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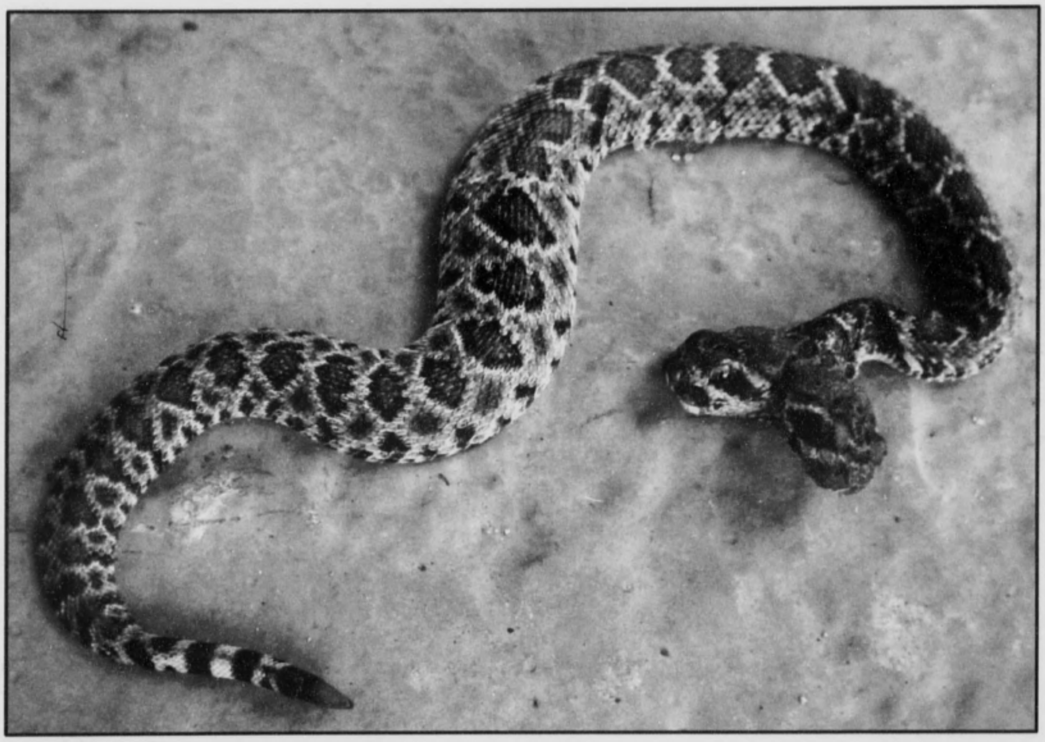

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COVER: Dicephalic western diamondback rattlesnake (*Crotalus atrox*). Photo by Chris T. McAllister.

ARKANSAS ACADEMY OF SCIENCE 2006

APRIL 7-8, 2006

90TH ANNUAL MEETING

Lyon College
Batesville, Arkansas

JOURNAL
ARKANSAS ACADEMY OF SCIENCE

Annual Meeting 7-8 April 2006
Lyon College

Stanley E. Trauth President	**David Saugey** President-Elect	**Jeff Robertson** Secretary
Joyce Hardin Treasurer	**Mostafa Hemmati** NAAS Delegate	**Henry Robison** Historian

Secretary's Report
MINUTES OF THE 90TH MEETING

ARKANSAS ACADEMY OF SCIENCE
SUMMARY OF 1ST AND 2ND BUSINESS MEETINGS
LYON COLLEGE
APRIL 7-8, 2006

1. With a crisp single BANG of the ceremonial gavel, Stan Trauth, President of the AAS called the meetings to order.

2. Local Arrangements Committee: Walter Roettger, President of Lyon College thanked the work of the Local Arrangements Chair Kurt Grafton and expressed his delight at the keynote speaker, Guy Consolmagno. Kurt Grafton then expressed his 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. Also presenting will be the winner of the Jr. Academy. He 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. Historian: Henry Robison reported that this is the 90th annual meeting of the Arkansas Academy of Science. This is the very 1st time that the annual meeting has been held at the Lyon College Campus.

4. Secretary: The minutes from 2005 Executive Committee business meeting in November 2005 were distributed and approved. These minutes will be going on the AAS website. Prior to this meeting, the current membership list included approximately 120 members (56 which are life members) of the Academy along with 14 institutional members. A request for $250 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. Treasurer/Auditor: The financial report was presented by Joyce Hardin. Page charges for Vol 59 were all e-mailed this year to save trees. Details on the income and expenses for the year were highlighted in addition to journal cost issues. The Academy posted a substantial gain of $7,000, a record this year. Mostafa Hemmati and Jeff Robertson were acting auditors and reviewed the financial statements. The "books" maintained by the Academy Treasurer were found to be good financial records kept with excellent integrity and showing no inconsistencies or irregularities. (The financial status of the Academy is found elsewhere in this volume for review).

6. *Journal* Editor-in-Chief: Stan Trauth reported that Vol. 59, 2005 of the journal is 223 pages. This is 79 more pages than last year's journal. 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 reported that the journal contained 29 papers, 8 notes, 21 features with 1 rejection. He noted that the peer review process for the journal was quite vigorous as can be attested to by the extensive list of reviewers, their locales and disciplines. Revised publication policies for the journal will be posted on the AAS website. He also requested $750 to cover incurred costs associated with managing editor duties and was approved.

8. Arkansas Science Fair Association: The state science fair is in its 52nd year and the annual winners go to an international fair that has 1000 students from 40 countries competing. The Association is looking for a replacement for the retiring director Micheael Rapp. A request and approval was obtained for $400 dollars to support the Arkansas Science Fair Association.

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: Will Slaton has replaced the outgoing Jim Murray as director for the talent search as this organization reorganizes.

11. Junior Science and Humanities Symposium: Linda Kondrick presented highlights from the 40th annual JSHS symposium and the 29th year hosted on the Arkansas Tech University campus (http://pls.atu.edu/jshs). Support from the military sponsors and the AAS help make the 3-day event and research paper presentations by students successful. Six students represent Arkansas in the national competition. A request and approval for continued support of $100 towards 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 on a lively discussion over the current debate on evolution in the classroom.
    c. Development Committee: Betty Crump related woes from an unsuccessful grant application to endow the journal but solicited other, "sexier" funding propositions to attract sponsors and hopefully produce successful grant applications.
    d. AAAS: Mostafa Hemmati reported that 4 students benefited from memberships to the AAAS and that due to snow and ice his attendance at the national meeting was cancelled.
    e. Arkansas Science Teachers Association: Tillman Kennon thanked the Academy for their continued support of the Science fair and the Jr. Academy. ASTA is hosting workshops for teachers in science lab safety and providing training to 7-12 grade teachers. They are also developing science benchmark tests for 5th and 7th grades.

13. New Business:

    a. The JAAS Editor-in-Chief and managing editor are retiring! It is paramount for the Academy to find replacements for these critical positions soon. The time to think is now about what the journal needs are going to be and any changes necessary about how the publication of the journal is handled.
    b. The nominations committee announced candidates they solicited for positions of Vice President and Treasurer. As there were no additional nominations from the floor, Joyce Hardin and Mostafa Hemmati were voted in by acclamation for V.P. and Treasurer, respectively.
    c. The development committee solicited input for ideas to fund scholarships, awards, the journal, etc, including but not limited to: better advertising of science education initiatives and programs by the AAS, AAS membership benefits promotion, corporate members and/or sponsors, "friends" of the Academy (philanthropic).
    d. Motion passed for the creation of a new poster award of $100 each for graduates and undergraduates as this presentation style has become more numerous and popular at the meeting.
    e. Motion passed for the Academy to adopt the position statement of our parent organization (AAAS) on intelligent design and a link to it and other information from the AAAS on our AAS website.
    f. Jamie Oliver of the Arkansas Environmental Federation who sponsors our student awards to the tune of $500 spoke about the participation of students at their convention and the availability of awards and scholarships.
    g. Desires were expressed by members to have the Academy:

        i. develop close contacts at each institution and promote the Academy. Department Chairs and Deans from science programs at each school should be educated and informed more about the Academy and the Journal,
        ii. develop a strategy to have the old journals scanned electronically and available to the members online,
        iii. find a way to reinstate journal distribution to student members,
        iv. think about establishing a permanent, recognizable web address (e.g. www.arkansasacademyofscience.org).

    h. Jeff Robertson from Arkansas Tech University announced the dates of the 91st annual meeting as April 13-14, 2007 on the ATU campus in Russellville, Arkansas. Locations for 2008 and beyond are solicited.
    i. Resolutions for the annual meeting were read to the membership (see Resolutions).

14. Closing: New president David Saugey accepted the ceremonial gavel from outgoing president Stan Trauth making him Past-President, Collis Geren President-Elect and Joyce Hardin Vice-President.

Meeting adjourned.

Jeff Robertson, AAS Secretary

ARKANSAS ACADEMY OF SCIENCE
2006 FINANCIAL STATEMENT

BALANCE - January 4, 2007	**$36,896.76**
BALANCE – January 17, 2006	**$36,174.06**
NET GAIN	**$722.70**

DISTRIBUTION OF FUNDS

Checking Account	**$8,610.08**
Bank of Ozarks, Russellville, AR	
Certificate of Deposit	**$13,000.00**
Life Membership Endowment	
Bank of Ozarks, Russellville, AR	
Dwight Moore Endowment	**$5,286.68**
Bank of Ozarks, Russellville, AR	
Phoebe and George Harp Endowment	**$5,959.51**
Bank of Ozarks, Russellville, AR	
Unrestricted	**$4,040.49**
Bank of Ozarks, Russellville, AR	
TOTAL	**$36,896.76**

INCOME:

1. ANNUAL MEETING		
a. Check from Lyon College	$2,981.44	
		$2,981.44
2. INTEREST		**$366.22**
3. JOURNAL		
a. Miscellaneous Sales	$247.17	
b. Page Charges	$11,350.25	
c. Subscriptions	$950.00	
		$12,547.42
4. JOURNAL CONTRIBUTION		**$3,200.00**
5. MEMBERSHIP		
a. Individual	$620.00	
b. Institutional	$660.00	
c. Life	$100.00	
		$1,380.00
6. MISCELLANEOUS INCOME		
a. Stan Trauth	$35.00	
b. Betty Crump	$35.00	
		$70.00
TOTAL INCOME		**$20,545.08**

EXPENSES:

1. STUDENT AWARDS		
a. Katie McLean	$100	
b. J.C. Wagnon	$50	
c. J.C. Douglas	$50	
d. Toby Ward	$100	
e. Laura McWilliams	$100	
f. Chris Wyatt	$50	
g. Mathew Lemay	$50	
h. Shara Jones	$50	
i. Marcy Bonitts	$50	
J. Bonnie Earleywine	$100	
k. Marcy Bonwits	$100	
l. Scott Jordan	$50	
m. Rex Medlin	$50	
n. Taldi Walter	$50	
		$950.00
2. AWARDS		
a. Junior Science and Humanities Sym.	$100	
b. Arkansas Science Fair	$400	
c. Arkansas Junior Academy of Science	$250	
		$750.00
3. JOURNAL		
a. Journal Expenses – Stan Trauth	$200	
b. Journal Expenses – Chris McAllister	$750	
c. Journal Charges (226 Pages + Cover)	$16,239.04	
d. Journal Expenses – Joy Trauth	$600	
e. Journal Expenses – Betty's stamps	$46.80	
		$17,835.84
4. MISCELLANOUS EXPENSES		
a. Jeff Robertson – Office Expenses	$62.37	
b. David Saugey	$182.12	
c. Bank Check Charges	$13.35	
d. Dues to the NAAS	$115.40	
		$373.24
TOTAL EXPENSES		**$19,909.08**

APPENDIX A

2006 AAS Award Winners

ORAL PRESENTATIONS

GRADUATE STUDENT AWARDS

Environmental Science

1st Place Joseph R. Milanovich / ASU
Effects of Prescribed Burning on Reproduction of the Western Slimy Salamander (*Plethodon albagula*) in Arkansas.

2nd Place Taldi Walter / UCA
The Role of Biology and Culture in Species Invasions: Contrasting Evidence From Two Invaded Ranges.

Life Science

1st Place Toby M. Ward / UAMS
Genetic Strategies for Improvement of Varicella Vaccine.

2nd Place Rex E. Medlin, Jr. / ASU
Distribution of Arkansas's Bottomland Bats.

3rd Place Jacy L. Wagnon / UAMS
Trans-Activation of Meiotic Recombination by mRNA.

UNDERGRADUATE STUDENT AWARDS

Environmental Science

1st Place Bonnie Earleywine / UCA
Population Status and Distribution of Spring Cavefish, Forbesichthys Agassizi, in Southeast Missouri.

2nd Place Shara Jones / HSU
Differences in Coral Community Structure Reflect Impacts of Indigenous Villages and Agriculture at 9 Reef Sites in Kuna Yala, Panama.

3rd Place Marcy Bonewits / UCA
Henderson Middle School Environmental Education Outreach.

Life Science

1st Place Katie McLean / Lyon
Developing a *C. Elegans*-Based Bioassay for Estrogenic Activity.

2nd Place J. C. Douglas / Lyon
A Mutation in Estrogen Receptor Alpha that Selectively Attenuates Activation by Phytoestrogens.

3rd Place Scott Jordan / HSU
Ichthyofaunal Assemblages in Three Proximate But Ecologically Diverse Streams in Clark County, Arkansas.

Physical Science

1st Place Laura McWilliams / Lyon
Response Surface Optimization of a Microwave Assisted Perkin Reaction.

2nd Place Chris A. Wyatt / UALR
When You Can't Take a Dust Mop to Space: A Solution to the Mars Rover Particle Adhesion Problem.

3rd Place Matthew Lemay / ASU
Investigation of Electromagnetic Shielding in Ferromagnetic Nano/Micro Powder/Polymer Composite Films.

The David M. Chittenden II
Undergraduate Green Chemistry Poster Award

Amon Holt
Lyon College
for
Ruthenium Complexes of Novel Thiosemicarbazones:
Synthesis, Characterization, and Antimicrobial Activity

APPENDIX B

RESOLUTIONS

BE IT RESOLVED that we, the membership of the Arkansas Academy of Science, offer our sincere appreciation to Lyon College for hosting the 90th annual Meeting of the Academy, held 7-8 April, 2006.

We thank our Local Arrangements Committee: Chair Dr. A. Kurt Grafton, Drs. Floyd Beckford, Ray Byler, Barry Gehm, Bob Gregerson, Stuart Hutton, David Koch, Tim Lindblom, David Pace, David Sonnier, Dave Thomas, and all of the student workers and staff who collectively contributed to such a successful meeting.

Appreciation is expressed for inviting us to this gorgeous campus and for use of these excellent facilities and the hospitality shown to us by Lyon personnel. We especially thank our keynote speaker Dr. Guy Consolmagno for his thought provoking presentation entitled, "Pluto and Planets X: Is Pluto a Planet? And Why Does It Matter?"

We thank Lyon College for their contributions to the Social and Banquet, which were both excellent and thoroughly enjoyed by all. And we thank Provost Vice President for Academic Affairs and Dean of the Faculty, Dr. John Peek. We sincerely appreciated and enjoyed the fine music provided by the Lyon College Pipe Band.

The Academy recognizes the important roles assumed by session chairs and expresses sincere appreciation to Mostafa Hemmati, James Engman, Wayne Wahls, Gus Williamson, Abul Kazi, Betty Crump, Scott Kirkconnell, Grover Miller, Steve Zimmer, Thomas Smith, Bill Shepard, Dave Thomas, Stuart Hutton, Brian Wagner, and Tim Lindblom.

A special appreciation is owed to those individuals who devoted considerable time and energy to judging student papers. They are Ginny Adams, Betty Crump, Steve Dinkelacker, James Kellum, Anwar A Bhuiyan, Mostafa Hemmati, Abul B. Kazi, Larry Lebofsky, William Slaton, Ying Hutton, David Thomas, Grover Paul Miller, Thomas Smith, Scott W. Kirkconnell, Deborly Wade, Bob Gregerson, and Tim Lindblom.

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: Stan Trauth (President), David Saugey President-Elect), Collis Geren (Vice President) and Betty Crump (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 8th day of April, 2006
Resolution Committee
David Saugey, Chair
Joyce Hardin
Mostafa Hemmati

2006 - 2007 MEMBERSHIP

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Phoebe	Harp	Arkansas State University
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Ronnie	Helms	
Mostafa	Hemmati	Arkansas Tech University
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Russellville, AR 72801

$100
Dr. Scott Kirkconnell
Department of Biology
Arkansas Tech University
Russellville, AR 72801

$50
Dr. William Shepherd
2805 Linden, Apt. 3
Little Rock, AR 72205

$100
Dr. James Wickliff
University of Arkansas-Fayetteville
1215 Columbus Blvd.
Fayetteville, AR 72701

$100
Dr. Arthur Fry
University of Arkansas-Fayetteville
1923 Joyce Street #323
Fayetteville, AR 72703

$200
Dr. Rudolph Eichenberger
Southern Arkansas University
Box 9176
Magnolia, AR 71754

$100
Dr. Stanley Trauth
Department of Biological Sciences
Arkansas State University
P.O. Box 599
State University, AR 72467

$100
Dr. George Templeton
University of Arkansas Fayetteville
PS 217
Fayetteville, AR 72701

$50
Mr. Brian Lockhart
USDA Forest Service
PO Box 227
Stoneville, MS 38776

$100
Mrs. Betty Crump
USDA Forest Service
P.O. Box 1270
Hot Springs, AR 71902

$900
Anonymous
USDA Forest Service
P.O. Box 1270
Hot Springs, AR 71902

$100
Dr. Henry Robison
2714 Chaffin LN.
Magnolia, AR 71753

$100
Dr. Wayne Gray
215 Colonial Ct
Little Rock, AR 72205

$100
Dr. Roger E. Koeppe
305 N. Washington
Fayetteville, AR 72701

$50
Dr. Barry Dean Gehm
3425 Juniper
Batesville, AR 72501

$1000
Dr. Douglas James
University of Arkansas
Department of Biological Science
Fayetteville, AR 72701

$25
Dr. Michael Looper
3214 Excelsior RD.
Greenwood, AR 72936

Arkansas Academy of Science
90th Annual Meeting
April 7-8, 2006

Welcome

I welcome all of you to the 90th annual meeting of the Arkansas Academy of Science. This year, we are very pleased to be meeting on the Lyon College campus for the very first time. On behalf of the AAS Executive Committee, I also thank all of you for your attendance, participation, and involvement in the AAS as well as the Arkansas Junior Academy of Science. We are looking forward to many diverse presentations during the next two days. This time will go quickly, but I hope you will have the opportunity to meet new acquaintances, visit with colleagues, and renew old friendships as we share and disseminate scientific knowledge. On behalf of the entire AAS membership, I want to extend a deep appreciation to Local Arrangements Committee and Lyon College for hosting this year's annual meeting.

Stanley E. Trauth
President, Arkansas Academy of Science

On behalf of Lyon College and the Local Arrangements Committee, I welcome you to our campus and the 90th Annual Meeting of the Arkansas Academy of Science. This is the first time, but hopefully not the last, that Lyon (once known as Arkansas College) has hosted the AAS. This year, we are pleased to be hosting over 100 presentations and posters from a wide variety of scientific disciplines. We hope you enjoy the meeting and your time on our campus as much as we've enjoyed planning and preparing for your visit. To help you find your way around Batesville and Lyon College, you'll find building, campus, and area maps in the back of this program. If you have any other questions or need any assistance during the meeting, just look for any member of the Local Arrangements Committee; we'll all have red ribbons on our name badges.

Anthony K. Grafton
Chair, Local Arrangements Committee

Keynote Speaker

PLUTO AND PLANETS X: IS PLUTO A PLANET? AND WHY DOES IT MATTER?

Guy J. Consolmagno, Vatican Observatory, V-00120 Città del Vaticano, Italy

Less than 15 years ago, the first "trans-Neptunian Object" besides Pluto was discovered. Within the past two years, a rush of new discoveries have revealed a number of bodies whose size rivals that of Pluto. What are these objects? Where did they come from, and what can they tell us about the origin and evolution of the solar system? How are these objects found, and why they only being discovered now? Finally, are they indeed new planets? Who gets to say, and how is this determined? Observing these objects, and observing their observers, is a revealing story in both the science and politics of planetary astronomy.

Dr. Guy Consolmagno, S.J., is a Jesuit monk, and an astronomer at the Vatican Observatory whose research includes observing the colors of trans-Neptunian Objects at the Vatican Observatory's Advanced Technology Telescope. He is also president of the International Astronomical Union Commission 16, Planets and Satellites, and a member of the IAU Working Group on Defining a Planet. Dr. Consolmagno also serves as curator of the Vatican Meteorite collection, one of the largest in the world. His research explores the connections between meteorites and asteroids, and the origin and evolution of small bodies in the solar system. Dr. Consolmagno divides his time between Tucson, Arizona, where he observes asteroids and Kuiper Belt comets with the Vatican's 1.8 meter telescope on Mt. Graham, and Castel Gandolfo, Italy, home of the Vatican meteorites. He has also traveled extensively around the world doing research and giving talks. A short biography (from which this was excerpted) is available from the Vatican Observatory facility in Arizona (http://clavius.as.arizona.edu/vo/R1024/GConsolmagno.html). Image courtesy of Alessia Giuliani.

PROGRAM

Arkansas Academy of Science
90th Annual Meeting
April 7-8, 2006
Lyon College

SCHEDULE OF EVENTS

Friday, April 7
- 8:00 a.m. - 4:00 p.m. Registration. Derby Center, 1st floor lobby
- 8:00 a.m. - 11:45 a.m. Workshops Derby Center (locations below)
- 10:00 a.m. - 11:30 a.m. AAS Executive Meeting Lyon Building, Worthen Room
- 12:00 p.m. - 2:00 p.m. Poster Setup Alphin Building, Kresge Gallery
- 12:15 p.m. - 12:45 p.m. Judges Meeting Derby Center, Room 007
- 1:00 p.m. - 2:30 p.m. Oral Presentations (Session 1) Derby Center, Rooms 009, 011, 016, 023
- 2:30 p.m. - 3:15 p.m. Poster Session, Alphin Building, Kresge Gallery
- 3:30 p.m. - 5:00 p.m. Oral Presentations (Session 2) Derby Center, Rooms 009, 011, 016, 023
- 4:00 p.m. - 5:00 p.m. AAS Science Education Committee Meeting Lyon Building, Worthen Room
- 5:15 p.m. - 6:15 p.m. First Business Meeting Lyon Building, Nucor Auditorium
- 6:15 p.m. - 7:00 p.m. Mixer Lyon Building, 2nd floor patio
- 7:00 p.m. - 8:00 p.m. Banquet Edwards Commons, Dining Hall
- 8:00 p.m. - 9:00 p.m. Keynote Address by Dr. Guy Consolmagno Edwards Commons, Dining Hall

Saturday, April 8
- 7:30 a.m. - 8:30 a.m. Continental Breakfast Derby Center, Basement
- 8:00 a.m. - 9:30 a.m. Registration Derby Center, 1st floor lobby
- 8:00 a.m. - 9:15 a.m. Oral Presentations (Session 3) Derby Center, Rooms 009, 011, 016, 023
- 9:30 a.m. - 10:30 a.m. UAMS Admissions Workshop Lyon Building, Nucor Auditorium
- 9:30 a.m. - 10:45 a.m. Oral Presentations (Session 4) Derby Center, Rooms 009, 011, 016
- 10:30 a.m. - 12:00 p.m. Poster Takedown Alphin Building, Kresge Gallery
- 11:00 a.m. - 11:15 a.m. AJAS Winner Oral Presentation Lyon Building, Nucor Auditorium
- 11:15 a.m. - 12:15 p.m. Second Business Meeting Lyon Building, Nucor Auditorium
- 12:15 p.m. Adjourn

SECTION PROGRAMS

*** Undergraduate **Graduate**

ORAL PRESENTATIONS

(Speakers' Underlined)

Session I: Friday April 7, 2006, 1:00 pm – 2:30 pm

Physics/Engineering I **Derby Room 009**

1: 00 pm **STUDY OF CAUSAL COMPONENT PLACEMENT IN AN ACTIVE SOUND CANCELLATION SYSTEM.** Eileen Anderson and Andrew B. Wright Department of Applied Science, University of Arkansas at Little Rock, 2801 S. University, Little Rock, AR 72204

1:15 pm **ANALYSIS OF BEARING VIBRATION SIGNATURES USING THE HILBERT-HUANG TRANSFORM.** Shivan Haran[1], Mario A. Davidson[1] and Liming W. Salvino[2] [1]College of Engineering, Arkansas State University, State University (Jonesboro), AR 72467 [2]Structures and Composite (Code 652), Carderock Division, NSWC, 9500 MacArthur Boulevard, West Bethesda, MD 20817-5700

1:30 pm **INVESTIGATION OF ELECTROMAGNETIC SHIELDING IN FERROMAGNETIC NANO/MICRO POWDER/ POLYMER COMPOSITE FILMS.** Matthew Lemay, Robert Engelken, David Harlan, W. Clark Marler and Michael Sattler Optoelectronic Materials Research Laboratory, College of Engineering, Arkansas State University, P.O. Box 1740, State University, AR 72467

1:45 pm **ANTIFORCE BREAKDOWN WAVES.** Michael Weller and Mostafa Hemmati Physical Science Department, Arkansas Tech University, Russellville, AR 72801

2:00 pm **WHEN YOU CAN'T TAKE A DUST MOP TO SPACE: A SOLUTION TO THE MARS ROVER PARTICLE ADHESION PROBLEM.** Chris A. Wyatt Department of Applied Science, University of Arkansas at Little Rock, 2810 S. University Ave, Little Rock, AR 72204

2:15 pm **NANO-SCIENCE MATERIAL RESEARCH, DEVELOPING QUANTUM DOTS AS IR DETECTORS.** Kenauiya Strain[1], Mansour Mortazavi[1], Gregory Salamo[2] and Ziad Abu-Waar[2] [1]University of Arkansas at Pine Bluff, Pine Bluff, Arkansas 71601 [2]University of Arkansas, Fayetteville, Arkansas 72701

Forestry & Environmental Science I **Room 011**

1:00 pm **HENDERSON MIDDLE SCHOOL ENVIRONMENTAL EDUCATION OUTREACH.** Marcy Bonewits, Souvidhya Khadka, Drew Pendergraft and Ginny Adams University of Central Arkansas, Environmental Science Program, 016 LSC, Conway, AR 72035-5003

1:15 pm **POPULATION STATUS AND DISTRIBUTION OF SPRING CAVEFISH, *FORBESICHTHYS AGASSIZI*, IN SOUTHEAST MISSOURI.** Bonnie Earleywine, Ginny Adams and S. Reid Adams Department of Biology, University of Central Arkansas, Conway, AR 72035

1:30 pm **DIFFERENCES IN CORAL COMMUNITY STRUCTURE REFLECT IMPACTS OF INDIGENOUS VILLAGES AND AGRICULTURE AT 9 REEF SITES IN KUNA YALA, PANAMA.** Shara Jones, Michelle Dare, April Helms and James Engman Henderson State University, Biology Department, Box 7520, Arkadelphia, AR 71999-0001

1:45 pm **ARKANSAS'AGRICULTURALBIOMASSREALLOCATION POTENTIALS.** Jason Self, Aaron Archer and Robert Engelken Arkansas State University, Department of Environmental Sciences, College of Engineering, P.O. Box 1740, State University, AR 72467

2:00 pm **THE ROLE OF BIOLOGY AND CULTURE IN SPECIES INVASIONS: CONTRASTING EVIDENCE FROM TWO INVADED RANGES** Taldi Walter and Katherine Larson Department of Biology, University of Central Arkansas, Conway, AR 72035

2:15 pm **FIVE YEARS OF CHANGE IN AN OLD-GROWTH PINE-HARDWOOD REMNANT IN ASHLEY COUNTY, ARKANSAS.** Don C. Bragg USDA Forest Service, Southern Research Station, P.O. Box 3516 UAM, Monticello, AR 71656

Biochemistry/Cell Biology **Derby Room 016**

1:00 pm **STRUCTURAL STUDIES OF THE BACTERIOPHAGE T4 HELICASE DDA.** Lauren P. Blair University of Arkansas for Medical Sciences, 4301 W. Markham, Little Rock, AR, 72205

1:15 pm **A MUTATION IN ESTROGEN RECEPTOR ALPHA THAT SELECTIVELY ATTENUATES ACTIVATION BY PHYTOESTROGENS.** Barry D. Gehm, J. C. Douglas and Justin Holt Science Division, Lyon College, Batesville, AR 72501

1:30 pm ***TRANS*-ACTIVATION OF A MEIOTIC HOMOLOGOUS RECOMBINATION HOTSPOT IN *ADE6* OF *SCHIZOSACCHAROMYCES POMBE* BY CHIMERIC PROTEIN GAL4DBD-ATF1.** Jun Gao, Mari K. Davidson and Wayne P. Wahls Department of Biochemistry and Molecular Biology, University of Arkansas for Medical Sciences, Little Rock, Arkansas 72205

1:45 pm **DEVELOPING A *C. ELEGANS*-BASED BIOASSAY FOR ESTROGENIC ACTIVITY** Barry D. Gehm, Katie McLean, Justin Holt, Ashley Turensky, Allyn Dodd and Tim Lindblom Science Division, Lyon College, Batesville, AR 72501

2:00 pm ***TRANS*-ACTIVATION OF MEIOTIC RECOMBINATION BY mRNA** Jacy L. Wagnon, Mari K. Davidson and Wayne P. Wahls Department of Biochemistry and Molecular Biology, University of Arkansas for Medical Sciences, Little Rock, AR 72205

2:15 pm **FORMATION OF MULTIPLE CYP2E1 COMPLEXES AFFECTS ACTIVITY** Arvind P. Jamakhandi[1], Daniel E. Sanders[2] and Grover P. Miller[1] [1]Department Biochemistry and Molecular Biology, University of Arkansas for Medical Sciences, Little Rock, AR 72205 [2]Southern Arkansas University, Magnolia, AR 71753

General Biology I **Derby Room 023**

1:00 pm **POLYUNSATURATED FATTY ACIDS METABOLIC ABNORMALITIES IN EXPERMENTAL HYPOTHYROIDISM** Lawrence M. Mwasi University of Arkansas at Pine Bluff, Department of Biology, Mail Slot 4972, 1200 North University Drive, Pine Bluff, AR 71601

1:15 pm **THE VERTICAL DISTRIBUTION OF MYXOMYCETES WITHIN FOREST FLOOR LITTER** Adam W. Rollins and Steven L. Stephenson Department of Biological Sciences, University of Arkansas, Fayetteville, AR 72701

1:30 pm **FISH COMMUNITY STRUCTURE IN FLOODPLAIN WETLANDS OF THE ARKANSAS RIVER** Matt D. Schroeder, Robert L. Clark and S. Reid Adams University of Central Arkansas, Conway, AR 72034

1:45 pm **GENETIC STRATEGIES FOR IMPROVEMENT OF VARICELLA VACCINE** Toby M. Ward and Wayne L. Gray University of Arkansas for Medical Sciences, 4301 W. Markham Street, Little Rock, AR 72205

2:00 pm **MYCETOZOANS OF THE NATIONAL PARKS** Katherine E. Winsett[1], Sally Edwards[1], Lora Lindley[1], Melissa McElderry[1], Rodney K. Nelson[2], Fred Spiegel[1] and Steven L. Stephenson[1] [1]University of Arkansas, Department of Biological Sciences, SCEN 601, Fayetteville AR, 72701 [2]University of Arkansas, Fort Smith, Math-Science 226, 5210 Grand Avenue, PO Box 3649, Fort Smith AR, 72913

2:15 pm **THE SIMIAN VARICELLA VIRUS GENE 61 IS A VIRAL TRANSACTIVATOR BUT IS NOT ESSENTIAL FOR REPLICATION IN CELL CULTURE** Kara Davis, Toby Ward, Yang Ou and Wayne L. Gray University of Arkansas for Medical Sciences, Little Rock, AR 72205

Session II: Friday April 7, 2006, 3:30 pm – 5:00 pm

Chemistry — Derby Room 009

3:30 pm **RESPONSE SURFACE OPTIMIZATION OF A MICROWAVE ASSISTED PERKIN REACTION** Laura McWilliams and R. David Pace Science Division, Lyon College, P.O. Box 2317, Batesville, AR 72503

3:45 pm **DEPOSITION OF NOVEL SEMICONDUCTOR/ POLYMER NANOCOMPOSITE PHOTOCONDUCTOR CONFIGURATIONS** Raphael Küeff, David Harlan, Robert Engelken, W. Clark Marler and Michael SattlerOptoelectronic Materials Research Laboratory, Electrical,Computer, and Information Engineering Program, Arkansas State University, P.O. Box 1740, State University (Jonesboro), AR 72467

4:00 pm **SYNTHESIS, CHARACTERIZATION, AND ANTIMICROBIAL ACTIVITY OF NOVEL THIOSEMICARBAZONES AND THEIR TRANSITION METAL COMPLEXES** Floyd A. Beckford and Amon Holt Science Division, Lyon College, Batesville, AR 72501

4:15 pm **ACETATE INDUCED REARRANGEMENT OF METAL-BOUND PYBOX LIGANDS** Abul B. Kazi[1], David A. Vicic[2] and Gavin D. Jones[2] [1]Department of Chemistry and Physics, University of Arkansas at Pine Bluff, Pine Bluff, AR 71601 [2]Department of Chemistry and Biochemistry, University of Arkansas, Fayetteville, AR 72701

4:30 pm **PHOTOCONDUCTANCE PROPERTIES OF CHEMICALLY DEPOSITED FILMS OF LOW HAZARD, ENVIRONMENTALLY BENIGN BISMUTH SULFIDE FILMS** W. Clark Marler, Robert Engelken, Michael Sattler, David Harlan, Matthew Lemay and Raphael Küeff Optoelectronic Materials Research Laboratory, Electrical, Computer, and Information Engineering Program, Arkansas State University, P.O. Box 1740, State University (Jonesboro), AR 72467

4:45 pm **MEASUREMENT OF ANTIOXIDANT CAPACITY OF AMINOTHIOLS AND THIAZOLIDINES USING THE TEAC ASSAY** Richard Walker, Yvonne Abbey, Malcom Mathis II and Janeé Adams Department of Chemistry and Physics, University of Arkansas at Pine Bluff, Slot 4941, Pine Bluff, AR 71601

Forestry/Environmental Science II — Derby Room 011

3:30 pm **INDIVIDUAL-TREE, OUTSIDE-BARK, GREEN SAWLOG WEIGHT EQUATIONS FOR THREE HARDWOOD SPECIES IN NORTHWEST ARKANSAS** Paul F. Doruska, Jonathan I. Hartley, Matthew B. Hurd and David W. Patterson Arkansas Forest Resources Center, University of Arkansas - Monticello School of Forest Resources, P.O. Box 3468, Monticello, AR 71656

3:45 pm **INDIVIDUAL-TREE, OUTSIDE-BARK, MERCHANDIZED STEM LENGTH GREEN WEIGHT EQUATIONS FOR SAWTIMBER-SIZED HARDWOOD SPECIES IN SOUTHERN ARKANSAS** Matthew B. Hurd, Paul F. Doruska, David W. Patterson, and Jonathan I. Hartley Arkansas Forest Resources Center, University of Arkansas - Monticello School of Forest Resources, P.O. Box 3468, Monticello, AR 71656

4:00 pm **TONS PER THOUSAND BOARD FEET CONVERSION FACTORS FOR ARKANSAS HARDWOODS** Jonathan I. Hartley, Paul F. Doruska, David W. Patterson and Matthew B. Hurd Arkansas Forest Resources Center, University of Arkansas-Monticello School of Forest Resources, P.O. Box 3468, Monticello, AR 71656

4:15 pm **EFFECTS OF PRESCRIBED BURNING ON REPRODUCTION OF THE WESTERN SLIMY SALAMANDER (*PLETHODON ALBAGULA*) IN ARKANSAS** Joseph R. Milanovich and Stanley E. Trauth Department of Biological Sciences, Arkansas State University, State University, AR 72467-0599

4:30 pm **A COMPLEX FOREST ON THE WHITE RIVER NATIONAL WILDLIFE REFUGE: IMPLICATIONS FOR BOTTOMLAND HARDWOOD OLD GROWTH** Brian Roy Lockhart[1] and Jamie E. Kellum[2] [1]U.S. Forest Service Southern Research Station, Center for Bottomland Hardwoods Research, P.O. Box 227, Stoneville, MS 38776 [2]U.S. Fish and Wildlife Service, White River National Wildlife Refuge, P.O. Box 47, St. Charles, AR, 72140

4:45 pm **ECONOMIC IMPACTS OF FUTURE BIOREFINERIES IN THE STATE OF ARKANSAS** Sayeed R. Mehmood and Matthew Pelkki Arkansas Forest Resource Center, School of Forest Resources, University of Arkansas at Monticello, P.O. Box 3468, Monticello, AR 71656

General Biology II — Derby Room 016

3:30 pm **A HYBRID ALGORITHM BASED ON MIE THEORY AND EVOLUTION STRATEGY FOR BREAST CANCER IMAGING** Shruti Pandalraju[1], Payam Rashidi[1], Magda El-Shenawee[1] and Demetrio Macias[2] [1]Department of Electrical Engineering, University of Arkansas, Fayetteville, AR 72701, USA [2]Laboratoire de Nanotechnologie et d'Instrumentation Optique -CNRS FRE 2671, Université de Technologie de Troyes, 12 rue Marie Curie, B.P. 2060, 10010 Troyes, France

3:45 pm **IMPROVED MICROSTRIP PATCH ANTENNA FOR BREAST CANCER DETECTION** Gokul Nanda Talapanuri and Magda El-Shenawee Department of Electrical Engineering, University of Arkansas, Fayetteville, AR 72701

4:00 pm **NEURAL NETWORKS AS STATISTICAL INDICATOR OF BREAST CANCER USING SCATTERED ELECTROMAGNETIC DATA** Douglas Woten[1], Payam Rashidi[2], John Lusth[3] and Magda El-Shenawee[2] [1]Microelectronics – Photonics Program (MicroEP); [2]Department of Electrical Engineering; [3]Department of Computer Science and Computer Engineering, University of Arkansas, Fayetteville, AR 72701

4:15 pm **MYXOMYCETES AS POTENTIAL BIOMONITORS OF AERIAL POLLUTANTS – A PRELIMINARY INVESTIGATION IN ROCKY MOUNTAIN NATIONAL PARK** Rodney K. Nelson[1] and Steve L. Stephenson[2] 1Department of Biology, University of Arkansas–Fort Smith, Fort Smith, Arkansas 72913 [2]Department of Biological Sciences, University of Arkansas–Fayetteville, Fayetteville, Arkansas 72701

4:30 pm **MYXOMYCETES OF THE TALAMANCA RANGE, COSTA RICA** Carlos Rojas[1], Steven L. Stephenson[1] and Martin Schnittler[2] [1]Department of Biological Sciences, University of Arkansas, Fayetteville, AR 72701

4:45 pm **HISTORY AND CURRENT STATUS OF CHANNEL CATFISH POPULATIONS WITHIN BUFFALO NATIONAL RIVER (BUFF)** Faron D. Usrey[1], Shawn Hodges[1], Mark Oliver[2] and Ken Shirley[2] [1]Buffalo National River, 405 North Main, Harrison, AR 42601 [2]Arkansas Game and Fish Commission, Fisheries District 2, Mountain Home, AR 72653

General Biology III **Derby Room 023**

3:30 pm **REPORT OF TRAPPING RATES FOR TURTLES OF THREE NORTHWESTERN LOUISIANA LACUSTRINE SYSTEMS, WITH BIOTELEMETRY OF NATIVE VS. NON-NATIVE ALLIGATOR SNAPPING TURTLES (*MACROCHELYS TEMMINCKII*)** V. Bogosian and L. M. Hardy Museum of Life Sciences, Louisiana State University in Shreveport, One University Place, Shreveport, Louisiana 71115

3:45 pm **NEW EVIDENCE FOR MATERNITY COLONIES OF THE INDIANA BAT (*MYOTIS SODALIS*) IN THE DELTA OF ARKANSAS** Stephen C. Brandebura, Rex E. Medlin and Thomas S. Risch Department of Biological Sciences, Arkansas State University, Jonesboro, AR, 72467

4:00 pm **ICHTHYOFAUNAL ASSEMBLAGES IN THREE PROXIMATE BUT ECOLOGICALLY IVERSE STREAMS IN CLARK COUNTY, ARKANSAS** Scott Jordan[1], Renn Tumlison[1], Lesley Self[1] and Henry W. Robison[2] [1]Department of Biology, Henderson State University, Arkadelphia, AR 71999 [2]Department of Biology, Southern Arkansas University, Magnolia, AR 71754

4:15 pm **DISCRIMINATION OF COLIFORM SOURCES IN ARKANSAS RIVER VALLEY WATERWAYS** Anna Goodwin, Jennifer Looper, Callie Causey, Amanda Ingle, Sarah Norman, Scott Kirkconnell and Tsunemi Yamashita Biology Department, Arkansas Tech University, Russellville, AR 72801

4:30 pm **DISTRIBUTION OF ARKANSAS'S BOTTOMLAND BATS** Rex E. Medlin, Jr., Stephen C. randubura, H. Bobby Fokidis and Thomas S. Risch Arkansas State University, Department of Biological Sciences, State University, AR 72467

4:45 pm **THE IMPACTS OF ROADWAYS ON THE POPULATION DEMOGRAPHY OF TURTLE POPULATIONS** Sara Ruane and Stephen A. Dinkelacker University of Central Arkansas, Department of Biology, Conway, Arkansas, 72035

Session III: Saturday April 8, 2006, 8:00 am – 9:15 am

Computer Science/Science Education **Derby Room 009**

8:00 am **INNOVATIVE APPLICATIONS OF CLASSROOM RESPONSE SYSTEMS: INVESTIGATING STUDENTS' ITEM RESPONSE TIME** Wilson J. Gonzalez-Espada and Daniel W. Bullock School of Physical and Life Sciences, Arkansas Tech University, 1701 North Boulder Avenue, Russellville, AR 72801

8:15 am **DUELING DISCIPLINES: TEACHING PHYSICS THROUGH MUSIC** Joel Plaag[1], Stuart Hutton[2] and Ying Hutton[3] [1]Fine Arts Division and [2]Science Division, Lyon College, Batesville, AR 72503 [3]Department of Physics and Astronomy, University of Central Arkansas, Conway, AR 72035

8:30 am **LEVELS OF UNDERSTANDING OF PHYSICAL SCIENCE CONCEPTS OF COLLEGE STUDENTS ENROLLED IN GENERAL EDUCATION PHYSICAL SCIENCE COURSES** Tillman Kennon Arkansas State University, Chemistry and Physics Department, PO Box 419, State University, AR 72467-0419

