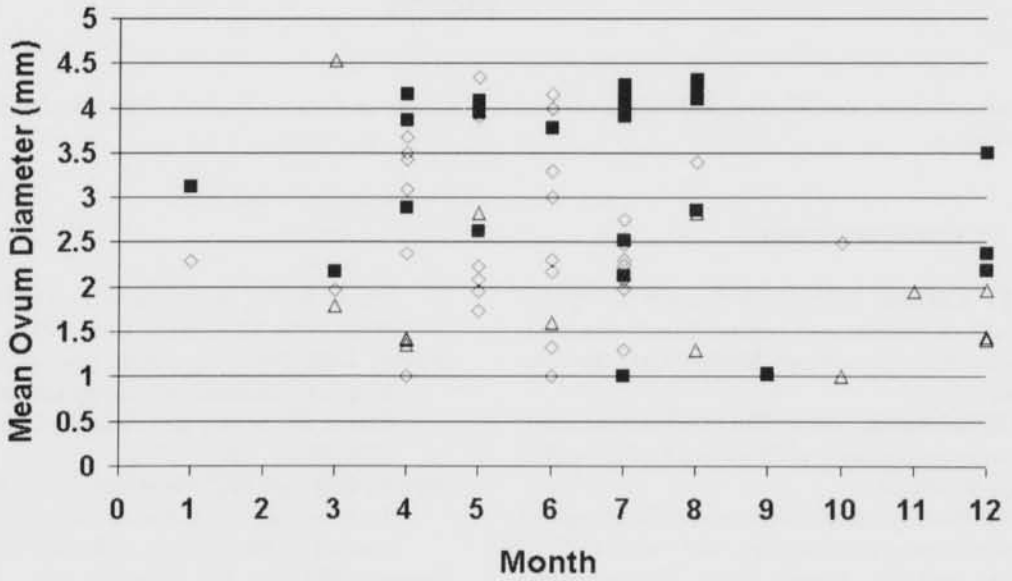

Fig. 2. Presence of sperm (solid squares), trace of sperm (triangles), and absence of sperm (diamonds) within spermatheca vs. mean ovum diameter (mm) in monthly samples of *Desmognathus brimleyorum* from Arkansas. (Some ovarian data were extracted from Trauth et al. [1990]).

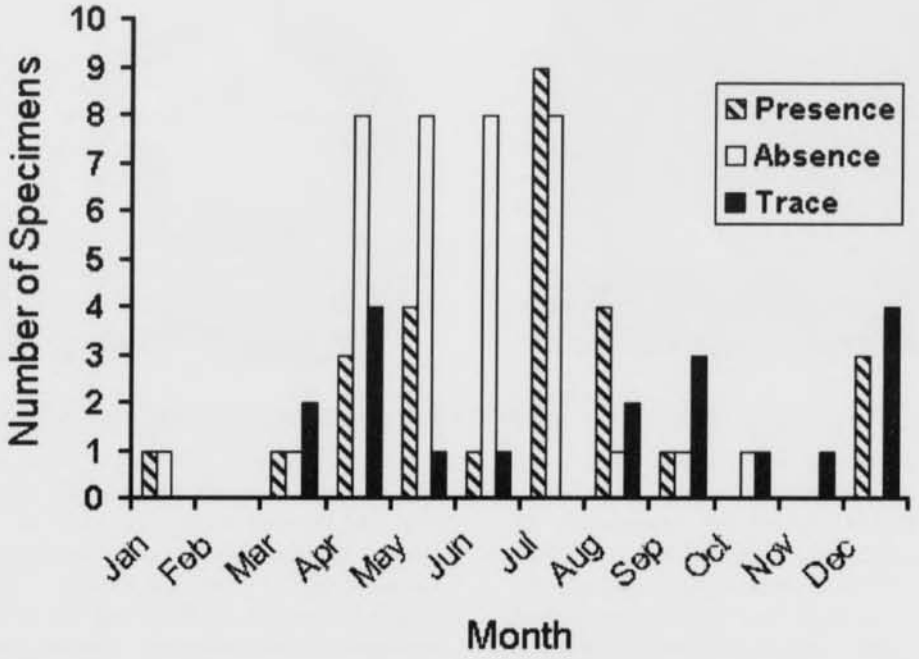

Fig. 3. Monthly spermathecal condition regarding the presence, absence, or a trace of sperm of female *Desmognathus brimleyorum* collected from Arkansas.

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Mapping of ssDNA Nicks Within dsDNA Genomes by Two-dimensional Gel Electrophoresis

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Abstract

DNA molecules within chromosomes undergo constant, dynamic changes yet maintain the integrity of the primary DNA sequence. DNA replication, adjustment of helical density, resolution of catenenes, repair of DNA damage, and homologous recombination each involve breakage and religation of the phosphate backbone of the double helix. Although the analysis of dsDNA breaks is facile, the analysis of ssDNA nicks is not. The principal impediment is that conventional, one-dimensional electrophoresis methods cannot readily detect ssDNA nicks in the context of dsDNA breaks. We therefore developed a two-dimensional (native/denaturing) gel electrophoresis approach to map the positions of ssDNA nicks. Analysis of cohesive ends of lambda phage DNA, UV-nicked DNA molecules, and DNA treated with ssDNA nicking endonuclease N-BbvcIB revealed that the method can detect and map with precision the positions of ssDNA nicks. Titration experiments revealed the ability to detect and quantitate nicked DNA molecules present at a frequency of 1% of total DNA molecules. This method can be used both to scan rapidly through large regions of the genome of interest and to map with high-resolution the location of ssDNA nicks in populations of dsDNA molecules. It is of utility for the analysis of ssDNA nicks involved in a variety of chromosomal processes.

Introduction

Meiosis produces 4 haploid cells from a diploid premeiotic cell (Roeder, 1997; Wahls, 1998; Hassold and Hunt, 2001; Lichten, 2001; Hunt and Hassold, 2002). This is achieved by coupling 1 round of DNA replication with 2 rounds of chromosome segregation. After DNA replication, homologous chromosomes pair and undergo a high rate of homologous recombination. Proper segregation of homologous chromosomes in the first (reductional) division requires that the paired homologs be held together and aligned on the metaphase plate of meiosis I in opposition to spindle tension (Page and Hawley, 2003). In most organisms, a combination of crossover recombination structures (chiasmata) and sister chromatid cohesion distal to chiasmata provide this glue.

Extensive genetic studies have led to the development of models for meiotic recombination (Holliday, 1964; Meselson and Radding, 1975; Szostak et al., 1983). These models each posit that cleavage of DNA strand(s) on one chromatid is followed by recombinational repair using a homologous chromatid as a template. Notably, both ssDNA nicks and dsDNA breaks are implicated. Confirmation of one of these models came from studies of budding yeast mutants (such as rad50S) that accumulate unrepaired, meiotically-induced, dsDNA breaks (Sun et al., 1989; Cao et al., 1990). The distribution and density of the dsDNA breaks in budding yeast correlates well with the distribution and density of recombination (Baudat and Nicolas, 1997; Gerton et al., 2000). However, the types of DNA breaks in vivo have not been adequately characterized for any other organism and artificially-introduced ssDNA nicks can be potent inducers of recombination (Strathern et al., 1991; Lee et al., 2004).

Using 2 independent criteria (one genetic and one biochemical) we obtained evidence for the presence of meiotically-induced, recombinase-dependent, ssDNA nicks in the fission yeast genome (our unpublished observations). This prompted us to develop a powerful, yet simple electrophoresis method for the detection and mapping of ssDNA nicks within dsDNA molecules spanning large distances (up to 1,000 kbp) along chromosomes.

Materials and Methods

Reagents.–Lambda bacteriophage DNA, 1 kbp ladder DNA, and restriction endonucleases were obtained from New England Biolabs. DNA modification enzymes were used according to the instructions of the manufacturer.

Two-dimensional Agarose Gel Electrophoresis.–All electrophoresis was conducted using 10 cm mini gel rigs (Ellard Scientific). DNA samples were fractionated for 200 Vh (40 V for 5 hr) in the first native dimension in 1% agarose gels containing Tris/Acetate/EDTA (TAE) buffer and TAE running buffer (Sambrook et al., 1989). Gels were stained for 15 min in 0.5-µg/ml EtBr and destained 3 times for 15 min each in H_2O prior to being photographed on a UV light box. In cases where exposure to UV light was undesirable the staining and visualization steps were omitted. Gel lanes were excised with a clean razor blade,

nd each lane strip was cast across the top (origin) of a gel ontaining 1% agarose, 50-mM NaCl, 1-mM EDTA. The els were soaked for 60 min in denaturing electrophoresis uffer (30-mM NaOH; 1mM-EDTA) and then subjected to lectrophoresis for 200 Vh (40 V for 5 hr) in denaturing lectrophoresis buffer. Gels were then stained for 15 min in .5-µg/ml EtBr and destained 3 times 15 min each in H_2O rior to being photographed on a long-wave UV light box.

Southern Blotting.–Procedures for Southern blotting were as described (Sambrook et al., 1989; Kon et al., 1997). The lambda phage DNA hybridization probe was prepared using α32P-dCTP (Perkin Elmer) and the RediPrime II prime labeling system (Amersham Biosciences) (Davidson et al., 2004).

Results

Conceptual Development of a Two-dimensional Gel Electrophoresis Method to Detect ssDNA Nicks Within dsDNA Molecules.–Although the analysis and mapping of dsDNA breaks is facile, the analysis and mapping of ssDNA nicks is not. The principal impediment is that conventional, one-dimensional electrophoresis approaches cannot readily detect ssDNA nicks in the context of dsDNA breaks. One must compare in parallel native and denaturing gels using hybridization with strand-specific probes, and such comparisons are restricted to analysis of one restriction fragment in each experiment (lane/hybridization/DNA strand). Because we wish to study the broad distribution of ssDNA nicks in chromosomes, and those nicks may not occur uniformly through the genome, one-dimensional analyses were inadequate.

We developed a 2-dimensional (native/denaturing) agarose gel electrophoresis method to discriminate between ssDNA nicks and dsDNA breaks simultaneously on a single gel for multiple restriction fragments spanning a genomic region. A schematic representation of the method is provided in Fig. 1. For the sake of illustration, we consider the fates of 3 dsDNA molecules–1 that is intact, 1 that contains a ssDNA nick, and 1 that has been bisected into 2 fragments by cleavage of both DNA strands (Fig. 1A). The DNA molecules are fractionated first under native conditions. Because the individual DNA strands of dsDNA are held together by hydrogen-bonding, base-pairing interactions, the dsDNA molecules will migrate proportional (inversely) to their mass, regardless of whether or not ssDNA nicks are present (Fig. 1B). However, if the dsDNA molecules are denatured into their ssDNA strands prior to electrophoresis, then the individual ssDNA strands derived from dsDNA molecules that contain nicks will migrate more rapidly than the ssDNA strands derived from intact dsDNA molecules. Thus, by fractionating a population of DNA molecules on one dimension under native conditions, then in a second dimension under denaturing conditions, it should be possible to resolve nicked dsDNA molecules from intact dsDNA molecules (Fig. 1C).

Detection of ssDNA Nicks Introduced by Ultraviolet Light.–For the initial characterization of the assay, we analyzed the migration pattern of a commercial, 1 kbp ladder typically used as a molecular weight marker for native electrophoresis. The samples were fractionated in the first dimension using conventional agarose gel electrophoresis in native (TAE) gel running buffer. The gels were stained with EtBr and subjected to short-wavelength UV light in order to visualize (and photograph) the DNA bands and to guide the excision of the portion of the gel (lane) containing DNA. The excised lane was cast into a second gel then denatured and subjected to electrophoresis in the second dimension in the presence of denaturant (NaOH). As expected, most of the DNA molecules were distributed along a diagonal arc of the gel (Fig. 2). However, a significant portion of the DNA migrated in a smear below the arc of intact dsDNA molecules (Fig. 2).

Material migrating as a smear in the ssDNA nick area could be due to the presence of pre-existing ssDNA nicks in the dsDNA molecules applied to the gel, or due to the introduction of ssDNA nicks by UV light used to visualize bands after running the first dimension, or due to nicking of DNA molecules by the denaturing conditions used to run the second dimension. To distinguish between these possibilities, we loaded on the gel, just prior to running the second dimension under denaturing conditions, an additional DNA sample that had not been exposed to UV light. DNA molecules in that control sample migrated to discrete (i.e., not smeared) positions in the second dimension (Fig. 2). Thus, ssDNA nicks were not present in the initial DNA molecules and they were not induced by the denaturing conditions. We conclude that the material migrating in the ssDNA nick area was due to exposure of the DNA to UV light. The UV light either introduced ssDNA nicks or some other type of lesion (e.g., pyrimidine dimers) that caused some of the DNA molecules to migrate below the intact dsDNA arc.

Detection of Staggered ssDNA Nicks Held Together by Compatible, Cohesive Ends.–As an independent test of the assay, we examined the fractionation of DNA molecules produced by digestion of lambda bacteriophage DNA with restriction endonuclease HindIII. There are 7 lambda/HindIII DNA fragments (Fig. 3A), two of which (4 kbp and 23 kbp) contain protruding, complementary, ssDNA tails 12 nucleotides (nt) in length (cos sites). These tails can form hydrogen-bonding, base-pairing interactions with one another that are stable enough to be maintained during electrophoresis under native conditions (Fig. 3B). As

a consequence, the abundance of the 4 kbp band in the "expected" position is often reduced–those dsDNA molecules co-migrate with the 23 kbp molecules near the exclusion point of the gel. This provided an excellent way to determine whether two staggered ssDNA nicks (located 12 nt away from each other) could be resolved by two-dimensional (native/denaturing) electrophoresis.

In the first (native) dimension, the 4 kbp band was reduced in abundance, as expected (Fig. 3C). In the second (denaturing) dimension, a band of 4 knt appeared in the ssDNA nick area below the position at which the 23 kbp band migrated in the first dimension (Fig. 3C). We make two conclusions from these data: First, base pairing between the 12 nt long, complementary, ssDNA tails of the cos sites on the 23 kbp and 4 kbp restriction fragments is sufficiently strong to be maintained under native electrophoresis conditions. Second, the two-dimensional approach can melt cohesive ends with 12 nt of base pairing and resolve the products as ssDNA molecules.

Mapping of ssDNA Nicks Within Multiple dsDNA Fragments Treated with Nicking Endonuclease.–Analysis of UV-treated DNA molecules (Fig. 2) and the DNA molecules held together by their cos sites (Fig. 3) indicated that the two-dimensional electrophoresis method can identify ssDNA nicks in dsDNA molecules. However, for the method to be practical it would have to be able to identify and map the positions of ssDNA nicks located long distances from the ends of dsDNA restriction fragments. We therefore compared the migration pattern of lambda/HindIII DNA fragments to the migration pattern of lambda/HindIII DNA fragments that had been treated with nicking restriction endonuclease N.BbvCIB (New England Biolabs). This enzyme has an asymmetric recognition site and nicks specifically one of the two DNA strands at that site. Based upon the DNA sequence of bacteriophage lambda, we would expect N.BbvCIB to introduce nicks at specific locations on three of the seven lambda/HindIII DNA fragments (Fig. 4A).

The lambda/HindIII DNA fragments migrated predominantly along the linear, intact dsDNA arc, except for the 4 kbp fragment, which migrated with the 23 kbp fragment in the first dimension due to its cos site (Fig. 4B). Lambda/HindIII DNA fragments that had been treated with nicking restriction endonuclease N.BbvCIB produced a distinct pattern: All of the fragments that lacked a recognition site for N.BbvCIB (except the 4 kbp fragment with cos) migrated on the linear, intact dsDNA arc (Fig. 4B). All of the fragments that contained a recognition site for N.BbvCIB migrated to their expected positions in the ssDNA nick area below the dsDNA arc (Fig. 4C). We conclude that two-dimensional (native/denaturing) electrophoresis analysis can detect and map the positions of ssDNA nicks located long distances (≥10 knt) away from the ends of dsDNA fragments.

Detection and Quantitation of Low-abundance ssDNA Nicks Within a Large Excess of Intact dsDNA Molecules.– The preceding experiments demonstrated that the positions of several ssDNA nicks can be mapped simultaneously within a population of dsDNA molecules. To determine the resolution of the assay, we mixed varying amounts of nicked and intact DNA fragments, ran the two-dimensional (native/denaturing) gels, and conducted Southern blotting. We were able to detect the nicked DNA molecules present at a frequency of about 1% of total DNA molecules (Fig. 4D and data not shown). By using phosphorimage analysis or similar methods, the amount of hybridization signal from nicked DNA molecules can be standardized relative to the signal intensity from intact dsDNA molecules. Thus, the assay can detect the presence of low abundance ssDNA nicks, map their positions, and determine their frequencies relative to those of intact dsDNA molecules (internal controls).

Discussion

Genetic models suggest that ssDNA nicks may initiate meiotic recombination, but molecular evidence for such lesions is lacking. Formal demonstration that ssDNA nicks initiate recombination will require, among other things, high-resolution mapping to determine whether the distribution of ssDNA nicks is coincident with the local frequencies of recombination. Unfortunately, available methods will not permit large-scale mapping of ssDNA nicks. We therefore developed a simple, inexpensive approach for the identification and mapping of ssDNA nicks. By fractionating a population of dsDNA molecules on one dimension under native conditions, then in a second dimension under denaturing conditions, it is possible to resolve ssDNA molecules from dsDNA molecules. Importantly, this method can detect ssDNA nicks located at least 10 kbp from the ends of dsDNA molecules (Fig. 4). Furthermore, it can identify specifically which dsDNA restriction fragment harbors ssDNA nick(s) (Fig. 4).

The finding that 12 nt of cohesive, ssDNA tails is sufficient to render a dsDNA break invisible in native electrophoresis (Fig. 3) has implications for data and conclusions published previously. The 501 kbp NotI-NotI restriction fragment "J" of fission yeast is located on the left arm of chromosome I (Fan et al., 1989). On this restriction fragment, the density of recombination (per unit distance) for numerous subintervals and the distribution of dsDNA breaks have been mapped (Young et al., 2002). Recombination density varies about 2.5-fold for different subintervals, but dsDNA breaks are reportedly only detectable at two major break sites, mbs1 and mbs2, which are located 105 kbp from one another. Thus, the frequency

nd distribution of dsDNA breaks is inadequate to account or the frequency of recombination. We suggest that ecombinogenic dsDNA breaks in fission yeast may be nore abundant than previously thought, but they may have ohesive ends. In other words, they would be "invisible" to he 1-dimensional, native gel electrophoresis approaches pplied to date. Alternatively, ssDNA nicks may initiate ecombination in fission yeast. Our 2-dimensional assay llows us to test these hypotheses.

Finally, we note that the method allows for elegantly simple scanning of large genomic regions for the presence of invisible dsDNA breaks (i.e., those with cohesive ends) and ssDNA nicks. One can fractionate total genomic DNA that has been digested with a restriction endonuclease and use a series of overlapping cosmids as probes. Each cosmid probe will therefore detect all restriction fragments in a region of approximately 50 kbp, and any ssDNA nicks or invisible dsDNA breaks above the limit of detection will be apparent as signals in the ssDNA nick area of the gels. Similarly, 1 could analyze regions spanning about 1,000 kbp by using hybridization probes derived from bacterial artificial chromosomes (BACs). Any dsDNA restriction fragment that contains a ssDNA nick could then be examined using fragment-specific and strand-specific hybridization probes.

In summary, the method described here can be used both to scan rapidly through large regions of the genome of interest and to map with high-resolution the location of ssDNA nicks in populations of dsDNA molecules. It will likely be of utility for the analysis of ssDNA nicks involved in a variety of chromosomal processes including meiotic recombination.

ACKNOWLEDGMENTS.– Work in our laboratory is supported by funds from the National Institutes of Health (GM62244). We thank our colleagues for their support and helpful suggestions.