8:45 am **SCIENCE LABORATORY SAFETY TRAINING FOR 7-12 ARKANSAS TEACHERS** Tillman Kennon and Ann Ross Arkansas State University, Chemistry & Physics and Teacher Education Departments, PO Box 419, State University, AR 72467-0419

9:00 am **PARALLEL ALGORITHMS FOR MULTICRITERIA SHORTEST PATH PROBLEMS** David L. Sonnier Lyon College, P.O. Box 2317, Batesville, Arkansas, 72503

Aquatic Biology I **Derby Room 011**

8:00 am **DISTRIBUTION OF BASS TAPEWORM AND YELLOW GRUB INFECTIONS IN ARKANSAS IMPOUNDED RESERVOIRS AND UPLAND STREAMS** James J. Daly, Sr.[2], Randall J. Keller[1], and Bruce DeYoung[2] [1]Department of Occupational Safety and Health, P.O. Box 9, Murray State University, Murray, KY, 42071 [2]Department of Microbiology and Immunology, University of Arkansas for Medical Sciences, Little Rock, AR 72205

8:15 am **A NON-INVASIVE TECHNIQUE FOR ASSESSING THE POPULATION PARAMETERS OF *CLINOSTOMUM* METACERCARIA IN SMALLMOUTH BASS (*MICROPTERUS DOLOMIEU*)** James J. Daly, Sr.[2], Randall J. Keller[1], and Bruce DeYoung[2] [1]Department of Occupational Safety and Health, P.O. Box 9, Murray State University, Murray, KY, 42071 [2]Department of Microbiology and Immunology, University of Arkansas for Medical Sciences, Little Rock, AR 72205

8:30 am **THE USE OF PARAMETRIC STATISTICAL METHODS FOR DETERMINING SIGNIFICANT DIFFERENCES BETWEEN POPULATIONS OF *CLINOSTOMUM* METACERCARIA IN SMALLMOUTH BASS** James J. Daly, Sr.[1] and Randall J. Keller[2] [1]Department of Microbiology and Immunology, University of Arkansas for Medical Sciences, Little Rock, AR 72205 [2]Department of Occupational Safety and Health, P.O. Box 9, Murray State University, Murray, KY, 42071

8:45 am **SITE DENSITY DISTRIBUTION OF "YELLOW GRUB" (*CLINOSTOMUM MARGINATUM*) IN PONDRAISED CHANNEL CATFISH (*ICTALURUS PUNCTATUS*)** Jeurel Singleton[1], James J. Daly Sr.[2] and Randall J. Keller[3] [1]Department of Natural Sciences, University of Maryland Eastern Shore, Princess Anne, MD [2]Department of Microbiology, University of Arkansas for Medical Sciences, Little Rock, AR [3]Department of Occupational Safety and Health, Murray State University, Murray, KY

9:00 am **NOTEWORTHY GEOGRAPHIC DISTRIBUTION RECORDS FOR THE GOLDEN TOPMINNOW, *FUNDULUS CHRYSOTUS* (CYPRINODONTIFORMES: FUNDULIDAE), FROM ARKANSAS** Chris T. McAllister[1], Henry W. Robison[2], and Thomas M. Buchanan[3] [1]Department of Biology, Angelo State University, San Angelo, TX 76909 [2]Department of Biology, Southern Arkansas University, Magnolia, AR 71754 [3]Department of Biology, University of Arkansas-Fort Smith, Fort Smith, AR 72913

Zoology I **Derby Room 016**

8:00 am **OBSERVATION OF AN UNUSUAL PREY ITEM DELIVERED TO AN EASTERN BLUEBIRD NEST** Sarah A. De Viney, Richard J. Baxter and Thomas S. Risch Arkansas State University, P.O. Box 599, State University, AR 72467

8: 15 am **PRELIMINARY RESULTS OF SURVEYS FOR WINTERING SMITH'S LONGSPURS AT AIRPORTS ACROSS ARKANSAS** William C. Holimon[1] and Catherine W. Rideout[2] [1]Arkansas Natural Heritage Commission, 1500 Tower Building, 323 Center Street, Little Rock, AR 72201 [2]Arkansas Game and Fish Commission, #2 Natural Resources Drive, Little Rock, AR 72205

8:30 am **DOCUMENTATION OF THE FIRST KNOWN OCCURRENCE OF BROAD-BILLED HUMMINGBIRD (*CYNANTHUS LATIROSTRIS*) IN ARKANSAS** Robert H. Doster[1], Dan Scheiman[2], Max D. Parker[3], E. Pershing Floyd[3], Kenny Nichols[3] and LaDonna Nichols[3] [1]U.S. Department of the Interior, Bureau of Reclamation, 555 Broadway Ave. NE, Suite 100, Albuquerque, NM 87102 [2]Audubon Arkansas, 201 East Markham St., Suite 450, Little Rock, AR, 72201 [3]Arkansas Audubon Society, 14300 Chenal Pkwy., #7473, Little Rock, AR 72211

8:45 am **SPERM MORPHOLOGY AND MORPHOMETRICS IN THE RINGED SALAMANDER, *MBYSTOMA ANNULATUM* (CAUDATA: AMBYSTOMATIDAE), USING SCANNING ELECTRON MICROSCOPY** Stanley E. Trauth[1], Malcolm L. McCallum[2] and Joseph R. Milanovich[1] [1]Department of Biological Sciences, Arkansas State University, P.O. Box 599, State University, AR 72467 [2]Department of Biological Sciences, Texas A&M University-Texarkana, 2600 Robison Rd., Texarkana, TX 75501

9:00 am **A PRELIMINARY CHECKLIST OF THE MOTHS (LEPIDOPTERA) OF CLARK COUNTY, ARKANSAS** Renn Tumlison and Kristen Benjamin Department of Biology, Henderson State University, Arkadelphia, AR 71999

Botany **Derby Room 023**

8:00 am **THE VASCULAR FLORA OF THE SOUTH FORK NATIVE PLANT PRESERVE, VAN BUREN COUNTY, ARKANSAS** Theo Witsell[1] and Brent Baker[2] [1]Arkansas Natural Heritage Commission, 1500 Tower Building, 323 Center Street, Little Rock, AR 72201 [2]Department of Biology, Lewis Science Center, University of Central Arkansas, 201 Donaghey Avenue, Conway, AR 72035

8:15 am **CHINESE FLAME TREE (*KOELREUTERIA BIPINNATA* FRANCH., SAPINDACEAE) NEW TO THE ARKANSAS FLORA** Nicole Freeman, Sara Melancen and Brett Serviss Department of Biology, Henderson State University, Arkadelphia, AR 71999–0001

8:30 am **A PRELIMINARY INVESTIGATION INTO THE OCCURRENCE, ESTABLISHMENT, AND BIOLOGY OF SEVEN, NON–NATIVE, WOODY ANGIOSPERMS IN SOUTHWEST ARKANSAS** Sara Melancen, Nicole Freeman, Johnathan Fuell, Allen Leible and Brett E. Serviss Department of Biology, Henderson State University, Arkadelphia, AR 71999–0001

8:45 am **STATUS OF WOLF'S SPIKERUSH (*ELEOCHARIS WOLFII*), CYPERACEAE, IN ARKANSAS AND NORTH AMERICA** Theo Witsell[1] and Paul McKenzie[2] [1]Arkansas Natural Heritage Commission, 1500 Tower Building, 323 Center Street, Little Rock, AR 72201 [2]U.S. Fish and Wildlife Service, 608 E. Cherry Street, Room 200, Columbia, MO 65201-7712

Session IV: Saturday April 8 2006, 9:30 am – 10:45 am

Physics/Engineering II **Derby Room 009**

9:30 am **SAFETY OF A RED DIODE LASER SOURCE USED IN FETAL RETINAL STIMULATION STUDIES** Al Adams Department of Physics and Astronomy, University of Arkansas at Little Rock, 2801 South University Ave, Little Rock, AR 72204

9:45 am **UNDERSTANDING GLOBAL POSITIONING SYSTEM LIMITATIONS: A CASE STUDY OF MAPPING AND SURVEY GRADE GPS** Buren B. DeFee, II and Chris Stuhlinger University of Arkansas at Monticello, School of Forest Resources, P.O. Box 3468, Monticello, AR 71656

10:00 am **AC IMPEDANCE SPECTROSCOPY IN SOLID SOLUTIONS OF CuXFeI-XTAC** Stuart Hutton and Yagya Regmi Science Division, Lyon College, Batesville AR 72503

10:15 am **VARIATIONAL SYMMETRIES AND CONSERVATION LAWS IN LINEARIZED GRAVITY** Balraj Menon Department of Physics and Astronomy, University of Central Arkansas, Conway, AR 72035

Aquatic Biology II **Derby Room 011**

9:30 am **AQUATIC MACROINVERTEBRATES OF THE STRAWBERRY RIVER SYSTEM, NORTHCENTRAL ARKANSAS** George L. Harp[1] and Henry W. Robison[2] [1]Department of Biological Sciences, Arkansas State University, AR 72467 [2]Department of Biology, Southern Arkansas University, Magnolia, AR 71754-9354

9:45 am **BIOMASS DYNAMICS OF *TIPULA* (INSECTA:DIPTERA) IN FORESTED STREAMS OF THE INTERIOR HIGHLANDS, ARKANSAS** S. B. McCord, A. D. Christian, and R. S. Grippo Department of Biological Sciences, Arkansas State University, State University, AR 72467

10:00 am **INVESTIGATIONS ON THE LEECHES OF ARKANSAS** William E. Moser[1], Donald J. Klemm[2], Dennis J. Richardson[3], Benjamin A. Wheeler[4], Stanley E. Trauth[5] and Bruce A. Daniels[6] [1]Department of Invertebrate Zoology, P.O. BOX 37012 National Museum of Natural History, Smithsonian Institution, Washington, DC 20013-7012 [2]Ecosystems Research Branch (MS-642), U.S. Environmental Protection Agency, Cincinnati, OH 45268- 0001 [3]Quinnipiac University, 275 Mount Carmel Avenue, Campus Box 71, Hamden, CT 06518 [4]Arkansas State University, Environmental Sciences Program, P.O. Box 847, State University, AR 72476 [5]Arkansas State University, Department of Biological Sciences, State University, AR 72467 [6]Smithsonian Institution, Office of Information Technology, PO Box 37012, Washington, DC 20013-7012

10:15 am **THE CAJUN DWARF CRAWFISH (*CAMBARELLUS SHUFELDTII*): AN INTERMEDIATE HOST FOR *SOUTHWELLINA DIMORPHA* (ACANTHOCEPHALA)** Dennis J. Richardson[1] and William F. Font[2] [1]Quinnipiac University, Box 71, 275 Mt. Carmel Avenue, Hamden, CT 06518 [2]Department of Biological Sciences, Southeastern Louisiana University, Hammond, Louisiana 70402

10:30 am **STATUS AND DISTRIBUTION OF THE ARKANSAS DARTER (*ETHEOSTOMA CRAGINI*) IN ARKANSAS** Brian K. Wagner and Mark D. Kottmyer Arkansas Game and Fish Commission, 2 Natural Resource Drive, Little Rock, AR 72205

Zoology II **Derby Room 016**

9:30 am **DISCOVERY OF A DICEPHALIC WESTERN DIAMONDBACK RATTLESNAKE, *CROTALUS ATROX* (SERPENTES: VIPERIDAE), FROM WEST TEXAS, WITH COMMENTS ON OTHER MONSTERS** Chris T. McAllister[1] and Van Wallach[2] [1]Department of Biology, Angelo State University, San Angelo, TX 76909 [2]Museum of Comparative Zoology, Harvard University, Cambridge, MA 02138

9:45 am **DISTRIBUTION AND STATUS OF THE PEPPERED SHINER, *NOTROPIS PERPALLIDUS* HUBBS, IN ARKANSAS AND OKLAHOMA.** Henry W. Robison Department of Biology, Southern Arkansas University, Magnolia, AR 71754-9354

10:00 am **NEW DISTRIBUTIONAL RECORDS FOR LAMPREYS (PETROMYZONTIDAE) FROM ARKANSAS** Renn Tumlinson[1], Henry W. Robison[2] and James Petersen[3] [1]Department of Biology, Henderson State University, Arkadelphia, AR 71999 [2]Department of Biology, Southern Arkansas University, Magnolia, AR 71754 [3]U. S. Geological Survey, Little Rock, AR 70000

10:15 am **GEOGRAPHIC DISTRIBUTION AND HARVEST DYNAMICS OF THE EASTERN SPOTTED SKUNK IN ARKANSAS** D. Blake Sasse[1] and Matthew E. Gompper[2] [1]Arkansas Game and Fish Commission, Little Rock, AR 72205 [2]Department of Fisheries and Wildlife Sciences, University of Missouri, Columbia, MO 65211

10:30 am **NOTES ON AN URBAN POPULATION OF THE WESTERN LESSER SIREN (*SIREN INTERMEDIA NETTINGI*), WITH RESPECT TO SIZE, ACTIVITY PERIODS, AND ECOLOGICAL ASSOCIATES** Jacob A. Sawyer and Stanley E. Trauth Department of Biological Sciences, Arkansas State University, State University, AR 72467-0599

POSTER PRESENTATION ABSTRACTS

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Poster 2 **A STUDY OF ARKANSAS DRINKING WATER CONTAMINATION BY MICROENVIRONMENT IN HOME WATER FILTERS** Nathan Avaritt and Olga Tarasenko Department of Biology, University of Arkansas at Little Rock, Little Rock, AR 72204

Poster 3 **DENSITY FUNCTIONAL STUDIES OF NITROSYL METALLOPORPHYIN COMPLEXES** John P. Graham and Robbie F. Davis Department of Physical Science, Arkansas Tech University, Russellville, AR 72801

Poster 4 **ROTATIONAL BARRIERS IN TRANSITION METAL CARBENE COMPLEXES** John P. Graham and Robbie F. Davis Department of Physical Science, Arkansas Tech University, Russellville, AR 72801

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Poster 7 **A RELIABLE, INEXPENSIVE METHOD FOR CREATING SAMPLES OF KNOWN C AND N CONCENTRATIONS FOR USE IN DRY COMBUSTION C-N ANALYZERS** Robert L. Ficklin[1], Russell I. Dresbach[2] and Joshua D. Richardson[1] [1]University of Arkansas - Monticello, Arkansas Forest Resources Center, School of Forest Resources, P.O. Box 3468, Monticello, AR 71656 [2]University of Missouri - Columbia, Department of Civil and Environmental Engineering, Soil Characterization Laboratory E2509, Columbia, MO 65201

Poster 8 **UPDATED LIST OF THE ANTS OF ARKANSAS** David M. General and Lynne C. Thompson University of Arkansas at Monticello, School of Forest Resources, Arkansas Forest Resources Center, Monticello AR 71656

Poster 9 **A COMPARISION OF EMG DURING A NORMAL SUBJECT'S VS. A SUBJECT WITH AN INCOMPLETE SPINAL CORD INJURY'S GAIT** Shivan Haran[1], Shawn M. Drake[2] and Valerie Kerperien[2] [1]College of Engineering; [2]MPT Program, Programs in Physical Therapy, College of Nursing and Health Professions, Arkansas State University, State University (Jonesboro), AR 72467

Poster 10 **RAMAN SPECTROSCOPY OF JSC MARS-1 SIMULATED MARTIAN DUST** Franklin D. Hardcastle[1], and Alexandru S. Biris[2] [1]Department of Physical Sciences, Arkansas Tech University, Russellville, AR 72801 [2]University of Arkansas at Little Rock, UALR Center of Nanotechnology, Graduate Institute of Technology, 2801 S. University Ave., Little Rock, Arkansas 72204

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Poster 17 **CURRENT RESULTS OF ECOPOESIS EXPERIMENTS IN THE SHOT MARTIAN ENVIRONMENT SIMULATOR** Carl Rector[1], Paul Todd[2], Penelope J. Boston[3], John Boling[1], Kathy A. Campbell[4], Tiffany McSpadden[1], Laura McWilliams[1], Jaime Warrington[1] and David J. Thomas[1] [1]Lyon College, Science Division, Batesville, AR 72501 [2]Space Hardware Optimization Technology, Inc., Greenville, IN 47124 [3]New Mexico Institute of Mining and Technology, Department of Earth and Environmental Science, Socorro, NM 87801 4Cedar Ridge High School, Newark AR 72562

Poster 18 **BIOTECHNOLOGY ADVANCES FOR ARKANSAS RICE VARIETIES (*ORYZA SATIVA* L.)** Moytri RoyChowdhury, Audrei Dabul, John Hubstenberger and Gregory Phillips Arkansas BioSciences Institute at Arkansas State University, State University AR-72467

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Poster 20 **THE ARKANSAS CENTER FOR SPACE AND PLANETARY SCIENCES** Derek W. G. Sears, Hazel Sears and Jessica Park Arkansas Center for Space and Planetary Sciences, University of Arkansas, Fayetteville, Arkansas 72701

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Poster 23 **GREEN CHEMISTRY: FROM RICE HULLS TO POLYFURAN** Carol J. Trana, Matthew McConnell, Adam Green, James Lindley, Lindsay Rymes, Shuneize Lowe, Maranda Henley, Kelley Sayyar, Walter E. Godwin and Rose McConnell School of Math & Natural Sciences, University of Arkansas at Monticello, Monticello, AR 71656.

Poster 24 **EXPOSURE TO SUB-LETHAL DOSE OF MALATHION IS MORE DETRIMENTAL TO BRAIN CHOLINESTERASE OF MATURE, REPRODUCTIVE MINNOW (*PIMEPHALES PROMELAS)* THAN THE YOUNG** Eric Duncan1, Jonathan Treece[2] and Malathi Srivatsan[2] [1]University of North Carolina at Pembroke, Pembroke, NC 28372 and [2]Department of Biological Sciences, Arkansas State University, Jonesboro, AR72401

Study of Causal Component Placement in an Active Sound Cancellation System

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Abstract.—In a feedforward Active Sound Cancellation (ASC) system, the acoustic delay between a primary source and an error microphone must be greater than the delay between the measurement of that source by the reference microphone and the arrival of the secondary source's wave at the error microphone. Such a configuration is called a causal configuration. For periodic disturbances, cancellation can still be achieved in a non-causal configuration. Since the waveform is periodic, each cycle of the waveform is identical, and the cycle being canceled is not the measured part of the waveform, but a subsequent cycle in the waveform. Non-periodic sources cannot be cancelled by a non-causal ASC system, and convergence of the Least Mean Squares algorithm is not as effective in a non-causal configuration as in a causal configuration. The ASC system was implemented to create a local zone of silence inside a reverberant enclosure. The primary source was a 125 Hz sinusoid generated outside of the enclosure. System delays were calculated and a causal component configuration was chosen. System performance under both causal and non-causal component configurations was examined. The system was able to create a maximum attenuation of >18 dB in both the causal and non-causal configurations. However, it was discovered that in the non-causal configuration, the computation of the optimal inverse signal was much slower than in the causal configuration.

Key words:— Active Sound Cancellation (ASC), acoustic delay, waveform, Least Mean Squares algorithm.

Introduction

Active Sound Cancellation (ASC), the attenuation of a sound field by constructive interference, has been proven to be a fertile area for research in recent years. A quiet space can be created in a noisy environment without foam, padding or acoustic tiles. ASC has been successfully tested in such diverse settings as mining vehicles (Stanef et al. 2004), high-rise apartments (Zhang et al. 2002), and MRI units (McJury et al. 1997). There are still many unanswered questions regarding the implementation of ASC, such as the ramifications of system causality.

In a feedforward ASC system, a reference microphone is placed where it can sense an unwanted acoustic noise, which is called the primary source. The microphone signal is sent to a controller. The controller computes an inverse signal and outputs it to a speaker located acoustically "downstream," near an area where the sound is to be attenuated. This speaker is called the secondary source. Another microphone, called the error microphone, is placed where the zone of attenuation is desired (see Fig. 1).

The time required to measure the primary source, compute the inverse signal, broadcast the signal, and propagate the acoustic wave to the error microphone must be less than the time required for the primary source to propagate to the error microphone (Ffowcs Williams et al. 1985). Otherwise, the secondary source waveform will arrive after the primary source waveform has passed the error microphone. This constraint in active sound cancellation is called the causality constraint.

The constraint of causality has been acknowledged since ASC was first conceived. Lueg (1936) mentioned that active sound cancellation is dependent on the fact that "the speed of sound is very much less than the speed of electrical impulses" and that there should be ample time for activation of control elements within a circuit before the sound wave to be cancelled arrives. Ffowcks Williams et al, (1985) described the constraint of causality and discussed the causes of electronic delay. Nelson and Elliott (1992) presented a method for approximation of electronic delay based on the number of poles in the low-pass filters.

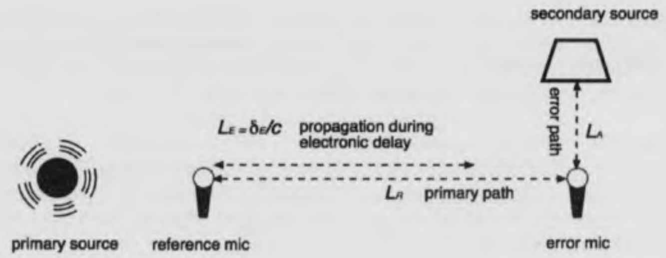

Fig. 1. Feedforward Active Sound Cancellation.

Several studies have demonstrated the effects of a causal configuration when canceling random noise. Tseng et al. (1998) moved the primary source in an arc while leaving the error and reference microphones stationary as the performance of an ASC system was recorded. When the primary source reached the bounds of a causal configuration, performance started to decline, and declined further as the configuration grew more non-causal.

Janocha and Liu (1998) showed in a simulation the deterioration in performance as increasing delay resulted in non-causality. Both of these studies presented results in terms of sound attenuation (dB) at the error microphone (real or simulated), but did not state the length of time that it took the system to reach the given attenuation.

Kong and Kuo (1999) used system efficiency as their metric. Efficiency reflects the percentage of the energy in the primary source that the ASC system is able to reduce. They showed theoretically and by simulation that the efficiency with which an ASC system cancels white noise decreases exponentially as a function of the degree of non-causality. Their simulation predicted that the system converges (albeit at different attenuation levels) in the same time whether the configuration is causal or non-causal.

Feedforward ASC can be performed with some success in a non-causal setting if the primary source is periodic and non-varying. Insufficient attention has been paid to causality, and many systems designed to cancel periodic noise are non-causal. Many researchers have claimed that causality is not important in such a system (Burdisso et al. 1993, Kuo and Morgan 1996, Kang and Kim 1997, Bai et al. 2002). However, the effects of causality, when canceling periodic disturbances, have not been thoroughly investigated.

The work presented in this paper compares the performances of an ASC system canceling a periodic disturbance under both causal and non-causal conditions. A clear benefit of a causal ASC system for periodic disturbances will be demonstrated, and an "easy to use" test to determine whether a system is causal when it is set up will be suggested.

Causality

Causal feedforward ASC depends upon the ability of the system to perform quickly enough to output the secondary source before the primary source's sound wave has propagated to the error microphone. While the sound waves generated by the primary source are travelling to the error microphone, the following events must take place:

1. The reference microphone must sense the sound from the primary source.
2. The reference microphone's signal must travel through the electronic filters and arrive at the controller.
3. The controller must compute the value of the primary source at the error microphone, using the reference microphone signal and the error microphone signal.
4. The controller must compute and output the secondary source signal.
5. This signal must travel through the electronic filters and arrive at the secondary source.
6. The sound must travel from the secondary source to the error microphone.

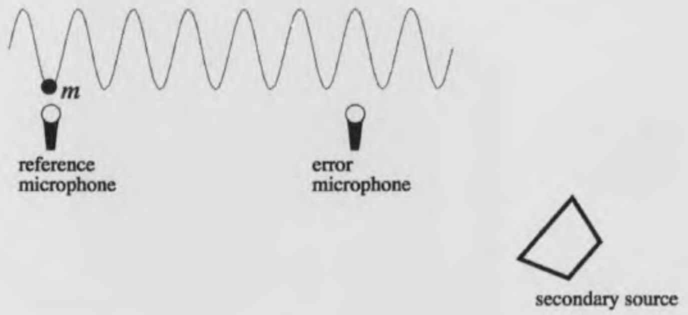

Fig. 2a. Element of acoustic wave at time = t_0.

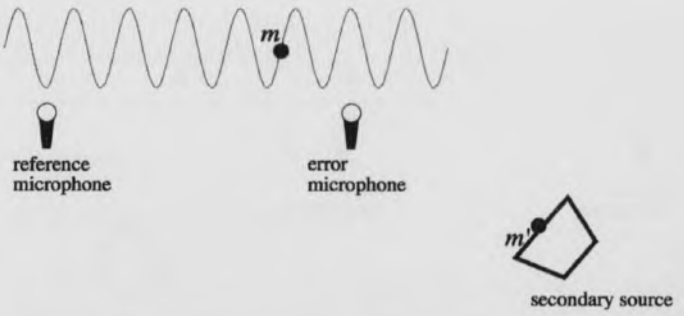

Fig. 2b. Element of acoustic wave at time = $t_0 + \delta_E$.

Each of these events takes time. The combined times for the first 5 items comprise the electronic delay, δ_E. Let the time of the sixth item be δ_A and the time necessary for the primary source to propagate acoustically from the reference microphone to the error microphone be δ_R.

Both δ_A and δ_R are acoustic delays, and the lengths of the respective acoustic paths are L_A/c and L_R/c, where c is the speed of sound (see Fig. 1). To fulfill the constraint of causality,

$$\delta_R - \delta_A \geq \delta_E . \qquad (1)$$

For a causal configuration, the components must be arranged such that their respective distances and the electronic delay allow Equation 1 to be satisfied.

Periodic noise can be cancelled by a non-causal system. Consider an element, m, of an acoustic wave propagating from the reference microphone to the error microphone.

The reference microphone senses an acoustic wave at time t_0 (see Fig. 2a). When the electronic delay has ended at $t_0 + \delta_E$, the wave has propagated through a distance $c(t_0 + \delta_E)$ as shown in Fig. 2b. The controller has finished calculating the inverse wave, and the output signal has reached the secondary source. The inverse wave, with element m' similarly marked, begins to travel. The primary source arrives at the error microphone at t_0

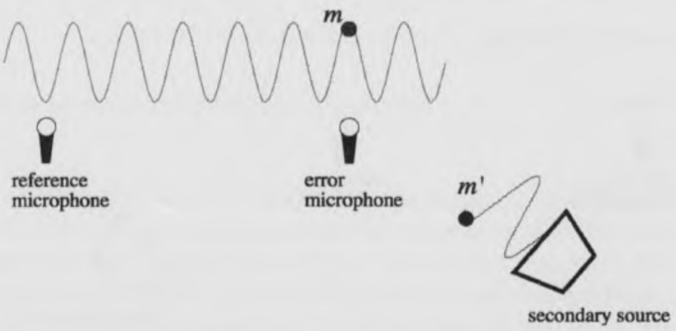

Fig. 2c. Element of acoustic wave at time = $t_0 + \delta_R$.

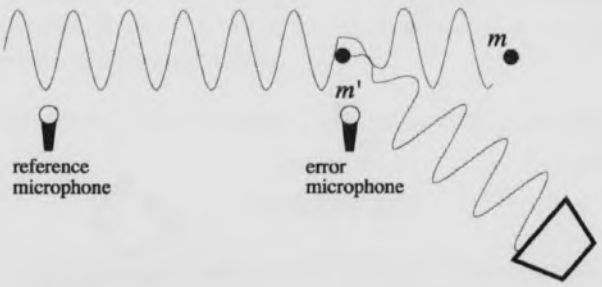

Fig. 2d. Cancellation occurring in non-causal configuration at time = $t_0 + \delta_E + \delta_A$.

+ δ_R (see Fig. 2c). In this example, the electronic delay was too long, and the point *m'* on the inverse signal that was computed to cancel *m* didn't reach the error microphone in time.

However, at $t_0 + \delta_E + \delta_A$ (see Fig. 2d), when the wave from the secondary source reaches the error microphone, the phase of the primary source will approximate its value at $t_0 + \delta_R$. If the difference is only a few cycles, the controller can make the necessary adjustments to cancel the sound at the error microphone.

The secondary source output was calculated to cancel the sound one or more cycles previous to the cycle actually present at the error microphone. The sound is still canceled but only because of the periodicity, not because the system can predict the sound present at the error microphone.

A non-causal system synchronizes the secondary source phase with the primary source waveform at the error microphone modulo 2π, if the primary source waveform is sinusoidal. If the primary source waveform is not truly sinusoidal, such synchronization cannot occur. This situation arises in random signals and non-periodic signals, such as speech.

Should a non-causal system be applied to cancel a non-periodic disturbance, the cancellation would not occur. Since real world acoustic disturbances are rarely completely static, a non-causal system is not reliable.

Materials and Methods

ASC System.—The causality experiments were conducted using the ASC system at the University of Arkansas at Little Rock (UALR). The system uses an Integrated Motions, Inc. MX31 embedded controller with preamplifiers and anti-aliasing filters to condition the signals from the microphones and anti-imaging filters to condition the signals to the speakers (see Fig. 3).

The ASC algorithm was designed in block diagram form using Simulink software and converted into C code using Real-Time Workshop. The controller sample rate was 1000 Hz. Both the primary and secondary sources were generated by JBL J520M speakers with a flat frequency response from 70 Hz to 20 kHz. An ADCOM GFA-6000 five-channel 100 Watt-per-channel audio power amplifier powered the speakers.

The frequency of the primary source was chosen as 125 Hz. A Galois test signal was added to the output of the secondary source to improve system identification. The test signal was composed of 127 harmonics from 3.906 to 500 Hz (Xie 1997). The frequency components of the Galois signal actually generated ranged from approximately 70 Hz to 500 Hz because of the lower limit of the speaker.

Custom circuits were used to condition the signals. Two microphone preamplifier circuits amplified the low-level microphone output signal and limited the radio frequency noise in the system. Two amplifier-filter circuits further amplified the microphone signals to occupy the range of the ± 10 V ADC channels in the MX31 controller, and implemented a low-pass anti-aliasing filter. Two attenuator circuits reduced the output of the MX31 to the ±5 V input range of the power amplifier and also implemented a low-pass anti-imaging filter. The low-pass filters had a cut-off frequency of 693 Hz.

The error and reference microphones were both Radio Shack

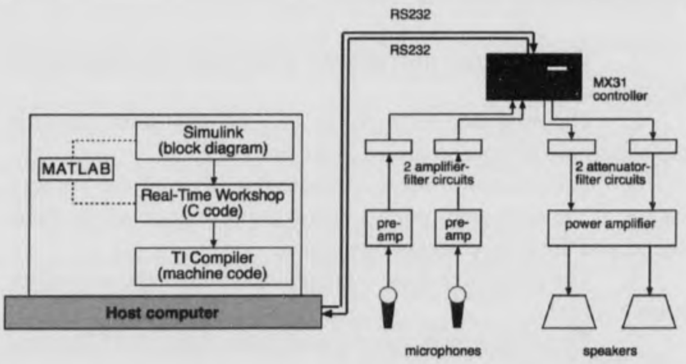

Fig. 3. Experimental hardware.

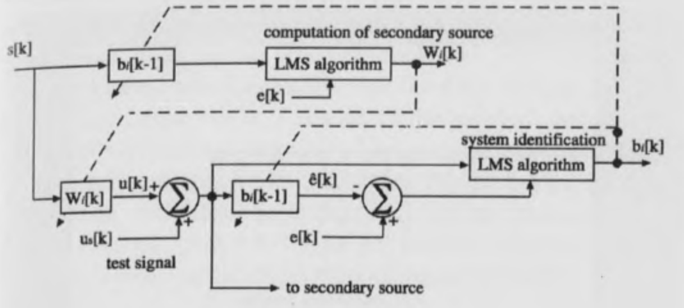

Fig. 4. Controller structure.

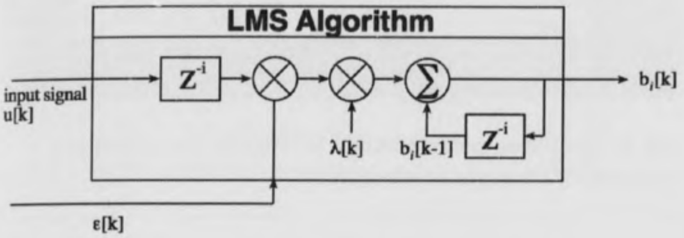

Fig. 5. Block diagram of least mean square algorithm.

No. 270-090 condenser type microphones with a flat frequency response from 20 Hz - 20 kHz.

Algorithm.—The controller performed two adaptive processes: system identification and computation of the filter coefficients used to generate the secondary source (see Fig. 4). The normalized Least Mean Square (LMS) algorithm was used in both of these processes (Fig. 5). It uses a gradient descent mechanism with a convergence parameter *λ[k]* to minimize an error, *ε[k]*. The convergence parameter *λ[k]* is adaptive (Goodwin and Sin 1984), modified by the magnitude of the filter coefficients, b_i:

$$\lambda = \mu\left(\sum_{i=1}^{N_b} b_i^{\,2}\right)^{-1}, \tag{2}$$

where N_b is the length of the filter and μ is a fixed convergence parameter.

In the system identification process, a finite impulse response (FIR) filter generates an estimate, *ê[k]*, of the error signal *e[k]* sensed by the error microphone:

$$\hat{e}[k] = \sum_{i=0}^{N-1} b_i[k]u[k-i], \tag{3}$$

where *N* is the number of filter coefficients and *u[k]* is the generated control signal. The algorithm to update the *N* coefficients is

$$b_i[k] = b_i[k-1] + \lambda\varepsilon\mu[k-1]. \tag{4}$$

In the controller's computation of the filter coefficients to generate the secondary source, the LMS algorithm minimizes the signal *e[k]* from the error microphone.

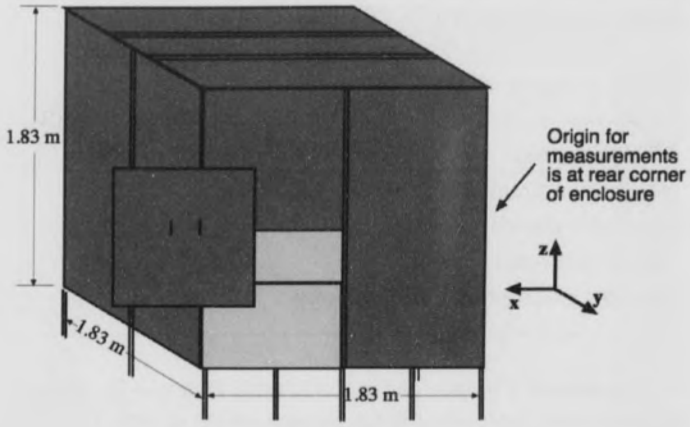

Fig. 6. Acoustic enclosure.

Enclosure.—The experiments were performed in UALR's acoustic enclosure. It is constructed of Unistrut™ steel framing and 1/4-inch exterior grade plywood (see Fig. 6). The floor of the structure is elevated with a clearance of 0.305 m to prevent coupling with the room floor.

The enclosure's internal dimensions are 1.83 m x 1.83 m x 1.83 m. The enclosure has no soundproofing, so external sounds are admitted. Extraneous sounds during the experiments were mainly low frequencies generated by the building ventilation system.

The first set of normal modes of the enclosure occurs at 94 Hz and the Schroeder frequency is approximately 400 Hz. The 125-Hz primary source generated a reverberant but non-diffuse sound field in the enclosure.

Computation of Causal Configuration

To establish a causal system, it was necessary to measure the delay of all system components. The components of the ASC system were tested, separately and in combination, for delays so that the total delay of the system could be known (Anderson 2004). The acoustic delays were known from the distance of the reference microphone to the error microphone and the speed of sound.

Component Delays.—The system components were tested with square waves at several frequencies and several sample times. A 125-Hz square wave sampled at 1 kHz yielded the results shown in Table 1.

Table 1. Component Delays

Component(s)	Delay
Attenuator circuit	2 ms
Amplifier circuit	2 ms
MX31 controller	1 ms
Speaker, microphone, and pre-amp	2 ms

Combined Component and Acoustic Delay.—To measure the combined component delay, square waves at 100, 125, 200, and 250 Hz were generated in a Simulink model, output through a DAC block to the MX31, and output from the MX31 to the attenuator circuit. The signal from the attenuator circuit was fed to the power amplifier, which generated an output to the speaker. The acoustic waves propagated 15 cm to the microphone. The microphone signal was fed to the pre-amplifier, then to the amplifier circuit. The output of the amplifier circuit was connected to the MX31, then fed to the workspace through an A/D block (see Fig. 7).

The combined delay at 125 Hz sampled at 1 kHz was approximately 8 ms. This result agrees well with the sum of the delays of the separate components. The 8 ms delay included an acoustic propagation time of 43 ms, low enough to be ignored when computing the electronic delay alone. The uncertainty due to component placement was about 15 ms for each component, also low enough to be ignored. However, there was an uncertainty of half of the sampling time in the delay. Therefore, the value of the electronic delay used for calculating component placement was 8 ± 0.5 ms.

Placement of Microphones and Secondary Source.—The primary source speaker was placed outside of the enclosure. This placement emulated the common real-world situation of break-in noise, wherein an unpleasant external noise source enters a vehicle, control room, or other enclosed area intended for human use.

Possible placements of components to achieve a causal system, given the location of the primary source, were calculated using a custom MATLAB program. The program used Equation 1 to check the causality at discrete points on a horizontal plane in the enclosure (see Fig. 8).

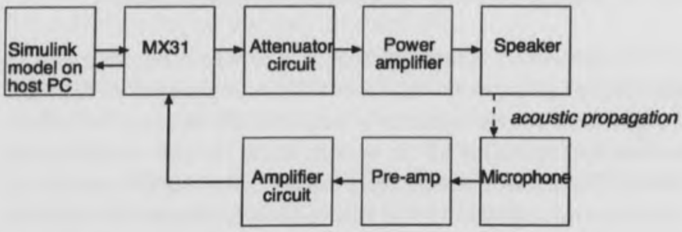

Fig. 7. Combined delay testing set-up.

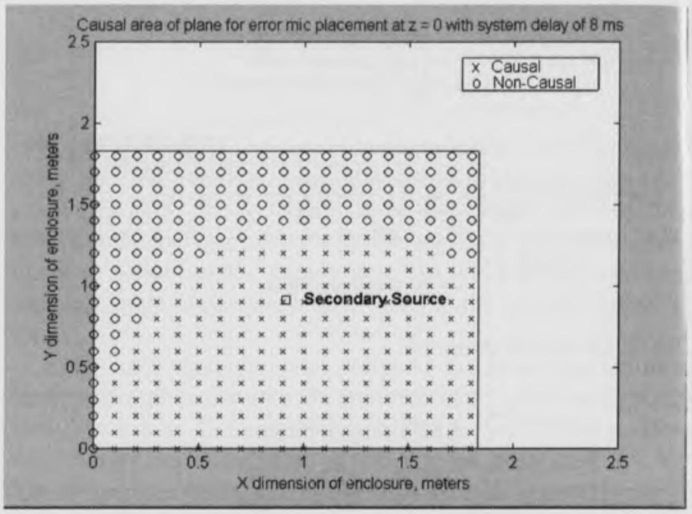

Fig. 8. Sample output of MATLAB program for computing causal error microphone placement.

The reference microphone was placed 10 cm from the primary source. Causal placements for the error microphone and secondary source on the floor of the enclosure were chosen from possible values shown by the program (see Table 2).

Table 2. Location of ASC components

Component	Location (m)
Primary source location	(0.03, 3.28, 2.52)
Reference microphone location	(0.02, 3.18, 2.52)
Secondary source location	(0.91, 0.91, 0.23)
Error microphone location	(1.25, 0.66, 0.22)

The minimum acoustic delay necessary between the reference microphone and error microphone to fulfill the constraint of causality in the chosen configuration was computed. The minimum delay was a sum of the electronic delay, δ_E, and the propagation time from the secondary source to the error microphone, δ_A. With $\delta_E = 8 \pm 0.5$ ms and $\delta_A = 1.2$ ms, the necessary delay was $\geq 9.2 \pm 0.5$ ms. The component placements in Table 2 fulfill causality with $\delta_R = 10.5$ ms.

Experiments

ASC was performed with the components in the chosen causal configuration. System identification was performed for 35 seconds with the Galois noise power level set at 0.1. This vel was chosen after testing the coherence between the Galois t signal and the error microphone (Wright and Craig 1998). stem identification and the computation of the inverse signal re performed simultaneously with the filter weights reset to ro for each experiment. The sound from the error microphone d the behavior of the algorithm filter weights were recorded.

The reference microphone was then moved toward the error crophone such that the distance L_R and the delay δ_R grew orter. ASC was performed again and the same parameters were corded. Table 3 summarizes the experimental results.

ble 3. Results of Causality Experiments

Exp	L_R (m)	δ_R (ms)	Attenuation in 35 seconds
1	3.61	10.5 (causal)	18.4 dB
2	3.19	9.3 (near causal threshold)	17.2 dB
3	3.08	9.0 (non-causal)	6.1 dB
4	2.90	8.4 (non-causal)	0 dB

The computation of the electronic delay predicted a threshold for a causal configuration between 8.7 and 9.7 ms. The δ_R value of 9.3 ms in Experiment 2 is near that threshold. In that experiment, the error signal attenuation settled to slightly less than the value for the causal configuration within 35 seconds. Experiment 3, with a δ_R value of 9.0 ms, had much slower attenuation. The convergence of the control algorithm was so slow during the other experiments that no attenuation was achieved in the first 35 seconds.

Other experiments with convergence times of several minutes showed that the maximum attenuation reached in the causal configuration could eventually be reached in the non-causal configurations. This effect is not mentioned in the studies using random noise (Tseng et al. 1998, Janocha and Liu 1998) and is probably not possible with random noise.

The system identification filter convergence time was unaffected by causality, as predicted by Kong and Kuo (1999). However, the convergence of the control filter taps appeared to be affected by causality, with convergence occurring more slowly in the non-causal configurations.

Fig. 9 shows the attenuation at the frequency of the primary source over 35 seconds for Experiments 1 (causal) and 3 (non-causal). FFTs were performed for each second of error microphone data to isolate the primary source frequency from the Galois noise (recall that the calculation of the inverse signal and system identification are performed simultaneously). The increase in sound pressure level (SPL) at the beginning of the causal plot is caused by the secondary source. Its output briefly increases the SPL at the error microphone before the optimal inverse wave is computed. The brief but audible increase in SPL proved to be present whenever ASC was performed in a causal configuration and served as a convenient indicator of a configuration's causality.