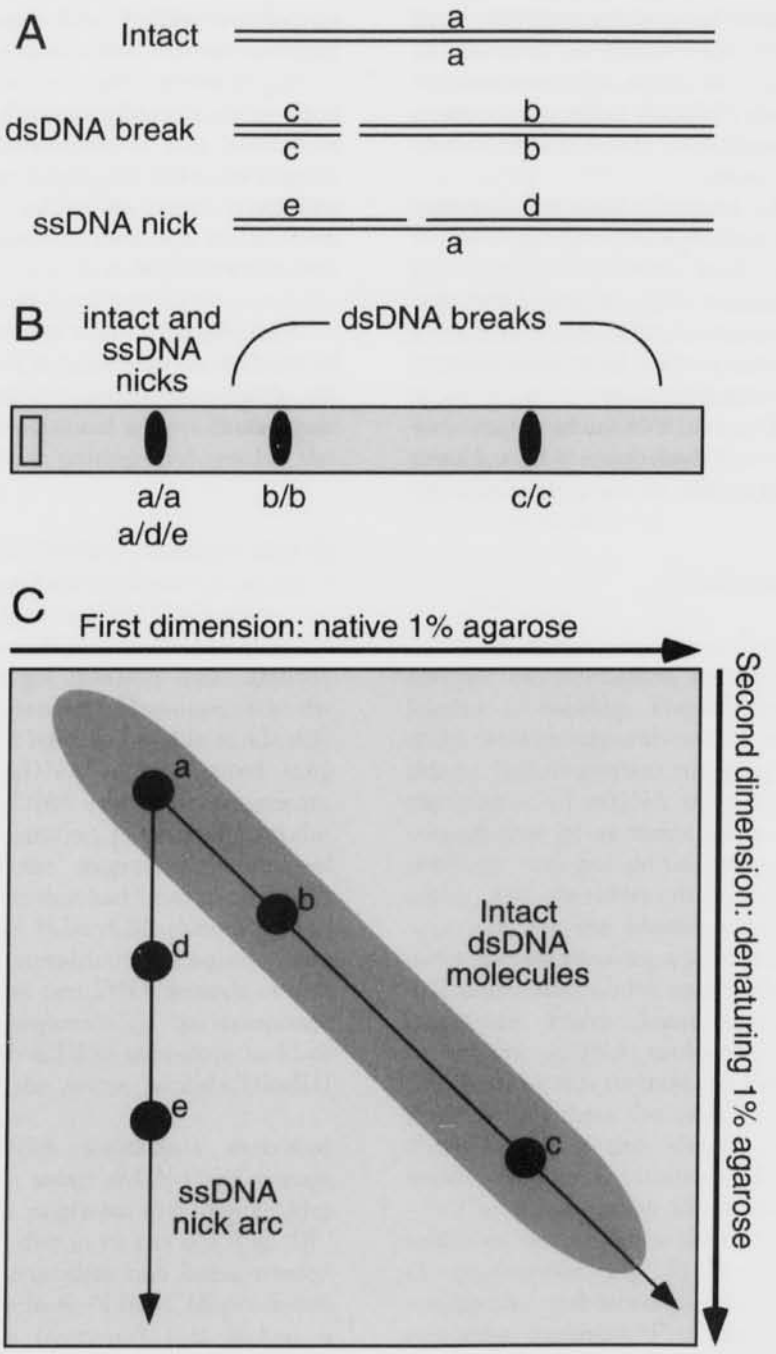

Fig. 1. A 2-dimensional assay for mapping ssDNA nicks in dsDNA molecules. (A) Schematic diagram of intact, broken, and nicked dsDNA molecules with individual strand molecules labeled (a-e). (B) In the first native dimension, nicked and intact molecules are not resolved from one another. (C) In the second denaturing dimension, nicked molecules migrate away from intact molecules.

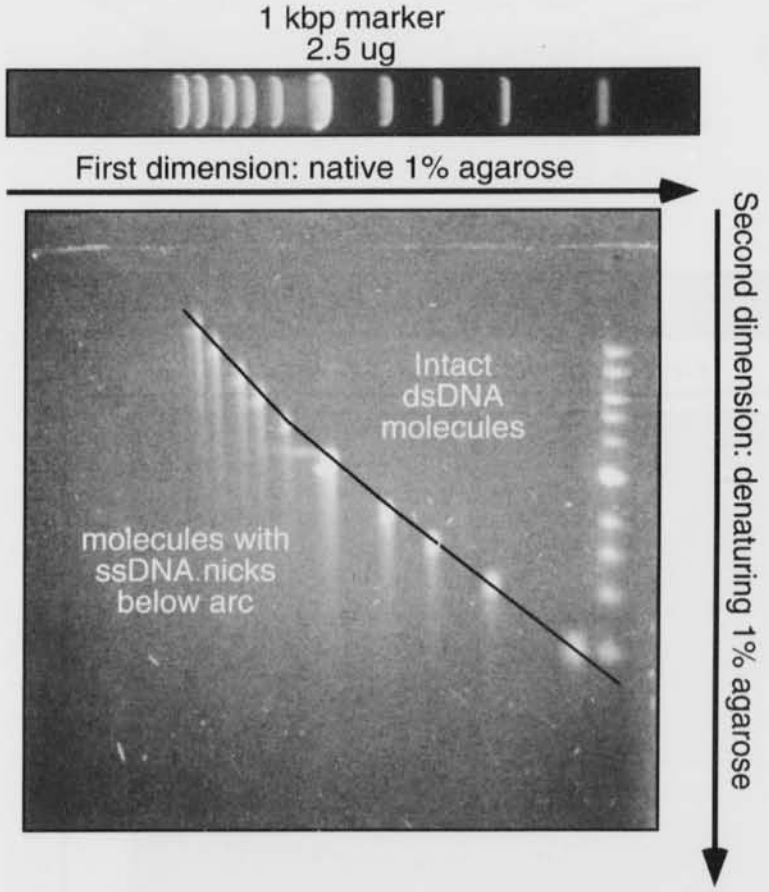

Fig. 2. Effects of UV-induced ssDNA nicks upon migration of dsDNA molecules in 2-dimensional gels. dsDNA molecules were fractionated in the native dimension, then subjected to intense UV light to visualize DNA bands (EtBr fluorescence) and to guide excision of the lane. The lane was cast into a second, denaturing gel, and additional DNA molecules (not exposed to UV) were loaded in a well on the right-hand side of the gel. Following electrophoresis in the second (denaturing) dimension, the gel was stained with EtBr and photographed again. Intact dsDNA molecules migrate to positions along a linear arc (line). Note that the DNA molecules exposed to UV light contain nicks (smearing below dsDNA arc), whereas the marker molecules not exposed to UV light do not contain nicks (rightmost lane).

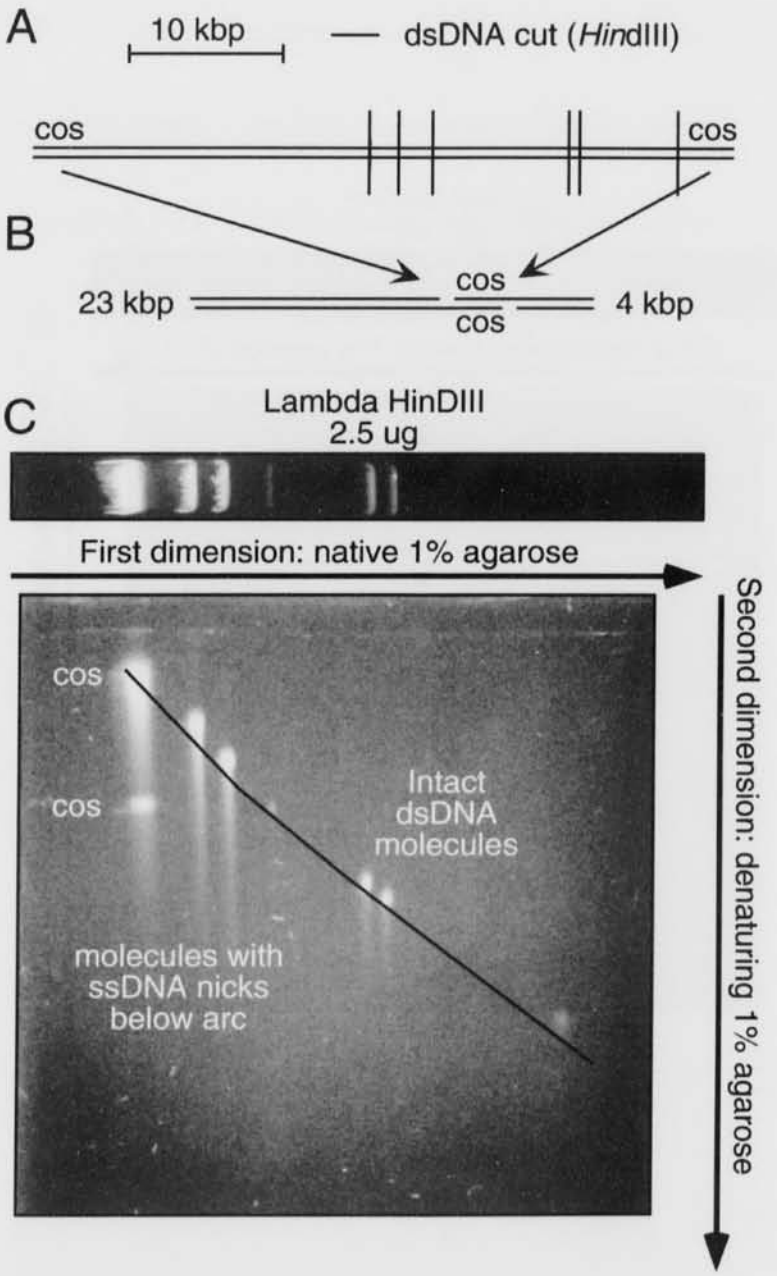

Fig. 3. Effect of compatible, cohesive ends of dsDNA upon the migration of DNA molecules in 2-dimensional gels. (A) Map of bacteriophage lambda chromosome showing positions at which restriction endonuclease HindIII cleaves. (B) The 23 kbp and 4 kbp restriction fragments contain 12 nt long, overhanging, complementary, ssDNA tails from the cos sites on the ends of the lambda chromosome. These can form hydrogen-bond, base-pairing interactions stable enough to hold the two fragments together in native gel electrophoresis. (C) Lambda HindIII fragments were subject to 2-dimensional analysis. Note that the bulk of the 4 kbp restriction fragment migrates with the 23 kbp restriction fragment in the first dimension, but the 4 knt ssDNA fragments migrate away from the 23 knt fragments in the second dimension (cos).

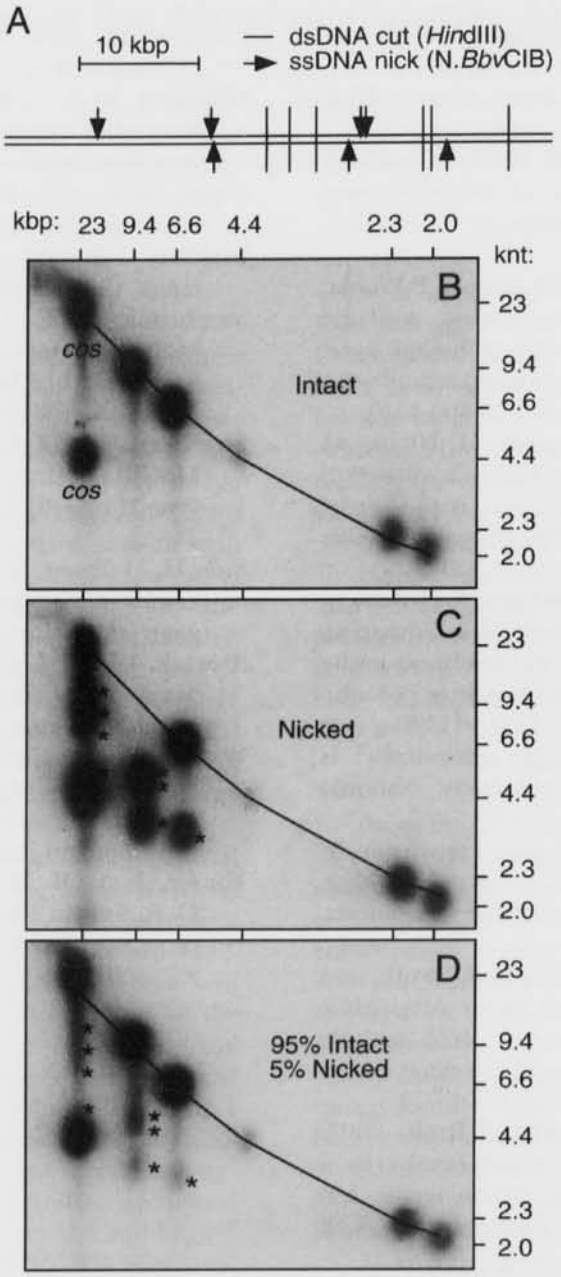

Fig. 4. Mapping the positions and relative abundance of multiple, ssDNA nicks within a population of dsDNA molecules. (A) Map of bacteriophage lambda chromosome showing positions at which restriction endonucleases HindIII and nicking restriction endonuclease N.BbvCIB cleave and nick DNA, respectively. Note that only 3 of 7 HindIII fragments will be nicked. (B-D) Two-dimensional gels of lambda phage genome were subject to Southern blotting using radiolabeled lambda phage DNA as a probe. For the sake of clarity, only a portion (~1/4) of each gel is shown. (B) Analysis of intact dsDNA molecules. With the exception of dsDNA molecules that harbor cohesive termini (cos), all dsDNA molecules migrate on a linear arc. (C) After treatment with nicking endonuclease N.BbvCIB, some of the individual DNA strands become nicked and thus migrate below the intact dsDNA arc (*). One can map the relative positions of ssDNA nicks. (D) DNA molecules as used in panels "B" and "C" were mixed in varying proportions prior to analysis in order to determine the resolution of the assay. The 5% example is shown in order to withstand reproduction, but ssDNA nicks can be detected at a frequency of 1% or less of total DNA molecules (not shown).

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GENERAL NOTES

Presettlement *Pinus taeda* in the Mississippi Valley Alluvial Plain of the Monroe County, Arkansas Area

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Introduction

Loblolly pine (*Pinus taeda*) is the most dominant conifer in the southeastern United States (Baker and Langdon, 1990). However, loblolly pine was conspicuously absent from virtually the entire Mississippi Valley Alluvial Plain during presettlement times. A map (Fig. 1) of the native distribution of loblolly from Baker and Langdon (1990) identifies 2 exceptions to this gap–a narrow strip of land (Macon Ridge) in northeastern Louisiana corresponding to Quaternary-period braided stream terraces left by the ancestral Arkansas River and a small pocket of braided stream terraces from the ancestral Missouri and Mississippi rivers in Arkansas (Saucier, 1974). Unlike Macon Ridge (a noticeably elevated landform), the Arkansas terraces are flat to very gently rolling plains subject to frequent, long-term and large-scale inundation (at least before modern drainage and flood control projects).

Although many hectares of Mississippi Valley Alluvial Plain have been planted in conifers during the last century, the pine found in the Monroe County, Arkansas area is of natural and prehistoric origin. Between 1815 and 1842, General Land Office surveyors traversed this area and reported abundant pine. Soon afterward, portions of the study area became the property of the American Land Company, which offered them for sale (American Land Company, 1844). A quarter-section (64.8 ha) in Township 1 North, Range 1 West (T1N R1W) was described as "...all post oak and pine glade, wet and boggy. Worth nothing" and a different parcel in T2S R1W was similarly recounted as a "...poor post oak and pine glade, very wet and boggy" (American Land Company, 1844, pp. 11, 16). Decades later, botanist Roland Harper journeyed through eastern Arkansas and reported a "good deal" of loblolly pine in Monroe County east of Brinkley; he further noted that "...this was the only place where I saw this pine between Little Rock and Memphis" (Harper, 1914, pp. 43-44). As the 20th Century progressed, this region was drained and/or cleared for lumber, agriculture, pasture, and home sites, but many of the marginal areas reverted to forest cover after other land use practices failed (Harrison, 1954).

Indeed, modern ecological investigation shows that the pine-dominated forests of the area arose from an unusual suite of environmental conditions driven largely by soils and disturbance regimes (Arkansas Natural Heritage Commission, 2002). These site conditions also support several endangered plant and animal species, which in turn has led to growing conservation efforts in the area. For example, the Arkansas Natural Heritage Commission recently purchased a number of small tracts in Monroe County in part to protect some of the second-growth remnants of the pine-dominated portions of this landscape. To improve our understanding of the complex interactions that produced this unique area, this paper describes the presettlement vegetation patterns reported in the public land survey records with additional materials from other historical descriptions.

Materials and Methods

From the GLO survey notes (Daniels, 2000), I analyzed all or part of 14 townships in east-central Arkansas for the abundance and distribution of tree species, ecological communities, and any other natural features. The study area encompasses most of eastern and central Monroe County and portions of Lee, Phillips, and St. Francis counties, an area of approximately 130,000 hectares (Fig. 2). Throughout the region, soils tend to be poorly or somewhat poorly drained and wet throughout much of the winter and spring. Locally, soils of the Foley-Calhoun-Bonn and Lafe-Bonn complex are of particular interest, as their high levels of sodium and magnesium help to structure the complex mosaic of unique plant communities (Arkansas Natural Heritage Commission, 2002).

Most information for this paper is derived from the individual trees used by the surveyors to witness their efforts. GLO surveyors recorded the names, diameters, distance, and bearings of 2 to 4 witness trees at each section, quarter-section, and meander corner on an approximately 1.61 km by 1.61 km lattice throughout the study region. In addition, surveyors also recorded the name, diameter, and distance from corner of 1 to 3 line trees for each 1.61 km of section line established.

Species identifications in the GLO and other forms of historical plant records are often only approximate at best, and often the common (surveyor-given) name is vague and

may represent multiple species, making identification sometimes no more precise than genus (MacRoberts et al., 1997; Bragg, 2002). For continuity, taxonomic identifications provided by the surveyors will primarily follow the interpretations of Bragg (2002) with modifications based on regional species occurrences.

Results and Discussion

Evidence of Human Influence.– In the study area, very few indications of Euroamerican settlement were given in the GLO survey notes, suggesting that little environmental modification had occurred immediately before and during the surveying period of 1815 to 1842. Some of the later resurveys (conducted in the 1840s) mention roads or trails, which is not surprising given that Monroe County was along one of the major routes between Memphis and Little Rock. A handful of pioneers and their clearings (sometimes called "improvements") were also reported in the 1840s resurveys, indicating that permanent settlement and subsistence agriculture had begun. There is virtually no mention of other forms of land clearing or disturbance, which suggests that the vegetation patterns reported by the GLO surveyors should be consistent with the virgin forests of the region.

Numerous openings identified as prairies were reported throughout the study area. Undoubtedly, many of these represent grasslands of natural origin, maintained by extreme site conditions unfavorable for tree growth. Other, more transitory grasslands may have been kept open by frequent fires, perhaps set by Native Americans and Euroamerican hunters. There is no direct evidence that any of these openings were the abandoned remnants of Native American agricultural practices. However, there are locations from the nearby Crowley's Ridge area where Indian fields were still being specifically identified by the GLO surveyors in the 1810s and 1820s.

Taxonomic Abundance.–The GLO records produced 3,458 trees from about 40 taxa (Table 1). Individuals labeled white oak (probably *Quercus alba* and/or *Quercus michauxii*) comprised 18.05% of witness trees, followed by black oak (various *Quercus* spp.; 16.14%), hickory (*Carya* spp.; 10.47%), elm (*Ulmus* spp.; 6.30%), and pine (probably loblolly; 5.67%). No other single taxon contributed more than 5% of the total number of witness trees, and 4 were represented by a lone tree. Because of biases in how they were chosen, the frequencies in Table 1 do not directly translate to species dominance. However, witness tree counts broadly reflect the patterns of taxonomic abundance in the Monroe County study area during the historical surveying period. In other words, infrequently reported species were probably not common on the landscape (or were too small on average to be regularly used as witness trees).

Black oak was not more precisely defined than *Quercus* spp. in Table 1 because of known issues with the GLO surveyors' identification of the taxon compared to modern interpretations. Contemporarily, black oak is *Quercus velutina*, but this species is most prominent in drier, rockier hills and slopes in parts of northern Arkansas and the central United States and is increasingly uncommon as one heads southward or onto the major floodplains (Sargent, 1947). Bragg (2003) also reported unusually high levels of black oak in the GLO survey records from Ashley County, Arkansas, suggesting that a wide range of oaks were probably lumped into the black oak group. In addition to some *Quercus velutina*, other probable taxa placed into this group by the surveyors may include southern red oak (*Quercus falcata*), cherrybark oak (*Quercus pagoda*), Shumard oak (*Quercus shumardii*), Nuttall oak (*Quercus nuttallii*), and perhaps even water oak (*Quercus nigra*).

Pine was not identified to species in the GLO work conducted in Arkansas, although only two distinct species (*Pinus taeda* and *Pinus echinata*) are native to the state. The best available evidence suggests that the pines the surveyors encountered were loblolly. For example, Harper (1914) reported only loblolly pine in his travels through this part of Arkansas. Shortleaf pine, though common in the uplands of presettlement Arkansas (including the nearby Crowley's Ridge), fares much more poorly than loblolly on wet sites and is very rarely found in bottomlands. However, the presence of loblolly pine in this portion of Arkansas is also highly unusual (Grimmett, 1989).

Witness Tree Size.–For a region with extensive poor soils, a surprising number of very large trees was found (Table 2). As an example, the largest witness tree was a 203 cm diameter white oak found in T1N R1W by one of the first surveyors to traverse the area. Oaks and baldcypress (*Taxodium distichum*) dominated the big witness trees, with only a few other taxa exceeding 100 cm in diameter. Baldcypress witness trees were particularly large, averaging 86 cm in diameter with a maximum of 183 cm (Table 1). Baldcypress also constituted 35% of the largest trees recorded in the GLO notes of the study area (Table 2). However, given the commercial interest in baldcypress during the early 19th Century, it is not surprising that large cypress trees were noted (Bragg, 2003). Only 1 pine exceeded 100 cm in diameter–most were less than 50 cm (Fig. 3). Unlike some of the hardwood species that showed an affinity for better quality locations, pine was most prevalent in the poor to moderate sites. Therefore, it is noteworthy that a species like loblolly that usually reaches a very large size on low terraces only infrequently exceeded 50 cm in diameter (Table 1, Fig. 3).

On average, most (74.7%) witness trees ranged from 12 to 51 cm in diameter (Table 1, Fig. 3). Some taxa provided very small diameter witness trees–down to 3 cm (elm) and 5 cm (white oak), although most exceeded 10 cm. These

minimums do not reflect the true distribution of small diameter stems on the landscape because surveyors avoided diminutive individuals. This bias by omission arose in part because surveyors needed to scribe a lot of information on the boles (a difficult task on a small tree), and small diameter trees were also considered more prone to mortality, given the degree of bark injury inflicted upon them. Small tree bias also means that species that rarely exceed 12 cm in maximum diameter are almost never used as witness corners, even though they may be fairly common across the landscape.

Other Surveyor Observations on Pine.– The GLO notes of the study region recorded numerous areas as "pine woods" or "pine land", suggesting that loblolly was the dominant overstory species in some stands. More often, pine was reported as mixed with oak and other hardwoods, sometimes with prominence given to pine (i.e., pine was listed first) or as a subordinate (e.g., "oak and pine"). In these areas, it is not unusual to see "huckleberry" (*Vaccinium* spp.), "briers" (possibly *Rubus* or *Smilax* spp.), and "swamp spice" (probably *Lindera benzoin*) listed by the surveyors as understory associates.

Loblolly plantations in the Mississippi Valley Alluvial Plain can be successful if they are not flooded too frequently or for too long of a duration. It is rare to see much natural regeneration under these plantations, although loblolly and other conifers have shown some ability to naturalize in the region under certain conditions. Nevertheless, the surveyors reported abundant natural loblolly pine regeneration in portions of Monroe County, indicating the potential for long-term persistence of loblolly pine in this seasonally flooded alluvial landscape. For instance, in 1820 deputy surveyor Nicholas Rightor identified the undergrowth in T2S R1W as "small Pine [and] Huckleberry". Another surveyor, John Garretson, frequently reported "oak and pine bushes" in the portions of T4N R2W where pine was a prominent species. Presumably, "bushes" referred to thickety patches of oak and pine regeneration, possibly stunted by long-term overstory suppression, repeated fire injury, or severe soil conditions.

Conclusions

In presettlement times (before 1850), this portion of Monroe County was a complex mosaic of hardwood swamps and flatwoods, scattered prairies and other openings, and occasional conifer-dominated stands. In a landscape covered with bottomland oaks, gums, hickories, other hardwoods, and baldcypress swamps, loblolly pine-dominated communities are unexpected elements of structural, functional, and compositional diversity. Thus, modern-day analogs of these loblolly pine forests are not artifacts of recent human influence, but rather self-replacing components of the ecosystem.

ACKNOWLEDGMENTS.– I would like to thank those who assisted with the analysis and preparation of the materials for this paper, including Jeff Barton, Wes Fletcher, Tom Foti, Kevin Hicks, Mike Shelton, Eric Sundell, Bruce Walsh, Mickie Warwick, and Theo Witsell. Two anonymous reviewers helped improve the quality of this paper.

Table 1. Probable species and diameter attributes of the witness trees collected from the Monroe County, Arkansas, study area.

GLO surveyor name	Probable taxa	Number of witness trees	% total witness trees	Min. diam. (cm)	Ave. diam. (cm)	Standard deviation (cm)	Max. diam. (cm)
White (W.) oak	*Quercus alba, Q. michauxii*	624	18.05	5	48	21.3	203
Black (B.) oak	*Quercus* spp.	558	16.14	10	46	21.3	127
Hickory	*Carya* spp.	362	10.47	8	33	10.9	76
Elm	*Ulmus* spp.	218	6.30	3	28	13.5	122
Pine	*Pinus taeda*	196	5.67	8	43	19.3	127
Sweetgum	*Liquidambar styraciflua*	162	4.68	8	46	21.3	122
Post oak	*Quercus stellata, Q. michauxii*	152	4.40	8	48	19.8	102
Pin oak	*Quercus nigra, Q. phellos, Q. nuttallii*	151	4.37	8	36	19.6	127

Table 1. Continued.

GLO surveyor name	Probable taxa	Number of witness trees	% total witness trees	Min. diam. (cm)	Ave. diam. (cm)	Standard deviation (cm)	Max. diam. (cm)
Gum	*Nyssa* spp., *Liquidambar*	150	4.34	15	48	21.6	122
Overcup oak	*Quercus lyrata*	116	3.35	10	43	20.8	102
Red (R.) oak	*Quercus falcata, Q. pagoda*	93	2.69	8	46	21.8	122
Tupelo (white) gum	*Nyssa aquatica*	91	2.63	15	43	18.0	102
Ash	*Fraxinus* spp.	75	2.17	10	36	18.8	97
Maple	*Acer* spp.	75	2.17	10	30	11.2	61
Dogwood	*Cornus florida*	67	1.94	8	18	5.3	30
Black gum	*Nyssa sylvatica*	58	1.68	8	36	13.5	76
Willow oak	*Quercus phellos*	45	1.30	15	41	15.2	76
Cypress	*Taxodium distichum*	36	1.04	15	86	45.2	183
Swamp white oak	*Quercus michauxii*	32	0.93	18	48	20.8	102
Pecan	*Carya illinoensis*	29	0.84	10	33	17.3	91
Persimmon	*Diospyros virginiana*	26	0.75	8	25	9.4	51
Hackberry	*Celtis laevigata*	24	0.69	10	28	13.7	76
P. oak	*Q. stellata, Q. nigra, Q. phellos Q. michauxii, Q. nuttallii*	21	0.61	10	43	13.2	76
Sassafras	*Sassafras albidum*	21	0.61	10	25	9.7	51
Horn beme	*Carpinus caroliniana*	8	0.23	8	15	4.1	20
Locust	*Gleditsia* spp., *Robinia pseudoacacia*	8	0.23	13	28	13.2	51
Mulberry	*Morus rubra*	7	0.20	10	23	7.6	36
Oak	*Quercus* spp.	7	0.20	25	46	9.1	51
Privey (white or red)	*Forestiera acuminata*	7	0.20	10	15	3.0	20
Cottonwood	*Populus deltoides, Populus heterophylla*	6	0.17	25	38	13.0	53
Honey locust	*Gleditsia triacanthos, Gleditsia aquatica*	6	0.17	20	33	21.3	76

Table 1. Continued.

GLO surveyor name	Probable taxa	Number of witness trees	% total witness trees	Min. diam. (cm)	Ave. diam. (cm)	Standard deviation (cm)	Max. diam. (cm)
Swamp oak	*Quercus michauxii, Q. nuttallii*	6	0.17	51	61	12.4	76
Ironwood	*Ostrya virginiana*	5	0.14	13	18	3.8	23
Boxelder (maple ash)	*Acer negundo*	4	0.12	23	30	6.4	38
Water oak	*Quercus nigra*	3	0.09	30	38	11.7	51
Willow	*Salix nigra*	3	0.09	15	28	10.7	36
Prickle sumac	*Aralia spinosa, Zanthoxylum clava-herculis*	2	0.06	10	13	3.6	15
Haw	*Crategus* sp.	1	0.03	13	13	–	13
Holly	*Ilex opaca*	1	0.03	36	36	–	36
Black walnut	*Juglans nigra*	1	0.03	56	56	–	56
Red bud	*Cercis canadensis*	1	0.03	15	15	–	15
						TOTAL:	**3,458**

Table 2. Trees greater than 102 cm in diameter by surveyor names for the Monroe County, Arkansas, study area.

Surveyor name	Diameter (cm)	Township & Range	Year
White oak	203	1N 1W	1815
Cypress	183	4N 2W	1842
Cypress	183	4N 2W	1842
Cypress	152	4N 2W	1842
Cypress	152	4N 2W	1842
White oak	140	2S 1E	1820
Cypress	137	4N 2W	1842
Black oak	127	1S 2W	1819
Black oak	127	2S 1E	1820
Black oak	127	2S 1W	1820
Black oak	127	3N 1W	1816
Cypress	127	2S 1E	1820
Cypress	127	3S 1W	1825

Table 2. Continued.

Surveyor name	Diameter (cm)	Township & Range	Year
Cypress	127	3S 1W	1825
Cypress	127	3S 1W	1825
Cypress	127	3S 1W	1825
Pin oak	127	3S 1W	1825
Pine	127	1S 1W	1815
Black oak	122	2S 1E	1820
Cypress	122	1S 2W	1819
Elm	122	3S 1W	1825
Gum	122	1S 1E	1815
Red oak	122	1S 1E	1815
Sweetgum	122	3S 1W	1825
White oak	122	1S 1E	1815
White oak	122	1S 2W	1819
White oak	122	4N 2W	1842
Black oak	114	1N 1W	1815
White oak	114	1S 1W	1815
Black oak	112	3N 2W	1842
Sweetgum	112	4N 2W	1842

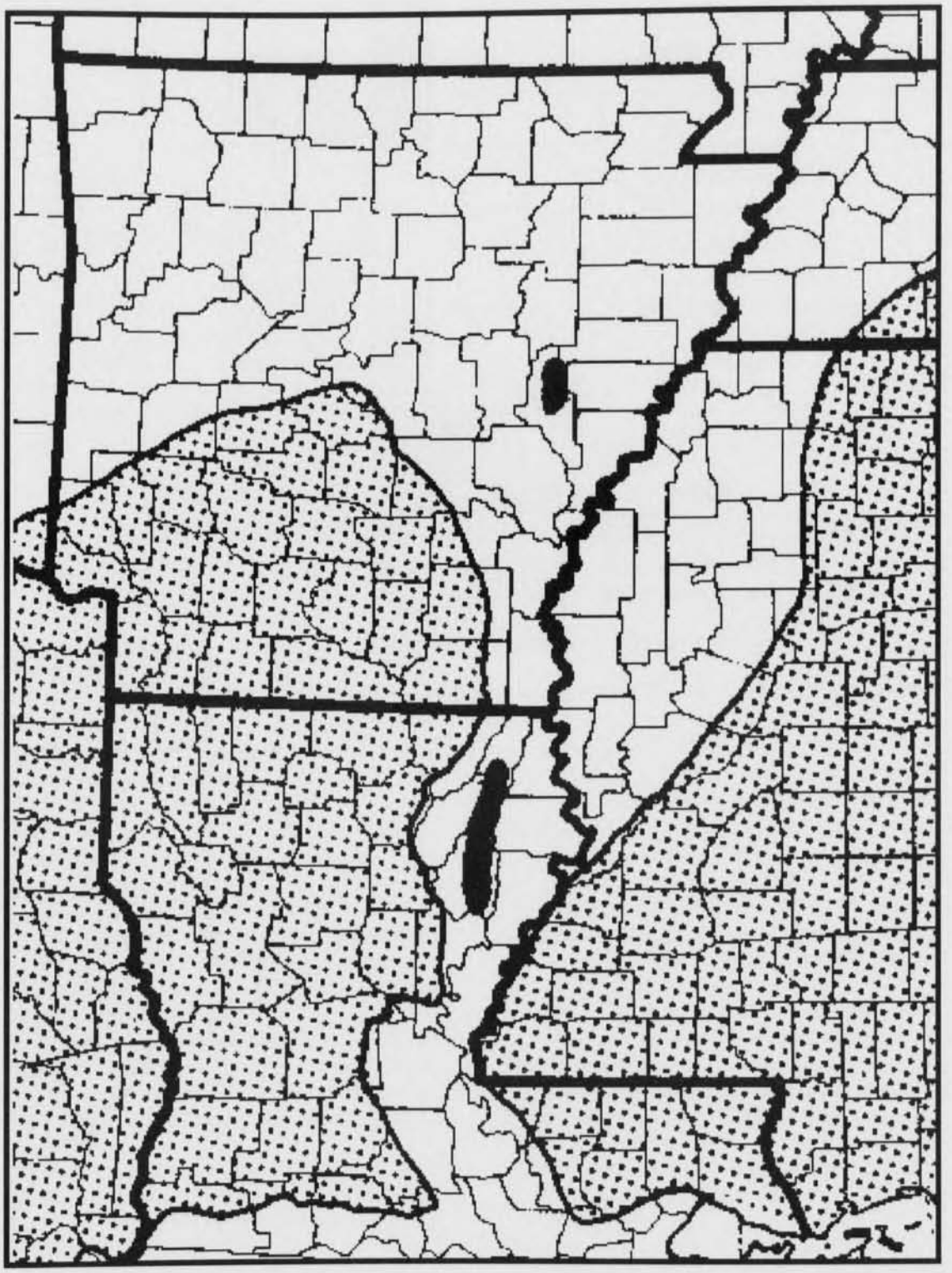

Fig. 1. Natural distribution of loblolly pine in the lower Mississippi River Valley indicated by the stippled pattern. The 2 dark shaded areas are the Monroe County, Arkansas, study area (north) and Macon Ridge in Louisiana (south). Figure adapted from Baker and Langdon (1990).

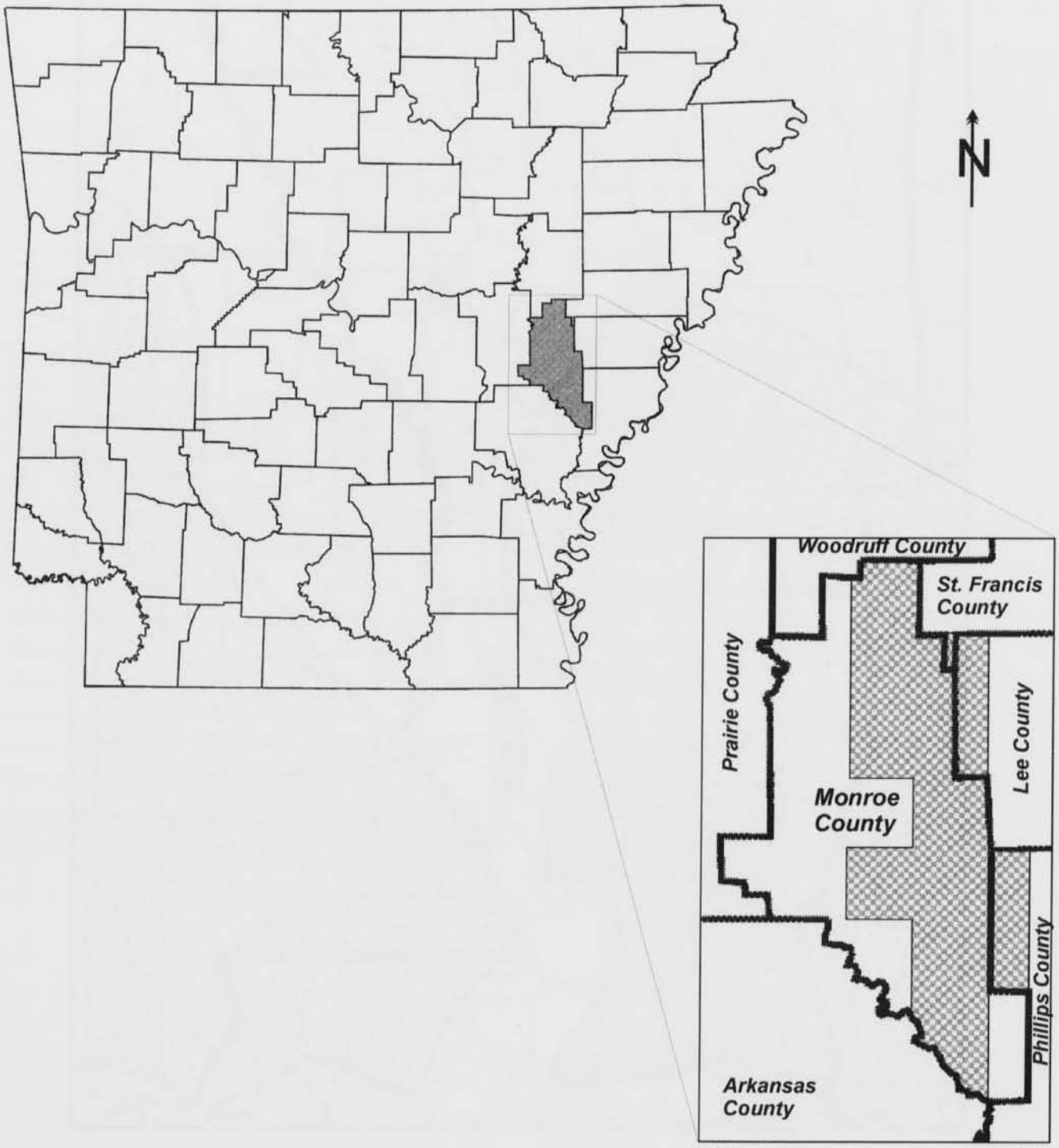

Fig. 2. Study area map. Shaded area in inset map approximates the townships selected for this analysis.

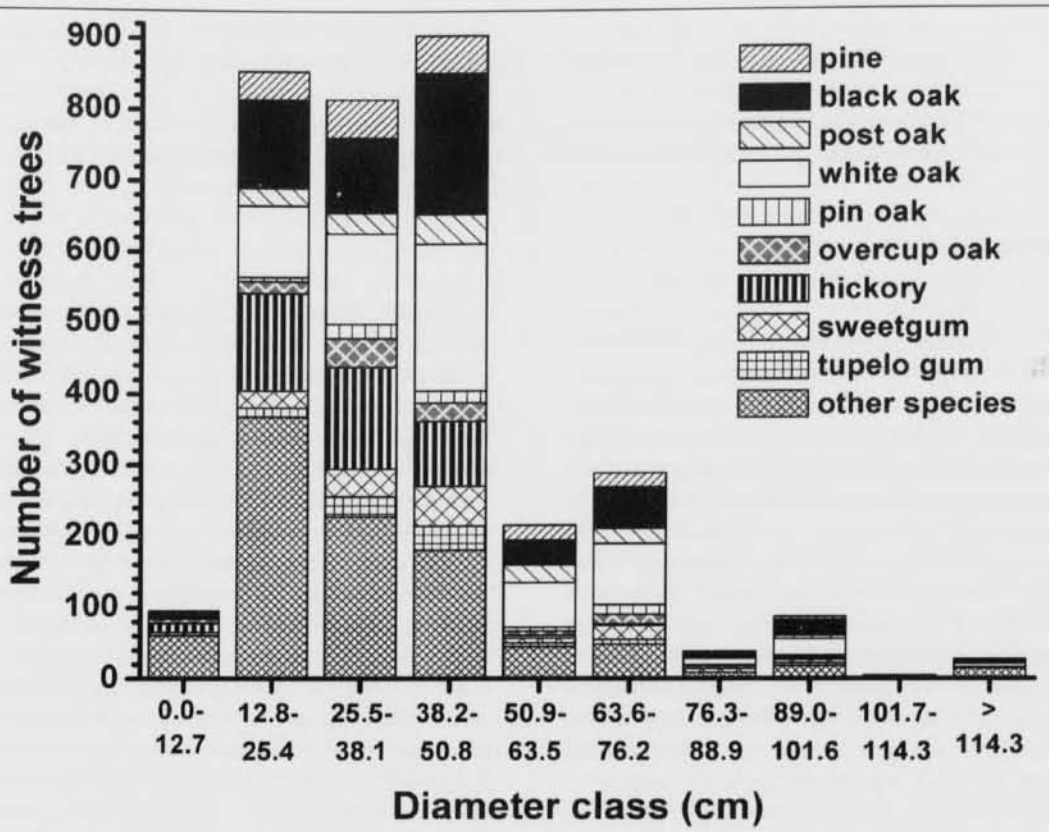

Fig. 3. Diameter distribution by major species or species group as identified by GLO surveyors in the Monroe County, Arkansas, area.