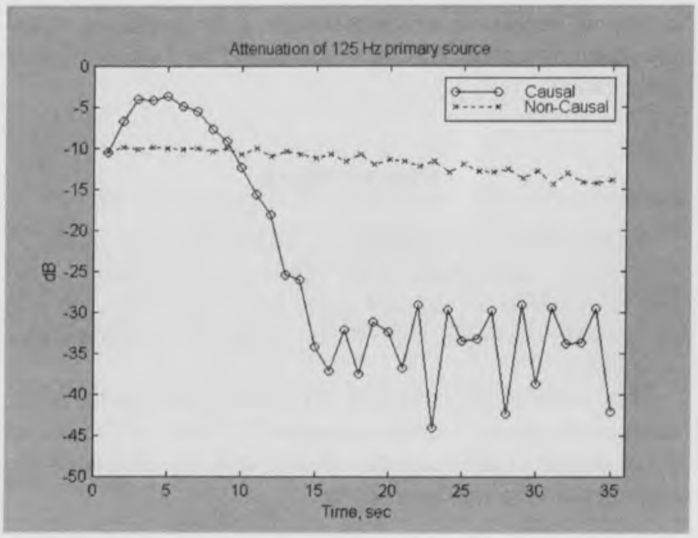

Fig. 9. Attenuation of 125 Hz primary source in causal and non-causal configurations.

The attenuation of the sound at the error microphone was much faster in the causal experiment and reached steady state in about 15 seconds. In the near-causal experiment, attenuation was nearly complete at 35 seconds. During the non-causal experiments, it took several minutes. This demonstrates that the computation time for the inverse signal in the non-causal system is much too long to be practical in most real-world applications. Since an ASC system must adapt to environmental changes, the very slow non-causal response may lead to poor performance or instability.

Conclusions

An ASC system was implemented to cancel a periodic disturbance in an enclosure. After computing causal positions for the components, and testing the system in that configuration, the reference microphone was moved into non-causal positions. The performance of the system in the non-causal configurations with increasing delays was recorded. Three potential contributions to knowledge are suggested by the results. First, a clear benefit of a causal ASC system for periodic disturbances was demonstrated: the much greater speed of computation of the inverse signal. This is contrary to previous comments about the irrelevance of causality when canceling periodic disturbances. Second, this study may be the first to verify, through experiment, the role of causality in the speed of algorithm convergence. The system identification filter convergence time was unaffected by causality, as predicted by Kong and Kuo (1999). However, the convergence of the control filter was affected by causality. Finally, a mark of system causality was found that may be previously unreported. An audible increase in SPL was present at the beginning of the experiment whenever ASC was performed in a clearly causal configuration.

Future Work

Experiments implementing an added electronic delay of the reference microphone signal rather than movement of the error microphone are already under way. They will eliminate any possible environmental effects caused by the microphone movements.

The cause of the increased SPL before convergence in the causal configuration will be determined. A limit will be placed on the output of the secondary source and any effects on the convergence time will be observed.

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Organometallic Ruthenium Complexes of Novel Thiosemicarbazones

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Abstract.—We present the preliminary results of a study of two novel thiosemicarbazones (TSCs) and their ruthenium complexes. The TSCs were prepared by refluxing thiosemicarbazide with 9-anthraldehyde or benzanthrone in ethanol for 3 hours. The metal complex of each ligand (complex **I** = $[(\eta^6\text{-}C_6H_6)Ru(9\text{-ant-TSC })(Cl)]Cl$ and complex **II** = $[(\eta^6\text{-}C_6H_6)Ru(benz\text{-}TSC)(Cl)]Cl)$ was prepared by refluxing the appropriate TSC with $[(\eta^6\text{-}C_6H_6)RuCl_2]_2$. The compounds were characterized using infrared, ultraviolet-visible, and NMR spectroscopy. Two different methods, the disk diffusion test and luminometry, were used to test the compounds against a variety of different bacterial strains for antibacterial activity. The most optimistic results were obtained for the 9-ant-TSC ligand, especially in relation to activity against Gram (+) bacteria. The metal complexes showed no measurable activity and further biological testing of the metal complexes is currently being conducted.

Key words:— thiosemicarbazones (TSCs), ruthenium complexes, bacterial strains.

Introduction

In modern chemotherapy the aim is to use a chemical compound that kills the offending organism or cells while having minimal impact of other cells. While most of the current chemotherapeutic agents are organic compounds, the use of inorganic (defined as metal-based) compounds has been growing in importance over the last 30 years. Indeed one of the most important anticancer drugs is *cis*-diamminedichloroplatinum (II), cisplatin, which is especially useful for the treatment of solid malignancies. Cisplatin however exhibits serious renal toxicity and has a narrow-spectrum of activity (being applicable to only a few tumor types). This has led to a continuing effort to design transition metal-based drugs that improve on spectrum activity and are also less toxic when compared to cisplatin. In other biomedical spheres, metal-based compounds are also gaining prominence. For example, the activity of organic antimicrobials such as chloroquine (a drug used to treat malaria) has been enhanced by binding the organic molecule to a ruthenium center. The Ru(II)-chloroquine complex is 2-5 fold more effective than chloroquine alone (Dyson and Allardyce 2001).

Organometallic compounds exhibit different ligand kinetics in solution to coordination complexes, which could prove advantageous in the design of inorganic drugs. Metallocenes of the type $M(\eta^5\text{-}C_5H_5)_2X_2$ (M = Ti, V, Nb, and Hf) have shown moderate anticancer behavior (Clarke et al. 1999). Ru(II) arene complexes of the type $[(\eta^6\text{-arene})Ru(XY)(Z)]^+$ are cytotoxic to cancer cells including cisplatin-resistant cell lines (Morris et al. 2001, Aird et al. 2002).

Thiosemicarbazones (TSCs, Fig. 1) have received considerable attention because they present a wide range of bioactivities: antibacterial, antifungal, anti-neoplastic, and antiviral (Beraldo and Gambino 2004). They thus represent an important class of compounds that have aroused considerable interest in chemistry and pharmacology. The properties of TSCs are usually affected by metal coordination. Although the free uncomplexed TSCs show interesting biological activity, in a number of cases the transition metal complexes showed greater biological activity (Pandeya and Dimmock 1993, Quiroga and Ranninger 2004). This can be related to increased lipophilicity which controls entry into the cell. It has also been proposed that the mechanism of antibacterial activity involves electron transfer and/or oxidative stress (Kovacic et al. 1989). Other positive effects of metal coordination include potentially significant reduction of drug resistance and side effects (West et al 1991). It is conceivable that coordination to the metal serves to activate the biologically active TSC ligand. Also, the metal complex can exhibit different bioactivities than the free TSCs. By coupling the TSCs with the organometallic Ru(II) group, it may be possible to synthesize new complexes that have good biological activity due to the synergistic effectiveness.

In this paper we report the synthesis of organometallic ruthenium complexes with novel thiosemicarbazone ligands (Fig. 2) and describe their characterization and antimicrobial activity.

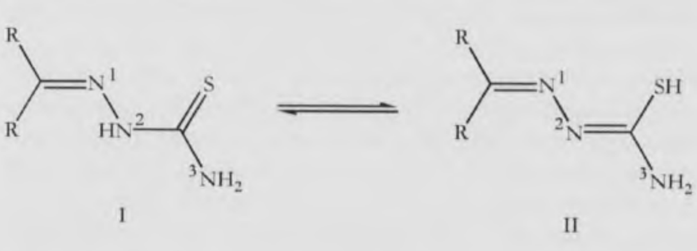
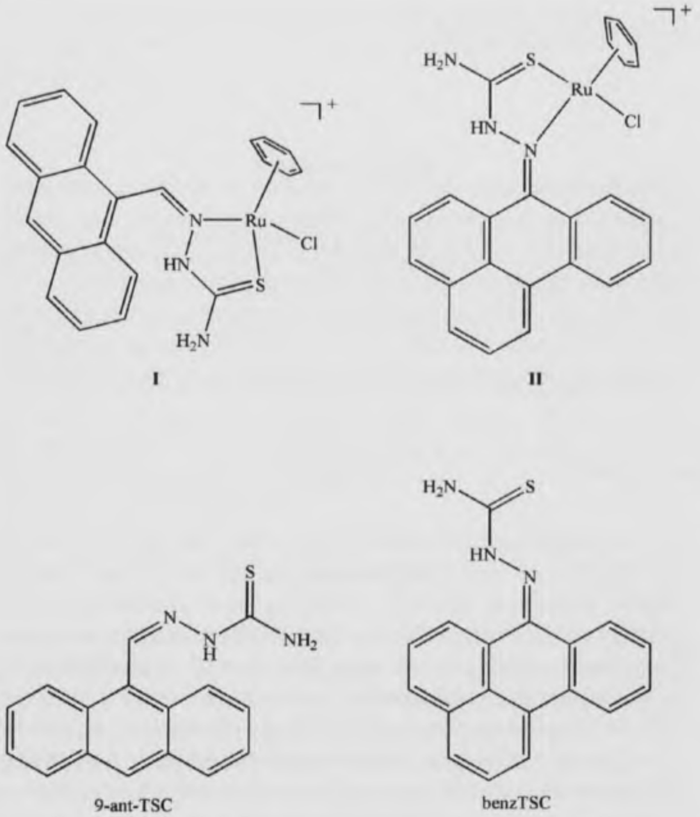


Fig. 1: Thiosemicarbazones showing thione (I) $\rightleftharpoons$ thiol (II) tautomerism



I

II

9-ant-TSC

benzTSC

Fig. 2: Structures of the thiosemicarbazones and complexes

Materials and Methods

Analytical or reagent grade chemicals were used throughout. Hydrated RuCl$_3$ was purchased from Strem (Newburyport, MA) and used as received. All other chemicals were obtained from Sigma-Aldrich (St. Louis, MO) or other commercial vendors and used as received. The luminometry studies were done using a BacTiter-Glo™ Microbial Cell Viability Assay (Promega, Madison, WI) on a Hidek Bioscan single-tube combination liquid scintillation counter and luminometer. Microanalyses (C, H, N) were performed by Desert Analytics (Tucson, AZ). ^{1}H and ^{13}C NMR spectra were recorded on a Bruker spectrometer operating at 300 MHz in chloroform-d_3 or dichloromethane-d_2. The chemical shifts were measured in ppm relative to TMS. IR spectra were recorded in KBr discs in the range 4000 – 450 cm^{-1} on a Mattson Satellite FTIR spectrophotometer, and the electronic spectra were recorded on an Agilent 8453 spectrophotometer in the range 190-1100 nm using quartz cuvettes. Melting points were determined in open capillaries and are uncorrected. The precursor complex $[(\eta^6\text{-benzene})RuCl_2]_2$ was prepared following the method of Bennett and Smith (1974).

Preparation of TSC.—9-anthraldehyde (or benzanthrone) was reacted with an equimolar amount of thiosemicarbazide in refluxing ethanol for 3 hours. The orange precipitate (yellow for benzanthrone) that formed was filtered, washed with copious amounts of ethanol, then ether, and dried at the vacuum pump. The yield was 82.7% for 9-ant-TSC and 43.4% for benz-TSC.

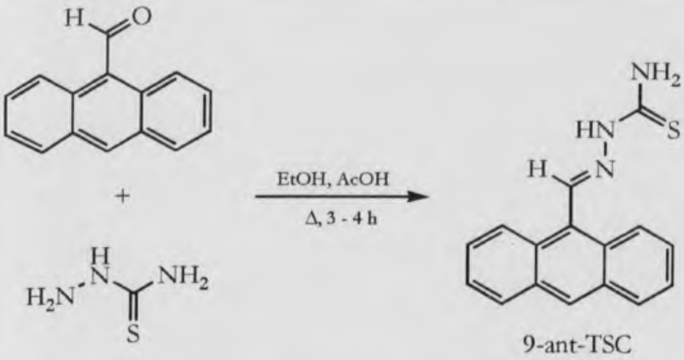
Preparation of complexes.—The complexes were prepared as follows: $[(\eta^6\text{-}C_6H_6)RuCl_2]_2$ (300 mg, 0.600 mmol) and the TSC (1.20 mmol) were dissolved in 40 mL of degassed toluene and the solution was stirred during reflux in an inert atmosphere for 4 hours. The brown precipitate (black for benzanthrone) was filtered, washed with copious amounts of pentane, and dried at the vacuum pump. The yield was 73.5% for **I** and 53.5% for **II**.

Antibacterial activity screens.—The ligands were screened against standard bacterial strains of *Staphylococcus aureus*, *Escherichia coli*, *Bacillus cereus*, *Enterococcus faecalis*, *Salmonella typhimurium*, and *Proteus vulgaris*. The antibacterial activity was studied by luminometry for the first 2 bacterial strains and by the disk diffusion method for the latter 4. For the luminescence assay, the bacteria were incubated in Mueller-Hinton broth at 37 °C for 20 hours. The cultures were diluted 1:100 in fresh M-H broth, and then 245 μl of the appropriate culture was added to 12 wells of a 24-well plate. Five microliters of the appropriate drug, 9-ant-TSC, benz-TSC, or chloramphenicol (the standard), all at 10^{-5} M were added to 4 wells of each culture, and the cultures were grown at 37 °C for an additional 5 hours. Luminescence was then measured as counts per second (CPS). For the disk diffusion test, small 7-mm-diameter circles of filter paper (P5) were saturated with 20 μl of the test solutions (10^{-3} M in DMSO). The disks were placed on agar plates that were inoculated with the bacterial cultures, and the plates were incubated at 37 °C for 20 hours. Chloramphenicol was used as a standard. Following incubation, each plate was checked for zones of inhibition (measured in mm). Antibacterial activity screens for **I** and **II** using disk diffusion were also conducted, but the results were inconclusive.

Results and Discussion

Syntheses.—Figure 2 shows the ligands and complexes prepared in this study. The ligands were made by condensing the aldehyde (9-anthraldehyde) or ketone (benzanthrone) with thiosemicarbazide as shown in Scheme 1 for 9-ant-TSC. The starting ruthenium dimer $[(\eta^6\text{-benzene})RuCl_2]_2$ was made following the method of Bennett and Smith (1974) by heating at reflux a methanolic solution of $RuCl_3.xH_2O$ with 1,3- or 1,4-cyclohexadiene. From this precursor compound, target complexes (Fig. 2) were synthesized according to Scheme 2. Generally, the ruthenium dimer is reacted with two equivalents of the ligand in toluene at elevated temperatures. The complexes precipitated directly from the reaction solution on cooling to ambient temperature.

Melting Points.—Shown in Table 1 are the melting points for the TSCs and their complexes. The TSCs melted over a narrow

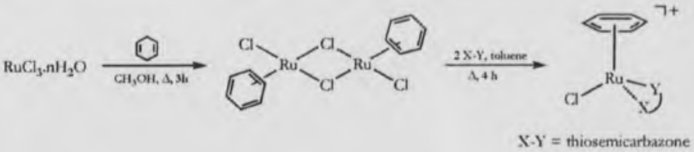


Scheme 1. Synthesis of the thiosemicarbazones

range (1.7-2.8°C), indicating relative purity of these compounds (as a melting range of about 2°C is generally considered normal for pure compounds). Neither complex showed any melting behavior below 350°C.

NMR Spectra.—The NMR data for the two ligands are consistent with the proposed structures. The 1H NMR spectrum of 9-ant-TSC in CD_2Cl_2 shows two singlets at 9.00 ppm and 9.74 ppm that are assigned to the $HC=N^1$ and HN^2 protons respectively. The azomethinic (HN^2) proton might be expected to show up at higher frequencies (11-14 ppm) but is shifted upfield in our spectrum. This is not unusual however as in a number of thiosemicarbazones the signal ascribed to this group show up in the range 8 – 11 ppm. The thione form of the ligand exists in solution as well as there is no peak at ~ 4.00 ppm in the ^1H NMR which corresponds to –SH. It has been reported that this resonance typically appears at ~ 4.0 ppm (Singh et al 2005). The aromatic resonances for both ligands appear at the expected positions (7.5 – 8.5 ppm). The ^{13}C NMR spectra show a high frequency signal at 180.4 ppm for 9-ant-TSC and 184.1 ppm for benz-TSC. These are assigned to the -C=S moiety. The -C=N signal occurs at 141.4 ppm for 9-ant-TSC and 136.4 ppm for benz-TSC. In both compounds there is a cluster of peaks in the 120 – 130 ppm range, which is typical for aromatic compounds.

Electronic Spectra.—UV-Vis electronic spectra of both TSCs and complexes **I** and **II** were analyzed using 10^{-5} M DMSO solutions in the 190-900 nm region. Data from the analysis is presented in Table 1. The bands occurring at <300 nm can most likely be attributed to π-π* transitions, while the bands between 350-400 nm are most likely due to n-π* transitions of the thiosemicarbazone ligands. While the major absorbance wavelengths for the TSCs didn't change much (0-10 nm) on formation of their respective complexes, there were hypochromic changes at each peak (0.02-0.73). This clearly indicates electron density movement between the ligand and the metal. The π-π* transitions also show small changes in molar absorptivity (a general decrease of about 0.05 from the TSC to the complex), which is likely a result of a weakening of the C=S bond. This indicates that the thione group is probably involved in metal coordination.



Scheme 2. Synthesis of the metal complexes

Vibrational Spectra.—The infrared spectral data for the ligands and complexes are shown in Table 2. Despite the fact that the HN-C=S group can undergo thione-thiol tautomerization (Fig. 1), the lack of a band around 2570 cm^{-1} (characteristic of S-H bonds) indicates that the TSCs are in the thione form in the solid state. The presence of bands around 3150 cm^{-1} indicates the presence of the –NH moiety, which further supports the thione coordination. These bands shift somewhat from the TSC to the related metal complex, possibly indicating involvement of the azomethinic nitrogen in complex formation. However, the N-N stretching (which occurs around 1015 cm^{-1}) does not vary much between TSC and the metal complex (2-7 $cm-^{1}$), which leads to the conclusion that the azomethinic nitrogen is not involved in formation of the complexes. Instead, the C=N band experiences a greater degree of shift between the TSC and metal complex (~14 cm^{-1}). Thus, it is concluded that the imine nitrogen is involved in metal coordination. The C=S bands for the TSCs is also shifted upon complex formation (from 827 to 809 cm^{-1} for 9-ant-TSC and from 1279 to 1297 cm^{-1} for benz-TSC), further indicating thione involvement in binding to the metal. The NH_2 bands show some amount of shift (as high as 24 cm^{-1}), but this occurred as a consequence of the coordination of the $-S=C-NH_2$ to the metal.

Antimicrobial Studies.—*In vitro* antibacterial properties of the TSCs are shown in Tables 3 and 4. Both TSCs showed little or no effect on Gram (-) bacteria, as determined from both the luminometry and disk-diffusion tests. In fact, no activity was seen for either TSC against the two Gram (-) bacteria in the disk-diffusion test, and both were about 4 times less effective against *E. coli* than against *S. aureus* in the luminometry assay. Benz-TSC also showed little effect against Gram (+) bacteria in the disk-diffusion test (0.5 mm), but had slightly higher effectiveness against *S. aureus* in comparison with 9-ant-TSC. Optimistic results for the 9-ant-TSC, especially in relation to Gram (+) bacteria, were obtained from the disk-diffusion test. Against *E. faecalis,* 9-ant-TSC was even more active than the chloramphenicol standard (bacteriostatic diameter of 10 mm, in comparison with 4 mm for the standard). Further studies are being done on this compound in an effort to establish minimum inhibitory concentration (MIC) values. Further biological

Table 1. Solution Electronic Spectral Data (nm) for Selected Thiosemicarbazones and their Ruthenium Arene Complexes (10^{-5} M DMSO solutions)

Compounds	Wavelength (molar absorptivity)			
9-ant-TSC[a] (208.0 – 210.8°C)[b]	231 (4.53)[c]	264 (4.68)	373 (3.76)	392 (3.84)
I (>350°C)	231 (4.49)	263 (4.56)	375 (3.72)	393 (3.82)
benz-TSC (154.1 – 155.8°C)	231 (4.49)	266 (4.15)	276 (4.15)	393 (4.05)
II (>350°C)	231 (4.54)	262 (4.10)	398 (3.31)	405 (3.32)

[a] =1 x 10^{-5} M in DMSO; [b] = Melting Points; [c] = log (ε)

Table 2. Selected Vibrational Bands (cm^{-1}) of Thiosemicarbazones and Complexes **I** and **II**

Assignment	9-ant-TSC	**I**	benz-TSC	**II**
υ (C=N)	1600	1614	1620	1622
υ (N-N)	1019	1017	1001	1008 (w)
υ (C=S)	1286	1285	1279	1297 (w)
	827	809 (w)	842	843
υ (NH)	3157	3146	3179	3168
υ (NH_2)	3440	3417	3385	3386
	3263	3287	3264	3283

w = weak

Table 3. Antibacterial Activity of the Thiosemicarbazones and Complexes **I** and **II** (10^{-3} M) - Bacteriostatic Diameter (mm)

Compound	Bacterial strain			
	Gram (+)		Gram (-)	
	Bacillus cereus	*Enterococcus faecalis*	*Proteus vulgaris*	*Salmonella typhimurium*
9-ant-TSC	3	10	0	0
I	-[a]	-	-	-
benz-TSC	0.5	0.5	0	0
II	-	-	-	-
Chloramphenicol	9.5	4	4.5	8.5

[a] = Results inconclusive

Table 4. Antibacterial Activity of the Thiosemicarbazones and Complexes **I** and **II** (10^{-3} M) – Luminescence (CPS)

Compound	Bacterial strain	
	Gram (+) *Staphylococcus aureus*	Gram (-) *Escherichia coli*
9-ant-TSC	1.30 x 10^7	5.72 x 10^7
I	-[a]	-
Benz-TSC	1.18 x 10^7	5.72 x 10^7
II	-	-
Chloramphenicol	3.22 x 10^6	3.39 x 10^6
Negative Control (Background)	1.13 x 10^5	1.13 x 10^5

[a] = Results inconclusive

testing of the metal complexes is being conducted, but the preliminary results have been inconclusive.

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Five Years of Change in an Old-Growth Pine-Hardwood Remnant in Ashley County, Arkansas

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Abstract.—The Levi Wilcoxon Demonstration Forest near Hamburg, Arkansas is an industrially-owned remnant of old-growth pine and hardwoods. Some of the loblolly (*Pinus taeda* L.) and shortleaf (*Pinus echinata* Mill.) pine in this stand are over 200 years old, and numerous individuals exceed 90 cm in diameter and 30 m in height. A 2000 survey of a portion of this tract found that 27 tree species contributed an average of 387.5 live stems/ha and 31.8 m^2/ha of basal area. An inventory of the same plots in 2006 yielded noticeable declines in density (now down to 342.5 stems/ha) and basal area (now 28.2 m^2/ha). Much of this loss came in the aftermath of a windstorm in May 2003, which felled a number of overstory pines. Loblolly pine decreased from 49.6 stems/ha and 13.2 m^2/ha in 2000 to 42.1 trees/ha and 11.2 m^2/ha in 2006, while shortleaf pine declined from 21.7 trees/ha and 5.0 m^2/ha to 14.6 trees/ha and 3.5 m^2/ha. Further pine mortality came from smaller-scale windthrow, lightning, and bark beetle infestations. Some hardwoods were also toppled by storms or crushed by falling trees, but most appear to have succumbed to drought, competition, and salvage logging. However, hardwood basal area remained virtually unchanged over this period, signifying adequate diameter growth and midstory recruitment. In particular, shade-tolerant hardwood species showed notable gains. Even though most overstory pines currently appear healthy, natural catastrophes and the lack of new canopy recruits may eradicate virtually all pines from this stand within 30 to 50 years.

Key words:—Levi Wilcoxon Demonstration Forest, loblolly pine, natural disturbance, shortleaf pine, windthrow.

Introduction

Very few stands of pine-dominated old-growth remain in the Upper West Gulf Coastal Plain of Arkansas. Extensive lumbering and agricultural clearing, coupled with other large-scale catastrophic disturbances, have converted millions of hectares of virgin forest into stands of young timber, pastureland, row crops, and commercial and residential developments. The remaining old-growth is found in a few small tracts that escaped conversion. Most prominent of the south Arkansas pine-hardwood old-growth sites are the "Lost Forty" in Calhoun County (Heitzman et al. 2004) and the Levi Wilcoxon Demonstration Forest (LWDF) in Ashley County (Georgia-Pacific, n.d., Allen 1985, Bragg 2004a), both of which are currently owned by large companies (Potlatch Corporation and Plum Creek Timber Company, respectively).

Although these sites currently receive some degree of protection from perturbations, they are still subject to forest succession and certain disturbances. For example, the LWDF is periodically salvaged to remove dead and dying pines. These mechanisms of change, coupled with decades of fire exclusion, forest fragmentation, and invasion by exotic species, have noticeably altered the composition, structure, and dynamics of old forest remnants across the South (e.g., Jones et al. 1981, Shelton and Cain 1999, Harrington et al. 2000, Bragg 2002, Harcombe et al. 2002).

The LWDF was ecologically described using field data collected between 2000 and 2003 (Bragg 2004a). Since this initial measurement, the stand has been affected by both catastrophic (primarily from a single windstorm and the resultant salvage) and individualistic (e.g., lightning strikes, beetle kills, drought) mortality of the mid- and overstory trees. The preservation and long-term management of the LWDF depends on our ability to anticipate change, which in turn requires a better understanding of short-term stand dynamics.

Materials and Methods

Site Description.—The LWDF (Fig. 1) is located in Ashley County, approximately 6 km south of Hamburg, Arkansas (Fig. 2). Most of the LWDF is gently rolling (0 to 2% slopes) and dominated by Calloway and Grenada silt loams (Glossic Fragiudalfs) on the higher ground and Arkabutla silt loams (Aeric Fluvaquents) along minor stream drainages (Gill et al. 1979). The mean elevation of the LWDF is 45 m, and the stand is located on a landform identified by Saucier (1974) as the Prairie Terrace Formation. The abundantly distributed "pimple" or "prairie" mounds throughout the stand provide further evidence of its association with the Pleistocene-period Prairie Terrace. The study site averages 140 cm of precipitation and 200 to 225 frost-free days annually (Gill et al. 1979).

Historically, the presettlement upland vegetation of southern Arkansas was pine, pine-oak, and oak-hickory-gum forests, pine-oak-hickory woodlands, and scattered prairies (Vanatta et al. 1916, Turner 1937, Bragg 2002 2003). When first reserved by the Crossett Lumber Company, the LWDF was overwhelmingly pine-dominated (Anonymous 1948). Over the decades, mortality and salvage removed many of the large loblolly (*Pinus taeda* L.) and shortleaf (*Pinus echinata* Mill.) pines (Bragg 2004a).

Fig. 1. View of a portion of the Levi Wilcoxon Demonstration Forest (LWDF) looking north towards Hamburg, Arkansas, from the LWDF parking area near the corner of Highways 425 and 52. The stand is dominated by supercanopy pines with lower canopy levels comprised almost entirely of a variety of hardwood species.

Hardwoods have grown increasingly numerous, although they do not yet constitute a majority of stand basal area.

Fire, glaze, and windthrow were the primary presettlement disturbances of the study area, with insect and disease outbreaks, lightning, and drought also impacting forested areas (Turner 1937, Bragg 2002). Frequent fires helped maintain relatively open understories in upland forests, conditions that changed as forestry and fire control were implemented by the 1930s. Logging and agriculture spread rapidly across the region beginning in the mid-1800s. However, most farming operations failed, and much of the cleared land in Ashley County quickly reverted back to forest (Vanatta et al. 1916). The post-fire control forests that seeded into the cut-over lands, abandoned farms, and neglected pastures were considerably denser, younger, and more even-aged than the original forests, with greater numbers of briars, vines, shrubs, and shade-tolerant tree species in the understory (Bragg 2002). Over time, stand composition of the old-growth LWDF remnant has also shifted toward a dense, woody, shade-tolerant understory.

Mid- and Overstory Remeasurement.—To ensure continuity, this paper will follow the same live tree sampling protocols of Bragg (2004a). Only the 6-ha reserved area of the LWDF was re-evaluated for overstory compositional and structural dynamics, using the same twenty-four 0.1-ha circular plots (17.84 m radius) established in the summer of 2000. In the original study, 8 plots were established on every transect, and transects were located 40 m from the next to avoid overlap. Plot centers were spaced 100 m apart along each transect, and every live tree > 9 cm in diameter at breast height (DBH) was tallied for species (Table 1) and DBH (measured to the nearest 0.25 cm using a diameter tape).

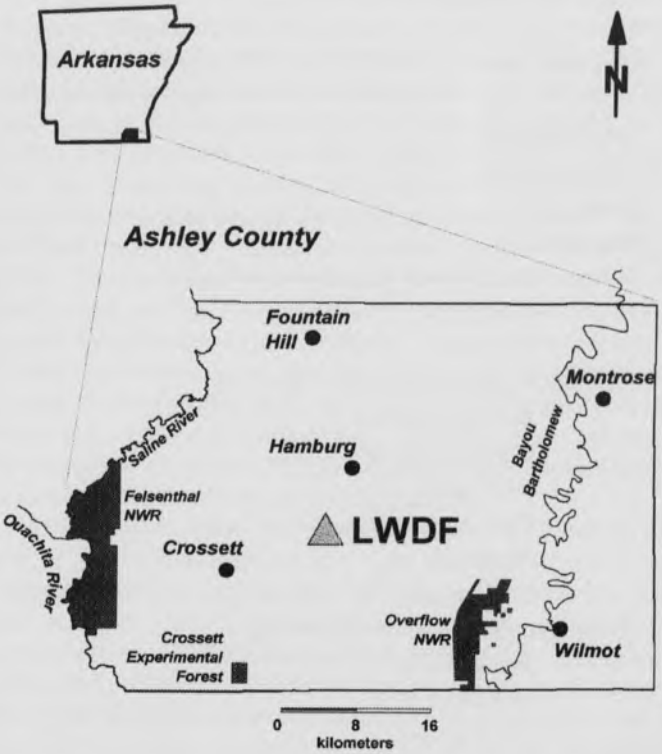

Fig. 2. Map of Ashley County, Arkansas, showing the location of the Levi Wilcoxon Demonstration Forest (LWDF) relative to other geographic features.

Species abundances and stand stocking (number of trees and basal area per hectare) were derived from the plot-level information. The analysis of species dynamics in this paper sometimes includes the use of functional groups rather than individual taxa. Occasionally, this aggregation was used to facilitate the graphical display of data. However, in the case of the red oak subgroup, the lumping of southern red oak (*Quercus falcata* Michx.), cherrybark oak (*Quercus pagoda* Raf.), and black oak (*Quercus velutina* Lam.) was done to avoid misidentification of these visually similar oaks in a less-than-ideal dormant leaf-off state.

Select individuals from the entire 20+ ha LWDF were incorporated in the sections of this paper that refer to tree size or age. Tree heights were originally measured using a cloth tape and percent-baseline clinometer using the tangent method. The 2006 heights were determined with a Laser Technology Impulse 200LR^{TM} laser rangefinder and the sine method of height calculation (Blozan 2004, Bragg, in press). Age data for the LWDF were supplemented by ring counts made at stump height (approximately 45 to 60 cm above groundline) on four recently felled snags. In addition, Dr. Brian R. Lockhart of

Table 1. Scientific and common names of mid- and overstory tree species mentioned in this paper. Species are grouped according tc the categories used in Table 2 and Fig. 3.

Common name	Scientific name[a]
Shortleaf pine	*Pinus echinata* Mill.
Loblolly pine	*Pinus taeda* L.
Sweetgum	*Liquidambar styraciflua* L.
Blackgum	*Nyssa sylvatica* L.
White oaks (grouped from the following)	*Quercus* spp.
White oak	*Quercus alba* L.
Post oak	*Quercus stellata* Wang.
Red oaks (grouped from the following)	*Quercus* spp.
Southern red oak	*Quercus falcata* Michx.
Cherrybark oak	*Quercus pagoda* Raf.
Black oak	*Quercus velutina* Lam.
Red oak subgroup	*Quercus falcata* + *Quercus pagoda* + *Quercus velutina*
Water oak	*Quercus nigra* L.
Willow oak	*Quercus phellos* L.
Elms (grouped from the following)	*Ulmus* spp.
Winged elm	*Ulmus alata* Michx.
American elm	*Ulmus americana* L.
Slippery elm	*Ulmus rubra* Muhl.
Other hardwoods (grouped from the following)	--
Red maple	*Acer rubrum* L.
American hornbeam	*Carpinus caroliniana* Walt.
Bitternut hickory	*Carya cordiformis* (Wang.) K. Koch
Mockernut hickory	*Carya tomentosa* Nutt.
Sugarberry	*Celtis laevigata* Willd.
Flowering dogwood	*Cornus florida* L.
Persimmon	*Diospyros virginiana* L.
Green ash	*Fraxinus pennsylvanica* Marsh.
American holly	*Ilex opaca* Ait.
Red mulberry	*Morus rubra* L.
Eastern hophornbeam	*Ostrya virginiana* (Mill.) Koch
Black cherry	*Prunus serotina* Ehrh.
Sassafras	*Sassafras albidum* (Nutt.) Nees.

[a] Species nomenclature from Harlow et al. (1979), Smith (1988), and Moore (1999).

the USDA Forest Service also contributed stump dimensions and ring counts for the LWDF collected by students from 36 pines salvaged following an insect outbreak and other mortality events in the latter half of the 1980s.

Results and Discussion

In the 5 years since the original forest survey of the LWDF was completed, a number of events affected stand structure and composition. A severe straight-line windstorm struck the area in May of 2003, felling many of the larger pines. The LWDF has been isolated in recent years as adjacent mature forests have been clearcut, accentuating storm loss-mediated structural changes. Thus, as the winds of this storm swept across the area from west-to-east, there were few obstructions to dissipate their energy before they struck the north-south oriented LWDF (Bragg 2004a). In addition to direct mortality from this particular windstorm, insects (primarily bark beetles (*Dendroctonus* spp.)) attracted to the fallen pines killed other nearby pines in the months following the storm. Other isolated storms produced further overstory losses via windthrow, lightning, and post-storm insect attack. Not surprisingly, overall tree density in the LWDF decreased from 387.5 stems/ha in the fall of 2000 to 342.5 trees/ha by February of 2006. Over this same time period, average basal area in the LWDF declined from the 31.8 m^2/ha initially reported to 28.2 m^2/ha in 2006, a reduction of 11%.

Species Composition Trends.—Table 2 provides a comparison of the species composition between the first inventory and this effort. Bragg (2004a) reported 27 tree species on the study plots in 2000, but the 2006 remeasurement yielded only 24. This discrepancy is not due to identification errors, but rather to the loss of a handful of tree-sized specimens on the plots. The three taxa absent from the 2006 inventory (persimmon (*Diospyros virginiana* L.), willow oak (*Quercus phellos* L.), and American elm (*Ulmus americana* L.)) were represented by 1, 4, and 1 individuals, respectively, in the original survey. Though specifically searched for, these individuals were not found and appear to have perished from drought, salvage logging, or as in the case of one willow oak, from being crushed by a falling tree. Another species, sugarberry (*Celtis laevigata* Willd.), appears poised to join the ranks of the missing taxa, as the single individual noted in 2000 was barely clinging to life after a large white oak (*Quercus alba* L.) fell onto it in 2005. Such is the ecological role of uncommon understory species—they are noticeably more volatile, and thus can have a dramatic impact on taxonomic richness. However, their disappearance from the study plots does not mean that these species vanished from the LWDF, as all of these species are still found in the forest encompassing the reserved area.

The absolute values and relative dominance of species fluctuated over the last 5 years (Table 2). The pines declined in prominence, especially following the windstorm. The most abundant taxon in the 2000 inventory, sweetgum (*Liquidambar styraciflua* L.), also decreased appreciably, losing over 16% of its number, primarily in the smallest diameter classes. Other taxa experiencing substantial (>10%) decreases included white oak (down 19%), post oak (*Quercus stellata* Wang., -17%), the red oak subgroup (-10%), slippery elm (*Ulmus rubra* Muhl., -36%), mockernut hickory (*Carya tomentosa* Nutt., -18%), flowering dogwood (*Cornus florida* L., -35%), red mulberry (*Morus rubra* L., -20%), and black cherry (*Prunus serotina* Ehrh., -22%). Most of these were in subordinate canopy positions and did not directly suffer from the severe winds or lightning faced by the emergent pines. Rather, falling trees, post-storm salvage operations, moisture extremes, light competition, and decay coupled with wind or glaze have killed hardwoods throughout the LWDF. Flowering dogwood, for instance, is particularly drought sensitive and died in large numbers during prolonged dryness in 2000 and 2001.

Some species increased their abundance over the last 5 years. Blackgum (*Nyssa sylvatica* L., up 7%), winged elm (*Ulmus alata* Michx., +15%), red maple (*Acer rubrum* L., +21%), bitternut hickory (*Carya cordiformis* (Wang.) K. Koch, +50%), and eastern hophornbeam (*Ostrya virginiana* (Mill.) Koch, +198%) all produced noticeable density increases. However, the large percentage growth for bitternut hickory and eastern hophornbeam does not translate to great numbers of new stems, as these were very uncommon species when inventoried in 2000. The increasers weathered the drought and storms of the last 5 years, and their higher shade tolerance allows for them to persist longer under a closed canopy. They are also capable of exploiting relatively small canopy gaps produced by disturbance events, so long as they can survive the proliferation of woody vines (e.g., *Vitis* spp., *Smilax* spp., *Lonicera* spp., *Gelsemium sempervirens* (L.) Jaume St.-Hil., *Lygodium japonicum* (Thunb. ex. Murr.) Sw.) following overstory removal.

Changes in Pine Dominance.—Pine dominance in the LWDF has varied considerably over the last 50 years and especially since the initial 2000 inventory. In 1948, a picture was taken of Levi Wilcoxon standing next to a sign at the entrance to the LWDF (Johnson et al. 1994, p. 58). Though not particularly detailed, the sign had basic statistics on the natural area, including that there were about 193 trees/ha on this site 15 cm DBH or greater, most (if not all) of which were loblolly or shortleaf pine. Although no longer the most common species, loblolly pine still dominates the stand, contributing 42.1 stems/ha and 11.2 m^2/ha to the stand totals (approximately 12% and 40%, respectively). The change in density and basal area for loblolly pine represent decreases of 15% and 16%, respectively, over the last 5 years. Shortleaf pine has declined even more precipitously since 2000, losing 33% and 30% of its density and basal area totals, respectively (Table 2). Pine mortality in some of the largest diameter classes was the primary cause of the declines in stand density and basal area (Fig. 3).

The rapid decline of the LWDF pine overstory parallels that of a nearby old forest. The tree component of the Reynolds Research Natural Area (RRNA) on the Crossett Experimental Forest south of Crossett, Arkansas has been monitored since the late 1930s (e.g., Cain and Shelton 1996, Shelton and Cain 1999). From 1935 to 1965 (Fig. 4), loblolly and shortleaf pine basal area increased from 13 m^2/ha to between 21 and 23 m^2/ha and was sustained at this level for the next 30 years (Cain and Shelton 1996, Shelton and Cain 1999). During this period, pine basal area was maintained by aggregate growth slightly higher or equal to mortality losses, not by the recruitment of new pines into the canopy. Pine abundance in the RRNA eventually dropped to the point that mortality losses could not be made up for by growth, and thus its basal area fell rapidly—by 2000, only 18.7 m^2/ha of live pines remained (Bragg 2002). Almost 6 years later, a follow-up cruise noted a further reduction in pine basal area on the RRNA to approximately 14 m^2/ha. This decline is also being experienced for most of the same reasons

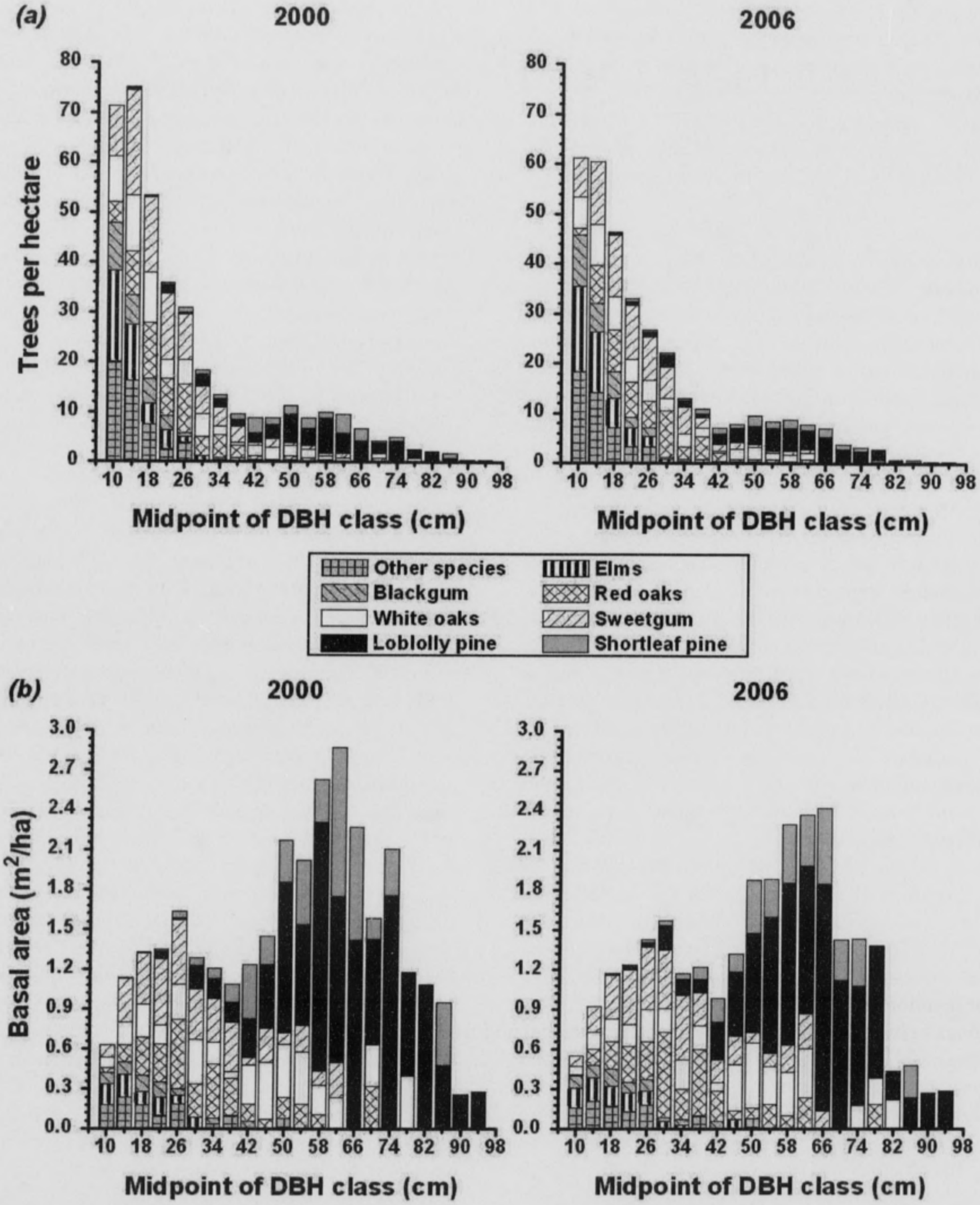

Fig. 3. Density *(a)* and basal area *(b)* distributions by size class of major species groups in the LWDF sampled in 2000 and 2006.