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The Effect of Ionizing Radiation on *Trichomonas vaginalis*: Increase in Red Pigmented Intracellular Bodies Associated with Irradiated Cells

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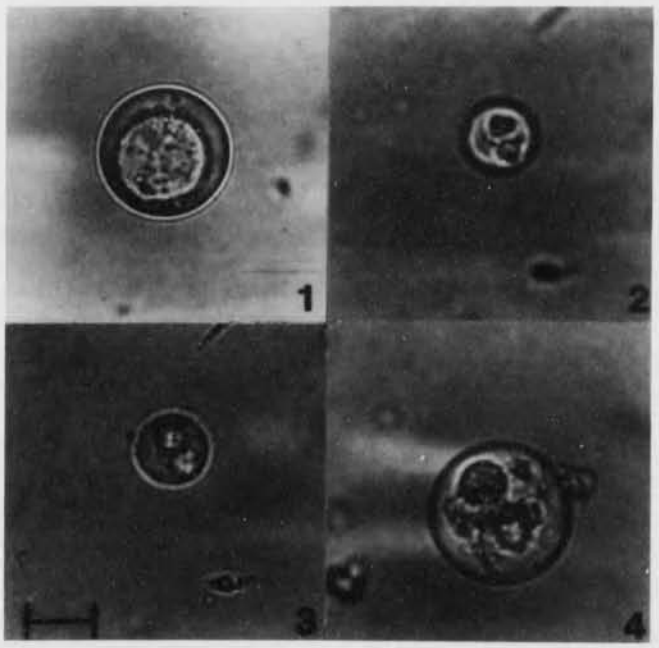
In the course of studies on the effect of ionizing radiation on the viability of the human urogenital parasite, *Trichomonas vaginalis*, red pigmented bodies began to appear in noticeable numbers in the organism's cytoplasm after exposure to high dosages of radiation. These red-pigmented bodies, ranging in appearance from dense granules to vacuolar-like structures, were easily visualized by light microscopy. Further, the number of cells containing these bodies seemed to increase with continued radiation. Review of the literature did not reveal a description of any trichomonad intracellular organelles that were as large as or as densely pigmented as the bodies we found associated with the exposure to ionizing radiation. The increase in the number of such bodies in the cell population in the present study corresponded with the reproductive death associated with an increase in radiation dosage found by Daly et al. (1991) and Hostetler et al. (2004).

The present investigation reports on the general appearance of these pigmented intracellular granules and on a dose-response study of the increase in these bodies by radiation. All methods and materials for culture maintenance, irradiation, radiation source, cell counts, and viability determinations can be found in Daly et al. (1981) and Hostetler et al. (2004). The presence and number of intracellular pigmented bodies were determined by examining trichomonads using wet slide preparations with bright-field light microscopy using oil (970X). For each radiation dose 200-250 cells were examined at a timed interval and compared with the control (unirradiated) cells kept in the dark at ambient temperature.

Initial studies of colony formation determined that a sterilizing dose of gamma radiation for 106 cells of Trichomonas vaginalis occurred at 1600 to 1800 Gy. It was noted that although the trichomonads were no longer viable, as determined by colony counts, they were still intact and highly motile. By contrast, trichomonads kept in the dark became round and were markedly less motile. The viability of this rounded-up control population, however, was close to 100%, and this did not begin to decrease until after 40 hr in the dark at ambient temperature. Trichomonads that were irradiated with more than 1600 Gy possessed a large number of red-pigmented intracellular bodies. These ranged from small (approximately 1 μm: or larger) dark-red objects to large vacuole-like structures that were pink. The larger structures, at times, occupied a major portion of the intracellular space and could be paired or exist as a single body (Fig. 1). Cells with the dense red granules tended to be much smaller than those cells with the larger pink structures. Both of these types of bodies also could be found, although in fewer numbers, in control populations that had not been irradiated. The appearance of these bodies was followed throughout the course of four experiments that extended the radiation dosage to 6100 Gy for the experimental population and 61 hr of testing for the control population in the dark (Fig. 2). The irradiated trichomonads remain actively motile up to 5100 Gy, and the total cell numbers did not begin to decrease until 4600 Gy. After 5100 Gy, no intact cells were found in the irradiated medium. The production of the pigmented bodies in both the irradiated and control cells appeared exponential. The increase in inclusions in the irradiated group coincided with 100% inability to reproduce. The increase in inclusions in the control group also was associated with a diminished reproductive potential as well as lack of motility. The population viability of unirradiated cells after 40 hrs in the dark was reduced to 50-70%. Interestingly, the increase of pigmented intracellular bodies in irradiated cells could be realized only by using cells that were taken from the exponential phase of growth. Cultures that were well into the stationary phase of growth would not produce cell populations that, when irradiated, would contain more inclusions than the unirradiated controls.

The occurrence of the large, pink, vacuole-like, interacellular inclusions in *Trichomonas vaginalis* that had been exposed to high doses of gamma irradiation was an unexpected finding. Retrospectively, these bodies were also found in the unirradiated populations, but with much less frequency. The large inclusions appeared, in terms of sequence and absence, to have formed from smaller, dense, red-pigmented bodies that were easily seen at X470. The vacuole-like pink bodies were larger and more prominent than the largest spherical organelles previously described in trichomonads. We have not found any description in the literature of intracellular bodies produced in cells by chemical or physical agents that correspond to the large pink structures associated with ionizing radiation in this study. In studies involving the effect of certain drugs on *T. vaginalis*, excessive cytoplasmic vacuolation may be found, but these vacuoles do not contain noticeable pigment nor

are they as large as the inclusions described herein (Buchner and Rummel, 1975; Carosi et al., 1977; Ings and Constable, 1975). The smaller, dense, red inclusions may be the same as the presumptive "apoptotic bodies" found within *T. vaginalis* that had been exposed to apoptotic-stimulating drugs (Chose et al., 2002). These bodies were condensed and aggregated nuclear material. Those findings imply that the bodies formed by radiation exposure are also of nuclear origin. The differences that we have found concerning trichomonad bodies of both types are presence of the red pigment and the size of the large bodies. Chose et al. (2002) were reluctant to define their bodies as completely apoptotic in origin because apoptosis originates in the mitochondria, and trichomonads are amitochondrite organisms. Chose and colleagues found no biochemical evidence to substantiate apoptosis as found in other cells. They have suggested that the process reported by them may be a different form of apoptosis not yet elucidated or even paraptosis, a non-apoptotic variant of programmed cell death found in multicellular organisms. Trichomonads are anaerobes and produce hydrogen in organelles called hydrogenosomes (hydrogenosomes are found in a variety of anaerobic free-living and parasitic protozoa–Embley et al., 2003). Iron compounds, such as ferredoxin, are necessary for the hydrogen metabolism of trichomonads (Marczak et al., 1983; Peterson and Alderete, 1984), and it can be posited that the red pigment is a result of a combining of this iron compound with nuclear material. Radiation is also known to affect membrane permeability and cause loss of membrane function, which might allow ferredoxin to enter the nucleus from the cytoplasm. The effect of radiation on the permeability of the outer cell membrane might also explain the larger size of trichomonads with the larger pink bodies. The hydrogenosomes themselves might also be considered as a possible source of the red pigmented bodies because of their requirement for ferredoxin in their metabolic activity. However, given their size and number, it would require coalescence of these bodies and formation of a maximum of 2 such units. Further studies with cytochemical analysis may resolve the genesis of radiation-induced pigmented organelles. We are reluctant to consider at this time that the production of the pigmented bodies is necessarily due to a process of apoptosis, natural or accelerated. Radiation damage may mimic apotic events and result in cell death, but it may not be the same as programmed cell death. As a temporary means of identification until further data is available, we are suggesting the term "thanatic bodies" since they are associated with the inability of *T. vaginalis* to reproduce in culture.



Figs. 1-4. Inclusion bodies found in *Trichomonas vaginalis* irradiated with greater than 1600 Gy of gamma radiation. The trichomonads were actively motile but incapable of reproducing. Large, vacuolar, pink body inside cell (1). Small, dense, reddish bodies (2 & 3). Speculative intermediate phase between small red bodies and large pink ones (4) Other trichomonad organelles are not visible because the plane of focus was on the pigmented bodies. The scale bar represents 10 µm.

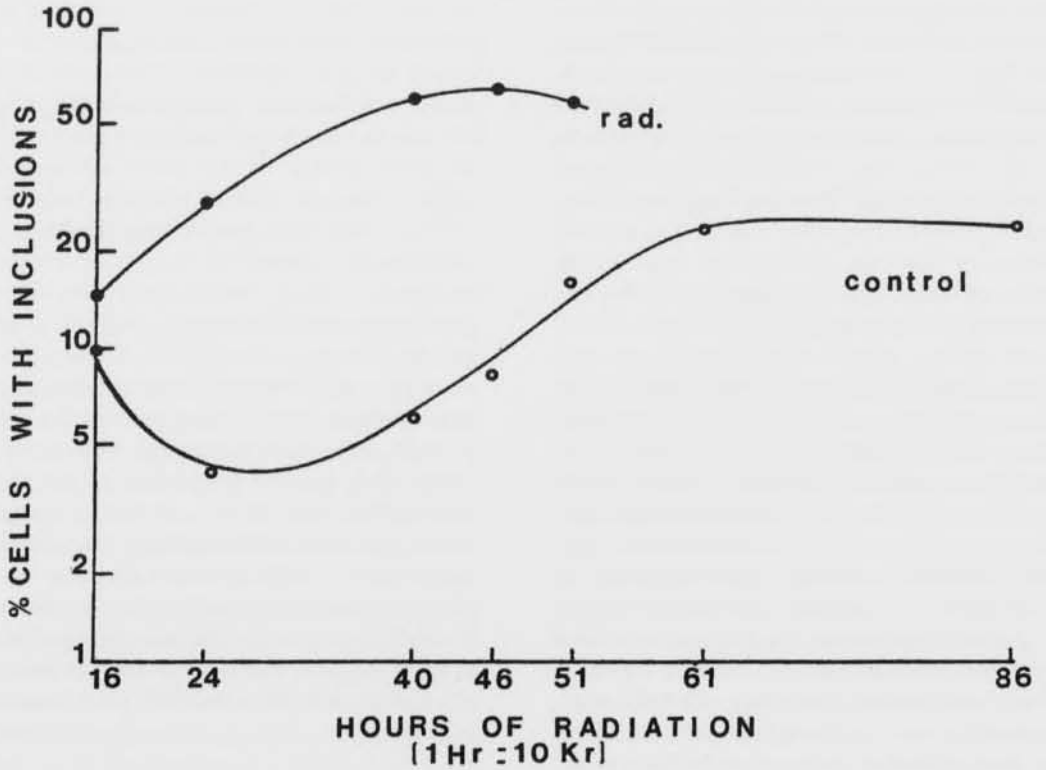


Fig. 5. Semi-logarithmic relationship between the appearance of inclusion bodies (dark red granules and pink vacuole-like structures) in the cytoplasm of Trichmonas vaginalis and gamma radiation greater than 1600 Gy. Unirradiated (control) cells were kept in the dark for a period equivalent to the time needed for a given radiation dose (100 Gy/hr). Points represent the averages for 4 experiments.

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Response of an Arkansas White-tailed Deer Population to Harvest

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Harvest management of game species for the purpose of maintaining or manipulating populations requires estimation of at least 3 population parameters: population size (density), recruitment, and mortality. Knowledge of these parameters is important because hunters, as well as anti-hunters, and the general public call for managers be able to defend their management activities (Lancia et al., 2000). Managers most often use indices, such as number of deer observed per unit distance driven during a spot-light survey, in place of population parameters. In doing so, the assumption is made that a constant, linear relationship exists between the index and the population parameter of interest (Lancia et al., 1996). Use of such indices is based on tradition, simplicity, and low cost. However, few indices have been validated (Rotella and Ratti, 1986), and none serve as a perfect substitute for population parameter estimation.

Different approaches are available to estimate density, recruitment, and mortality (Johnson, 1996; Lancia et al., 1996). Density estimation has been conducted using mark-recapture techniques (Peterson, 1896; Lincoln, 1930; Bartmann et al., 1987), distance sampling (Buckland et al., 1993), area sampling (Seber, 2002), and removal techniques (Zippin, 1958; Lancia et al., 1988). For white-tailed deer in the southeastern U.S., density estimates are most often derived using mark-recapture approaches, but distance sampling is becoming more widely used (Langdon, 2001; Lopez et al., 2004). Area sampling, which is most commonly used for large ungulates in the western U.S., is not very applicable in the southeastern U.S. due to limited visibility using traditional techniques (e.g., helicopter surveys). However, advances in the use and technology of thermal infrared imagery are changing the ability to employ area sampling during leaf-off conditions in deciduous hardwood areas of the Southeast.

Recruitment is typically estimated using a sex/age ratio approach (Downing, 1980; Ginnett and Butch-Young, 2000). Population size is estimated and an independent estimate of the number of young per adult female (e.g., fawns/100 does) is applied to the population estimate. The manner in which the ratio is obtained varies depending upon location. Spotlighting is most commonly used to obtain ratios in the southeastern U.S., whereas visual aerial surveys are used in the western U.S.

Estimation of mortality is directly provided by radio-telemetry (Lancia et al., 2000). Mortality estimation using radio-telemetry, however, requires a great amount of time and is financially expensive. Most mortality of white-tailed deer is due to harvest (Dusek et al., 1989), and harvest is usually used as an index to mortality.

Population-parameter estimation has not been used often by managers for white-tailed deer in Arkansas. With implementation of new management strategies for white-tailed deer or acquisition of new properties, baseline data that includes population parameters are useful to determine the efficacy of the management strategies employed. The Arkansas Game and Fish Commission (AGFC) purchased a new property, the Choctaw Island Wildlife Management Area (CIWMA), in October 2001. The deer population was not hunted for 2 seasons following the purchase. The first hunting season under the direction of the AGFC was during fall 2003. Because the CIWMA was a newly acquired wildlife management area, AGFC desired baseline data for white-tailed deer management. Our objectives were to 1) estimate winter deer density on Choctaw Island Wildlife Management Area, and 2) determine if the population was reduced based on harvest.

The study was conducted on the CIWMA located in Desha County, Arkansas (Lat. 33°35′ 47″ N, Long. 91°11′20″ W). The CIWMA was approximately 3268 ha in size and lies within the levee of the Mississippi River. The area was divided into two parts, the mainland (2361 ha) and an island (907 ha) in the Mississippi River, but was managed as one property and one population. Topography was flat and elevation ranged from 33.5 to 46.0 m. The entire area was subject to seasonal (winter-spring) flooding. Cover types were bottomland hardwood forests, cottonwood (*Populus deltoides*) plantations, open fields, and food plots. Dominant tree species were oaks (*Quercus* spp.), pecan (*Carya illinoensis*), and cottonwood. Mean total precipitation for February and March was 13.34 cm and 13.46 cm, respectively. The mean minimum temperature during February and March was 2°C and 6.5°C, respectively (Dermott, AR, Station 031962).

Non-overlapping, parallel transects were established and surveyed from a Cessna 182 fixed-wing aircraft. Transects were approximately 400 m apart. The site was surveyed once each night on 8 and 9 March 2003 and on 20, 21, 22, and 27 February 2004. Flights were conducted at approximately 457 m above ground level (AGL), and height AGL was maintained using an on-board altimeter; resulting strip transects were approximately 110 m wide. Flights were conducted between 2000 and 2300 hrs in 2003 and between 2300 hrs and 0600 hrs in 2004. Flights were conducted at

different times the second year in an attempt to maximize the detection of thermal signatures. Flight paths (lat., long., WGS84), altitude (ft), speed (mph), date and time were collected by an onboard Global Positioning System (GPS) unit. The GPS signal was routed through a video encoder-decoder (VED). Locations of the plane obtained from the GPS unit were recorded on the audio portion of the video tape. The VED continuously labeled the video tape with position, time, date, speed, and altitude information.

Surveys were conducted using an IR-M700 thermal infrared imager (Mitsubishi, Inc., Ontario, Canada) with a 50 mm lens mounted in the belly of the plane in a fixed, vertical position. Wavelengths ranging from 1.2 to 5.9 _m were used. The detector array size was 801 x 512 pixels with a sensitivity of 0.08 oC. Output was conducted through an RS170, 75 _ connection to a digital video camera (Sony DCR-TRV1000). Video was reviewed using a 33 cm, 1000 line resolution, black and white monitor (Sony PVM-137).

We used area sampling to estimate deer density in the CIWMA. The assumption made when using area sampling is that all animals are detected within the area surveyed. A high rate of detection (94%) was reported for thermal targets in associated research (Kissell and Tappe, 2004). The number of deer observed in each strip transect was recorded. Double counting was prevented by use of GPS locations integrated with videography and spacing of transects (Naugle et al., 1996). GPS data were transferred into a geographic information system (GIS). Length and width of transects were used to compute the area sampled during each survey. We assumed an average altitude of 457 m AGL for the purpose of calculating the area surveyed. Density was calculated as the number of deer recorded per unit area, and nights were used as replicates.

A comparison of density estimates between years for the mainland and the island was conducted using t-tests adjusted for unequal variance when necessary (SAS Institute, Inc., Cary, N.C.). All analyses were conducted with _ = 0.05.

Recruitment was assessed using spotlight surveys conducted from the back of a pickup truck moving ≤ 8 km/hr. Surveys were conducted during January 2004 following the hunting season. Both sides of the road were scanned using a 750,000 candle power spotlight. Upon detection of animals, the age (adult or juvenile), sex, time, and group number were recorded. Surveys began at least 1 hr after sundown and continued until a pre-determined route was completed. Surveys were conducted nightly until 100 does were observed. The number of fawns per 100 does was calculated to represent recruitment. Harvest data were provided by the AGFC. During 2003, 33 transects on the mainland and 18 transects on the island were surveyed, and in 2004, 84 transects on the mainland and 47 transects on the island were surveyed. The strip transect area covered during the surveys in 2003 varied from 434 ac (175.6 ha) to 1248 ac (505.1 ha), and from 621 ac (251.3 ha) to 1344 ac (543.9 ha) in 2004 (Table 1). The mean density was 1 dee per 7.4 ac (3.0 ha) and 1 deer per 8.8 ac (3.6 ha) on th mainland in 2003 and 2004, respectively. The mea population sizes on the mainland during the winters of 200 and 2004 were estimated to be ~ 788 and ~ 659 dee respectively.

No significant differences were found between years fo the densities on the mainland ($p = 0.300$, $t = 1.19$) or th island ($p = 0.397$, $t = 1.38$). Only two replicate flights wer obtained during 2003 and this resulted in more variance compared to that obtained from 4 replicates in 2004. A difference between densities on the island and mainland during 2004 ($p = 0.003$, $t = 4.96$) existed but was not detected in 2003 ($p = 0.205$, $t = 1.86$) due to the variance. Though conditions were good during both nights that transects were flown in 2003, slightly poorer conditions were experienced the second night. Daytime heating translated into greater thermal loading of vegetation and water. This may partially explain why density estimates from the second flight were slightly lower than those from the first flight. We flew after sunset and prior to 0100 hrs. To minimize the effects of thermal loading, flights were conducted later in the night in 2004. Variability of detection was much higher on the island compared to the mainland. We believe this may have been due to either the poorer conditions experienced during the second flight, a change in deer behavior (e.g., animals moving off the island or being bedded under vegetation), or a combination of these factors.

While there was no difference in deer density between years, more deer were observed on the island in 2004 (Table 1), the year following the first deer harvest season. Deer are known to increase movements, increase home range size, and even shift activity centers in response to hunting pressure (Root et al., 1988). It is possible that deer moved to the island in response to hunting pressure on the mainland and remained there at least through February when the flights were conducted. Assuming that the increase in numbers observed on the island was due to movement, it is not known whether the movements were permanent because no active radio-telemetry work was under way at the time. Another possible explanation is that the deer population on the island increased from one year to the next. We believe this is very unlikely, however, due to the relatively poor habitat on the island.

In addition to the density data, spotlighting surveys indicated 32.2 fawns:100 does (recruitment) and 15.7 bucks per 100 does. The level of recruitment suggested a population above carrying capacity. Poor recruitment is a function of population density (Gilbert and Raedeke, 2004) and nutrition (Fryxell et al., 1991; McCullough, 2001). Nutrition most likely has a time-lag effect on recruitment (Fryxell et al., 1991), and managers should expect a slow recovery due to nutrition-mediated recruitment when densities have been high for a prolonged period of time.

The CIWMA deer population is an exemplary model of these conditions.

The AGFC reported a harvest of 181 deer during the 2003-2004 season, of which 157 were does (C. Gray, Arkansas Deer Program Coordinator, pers. comm.). In response to the failure of harvest to initially reduce the population, AGFC changed their harvest strategy to encourage the harvest of more does. During the 2004-2005 deer season, a total of 269 deer was harvested, of which 189 were does (C. Gray, pers. comm.). While a density estimate was not provided during winter 2005, it is believed that a sufficient number of animals were harvested to begin decreasing the population.

We recommend further estimation of population parameters annually because harvest is skewed toward does, a sizeable proportion of the population is being harvested, and the duration of a nutrition-mediated time-lag in recruitment is unknown. This work serves as a model for white-tailed deer management in Arkansas and encourages managers and biologists to make decisions based on population parameter estimation and not indices.

ACKNOWLEDGMENTS.—The authors wish to thank L. Davis for providing valuable information, assistance, and professionalism in flying. We also thank C. Gray and other personnel of the Arkansas Game and Fish Commission (AGFC) Southeast Regional Office for providing in-kind support of this and associated research. Funding was provided by AGFC and the Arkansas Forest Resources Center.

Table 1. Density estimates (number of deer per acre; ha are in parentheses) of white-tailed deer on Choctaw Island Wildlife Management Area, Desha County, Arkansas, calculated from aerial thermal infrared videography during 8-9 March 2003 and 20-27 February 2004. See text for description of Island and Mainland.

Site	Date flown	Acres sampled (ha)	Number of deer observed	Density #deer/acre	Mean(ha)	S.E.(ha)	Estimated number of deer
Island	8 March 2003	434 (176)	20	0.046 (0.114)			
	9 March 2003	660 (267)	12	0.018 (0.045)	0.026 (0.065)	0.014 (0.035)	58
Mainland	8 March 2003	1163 (471)	194	0.167 (0.417)			
	9 March 2003	1248 (501)	144	0.115 (0.286)	0.135 (0.333)	0.026 (0.066)	788
Island	20 February 2004	621 (251)	50	0.081 (0.200)			
	21 February 2004	642 (260)	44	0.068 (0.169)			
	22 February 2004	676 (274)	40	0.059 (0.147)			
	27 February 2004	673 (272)	39	0.058 (0.143)	0.065 (0.161)	0.005 (0.013)	147
Mainland	20 February 2004	1332 (539)	148	0.111 (0.278)			
	21 February 2004	1328 (537)	149	0.112 (0.278)			
	22 February 2004	1333 (540)	185	0.139 (0.345)			
	27 February 2004	1344 (544)	131	0.097 (0.238)	0.114 (0.278)	0.009 (0.022)	659

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Second Record of the Dipluran, *Occasjapyx carltoni* Allen, 1988 (*Japygidae*), from Arkansas

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In North America, the genus *Occasjapyx Silvestri*, 1948 (Diplura:Japygidae) currently includes 5 species, *O. americanus* (MacGillivary, 1893), *O. californicus Silvestri*, 1948, *O. carltoni* Allen, 1988, *O. koboidi* Silvestri, 1928, and *O. sierrensis* Smith, 1959 (Smith, 1959; Reddell, 1983; Allen, 2002). Four of these taxa occur in the far western United States in various parts of California, whereas *O. carltoni* is found in the east-central U.S. in the Ozark Mountains of northern Arkansas. Allen (1988) described *O. carltoni* based on 2 specimens collected near the Buffalo River at Kyle's Landing, Indian Creek, Newton County, Arkansas on 7 March 1988. The specimens were collected under rocks along a creek bank. Allen (1988) reported that both types were deposited in the University of Arkansas Insect Collection, but Robison and Allen (1995) gave the American Museum of Natural History, New York, NY as the holotype repository and Arkansas as the paratype repository. The latter specimen was apparently later transferred to the Academy of Natural Sciences, Philadelphia (R. T. Allen, pers. comm.). Since the original description, we are unaware of additional reports of the species in the literature other than a color photograph, a line drawing, and additional commentary of this Arkansas endemic in Robison and Allen (1995).

On 30 December 2004, the first author collected a single specimen of an unknown japygid species in muddy substrate within the twilight zone of Blevins Cave (formerly Cave Creek Spring Cave), 9.8 km (6.1 mi) north of Pleasant Plains off US 167 along Powers Creek, Independence County, Arkansas. The specimen was placed in a vial containing 70% ethanol and sent to the second author for identification. Based on the terminal lamina of the lacinia, antennae, cerci, and dorsal chaetotaxy, the specimen was tentatively identified as *O. carltoni*. A voucher specimen is deposited in the invertebrate collection of the Louisiana State Arthropod Museum.

The new collection site is approximately 167 km (104 mi) southeast of the type locality (Fig. 1). Interestingly, records of diplurans from Arkansas caves are rare (see McDaniel and Smith, 1976; McDaniel et al., 1979; Dunivan et al., 1982; Graening et al., 2003). As such, we suggest additional collecting of diplurans in epigean habitat in the state.

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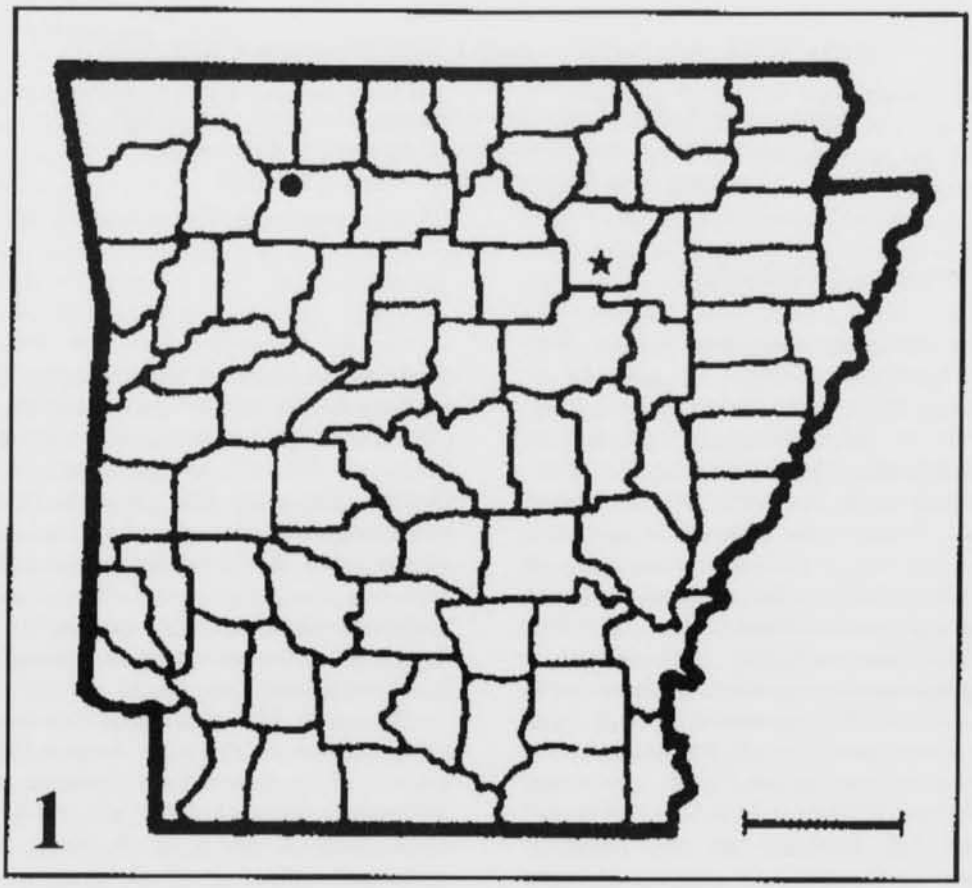

Fig. 1. County outline map of Arkansas showing localities for *O. carltoni*. Type locality in Newton County (dot), new locality in Independence County (star). Scale bar = 81 km (50 mi).

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Identification of Cystacanths and Adults of *Oligacanthorhynchus tortuosa*, *Macracanthorhynchus ingens*, and *Macracanthorhynchus hirudinaceus* Based on Proboscis and Hook Morphometrics

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Identification of cystacanths of certain acanthoce-phalans belonging to the family Oligacanthorhynchidae has been difficult due to discrepancies in the literature concerning proboscis and hook morphometrics (Meyer, 1933; Moore, 1946; Van Cleave, 1953; Schmidt, 1972; Elkins, 1981). The purpose of this study was to conduct direct comparison of proboscis and hook morphometrics of *Oligacanthorhynchus tortuosa* (Leidy, 1850) Schmidt, 1972, *Macracanthorhynchus ingens* (Linstow, 1879) Meyer, 1932, and *Macracanthorhynchus hirudinaceus* (Pallas, 1781) Travassos, 1917. Recent acquisition of cystacanths of *O. tortuosa* and young juveniles of *M. ingens* provided material for comparison of cystacanths and adults of these 2 species. Resultant data make the identification of both cystacanths and adults of these acanthocephalans of North American mammals possible, greatly facilitating epizootiological investigations.

Adults and very young juveniles of *M. ingens* were acquired from raccoons utilized in a study of the population structure and dynamics of *M. ingens* from Ossabaw Island, Georgia (Richardson and Barger, 2005). Adult *M. hirudinaceus* from domestic swine were acquired from a biological supply company. Adult *O. tortuosa* were acquired from Virginia opossums collected in Pope, Searcy, and Van Buren counties in Arkansas (Richardson, 1993; Richardson and Barnawell, 1995). Data for cystacanths of *O. tortuosa* were taken from Richardson (in press) who demonstrated the life cycle of *O. tortuosa* using cystacanths from millipedes (*Narceus americanus*) collected in St. Tammany Parish, Louisiana. Voucher specimens were deposited in the Harold W. Manter Laboratory, Lincoln, Nebraska, and assigned accession numbers as follows: proboscides of adult *M. ingens* HWML48143); proboscides of adult *M. hirudinaceus* HWML48144); proboscides of adult *O. tortuosa* HWML48145); juvenile *M. ingens* (HWML48146); cystacanths of *O. tortuosa* (HWML48149).

Proboscides were removed from adult worms. All specimens were treated and microscopically examined and drawn according to Richardson (in press). All measurements were made as prescribed by Van Cleave 1953) as follows. Hook numbers were ascribed considering a hook arrangement of 6 diagonal rows of 6 hooks each or 6 circular rows of 6 hooks each. Either arrangement results in the same numerical hook assignments (see Text Fig. C of Van Cleave (1953)). Measurements of hook length were conducted on hooks in full lateral view as shown in Fig. 1 being measured as a straight line connecting the free point of the thorn with the point where the thorn joins the root. Proboscis length was measured from the anterior end of the proboscis to the insertion of the hook blade of hook number 6. Proboscis width was measured at the widest point (Fig. 2). All measurements are given in µm with the range followed by the mean in parentheses. Statistical analyses were conducted using a Student's 2-tailed t-tests (Microsoft® Excel 2002). Significant differences assume $p < 0.05$.

Proboscides and hooks of *M. hirudinaceus* (Fig. 3) are larger than those of *M. ingens* (Fig. 4), which in turn are larger than those of *O. tortuosa* (Fig. 5). No significant differences were detected in proboscis length and hook length between cystacanths and adults of *M. ingens* and *O. tortuosa*. Barbs (Fig. 7) were observed inconsistently among hooks for all 3 species. Proboscis and hook morphometrics are summarized in Tables 1 and 2.

Both cystacanths and adults of *O. tortuosa*, *M. ingens*, and *M. hirudinaceus* may be easily identified based on proboscis and hook morphometrics. Differences in hook size among the 3 species are most dramatically exhibited by hook number 3 (Figs. 6-8).

Hook size and proboscis length appear to remain stable through development from cystacanth to adult. The increase in proboscis width observed may reflect changes in musculature as opposed to true growth of the proboscis. These data support the assertion of Moore (1962) in regard to *Mediorhynchus grandis* that proboscis and hook morphometrics are fixed by the time worms become infective cystacanths. Van Cleave (1941) and Elkins

(1981) made the same observation in regard to hook morphometrics. Cystacanths of *M. ingens* and *O. tortuosa* are shown in Figs. 9 and 10.

It is well established that adult female acanthocephalans attain much greater sizes than adult males. Richardson (in press) found that female cystacanths of *O. tortuosa* are significantly more robust than males and have significantly larger proboscides and hooks. Thus, it appears that the size difference between sexes is apparent by the time worms become infective cystacanths.

ACKNOWLEDGMENTS.– Michael A. Barger, Peru State University assisted in specimen preparation. This study was funded by a Summer Research Grant from the School of Health Sciences, Quinnipiac University.

Table 1. Summary of proboscis morphometrics for adult *Macracanthorhynchus ingens, Macracanthorhynchus hirudinaceus,* and *Oligacanthorhynchus tortuosa* and cystacanths of *M. ingens* and *O. tortuosa.* All measurements are in µm. Range is followed by mean in parentheses.

Species and Ontogenetic Stage	Length	Width	Length:Width Ratio
M. ingens Adult	405-459 (437)	653-729 (683)	0.62-0.68 (0.64)
M. ingens Cystacanth	390-546 (467)	504-700 (590)	0.73-0.87 (0.79)
M. hirudinaceus Adult	716-952 (794)	873-1260 (1119)	0.62-0.88 (0.72)
O. tortuosa Adult	248-315 (282)	257-325 (291)	0.86-1.13 (0.97)
O. tortuosa Cystacanth	239-324 (282)	238 311 (277)	0.90-1.10 (1.00)

Table 2. Summary of hook lengths for adult *Macracanthorhynchus ingens, Macracanthorhynchus hirudinaceus,* and *Oligacanthorhynchus tortuosa* and cystacanths of *M. ingens* and *O. tortuosa.* All measurements are in µm. Range is followed by mean in parentheses.

Species and Ontogenetic Stage	Hook 1	Hook 2	Hook 3	Hook 4	Hook 5	Hook 6
M. ingens Adult	160-212 (185)	149-207 (182)	104-158 (135)	108-158 (123)	86-106 (96)	72 99 (86)
M. ingens Cystacanth	153-212 (182)	151-196 (173)	117-158 (137)	95-133 (114)	86-104 (95)	59-90 (82)
M. hirudinaceus Adult	185-325 (254)	196-291 (241)	225-302 (268)	160-218 (192)	131-221 (156)	95-162 (137)
O. tortuosa Adult	65-101 (89)	63-72 (67)	52-81 (61)	43-73 (57)	45-54 (50)	36-50 (42)
O. tortuosa Cystacanth	78-104 (90)	59-89 (74)	55-74 (62)	48-76 (57)	36-56 (47)	34-50 (41)

Fig. 1. Proper measurement of hook length indicated by dotted line.

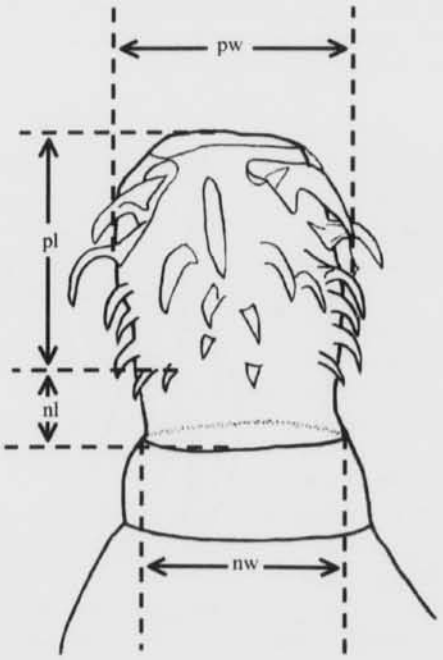
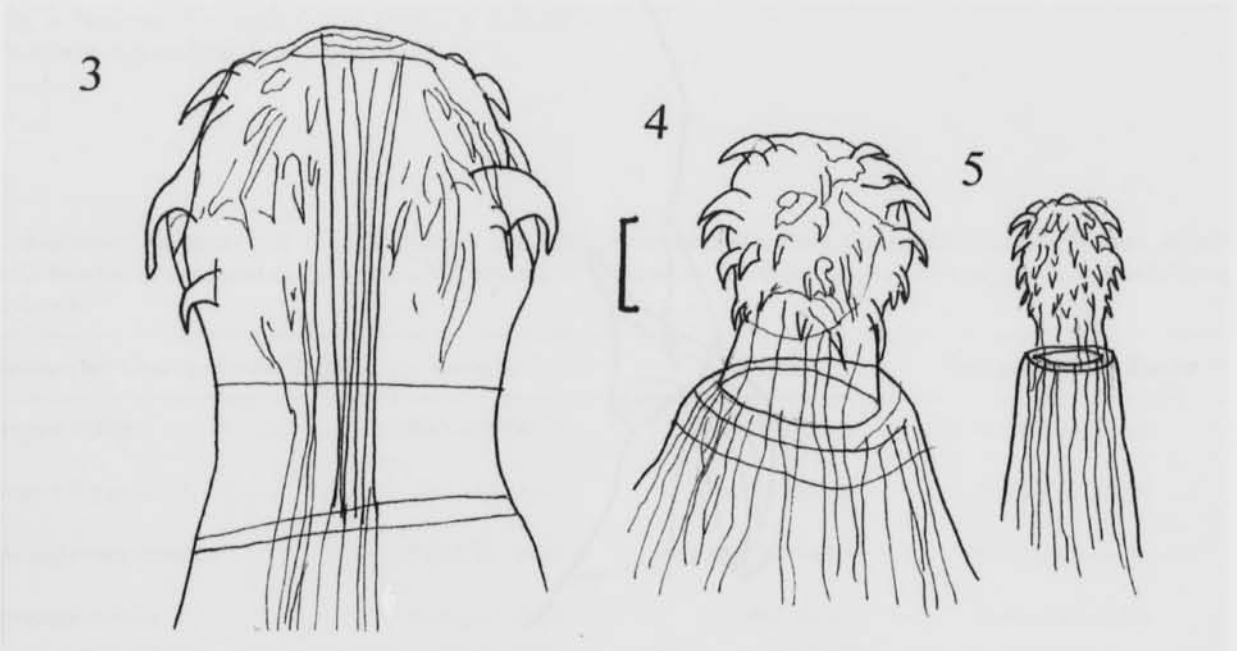
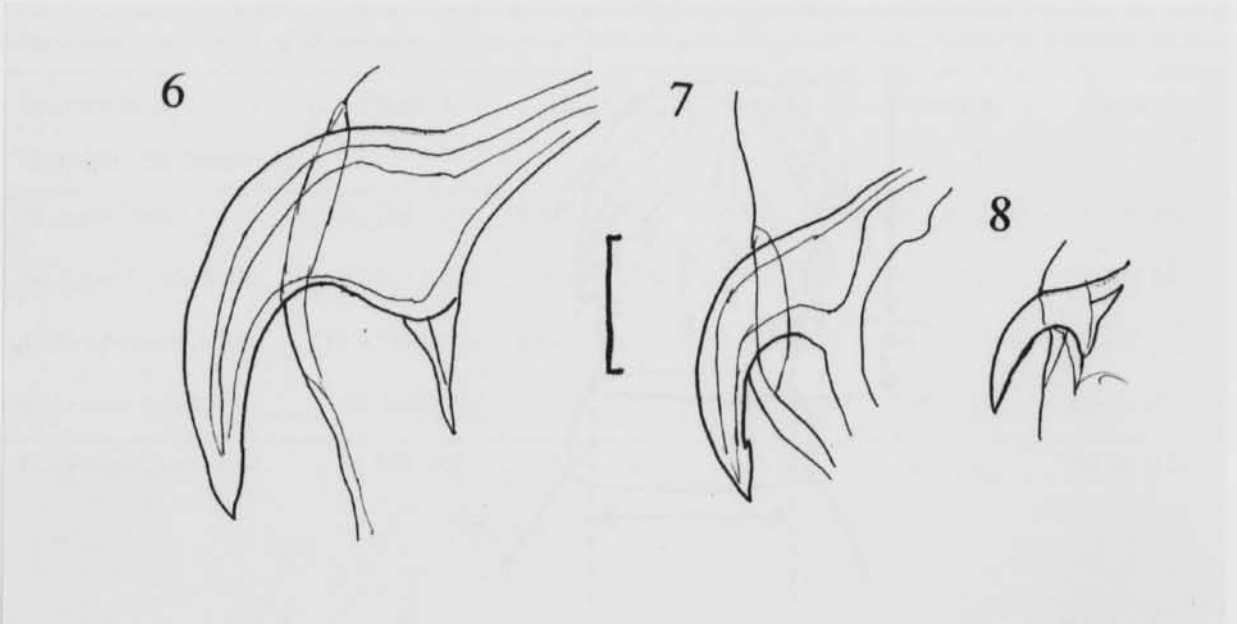


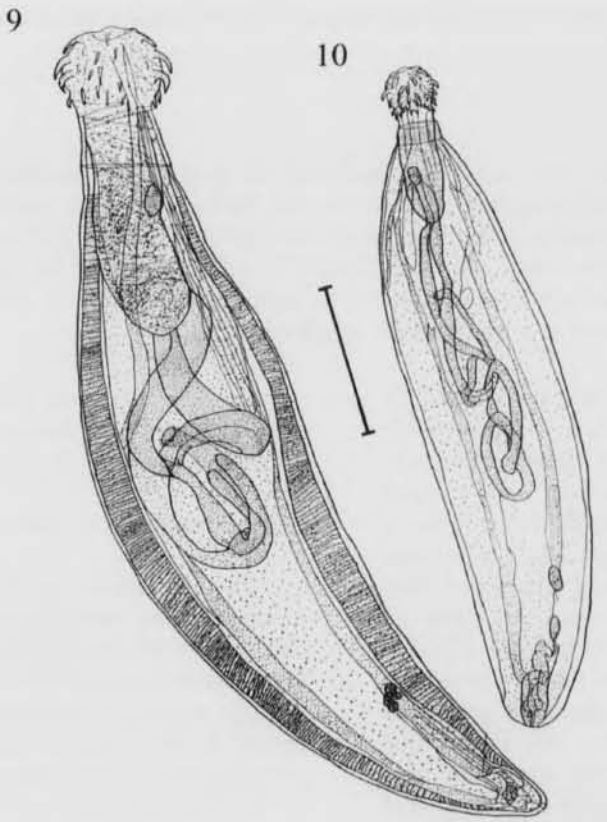
Fig. 2. Proper measurement of proboscis length (pl), proboscis width (pw), neck length (nl), and neck width (nw).