Table 2. Initial (Bragg 2004a) versus current mid- and overstory inventories of live trees in the reserved portion of the Levi Wilcoxon Demonstration Forest in Ashley County, Arkansas.

	Density (trees/ha)			Basal area (m²/ha)			Mean DBH (cm)		
Species/species group [a]	2000	2006	Diff. [b] (%)	2000	2006	Diff. (%)	2000	2006	Diff. (%)
Shortleaf pine	21.67	14.58	-33	5.02	3.51	-30	52.4	53.5	2
Loblolly pine	49.58	42.08	-15	13.23	11.15	-16	55.9	56.0	0
Sweetgum	85.83	71.67	-16	3.83	3.82	0	21.5	23.6	9
Blackgum	25.00	26.67	7	0.55	0.61	11	15.5	15.7	1
White oaks									
White oak	55.00	44.58	-19	3.56	3.38	-5	24.6	26.8	9
Post oak	2.50	2.08	-17	0.59	0.43	-27	49.9	44.5	-11
Red oaks									
Red oak subgroup [c]	47.50	42.92	-10	2.41	2.47	2	22.2	23.5	6
Water oak	8.75	7.92	-9	0.75	0.91	21	28.8	34.2	19
Willow oak	1.67	0.00	-100	0.06	0.00	-100	20.1	--	n/a [d]
Elms									
Winged elm	33.75	38.75	15	0.64	0.83	30	14.7	15.3	4
American elm	0.42	0.00	-100	0.01	0.00	-100	10.9	--	n/a
Slippery elm	5.83	3.75	-36	0.08	0.06	-25	12.7	13.3	5
Other hardwoods									
Red maple	13.75	16.67	21	0.18	0.22	22	12.6	12.5	-1
American hornbeam	1.25	1.25	0	0.02	0.03	50	14.4	16.1	12
Bitternut hickory	1.67	2.50	50	0.11	0.15	36	23.8	22.7	-5
Mockernut hickory	4.58	3.75	-18	0.20	0.17	-15	21.6	22.4	4
Sugarberry	0.42	0.42	0	0.01	0.01	0	12.4	12.7	2
Flowering dogwood	10.83	7.08	-35	0.14	0.09	-36	12.5	12.7	2
Persimmon	0.42	0.00	-100	0.01	0.00	-100	9.4	--	n/a
Green ash	0.42	0.42	0	0.01	0.01	0	9.9	10.7	8
American holly	0.83	0.83	0	0.01	0.01	0	13.3	14.5	9
Red mulberry	2.08	1.67	-20	0.05	0.06	20	16.7	21.2	27
Eastern hophornbeam	0.42	1.25	198	0.01	0.01	0	9.9	11.4	15
Black cherry	7.50	5.83	-22	0.19	0.13	-32	17.3	15.9	-8
Sassafras	5.83	5.83	0	0.13	0.15	15	15.9	17.4	9
TOTALS:	387.50	342.50	-12	31.80	28.21	-11			

[a] See Table 1 for taxonomic grouping details.

[b] Percent difference between 2000 and 2006 inventories, calculated from: ([2006 – 2000] / 2000) * 100.

[c] Due to the difficulty in differentiating southern red oak, cherrybark oak, and black oak in the dormant (leaf-off) period, these species were grouped into the "Red oak subgroup".

[d] Diameter change is undefined, therefore there is no applicable (n/a) measure of percent change in this case.

as in the LWDF (Fig. 4). However, the RRNA is not regularly salvaged to remove dead and dying pines, so this preserve has considerably more large woody debris than the LWDF.

Stand Structural Change.—Diameter class distributions in the LWDF were comparable to those reported in 2000 (Fig. 3). Hardwood dominance increased in virtually all size classes, and this trend will continue into the foreseeable future unless an adequately severe and extensive disturbance opens the overstory and reduces the duff enough to permit large-scale pine regeneration and canopy recruitment. In particular, large pines and shade-intolerant hardwoods—such as sweetgum and the red oak subgroup—will continue to decrease, while more shade-tolerant hardwoods (e.g., flowering dogwood, winged elm, blackgum, and hickory) occupy an increasing proportion

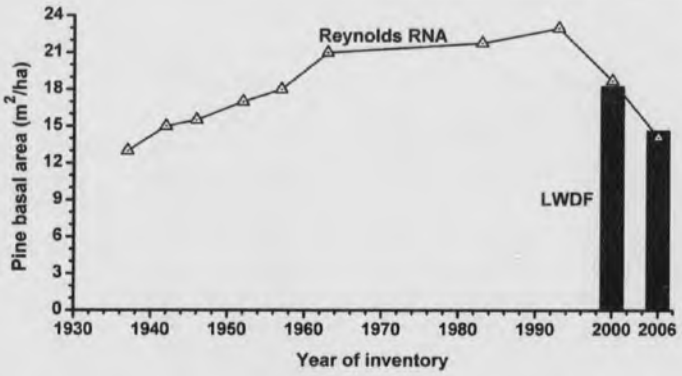

Fig. 4. Long-term trend of overstory pine basal area at the Reynolds RNA (line) and the LWDF (bars). Data compiled from Cain and Shelton (1996), Bragg (2002), and this study.

of the small- to medium-sized diameter classes.

Storm losses were not evenly distributed throughout the LWDF. A relatively large swath of damage perhaps a hectare in extent appeared in the middle of the reserved area. This, coupled with insect-related post-storm mortality, caused a considerable gap in the canopy to form. However, rather than providing an opportunity for the reestablishment of the current overstory species, the existing understory of American beauty berry (*Callicarpa americana* L.), woody vines (e.g., *Vitis* spp.), graminoids, and other exploiters of forest openings (e.g., *Rubus* spp.) quickly and almost completely occupied the larger openings. Shade-tolerant hardwood regeneration should gradually emerge from these thickets, but it is highly unlikely that pine seedlings will persist in the dense undergrowth long enough to ascend into the canopy. Few of the smaller gaps caused by individual trees being killed by storms or their aftermath provide adequate space to permit seedlings to reach the overstory, regardless of their shade tolerance. The limited amount of resources freed by these minor gaps will be appropriated by shrubs and vines in the understory and eventually lost to the lateral crown expansion of canopy trees.

Large Tree Attributes.—Bragg (2004a) also surveyed the entire LWDF for trees of exceptional dimensions. Though storms, lightning, and beetles have killed many large loblolly and shortleaf pines, including several >100 cm in DBH, the two most notable pines have survived to date. The Morris Pine, a 300+ year old loblolly named after a long-time Crossett Lumber Company employee (Anonymous 1950), was measured at almost 142 cm DBH in 2000. In 2006, this pine had not changed in diameter and was nearly 36 m tall (Table 3). The Morris Pine still appears healthy, although it is increasingly isolated as neighboring pines die.

The Walsh Pine, the current state and probable national champion shortleaf pine, measured 90.7 cm DBH and 43.3 m tall in 2001 and now scales 90.9 cm DBH and 41.5 m tall. The Walsh Pine has not become shorter over the years; rather, the 1.8 m height difference arose from the use of more accurate laser technology and a more dependable height determination technique (the sine method). The tangent method used in 2000 is prone to overestimate height, especially for large, wide-crowned individuals. As an example, the 120.7 cm DBH, 45.6 m tall loblolly pine reported in Bragg (2004a) was originally measured using the tangent method with a cloth tape and clinometer. This tree was blown over and partially salvaged in 2003. However, the base of the pine remains were it fell, and the top was also left in place, making it possible to measure its stem length along the ground. This tree turned out to be just over 40 m tall, or almost 6 m shorter than first thought.

Wind, decay, and drought also killed a number of large hardwoods, especially some hollow oaks, but in general these hardwoods were less impacted by the last 5 years of disturbances than the pines. White oak and post oak comprised the majority of the biggest hardwoods across the site, with a few sweetgum, southern red oak, and water oak (*Quercus nigra* L.) greater than 70 cm DBH scattered throughout the LWDF (Table 3). A relatively large (46.5 cm DBH and 27.6 m tall) winged elm was also located in 2006. Most hardwoods in the LWDF are noticeably shorter than the pines, which form a supercanopy above them. A few sweetgum exceed 35 m tall, but most overstory hardwoods are between 25 and 30 m.

Supplemental Pine Age Data.—In late 2005, four shortleaf pine snags were felled to minimize vehicular hazards along Highway 425 as it passes through the LWDF, and ring counts were made on the stumps left behind (Table 4). Due to pre-existing decay of the outer rings and heart rot, these ring counts are only approximate. Without more accurate cross-dating, we cannot specify exactly when these trees succumbed, except to say that they died from 2 to 4 years ago. These shortleaf pines ranged from 146 to 166 years old. Other sources have identified cohorts of similarly aged pines at a number of nearby sites (e.g., Jones 1971, Tompkins 2000, Heitzman et al. 2004, Bragg 2004b); the age of these pines coincide with the beginning of large-scale Euroamerican settlement in this portion of the Upper Gulf Coastal Plain.

Stump 4, though the hardest to age given its rotten heartwood, contained other important information. Two obvious fire scars dating to approximately 25 and 102 years ago were found on the cut face of the stump. It is possible other fire scars will be discovered on this tree once a section has been removed and sanded for more detailed observations. A number of fire scarred live pines can be found throughout the LWDF, including several within 50 m of this stump. Given this relative abundance, it should be possible to construct at least a partial fire chronology in this stand, which will prove helpful in understanding historic fire regimes.

Dr. Brian R. Lockhart of the USDA Forest Service provided additional data on 36 pine stumps from the LWDF, which were aged by students in 1988. Combined with the age records from Bragg (2004a) and those mentioned in this paper, a graph of

Table 3. Large tree dimensions across the entire LWDF measured in February-March 2006.

Species [a]	DBH (cm)	Height [b] (m)	Average [c] crown width (m)	Bigness Index [d]	Notes
Loblolly pine	141.7	35.6	17.4	306	Morris Pine
Loblolly pine	104.1	**42.2**	14.5	279	
Loblolly pine	109.0	37.2	16.4	270	
Loblolly pine	101.6	38.6	20.4	269	
White oak	114.8	**33.3**	20.9	268	
Shortleaf pine	111.5	35.4	16.9	--	double stem
Loblolly pine	106.9	35.6	16.8	263	
Shortleaf pine	90.9	**41.5**	15.2	261	Walsh Pine
Shortleaf pine	87.6	40.1	11.3	249	
White oak	92.7	33.5	23.4	244	
Southern red oak	97.8	**31.3**	24.4	244	
Shortleaf pine	83.8	36.9	13.8	236	
Shortleaf pine	74.9	39.5	11.4	232	
White oak	92.7	30.7	19.4	231	
Sweetgum	76.2	**36.8**	15.2	227	
White oak	81.5	32.0	22.5	224	
Shortleaf pine	78.2	34.3	11.9	219	
White oak	74.9	32.5	16.6	213	
Sweetgum	81.5	30.2	15.9	213	
Post oak	81.3	**30.4**	13.0	211	
Shortleaf pine	63.8	37.9	8.7	210	
Post oak	81.3	27.9	18.3	207	
Water oak	71.9	**31.2**	11.2	200	
Winged elm	46.5	**27.6**	14.2	160	

[a] Not every tree species present in the LWDF is represented in this table. The tallest example of each species is indicated by **bold-faced text**.
[b] The height reported in this table is calculated using the sine method, which is considerably more accurate for large dimension individuals, especially wide-crowned hardwoods (Blozan 2004, Bragg 2006).
[c] Average of the widest portion of the crown and the width perpendicular to this axis.
[d] Bigness Index (American Forests 2006) = circumference (in inches) + tree height (in feet) + ¼ average crown width (in feet)

establishment dates shows a long history of pine recruitment during the 19th Century and first half of the 20th Century (Fig. 5). There is a considerable range of pine ages in the LWDF, from an estimated 300+ years for the Morris Pine to approximately 50 years old (Fig. 5). The estimated age of the Morris Pine clearly isolates it temporally from the rest of the stand. However, this incomplete and non-random sample does not infer that there are no other pines in the stand that originated in the 18th Century—rather, it simply implies that we did not date any others to this period.

Even though precise dating was often complicated by extensive basal decay, most pines aged in the LWDF originated from 1840 to 1900. Since the LWDF was old-growth when established in 1948, the lack of old pines that would have dominated the canopy when the stand was reserved indicates that this cohort has almost completely succumbed. Pine recruitment

Table 4. Tree age dated from shortleaf pine stumps dated in March of 2006 on the LWDF.

Stump number	Stump diameter (cm)	Ring count	Comments
1	69.8	146	Ring count to pith; no obvious fire scars
2	77.5	150	Ring count to pith; no obvious fire scars
3	87.6	166	Ring count to pith; no obvious fire scars
4	86.4	151	Ring count to rotten core; fire scars from 25 and 102 years ago

has also been virtually non-existent since the 1950s, with the most recent canopy ascensions following the abandonment of the unpaved highway to Bastrop, Louisiana, decades ago. A few small pockets of young (<10 yr old) pine can be found along the edge of Highway 425, but regeneration conditions within the stand are too unfavorable to maintain pine dominance.

Using the pine stump ring counts from the 1988 data, the stump ages of Bragg (2004a), and the new data points collected in 2006, a linear regression model of pine age as a function of stump diameter was developed (Fig. 6). Loblolly and shortleaf pine were not distinguished from each other, partially because they both follow the same general allometric patterns and partially because a considerable number of the 1988 pines were not identified to species (shown as stars in Fig. 6). Although the slope of the equation is highly significant ($P < 0.0001$), the regression explained only a small portion of the overall variance in the data ($R^2 = 0.2355$). This is not surprising, given that 60 to 70 cm pine stumps in the LWDF ranged in age from less than 60 years to 160 years. Generally, there is a much stronger relationship between diameter and age in well-regulated loblolly/shortleaf pine forests, and the dispersed nature of the data in Fig. 6 is further evidence of the old-growth structure of the LWDF.

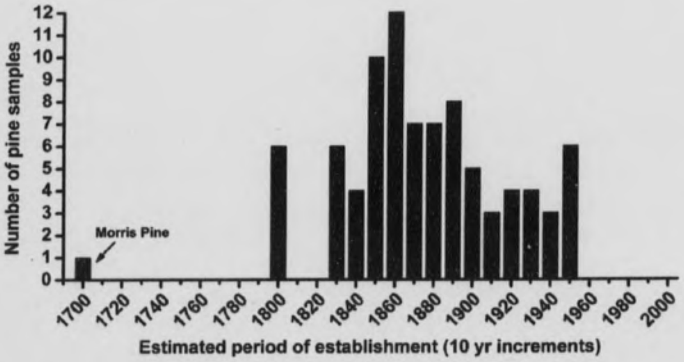

Fig. 5. Establishment pattern of selected pines in the LWDF taken from data in Bragg (2004a), the current study, and unpublished data collected by Dr. Brian Lockhart in 1988. Age of the Morris Pine and the individuals established in 1800 are estimates.

Conclusions

Five years, though a short period of time in the history of this old pine stand, has been a time of dramatic changes in species abundance and dominance. The strong windstorm that struck the LWDF, though not as devastating as a tornado or crown fire, had a disproportionate impact on the overstory pines and thus accelerated succession toward hardwoods. However,

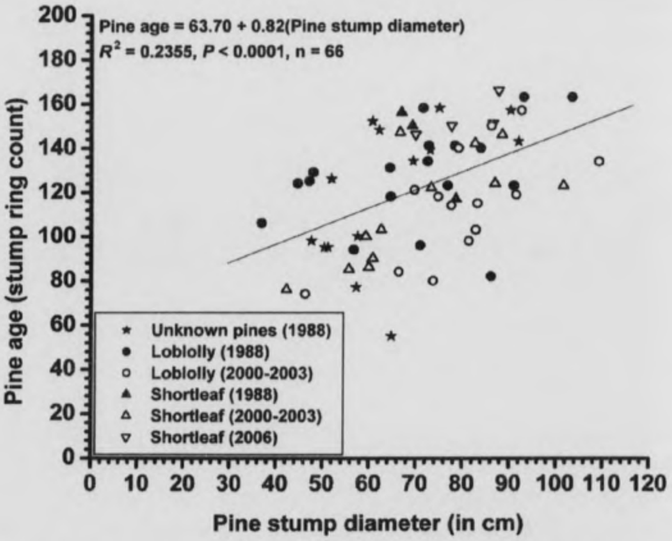

Fig. 6. Relationship between pine age and stump diameter at the LWDF using data from the present study, Bragg (2004a), and unpublished data from 1988.

even relatively brief periods of drought that occurred during the last few years were sufficient to at least temporarily impact many of the under- and midstory tree species, further altering the successional trajectory of this remnant old-growth stand. Under all of these pressures, long-term maintenance of a pine overstory will be virtually impossible in this preserve without deliberate human intervention to assure its recruitment.

ACKNOWLEDGMENTS.—I would like to recognize the contributions of the following: Mike Chain (USDA Forest Service), Conner Fristoe (Plum Creek Timber Company), Jim Guldin (USDA Forest Service), Brian Lockhart (USDA Forest Service), Mike Shelton (USDA Forest Service), Kirby Sneed (USDA Forest Service), and Bruce Walsh (USDA Forest Service, deceased). Brian Lockhart also graciously provided the previously unpublished pine stump ring counts for the LWDF.

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Density Functional Studies of the Structure and Bonding of Nitrosyl Metalloporphyrin Complexes

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Abstract.—Density functional calculations were used to determine optimized geometries for nitrosyl-metalloporphyrin complexes of Fe(II), Co(II) and Mn(II). The optimized structures were found to be consistent with experimental data and previous computational predictions using single point density functional calculations. Vibrational frequencies for the N-O stretching mode were also calculated and shown to be consistent with experimental data. The nature of the bonding between the metal center and nitrosyl ligand is discussed in relation to the structure of the M-N-O linkage. The results were found to be consistent with previous descriptions derived from the Fenske-Hall approximate molecular orbital method. Other interesting structural features in the optimized geometries are noted.

Key words:—Density functional calculations, optimized geometries, nitrosyl-metalloporphyrin complexes, Fe(II), Co(II), Mn(II), Fenske-Hall approximate molecular orbital method.

Introduction

The calculations presented in this paper focus on the structure of the metal-nitrosyl unit in porphyrin complexes of Fe(II), Co(II), and Mn(II) (indicated as M(PP)NO). It has been shown experimentally that the M-N-O linkage in Mn(II) porphyrin complexes is essentially linear (Scheidt and Frisse 1975) while the structures in Fe(II) and Co(II) complexes are bent (Scheidt and Hoard 1973, Scheidt and Piciulo 1976). Approximate molecular orbital calculations have been used to explain this trend, which primarily arises as a result of a change in the number of electrons available for bonding on the metal center (Graham and Brown 2001). Bending of the M-N-O linkage allows for orbital mixing, resulting in a lowering of the energy of the highest occupied molecular orbital (HOMO) in complexes of Iron (II) and Cobalt (II). A qualitative MO diagram describing the interactions between linear and bent NO and a metal porphyrin fragment is given in Fig. 1.

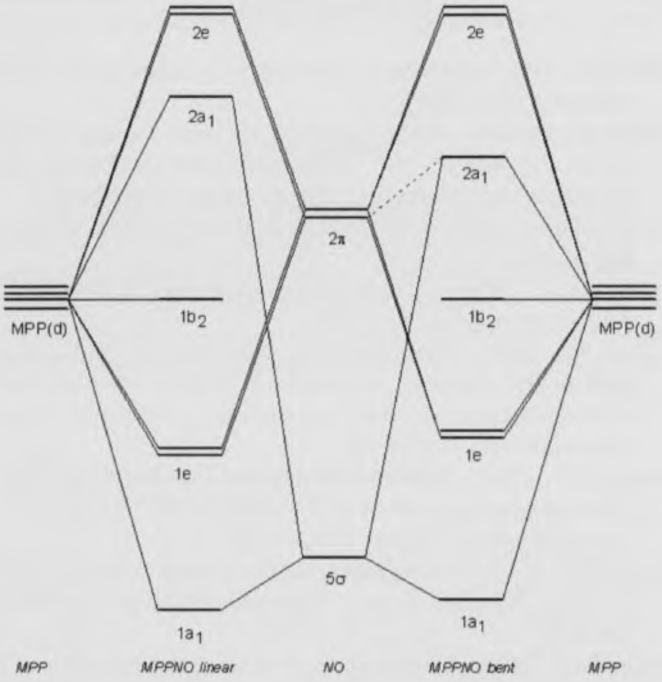

Fig. 1. Qualitative Molecular Orbital Description of Linear and Bent Nitrosyl Metalloporphyrin Complexes.

The $2a_1$ molecular orbital for the linear NO complex in Fig. 1 can be described as primarily the nitrosyl 5 – metal d_{z^2} antibonding interaction. For Co(II) and Fe(II) this is the HOMO of the complexes. In the case of Co(II) this orbital is doubly occupied, in the Fe(II) complex it is singly occupied, and in the Mn(II) complex it is empty. Bending of the M-N-O linkage allows mixing between the metal d_{z^2} and NO 2π orbitals and a decrease in overlap between the NO5 and metal d_{z^2} resulting in stabilization of the $2a_1$ orbital. Bending also results in some destabilization of the 1e orbitals (largely metal based orbitals with metal $d\pi$-NO2π back-donation) and hence is favored only when $2a_1$ is occupied. Qualitative molecular orbital theory predicts a linear NO in the Mn(II) complex and a bent NO in Fe(II) and Co(II) complexes. The degree of bending in the Co(II) complex is expected to be greater than that in the Fe(II) complex as the $2a_1$ orbital is only singly occupied in the Fe(II) case. These qualitative descriptions of bonding provide a simple explanation of the reasons for NO bending. However, the method used to generate these descriptions (Fenske-Hall Approximate Molecular Orbital Method) cannot be used to quantitatively predict the degree of bending that will occur.

Because of the large size of the systems of interest, full geometry optimizations using ab initio or DFT (Density Functional Theory) methods were not practical until recently. Hence to obtain a quantitative prediction of the degree of bending of the M-N-O linkage in each complex, previous studies used a series of single point density functional calculations to estimate the optimal M-N-O angle (Graham and Brown 2001). The degree of bending predicted in these studies was found to be in good agreement with experimental data. Due to the rapid development in processing speed of modern computers, full optimization calculations on large molecules are now possible even on relatively inexpensive personal computers. In the current paper we present the results of full geometry optimization and vibrational analysis calculations on the nitrosyl-porphyrin complexes of iron(II), manganese(II), and cobalt (II).

Methods

All calculations were performed using Gaussian 03W on IBM compatible PCs (Fritch et al. 2003). The non-local density functional method BPW91 was used for all calculations. Geometry optimizations were performed using both 6-31G and 6-31G(d) basis sets. Vibrational frequencies were calculated using the 6-31G(d) basis set, except for the manganese complex for which frequencies were calculated using 6-31G.

Results and Discussion

The geometries of the metal nitrosyl-porphyrin complexes were optimized using the non-local density functional method BPW91 and the basis sets 6-31G and 6-31G(d). Ball and stick representations of the optimized complexes of Mn, Fe and Co are given in Figs. 2, 3 and 4, respectively.

Table 1. Calculated M-N-O angles for nitrosyl porphyrin complexes of Mn(II), Fe(II) and Co(II)

M-N-O angle	Mn	Fe	Co
Experiment	*176*	*149*	*135*
ADF single point calculations	180.0	144	128
BPW91/6-31G optimization	180.0	143	125
BPW91/6-31G(d) optimization	180.0	144	123

A comparison of calculated and experimental M-N-O angles for each complex is given in Table 1. It is observed that the predicted M-N-O angles are in good agreement with both experimental data and previously predicted values from single point DFT calculations. The addition of d-polarization functions to the 6-31G basis set (6-31G(d)) did not significantly change the calculated M-N-O angles. Both the single point DFT calculations and the full geometry optimization calculations appear to slightly overestimate the degree of bending in the iron and cobalt complexes. However, the experimental numbers were determined for complexes in which there are additional functional groups attached to the porphyrin ring (such as in protoporphyrin IX) and also under conditions where intermolecular interactions could produce some variation in structure. Hence, an exact agreement is not expected.

As illustrated in Figs. 3 and 4, bending of the NO ligand in the Fe(II) and Co(II) complexes occurs in a plane between nitrogen atoms of the porphyrin ring (a dihedral angle of 45°).

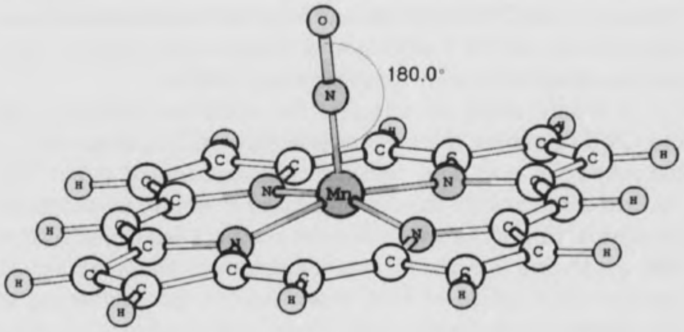

Fig. 2. BPW91/6-31G(d) Optimized Geometry for Mn(PP)NO.

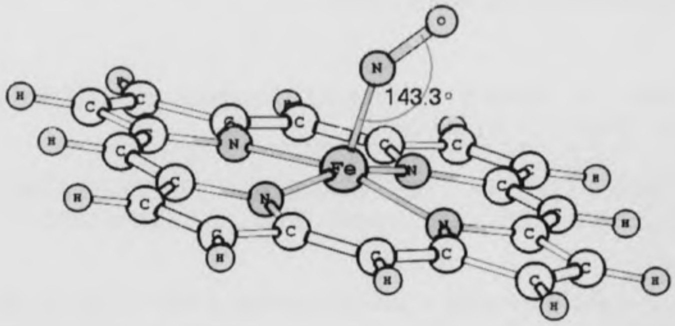

Fig. 3. BPW91/6-31G(d) Optimized Geometry for Fe(PP)NO.

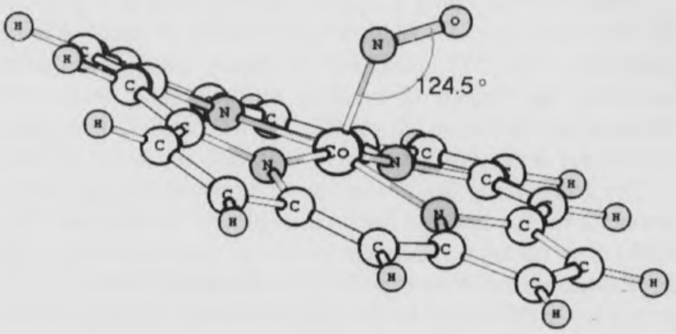

Fig. 4. BPW91/6-31G(d) Optimized Geometry for Co(PP)NO.

This minimizes repulsive interactions between the NO and ring nitrogen atoms. This is the mode of bending assumed in previous approximate and DFT calculations (Graham and Brown 2001) and consistent with known experimental results.

It is interesting to note that the optimized structure for the Co(II) complex exhibits considerable ruffling distortion of the porphyrin ring. This structure was determined to be 7 kJ/mole lower in energy than an optimized structure in which the porphyrin ring is forced to remain planar. Distortions of this type are known to occur for smaller metal ions and it is believed that the main cause of such distortions is the shortening of the distance between the metal center and porphyrin nitrogen atoms. Table 2 lists metal-porphyrin (N) distances for the three complexes. It is observed that the average metal-N distance in the Co(II) complex is indeed shorter than in the complexes of Fe(II) and Mn(II). The porphyrin rings in the Mn(II) and Fe(II) complexes are essentially planar.

Table 2. Metal-N (porphyrin) distances in M(PP)NO, M = Mn(II), Fe(II) and Co(II)

Mn-N	Fe-N	Co-N
1.997, 1.997, 1.975, 1.975 Average = 1.986	2.027, 2.027, 1.999, 1.999 Average = 2.013	2.018, 2.018, 2.018, 2.018 Average = 2.018

Other interesting structural features noted are that in the complexes of Fe and Co, the metal is slightly displaced from the center of the porphyrin ring and the M-N bond of M-NO is slightly tilted from perpendicular toward the closer side of the porphyrin ring.

The N-O stretching frequency is often used as a probe in the determination of structure and bonding of metal nitrosyl complexes. The NO stretching frequency gives information regarding the degree of bending of the ligand with lower frequencies indicative of smaller M-N-O angles. Calculated vibrational frequencies for the N-O stretch are given in Table 3. The frequencies are scaled using standard scaling factors related to the method and basis set employed (Radom and Scott 1996). The calculated frequencies are in good agreement with experimental values. As expected, the calculated N-O stretching frequency in the series Mn-Fe-Co decreases as the M-N-O angle decreases.

The variation of Co(PP)NO HOMO energy and total energy with M-N-O angle is illustrated in Fig. 5. The energies are relative values, scaled for visualization purposes, and hence no units are given. Consistent with previous calculations, the energy of the HOMO decreases approximately linearly with decreasing M-N-O angle. The total energy of the complex exhibits a minimum at approximately 123°, beyond which further stabilization of the HOMO no longer results in stabilization of the complex. This is expected as bending of the NO results in some destabilization of the $1a_1$ and 1e molecular orbitals (Fig. 1). The resulting optimal angle arises from competing effects of stabilization of $2a_1$ and destabilization of $1a_1$ and 1e. In the Iron (II) complex, a larger M-N-O angle (relative to Co(II)) is expected as the singly occupied $2a_1$ HOMO contributes less influence on the total energy of the molecule.

Table 3. Calculated Vibrational Frequencies for the N-O stretch in M(PP)NO, M = Mn(II), Fe(II) and Co(II).

	Experimental	Calculated Gaussian 03
Co	1680	1661 (6-31G(d))
Fe	1700	1691 (6-31G(d))
Mn	1730	1744 (6-31G)

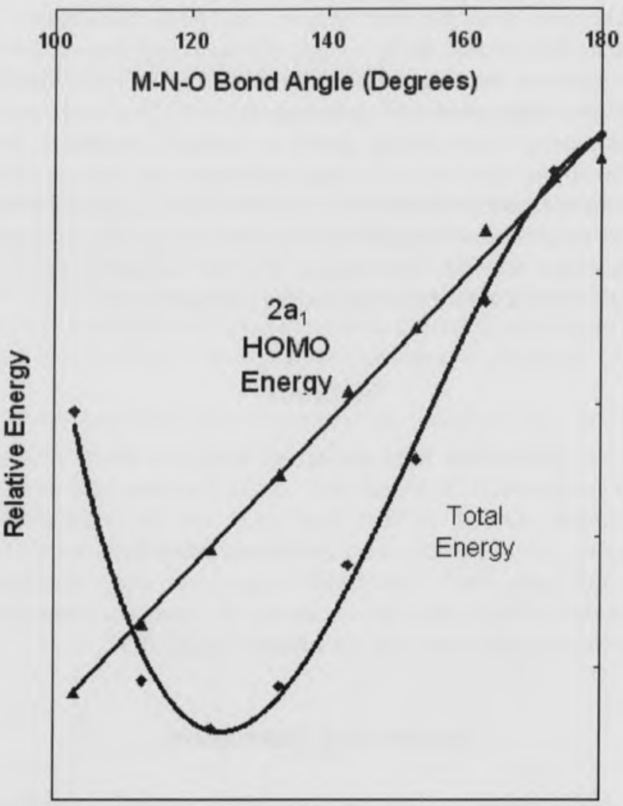

Fig. 5. Variation of Relative Total Energy and HOMO Energy with M-N-O angle for Co(PP)NO.

Conclusions

The results obtained using Gaussian 03 density functional calculations are consistent with those previously derived from Fenske-Hall approximate molecular orbital calculations and Amsterdam Density Functional single point energy calculations

(Graham and Brown 2001). Good agreement is obtained between optimized geometries, previously predicted structures, and experimental structures for the complexes of interest. Calculated vibrational frequencies are also found to be in good agreement with available experimental data. The general nature of the bonding and the influence of the HOMO energy on the structures derived from Gaussian calculations are consistent with previous descriptions derived from approximate molecular orbital calculations.

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Aquatic Macroinvertebrates of the Strawberry River System in North-central Arkansas

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Abstract.—The Strawberry River has been designated an Extraordinay Resource Water, an Ecologically Sensitive Water Body, and a Natural and Scenic Waterway. As such, it is particularly important that the biodiversity of this river system be documented thoroughly. The purpose of this research was to develop a comprehensive list of the aquatic macroinvertebrates of the Strawberry River and its major tributaries. The information was developed from a thorough literature review and by examining specimens housed in various collections of the Arkansas State University Museum of Zoology and collections of the authors. The latter included 9 collections at 4 sites along the mainstream and 17 collections from 8 tributaries. To date, 313 species of aquatic macroinvertebrates are known to occur in the Strawberry River system. Among the freshwater mussels, 1 is listed and 7 are ranked: *Leptodea leptodon* (Federal list-endangered, S1), *Cyprogenia aberti*, *Lasmigona costata*, and *Quadrula cylindrica* (S2), *Cyclonaias tuberculata, Strophitus undulatus*, and *Villosa lienosa* (S3). Of 25 stonefly species known to be endemic to the Interior Highlands, 7 occur in this system.

Key words:—Strawberry River, aquatic macroinvertebrates, freshwater mussels, stonefly, Federal list-endangered, *Leptodea leptodon, Cyprogenia aberti, Lasmigona costata, Quadrula cylindrica, Cyclonaias tuberculata, Strophitus undulatus, Villosa lienosa.*

Introduction

The Strawberry River has been designated an Extraordinary Resource Water and an Ecologically Sensitive Water Body, as outlined by the Arkansas Water Quality Standards. It is also designated as a Natural and Scenic Waterway (ADPCE 1996). As such, it is particularly important that the biodiversity of this river system be documented thoroughly.The Strawberry River is a spring-fed, relatively clear stream in north-central Arkansas, typically consisting of wide, shallow pools separated by riffles. The river originates near Viola, Arkansas, and flows 177 km before entering the Black River at BR km 56.2, approximately 10.5 km above Lockheart Ferry. The drainage area is 2,100 km^2 (Beadles 1972). Principal tributaries are the Little Strawberry River, Piney Fork, North Big Creek, South Big Creek, Cooper Creek, Dry Creek, and Caney Creek. The Strawberry River arises in Ordovician Calico sandstone of lower Fulton County and winds through Cotter dolomite in Izard and Sharp counties. Midway into Lawrence County, the river passes through Powell limestone, then in rapid succession through Smithville and Black Rock limestone. As it nears its confluence with the Black River, the Strawberry River drops into the Quaternary Alluvium (Croneis 1930). Major soils adjacent to the river are chiefly of the Huntington and Elk series (SCS 1964). Mean annual rainfall is 112, 119, and 138 cm in Izard, Sharp and Lawrence counties, respectively. Air temperatures range from –25 to 40° C (Hickmon 1941).

No previous studies dealing with aquatic macroinvertebrates of the Strawberry River system have been comprehensive. Rather, they have been restricted to a few collections at a limited number of locations (e.g. Robison 1968; Robison and Harp 1971; Beadles 1972; Harp 1972; Moss and Harp 1993; ADPCE 1996) or have focused on a target group of organisms (e.g. Harp 1989; Poulton and Stewart 1991; Rust 1993; Moulton and Stewart 1996).

The purpose of this research was to develop a comprehensive list of the aquatic macroinvertebrates of the Strawberry River and its major tributaries. Species of particular interest are discussed.

Materials and Methods

The information to develop a list of the aquatic macroinvertebrates known to occur in the Strawberry River system was derived primarily from 4 sources (Table 1). A comprehensive review of the literature provided the nucleus. Aquatic macroinvertebrates previously collected and housed in the Adult Odonata Collection, Freshwater Mussel Collection and George L. Harp Aquatic Macroinvertebrate Collection of the Arkansas State University Museum of Zoology were inventoried. Mollusca collected from the Strawberry River mainstream in the 1970s by one of us (HWR) provided additional information. Both authors collected a series of samples during 1988-89 from tributaries that had received little or no previous attention.

Collections were made with a Turtox Indestructible™ dip net. Taxonomic usage follows Merritt and Cummins (1996) for aquatic insects, Gordon (1980) for Mollusca, and Smith (2001) for other aquatic macroinvertebrates. Common names of crayfishes follow Pflieger (1996), those of Mollusca follow Harris and Gordon (1990), and those of Odonata follow Paulson and Dunkle (1999).

We have endeavored to use the most current taxonomy. However, taxonomy, particularly that of aquatic macroinvertebrates, is dynamic. To minimize confusion, older names under which species have occurred in older literature have been noted (Table 2). For example, a small clubtail dragonfly that occurs in Arkansas, Kentucky, Missouri, and Tennessee, previously has been identified as *Stylogomphus albistylus*, an eastern species, but it was in fact an undescribed species. Cook and Laudermilk (2003) have recently described it as *Stylogomphus sigmastylus.*

Results and Discussion

This survey encompassed 9 collections at 4 sites along the Strawberry River mainstream and 14 collections on tributaries (Table 1). The authors or their former students were associated with nearly all collections made. The notable exception was the Plecoptera survey by Poulton and Stewart (1991), but even this comprehensive study began with verification of the identities of the stoneflies in the possession of the authors.

Because the purposes and methodologies varied greatly among studies, as well as number of times a given site was visited, analyses of data are restricted.

To date, 313 species of aquatic macroinvertebrates are known to occur in the Strawberry River system (Table 2). Significant records are as follows.

Among the Unionoida, *Leptodea leptodon*, the scaleshell, was reported from the Strawberry River by Harris et al. (1997). This species is on the federal list of endangered species and is ranked in Arkansas as S1, defined as typically 5 or fewer estimated occurrences in the state or only a few remaining individuals, may be vulnerable to extirpation. Additionally, in Arkansas 3 Strawberry River species are ranked as S2, *Cyprogenia aberti, Lasmigona costata, and Quadrula cylindrica.* Individuals of these species are very rare with typically 5-20 estimated occurrences or with many individuals in few occurrences; they are often susceptible to becoming extirpated. Finally, *Cyclonaias tuberculata, Strophitus undulatus, and Villosa lienosa* are ranked as S3. Individuals of these species are rare to uncommon with typically 20-100 estimated occurrences or fewer occurrences but with large numbers of individuals in some populations. These populations may be susceptible to large-scale disturbances.

Baetisca obesa was collected from the lower Strawberry River, at St. Hwy. 25. This is only the second record for this species in Arkansas. Cochran and Harp (1990) first reported this mayfly from the St. Francis Sunken Lands. At both locations, the rivers are of medium size and have moderate current over a sand substrate. The Strawberry River site is the only location reported for Arkansas where *B. lacustris* and *B. obesa* occur together. *B. lacustris* characteristically inhabits smaller, less turbid streams with rubble substrate.

Of the 25 stonefly species listed as regional endemics by Poulton and Stewart (1991), 7 are known to occur in the Strawberry River system. These are *Allocapnia mohri, Zealuctra warreni, Strophopteryx cucullata, Neoperla falayah, N. osage, N. robisoni* and *Isoperla ouachita.* Additionally, *Attaneuria ruralis*, which occurs only in Arkansas, Illinois, Kansas, and Missouri, is a species rarely collected.

Seven specimens of the predaceous diving beetle *Heterosternuta ouachitus* were collected, 2 from the Strawberry River at St. Hwy. 354 (Harp 1989), 2 at Hars Creek and 3 at McJunkins Branch. Prior to that date, the collection of only 21 specimens had ever been reported. Unlike most predaceous diving beetles, *Heterosternuta* species prefer the interstices of rocks at the edges of riffles in headwater streams, a microhabitat that is rarely sampled. The center of distribution for *H. ouachitus* is the Interior Highlands. While this species is widely distributed, it is quite uncommon. Of 30 samples taken in its preferred habitat, 9 contained a total of 34 specimens among 981 total *Hydroporus* (general sense) individuals (Harp 1989). The 5 specimens collected at Hars Creek and McJunkins Branch constituted only 2.7% of the *Hydroporus* from those streams.

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Table 1. Collecting sites (noted by date and collectors) and literature sources for aquatic macroinvertebrate taxa, Strawberry River System.