Figs. 3-5. 3. Proboscis of *Macracanthorhynchus hirudinaceus*, 4. *Macracanthorhynchus ingens*, and 5. *Oligacanthorhynchus tortuosa.* Scale bar = 250 µm.



Figs. 6-8. 6. Hook number 3 of *Macracanthorhynchus hirudinaceus*, 7. *Macracanthorhynchus ingens*, and 8. *Oligacan- thorhynchus tortuosa.* Scale bar = 50 µm.



Figs. 9 and 10. Cystacanths of 9. *Macracanthorhynchus ingens* (HWML48147) and 10. *Oligacanthorhynchus tortuosa* (HWML48148), respectively, removed from the hemocoel of a millipede (*Narceus americanus*) Scale bar = 1 mm.

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New Pulsating Variable Discovered In The Constellation Andromeda

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A new pulsating variable star, HH95 HV And-7, is found near the cataclysmic variable HV And, which is a part of the Indiana University RoboScope observing program (Honeycutt and Turner, 1992). A finding chart generated with Aladin software (Bonnarel et.al., 2000) is shown in Fig. 1. Its coordinates are (J2000) 00°40'46.23" +43°23'57.9". This star was initially calibrated as a secondary photometric standard star with V=15.277 and B-V=0.281 for the field of HV And (Henden & Honeycutt, 1995), but it suspiciously had the largest standard deviation of the group of standards ($\partial_v = 0.14$). Its variability detailed here means that it can not be used as a photometric standard.

RoboScope is an 0.41-m telescope fully automated for unattended differential CCD photometry of cataclysmic variables and related objects located in the Morgan-Monroe state forest north of Bloomington Indiana. It uses a liquid nitrogen cooled, thinned SITe CCD with 24 micron pixels in an array of size 512_512 pixels yielding a 14x14 arcminute field of view and typically obtains 1-2 exposures per clear night for ~150 objects. The field of HV And was observed on 318 separate nights from HJD 2448473 (UTD 1991-08-04) through HJD 2450141 (UTD 1996-02-27) through a standard Johnson V filter. The inhomogeneous ensemble photometry (Honeycutt, 1992) used on the RoboScope database can yield the light curve for every star in the field of interest. The variability was revealed by the large 0.14 sigma uncertainty in its instrumental ensemble magnitude shown in Fig. 2.

A period search of the RoboScope data for this star using the method of Horne and Baliunas (1986) is shown in Fig. 3. A total of 5000 individual frequencies were tested from 0.1 to 2.0 days. The strongest peak at 0.4661 day is flanked by spurious peaks at 0.32, 0.87, 0.34, 0.53 days. These represent aliases of the true period (P) due to the time sampling frequency (t) of the data (~1 day). They correspond to spurious periods (P) found from

$$\mathbf{1/P = 1/P \pm e/t}$$

(Lafler & Kinman, 1965) for values of e = +1, -1, +1/2, and -1/4 respectively. Power spectra were also generated for several data sets constructed such that the time of true observations was preserved, but the magnitudes were randomly shuffled and assigned to these times. This has the effect of evaluating the "windowing" function for the period search, discriminating against periods associated with the time sampling of the data and further checking the validity of the power found in the strongest peak. No peaks with power larger than ~5 were found compared to ~120 for the true period.

The search results reveal a pulsating variable with a period of 0.46614(5) days as shown in the phased light curve for integer cycle (N) in Fig. 4 using the ephemeris

$$\mathbf{Maximum = 2448400.0(0) + 0.46614(5) \_ N.}$$

The accuracy in the period was estimated by minimizing the total distance (string-length) between phased data points for various periods near the peak period. Since the observational data is over a timescale much larger than a single period, the folding of the data in phase allows the period to be obtained to high accuracy since different periods would cause larger string-lengths in the phased light curve. The taxonomy of its light curve, namely the 0.5 magnitude amplitude, the ~1/2 day period and symmetric sinusoidal shape is indicative an RR Lyrae pulsating variable, most likely RRc (Feast, 1996).

Since it is not associated with a known globular cluster, it is likely a galactic halo object or old, thick disk member of our galaxy (Feast, 1999). Using its mean magnitude of mv=15.3 and a simple Period-Luminosity relationship for RR Lyrae stars MV = -2.8 log P - 0.6 (Eggen, 1994), yields an absolute magnitude of MV = 0.328 and a distance of ~10,000 parsecs (=32,600 Ly).

ACKNOWLEDGMENTS.—I gratefully acknowledge the ever-changing vanguard of RoboScope "baby-sitters" who keep the RoboScope system operational and always include Kent Honeycutt, Brice Adams, George Turner, and Bill Kopp.

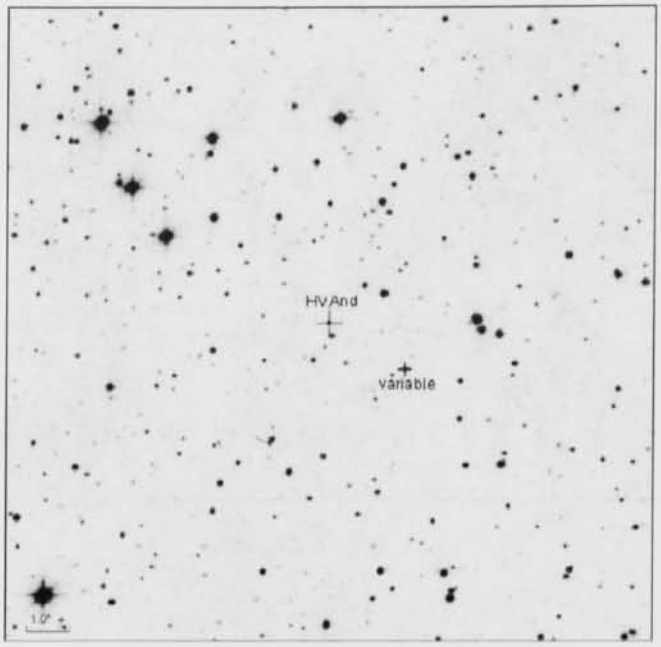

Fig. 1. A 14x14 arcminute finding chart for the new variable, HH95 HV And-7, near the cataclysmic HV And. North is up and East is to the left in the figure.

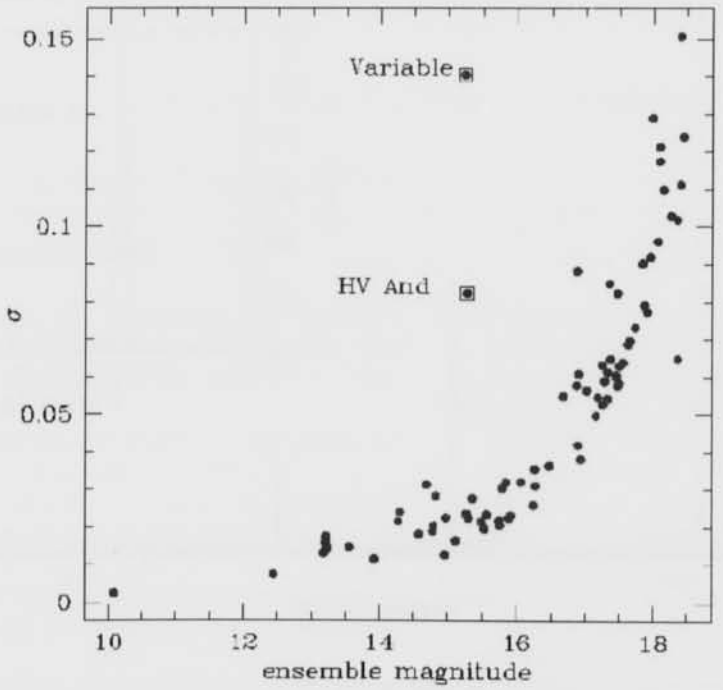

Fig. 2. Standard deviation of ensemble magnitudes for nearby stars in the field revealing variability.

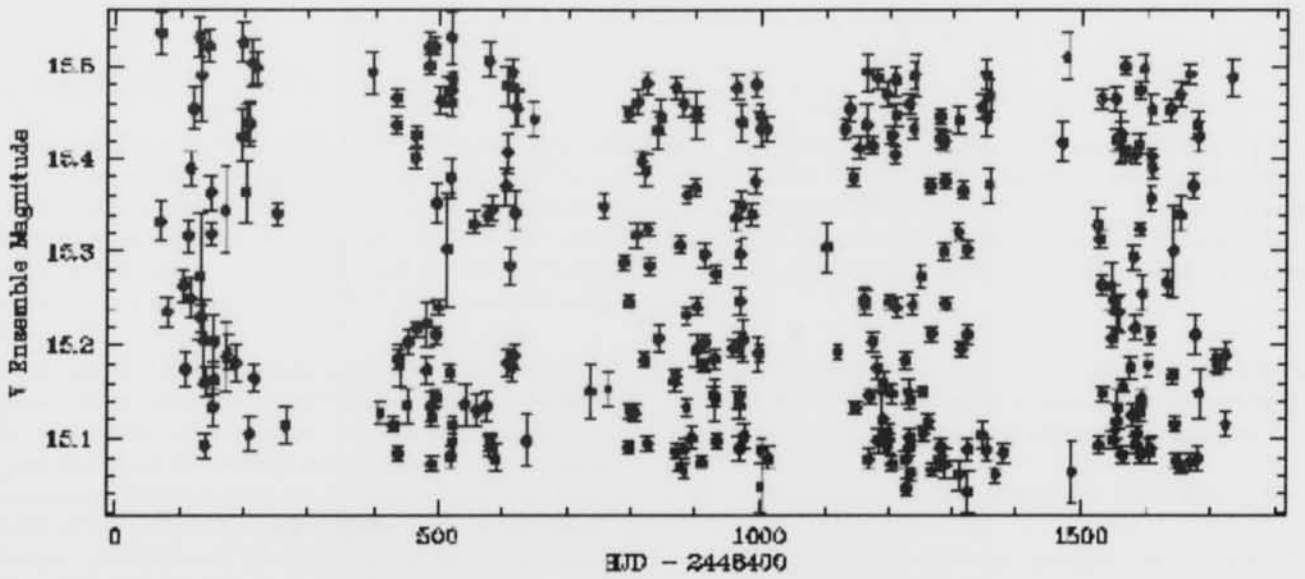

Fig. 3. Differential ensemble photometry of HH95 HV And-7.

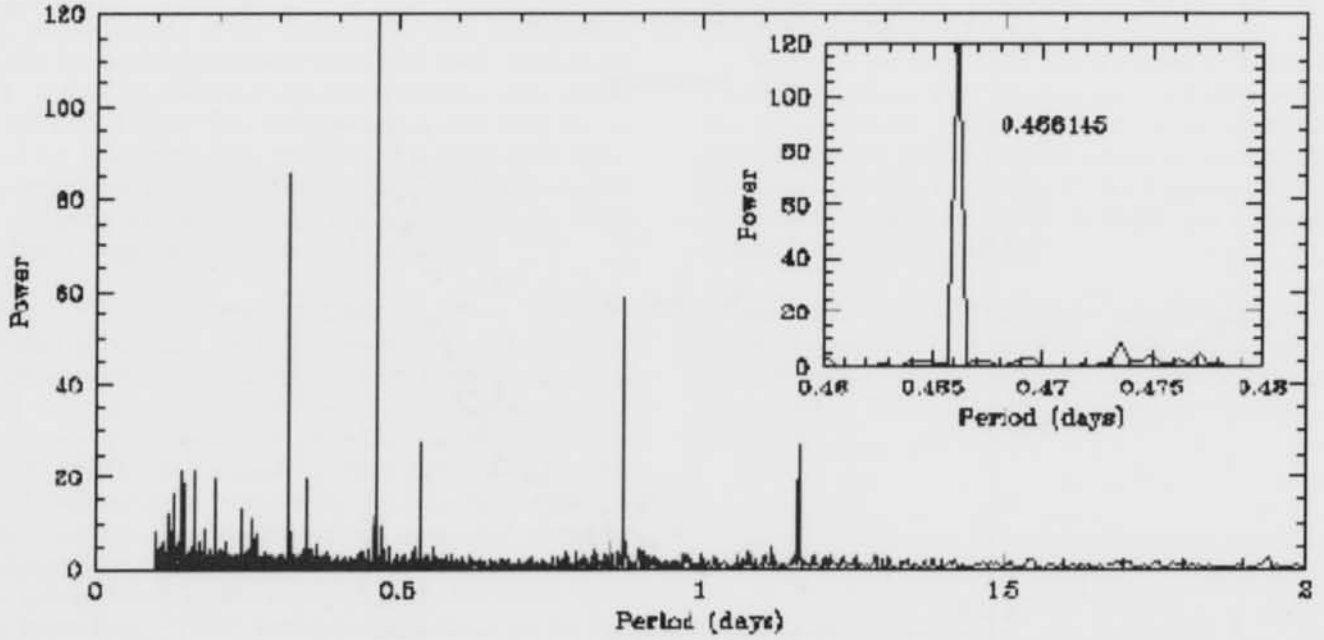

Fig. 4. A power spectrum period search of the light curve data in Fig. 3.

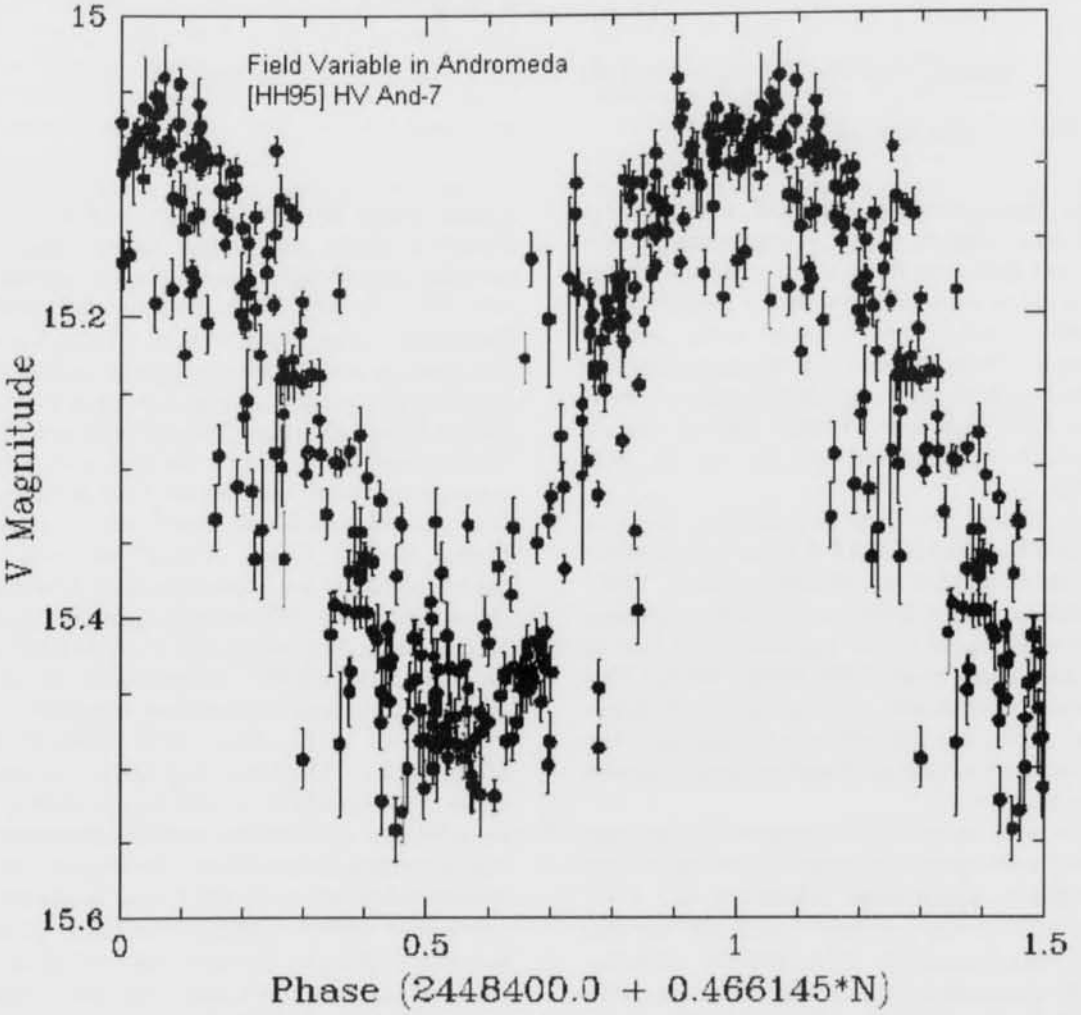

Fig. 4. A power spectrum period search of the light curve data in Fig. 3.

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Pesticide Residues in Guano of Gray Bats (*Myotis grisescens*) in Arkansas

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The gray bat, *Myotis grisescens*, is a small, insectivorous bat found in the karst regions of the southeastern United States. In the winter gray bats form a few extremely large colonies in cooler caves, while in the summer they disperse to more numerous caves over a large area, usually segregated by gender. Primarily due to population declines thought to be the result of excessive disturbance of these colony sites, the U.S. Fish and Wildlife Service placed the gray bat on the endangered species list in 1976 (Brady et al., 1982).

Subsequent to listing, gray bats in maternity caves in Missouri were killed by dieldrin (CAS # 60-57-1), a product of the insecticide aldrin (CAS # 309-00-2)(Clark et al., 1978). In addition to dieldrin, lethal levels of heptachlor epoxide (CAS # 76-44-8) were found in the brains of gray bats at other maternity sites (Clark et al., 1980, 1983a, 1983b). This mortality probably contributed to population declines observed at these caves, but it is difficult to determine the role of other factors such as human disturbance and normal population fluctuations.

Contaminants were most likely acquired by the bats while feeding on arthropods exposed to the pesticides following agricultural application (Clark et al., 1978, Clawson, 1989, 1991). The primary pesticide causing mortality, aldrin, was banned in 1974, and the chemical substituted for it, hephachlor, was made illegal in 1979, however these and the breakdown products of DDT, such as DDE, have been found to be extremely persistent in the environment (Clawson, 1991). Significant population declines of other bat species such as the Brazilian free-tailed bat (*Tadarida brasiliensis*) have also been traced to DDT poisoning including intentional applications of DDT to nursery colonies (Clark et al., 1978; Clark, 2001).

Although 1 prior study found that developing a statistically significant relationship between pesticide residues in bat guano and that in bat carcasses may not be possible, pesticide presence in guano is at least an indication that there may be more significant problems in the population. Samples taken from guano piles may represent a broad cross-section of the population as they include guano from large numbers of individuals; guano sampling also avoids the need to collect bat carcasses, which are often too decomposed for testing. Dieldrin concentrations in guano above 0.38 ppm have been found at caves where pesticide-related mortality has occurred (Clark et al., 1981).

The study was conducted at 4 gray bat caves in Arkansas: Bone and Dodd caves (Independence County), Logan Cave (Benton County), and Morris Cave (Sharp County). Bone and Logan caves are both maternity colonies, and though Dodd Cave was a maternity colony in the 1970's (Saugey, 1978), since the early 1980s, it has only been used in the spring and fall. Morris Cave is a transient site used by gray bats in the spring. Guano from the upper layers of guano piles in these caves was collected in January (Logan Cave), February (Morris Cave), and April (Bone and Dodd caves), 2004 using a stainless steel spoon and kept at room temperature until tested. Four samples were taken in Bone, Dodd, and Logan caves, and 3 samples were taken from Morris Cave. Using electron capture gas chromatography the Mississippi State Chemical Laboratory (Mississippi State, MS) analyzed samples for the presence of 25 chemical compounds: HCB, alpha BHC, gamma BHC, beta BHC, delta BHC, oxychlordane, heptachlor epoxide, gamma chlordane, t-nonachlor, toxaphene, PCB – 1242, PCB – 1248, PCB – 1254, PCB – 1260, PCB – Total, o,p'-DDE, alpha chlordane, p,p'-DDE, dieldrin, o,p'-DDD, endrin, cis-nonachlor, o,p'-DDT, p,p'-DDD, and mirex.

Thirteen of 15 (87%) samples contained p,p'-DDE with values ranging from 0.011 to 0.057 ppm (dry weight). This breakdown product of DDT was found in samples from every cave whereas dieldrin was found in only 2/15 (13%) samples (Table 1). Dieldrin concentrations at Bone and Dodd caves were 0.012 and 0.014 ppm respectively in the two samples in which dieldrin was detected. The remaining 23 compounds were not found in any samples.

This study confirms that gray bats in Arkansas caves have been and are continuing to be exposed to pesticide residues that are potentially fatal. However, the pesticide concentrations found in guano from this small sample of Arkansas maternity caves are low in comparison to those found in previous studies (Table 2). Dieldrin was found at 2 caves at low concentrations. Levels of dieldrin previously associated with bat mortality (> 0.38 ppm) are 27 times higher than the most contaminated sample observed in Arkansas, and heptachlor epoxide was not found in any of the samples. Though there is a less reliable relationship between DDE and bat mortality, that it was present in all caves indicates that it could have played a role in bat deaths in the past.

Despite the ban on the use of DDT and dieldrin in the 1970s, the pesticide residues found in this study are still commonly detected in surface and groundwater samples in the Ozarks (Adamski and Pugh, 1996; Bell et al., 1996; Adamski, 1997; Bell et al., 1997). Pesticides continue to play

an important role in agriculture and their use is increasing in the study area. Though harvested cropland represents only 5-18% of the land base in Benton, Independence, and Sharp counties, there has been a sharp increase in the use of pesticides in recent years. From 1997 to 2002 there was a 240% increase in land treated with chemicals to control insects, weeds, grass, brush, nematodes, and diseases in crops and orchards in Benton County. Independence County and Sharp County had smaller increases of 6% and 21% over the same period (U.S. Department of Agriculture, 2004).

In conclusion, given the continuing influence of banned pesticides and the increasing use of new compounds in agriculture in the region, periodic monitoring of pesticide concentrations in guano and carcasses of dead bats is recommended.

ACKNOWLEDGMENTS.–I thank Steve Hensley and Carla Mitchell of the U.S. Fish and Wildlife Service for access to Logan Cave National Wildlife Refuge and for their assistance in providing unpublished reports on this subject. I also thank the private landowners who provided access to the other caves that were a part of this study. Rick Clawson of the Missouri Department of Conservation and Donald Clark were extremely helpful in explaining aspects of their extensive work on pesticides and gray bats.

Table 1. Contaminants detected (ppm by dry weight) in gray bat guano in Arkansas, 2004.

CAVE	SAMPLE #	p,p'-DDE	Dieldrin
Bone Cave (Independence Co.)	1	0.028	ND*
	2	0.037	0.012
	3	0.036	ND
	4	0.032	ND
	Average	0.033	n/a
Dodd Cave (Independence Co.)	1	0.015	ND
	2	ND	ND
	3	0.040	0.014
	4	0.020	ND
	Average	0.019	n/a
Logan Cave (Benton Co.)	1	0.057	ND
	2	ND	ND
	3	0.033	ND
	4	0.051	ND
	Average	0.035	n/a
Morris Cave (Sharp Co.)	1	0.015	ND
	2	0.011	ND
	3	ND	ND
	Average	0.009	n/a

***ND = Not Detected**

Table 2. Comparison of three contaminants found in guano by this and other studies.

Study	DDE		Dieldrin		Heptachlor epoxide	
State – Year(s) studied, reference (sample size)	**% colonies in which found**	**Range (ppm) (dry)**	**% colonies in which found**	**Range (ppm) (dry**	**Range colonies in which found**	**% (ppm) (dry)**
Virginia–Historic Ryan et al. 1992 (n = 2)	100	0.08-0.63	100	0.08-0.46	50	0.25
Alabama–1976 Clark et al. 1988 (n = 8)	100	0.122-1.563	75	2.632 - 10	38	2.44 – 6.667
Alabama–1985 Clark et al. 1988 (n = 1)	100	0.5	0	n/a	0	n/a
Missouri–1982 Clawson 1989 (n = 4)	–	–	75	0.31-0.61	25	0.15
Missouri -1988-89 Clawson 1991 (n = 5)	–	–	100	Note 1	100	Note 1
Virginia–1989 Ryan et al. 1992 (n = 3)	67	0.03-0.04	0	n/a	0	n/a
Oklahoma–1990 Martin 1992 (n = 5)	100	0.05-0.14	0	n/a	0	n/a
Arkansas– 2004 This study (n = 4)	100	0.011-0.057	50	0.012- 0.014	0	n/a

Note 1 = Insufficient information is available to transform reported contaminant values to be directly comparable with other studies.

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Deer-Vehicle Collisions in Arkansas

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Between 1990 and 2000, the human population in Arkansas increased 13.7%. The northwest corner of the State had the greatest increase with the population in Fayetteville-Springdale-Rogers metropolitan area growing by 47.5% (Perry and Mackun, 2001). Arkansas also harbors an abundant white-tailed deer (*Odocoileus viginianus*) herd. No reliable statewide estimates of the size of Arkansas's deer population are available; however, legal statewide deer harvests have ranged from 90,910 in 1990 to 194,687 in 1999 (AGFC, 2004). As human populations continue to increase, encroachment into areas populated by deer is inevitable, thus increasing deer-human interactions and conflicts. Arkansas' first attempt to reduce the size of a nuisance urban deer herd via an archery hunt occurred in 2002 at Bull Shoals in Marion County.

In Arkansas, deer-vehicle collisions are a very visible negative consequence of an increasing human population combined with an abundant population of white-tailed deer. Farrell (2003) found that deer-vehicle accident occurrence in Arkansas counties was influenced more by roadway features, level of urbanization, and human population densities than by deer densities or landscape characteristics. However, landscape characteristics in Arkansas were useful in predicting site-specific probabilities of deer-vehicle collisions (Enderle, 2003).

There is no nationwide data clearinghouse for reporting deer-vehicle collision information. However, several studies have reported information, or estimated the effects, of deer-vehicle collisions. It is estimated that nationwide, at least 1.5 million deer-vehicle collisions occur annually (Conover et al., 1995). These accidents result in about $1.1 billion of damage to vehicles ($\bar{x}$ = $1,577/vehicle) and at least $200 million in loss of life or injury (Conover et al., 1995). Human injury rates have been reported at 4% (Conover et al., 1995; Hansen, 1983) and death rates at 0.03% (Conover et al., 1995). Peaks in deer-vehicle collisions typically occur late in the evening, at night, and in the early morning. Seasonally, they peak in the fall with a smaller peak in the spring (Allen and McCullough, 1976; Carbaugh et al., 1975).

In Arkansas, records on deer-vehicle collisions are not readily available or do not exist. Vehicle accident reports filed with the Arkansas State Police are the most extensive and reliable source of information on deer-vehicle collisions available in Arkansas, and thus, these reports were used to provide the following descriptive statistics on deer-vehicle collisions in the state.

Vehicle accident reports involving deer from 1998-2001 were obtained from the Arkansas State Police. These reports were of accidents that occurred on state and federal highways and were of a serious enough nature to require a response from the state police. Thus, these accidents were not representative of all deer-vehicle collisions in Arkansas. Information was not available on accidents that occurred on roads maintained by a county or municipality or on any accident that was not reported. While it is unknown what percentage of deer-vehicle collisions the vehicle accident reports represented, the information that was available likely represented deer-vehicle collisions that were the most serious and costly.

Accident reports were available only as hardcopy reproductions. Every report filed with the Arkansas State Police during 1998-2001 was inspected to identify accidents that involved a deer. Identified reports were photocopied at the Arkansas State Police headquarters in Little Rock, and

Table 1. Summary of deer-vehicle collision information obtained from Arkansas State Police vehicle accident reports.

Parameter	Year				Overall Total	Overall Mean
	1998	1999	2000	2001		
Number of collisions	1,420	1,618	1,248	1,572	5,858	1,465
Number of human injuries	15	7	6	12	40	10
Percent with human injuries	1.1	0.4	0.5	0.8	—	0.7
Number with deer deaths[1]	478	575	419	416	1,888	472
Percent with deer deaths	69.6	69.0	67.4	63.3	—	67.5
Mean damage estimate per vehicle	$1,868	$1,918	$1,975	$1,947	—	$1,926
Total damage estimates per year	$2,554,215	$2,938,773	$2,334,045	$2,945,345	$10,762,278	$2,690,570

[1] Fate of deer was determined in a total of 2,799 deer-vehicle collisions.

then pertinent information was entered into a database. This information included the year, date, time, location, gender of deer involved, fate of deer (death or ran away injured), fate of vehicle occupant(s), and a monetary estimate of vehicle damage.

Annual means were computed for vehicle-damage estimates. Numbers of human injuries and deer deaths were summarized by year and averaged across years. Numbers of collisions were averaged across years by time and month. Proportions of bucks and does involved in collisions were averaged across years by month.

A total of 5,858 vehicle accident reports, averaging 1,465 per year, indicated the occurrence of a deer-vehicle collision (Table 1).

Collisions were recorded in all months, but most (>50%) occurred during October – December with a peak in November (Fig. 1). This time period coincides with white-tailed deer breeding activity in Arkansas which also peaks in November (AGFC, 2004). The number of collisions was greatest between 5:30 p.m. and midnight with a smaller peak occurring between 5:00 – 7:00 a.m. (Fig. 2). These time periods are consistent with diel activity patterns documented for deer in Arkansas (Cartwright, 1975; Pledger, 1975). Most deer (67.5%) were killed as a result of the collisions; 32.5% were injured and fled the collision site. The ultimate fate of these animals is not known. Overall, 48.3% of the collisions were with bucks and 51.7% were with does. However, this proportion varied by month, ranging from 24.1% bucks and 75.9% does in June to 64.7% bucks and 35.3% does in November (Fig. 3). The larger proportion of bucks involved in collisions during October – December coincides with buck rutting activity in Arkansas (AGFC, 2004). Annually, the human injury rate averaged 0.7% with 6 – 12 vehicle occupants being injured per year (Table 1). Estimated damage to individual vehicles ranged from $0 - $20,000. Total estimated damage averaged almost $2.7 million/year with a mean of $1,926 per collision.

Decker et al. (1990) found that only 17-25% of deer-vehicle accidents are reported. If this is the case in Arkansas, then deer-vehicle collisions on state and federal highways may be as great as 9,000 annually. Including accidents that occur on roadways maintained by counties and municipalities could potentially double that number, resulting in an estimated 18,000 deer-vehicle collisions with

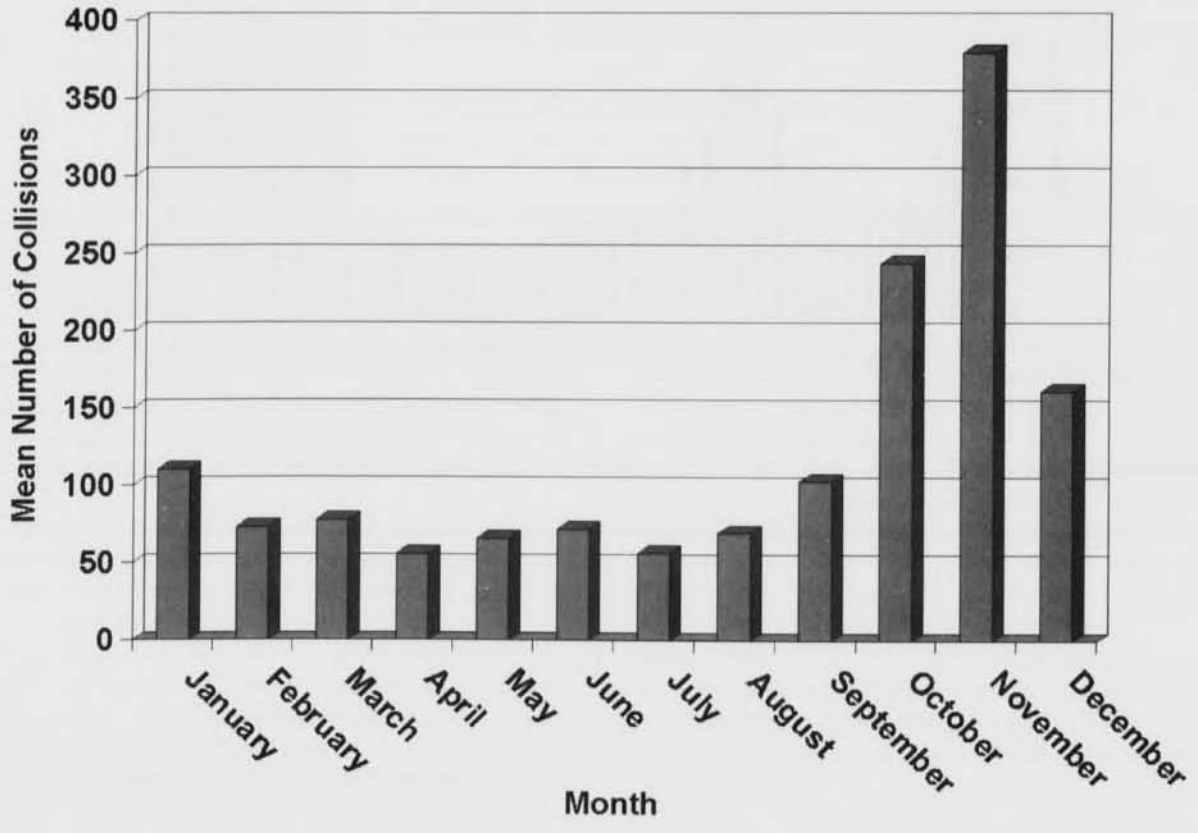

Fig 1. Mean monthly numbers of traffic accident reports filed with the Arkansas State Police during 1998 – 2001 that indicated the occurrence of a deer-vehicle collision.

an estimated loss of almost $35 million in vehicle damage annually. Given the potential economic impact, a statewide collaborative effort involving the Arkansas Game and Fish Commission, Arkansas State Police, Arkansas Department of Transportation, and county and local governments is needed to adequately address the issues surrounding deer-vehicle collisions. In addition to a unified, consistent effort to collect information and institute mitigation measures, educational efforts should be focused at both policy makers and the general public.

ACKNOWLEDGMENTS.–Thanks to Donald Enderle and Chris Farrell, graduate students at the University of Arkansas – Monticello, for their many hours perusing vehicle accident reports and to Susan Enderle for entering thousands of lines of data. Also, thanks to the Arkansas Forest Resources Center for project funding.

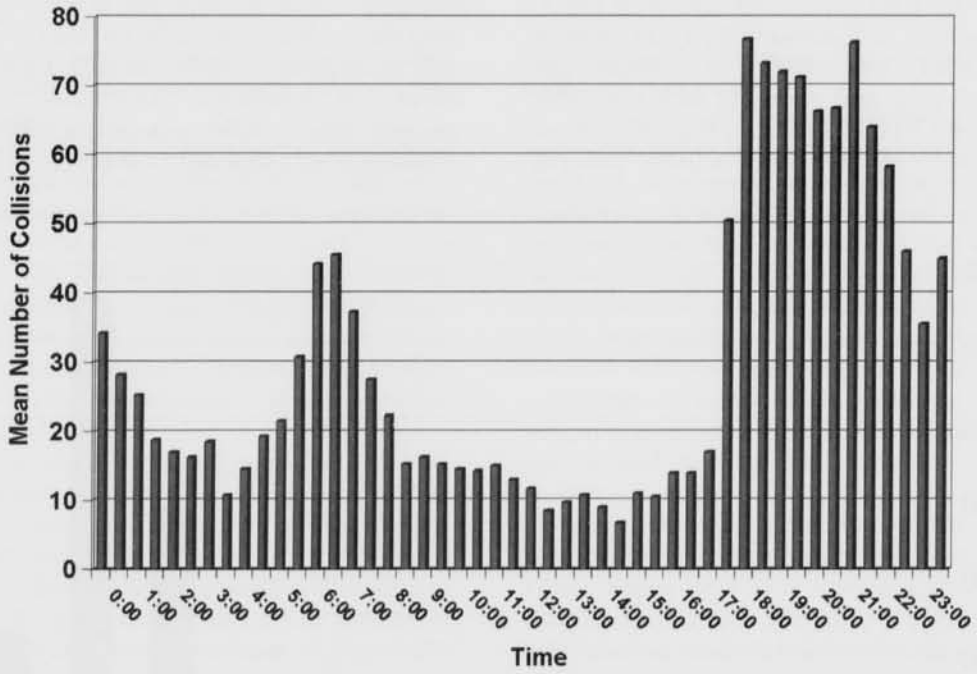

Fig 2. Numbers of deer-vehicle collisons by time of day reported to the Arkansas State Police in a traffic accident report during 1998-2001.

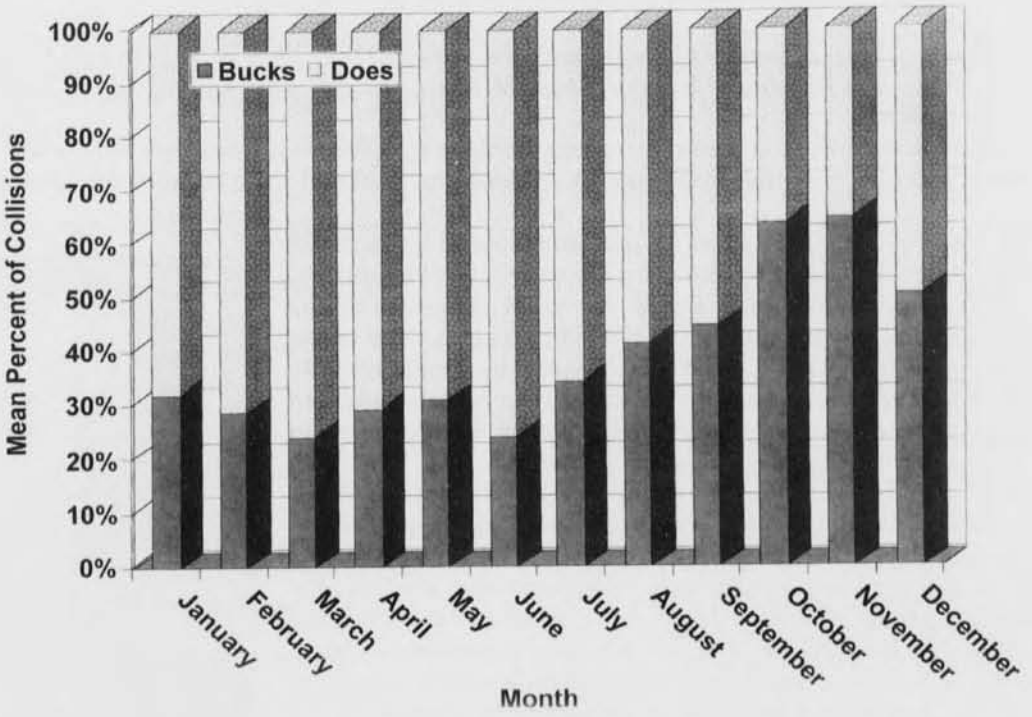

Fig 3. Sex ratios of deer involved in deer-vehicle collisions reported to the Arkansas State Police in a traffic accident report by month during 1998-2001.