1. Strawberry R. at St. Hwy. 354, 5mi W Horseshoe Bend, SW¼Sec7, T18N, R8W, Izard Co. 4 Sep 88, GL Harp (1a); 1995, Richard Mitchell (1b).
2. South Big Cr. at St. Hwy. 115, 1.5mi NE Calamine, NE¼Sec23, T16N, R4W, Sharp Co. 19 Aug 86, GL Harp (2a); 5 Sep 88, Billy Justice & GL Harp (2b); 1 Apr 95, Bill Posey and Aquatic Entomology Class (2c).
3. Cooper Cr., 1.5mi W Smithville, NW¼Sec32, T17N, R3W, Lawrence Co. 1 Jul 64, L Tucker (3a); 25 Jun 71, GL Harp (3b), 11 Sep 71, GL Harp (3c); 1974, Herb Athearn (3d); 6 Oct 78, T Pickett (3e); 10 Jul 79, HE Barton (3f); 5 Sep 88, B Justus & GL Harp (3g); 19 Feb 95, B Posey & GL Harp (3h).
4. Greasy Cr., 3mi NNW Union, SE¼Sec21, T19N, R9W, Fulton Co. 23 Sep 89. HW Robison & GL Harp.
5. Unnamed tributary of the Strawberry R. at a section road ford, 5mi WNW Franklin, SE¼Sec30, T18N, R8W, Izard Co. 23 Sep 89. HW Robison & GL Harp.
6. McJunkins Branch, 2.2mi SE Franklin, SE¼Sec4, T17N, R7W, Izard Co. 23 Sep 89. HW Robison & GL Harp
7. Strawberry R. at St. Hwy. 395, 7mi W Salem, SW¼Sec10, T19N, R9W, Fulton Co. 23 Sep 89. HW Robison & GL Harp
8. Hars Cr. 0.7mi S St. Hwy. 56, 0.2mi E Izard Co line, SW¼Sec30, T18N, R6W, Sharp Co. 23 Sep 89. HW Robison & GL Harp
9. Strawberry R. at U.S. Hwy. 167, 2mi N Evening Shade, NE¼Sec 27, T17N, R6W, Sharp Co. 21 Aug 76, GL Harp (9a); 7 Aug 78, GL Harp (9b); 24 Sep 89, HW Robison & GL Harp (9c); 3 Jun 90, GL Harp (9d).
10. Mill Cr. at St Hwy 56, Evening Shade, NE¼Sec3, T16N, R6W, Sharp Co. 24 Sep 89, HW Robison & GL Harp.
11. Strawberry R. at St. Hwy. 25 bridge, ~2mi SW Lynn, NE¼Sec34, T16N, R3W, Lawrence Co. 22 Jul 90, GLH (11a); 1 Apr 95, B Posey and Aquatic Entomology Class (11b).
12. Strawberry R. ~1.3mi N Poughkeepsie, NE¼Sec26, T17N, R5W, Lawrence Co. 1 Apr 95, B Posey and Aquatic Entomology Class.
13. North Big Cr. ~1mi SE Center, NW¼Sec10, T17N, R5W, Sharp Co. 31 May 05. GL Harp.
14. Aquatic macroinvertebrates documented from the Strawberry R. by Robison and Harp (1971).
15. Aquatic macroinvertebrates documented from three stations along the Strawberry R. by Harp (1972).
16. Freshwater mussels documented from the Strawberry R. mainstream during the 1970s by HW Robison.
17. Stoneflies documented from the Strawberry R. System by Poulton and Stewart (1991).
18. Aquatic macroinvertebrates documented from North Big Cr. by Moss and Harp (1993).
19. Freshwater mussels documented from the Strawberry R. mainstem by Rust (1993).
20. Caddisflies documented from the Strawberry River System by Moulton and Stewart (1996).
21. Aquatic macroinvertebrates documented from the Strawberry River System by ADPCE (1996).
22. Arkansas crayfishes database.
23. Report listing rare and endangered Unionacea by Harris et al. 1997.

Table 2. Aquatic macroinvertebrate taxa and distributions within the Strawberry River System.

Scientific Name	Common Name	Location (Table 1)
PORIFERA	Sponges	
Spongillidae	Freshwater sponges	
Sp. 1	Freshwater sponges	18
PLATYHELMINTHES	Unsegmented flatworms	
Class Turbellaria	Turbellarians	
Order Tricladida	Triclads	
Planariidae	Planarians	14
Dugesia sp.	Planarian	15, 18
NEMATOMORPHA	Horsehair worms	4, 10
Gordiidae	Gordian worms	
ANNELIDA	Segmented roundworms	
Class Oligochaeta	Oligochaetes	14, 15, 21
Order Lumbriculida	Lumbriculids	
Lumbriculidae	Lumbriculids	
Sp. 1	Lumbriculid	18
Order Branchiobdellida	Branchiobdellid worm	
Branchiobdellidae	Branchiobdellid worm	
Sp. 1	Branchiobdellid worm	18
Class Hirudinea	Leeches	
Order Rhynchobdellida	Rhynchobdellid leeches	14
Glossiphoniidae	Glossiphoniid leeches	
Batrachobdella sp.	Glossiphoniid leech	18
Helobdella sp.	Glossiphoniid leech	4, 18
Placobdella sp.	Glossiphoniid leech	17
MOLLUSCA	Molluscs	
Class Gastropoda	Snails, limpets (Univalves)	21
Order Limnophila	Pulmonate snails	
Ancylidae	Limpets	
Ferrisia rivularis	Limpet	9c, 15, 18
Planorbidae	Planorbids	
Gyraulus sp.	Planorbid	18
Helisoma sp.	Planorbid	9c
Pleuroceridae	Pleurocerids	14
Elmia (=*Goniobasis*) *ovoidea*	Pleurocerid	9c, 10, 15
Pleurocera acuta	Pleurocerid	3d
Viviparidae	Viviparids	
Campeloma sp.	Viviparid	9c, 18
Physidae	Physids	
Physella (=*Physa*) sp.	Physid	4, 5, 6, 7, 8, 9c, 10, 18

Table 2. Aquatic macroinvertebrate taxa and distributions within the Strawberry River System. (cont.)

Scientific Name	Common Name	Location (Table 1)
Class Pelecypoda	Clams, mussels (Bivalves)	
Unionoida	Unionoid mussels	
Sphaeriidae	Fingernail clams	18
Musculium sp.	Fingernail clam	4
Sphaerium sp.	Fingernail clam	7
S. striatinum	Fingernail clam	15
Unionidae	Unionids	
Actinonaias ligamentina	Mucket	16, 19
Amblema plicata	Threeridge	16, 19
Cyclonaias tuberculata	Purple wartyback	16, 19
Cyprogenia aberti	Western fanshell	19
Elliptio dilata	Spike	19
Fusconaia ebena	Ebonyshell	19
F. flava	Wabash pigtoe	19
Lampsilis teres	Yellow sandshell	16
Lasmigona complanata	White heelsplitter	19
L. costata	Fluted-shell	19
Leptodea fragilis	Fragile papershell	16, 19
L. leptodon	Scaleshell	23
Legumia recta	Black sandshell	19
Obliquaria reflexa	Threehorn wartyback	19
Potamilus purpurata	Bleufer	16, 19
Pyganodon grandis	Giant floater	16, 18
Quadrula cylindrica	Rabbitsfoot	16
Q. metanevra	Monkeyface	16, 19
Q. pustulosa	Pimpleback	16, 19
Strophitus undulatus	Squawfoot	16
Tritogonia verrucosa	Pistolgrip	16, 19
Truncilla truncata	Deertoe	16. 19
Villosa lienosa	Little spectaclecase	16
Veneroida	Veneroid mussels	
Corbiculidae	Asiatic clam	
Corbicula fluminea	Asiatic clam	9c, 16, 18, 21
ARTHROPODA		
Class Arachnida	Arachnids	
Hydracarina	Water mites	14, 18
Lebertia sp.	Water mite	15
Class Crustacea	Crustaceans	
Order Isopoda	Isopods	
Asellidae	Sow bugs or pill bugs	
Caecidotea sp.	Sow bug	11b
C. militaris	Sow bug	15

Table 2. Aquatic macroinvertebrate taxa and distributions within the Strawberry River System. (cont.)

Scientific Name	Common Name	Location (Table 1)
Lirceus sp.	Sow bug	2c, 3h, 6, 11b, 15, 18, 21
Order Amphipoda	Amphipods	
Crangonyctidae	Sideswimmers, scuds	
Crangonyx gracilis	Sideswimmer	15
Synurella sp.	Sideswimmer	11b, 12
Gammaridae	Sideswimmers, scuds	
Gammarus sp.	Sideswimmer	3g, 3h
G. minus	Sideswimmer	10
Talitridae	Sideswimmers, scuds	
Hyalella azteca	Sideswimmer	6, 9c, 12, 15, 18
Order Decapoda	Decapod crustaceans	
Palaemonidae	Freshwater shrimp	
Palaemonetes kadiakensis	Freshwater shrimp	11b, 15
Cambaridae	Crayfishes	11b
Fallicambarus fodiens	Digger crayfish	22
Orconectes sp.	Crayfish	3g, 4, 5, 6, 10, 15, 18
O. eupunctus	Coldwater crayfish	22
O. ozarkae	Ozark crayfish	22
O. punctimanus	Spothanded crayfish	22
O. virilis	Northern crayfish	22
Class Insecta		
Order Ephemeroptera	Mayflies	
Baetidae	Baetid mayflies	
Acentrella sp.	Baetid mayfly	1b
Baetis sp.	Baetid mayfly	1b, 2c, 10, 11b, 12, 18, 21
Callibaetis fluctuans	Baetid mayfly	8
Diphetor hageni	Baetid mayfly	12
Procloeon rubropictum	Baetid mayfly	2c
P. rufostrigatum	Baetid mayfly	11b
Pseudocloeon sp.	Baetid mayfly	14, 15, 21
Baetiscidae		
Baetisca sp.	Baetiscid mayfly	11b
B. lacustris (=*bajkovi*)	Baetiscid mayfly	2c, 11b, 15
B. obesa	Baetiscid mayfly	11b
Caenidae	Caenid mayfly	
Bracycercus sp.	Caenid mayfly	14
Caenis sp.	Caenid mayfly	2c, 8, 12, 14, 18, 21
Ephemeridae	Burrowing mayfly	
Hexagenia sp.	Burrowing mayfly	18
H. atrocaudata	Burrowing mayfly	8c
H. limbata	Burrowing mayfly	15
Ephemerellidae	Ephemerellid mayfly	12
Ephemerella sp.	Ephemerellid mayfly	18

Table 2. Aquatic macroinvertebrate taxa and distributions within the Strawberry River System. (cont.)

Scientific Name	Common Name	Location (Table 1)
E. argo	Ephemerellid mayfly	15
E. deficiens	Ephemerellid mayfly	15
E. dorothea	Ephemerellid mayfly	1b, 2c, 3h, 11b, 12, 15
E. invaria	Ephemerellid mayfly	11b
E. needhami	Ephemerellid mayfly	2c, 11b, 15
E. rotunda	Ephemerellid mayfly	11b
E. simplex	Ephemerellid mayfly	15
E. sordida	Ephemerellid mayfly	15
Seratella sp.	Ephemerellid mayfly	10b
S. sordida	Ephemerellid mayfly	12
Heptageniidae	Heptageniid mayfly	
Heptagenia sp.	Heptageniid mayfly	18
Nixe sp.	Heptageniid mayfly	18
Stenacron interpunctatum	Heptageniid mayfly	1b, 3g, 18
Stenonema sp.	Heptageniid mayfly	10, 14, 18, 21
S. ares	Heptageniid mayfly	15
S. femoratum	Heptageniid mayfly	1a, 3h, 6, 7, 9c, 11b, 15, 18
S. luteum	Heptageniid mayfly	2c, 3h, 11b
S. mediopunctatum	Heptageniid mayfly	1b, 2c, 3h, 11b, 12, 18
S. nepotellum	Heptageniid mayfly	15
S. pulchellum	Heptageniid mayfly	18
S. terminatum	Heptageniid mayfly	7, 11b, 12, 18
Isonychiidae	Isonychiid mayfly	
Isonychia sp.	Isonychiid mayfly	1b, 2b, 2c, 3g, 3h, 5, 6, 10, 11b, 12, 14, 15, 18, 21
Leptophlebiidae	Leptophlebiid mayfly	2c
Choroterpes sp.	Leptophlebiid mayfly	18
C. basalis	Leptophlebiid mayfly	5, 6
Paraleptophlebia sp.	Leptophlebiid mayfly	15
P. guttata	Leptophlebiid mayfly	1b, 2c, 3h, 11b
Polymitarcyidae	Burrowing mayfly	
Ephoron sp.	Burrowing mayfly	14, 21
Potamanthidae	Burrowing mayfly	
Anthopotamus (=Potamanthus) sp.	Burrowing mayfly	12, 14, 15
Siphloneuridae	Siphloneurid mayfly	
Ameletus sp.	Siphloneurid mayfly	3h
Siphloneurus sp.	Siphloneurid mayfly	11b
S. marshalli	Siphloneurid mayfly	2c
Tricorythidae	Tricorythid mayfly	
Tricorythodes sp.	Tricorythid mayfly	6, 18, 21
T. atratus	Tricorythid mayfly	3h, 11b, 15
Order Odonata	Dragonflies, damselflies	
Calopterygidae	Broad-winged damsels	
Calopteryx maculata	Ebony jewelwing	1b, 2a, 2b, 2c, 3g, 5, 10, 11b, 18

Table 2. Aquatic macroinvertebrate taxa and distributions within the Strawberry River System. (cont.)

Scientific Name	Common Name	Location (Table 1)
Hetaerina sp.	Rubyspot damselfly	2b, 11b
H. americana	American rubyspot	2c, 3e, 9b, 9c, 18
H. titia	Smoky rubyspot	2a
Coenagrionidae	Pond damsels	
Amphiagrion sp.?	Red damsel	15 (probably *Ischnura)*
Argia sp.	Dancer	1a, 2b, 2c, 4, 5, 6, 9c, 10, 11b, 12, 15, 18
A. apicalis	Blue-fronted dancer	9b
A. fumipennis violacea	Variable dancer	9b
A. moesta	Powdered dancer	9b
A. plana	Springwater dancer	9b, 10
A. sedula	Blue-ringed dancer	2a, 3e, 9b
A. tibialis	Blue-tipped dancer	9b, 9d
A. translata	Dusky dancer	9b
Enallagma sp.	Bluet	1a, 2b, 2c, 4, 6, 7, 8, 9c, 11b, 12, 18
Ischnura sp.	Forktail	4, 6, 7, 15, 18
I. posita	Fragile forktail	9b
Aeshnidae	Darners	14, 18
Basiaeschna janata	Springtime darner	2b, 3g, 3h, 4, 5, 6, 9c, 10, 18
Boyeria vinosa	Fawn darner	2b, 10, 12
Gomphidae	Clubtails	
Arigomphus lentulus	Stillwater clubtail	9c
Dromogomphus sp.	Spinyleg clubtail	1a, 2b, 3g, 12
D. spinosus	Black-shouldered spinyleg	3e, 3h, 4, 9a, 9c, 15
D. spoliatus	Flag-tailed spinyleg	3h
Erpetogomphus designatus	Eastern ringtail dragonfly	9c
Gomphus sp.	Clubtail	2b, 2c, 4, 9c, 10, 11b
G. externus	Plains clubtail	12
G. graslinellus?	Pronghorn clubtail	5
G. lividus	Ashy clubtail	11a
G. ozarkensis	Ozark clubtail	13
Hagenius brevistylus	Dragonhunter	2c, 3c, 3g, 9a, 11b, 12
Ophiogomphus westfalli	Westfall's snaketail	1a, 2b
Progomphus obscurus	Common sanddragon	2b, 2c, 3b, 3g, 3h, 11b
Stylogomphus sigmastylus	Western least clubtail	2c, 3g, 6, 7, 9c, 10, 18
Stylurus plagiatus	Russet-tipped clubtail	11a
Corduligastridae	Spiketails	
Cordulegaster maculata?	Twin-spotted spiketail	10
Macromiidae	Cruisers	
Didymops transversa	Stream cruiser	11b
Macromia sp.	River cruiser	1a, 2b, 2c, 3g, 9c, 11b, 12, 14, 18
M. illinoiensis	Illinois river cruiser	11a, 15
Corduliidae	Emeralds	
Epitheca cynosura	Common baskettail	1a, 4

Table 2. Aquatic macroinvertebrate taxa and distributions within the Strawberry River System. (cont.)

Scientific Name	Common Name	Location (Table 1)
E. princeps	Prince baskettail	2b, 4, 9c
Neurocordulia xanthosoma	Orange shadowdragon	3a, 9d
Somatochlora linearis	Mocha emerald	5, 8
Libellulidae	Skimmers	15, 18
Dythemis velox	Swift setwing	9a
Erythemis simplicicollis	Eastern pondhawk	3c, 4
Libellula sp.	Skimmer	1a, 11b
L. cyanea	Eastern spangled skimmer	3b, 4
L. luctuosa	Pied skimmer	4, 8
L. vibrans	Great blue skimmer	3c, 4, 9a
Plathemis lydia	Common whitetail	4, 5, 9a
Sympetrum sp.	Meadowhawk	1a
S. ambiguum	Blue-faced meadowhawk	9b
Order Plecoptera	Stoneflies	
Capniidae		
Allocapnia granulata	Capniid stonefly	17
A. mohri	Capniid stonefly	17
A. mystica	Capniid stonefly	17
A. rickeri	Capniid stonefly	17
Leuctridae	Winter stoneflies	
Leuctra tenuis	Winter stonefly	17
Zealeuctra classeni	Winter stonefly	17
Z. narfi	Winter stonefly	17
Z. warreni	Winter stonefly	17
Taeniopterygidae	Taeniopterygid stoneflies	
Strophopteryx sp.	Taeniopterygid stonefly	2c
S. cucullata	Taeniopterygid stonefly	17
S. fasciata	Taeniopterygid stonefly	17
Taeniopteryx sp.	Taeniopterygid stonefly	3h
T. burksi	Taeniopterygid stonefly	17
Nemouridae	Nemourid stoneflies	
Amphinemura sp.	Nemourid stonefly	1b
A. delosa	Nemourid stonefly	17
Prostoia sp.	Nemourid stonefly	1b
P. completa	Nemourid stonefly	17
Chloroperlidae	Chloroperlid stoneflies	
Haploperla sp.	Chloroperlid stonefly	1b
H. brevis	Chloroperlid stonefly	17
Perlidae	Perlid stoneflies	
Acroneuria sp.	Perlid stonefly	2c, 3h, 18
A. evoluta	Perlid stonefly	17
A. perplexa	Perlid stonefly	17
Agnetina capitata	Perlid stonefly	11b

Table 2. Aquatic macroinvertebrate taxa and distributions within the Strawberry River System. (cont.)

Scientific Name	Common Name	Location (Table 1)
Anacroneuria sp.	Perlid stonefly	18
Attaneuria ruralis	Perlid stonefly	2c, 12
Neoperla sp.	Perlid stonefly	17, 18, 21
N. choctaw	Perlid stonefly	17
N. clymene	Perlid stonefly	14, 15, 17
N. falayah	Perlid stonefly	17
N. osage	Perlid stonefly	2c, 17
N. robisoni	Perlid stonefly	17
Paragnetina kansensis	Perlid stonefly	17
Perlesta sp.	Perlid stonefly	1b, 15
P. cinctipes	Perlid stonefly	17
P. decipiens	Perlid stonefly	17
P. shubuta	Perlid stonefly	17
Perlinella drymo	Perlid stonefly	17
P. ephyre	Perlid stonefly	17
Phasganophora sp.	Perlid stonefly	18
Perlodidae	Perlodid stoneflies	
Hydroperla crosbyi	Perlodid stonefly	17
Isoperla sp.	Perlodid stonefly	1b, 2c, 3h, 15
I. coushatta	Perlodid stonefly	2c, 11b, 17
I. dicala	Perlodid stonefly	2c, 12
I. mohri	Perlodid stonefly	2c, 17
I. namata	Perlodid stonefly	17
I. ouachita	Perlodid stonefly	17
Order Hemiptera	True bugs	
Belostomatidae	Giant water bugs	
Sp. 1	Giant water bug	18
Corixidae	Water boatmen	15
Hesperocorixa sp.	Water boatman	5, 8, 18
Sigara sp.	Water boatman	5, 7, 8, 10, 18
S. modesta	Water boatman	1a, 18
Trichocorixa sp.	Water boatman	4, 9c, 18
Gelastocoridae	Toad bugs	
Gelastocoris oculatus oculatus	Toad bug	4, 5, 18
Gerridae	Water striders	
Gerris marginatus	Water strider	1a, 15
G. nebularis	Water strider	15
G. remigis	Water strider	7, 10
Limnoporus canaliculatus	Water strider	5, 6, 8, 9c, 10
Metrobates hesperius	Water strider	2b
Neogerris sp.	Water strider	18
Rheumatobates sp.	Water strider	6, 18
Trepobates sp.	Water strider	1a, 8

Table 2. Aquatic macroinvertebrate taxa and distributions within the Strawberry River System. (cont.)

Scientific Name	Common Name	Location (Table 1)
T. pictus	Water strider	4
T. subnitidus?	Water strider	6
Hebridae	Velvet water bugs	
Hebrus sp.	Velvet water bug	18
H. concinnus	Velvet water bug	6
Hydrometridae	Water measurers	
Hydrometra hungerfordi	Water measurer	5
H. martini	Water measurer	2b, 3f
Mesoveliidae	Water treaders	
Mesovelia sp.	Water treader	1a, 2b, 3g, 18
M. mulsanti	Water treader	6
Naucoridae	Creeping water bugs	
Pelocoris sp.	Creeping water bug	2c
Nepidae	Water scorpions	
Ranatra kirkaldyi	Water scorpion	1a, 4
Notonectidae	Backswimmers	
Notonecta indica	Backswimmer	5
Pleidae	Pygmy backswimmers	
Neoplea sp.	Pygmy backswimmer	4, 18
Veliidae	Broad-shouldered water strider	
Microvelia sp.	Small water strider	1a, 2c, 3g, 18
M. americana	Small water strider	5, 7, 10
M. hinei	Small water strider	8
Rhagovelia sp.	Broad-shouldered water strider	2b, 3g, 18
R. knighti	Broad-shouldered water strider	6, 10
Steinovelia stagnalis	Small water strider	4
Order Megaloptera	Alderflies, dobsonflies, fishflies	
Corydalidae	Dobsonflies, fishflies	
Chauliodes pectinicornis	Fishfly	7, 9c
Corydalus cornutus	Hellgrammite	2b, 2c, 3h, 7, 12, 14, 15, 18
Nigronia serricornis	Fishfly	3g
Sialidae	Alderflies	
Sialis sp.	Alderfly	1a, 4, 5, 7, 9c, 10, 14, 18
Order Trichoptera	Caddisflies, caddisworms	
Glossosomatidae	Glossosomatid caddisflies	
Agapetus illini	Glossosomatid caddisfly	20
Helicopsychidae	Helicopsychid caddisflies	
Helicopsyche sp.	Helicopsychid caddisfly	2c, 10, 18, 21
H. borealis	Helicopsychid caddisfly	20
Hydropsychidae	Hydropsychid caddisflies	
Ceratopsyche bronta	Hydropsychid caddisfly	20
C. morosa	Hydropsychid caddisfly	20
Cheumatopsyche sp.	Hydropsychid caddisfly	1b, 2b, 10, 14, 15, 18, 21

Table 2. Aquatic macroinvertebrate taxa and distributions within the Strawberry River System. (cont.)

Scientific Name	Common Name	Location (Table 1)
C. campyla	Hydropsychid caddisfly	20
C. miniscula	Hydropsychid caddisfly	20
C. oxa	Hydropsychid caddisfly	20
C. pettiti	Hydropsychid caddisfly	20
Hydropsyche sp.	Hydropsychid caddisfly	2c, 11b, 12
Macrostemum (=Macromia) sp.	Hydropsychid caddisfly	15
Hydroptilidae	Hydroptilid caddisflies	
Sp. 1	Hydroptilid caddisfly	18
Leptoceridae	Leptocerid caddisflies	
Ceraclea cancellata	Leptocerid caddisfly	20
C. tarsipunctata	Leptocerid caddisfly	20
C. transversa	Leptocerid caddisfly	20
Oecetis sp.	Leptocerid caddisfly	15
O. inconspicua	Leptocerid caddisfly	20
O. persimilis	Leptocerid caddisfly	20
Triaenodes flavescens	Leptocerid caddisfly	20
T. ignitus	Leptocerid caddisfly	20
Limnephilidae	Limnephilid caddisflies	
Pycnopsyche sp.	Limnephilid caddisfly	2b, 3g, 3h, 18
Philopotamidae	Philopotamid caddisflies	
Chimarra sp.	Philopotamid caddisfly	1b, 3h, 5, 7, 10, 14, 15, 18, 21
C. feria	Philopotamid caddisfly	20
Polycentropodidae	Polycentropodid caddisflies	
Cyrnellus sp.	Polycentropodid caddisfly	3h
Neureclipsis sp.	Polycentropodid caddisfly	3h
Polycentropus sp.	Polycentropodid caddisfly	15, 20, 21
Psychomyiidae	Psychomyiid caddisflies	
Psychomyia sp.	Psychomyiid caddisflies	21
P. flavida	Psychomyiid caddisfly	20
Rhyacophilidae	Rhyacophilid caddisflies	
Rhyacophila sp.	Rhyacophilid caddisfly	1b
R. fenestra	Rhyacophilid caddisfly	20
Order Lepidoptera	Butterflies, moths	
Pyralidae	Pyralid moths	
Petrophila (=*Elophila*) sp.	Pyralid moth	14, 15, 18
Order Coleoptera	Beetles	
Anthicidae	Anthicid beetles	
Sp. 1	Anthicid beetle	7
Curculionidae	Weevils	
Lixus sp.	Weevil	18
Dryopidae	Long-toed water beetles	
Helichus sp.	Long-toed water beetle	1a, 1b, 2b, 2c, 3g, 10, 11b, 18
H. lithophilus	Long-toed water beetle	5, 6, 7, 8, 9c

Table 2. Aquatic macroinvertebrate taxa and distributions within the Strawberry River System. (cont.)

Scientific Name	Common Name	Location (Table 1)
Dytiscidae	Predaceous diving beetles	
Agabus ambiguous?	Predaceous diving beetle	7
Copelatus chevrolati renovatusi	Predaceous diving beetle	6
Coptotomus venustus	Predaceous diving beetle	7, 9c
Heterosternuta ouachitus	Predaceous diving beetle	1a, 6, 8
H. pulcher	Predaceous diving beetle	1a, 6, 8
H. wickhami	Predaceous diving beetle	1a, 2b, 6, 8
Hydroporus sp. 1	Predaceous diving beetle	2b, 3g, 3h, 4, 5, 7, 9c, 10, 18
H. sp. 2	Predaceous diving beetle	4, 5, 7, 9c, 10
H. sp. 3	Predaceous diving beetle	4, 5
H. sp. 4	Predaceous diving beetle	4
H. sp. 5	Predaceous diving beetle	4
H. sp. 6	Predaceous diving beetle	4
H. rufilabris	Predaceous diving beetle	1a, 6
Hydrovatus sp.	Predaceous diving beetle	4
Ilybius biguttulus	Predaceous diving beetle	5
Laccophilus sp.	Predaceous diving beetle	3h
L. fasciatus rufus	Predaceous diving beetle	5, 7, 10
L. maculatus maculatus	Predaceous diving beetle	4, 5
L. proximus proximus	Predaceous diving beetle	4, 6
Neobidessus sp.	Predaceous diving beetle	5
Neoporus blanchardi	Predaceous diving beetle	2b
N. clypealis	Predaceous diving beetle	1a, 8
N. dimidiatus	Predaceous diving beetle	6, 8
N. shermani	Predaceous diving beetle	1a, 2b, 6, 8
N. striatopunctipennis	Predaceous diving beetle	2b
N. undulatus	Predaceous diving beetle	8
Thermonectus basillaris	Predaceous diving beetle	8
Uvarus sp.	Predaceous diving beetle	4, 11b, 18
Elmidae	Riffle beetles	3h, 14, 15
Ancyronyx variegata	Riffle beetle	2c
Dubiraphia sp. 1	Riffle beetle	2b, 9c, 18
D. sp. 2	Riffle beetle	18
Macronychus glabratus	Riffle beetle	1b, 2b, 2c, 3g, 7
Optioservus sp.	Riffle beetle	18
Stenelmis sp.	Riffle beetle	1a, 1b, 2b, 3g, 11b, 18, 21
S. crenata	Riffle beetle	6
Gyrinidae	Whirligig beetles	
Dineutus sp.	Whirligig beetle	18, 21
Dineutus carolinus	Whirligig beetle	9c
D. ciliatus	Whirligig beetle	1a, 3h, 5, 6, 7
D. discolor	Whirligig beetle	15
D. emarginatus	Whirligig beetle	9c

Table 2. Aquatic macroinvertebrate taxa and distributions within the Strawberry River System. (cont.)

Scientific Name	Common Name	Location (Table 1)
D. serrulatus	Whirligig beetle	2c, 3h
Gyretes sinuatus	Whirligig beetle	15
Gyrinus sp.	Whirligig beetle	1a, 15
G. woodruffi	Whirligig beetle	9c
Haliplidae	Crawling water beetles	
Haliplus fasciatus	Crawling water beetle	8
Peltodytes dispersus	Crawling water beetle	8
P. dunavani	Crawling water beetle	1a, 4, 6, 8, 18
P. duodecimpunctatus	Crawling water beetle	1a, 2c, 3h, 4, 5, 6, 7, 8, 12, 18
P. festivus	Crawling water beetle	4
P. litoralis	Crawling water beetle	1a, 4, 18
P. muticus	Crawling water beetle	4
P. sexmaculatus	Crawling water beetle	1a, 4, 11b, 18
Helophoridae	Water scavenger beetle	
Helophorus sp.	Water scavenger beetle	18
Hydrophilidae	Water scavenger beetles	
Sphaeridiinae	Water scavenger beetle	1a, 3g
Berosus sp. 1	Water scavenger beetle	1a, 4, 18
B. sp. 2	Water scavenger beetle	4
Crenitus? sp.	Water scavenger beetle	5
Enochrus sp.	Water scavenger beetle	4, 9c, 18
E. ochraceus	Water scavenger beetle	1a, 8
E. perplexus	Water scavenger beetle	6
E. pygmaeus nebulosus	Water scavenger beetle	1a, 2b, 8, 18
Helochares sp.	Water scavenger beetle	1a, 4, 5, 18
Hydrochus sp.	Water scavenger beetle	1a, 4
Laccobius sp.	Water scavenger beetle	1a, 4, 7
Paracymus sp.	Water scavenger beetle	1a, 18
Tropisternus sp.	Water scavenger beetle	18
T. collaris mexicanus	Water scavenger beetle	4, 5
T. ellipticus	Water scavenger beetle	7, 18
T. glaber	Water scavenger beetle	7, 9
T. lateralis nimbatus	Water scavenger beetle	4, 8
T. natator	Water scavenger beetle	1a, 4, 5, 6, 7, 8, 9c, 10
Lutrochidae	Marsh-loving beetles	
Lutrochus laticeps	Marsh-loving beetle	18
Noteridae	Burrowing water beetles	
Hydrocanthus atripennis	Burrowing water beetle	2b
Psephenidae	Water pennies	
Ectopria nervosa	Water penny	3g, 3h
Psephenus herricki	Water penny	3g, 14, 15, 18
Scirtidae (=Helodidae)	Marsh beetles	15
Scirtes sp.	Marsh beetle	6

Table 2. Aquatic macroinvertebrate taxa and distributions within the Strawberry River System. (cont.)

Scientific Name	Common Name	Location (Table 1)
Order Diptera	True flies	
Athericidae	Athericid flies	
Atherix sp.	Athericid fly	10
Ceratopogonidae	Biting midges	15
Sp. 1	Biting midge	18
Bezzia or *Probezzia*	No see'um, punkie	4
Chaoboridae	Phantom midges	
Chaoborus sp.	Phantom midge	18
Chironomidae	Non-biting midges, bloodworms	
Procladius sp.	Bloodworm	3h
Sp. 1	Bloodworm	1a, 2b, 3g, 4, 5, 6, 7, 8, 9c, 10, 11b, 12, 14, 15, 18, 21
Dixidae	Dixid midges	
Dixella sp.	Dixid midge	18
Dolichopodidae	Dolichopodid fly	
Rhaphium sp.	Dolichopodid fly	1a
Empididae	Dance flies	
Hemerodromia	Dance fly	21
Ptychopteridae	Phantom craneflies	
Bittacomorpha clavipes	Phantom cranefly	7
Sciomyzidae	Sciomyzid flies	
Sepedon sp.	Sciomyzid fly	9c
Simuliidae	Blackflies	2c
Prosimulium sp.	Blackfly	3h
Simulium sp.	Blackfly	1b, 14, 15, 18, 21
Stratiomyidae	Soldier flies	
Stratiomys sp.	Soldier fly	1a, 2b, 4, 10, 18
Tabanidae	Horseflies, deerflies	2c, 14
Apatolestes sp.	Horsefly	11b, 12
Chrysops sp.	Horsefly	3g, 6
Hybomitra sp.	Horsefly	1b
Tabanus sp.	Horsefly	9c, 21
Tipulidae	Craneflies	
Hexatoma sp.	Cranefly	1a, 21
Tipula sp.	Cranefly	1a, 1b, 3g, 3h, 10, 11b, 12, 18

Total taxa = 313

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Electron Shock Waves

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Abstract.—In this paper we describe numerical investigations of breakdown waves concentrating on antiforce waves. We employed one-dimensional electron fluid dynamical equations for a luminous pulse wave propagating into a neutral gas region and subjected to an applied electric field. We assumed that the electrons were the main element in the propagation of the wave and that the electron gas partial pressure provided the driving force. These waves are considered to be shock fronted and are composed of two regions: the thin sheath region behind the shock front and the thicker quasi-neutral region following the sheath region. Our set of equations, known as the electron fluid dynamical (EFD) equations, is composed of the equations of conservation of mass, momentum, and energy coupled with Poisson's equation. For antiforce waves, we were able to successfully integrate the set of EFD equations through the sheath region using a set of initial boundary conditions at the wave front. By using values of electron gas temperature, electron number density, ionization rate, and also the existing conditions at the end of the sheath region as initial boundary values for the thermal region of the gas, we were able to integrate the electron fluid dynamical equations, modified for the thermal region of the gas, through that region. Our results satisfy the required conditions at the end of the sheath and quasi-neutral regions. The wave profiles for electric field, electron velocity, electron number density, electron gas temperature, and ionization rate within the sheath and quasi-neutral regions were determined.

Key words.—Breakdown waves, one-dimensional electron fluid dynamical (EFD) equations, luminous pulse wave, electrons.

Introduction

Lightning has always been an awe-inspiring event that has intrigued and perplexed men since the beginning of history. This is the pinnacle example of luminous fronts, or pulses, generated by potential differences between two points in a gas. Von Zahn (1879) proposed lack of Doppler shift in the radiation emitted from breakdown waves, inferring negligible mass motion within the pulse. Thomson (1893) made the observation that breakdown waves moved at approximately half the speed of light, rather than instantaneously jumping from one point to another.

Beams (1930) then proved Thomson's observations correct and proposed an explanation for this phenomenon: that the gas behind the pulse was electrically conductive, thereby carrying a potential and creating a breakdown of the gas in the given area as the wave propagates. He also explained that because of the large mass difference between the positive ions and the electrons, the positive ions, as compared to the electrons, will have a negligible increase in speed. This explanation is still accepted today. Paxton and Fowler (1962) applied a three-fluid, hydrodynamical model to formulate a set of equations describing the wave propagation.

Shelton and Fowler (1968) continued this work and described the phenomena as "electron fluid dynamical" waves. They developed a set of one-dimensional equations describing the phenomena, deriving equations for energy and momentum loss and gain terms during the electron collisions with heavy particles. Their main concern was proforce waves, waves for which the electric field force on electrons is in the same direction as the direction of the propagation of the pulse. We will concentrate on antiforce waves, waves for which the electric field force on electrons is in the opposite direction of the propagation of the wave. Using an approximation method, Fowler and Shelton (1973) solved their set of electron fluid dynamical equations for the dynamical transition region of the wave. Their approximate solutions are in good agreement with experimental data available (Blais and Fowler 1973).

Later, Sanmann and Fowler (1975) tried to account for the propagation of antiforce waves. They considered the electron gas partial pressure to be much larger than that of the other species, therefore providing the driving force for the propagation of the wave. Fowler et al. (1984) completed the set of electron fluid dynamical equations by adding terms to the equation of conservation of energy, which proved to be essential for exact numerical solution of the set of electron fluid dynamical equations. They also developed a computer program to integrate the equations through the sheath region. Hemmati (1999) completed the set of electron fluid dynamical equations representing the antiforce waves. Rakov (2000) provided a complete set of experimental results for the wave speed, current, and charge in his review of positive and bipolar lightning discharges.

Analysis

The equations which were fully developed by Fowler et al. (1984), representing a one-dimensional, steady state, constant velocity, electron fluid dynamical wave propagating into a neutral medium, are the equations of conservation of mass, momentum, and energy coupled with Poisson's equation:

$$\frac{d(n\upsilon)}{dx} = n\beta, \qquad [1]$$

$$\frac{d}{dx}[mnv(\upsilon - V) + nkT_e] = -enE - Kmn(\upsilon - V), \quad [2]$$

$$\frac{d}{dx}[mn\upsilon(\upsilon - V)^2 + nkT_e(5\upsilon - 2V) + 2en\upsilon\Phi - \frac{5nk^2T_e}{mK}\frac{dT_e}{dx}]$$
$$= -3(\frac{m}{M})nkKT_e - (\frac{m}{M})Kmn(\upsilon - V)^2, \qquad [3]$$

$$\frac{dE}{dx} = \frac{e}{\varepsilon_0}n(\frac{\upsilon}{V} - 1); \qquad [4]$$

where n, v, T_e, e, and m are the electron number density, velocity, temperature, charge, and mass, respectively. M, E, E_o, V, k, K, x, β, and φ are neutral particle mass, electric field within the sheath region, electric field at the wave front, wave velocity, Boltzmann's constant, elastic collision frequency, position within the sheath region, ionization frequency, and ionization potential of the gas, respectively.

To reduce the set of electron fluid dynamical equations to non-dimensional form, the following set of dimensionless variables are introduced:

$$\eta = \frac{E}{E_o}, \nu = (\frac{2e\phi}{\varepsilon_o E_o^2})n, \psi = \frac{v}{V}, \theta = \frac{T_e k}{2e\phi}, \xi = \frac{eE_o x}{mV^2},$$
$$\alpha = \frac{2e\phi}{mV^2}, \kappa = \frac{mV}{eE_o}K, \mu = \frac{\beta}{K}, \omega = \frac{2m}{M},$$

where η, ν, ψ, θ, μ, and ξ represent the dimensionless net electric field (applied plus space charge field), electron number density, electron velocity, electron gas temperature, ionization rate, and position within the sheath region, respectively. α and κ are wave parameters.

Substituting the dimensionless variables in equations (1-4) reduces them to the following form:

$$\frac{d(\nu\psi)}{d\xi} = \kappa\mu\nu, \qquad [5]$$

$$\frac{d}{d\xi}[\nu\psi(\psi - 1) + \alpha\nu\theta] = -\nu\eta - \kappa\nu(\psi - 1), \qquad [6]$$

$$\frac{d}{d\xi}[\nu\psi(\psi - 1)^2 + \alpha\nu\theta(5\psi - 2) + \alpha\nu\psi + \alpha\eta^2 - \frac{5\alpha^2\nu\theta}{\kappa}\frac{d\theta}{d\xi}] =$$
$$-\omega\kappa\nu[3\alpha\theta + (\psi - 1)^2], \qquad [7]$$

$$\frac{d\eta}{d\xi} = \frac{\nu}{\alpha}(\psi - 1). \qquad [8]$$

Shelton and Fowler (1968) proposed the existence of two distinct regions within the wave: the sheath and the quasi-neutral region. In the sheath region, electron velocity, starting from an initial value at the shock front, reduces to speeds comparable to those of heavy particles. Also, the electric field, starting with its maximum value at the shock front, reduces to a negligible value at the trailing edge of the sheath. These conditions translate into the following equation form:

$$\psi_2 = 1, \eta_2 = 0, \psi_2' = 0, \text{ and } \eta_2' = 0, \qquad [9]$$

where, ψ_2, η_2, ψ_2', and η_2' are the non-dimensional electron velocity, electric field, electron velocity derivative, and electric field derivative at the end of the sheath region, respectively.

In the quasi-neutral region, through further ionization of neutral particles, the electron gas cools to near room temperatures. Therefore, the electric field energy present ahead of the wave is converted to ionization energy behind the wave. In non-dimensional form the expected conditions at the end of the quasi-neutral region are as follows:

$$\nu_f = 1 \text{ and } \theta_f = 0.065.$$

All of our attempts at integrating equations 5-8 through the quasi-neutral region failed. Equations 5-8 were derived by combining the primitive forms of the fluid equations, and since we were not using approximation methods for solving the set of fluid equations, there was no need for the combined form of the equations. Therefore, for our investigation of the quasi-neutral region of the wave, we chose the primitive form of the electron-fluid dynamical equations:

$$\frac{d(\nu\psi)}{d\xi} = \kappa\mu\nu, \qquad [10]$$

$$\frac{d}{d\xi}[\nu\psi^2 + \alpha\nu\theta] = -\nu\eta - \kappa\nu(\psi - 1) + \kappa\mu\nu, \qquad [11]$$

$$\frac{d}{d\xi}(\nu\psi^3 + 5\nu\psi\alpha\theta - \frac{5\alpha^2\nu\theta}{\kappa}\frac{d\theta}{d\xi}) = -2\nu\psi\eta - 2\kappa\nu(\psi-1) + \kappa\mu\nu(\psi-1) - \omega\kappa\nu[3\alpha\theta + (\psi-1)^2], \quad [12]$$

$$\frac{d\eta}{d\xi} = \frac{\nu}{\alpha}(\psi - 1). \quad [13]$$

By applying the expected conditions at the end of the sheath region [9] in the expanded forms of the equations of conservation of mass and momentum [10-11], the equations describing the quasi-neutral region become

$$\nu_2' = \kappa\mu_2\nu_2 \quad [14]$$

and

$$\theta_2' = -\kappa\mu_2\theta_2 \quad [15]$$

where v′2 and θ′ 2 are the electron number density derivative and electron gas temperature derivative in the quasi-neutral region.

By integrating equations 5-8 through the sheath region, one can find the electron number density, electron gas temperature, and ionization rate values at the end of the sheath region. These variables now become the initial boundary conditions for the quasi-neutral region. We have been able to successfully integrate equations 14-15 through the quasi-neutral region of the wave and our results meet the expected conditions at the trailing edge of the wave ($\nu_f = 1$ and $\theta_f = 0.065$).

For antiforce waves, slight changes in the electron fluid dynamical equations need to be made. For an observer stationary relative to the wave front, the heavy particles move in the negative x direction ($V < 0$, $E_o > 0$, and $K > 0$). Therefore, both κ and ξ will be intrinsically negative. For antiforce waves, therefore, the set of dimensionless variables is slightly different and has been derived by Hemmati (1999):

$$\eta = \frac{E}{E_o}, \nu = (\frac{2e\phi}{\varepsilon_o E_o^2})n, \psi = \frac{v}{V}, \theta = \frac{T_e k}{2e\phi}, \xi = -\frac{eE_o x}{mV^2},$$

$$\alpha = \frac{2e\phi}{mV^2}, \kappa = -\frac{mV}{eE_o}K, \mu = \frac{\beta}{K}, \omega = \frac{2m}{M}.$$

The equations describing antiforce waves in non-dimensional form are therefore

$$\frac{d}{d\xi}[\nu\psi] = \kappa\mu\nu, \quad [16]$$

$$\frac{d}{d\xi}[\nu\psi(\psi - 1) + \alpha\nu\theta] = \nu\eta - \kappa\nu(\psi - 1), \quad [17]$$

$$\frac{d}{d\xi}[\nu\psi(\psi-1)^2 + \alpha\nu\theta(5\psi - 2) + \alpha\nu\psi + \alpha\eta^2 - \frac{5\alpha^2\nu\theta}{\kappa}\frac{d\theta}{d\xi}] = -\omega\kappa\nu[3\alpha\theta + (\psi-1)^2], \quad [18]$$

$$\frac{d\eta}{d\xi} = -\frac{\nu}{\alpha}(\psi - 1). \quad [19]$$

Results

The electron velocity at the wave front is less than the wave velocity ($v_1 < V$). The dimensionless electron velocity at the wave front, ψ_1, therefore must be less than 1. As a result, according to the Poisson's equation, the electric field will have a positive slope behind the wave front resulting in an initial increase in the electric field. Heading through the sheath region following the shock front, the electric field will increase until the electrons gain speed in excess of ion speeds. The dimensionless electron velocity will then become larger than 1, making the electric field slope negative. The electric field therefore decreases (Hemmati 1995) until the electrons slow down to speeds comparable to ion speeds at the end of the sheath region ($\psi_2 \rightarrow 1$), requiring that the electric field and its slope approach zero at the end of the sheath as well ($\eta_2 \rightarrow 0, \eta_2' \rightarrow 0$).

We used a trial and error method to integrate equations 16-19. For a given wave speed, α, we chose a set of values for wave constant, κ, electron velocity, ψ_1, and electron number density, ν_1, at the shock front. We repeatedly changed the values of κ, ψ_1, and ν_1, in the process of integration of equations 16-19 so that the process led to a conclusion in agreement with the expected conditions [9] at the end of the sheath region. As in the proforce case, we used the conditions at the end of the sheath region to find the equations describing the quasi-neutral region. For integrating the set of equations describing the quasi-neutral region, we used electron temperature, electron number density, and ionization rate values at the end of the sheath region as initial boundary values for the quasi-neutral region. For antiforce waves, we were successful in integrating the electron-fluid dynamical equations through both the sheath and quasi-neutral regions for two values of wave speeds, $\alpha = 0.05$ and $\alpha = 1$, representing wave velocities of 1.33×10^7 m/s and 2.96×10^6 m/s, respectively. For $\alpha = 0.05$, the initial boundary conditions required were $\kappa = 0.35$, $\psi_1 = 0.95$, and $\nu_1 = 0.09$. For $\alpha = 1$, the conditions required at the shock front were $\kappa = 0.17$, $\psi_1 = 0.98$, and $\nu_1 = 0.40$.

Figure 1 represents electric field, η, as a function of position, ξ, inside the sheath region. As it can be seen, the electric field approaches zero as it nears the end of the sheath. For α =

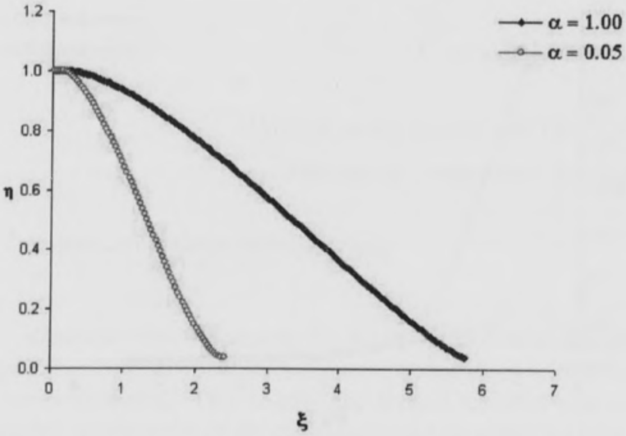

Fig. 1. Electric field, η, as a function of position, ξ, inside the sheath region for $\alpha = 0.05$ and $\alpha = 1$.

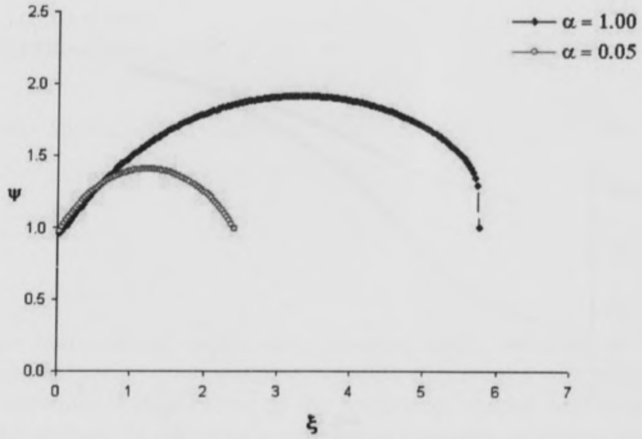

Fig. 2. Electron velocity, ψ, as a function of position, ξ, inside the sheath region for $\alpha = 0.05$ and $\alpha = 1$.

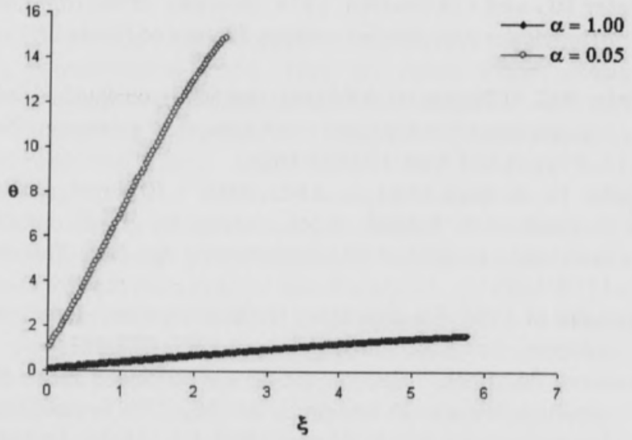

Fig. 3. Electron temperature, θ, as a function of position, ξ, inside the sheath region for $\alpha = 0.05$ and $\alpha = 1$.

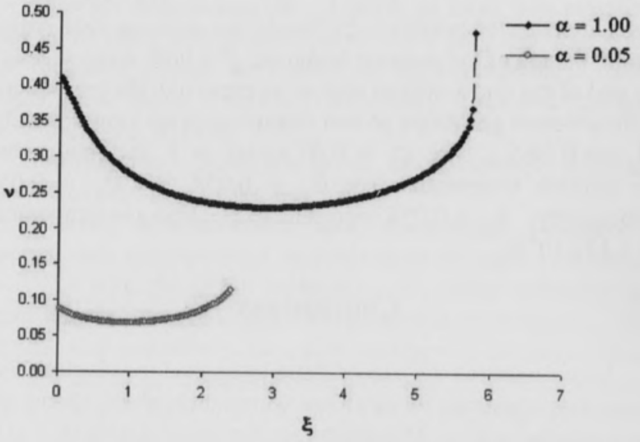

Fig. 4. Electron number density, ν, as a function of position, ξ, inside the sheath region for $\alpha = 0.05$ and $\alpha = 1$.

0.05 and $\alpha = 1$, the sheath region goes to $\xi = 2.41$ and $\xi = 5.75$, respectively; representing sheath thicknesses of 2.43×10^{-6} m and 2.87×10^{-5} m, respectively. Figure 2 represents electron velocity, ψ, as a function of position, ξ, inside the sheath region. As expected, the dimensionless velocity goes to one as it approaches the end of the sheath region.

Figure 3 represents electron temperature, θ, as a function of position, ξ, inside the sheath region. For $\alpha = 0.05$ and $\alpha = 1$, the electron temperature goes to $\theta = 14.868$ and $\theta = 1.623$, respectively at the end of the sheath region. $\theta = 14.868$ and $\theta = 1.623$ represent electron gas temperatures of 8.62×10^{6} K and 9.41×10^{5} K, respectively. Figure 4 represents electron number density, ν, as a function of position, ξ, inside the sheath region. For $\alpha = 0.05$ and $\alpha = 1$, the electron number density goes to $\nu = 0.1145$ and $\nu = 0.4726$, respectively at the end of the sheath region. $\nu = 0.1145$ and $\nu = 0.4726$ represent electron number densities of $1.26\times10^{19}\,/\mathrm{m}^3$ and $5.21\times10^{19}\,/\mathrm{m}^3$, respectively.

Figure 5 represents electron number density, ν, as a function of position, ξ, inside the quasi-neutral region. The log of position is graphed for simplification. As expected, the dimensionless electron number density approaches one ($\nu_f \to 1$) for both wave speeds at the end of the quasi-neutral region. $\nu_f = 1.0$ represents an electron number density of $1.10\times10^{20}\,/\mathrm{m}^3$. Figure 6 represents electron temperature, θ

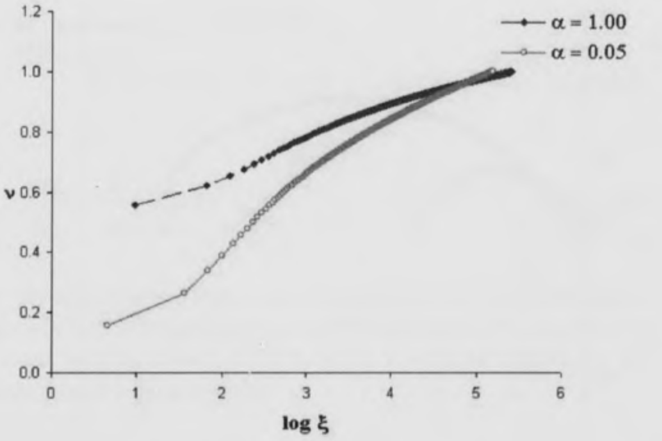

Fig. 5. Electron number density, ν, as a function of position, ξ, inside the QNR for α = 0.05 and α = 1.

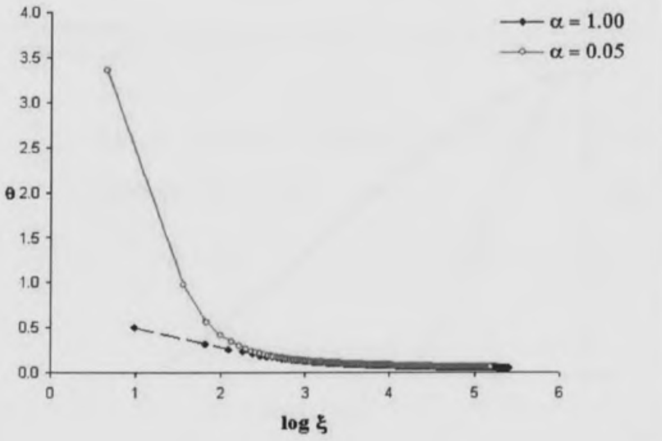

Fig. 6. Electron temperature, θ, as a function of position, ξ, inside the QNR for α = 0.05 and α = 1.

, as a function of position, ξ, inside the quasi-neutral region. Again, the log of the position is shown. For both wave speeds at the end of the quasi-neutral region, as expected, the temperature of the electron gas drops so that ionization is no longer possible ($\theta_f \rightarrow 0.065$). For α = 0.05 and α = 1, the final values for electron temperature are θ_f = 0.054 and θ_f = 0.052, respectively. θ_f = 0.054 represent an electron gas temperature of 3.13×10^4K.

Conclusions

In our research we successfully integrated the electron fluid dynamical equations for antiforce waves through the sheath and quasi-neutral region. The results for the wave speeds α = 0.05 and α = 1 conform to the expected conditions at the end of both the sheath and quasi-neutral region. Our selected wave speeds and calculated electron number densities and electron gas temperatures compare well with the observations of Uman et al. (1968), Rakov (2000), and Fujita et al. (2003). This is yet another confirmation of the validity of the fluid model used to describe breakdown waves.

ACKNOWLEDGMENTS.—The authors would like to express their gratitude to the Arkansas Space Grant Consortium for their continued financial support of this research.

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Discovery of a Dicephalic Western Diamondback Rattlesnake, *Crotalus atrox* (Serpentes: Viperidae), from Texas, with a Summary of Dicephalism Among Members of the Genus *Crotalus*

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Abstract.—We document the fourth record of a dicephalic western diamondback rattlesnake, *Crotalus atrox*, collected on 17 September 2005 in Tom Green County, Texas, along with features of its external and internal anatomy, which are compared with normal *C. atrox* neonates. This is only the second reported case of complete anatomical duplication of the respiratory system and nearly complete duplication of the circulatory and digestive systems in dicephalic snakes. In addition, we present a summary of dicephalism among 10 species within the genus *Crotalus*.

Key words:—dicephalism, western diamondback rattlesnake, *Crotalus atrox*, serpentes, viperidae, anatomy, Texas.

Introduction

Two-headed (or dicephalic) snakes are a curiosity of the herpetological world. They are rarely found in nature but frequently produced in captivity, probably as a result of inbreeding depression, temperature anomalies, and environmental toxins. Cunningham (1937) produced the first historical survey of dicephalic snakes and summarized data on 225 known specimens, including 134 figures on 12 plates. In the nearly 70 years since Cunningham's time, the number of verified specimens has risen to more than 500 (Smith and Pérez-Higareda 1987, Matz 1989, Payen 1991). These snakes occur in 8 families (Leptotyphlopidae, Pythonidae, Boidae, Tropidophiidae, Viperidae, Elapidae, Hydrophiidae, and Colubridae) and these 165 species are included within 92 genera (Wallach 2004). Herein, we report a noteworthy case of dicephalism in a western diamondback rattlesnake, *Crotalus atrox* Baird and Girard 1853, collected from the wild in western Texas, along with detailed information on its external and internal anatomy.

Materials and Methods

A dicephalic neonate male *C. atrox* (snout-vent length = 241 mm) was collected on 17 September 2005 by the senior author at the Head of the River Ranch in the vicinity of Christoval, Tom Green County, Texas (31° 08.1'N, 100° 29.2'W). It refused to eat on numerous occasions and following 2 weeks of captivity was euthanized with an overdose of sodium pentobarbital (Nembutal ®). The specimen was preserved in 10% formalin, transferred to 70% ethanol, and deposited in the Angelo State University Natural History Collection (ASNHC 14135). It was subsequently shipped to the junior author for a more detailed examination.

External Measurements.—Length of head was measured from tip of snout to posterior end of lower jaw, and head width was measured across the middle of the supraoculars. Scale counts were taken following previously described methods (Dowling 1951, Klauber 1956).

Radiographs.—Radiographs of the dicephalic specimen were taken while the snake was alive.

Internal Measurements and Anatomical Structure.—Visceral data are presented as percentage of snout-vent length (% SVL) with the organ midpoint (MP) often following the organ length. Paired organs are presented as left/right. The tracheal ring estimate is based on number of cartilages along a 5 mm segment at mid-tracheal position. The left systemic arch (LSA) and right systemic arch (RSA) are indicated herein. Since all of the blood vessels were not intact and injected with formalin, it was impossible to determine their exact identities and arrangement.

Internal viscera of the dicephalic specimen were compared with those of 2 normal male neonate *C. atrox* from Brownsville, Texas (Field Museum of Natural History, FMNH 27158 and University of Florida, UF 42587) and a specimen with no collection data exchanged to N. Ananjeva of St. Petersburg (Zoological Institute [ZISP], Akademia Nauk).

Results

External Anatomy.—The specimen is craniodichotomous (Figs. 1-2) with a tail length of 21 mm (excluding button, which is 7.7 mm long), total length of 262 mm, and a tail/total length ratio of 8.0%. Its midbody diameter is 16 mm and the scale row formula is 25-25-21, with 176 ventrals, 27 subcaudals (anterior 25 single,

Fig. 1. Dicephalic *C. atrox* from Tom Green County, Texas (in life). Photo by D. Elder.

terminal 2 paired). The left head is 18.1 mm long and 8.9 mm wide with 16/16 supralabials, 17/15 infralabials, 9/9 circumoculars, 5/4 interoculabials, and minimum intersupraoculars 6; the right head is 17.2 mm long and 8.8 mm wide with 17/15 supralabials, 16/16 infralabials, 9/9 circumoculars, 4/4 interoculabials; and minimum intersupraoculars 7. The ventrals number 158 to 163 paired with an umbilical scar between them; the umbilical scar-vent interval is 10.2% of the total ventrals. Externally the heads appeared united at the angle of the jaws (Fig. 2), but radiographs revealed short, symmetrical necks of approximately 6 vertebrae supporting each head.

Coloration.—The coloration (after preservation, Fig. 1) is typical of light specimens with a grayish-brown head dorsum, a pair of white lateral heads pre- and post-ocular oblique stripes, an immaculate white chin, 31 white-bordered gray dorsal diamonds on the body (1 at midbody paired and smaller than others), ground color lighter gray with a ventrolateral series of small blackish spots, venter white anteriorly, darkening to gray posteriorly, and 5 black crossbands on tail.

Internal Anatomy.—Duplication of the internal anatomy does not correlate well with the external bifurcation. All organs and glands except the most caudal viscera (gonads, adrenals, kidneys and large intestine) are duplicated and most, if not all, of the anterior blood vessels in ASNHC 14135 are duplicated.

The left hyoid is shorter than the right but its sternohyoideus muscle is longer, posterior tips of the hyoid are at 9.3%/10.0% and the sternohyoideus muscle is at 16.2%/15.8%. The thymus glands on the right side are cranial to those on the left: anterior thymus gland at 1.2% (MP = 38.8%)/0.8% (MP = 38.0%) and posterior thymus gland at 0.8% (MP = 39.8%)/0.4% (MP = 38.6%).

The left heart (3.3%, MP = 42.7%) is larger and more cranial than the right heart (2.5%, MP = 43.6%) and they are mirror images of one another with their right atria in contact along the midline and the left atria lateral (the left heart is normal in position, the right heart is reversed; the only instance of reversed symmetry of the viscera). Each heart has 2 systemic or aortic

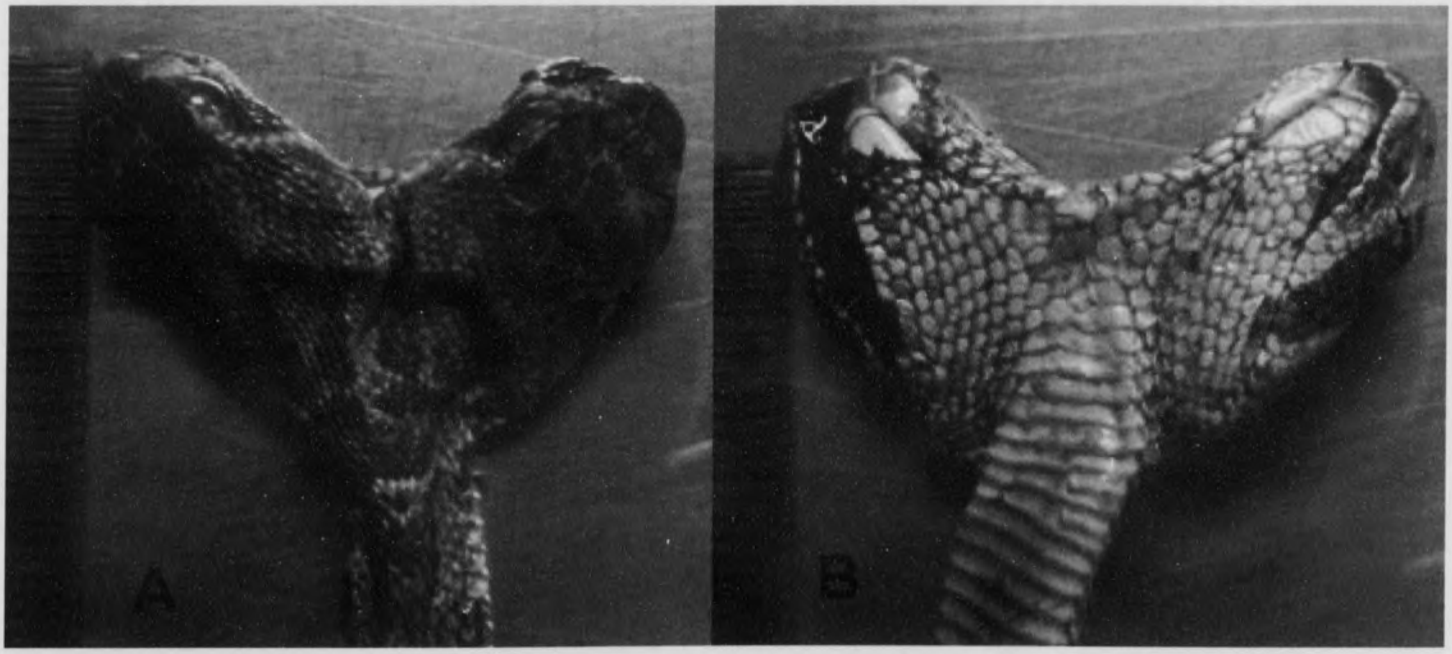

Fig. 2. Dicephalic *C. atrox* from Tom Green County, Texas, showing position of heads. A. Dorsal view. B. Ventral view. Scale bar (ruler) = 6 cm (Fig. 2A), 5 cm (Fig. 2B).

arches that form 3 junctions and an eventual single dorsal aorta, the RSA of the left heart joining the RSA of the right heart at 42.7%. The LSA of the right heart joins the combined RSA of the right and left hearts at 44.0%, and the LSA of the left heart joins the combined RSA of the right and left hearts at 47.3%. Every body segment receives a pair of intercostal arteries from the dorsal aorta in the posterior body cavities. The right systemic arch of each heart gives off left and right common carotids with a dorsal vertebral artery that bifurcates into anterior and posterior branches; the left carotid runs along the ventral surface of the esophagus and the right carotid extends along the ventral surface of the trachea. The pulmonary trunk of each heart gives rise to a large ventral anterior pulmonary (tracheal) artery and a smaller dorsal anterior (tracheal) artery; each has a weaker posterior branch that extends to the saccular lung. Each sinus venosus (and right atrium) receives a single posterior vena cava from the liver and a left and right anterior vena cava; the left vena cava extends along the ventral esophagus adjacent to the left carotid artery and the right vena cava runs along the ventral aspect of the trachea adjacent to the main branch of the pulmonary artery. Each left atrium receives a large anterior pulmonary (tracheal) vein and a smaller posterior pulmonary vein.

The respiratory system is lacking a left lung, bronchus, and orifice. The trachea, 26.1% (MP = 28.0%)/24.5% (MP = 30.1%), extends along the ventral surface of the tracheal lung in both cases. The estimated number of tracheal rings is 343/331 or 81.9/77.1 per 10% SVL with a tracheal-ring-width/tracheal-lung circumference at the mid-lung point of 3.0/3.5; tracheal rings have free tips. The right bronchus is 1.7% with a trachea-bronchus posterior tip at 44.0%/46.5%; the anterior lung tip is 14.9%/17.8% and tracheal lung 26.1% (MP = 28.0%)/24.5% (MP = 30.0%) with a maximum diameter of the tracheal lung at midpoint 4.0 mm/4.0 mm and maximum diameter just cranial of the heart at 4.0 mm/5.0 mm. The cardiac lung is 3.3%/2.5% with a saccular (right) lung which is much smaller on the left side of body, 6.2% (MP = 47.5%)/17.4% (MP = 53.5%); the maximum diameter of the saccular lung is 3.0 mm/10.0 mm and saccular lung is 8.3% (MP = 46.5%)/19.9% (MP = 52.3%); posterior lung tip is 50.6%/62.2% with a bronchus/right (saccular) lung at 8.3%. The left lung sack = 0.42 of the right lung sack, total lung/trachea-bronchus ratio 0.85/1.00.

The esophagus, 54.4% (MP = 29.3%)/55.0% (MP = 29.4%), lies on the left side of the tracheal lung in each case; it is lined internally with thin longitudinal ridges. The stomach, 13.3% (MP = 61.0%)/17.8% (MP = 59.1%), possesses distinctly heavier longitudinal and transverse rugae; the right stomach is larger and thicker-walled than the left; the maximum diameter of the stomach is 4.0 mm/6.0 mm with the pylorus at 67.6%/68.0%. The small intestine at 17.4% (MP = 76.3%)/17.0% (MP = 76.6%) and the paired anterior small intestine (duodenum) at 7.1%/6.6% before they fuse into single small intestine at 74.7%.

The heart-liver gap lies at 1.2%/0.6% with the liver at 18.3% (MP = 54.8%)/21.8% (MP = 56.3%). The 2 livers are mirror images although dissimilar in size and shape; the left liver is bipartite but artificially broken into 2 pieces (probably from the preservation process). The anterior section lies at 2.1% with a gap of 2.5% (probably artificial); the medial section is at 8.5% with another gap of 3.5% and a terminal sliver of 1.7%. The medial portion has a bifurcated fork; the medial fork is short (0.8% or 1 ventral); the lateral fork is the longer, forming a contiguous posterior "tail" (4.4% or 5 ventrals), but when including the disjunct posterior section, the "tail" measures 9.5% (or 7 ventrals). The left posterior vena cava is not visible ventrally; it extends along the dorsal surface of the left liver until it joins the right post cava at 54.4%. The right liver is also bipartite; the left lobe is unipartite, 4.1% (MP = 48.1%), but the right lobe is bipartite with a short (4.6%) posterior section separated by a gap of 0.4%. There is an anterior extension of the right lobe of the liver at 0.8%)1 ventral) and a posterior tail (right lobe) at 17.0% or 19 ventrals. The right posterior vena cava is visible ventrally, separating the left and right lobes and extending along the medial surface of the right lobe posteriorly. The left lobe = 0.19 of the right lobe on the right liver, 0.83 of the right liver on the left liver; the liver-gall bladder gap lies at 1.7%/0.8%; the gall bladders are large, 2.1% (MP = 66.6%)/2.5% (MP = 69.3%).

Both pancreas are of identical size, 1.0% (MP = 67.5%)/1.0% (MP = 67.7%) and are attached to their respective guts adjacent to the gall bladder; a single spleen is present on the right side at 0.8% (MP = 67.0%) with ducts entering the right pancreas and the left gut.

The posterior extent of duplication of the viscera occurs at 0.75 SVL, and caudad of this point the organs are normal and unduplicated. The left testis lies at 2.7% (MP = 85.0%); it is short and broad with numerous irregular creases (but is not segmented); the right testis was removed before measurements were taken. The left adrenal lies at 2.9% (MP = 82.8%), and the right adrenal is at 2.7% (MP = 79.1%). The kidneys are segmented; the left kidney lies at 8.7% (MP = 90.7%) and has 17 lobes; the right kidney lies at 11.6% (MP = 89.2%) and has 18 lobes. There is a single caudal portion of the small intestine at 10.4%; the ileocolic junction is at 85.1% and the rectal caecum is lacking. The large intestine and the cloaca lie at 14.9% (MP = 92.5%).

The thyroid gland(s) and left spleen are missing. The left liver was broken medially, and a number of blood vessels were broken, probably as a result of the original exploratory dissection just prior to preservation.

Discussion

The present specimen represents the 40th known dicephalic snake for which visceral anatomical data are known, previous reports include 1 boid, 26 colubrids, 1 elapid, 1 hydrophiid, and 10 viperids (Wallach 2004). There are 33 reported instances of dicephalism among 10 species of the genus *Crotalus* (Table 1). Of these reports, 4 cases of dicephalism are now known in *C. atrox* (Anonymous 1975, Engelmann and Obst 1981, Muir 1990, this report), including 1 wild and 3 captive cases. The rattlesnake for which the most cases of dicephalism (13 reports) have been reported is the timber rattlesnake, *Crotalus horridus* (see Table 1).

The late Jozsef Laszlo of the San Antonio Zoo mentioned a wild caught neonate craniodichotomous *C. atrox* from New Braunfels, Texas (Anonymous 1975) that survived for 7 weeks; it fed, digested, and defecated normally and was right head dominant (Murphy and Shadduck 1978). The disposition of this specimen is unknown.

A captive-born specimen of *C. atrox*, whose mother originated from the vicinity of Dallas, Texas, is shown in Engelmann and Obst (1981). It was stillborn on 10 February 1974 and is deposited in the Dresden Museum (MTKD D12509).

Three abnormal *C. atrox* offspring were produced in a litter of 11 albino neonates, 1 being a craniodichotomous dicephalic and 2 others eyeless with fused mouths (Muir 1990). The dicephalic individual survived 1 hr. Both parents were albinos and possibly littermates who originated from an unknown location, which would suggest the presence of the same recessive deleterious mutations in both parents. In addition, it would portend a tendency toward low genetic diversity and higher likelihood of negative recessive traits. Mating occurred on 22 March 1989 and the neonates were born 11 September 1989. The mother experienced abnormally cold temperatures in May, June, and August, another possible causative factor in the teratology. The 2 parents (mother ca. 1,200 mm, father ca. 1,500 mm) produced 12 offspring the previous year with 6 live and 6 stillborn, none of which were dicephalics (Muir 1990). The dicephalic specimen is preserved in the National Museum of Natural History (USNM 299806).

Lengths of known neonate *C. atrox* in Texas range from 229 to 330 mm (Werler and Dixon 2000) and from 214 to 367 mm throughout the species' entire range with a mean length of 285 mm (Ernst and Ernst 2003). Klauber (1956) earlier reported the average neonate length in *C. atrox* to be 350 mm with the smallest specimen he measured being 257 mm. Our specimen (ASNHC 14135), with a length of 262 mm, is well below the average size, a condition often documented among dicephalics and twin siblings. Lagerlund (1951) reported a dicephalic common adder (*Vipera berus*) to be 50 mm shorter than its siblings, and Petch (1990) found identical twins of twin-spotted ratsnakes (*Elaphe bimaculata*) measured only 111 mm while their normal siblings were 140 mm. In addition, Manimozhi et al. (2006) produced a dicephalic Burmese python (*Python molurus*) which measured 550 mm when its siblings averaged 611 mm.

Klauber (1956) listed the mean values for scale characters of *C. atrox* as follows (with corresponding values for ASNHC 14135 in parentheses): midbody scale rows 25 (25), ventrals 181.9 (176), subcaudals 25.7 (27), supralabials 15.5 (16.0), infralabials 16.8 (16.0), body blotches 35.4 (31), and relative tail length 7.9% (8.0%). Even though the present specimen was stunted in size, its scale counts are within normal limitations compared to the mean for the species.

Table 2 compares ASNHC 14135 with normal *C. atrox* neonates, and most of the traits are close to the values of normal individuals. Externally, it has a higher number of intersupraoculars. Internally, the major difference is in the shortened length of the saccular lung (with a more cranially positioned posterior tip). Also, the sternohyoideus muscle is longer and the right kidney is shorter than in the normal specimens.

The complete duplication of the respiratory system and nearly complete duplication of the circulatory and digestive systems is a rare event. Among 40 descriptions of the visceral anatomy in dicephalic snakes, only a single case of identical duplication of the viscera is known to the authors (Wallach, unpublished). A 162 mm craniodichotomous European asp (*Vipera aspis*) from Gugand, Vendée, France was reported to have 2 tracheae, 2 lungs, 2 hearts, 2 livers, 2 esophagi, 2 stomachs, and 2 small intestines that eventually fused embryologically into a single organ (Naulleau 1983, 1987). Other structures were not mentioned but it can be assumed that the gall bladder and pancreas also were duplicated.

In summary, we have provided, for the first time, a detailed description of the external and internal morphology of a dicephalic *C. atrox*. In addition, we have documented the fourth report of dicephalism in *C. atrox* and provided a summary of dicephalism among members of the genus *Crotalus*. When researchers find additional dicephalic snakes, attempts at collecting maximal data should be made along with documentation in the scientific literature.

Acknowledgments.—We thank the Boulware family for allowing CTM to collect on their properties, without which, this discovery would not have been possible. CTM also thanks Bob Dowler (ASNHC) for inviting him to take part in this fieldwork (mini-bioblitz) and Jim Murphy (National Zoological Park, Washington, DC), Hobart Smith (Univ. Colorado), and Stan Trauth (ASU) for providing technical information on dicephalics. Many thanks are also due to the curators who allowed dissection of specimens, H. K. Voris and A. Resetar (FMNH), D. L. Auth (UF), and N. Ananjeva (ZISP), and A. H. Wynn (USNM) for information on USNM 299806. Finally, much appreciation to D. Elder (Angelo State University Ram Page) for providing Fig. 1.

Table 1. Dicephalism in rattlesnakes of the genus *Crotalus*. Dashed lines represent unknown information.

Species	Country	State/Province	Locality	Date	Birth	Reference
C. adamanteus	USA	Florida	Gainesville	1967	captive	Murphy and Shadduck, 1978
			Odessa	1953	wild	Anonymous, 1954
C. atrox	USA	Texas	Dallas	1974	captive	Engelmann and Obst, 1981
			New Braunfels	1975	wild	Anonymous, 1975
		—	—	1989	captive	Muir, 1990
			Christoval	2005	wild	This report
C. basiliscus	Mexico	Colima	Colima	1928	captive	Wiley, 1930
C. durissus	Brazil	São Paulo	Araçatuba	1924	wild	Amaral, 1927
		Mato Grosso	—	—	wild	Vanzolini, 1947
		—	—	—	captive	Belluomini et al., 1974
		—	—	1891	wild	ER Brygoo *in* Matz, 1989
C. horridus	USA	Alabama	Bullock Co.	1978	captive	Lasher, 1980
			LaGrange	1995	wild	Sherer, 1995
		Arkansas	Paris	—	wild	Fred Lally, pers. comm.
		Iowa	Elkport	—	wild	C Nading *in* Cunningham, 1937
		Louisiana	LaPlace	—	captive	Wright, 1960
		Maryland	—	—	wild	Harris, 1968
		New York	New York	—	captive	RL Ditmars *in* Cunningham, 1937
		North Carolina	—	1869	wild	Anonymous, 1877b
		Oklahoma	Broken Bow	—	wild	Proctor, 1933
		Pennsylvania	Mount Sterling	—	wild	Anonymous, 1877a
			—	—	wild	Miller, 1938
		West Virginia	Logan	—	wild	Rimkus, 1947
		Wisconsin	Genoa	1915	wild	Anonymous, 1915
*C. lutosus**	USA	Idaho	Boise	—	wild	Todd, 1936
C. mitchellii	USA	Arizona	Camp Yuma	1855	wild	Baird, 1856
C. oreganus	USA	California	Fresno Co.	1926	wild	Bridges, 1926
C. scutulatus	USA	Arizona	Yuma	—	wild	Kelly, 1909
C. viridis	Canada	Alberta	Medicine Hat	1949	captive	MV Ratcliffe *in* Klauber, 1956
		Saskatchewan	Leader	2004	wild	Reid, 2005
	USA	Montana	Glendive	—	wild	McMullin, 1963
			—	1988	captive	Payen, 1991
		Texas	Pyote	1970	wild	Rogers, 1970

*Formerly *C. viridis lutosus* (see Douglas et al. 2002).

Table 2. Comparison of external and internal characteristics of ASNHC 14135 with normal neonates of *Crotalus atrox* from Texas. Organs and sides of head separated as left/right. Visceral data presented as % SVL, ALE = anterior liver extension in number of ventrals, ENTR = number of tracheal rings estimated for 10% SVL at midlung interval, JSA = junction of systemic arches in heart lengths posterior to heart, L = length, MP = midpoint, PLT = posterior liver tail in number of ventrals, PT = posterior tip, USVI = umbilical scar-vent interval as % total ventrals.

Character	ASNHC	FMNH	UF	ZISP
Sex	male	male	male	female
SVL	241 mm	320 mm	431 mm	350 mm
Tail/total length	8.0%	8.6%	7.9%	6.2%
Scale rows	25-25-21	27-27-21	27-25-21	27-25-21
Ventrals + subcaudals	176 + 27	175 + 29	177 + 30	180 + 21
Supralabials	16/16 / 17/15	17/16	17/15	16/15
Infralabials	17/15 / 16/16	17/16	16/18	16/16
Intersupraoculars	6/7	5	5	5
Circumoculars	9/9 / 9/9	8/8	8/9	8/9
USVI	10.2	9.7	9.0	8.3
Sternohyoideus PT	16.2 / 15.8	13.6	13.5	12.0
Trachea + bronchus	41.2 / 44.6	46.6	44.1	44.9
ENTR	81.9 / 77.1	99.2	94.7	88.8
TM/TR ratio	3.0 / 3.5	5.5	2.5	3.0
Bronchus/saccular lung	0 / 8.3	7.9	6.4	4.7
Tracheal + cardiac lungs	29.5 / 27.0	30.9	28.5	30.6
Saccular lung	6.2 / 17.4	23.8	29.0	30.6
Lung PT	50.6 / 62.2	70.3	69.6	70.6
Heart	3.3 / 2.5	3.8	2.8	3.7
Snout-heart interval	44.4 / 44.8	46.6	43.6	45.4
JSA	0.88 / – 0.33	0.85	0.75	0.56
Heart-liver gap	1.2 / 0.6	– 0.6	– 0.2	0
Liver	18.3 / 22.0	21.9	20.4	18.3
ALE	0 / 1	1	2	2
PLT	? / ?	14	13	12
Liver-gall bladder gap	1.7 / 0.8	2.2	1.2	2.9
Gall bladder MP	66.6 / 69.3	71.1	66.4	67.9
Right kidney (MP)	8.7 (89.2)	10.9 (88.9)	14.6 (88.7)	11.7 (89.9)
Left kidney (MP)	11.6 (90.7)	12.8 (90.2)	11.2 (89.8)	10.3 (90.9)

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Biomass Dynamics of *Tipula* (Insecta:Diptera) in Forested Streams of the Interior Highlands, Arkansas

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Abstract.—Abundance patterns of aquatic macroinvertebrates that utilize coarse particulate organic matter as a food source are important indicators of non-point source pollution associated with silviculture activities. This group, referred to as shredders, typically decreases in abundance as its food source—primarily leaves—is removed from the ecosystem. We tested whether larval biomass of the crane fly *Tipula*, a common member of the group, was an effective estimator of shredder abundance. Additionally, we used regression analysis to test whether the length to dry mass relationship of *Tipula* differed among geographic regions, between seasons, and between years. Results did not indicate significant differences in the relationship among these variables. Thus, we concluded that a general length to dry mass relationship was appropriate for *Tipula* in streams of the Interior Highlands. Our results were similar to those reported from North Carolina and Virginia. *Tipula* biomass was positively correlated with the total richness of the macroinvertebrate assemblage, a common measurement of stream quality, but was not correlated with the numerical abundance of the shredder functional feeding group. Thus, we concluded that *Tipula* biomass would not be an effective surrogate for whole assemblage metrics in biological assessments.

Key words:— aquatic macroinvertebrates, non-point source pollution, silviculture, *Tipula,* Interior Highlands, Arkansas.

Introduction

Silviculture is a major land use in Arkansas; over 55% of the state's land cover is commercial forest (AFC 2002). In Arkansas as in other states, biological assessments are performed to examine the impacts of non-point source (NPS) pollution, and one of the potential sources of NPS pollution is silviculture activity. Biological assessments can be effective in testing whether stream impairment is associated with NPS pollution (Rosenberg and Resh 1993). Standard biological assessment requires collecting samples representing the entire invertebrate community of streams in the watershed of interest and comparing characteristics of that community to an unimpaired reference condition. Whole community samples are recommended in order to make this sampling methodology adaptable enough to detect various types of environmental degradation (Barbour et al. 1999). However, these samples require significant investments of money and time to collect and analyze (Davidson and Clem 2002). An assessment procedure that reduces the time and cost spent per sample, but retains the ability to detect environmental impairment, is desirable. For assessments of potential silviculture impacts, focus upon the abundance and biomass dynamics of the shredder functional feeding group may be a solution.

Studies of the effects of forest clearing have frequently noted compositional shifts in aquatic invertebrate communities in streams of the cleared watershed, most often as a reduction in the abundance of the shredder functional feeding group, with corresponding increases in the collector and/or scraper groups (Newbold et al. 1980, Noel et al. 1986, Stone and Wallace 1998). The principal hypothesis of the cause of the shift is that removal of the forest canopy deprives the system of an important energy source—leaf inputs—and results in greater insolation of the stream bottom, stimulating algal growth (Campbell and Doeg 1989).

Important members of the shedder group are *Tipula* spp. (crane flies). This genus has more than 30 species in North America (Byers 1996), and as aquatic larvae, they primarily feed on decomposing plant material and associated microflora (Pritchard 1983). *Tipula* spp. are some of the major shredders, particularly in terms of biomass, in streams of the Interior Highlands (SBM pers. obs.). Most other regional representatives of this functional feeding group are either small taxa (e.g. capniid, leuctrid, and nemourid stoneflies and the chironomid genera *Cricotopus* and *Polypedilum*) or are larger taxa typically found in low numbers (e.g., limnephilid and phryganeid caddisflies; Merritt and Cummins 1996).

Tipula individuals go through four larval instars (Pritchard 1983). Mean length at the end of each instar has been reported for *Tipula sacra* from Alberta, Canada, as follows: first instar—5 mm, second instar—14 mm, third instar—20 mm; fourth instar females reached 50 mm and males reached 30 mm (Pritchard and Hall 1971). Pritchard (1976) reported adult *Tipula* emergence in Alberta through June and July with adults living less than a week. Eggs hatched in a few days, with second instar larvae appearing a few weeks later. They spent about 3 months as a second instar before molting to the third in November and overwintered as third instars. Most individuals molted to the fourth instar in April and May. He found first instar larvae in July through September, and they were the most abundant instar in July. Second instar larvae were present in all months except May and June and were most prominent in August and early September. Third instar larvae were present from August through the following June and were numerically dominant from mid-September to May. Fourth instar larvae were collected in all months and were numerically dominant in June. The typical *Tipula* life cycle is semivoltine with the second year spent as a fourth instar larva (Pritchard 1983). However, Pritchard (1980) hypothesized some cohort splitting may occur in the fall after hatching with most individuals overwintering as third instars, but some others grow rapidly to fourth instar and overwinter in that stage

Researchers addressing ecological questions at various scales recognize invertebrate biomass as an important variable (Benke et al. 1999). However, Rogers et al. (1976) noted that while insect biomass is an important piece of information, direct massing is too time consuming to be practical. Therefore, estimation of biomass is frequently performed using length to mass conversion ratios (Burgherr and Meyer 1997). A common method of performing this conversion is to use regression analysis, typically after log-transformation of raw data, and describe mass as a power function of length (Rogers et al. 1976, Sample et al. 1993, Hodar 1996). Length to mass power functions have been developed for aquatic dipterans (Burgherr and Meyer 1997), for other genera of the family Tipulidae (Meyer 1989), and for *Tipula abdominalis* (Smock 1980, Benke et al. 1999). However, Meyer (1989) questioned whether information from the equations was consistent across different geographical locations. It can be further questioned whether the information is consistent over time, i.e., between seasons and between years.

Tipula spp. are often the major holometabolous shredder in forested, low-order, Interior Highland streams, many of which are temporary streams (Poulton and Stewart 1991, Moulton and Stewart 1996). Holometabolous taxa have been observed to have higher growth ratios than hemimetabolous species (Cole 1980), which may be advantageous for exploiting ephemeral habitats like those prevalent in the Interior Highlands. That is, fast growth may be an adaptive advantage in streams that typically cease flowing for at least a few months every year.