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Farrell, MC. 2003. A multivariate analysis of broad-scale factors that contribute to deer-vehicle collisions in Arkansas. Unpublished MS thesis. University of Arkansas, Monticello. 93 p.

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Perry, MJ and **PJ Mackun.** 2001. Population change and distribution, 1990 to 2000. U.S. Department of Commerce, Economics and Statistics Administration, U.S. Census Bureau. Washington, D.C. 7 p.

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In Memoriam

David M. Chittenden, 1936 – 2004

David Morse Chittenden II, 67, of Jonesboro passed away on Friday, Jan. 2, 2004, at St. Bernard's Medical Center. He is survived by his wife, Ruby; two daughters, Jennifer Clack of Jonesboro and Julia Chittenden of Fayetteville; and one granddaughter, Madelyn Clack.

Born on Apr. 7, 1936, in Buffalo, N.Y., David received a bachelor's degree from Rensselaer Polytechnic Institute at Troy, N.Y., in 1958, a master's degree from the University of Arkansas in 1960 and a Ph.D. from the University of Arkansas in 1966.

David moved to Jonesboro in 1967, when he was appointed as an Assistant Professor of Chemistry. After 33 years of service to the Department and University, David retired in 2000. During that time, David served as chairman of the Department of Chemistry and Biochemistry from 1990-96, acting as dean of the College of Arts and Sciences from 1984-85, and was associate dean from 1984-86. David was a devoted teacher and researcher. His versatility as an educator was seen in the variety of classes he taught, from the non-majors Physical Science course to the graduate level Radiochemistry and Geochemistry courses.

David stayed active in research throughout his career. He was an Ames Associate at NASA Ames Research Center in Mountain View, Calif., from 1994-98 and served in the Faculty Research Participation Program at Oak Ridge National Laboratory in 1969. He had numerous publications, including several articles in the Journal of the Arkansas Academy of Science. David was a member of the American Chemical Society, Sigma Xi Research Society, the American Geophysical Union, the Arkansas Academy of Science, Council on Undergraduate Research, serving as ASU liaison in 1994-96, American Association of University Professors and the American Association for the Advancement of Science.

David was committed to the growth of science in the State of Arkansas. Not only did David serve as President of the Arkansas Academy of Science in 1989, he was also involved with the Arkansas EPSCoR committee, the SURF SILO program, and the Arkansas Space Grant Consortium. He also had a passion for the Arts, serving on the board of directors of the Jonesboro Fine Arts Council, was on the Guild of Opera Memphis and was the 2003 president of the Stage One Repertory Theater.

David touched the lives of many people and will be missed deeply. David loved a good pun and was happy to repeat it to any one who would listen. He was a good colleague to those that knew him, and an even better friend.

List of Associate Editors and Reviewers for the Journal of the Arkansas Academy of Science, Volume 59, 2005

The Arkansas Academy of Science gratefully acknowledges the following individuals who served as Associate Editors and outside reviewers of manuscripts for volume 59 of the *Journal* during 2005. The editorial staff extends our heartfelt appreciation for the expertise and assistance provided by our colleagues. Only through your diligent efforts can we continue to produce a high quality publication.

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The Journal of the Arkansas Academy of Science

Revised Publication Policies and Instructions for Prospective Authors

The *JOURNAL OF THE ARKANSAS ACADEMY OF SCIENCE* is published annually. It is the policy of the Arkansas Academy of Science that 1) at least one of the authors of a paper submitted for publication in the *JOURNAL* must be a member of Arkansas Academy of Science, 2) only papers presented at the annual meeting are eligible for publication, and 3) the manuscript is due at the time of presentation. In accordance with this policy, manuscripts submitted for publication should be given to the section chairman at the time the paper is being presented. Correspondence after that time should be directed to Dr. Chris T. McAllister, Managing Editor, *Journal of the Arkansas Academy of Science.*

Each submitted paper should contain results of original research, embody sound principles of scientific investigation, and present data in a concise yet clear manner. SCIENTIFIC STYLE AND FORMAT, The CBE Manual for Authors, Editors, and Publishers Sixth Edition, published by the Style Manual Committee, Council of Biology Editors, is a convenient and widely consulted guide for scientific writers and will be the authority for most style, format, and grammar decisions. Authors should use the active voice for directness and clarity. Special attention should be given to consistency in tense, unambiguous reference of pronouns, and to logically placed modifiers. All prospective authors are strongly encouraged to submit their manuscripts to other qualified persons for a friendly review of clarity, brevity, grammar, and typographical errors before submitting the manuscript to the *JOURNAL.*

Preparation of Manuscript

1. Use Microsoft Word 6.0 or better for preparation of the document.
2. Save figures as tif or jpeg files.
3. Double space the manuscript and all associated text including the Literature Cited on 8. 5 x 11 inch bond paper. SINGLE SPACED MANUSCRIPTS WILL BE REJECTED UNREAD.
4. Use 12 point font in Times New Roman for text.
5. Use one-inch margins.
6. Number pages.
7. Do not submit word-processed copy printed with justified right-hand margins.
8. Set words in italics that are to be printed in italics (e.g., scientific names).
9. Clip, do not staple, pages together.
10. Include a separate title page with authors' names and addresses.
11. Indicate on the title page which author is the correspondence author and include that author's email address, phone number, and fax number.
12. **An abstract** summarizing in concrete terms the methods, findings, and implications discussed in the body of the paper must accompany a feature article. The abstract should be completely self-explanatory.
13. Most **feature articles** should include the following sections: Abstract, Introduction, Materials and Methods, Results, Discussion, Conclusions, Acknowledgments, and Literature Cited. These **section headings** should be centered and in bold.
14. A **feature article** includes approximately 6 or more typewritten pages. A JOURNAL printed page is equal to approximately 3½ typewritten pages, and the author is assessed a **page charge** (see Review Procedure section).
15. Indent paragraphs and subheadings 5 spaces.
16. **Subheadings** should be italicized, in bold, and followed by .–
17. A **general note** is usually 1 to 5 typewritten pages and rarely utilizes subheadings. the first page with the body of the paper following. Abstracts are not used for general notes.
18. The **metric system of measurements** and weights must be employed. **Grams** and **kilograms** are units of **mass** not weight. Standard distance measurements are permitted in parentheses.
19. In scientific text, **Arabic numerals** should be used in preference to words when the number designates anything that can be counted or measured:3 hypotheses, 7 samples, 20 milligrams. However, numerals are not used to begin a sentence; spell out the number, reword the sentence, or join it to a previous sentence. Also, 2 numeric expressions should not be placed next to each other in a sentence. The pronoun "one" is always spelled out.
20. **Tables and figures** (line drawings, graphs, or black and white photographs) should not repeat data contained in the text. Tables and figures must be numbered and have short legends. Author(s) must place reference to each to them in the text. Tables should immediately follow the Literature Cited. Legends for figures should be typed on a separate page, which should follow the tables and precede the figures. **Do not run tables and figures in the text.** Illustrations must be of sufficient size and clarity to permit reduction to standard page (or ½ page) size; ordinarily they should be no larger than twice the size of intended reduction and no larger than a manuscript page for ease of handling. **Photographs** must be printed on glossy paper. Sharp focus and high contrast are essential for good reproduction. Figures and labeling must be of professional quality. Figure number, author's name, and top of figure must be written in pencil on the back of each figure. Tables must be of professional quality when submitted. Indicate preferred placement of figures and tables in the margins of the manuscript. Do not submit original artwork, photos, tables, or figures with the review copies of the manuscript.
21. **Literature Cited:** Authors should use the Name–Year format as illustrated in *The CBE Manual for Authors, Editors, and Publishers* and as shown below. The *JOURNAL* will deviate from the form given in the *CBE Manual* only in regard to placement of authors' initials and abbreviation of journal titles. Initials for second and following authors will continue to be placed before the author's surname. **Journal titles will no longer be abbreviated.** The general formats for a journal article and a book are shown below along with examples. Note that authors' names are in bold, double spacing occurs after periods, and second and following lines are indented 5 spaces. Do not cite abstracts and oral, unpublished presentations.

Author(s). Year. Article title. Journal title volume number (issue number):inclusive pages.

Author(s) [or editor(s)]. Year. Title of Book. Place of publication: publisher name. Number of pages.

<u>Standard Journal Article</u>

Davis, DH. 1993. Rhythmic activity in the short-tailed vole, *Microtus.* Journal of Animal Ecology 2:232-8.

Form of Citation: (Davis, 1993)

Steiner, U, JE Klein, and **LJ Fetters.** 1992. Complete wetting from polymer mixtures. Science 258:1122-9.

Form of Citation: (Steiner et al., 1992)

Zheng, YF and **JYS Luh.** 1989. Optimal load distribution for two industrial robots handling a single object. ASME Journal of Dynamic System, Measurement, and Control 111:232-237.

Form of Citation: (Zheng and Luh, 1989)

<u>Electronic Journal Articles and Electronic Books</u> should be cited as standard journal articles and books except add an availability statement and date of accession following the page(s).

. . . 653 p. Available at: <u>www.usfw.gov/ozarkstreams</u>. Accessed 2004 Nov 29.

<u>Books, Pamphlets, and Brochures</u>

Box, GEP, WG Hunter, and **JS Hunter.** 1978. Statistics for experimenters. New York: J Wiley. 653 p.

Form of Citation: (Box et al., 1978)

Gilman, AG, TW Rall, AS Nies, and **P Taylor,** editors. 1990. The pharmacological basis of therapeutics. 8th ed. New York: Pergamon. 1811 p.

Form of Citation: (Gilman et al., 1990)

Engelberger, JF. 1989. Robotics in Service. Cambridge (MA): MIT Press. 65 p.

Form of Citation: (Engelberger, 1989)

Book Chapter or Other Part with Separate Title but Same Author(s) – General format is given first.

Author(s) or editor(s). Year. Title of book. Place of publication: publisher's name. Kind of part and its numeration, title of part; pages of part.

Hebel, R and **MW Stromberg.** 1987. Anatomy of the laboratory cat. Baltiimore: Williams & Wilkins. Part D, Nervous system; p 55-65.

Singleton, S and **BC Bennett.** 1997. Handbook of microbiology. 2nd ed. Emmaus (PA): Rodale. Chapter 5, Engineering plasmids; p. 285-296.

Book Chapter or Other Part with Different Authors–General format is given first.

Author(s) of the part. Year. Title of the part. In: author(s) or editor(s) of the book. Title of the book. Place of publication: publisher. Pages of the part.

Weins, JA. 1996. Wildlife in patchy environments: Metapopulations, mosaics, and management. In: McCullough DR, editor. Metapopulations and wildlife conservation. Washington, DC: Island Press. p. 506.

Johnson, RC and **RL Smith.** 1985. Evaluation of techniques for assessment of mammal populations in Wisconsin. *In:* Scott Jr NJ, editor. Mammal communities. 2nd ed. New York: Pergamon. p. 122-130.

Dissertations and Theses–General format is given first

Author. Date of degree. Title [type of publication–dissertation or thesis]. Place of institution: name of institution granting the degree. Total number of pages. Availability statement.

The availability statement includes information about where the document can be found or borrowed if the source is not the institution's own library.

Millett, PC. 2003. Computer modeling of the tornado-structure interaction: Investigation of structural loading on a cubic building [MS thesis]. Fayetteville (AR): University of Arkansas. 176 p. Available from: University of Arkansas Microfilms, Little Rock, AR; AAD74-23.

Stevens, WB. 2004. An ecotoxicological analysis of stream water in Arkansas [dissertation]. State University (AR): Arkansas State University. 159 p.

Scientific and Technical Reports–General format is given first.

Author(s) (Performing organization). Date of publication. Title. Type report and dates of work. Place of publication: publisher or sponsoring organization. Report number. Contract number. Total number of pages. Availability statement if different from publisher or sponsoring organization. (Availability statement may be an internet address for government documents.)

Harris, JL and ME Gordon (Department of Biological Sciences, University of Mississippi, Oxford MS). 1988. Status survey of Lampsilis powelli (Lea, 1852). Final report 1 Aug 86–31 Dec 87. Jackson (MS): US Fish and Wildlife Service, Office of Endangered Species. Report nr USFW-OES-88-0228. Contract nr USFW-86-0228. 44+ p.

[USGS] US Geological Survey. 1979. Drainage areas of streams in Arkansas in the Ouachita River Basin. Open file report. Little Rock (AR): USGS. 87 p. Available from: www.usgs.gov/ouachita

Form of citation: (USGS, 1979)

Published Conference Proceedings – General format is given first.

Editor(s). Date of publication. Title of publication or conference. Name of conference (if not given in the 2nd element); inclusive dates of the conference; place of the conference. Place of publication: publisher. Total number of pages.

Vivian, VL, editor. 1995. Symposium on Nonhuman Primate Models for AIDS; 1994 June 10-15; San Diego, CA. Sacramento (CA): Grune & Stratton. 216 p.

Grammar and Usage –

The following is not comprehensive, but highlights common mistakes.

Numbers, units and symbols. Use digits for numbers unless a number is the first word of a sentence, or it is used as a pronoun (e.g., at least one was captured), in which case the number is spelled out. Avoid using introductory phrases such as "A total of . . .". Spell out ordinal numbers (e.g., first, fifth) in text, but use digits for adjectives such as 2-fold and 3-way.

Hyphenate number-unit phrases used as adjectives (e.g., 8-m^2 plots, 1-year-old-males) but not those used as predicate adjectives (e.g., the plots were 8 m^2). Do not insert a comma or hyphen between consecutive, separate numbers in a phrase (e.g., 25 2-m^2 plots). Do not use naked decimals (i.e., use 0.05, not .05).

Italicize Roman letters in the text used as symbols for statistics, tests, or variables. Insert symbols from your word processing program's symbols directory as opposed to creating the symbol with keyboard functions. Insert a space on either side of symbols when used in an equation (e.g., $n = 12$, $P = 0.002$), but not when used as "adjectives" (e.g., >20 observations).

Dates and years. Date sequence is day-month-year without punctuation (e.g., 4 Feb 1947). Spell out months, except in parentheses and table and figure bodies, where 3-letter abbreviations are used without a period. Do not use an apostrophe when referring to an entire decade (i.e., 1940s, not 1940's).

Punctuation. Commas.

1. Use a comma before the conjunction in a serial list of >2 items (e.g., red, black, and blue). Do not use a comma to separate 2 items in a series.
2. Use a comma to set off an introductory clause beginning with a subordinating conjunction (if, although, because, since, when, where, while).
3. Use a comma to set off a transitional or parenthetic word or phrase (to be sure, of course, after all, finally).
4. Use a comma to separate a nonrestriction clause or appositive from the rest of the sentence. Nonrestrictive clauses usually begin with "which." They provide additional information but are not necessary to understand the sentence (e.g., These fish, which were found in a cave, are blind and depigmented.) Commas do not separate restrictive clauses from the rest of the sentence. Restrictive clauses usually begin with "that" and are necessary for the meaning of the sentence (e.g., Fish that live in caves are usually blind and depigmented.)
5. Use a comma to separate different elements of an address or geographic designation (e.g., The frogs were collected in Conway County, Arkansas, on February 21.)

Unnecessary and Incorrect Uses of Commas

1. Do not use a comma to separate a compound sentence before the conjunction unless the sentence will be confusing otherwise (e.g., "Use an infrared scope at night and use a regular scope during the

day," not "Use an infrared scope at night, and use a regular scope during the day.").
2. Do not use a comma to set off a short introductory phrase or clause of the comma would not contribute to clarity or ease of reading.
3. Do not use a comma to set off a restrictive appositive (a defining word or phrase needed for the desired meaning). The species *Pseudacris streckeri* is a small burrowing frog.
4. Do not use commas to separate prepositional phrases, even those beginning with "with."
5. Do not separate a compound predicate with a comma. We captured 46 bats and tagged 38 of them.
6. Do not use a comma to separate name modifiers from the stem name. Franklin D Roosevelt Jr [not "Franklin D. Roosevelt, Jr."] Note the absence of periods also.

Hyphen

1. Do not hyphenate prefixes, suffixes, or combining forms (e.g., post partum) unless necessary to avoid misreading.
2. Hyphenate compounds used as adjectives (e.g., 1-m plot, 2-day period, 14-cm dbh).
3. Although the rules for hyphenation are complex, there are a few basic principles:
   a. a phrase containing a participle or an adjective is hyphenated as a compound when it precedes the word modified (e.g., home-range estimation) and is written without a hyphen when it follows the word modified (estimation of home range);
   b. a modifier containing a number is usually hyphenated (e.g., 3-month-old fawn); and
   c. a 2-word modifier containing an adverb ending in -ly is not hyphenated (e.g., publicly owned land).

Colon

1. A colon can only follow a complete independent clause.
2. A colon may be used to separate two independent clauses where the second clause amplifies or clarifies the first.
3. A colon may be used to introduce a list. We used 3 morphological measures in our analysis: snout-vent length, tibia length, and mass.
4. A colon should **not** be used after a title, text heading or subheading, equation, or formula standing separate from text.
5. A colon **may not** split an infinitive. The objectives of the study were to determine population heterozygosity, compare frequency of specific alleles in different populations, and estimate size of evolutionary units. (not "The objectives of the study were to: determine population . . .")
6. A colon **may not** separate a verb and its object. The 3 proteins studied were actin, keratin, and myosin. (not "The 3 proteins studied were: actin, keratin, and myosin.")

Possessives

The general principle of adding an apostrophe and "s" holds for most nouns, including proper nouns, that end in "s." Pronunciation can serve as a guide: if one would pronounce the possessive "s," it should appear in the written form.

the grass's texture (but better "the texture of the grass")
Williams's work on the topic
Charles's suggestion
Arkansas's lakes and mountains
Agassiz's theories on glaciation
Descartes's esssays

But
Archimedes' screw
Hippocrates' teachings
Rameses' tomb

Review Procedure

Evaluation of a paper submitted to the *JOURNAL* begins with a critical reading by the Managing Editor. The paper is then submitted to referees for checking of scientific content, originality, and clarity of presentation. Attention to the preceeding paragraphs will greatly speed up this process. Judgments as to the acceptability of the paper and suggestions for strengthening it are sent to the author. If the paper is tentatively accepted, the author will rework it, where necessary, and return two copies of the revised manuscript together with the original to the Managing Editor. Usually a time limit for this revision will be requested. If the time limit is not met, the paper may be considered to be withdrawn by the author and rejected for publication. All final decisions concerning the acceptance or rejection of a manuscript are made by the Managing Editor and/or Editor-in-Chief.

When a copy of the proof, original manuscript, and reprint order blanks reach the author, they should be carefully read for errors and omissions. The author should mark corrections on the proof and return both the proof and manuscript to the Managing Editor within 48 hours or the proof will be judged correct. Printing charges accruing from excessive additions to or changes in the proofs must be assumed by the author. Reprint charges are placed with the printer, not the Managing Editor. Page changes are $50 printed page. These changes and excessive printing charges will be billed to the author by the Academy of Science ($4.00 per word). A page charge will be billed to the author of errata.

ABSTRACT COVERAGE

Each issue of the *JOURNAL* is sent to several abstracting and review services. The following is a partial list of this coverage.

Abstracts in Anthropology
Abstracts of North America Geology
Biological Abstracts
Chemical Abstracts
Mathematical Reviews
Recent Literature of the Journal of Mammalogy
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Sport Fishery Abstracts
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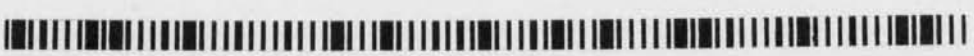

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