Thus, the purpose of this study was to address the following questions:

1) Do *Tipula* length to dry mass relationships differ among geographically separate streams, between seasons, or between years? Do they differ among larval instars?

2) Do power equations developed for Interior Highland *Tipula* spp. conform to those derived from other sources?

3) Would *Tipula* biomass be useful as a biomonitoring tool for detecting decreased CPOM inputs associated with silviculture?

4) Does seasonal growth for *Tipula* occur faster in temporary streams than in permanent streams?

Methods

Tipula specimens were collected from 5 stream locations in the Ouachita and Ozark highlands in Arkansas from January 2003 through March 2004 using a 23-cm x 46-cm long-handled kick net. Characteristics of the study streams are summarized

Table 1. Characteristics of the 5 study streams. At Caney Creek, 2 sampling stations were on a first order segment of the stream and 1 was on a second order segment. At Thompson Creek, 2 stations were permanently flowing and 1 was on a segment that ceased flowing.

Stream	Ecoregion (subregion)	Order	Flow permanence	Drainage Area (km^2)	Gradient (m/km)
Bailey Creek	Ozark Highlands (Springfield Plateau)	2	Temporary	22.3	10.1
Big Creek	Arkansas Valley --	4	Permanent	89.5	4.3
Caney Creek	Ouachita Highlands (Fourche Mountains)	1/2	Temporary	6.8	16.7
Harris Creek	Ouachita Highlands (Central Ouachitas)	3	Permanent	24.2	12.2
Thompson Creek	Ozark Highlands (Boston Mountains)	2	Permanent, but spring-influenced	10.3	15.8

in Table 1. Sample substrates were predominantly cobble and gravel. At each location, we performed surveys at 3 separate stations, whose lengths of 100 to 200 meters were approximately 20x the average stream width. Collections were made in the early winter and early spring of each year. We collected 3 replicates at each site, for a total of 9 samples (3 replicates x 3 stations) at each stream during each survey period. Samples were preserved in the field with 5 % formaldehyde and transported to the Arkansas State University aquatic biomonitoring laboratory for analysis.

Tipula larvae were identified using Byers (1996) and separated from debris and other invertebrates and transferred to 80% ethanol within one month of collection. Length and mass measurements were taken approximately five months later. It is likely that *Tipula* specimens lost a portion of their dry mass due to leaching (e.g., Howmiller 1972, Landahl and Nagell 1978, Leuven et al. 1985). Even so, this method of preservation, as opposed to live collecting or freezing, is commonly used by entomologists (Rogers et al. 1976, Hodar 1996). Measurements of total length were made with Mecanic Type 6911 calipers, which are accurate to 0.1 mm. Specimens that were obviously contracted from their normal length were infrequently noted (<<1 % of the total individuals measured) and were included in the analysis. Dry mass (DM) was obtained by drying the animals at ~105°C for 16 to 24 hours then massing them on a Mettler-Toledo AB204-S balance accurate to 0.1 mg.

We transformed length and dry mass to $\log_{10}$ values and used linear regression to determine the y-intercept and slope of the line of best fit. We initially performed separate regressions on data from each stream site (n = 5) from each survey season (n = 2) in each year (n = 2). Stations and replicates were pooled within these data sets. We examined the average y-intercepts and slopes and their 90 % confidence intervals, using a Bonferroni adjustment to protect the family of estimates from error inflation. If overlap occurred between confidence intervals, we concluded that the y-intercept and slope coefficients did not differ significantly. Potential experimental outcomes ranged between 20 separate regression lines, if the length to dry mass relationship differed between seasons, years, and between each study stream and 1 line, if the relationship did not differ between season, year, or stream.

Table 2. Mean values (± 1 SE) of y-intercepts and slopes from length vs. mass regressions for samples from all study streams in 2 seasons of 2 years. Regressions used log10-transformed data for total length (mm) and dry mass (mg).

Stream	Year	Season	Y-intercept	Slope	n	r^2
Bailey Creek	2003	Winter	-2.58 ± 0.10	2.57 ± 0.08	172	0.87
		Spring	-2.16 ± 0.11	2.34 ± 0.08	131	0.88
	2004	Winter	-2.76 ± 0.42	2.68 ± 0.55	26	0.50
		Spring	-1.69 ± 0.27	2.04 ± 0.19	38	0.76
Big Creek	2003	Winter	-2.33 ± 0.16	2.45 ± 0.11	62	0.89
		Spring	-2.16 ± 0.32	2.35 ± 0.21	40	0.77
	2004	Winter	-2.43 ± 0.11	2.51 ± 0.08	93	0.91
		Spring	-1.72 ± 0.59	2.18 ± 0.36	31	0.56
Caney Creek	2003	Winter	-2.46 ± 0.38	2.55 ± 0.32	10	0.89
		Spring	-2.02 ± 0.26	2.28 ± 0.17	54	0.78
	2004	Winter	-2.51 ± 0.09	2.53 ± 0.08	342	0.74
		Spring	-1.54 ± 0.21	2.01 ± 0.14	69	0.75
Harris Creek	2003	Winter	-2.36 ± 0.23	2.42 ± 0.17	36	0.85
		Spring	-2.43 ± 0.34	2.51 ± 0.21	24	0.86
	2004	Winter	-2.79 ± 0.17	2.80 ± 0.13	69	0.88
		Spring	-1.93 ± 0.25	2.23 ± 0.16	46	0.81
Thompson Creek	2003	Winter	-2.20 ± 0.26	2.20 ± 0.23	28	0.78
		Spring	-1.56 ± 0.20	1.84 ± 0.15	27	0.86
	2004	Winter	-3.33 ± 0.20	3.23 ± 0.18	99	0.78
		Spring	-1.77 ± 0.22	2.08 ± 0.15	52	0.80

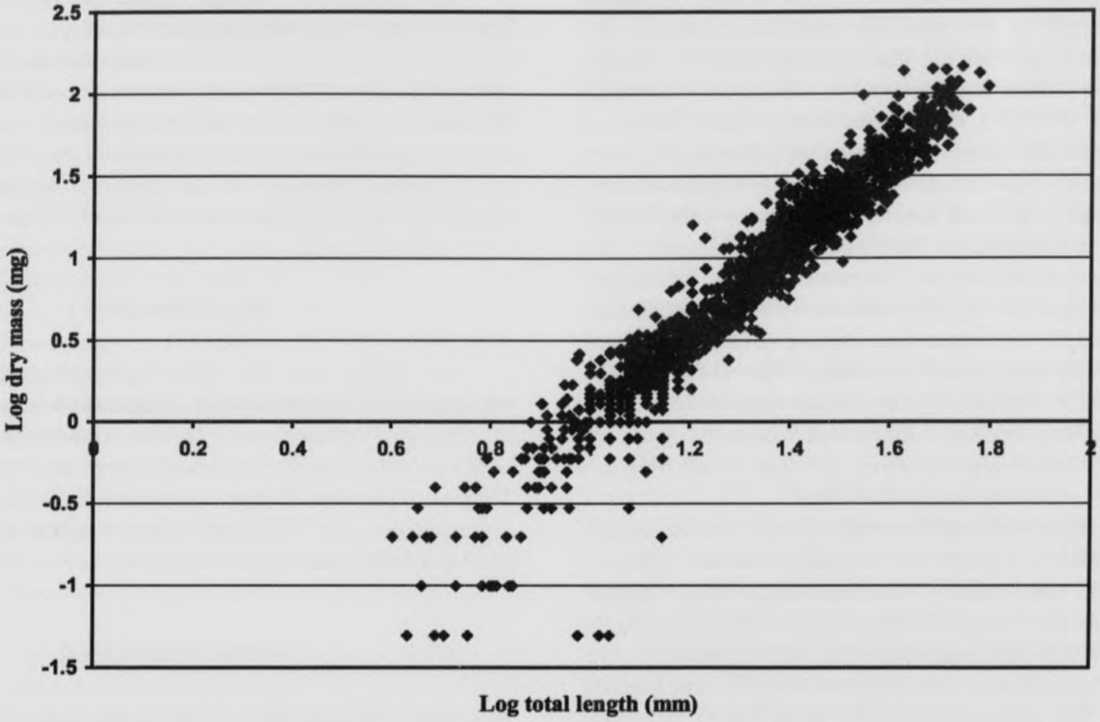

Fig. 1. Log dry mass vs. log length regression line for *Tipula* from 5 Interior Highlands study streams, winter and spring, 2003 – 2004.

Similarly, we used regression analysis to examine whether the length to dry mass relationship differed among the four larval instars. We separated data by total length, but pooled study streams, seasons, and years, using a modification of the instar length ranges reported by Pritchard and Hall (1971). We used 7.5 mm as the boundary between first and second instars and 25 mm as the boundary between third and fourth instars because these were the natural divisions indicated by a length vs. frequency histogram of our data.

If it was determined that annual, seasonal, and geographical differences were not significant, we pooled our data and compared our general length to dry mass relationship to those obtained for aquatic dipterans in other studies by converting the linear regression equation into a power function:

$$DM = aL^b$$

where DM is the dry mass of the organism (in mg), L is the total length (mm) and a and b are constants. The exponent b of the power model is the mean slope obtained in the linear regression, and a is the mean antilogarithm of the y-intercept.

We used correlation analysis to examine the relationship between *Tipula* biomass and two invertebrate community characteristics frequently used in biomonitoring studies—total taxa richness and the relative abundance of the shredder functional feeding group (% shredders). Total taxa richness is widely used as a metric in stream quality assessments (e.g., Barbour et al. 1999, OEPA 1987, ADEQ 2002). We chose % shredders as an additional community characteristic since it was expected to be the most sensitive to decreased CPOM inputs as well as being strongly associated with *Tipula* biomass.

We tested whether growth rates differed between *Tipula* collected from temporary streams and those from permanent streams using one-way analysis of variance. Data entered in the analysis were the average lengths for each sample (3 replicates x 3 stations x 2 years) for 4 of the 5 study streams. Bailey Creek and Caney Creek were considered temporary streams, whereas Big Creek and Harris Creek were considered permanent streams using information from 1:24,000 USGS topographic maps. Thompson Creek flowed throughout our study, but it was reported by an adjacent property owner to cease flow in particularly dry years. Since it was difficult to assign Thompson Creek to either treatment group, we excluded it from this analysis.

Results and Discussion

Of the 20 regressions (2 seasons x 2 years x 5 study streams) of *Tipula* total length vs. dry mass, the confidence intervals for the y-intercept and slope of the line overlapped for 17 (Table 2). Two of the 3 exceptions occurred in data from Thompson Creek; although they did not consistently indicate either a steeper or a shallower slope. Since so few regression lines differed significantly between seasons, years, or study streams, we concluded that a general length to dry mass relationship for *Tipula* spp. in the Interior Highlands is appropriate. The relationship for the pooled data is illustrated in Fig. 1. The power equation derived from the log dry mass to log length regression was $DM = 0.002\ L^{2.72}$, which was similar to those reported for *T. abdominalis* from North Carolina ($DM = 0.0015\ L^{2.81}$) and Virginia ($DM = 0.0054\ L^{2.46}$) by Benke et al. (1999). This indicated that the relationship is consistent within the genus, at least in the southeastern United States, and that *Tipula* biomass can be accurately estimated using total length.

Regression lines differed between *Tipula* instars (Table 3), with the shallowest slope for first instar larvae. Second and fourth instars had similar y-intercepts and slopes, whereas third instar larvae had a significantly steeper slope than any of the other instars. The high variability and low r^2 value for first instars likely reflected the low sample size (n = 37). The amount of variation in dry mass explained by total length increased progressively with larger instar larvae; each of the second, third, and fourth instar groups had n > 400.

Tipula biomass was positively correlated with the total richness of the macroinvertebrate assemblage ($r = 0.204$, $p = 0.006$) but was not correlated to the relative abundance of the shredder functional feeding group ($r = 0.021$, $p = 0.779$). The latter result suggests that the abundances of other shredder taxa obscure the presence of *Tipula* when the entire assemblage is considered. Also, the significant association with total richness likely reflects the large sample size (n = 180), as the correlation coefficient itself was low. Thus we could not conclude that *Tipula* biomass would be an effective surrogate for whole assemblage metrics in biological assessments. However, there was no impairment gradient in our samples; none indicated even moderate impairment from reference conditions. Therefore, it remains possible that *Tipula* biomass could be an effective indicator of reduced CPOM input into streams.

The increase in average length for *Tipula* did not differ between permanent and temporary streams ($F = 0.02$, $p = 0.886$). Mean length increases between winter and spring samples were nearly identical at 16.5 mm for permanent streams and 16.7 mm for temporary streams (n = 54 for each group). Perhaps this is not surprising, as the organisms in this study most likely represented closely related species. In any case, no increased growth rate adaptation in *Tipula* from intermittent streams was evident.

Conclusions

This study indicated that a general length to dry mass relationship is appropriate to estimate biomass for *Tipula* of the Interior Highlands, and that the confidence in the accuracy of the relationship increases with developmental stage. *Tipula* biomass was not strongly associated with whole community characteristics, and therefore does not appear to be an effective indicator of general macroinvertebrate community degradation.

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Table 3. Mean values (± 1 SE) of y-intercepts and slopes from length vs. mass regressions for the four instars of *Tipula* and for pooled data. Regressions used log10-transformed data for total length (mm) and dry mass (mg).

	Y-intercept	Slope	n	r^2
First instar (< 7.5 mm)	-2.08 ± 0.51	1.82 ± 0.66	37	0.17
Second instar (7.6 – 14 mm)	-2.45 ± 0.16	2.47 ± 0.14	413	0.41
Third instar (14.5 – 25 mm)	-3.00 ± 0.12	2.95 ± 0.09	454	0.69
Fourth instar (> 25 mm)	-2.36 ± 0.09	2.48 ± 0.06	545	0.75
All data	-2.72 ± 0.03	2.72 ± 0.02	1449	0.93

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Variational Symmetries and Conservation Laws in Linearized Gravity

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Abstract.—The methods of symmetry group analysis are applied to the action functional of linearized gravity to derive necessary conditions for the existence of variational symmetries. Two classes of variational symmetries of linearized gravity are discussed, and the local conservation laws associated with these variational symmetries are presented by applying Noether's theorem.

Key words.—Gravity, variational symmetries, linearized, conservation laws, Noether's theorem.

Introduction

Local conservation laws play a pivotal role in several branches of physics (Barrett and Grimes 1995, Goldberg 1958). The conserved quantities derived from conservation laws permit the characterization of a given physical system in terms of a relatively small number of physical quantities. For example, quantities such as energy, linear momentum, angular momentum, and charge, are often encountered in a wide range of classical and quantum systems because of the conservation laws associated with these quantities. In the quantum field theoretic description of the fundamental interactions, the existence of conservation laws is often the guiding principle that dictates the correct choice of the field theory that describes the fundamental interactions (Kaku 1993). In general relativity, it has often been argued that the existence of local conservation laws would lead to the construction of observables of the gravitational field, which could play a significant role in any quantum theory of gravity (Torre 1993). Conservation laws also play an important role in a variety of mathematical issues such as integrability, existence and uniqueness, and stability (Olver 1993).

In 1918, the German mathematician Emmy Noether proved two important theorems concerning the existence of conservation laws for physical systems that admit a Lagrangian formulation (Noether 1918). Her first theorem proved that if the Lagrangian admits a variational symmetry (a symmetry transformation that leaves the action functional invariant), then the system admits a local conservation law. The second theorem states that if a variational symmetry depends on arbitrary functions, then the differential equations governing the system must satisfy an identity.

In this paper, I discuss the variational symmetries and local conservation laws admitted by the linearized, vacuum Einstein equations of general relativity. I begin with a brief review of the Lagrangian formulation of linearized gravity and then proceed to apply methods of symmetry group analysis to derive necessary conditions for the existence of variational symmetries of linearized gravity. By applying Noether's theorems, the conservation laws associated with the variational symmetries are derived. I conclude with a discussion of two classes of variational symmetries admitted by linearized gravity — the Poincaré group of symmetries and the gauge symmetry of linearized gravity.

Methods

Linearized gravity—In Einstein's general theory of relativity, spacetime is assumed to be a 4-dimensional manifold M endowed with a Lorentzian metric g_{ab}. The spacetime metric g_{ab} satisfies the Einstein equations (in units where $c = 8\pi G = 1$),

$$G_{ab} = T_{ab}, \tag{1}$$

where G_{ab} is the Einstein tensor and T_{ab} is the energy-momentum tensor of the matter distributions (Wald 1984). In the absence of any matter distributions (for example, outside a star or planet), $T_{ab} = 0$ and the metric g_{ab} solves the vacuum Einstein equations of general relativity,

$$G_{ab} = 0. \tag{2}$$

Furthermore, if we restrict attention to regions of spacetime where the gravitational field is weak (for example, very far away from a star or planet), we can always choose local coordinates on spacetime such that the metric g_{ab} takes the form

$$g_{ab} = \eta_{ab} + h_{ab}, \tag{3}$$

where h_{ab} can be viewed as a symmetric $(0,2)$-type tensor field propagating on a flat spacetime endowed with the Minkowski metric η_{ab}. The tensor h_{ab} is assumed to be a small perturbation of the background Minkowski metric. This assumption allows us to restrict attention to terms linear in h_{ab}, which in turn implies

$$g^{ab} = \eta^{ab} - h^{ab}, \tag{4}$$

where $g^{ac} g_{cb} = \delta^{a}_{b}$ and $h^{ab} = \eta^{ac}\eta^{bd} h_{cd}$.

The linearized, vacuum Einstein equations are obtained by substituting equations (3) and (4) in equation (2) and expanding G_{ab} to linear order in h_{ab}. This yields

$$G_{ab} = \frac{1}{2}\left(\partial_c \partial_a h^{c}{}_{b} + \partial_c \partial_b h^{c}{}_{a} - \partial_a \partial_b h - \partial_c \partial^c h_{ab} - \eta_{ab} \partial_c \partial_d h^{cd} + \eta_{ab} \partial_c \partial^c h\right) = 0, \tag{5}$$

where $\partial_a := \frac{\partial}{\partial x^a}$ and $h := \eta_{cd} h^{cd} = h^{c}{}_{c}$ (Carroll 2004).

The linearized, vacuum Einstein equations do admit a Lagrangian formulation; they can be derived from a variational principle. The action functional $S[h^{ab}]$ of the variational problem is

$$S[h^{ab}]=\int_{\Omega}L(h,\partial h)\,d^4x, \tag{6}$$

where the Lagrangian density,

$$L(h,\partial h)=\frac{1}{2}\Big[(\partial_c h^{cd})(\partial_d h)-(\partial_c h^{de})(\partial_d h^c{}_e)+\frac{1}{2}\eta^{cd}(\partial_c h^{ef})(\partial_d h_{ef})-\frac{1}{2}\eta^{cd}(\partial_c h)(\partial_d h)\Big] \tag{7}$$

is defined on a compact region Ω of the spacetime manifold M with a smooth boundary $\partial\Omega$ (Carroll 2004). Let

$$h_{\varepsilon}^{ab}=h^{ab}+\varepsilon\delta h^{ab} \tag{8}$$

represent a one-parameter family of symmetric tensor fields on Ω. The tensor field δh^{ab} satisfies the boundary condition

$$\delta h^{ab}\,|_{\partial\Omega}=0. \tag{9}$$

In order to derive the linearized equations, we assumed the tensor field $h_0^{ab}=h^{ab}$ extremizes the action functional $S[h^{ab}]$. That is,

$$\delta S:=\frac{d}{d\varepsilon}\Big|_{\varepsilon=0}S[h_{\varepsilon}^{ab}]=0. \tag{10}$$

Equations (6) and (10) imply

$$\delta S=\int_{\Omega}\delta L\,d^4x=\int_{\Omega}\frac{d}{d\varepsilon}\Big|_{\varepsilon=0}L\,d^4x=0. \tag{11}$$

By virtue of equations and , equation can be written as

$$\int_{\Omega}G_{ab}\,\delta h^{ab}\,d^4x+\int_{\Omega}\partial_a J^a\,d^4x=0, \tag{12}$$

where G_{ab} is the linearized Einstein tensor given in equation (5), the vector field

$$J^a=\frac{1}{2}\Big[(\partial_b h)\delta h^{ab}+(\partial_b h^{ab})\delta h-2(\partial_c h^a{}_b)\delta h^{cb}+(\partial^a h_{bc})\delta h^{bc}-(\partial^a h)\delta h\Big] \tag{13}$$

and the trace of the variation δh^{ab} is $\delta h=\eta_{ab}\delta h^{ab}$ (Wald 1984). The divergence theorem implies

$$\int_{\Omega}\partial_a J^a\,d^4x=\int_{\partial\Omega}n^a J_a\,d^3x, \tag{14}$$

where n^a is the unit outward normal to the three-dimensional boundary $\partial\Omega$. Equations (9) and (13) imply

$$\int_{\partial\Omega}n^a J_a\,d^3x=0,$$

which forces the second integral in equation (12) to vanish, leaving

$$\int_{\Omega}G_{ab}\,\delta h^{ab}\,d^4x=0 \tag{15}$$

to hold for all symmetric tensor fields δh^{ab} satisfying the boundary condition. The fundamental lemma in the calculus of variation (Courant and Hilbert 1989) implies that equation (15) holds in Ω only if the linearized Einstein tensor G_{ab} vanishes in Ω, proving the result that a necessary condition for the tensor field h^{ab} to extremize the action functional $S[h^{ab}]$ is that it satisfy the linearized, vacuum Einstein equations.

Variational symmetries and conservation laws—The linearized, vacuum Einstein equations are a system of ten differential equations $G_{ab}=0$, involving 4 independent variables, or spacetime coordinates x^a, and ten dependent variables h_{ab} representing the components of a symmetric $(0,2)$-type tensor field. Let M represent the space of independent variables (or coordinates) x^a and U represent the space of dependent variables with coordinates h_{ab}. Consider a one-parameter family of infinitesimal transformations on the product space $M\times U$ given by

$$\widetilde{x}^a=x^a+\varepsilon\xi^a(x,h)+O(\varepsilon^2)\quad\text{and} \tag{16}$$

$$\widetilde{h}_{ab}=h_{ab}+\varepsilon\gamma_{ab}(x,h)+O(\varepsilon^2), \tag{17}$$

where $\varepsilon \ll 1$, and the functions $\xi^a(x,h)$ and $\gamma_{ab}(x,h)$ are the components of a smooth vector field and a smooth, symmetric, $(0,2)$-type tensor field, respectively, on $M\times U$. The infinitesimal transformations (16) and (17) transform the action functional given in equation (6) to

$$S[\widetilde{h}^{ab}]=\int_{\Omega}L(\widetilde{h},\widetilde{\partial}\widetilde{h})\,d^4\widetilde{x}, \tag{18}$$

where

$$\begin{aligned}L(\widetilde{h},\widetilde{\partial}\widetilde{h})=\frac{1}{2}(\widetilde{\partial}_c\widetilde{h}^{cd})(\widetilde{\partial}_d\widetilde{h})-\frac{1}{2}(\widetilde{\partial}_c\widetilde{h}^{de})(\widetilde{\partial}_d\widetilde{h}^c{}_e)+\frac{1}{4}\eta^{cd}(\widetilde{\partial}_c\widetilde{h}^{ef})(\widetilde{\partial}_d\widetilde{h}_{ef})\\-\frac{1}{4}\eta^{cd}(\widetilde{\partial}_c\widetilde{h})(\widetilde{\partial}_d\widetilde{h}),\end{aligned} \tag{19}$$

and $\widetilde{\partial}_a:=\frac{\partial}{\partial\widetilde{x}^a}$ represents partial derivatives with respect to the transformed coordinates $\widetilde{x}^a$. Expanding equations (18) and (19) in a Taylor series about $\varepsilon=0$ and reorganizing the resulting expression gives

$$S[\widetilde{h}^{ab}]=S[h^{ab}]+\varepsilon\int_{\Omega}\Big[\delta L+\partial_c\big(\xi^c L\big)\Big]d^4x+O(\varepsilon^2), \tag{20}$$

where

$$\begin{aligned}\delta L=\frac{1}{2}\Big[(\partial_c h^{cd})(\partial_d Q)+(\partial_c Q^{cd})(\partial_d h)-(\partial_c Q^{de})(\partial_d h^c{}_e)-(\partial_c h^{de})(\partial_d Q^c{}_e)\\+\eta^{cd}(\partial_c Q^{ef})(\partial_d h_{ef})-\eta^{cd}(\partial_c Q)(\partial_d h)\Big]\end{aligned} \tag{21}$$

and the characteristic Q_{ab} is defined as

$$Q_{ab}=\gamma_{ab}-\xi^c\partial_c h_{ab} \tag{22}$$

with $Q^{ab} = \eta^{ac}\eta^{bc}Q_{cd}$ and $Q = \eta^{ab}Q_{ab}$. In deriving equation (20) we have also used the fact that, to first order in ε, the volume element $d^4\widetilde{x}$ transforms as

$$d^4\widetilde{x} = \left(1 + \varepsilon\, \partial_c \xi^c\right) d^4 x + O(\varepsilon^2).$$

The infinitesimal transformations (16) and (17) represent a variational symmetry of the action functional if they leave the action functional invariant up to an overall surface term for all symmetric tensor fields h_{ab} on M (Olver 1993). In other words,

$$S[\widetilde{h}^{ab}] = S[h^{ab}] + \varepsilon \int_{\partial\Omega} n_a \Lambda^a d^3 x + O(\varepsilon^2), \quad (23)$$

where Λ^a are the components of a vector field on M and n_a is a one-form field normal to the boundary $\partial\Omega$. The divergence theorem applied to the surface term in Equation (23) implies that if the fields ξ^a and γ_{ab} represent variational symmetries of linearized gravity then they must satisfy the condition

$$\delta L + \partial_c \left(\xi^c L\right) = \partial_c \Lambda^c. \quad (24)$$

In equation if the vector field $\Lambda^a = 0$, the variational symmetry is called a strict variational symmetry; otherwise the symmetry is referred to as a divergence symmetry (Olver 1993).

A local conservation law of the linearized, vacuum Einstein equations is a vector field P^a built from the coordinates x^b, the tensor field h_{ab}, and derivatives of the tensor field h_{ab} to any arbitrary but finite order that satisfies the condition

$$\partial_a P^a = 0$$

on solutions of the field equations (5) of linearized gravity. In order to elicit the relationship between variational symmetries and local conservation laws established in Noether's theorems, we rewrite equation (24) by integrating the term δL by parts. This yields, after some algebra,

$$\delta L = Q^{ab} G_{ab} + \partial_a S^a, \quad (25)$$

where G_{ab} is the linearized Einstein tensor defined in equation (5) and

$$S^a = \frac{1}{2}\left[(\partial_b h)Q^{ab} + (\partial_b h^{ab})Q - (\partial_c h^a{}_b)Q^{cb} - (\partial_b h^a{}_c)Q^{cb} + (\partial^a h_{bc})Q^{bc} - (\partial^a h)Q\right] \quad (26)$$

Substituting equation in equation and reorganizing the terms gives

$$\partial_a P^a = -Q^{ab} G_{ab}, \quad (27)$$

where

$$P^a = \Lambda^a - S^a - \xi^a L. \quad (28)$$

It is clear that on solutions of equation (5), if Q_{ab} represents the characteristic of a variational symmetry, then P^a represents a conserved vector field of linearized gravity.

We now explore two distinct types of variational symmetries that are significant in linearized gravity - the Poincaré symmetries and gauge symmetries. Assume the vector field ξ^a depends only on the coordinates on the manifold M (i.e., $\xi^a = \xi^a(x)$). Consequently, the infinitesimal transformations (16) represents a one-parameter family of coordinate transformations on M. Since h_{ab} is a tensor field on M, it must transform according to the tensor transformation law, namely,

$$\widetilde{h}_{ab} = \widetilde{\partial}_a x^c \widetilde{\partial}_b x^d h_{cd} = h_{ab} - \varepsilon\left(\partial_a \xi^c h_{cb} + \partial_b \xi^c h_{ac}\right) + O(\varepsilon^2). \quad (29)$$

Equations (17) and (29) imply

$$\gamma_{ab} = -\left(\partial_a \xi^c h_{cb} + \partial_b \xi^c h_{ac}\right)$$

and the characteristic Q_{ab} takes the form

$$Q_{ab} = -L_\xi h_{ab}, \quad (30)$$

where $L_\xi h_{ab}$ is the Lie derivative of the tensor field h_{ab} with respect to the vector field ξ^a (Wald 1984). Furthermore, let us assume that the infinitesimal coordinate transformations generated by the vector field ξ^a leave the Minkowski metric η_{ab} invariant. In other words, ξ^a is a Killing vector field of the Minkowski metric and hence satisfies the Killing equation

$$L_\xi \eta_{ab} = \partial_a \xi_b + \partial_b \xi_a = 0. \quad (31)$$

Substituting equations (30) and (31) in equation (21) and simplifying the resulting equation using the properties of the Lie derivative gives

$$\delta L = -\xi^c \partial_c L + F^{ab}{}_c \partial_a \partial_b \xi^c, \quad (32)$$

where

$$\begin{aligned} F^{ab}{}_c = \frac{1}{2}\Big\{ & \delta^a_c h^{bd} \partial_d h + \delta^d_c h^{ab} \partial_d h - h^{bd} \partial_c h^a{}_d - h^{bd} \partial_d h^a{}_c \\ & - h^a{}_d \partial_c h^{bd} + h^d{}_c \partial_d h^{ab} + 2\eta^{ad} h^{be} \partial_d h_{ce} \Big\} \end{aligned}$$

Substituting equation (32) back into equation (24) yields

$$-\xi^c \partial_c L + F^{ab}{}_c(h, \partial h) \partial_a \partial_b \xi^c + \partial_c \left(\xi^c L\right) = \partial_c \Lambda^c.$$

Since ξ^a is a Killing vector field, it follows that $\partial_c \xi^c = 0$ and $\partial_a \partial_b \xi^c = 0$ (Crampin and Pirani 1994). Setting $\Lambda^a = 0$ proves that all coordinate transformations on the spacetime manifold M that leaves the Minkowski metric invariant are strict variational symmetries of the action functional of linearized gravity. These coordinate transformations are the ten-parameter group of Poincaré symmetries.

To determine the conserved vector field associated with the Poincaré symmetries, we substitute equation (30) in equation (28) and set $\Lambda^a = 0$ to obtain

$$P^a = \frac{1}{2}(\partial_b h)L_\xi h^{ab} + \frac{1}{2}(\partial_b h^{ab})L_\xi h - (\partial_c h^a{}_b)L_\xi h^{cb} + \frac{1}{2}(\partial^a h_{bc})L_\xi h^{bc} - \frac{1}{2}(\partial^a h)L_\xi h$$
$$+\frac{1}{2}\xi^a\left[(\partial_c h^{cd})(\partial_d h) - (\partial_c h^{de})(\partial_d h^c{}_e) + \frac{1}{2}\eta^{cd}(\partial_c h^{ef})(\partial_d h_{ef}) - \frac{1}{2}\eta^{cd}(\partial_c h)(\partial_d h)\right].$$

The other variational symmetry is the infinitesimal gauge transformation obtained by setting $\xi^a = 0$ and $\gamma_{ab} = \partial_a X_b + \partial_b X_a$ in equations (16) and (17), where X^a is an arbitrary vector field on the manifold M. Substituting the characteristic $Q_{ab} = \partial_a X_b + \partial_b X_a$ in equation (21) and recursively applying the integration by parts formula to the resulting equation gives

$$\delta L = \partial_a \Lambda^a - 2\partial_a G^{ab} X_b, \qquad (33)$$

where

$$\Lambda^a = S^a + 2G^a{}_b X^b, \qquad (34)$$

and S^a is the vector field defined in equation (26) with $Q_{ab} = \partial_a X_b + \partial_b X_a$. The linearized, vacuum Einstein equations satisfy the contracted Bianchi identity

$$\partial_a G^a{}_b = 0, \qquad (35)$$

which reduces equation (33) to a form that clearly indicates that the gauge transformation is not a strict variational symmetry of the linearized theory, but instead a divergence symmetry. Equations (28) and (34) imply that the conserved vector field is

$$P^a = 2G^a{}_b X^b.$$

However, note that the conserved vector field $P^a = 0$ on solutions of equation (5) and hence defines a trivial conservation law. This is because the gauge symmetry depends on an arbitrary vector field X^a and hence falls under the purview of Noether's second theorem, which states that the linearized Einstein equations must satisfy a constraint equation. This constraint equation is the contracted Bianchi identity given in equation (35).

Conclusions

I derived necessary conditions that must be satisfied by a variational symmetry of the linearized, vacuum Einstein equations and investigated two classes of variational symmetries: the ten-parameter group of Poincaré symmetries and the gauge symmetry. I showed that the Poincaré symmetries admit a ten-parameter family of local conservation laws, while the gauge symmetry is a divergence symmetry admitting a trivial conservation law. Looking ahead, it would be interesting to investigate the various conservation laws associated with the Poincaré symmetries and explore their geometric and physical significance. Another interesting research direction is the classification of all local conservation laws of the linearized, vacuum Einstein equations. This is achieved by investigating solutions of equation , where the characteristic Q_{ab} depends on derivatives of the the tensor field h_{ab} to any arbitrary but finite order.

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Leeches (Annelida: Hirudinida) of Northern Arkansas

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Abstract.—Twenty-one lotic and lentic environments throughout central and northern Arkansas were surveyed for the presence of leeches during June 2004 and April, July – October 2005. Fourteen species of leeches (*Helobdella elongata, Helobdella papillata, Helobdella stagnalis, Placobdella cryptobranchii, Placobdella multilineata, Placobdella ornata, Placobdella papillifera, Placobdella parasitica, Placobdella phalera, Placobdella picta, Haemopis marmorata, Erpobdella fervida, Erpobdella microstoma*, and *Erpobdella punctata*) representing 3 families were collected. Five species (*H. elongata, P. cryptobranchii, P. multilineata, H. marmorata*, and *E. fervida*) are reported from Arkansas for the first time. The natural history of the 22 species of leeches now known from Arkansas is reviewed.

Key words.—Leeches, *Helobdella elongata, Helobdella papillata, Helobdella stagnalis, Placobdella cryptobranchii, Placobdella multilineata, Placobdella ornata, Placobdella papillifera, Placobdella parasitica, Placobdella phalera, Placobdella picta, Haemopis marmorata, Erpobdella fervida, Erpobdella microstoma, Erpobdella punctata,* Arkansas, lotic, lentic.

Introduction

There are approximately 80 species of leeches known from North America; however, limited data are available on their distribution (Klemm 1985, Sawyer 1986). The leech fauna of the southern United States is particularly understudied with only a few studies in the region (Meyer 1937, Sawyer 1967, Sawyer and Shelley 1976). Although several new leech species have been described from the southern United States, many have not been found since their original description (Sawyer and Shelley 1976, Johnson and Klemm 1977, Klemm 1985). Little is known about the leech fauna of Arkansas. Published accounts have described parasitism and the life histories of a few leech species or have documented the fauna in a very limited geographic area (Becker et al. 1966, Curry 1976, Harp and Harp 1980, Cochran and Harp 1990, Chordas et al. 1996, Briggler et al. 2001, Turbeville and Briggler 2003). There has not been a holistic survey of the state.

This study presents new survey data along with a summary of information gleaned from museum specimens and published records to serve as a baseline for further comprehensive study of the leech biodiversity of Arkansas.

Materials and Methods

Twenty-one lotic and lentic localities in central and northern Arkansas were surveyed for leeches during June 2004 and April, July – October 2005. Leeches were hand-collected from beneath submerged rocks, branches, logs, and vegetation, and by dip-net. The latitude and longitude of the collecting sites were recorded with a Garmin Vista receiver (datum: WGS 84), and published specimen records and unpublished museum specimens were geocoded with Topozone, Delorme Street Atlas, and the US Geological Survey Geographic Names Information System (GNIS) gazetteer. Data points were plotted using ArcView 3.3 (ESRI Corporation, Redlands, California).

Leech specimens were relaxed in 5-10% ethanol (added dropwise in a vessel until the leech no longer reacted to a probe), fixed in 10% buffered formalin, preserved in 70% ethanol, and examined under a dissecting microscope. Additional leech specimens were preserved in 95% ethanol. Voucher specimens of leeches were deposited in the Invertebrate Zoology collections of the National Museum of Natural History, Smithsonian Institution (accession no. 2041351).

Table 1. Checklist of the Leech Fauna of Arkansas

- Superorder Euhirudinea
  - Order Rhynchobdellida
    - Family Glossiphoniidae
      - *Helobdella elongata* Castle 1900 *†
      - *Helobdella papillata* Moore 1952 †‡
      - *Helobdella stagnalis* (Linnaeus 1758) †‡
      - *Placobdella cryptobranchii* (Johnson and Klemm 1977) *†
      - *Placobdella montifera* Moore 1906 ‡
      - *Placobdella multilineata* Moore 1953 *†
      - *Placobdella ornata* (Verrill 1872) †‡
      - *Placobdella papillifera* (Verrill 1872) †‡
      - *Placobdella parasitica* (Say 1824) †‡
      - *Placobdella phalera* (Graf 1899) †‡
      - *Placobdella picta* (Verrill 1872) †‡
    - Family Piscicolide
      - *Cystobranchus verrilli* Meyer 1940 ‡
      - *Gonimosobdella klemmi* Williams and Burreson 2005 ‡
      - *Myzobdella lugubris* Leidy 1851 ‡
      - *Piscicolaria reducta* Meyer 1940 ‡

- Order Arhynchobdellida
  - Family Haemopidae
    - *Haemopis terrestris* (Forbes 1890) ‡
    - *Haemopis marmorata* (Say 1824) *†
  - Family Hirudinidae
    - *Macrobdella diplotertia* Meyer 1975 ‡
    - *Macrobdella ditetra* Moore 1953 ‡
  - Family Erpobdellidae
    - *Erpobdella fervida* (Verrill 1871)*†
    - *Erpobdella microstoma* (Moore 1901) †‡
    - *Erpobdella punctata* (Leidy 1870) †‡

* - Arkansas State Record
† - This study
‡ - Published records

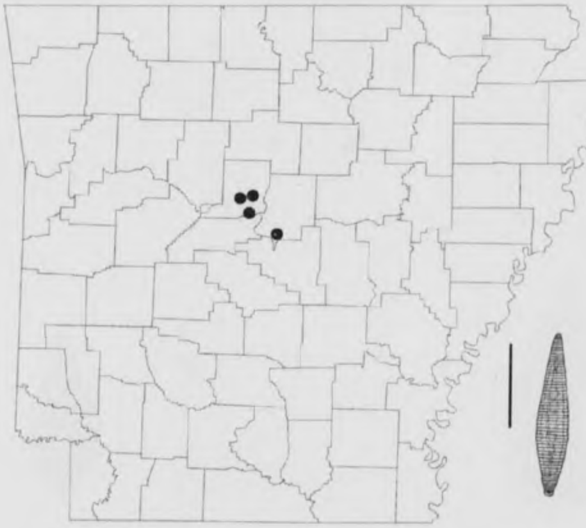


Fig. 1. Distribution map and line drawing of *Helobdella elongata* from Arkansas (circles – current study; scale bar is 0.5 cm).

Results and Discussion

Fourteen species of leeches representing 3 families were found, of which 5 are reported from Arkansas for the first time (Table 1).

Order Hirudinida
Suborder Rhynchobdellida
Family Glossiphoniidae

Helobdella elongata
Castle, 1900* (Fig. 1)

Locality: Conway County, Brewer Lake (35 13.661 N 92 36.661 W), 1 spm; Conway County, Overcup Lake (35 12.613 N 92 42.712 W), 1 spm; Faulkner County, Lake Conway, Mayflower, off Route 40 (35 58.2466 N 92 25.314 W), 3 spms; Perry County, Plummerville Bottoms, Arkansas River (35 06.663 N 92 38.350 W), 1 spm.

In a recent phylogenetic analysis of the genus *Helobdella*, Siddall and Borda (2003) transferred *H. elongata* from the genus *Gloiobdella. H. elongata* is infrequently found, and has a scattered distribution throughout North America, including Louisiana, Mississippi, Missouri and Texas (Klemm 1985). This study is the first report of this species from Arkansas.

Helobdella elongata is a predaceous leech, feeding on the soft body tissues/fluids of insect larvae, aquatic oligochaetes and snails (Sawyer 1974, Klemm 1985, 1991). This leech is small, elusive, and frequently overlooked in studies. *H. elongata* is often associated with organically polluted water (Sawyer 1974, Klemm 1991), which is probably a response to an abundance of potential prey.

Helobdella papillata
Moore, 1952 (Fig. 2)

Locality: Van Buren County, South Fork of Little Red River, South of Clinton (35 35.0218 N 92 26.5145 W), 1 spm; Van Buren County, McIntire Pond no.2 (35 36.585 N 92 28.824 W), 3 spms.

In previous keys, specimens collected in this study would fall under the description of *Helobdella triserialis* (Klemm 1982, 1985, Sawyer 1986). Based upon the original species descriptions and the results of a phylogenetic analysis, Siddall and Borda (2003) differentiated *H. papillata* (three irregular rows of papillae irrespective of size and pigmentation) and *H. triserialis* (cephalic transverse banding). *Helobdella papillata*, as it is now defined, is common and widely distributed throughout eastern North America. Additional records of *H. papillata* from

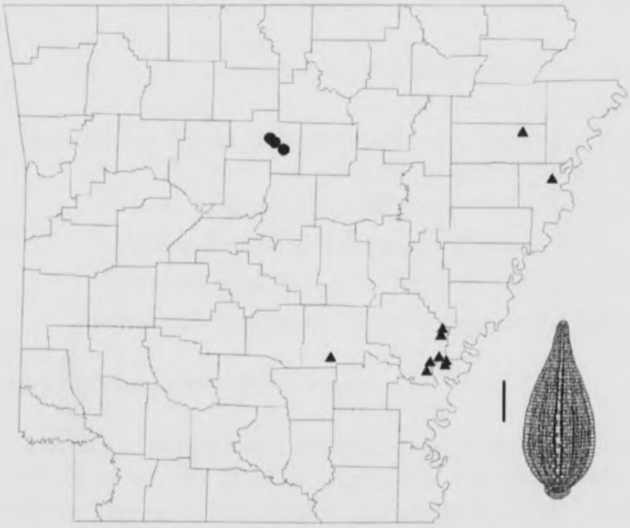


Fig. 2. Distribution map and line drawing of *Helobdella papillata* from Arkansas (circles – current study; triangles – published records; scale bar is 0.5 cm).

Arkansas have been reported by Curry (1976) from Bayou Bartholomew, Harp and Harp (1980) from Wapanocca National Wildlife Refuge, Cochran and Harp (1990) from St. Francis Sunken Lands, and Chordas et al. (1996) from White River National Wildlife Refuge.

Helobdella papillata is predaceous, feeding on the soft body tissues of mollusks, especially gastropods (Sawyer 1974, 1986, Klemm 1985, 1995). It is typically found beneath submerged substrata or within snail shells.

Helobdella stagnalis
(Linnaeus 1758) (Fig. 3)

Locality: Conway County, Overcup Lake (35 12.613 N 9 42.712 W), 1 spm; Faulkner County, Lake Conway, Mayflower, off Route 40 (35 58.2466 N 92 25.314 W), 10 spms; Randolph County, Eleven Point River, 5 spms.

Helobdella stagnalis has a cosmopolitan distribution and is known from every continent, except for Australia and Antarctica. It is prevalent in northern North America and less abundant in the southern United States. Additional records of *H. stagnalis* from Arkansas have been reported by Curry (1976) from Bayou Bartholomew, Harp and Harp (1980) from Wapanocca National Wildlife Refuge, Cochran and Harp (1990) from St. Francis Sunken Lands, and Chordas et al. (1996) from White River National Wildlife Refuge. It has also been reported from Louisiana, Mississippi, Missouri, and Texas (Klemm 1985).

Helobdella stagnalis is predaceous, feeding on the soft body tissues/fluids of a variety of prey, including oligochaetes, aquatic insect larvae, crustaceans, and small mollusks (Sawyer 1972, 1974, 1986, Klemm 1985, 1991, 1995). *H. stagnalis* is more abundant in organically polluted waters (Sawyer 1974, 1986) and is commonly found beneath submerged substrata and between the leaves of reeds and cattails.

Placobdella cryptobranchii
(Johnson and Klemm 1977)* (Fig. 4)

Locality: Fulton County, Spring River, 193 spms; Randolph County, Eleven Point River, 62 spms.

Although not included in the phylogenetic analysis and

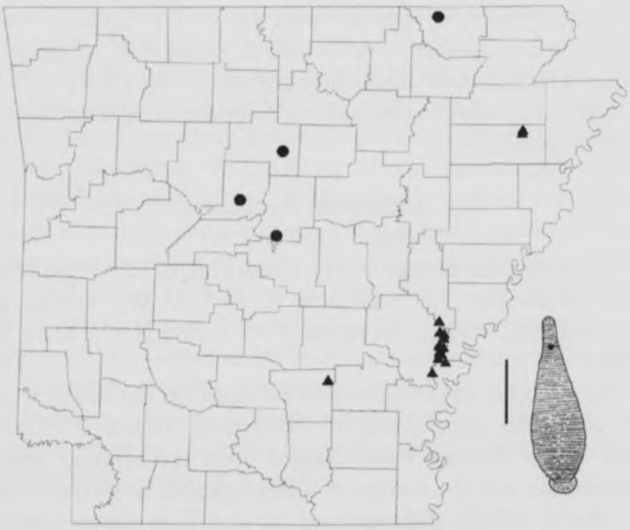


Fig. 3. Distribution map and line drawing of *Helobdella stagnalis* from Arkansas (circles – current study; triangles – published records; scale bar is 0.5 cm).

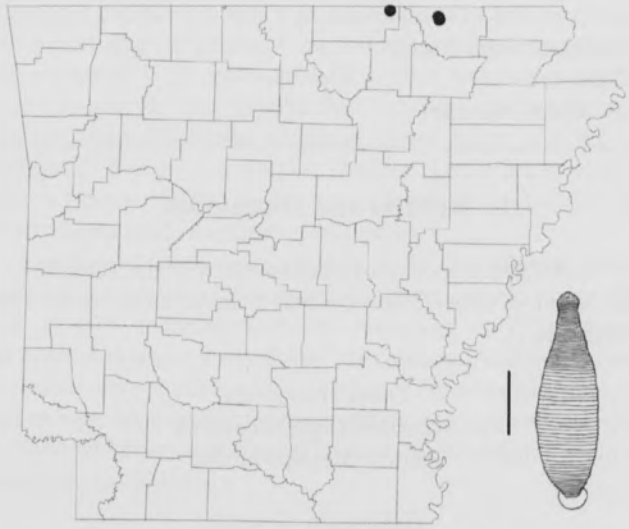


Fig. 4. Distribution map and line drawing of *Placobdella cryptobranchii* from Arkansas (circles – current study; scale bar is 0.5 cm).

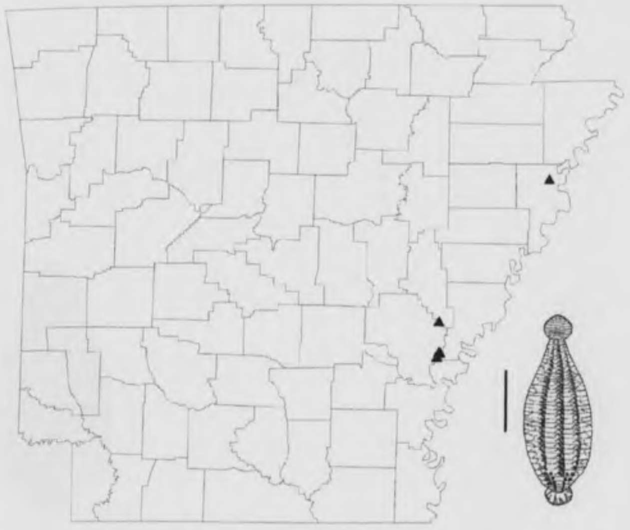


Fig. 5. Distribution map and line drawing of *Placobdella montifera* from Arkansas (triangles – published records; scale bar is 0.5 cm).

subsequent suppression of the genus *Desserobdella* by Siddall et al. (2005), *Placobdella cryptobranchii* represents a new combination due to its morphological similarity to the other members of the former genus *Desserobdella*. *P. cryptobranchii* was previously only known from its type locality in the North Fork of the White River (Ozark County, Missouri) and had not been collected since its description. This study is the first report of *P. cryptobranchii* from Arkansas. It is also known from several localities in Missouri (Moser et al. unpublished data). The only known host of *P. cryptobranchii* is the Ozark Hellbender, *Cryptobranchus alleganiensis bishopi*, and it has never been collected detached from its host (Johnson and Klemm 1977, Moser et al. unpublished data).

Placobdella montifera
Moore, 1906 (Fig. 5)

Placobdella montifera has been reported throughout Canada and the eastern and midwestern United States, including Louisiana, Missouri, and Texas (Klemm 1985). It was reported from Arkansas by Harp and Harp (1980) in the Wapanocca National Wildlife Refuge and by Chordas et al. (1996) in the White River National Wildlife Refuge. *P. montifera* is an opportunistic blood-feeding leech with a variety of fish species being the most commonly described hosts, including members of the families Lepisosteidae, Acipenseridae, Percidae, Centrarchidae, Ictaluridae, Catostomidae, Cyprinidae, Salmonidae, and Esocidae (Hoffmann 1967, Sutherland and Holloway 1979, Amin 1981, Sawyer 1986). It also has been regularly found in the mantle cavity of freshwater clams, but it is unknown if the leech feeds on the clam or if the clam is clandestine shelter (Curry and Vidrine 1976, 1977, Curry 1977, 1979).

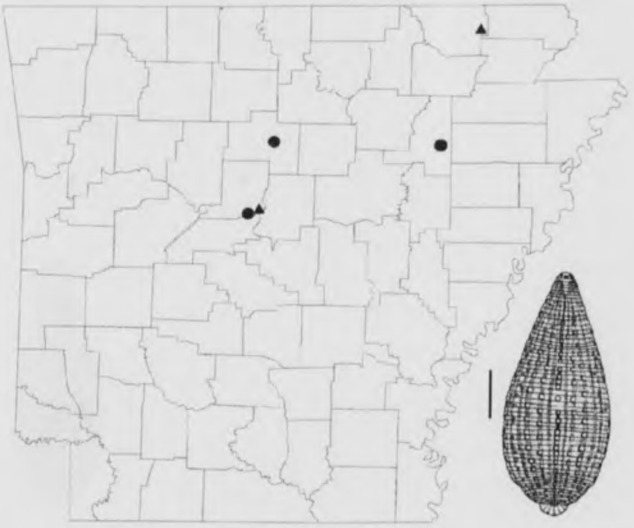


Fig. 6. Distribution map and line drawing of *Placobdella multilineata* from Arkansas (circles – current study; triangles – museum specimens; scale bar is 1.0 cm).

Placobdella multilineata
Moore, 1953* (Fig. 6)

Locality: Jackson County, Roadside ditch off US 14, 1.4 km W of Amagon (35 33.780 N 91 07.664 W), 1 spm; Jackson County, Roadside ditch off US 14, southside of road, 0.3 km W of Amagon (35 33.716 N 91 06.936 W), 2 spms; Perry County, Plummerville Bottoms, Arkansas River (35 06.663 N 92 38.350 W), 2 spms; Van Buren County, Honey Hill Pond (35 35.5407 N 92 26.1546 W), 1 spm.

Placobdella multilineata is a common species in the southern United States whose range extends northward through the Mississippi Valley. An additional two specimens of *P. multilineata* from Randolph County (Current River) and Conway County (near Menifee) also were found in the USNM collections (USNM 50221-50222). This study is the first published report of *P. multilineata* from Arkansas.

Placobdella multilineata is a blood-feeding leech on reptiles and amphibians. Reported hosts include turtles, alligators, and amphiumas (Sawyer and Shelley 1976, Forrester and Sawyer 1974, Saumure and Doody 1998). It is unknown whether *P. multilineata* exhibits seasonal host attachment. Specimens were found free-living on 31 July 2005 and 2-3 August 2005 in this study.

Placobdella ornata
(Verrill 1872) (Fig. 7)

Locality: Randolph County, Eleven Point River, 1 spm; White County, Lake Bald Knob (35 20.4069 N 91 35.2448 W), 1 spm.

Placobdella ornata is common and has a widespread distribution throughout northern North America but is less well

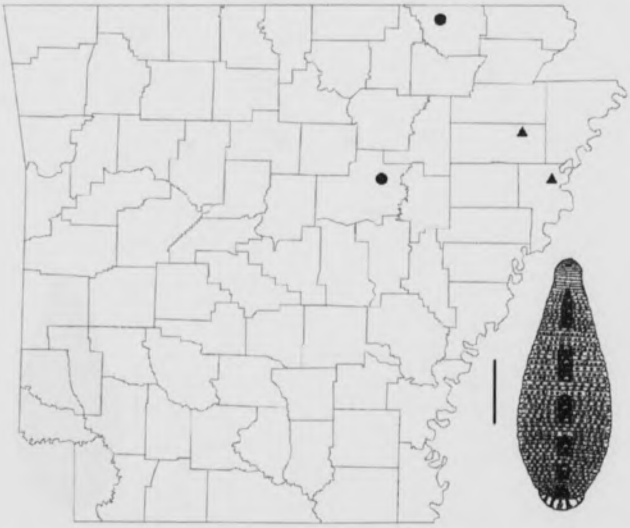


Fig. 7. Distribution map and line drawing of *Placobdella ornata* from Arkansas (circles – current study; triangles – published records; scale bar is 1.0 cm).

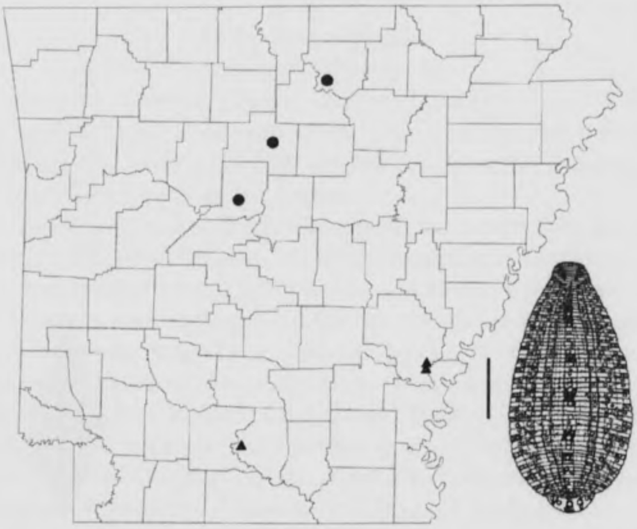


Fig. 8. Distribution map and line drawing of *Placobdella papillifera* from Arkansas (circles – current study; triangles – published records; scale bar is 1.0 cm).

known in the southern United States. It has been reported from Oklahoma and Texas (Klemm 1985) and in the Wapanocca National Wildlife Refuge and St. Francis Sunken Lands in Arkansas (Harp and Harp 1980, Cochran and Harp 1990).

Placobdella ornata is a opportunistic blood-feeding leech. It principally blood-feeds on turtles, but other reported hosts include fish, amphibians, birds, and humans (Moore 1964, Sawyer 1972, 1986, Klemm 1991, 1995, Moser 1991). *P. ornata* does not exhibit seasonal host attachment. After a blood-meal, it detaches from its host (Sawyer 1986). In this study, the specimen from the Eleven Point River was found free-living, and the specimen from Lake Bald Knob was found on the leg of one of the authors (WEM) after walking through aquatic vegetation.

Placobdella papillifera
(Verrill 1872) (Fig. 8)

Locality: Conway County, Overcup Lake (35 12.613 N 92 42.712 W), 4 spms; Independence County, Unamed creek NE of Batesville (35 47.6333 N 91 36.8666 W), 1 spm; Van Buren County, Honey Hill Pond (35 35.5407 N 92 26.1546 W), 1 spm.

Placobdella papillifera is widely distributed throughout North America, including sites in Arkansas, Louisiana, Mississippi and Texas (Klemm 1985, Chordas et al. 1996). It is a temporary blood-feeding leech on reptiles. Reported hosts include turtles and alligators, but the species is typically found free-living (Klemm 1985, Sawyer 1986). All specimens reported in this study were found free living and attached beneath submerged substrata.

Placobdella parasitica
(Say 1824) (Fig. 9)

Locality: Conway County, Brewer Lake (35 13.661 N 92 36.661 W), 2 spm; Conway County, Overcup Lake (35 12.613 N 92 42.712 W), 1 spm; Independence County, White River vic. Batesville (35 45.21N 91 37.94 W), 1 spm; Fulton County, Warm Fork River (36 29.63 N, 91 31.93 W), 3 spms; Van Buren County, Choctaw Lake, boat lauch (35 31.7887 N 92 22.7164

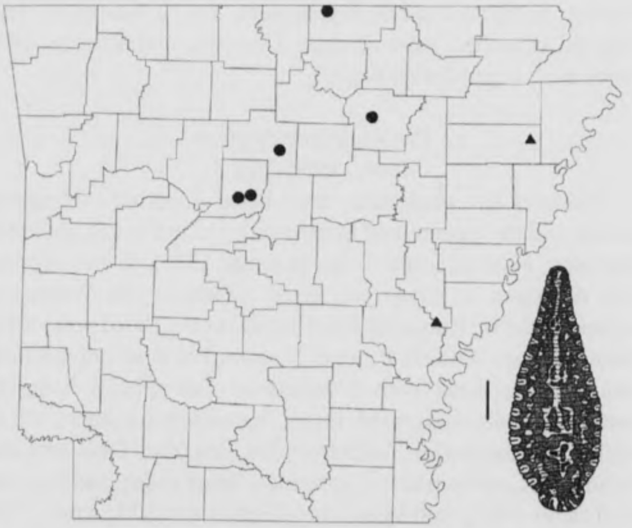


Fig. 9. Distribution map and line drawing of *Placobdella parasitica* from Arkansas (circles – current study; triangles – published records; scale bar is 1.0 cm).

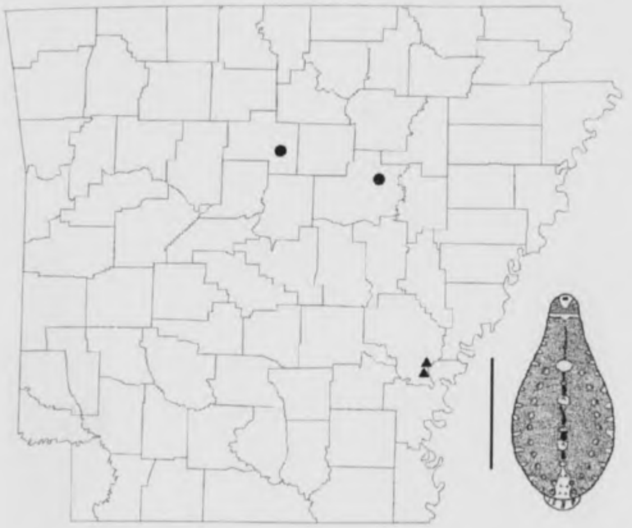


Fig. 10. Distribution map and line drawing of *Placobdella phalera* from Arkansas (circles – current study; triangles – published records; scale bar is 0.5 cm).

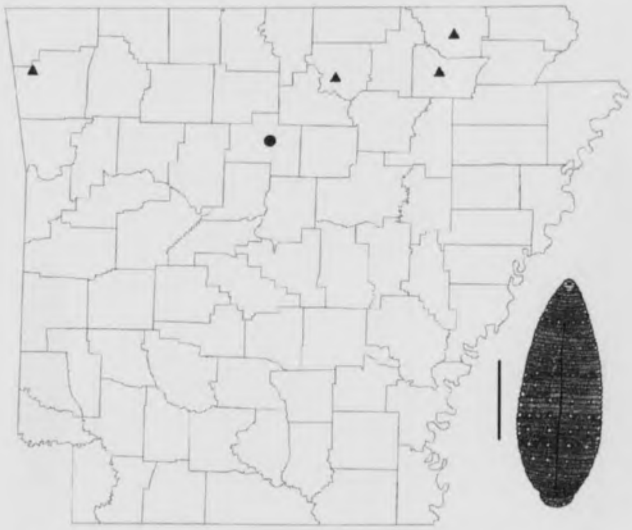


Fig. 11. Distribution map and line drawing of *Placobdella picta* from Arkansas (circle – current study; triangles – published records and museum specimens; scale bar is 1.0 cm).

W), 1 spm.

Placobdella parasitica has been reported throughout Canada and the eastern United States. Additional records of *P. parasitica* from Arkansas were reported by Cochran and Harp (1990) from St. Francis Sunken Lands and by Chordas et al. (1996) from White River National Wildlife Refuge. It was also reported from Arkansas by Sawyer (1972) but no locality data were given.

Placobdella parasitica is an opportunistic blood-feeding leech on turtles. Reported turtle host species are listed by Moser (1995) and Watermolen (1996). Any turtle species occurring in North America is considered a potential host. *P. parasitica* exhibits seasonal host attachment, detaching in the spring to fall in order to breed and brood (Sawyer 1972, Koffler et al. 1978, Moser 1993). Specimens were found attached to *Chelydra serpentina* (Common Snapping Turtle) on 22 September 2005 and *Pseudemys concinna* (Eastern River Cooter) on 3 October 2005. *P. parasitica* was also found free-living on 2-3 August 2005.

Placobdella phalera
(Graf 1899) (Fig. 10)

Locality: Van Buren County, Choctaw Lake, boat lauch (35 31.7887 N 92 22.7164 W), 1 spm; White County, Lake Bald Knob (35 20.4069 N 91 35.2448 W), 3 spms.

In a recent phylogenetic analysis of the family Glossiphoniidae, Siddall et al. (2005) reestablished the combination *Placobdella phalera* and suppressed the genus *Desserobdella*. *P. phalera* has a scattered distribution throughout eastern North America. It has been reported from Louisiana and Texas by Klemm (1985). Additional records of *P. phalera* from Arkansas have been reported by Chordas et al. (1996) from the White River National Wildlife Refuge.

Placobdella phalera is a blood-feeding leech on fish. It is opportunistic having been reported from fishes of the families Centrarchidae, Ictaluridae, Amiidae, and Acipenseridae in the field, and Anguillidae, Cyprinidae, Esocidae, Gasterosteidae, and Salmonidae in the laboratory (Smith and Taubert 1980, Amin 1981, Jones and Woo 1990). All specimens of *P. phalera* found in this study were free-living and attached beneath submerged substrata.

Placobdella picta
(Verrill 1872) (Fig. 11)

Locality: Van Buren County, McIntire Pond no.2 (35 36.585 N 92 28.824 W), 4 spms.

The former type species of the genus *Desserobdella*, *Clepsine picta*, was reclassified as *Placobdella picta* by Siddall et al. (2005). *P. picta* is widely distributed throughout northern North America. It has also been reported from Arkansas by Klemm (1982, 1985), McAllister et al. (1995) from SW of Melbourne (Izard County), Briggler et al. (2001) from a small pond in Washington County (36 05 N 94 23 W), and Turbeville and Briggler (2003) with no locality specified.

Placobdella picta is a temporary ectoparasite on amphibians (Sawyer 1972, Barta and Sawyer 1990, Klemm 1985) and is an important regulator of amphibian populations (Brockleman 1969, Berven and Boltz 2001). In this study, specimens were found in a small woodland pond, which is the typical habitat of *P. picta* (Sawyer 1972, Briggler et al. 2001).

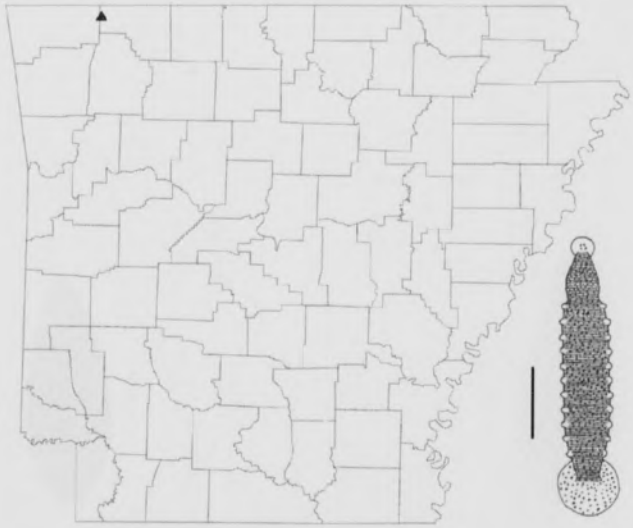


Fig. 12. Distribution map and line drawing of *Cystobranchus verrilli* from Arkansas (triangle – published record; scale bar is 0.5 cm).

Fig. 13. Distribution map and line drawing of *Gonimosobdella klemmi* from Arkansas (triangles – published records) (drawing from Williams and Burreson 2005; reprinted with permission; scale bar is 0.5 cm).

Family Piscicolide

Cystobranchus verrilli
Meyer, 1940 (Fig. 12)

Cystobranchus verrilli is infrequently collected and has a scattered distribution throughout eastern North America (Klemm 1985). Becker et al. (1966) reported this species from Arkansas in the White River drainage prior to the impoundment of Beaver Reservoir. *C. verrilli* is a blood-feeding leech on fish with members of the families Centrarchidae, Percidae, Ictaluridae, and Lotidae as reported hosts (Meyer 1940, Meyer and Moore 1954).

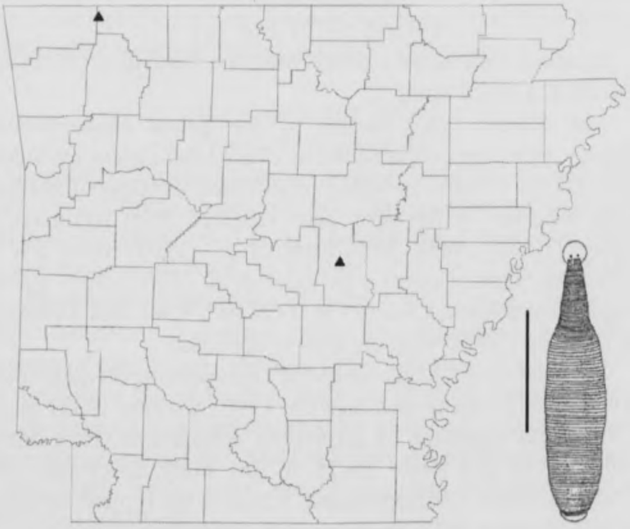


Fig. 14. Distribution map and line drawing of *Myzobdella lugubris* from Arkansas (triangles – published records; scale bar is 0.5 cm).

Gonimosobdella klemmi
Williams and Burreson, 2005 (Fig. 13)

Gonimosobdella klemmi is a new genus and species that was recently described by Williams and Burreson (2005) from cyprinid fishes in Arkansas (The type locality is the Middle Fork of the Little Red River in Searcy County, just N of the Van Buren County line), Illinois, and Missouri. The species superficially resembles the genus *Cystobranchus*. The full distribution and host preference of *G. klemmi* is not known.

Myzobdella lugubris
Leidy, 1851 (Fig. 14)

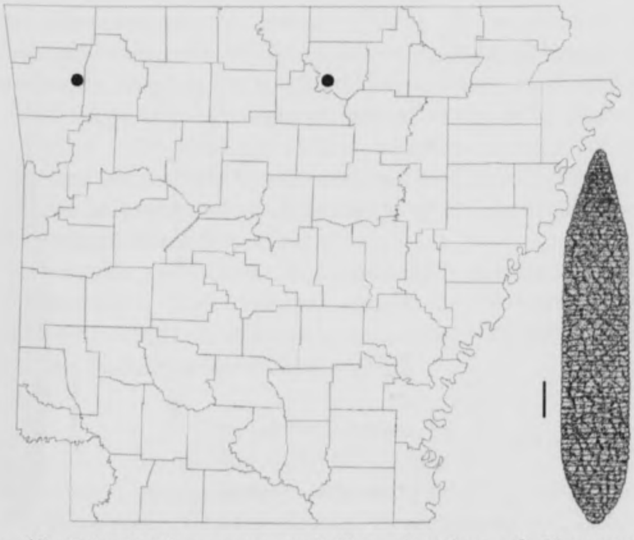
Myzobdella lugubris is common and widely distributed throughout North American fresh and brackish waters (Sawyer and Shelley 1976, Klemm 1985). Becker et al. (1966) reported this species from Arkansas in the White River drainage prior to the impoundment of Beaver Reservoir and Lonoke Hatchery. *M. lugubris* is an opportunistic blood-feeding leech on fish. Any fish species occurring in North America is considered a potential host. Reported fish host species are listed by Meyer (1940, 1946), Sawyer (1986), and Klemm (1982, 1995).

Piscicolaria reducta
Meyer, 1940 (Fig. 15)

Piscicolaria reducta is infrequently collected and has a scattered distribution in eastern North America (Great Lakes and Mississippi drainage systems; Klemm, 1985). This species was

Fig. 15. Distribution map and line drawing of *Piscicolaria reducta* from Arkansas (the specimen location is unknown; scale bar is 0.5 cm).

reported from Arkansas by Klemm (1982), however no locality data was given. *Piscicolaria reducta* is an opportunistic blood-feeding leech on fish. It has been reported from a wide variety of fish, including members of the family Cyprinidae, Percidae, Catostomidae, Centrarchidae, Ictaluridae, and Cichlidae (Meyer 1940, 1946, 1954, Booth and Aliff 1979, Klemm 1982, Price and Nadolny 1993).



ig. 17. Distribution map and line drawing of *Haemopis armorata* from Arkansas (circles – current study; scale bar 1.0 cm).

Fig. 16. Distribution map and line drawing of *Haemopis terrestris* from Arkansas (triangle – published record; scale bar is 1.0 cm).

Suborder Arhynchobdellida
Family Haemopidae

Haemopis terrestris
(Forbes 1890) (Fig. 16)

Haemopis terrestris is a terrestrial leech. It is rarely encountered and has a general mid-western distribution. *Haemopis terrestris* was reported from Arkansas by Klemm (1982, 1985) based on a specimen from Pine Bluff in the USNM (USNM 20804). *Haemopis terrestris* is typically found in damp soil beneath rocks and logs, where it feeds upon large earthworms (Sawyer and Shelley 1976, Shelley et al. 1979).

Haemopis marmorata
(Say 1824)* (Fig. 17)

Locality: Benton County: in a small tributary of Spavinaw Creek just off State Highway 279, at a point 2.95 km N of its junction with St. Hwy 102 (36 23.06 N, 94 19.55 W), 1 spm; Fulton County, Spring River - Bayou Access (36 27.79 N, 91 31.56 W), 1 spm.

Haemopis marmorata is widely distributed throughout North America but is less well known from the southern United States (Klemm 1985). This study is the first report of *H. marmorata* from Arkansas.

Haemopis marmorata is an opportunistic predaceous and scavengous feeder. Reported foods include earthworms, aquatic worms, insect larvae, small crustaceans, other leeches, dead vertebrates, and whole snails and clams (Sawyer 1972, 1974, 1986, Klemm 1995). It is an amphibious nocturnal leech that forages some distance from the water's edge (Moore 1912, Moser 1991). Specimens in this study were found in the shallow

Fig. 18. Distribution map and line drawing of *Macrobdella diplotertia* from Arkansas (triangles – published records; scale bar is 1.0 cm).

Fig. 19. Distribution map and line drawing of *Macrobdella ditetra* from Arkansas (triangle – published record; scale bar is 1.0 cm).

water of a small creek and crawling across an open gravel area between a river's bank and aquatic vegetation.

Family Hirudinidae

Macrobdella diplotertia

Meyer, 1975 (Fig. 18)

Macrobdella diplotertia is only known from Missouri, Arkansas and Kansas (Meyer 1975, Klemm 1985, Turbeville and Briggler 2003, Trauth and Neal 2004). *M. diplotertia* is an omnivore. It has been reported to feed on frog and salamander eggs, and human blood (Turbeville and Briggler 2003, Trauth and Neal 2004). *M. diplotertia* likely blood-feeds on amphibians as do other members of the genus.

Macrobdella ditetra

Moore, 1953 (Fig. 19)

Macrobdella ditetra is distributed throughout the southern United States (Sawyer 1967, Klemm 1985). It was reported from Arkansas by Klemm (1985) based on two specimens collected from Locust Bayou (USNM 56782). *M. ditetra* is an omnivore. Reported hosts include frogs, small fish, and frog eggs (Moore 1953, Beckerdite and Corkum 1973).

Family Erpobdellidae

Erpobdella fervida

(Verrill 1871)* (Fig. 20)

Locality: Faulkner County, Beaver Fork Lake, near Conway (35 08.140 N 92 27.523 W), 6 spms; Jackson County, Roadside ditch off US 14, southside of road, 0.3 km W of Amagon (35 33.716 N 91 06.936 W), 1 spm.

Siddall (2002) suppressed the genus *Mooreobdella* and made the combination *Erpobdella fervida. E. fervida* is widely distributed throughout northern North America but is less well known from the southern United States (Klemm 1985). It has been reported from Kansas and Missouri (Sawyer 1967, Klemm 1985). This study constitutes the first report of this species from Arkansas.

Fig. 20. Distribution map and line drawing of *Erpobdella fervid* from Arkansas (circles – current study; scale bar is 1.0 cm).

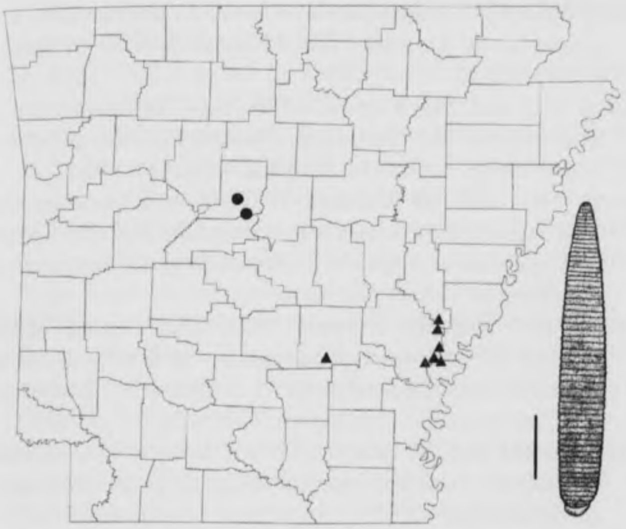


Fig. 21. Distribution map and line drawing of *Erpobdella microstoma* from Arkansas (circles – current study; triangle – published record; scale bar is 1.0 cm).

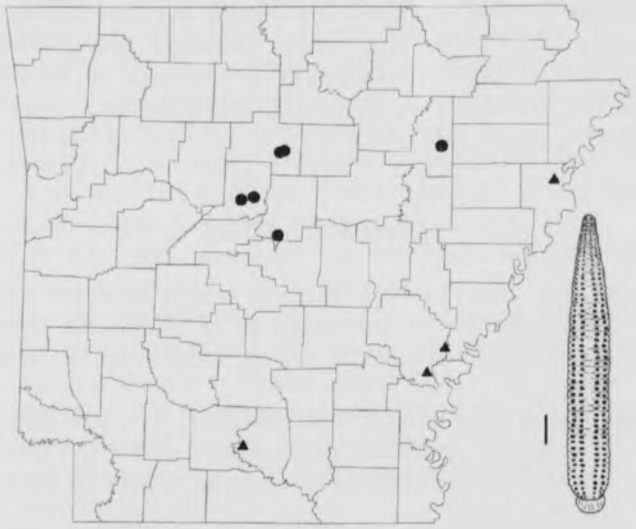


Fig. 22. Distribution map and line drawing of *Erpobdella punctata* from Arkansas (circles – current study; triangles – published record and museum specimens; scale bar is 1.0 cm).

Erpobdella fervida is a predaceous and scavengous feeder. It has been reported to feed on oligochaetes and insect larvae and is commonly associated with dead animals at the shoreline (Moore 1912, Sawyer 1972).

Erpobdella microstoma

(Moore 1901) (Fig. 21)

Locality: Conway County, Overcup Lake (35 12.613 N 92 42.712 W), 3 spms; Perry County, Plummerville Bottoms, Arkansas River (35 06.663 N 92 38.350 W), 4 spms.

Erpobdella microstoma was transferred from the suppressed genus *Mooreobdella* by Siddall (2002). *E. microstoma* is widespread throughout the United States and Eastern Canada. It is commonly found in the southern United States, especially in the Mississippi-Ohio drainage systems (Sawyer 1972, Sawyer and Shelley 1976, Klemm 1985). Additional records of *E. microstoma* from Arkansas were reported by Curry (1976) from Bayou Bartholomew and by Chordas et al. (1996) from the White River National Wildlife Refuge.

Erpobdella microstoma is a predaceous species, feeding on oligochaetes, insect larvae, and snails (Sawyer 1972, Klemm 1985, 1995). It seems to be associated with low-oxygen organically polluted water (Klemm 1985).

Erpobdella punctata

(Leidy 1870) (Fig. 22)

Locality: Conway County, Brewer Lake (35 13.661 N 92 6.661 W), 6 spms; Conway County, Overcup Lake (35 12.613 92 42.712 W), 4 spms; Faulker County, Lake Conway, ayflower, off Route 40 (35 58.2466 N 92 25.314 W), 10 spms; ackson County, Roadside ditch off US 14, 1.4 km W of Amagon (35 33.780 N 91 07.664 W), 6 spms; Van Buren County, Choctaw Lake (35 31.6007 N 92 24.4126 W), 1 spm; Van Buren County, Choctaw Lake, Bruce's Ferry Landing (35 32.1396 N 92 22.2735 W), 3 spms.

Erpobdella punctata is common throughout northern North America but is less well known from the southern United States (Klemm 1985). It has been reported from Louisiana (Sawyer 1967, Klemm 1985) and from Arkansas in the Wapanocca National Wildlife Refuge (Harp and Harp 1980) and the White River National Wildlife Refuge (Chordas et al. 1996). An additional specimen of *E. punctata* from Calhoun County (Ouachita River, Locust Bayou) also was found in the USNM collections (USNM 52048).

Erpobdella punctata is an opportunistic predator and scavenger. Reported food items include aquatic insect larvae, oligochaetes, small crustaceans, and snails (Sawyer 1970, 1972, Davies and Everett 1975, Klemm 1991, 1995). *E. punctata* is associated with organically enriched habitats where its food is in abundance (Klemm 1995).

Conclusions

Twenty-two species of leeches are now known from Arkansas. Additional studies are needed to elucidate the entire leech fauna of Arkansas, as many regions of the state remain unstudied. Based on their presence in surrounding states, an additional 8 species of leeches (*Actinobdella inequiannulata*, *Glossiphonia complanata*, *Placobdella nuchalis*, *Placobdella pediculata*, *Placobdella translucens*, *Piscicola punctata*, *Philobdella gracilis*, and *Erpobdella melanostoma*) likely occur in Arkansas but have not been collected.

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The Perkin Reaction: Rapid and Efficient Process Optimization Through a Microwave/Design of Experiments Couple

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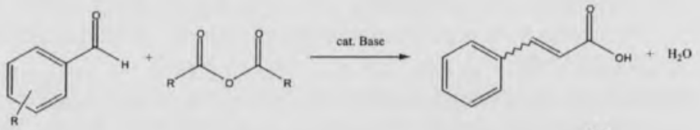
Abstract.—Microwave chemistry and a Design of Experiments (DOE) protocol were employed together in order to rapidly and efficiently optimize a modified Perkin reaction. Microwave heating significantly reduced the reaction time, and the DOE provided a statistically significant understanding of underlying process relationships in a minimum number of experimental runs. In all, the reaction time was reduced from 1 hour to 2 minutes, factors important to yield were identified, an interesting cross-term interaction was discovered, and it was demonstrated that the more economical sodium acetate trihydrate catalyst was a viable alternative to the more costly anhydrous sodium acetate.

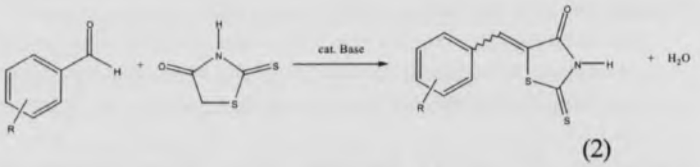
Key words:— Microwave chemistry, Design of Experiments (DOE) protocol, Perkin reaction, sodium acetate trihydrate catalyst, anhydrous sodium acetate.

Introduction

The base-catalyzed condensation of aromatic aldehydes with acid anhydrides, called the Perkin reaction, is a classical method for the synthesis of α,β-unsaturated carboxylic acids as shown in Equation 1 (March, 1985). A variation of this reaction involves the use of rhodanine instead of an acid anhydride as a route to a variety of biologically active compounds as shown in Equation 2 (Brown 1961, Foye and Torivich 1977). However, long reaction times and high temperatures (Mayo et al. 1994; Sykes, 1987) and/or exotic base catalysts (Vererkova et al. 1999) are required for the Perkin reaction.



(1)



(2)

Acceleration of the reaction rate using microwaves could potentially reduce reaction times from hours to just a few minutes. Additionally, microwave heating is an efficient energy alternative over the classical thermal sources, which are highly inefficient.

Verekova, et al. (1999) reported that microwaves do accelerate the Perkin reaction (Equation 1), but their best yields were obtained after 10 minutes at 800-Watts using toxic cesium catalysts. Although sodium acetate is the most common base catalyst, reports suggest that it must be anhydrous (Mayo et al. 1994). Given the previous long reaction time (10 minutes) at high microwave power (800 Watts) using a toxic catalyst (Vererkova et al. 1999), we decided to investigate optimizing this reaction using an efficient statistical approach called Design of Experiments (DOE). The acceleration of reactions using microwave chemistry combined with the resource efficiency of DOE constitutes a powerful process for optimizing chemical reactions. Herein, we demonstrate the utility of the microwave-DOE couple through the optimization of the Perkin reaction (Equation 2).

DOE is a large area of statistics that provides a way to consider the effects of all variables of a process on a set of outcomes. It does so in a uniform but simultaneous way through the construction of a mathematical model that has statistically significant predictive value within a defined design space (Box et al. 1978, Laird 2002). Chemists in the corporate sector have largely embraced DOE as a credible tool for optimizing processes, developing predictive models for reactions, and understanding complex variable interactions with a minimum of experiments (e.g., without having to run all possible combinations of variables and their levels) (Hendrix 1979, Owen et al. 2001). Historically, it appears that the academic sector has been reluctant (Lendrem et al. 2001) to employ DOE methods in chemical research, but this is beginning to change (Carlson et al. 2001, Carlson 2005). While DOE does not provide a comprehensive solution to process optimization, it does offer the chemist several advantages over the classical one-variable-at-a-time (OVAT) approach, including error analysis of the experimental process as well as the statistical model itself, detection of complex interactions between reaction parameters that influence experimental results, a finite number of experiments to reach research objectives, and the construction of a predictive mathematical model of the reaction within the experimenter-determined boundaries of the design (Bayne and ubin, 1986). Common objections chemists have to the use of OE for optimization of chemical processes include a lack of ufficient material resources or statistics trumping chemical ntuition. Along with these, a host of other perceived obstacles ave been adequately addressed in another forum (Lendrem et al. 2001).

Microwave assisted acceleration of organic reactions has emerged within the past twenty years as a viable alternative to conventional thermal methods (Hayes 2002, Marx 2004). Reactions carried out under conventional thermal conditions are often accompanied by long reaction times, undesired side product formation, and/or low yields. Microwave methodologies provide viable alternatives to classical thermal approaches for drug discovery efforts (Rose 2002), analytical chemistry (Kingston and Jassie 1988), protein synthesis (Yu et al. 1992), and green chemistry (Mingos 1994). Microwave techniques often provide the opportunity for carrying out organic transformations in a solventless or solid phase environment (Larhed et al. 2002).

Materials and Methods

Based on the fundamental chemical nature of this reaction, we believed that a 2-level designed experiment consisting of 4 factors (catalyst loading (**A**), acetic acid level (**B**), *o*-chlorobenzaldehyde level (**C**), and microwave time (**D**)) would provide the most information on underlying relationships affecting yield. A 2-level screening experiment is the best option for revealing all main and two-factor (cross-term) effects. This 2-level, 4-factor DOE translated into 2^4 (16) experiments to uniformly cover the design space as shown in Table 1.

Inclusion of 6 center point experiments (all factors at midrange settings) provided a way to measure variability due to experimenter/process error. In other words, with a minimum of 6 center points, total error can be separated into two components: pure error due to the experiment and error due to model lack-of-fit. Table 1 shows the experiments carried out.

Table 1. Factor Settings for the 2-Level Experiment at 330-Watts Microwave Power

Run	Factor Settings			
	mmol NaOAc	mL HOAc	mmol o-CB	MW Time sec
1	0.500	0.500	0.200	30
2	0.500	0.500	0.200	120
3	0.0100	0.500	0.600	30.0
4	0.0100	0.500	0.200	30.0
5	0.500	0.500	0.600	30.0
6	0.0100	2.00	0.200	120
7	0.500	2.00	0.600	30.0
8	0.0100	0.500	0.600	120
9	0.255	1.25	0.400	75.0
10	0.255	1.25	0.400	75.0
11	0.500	2.00	0.200	30.0
12	0.0100	0.500	0.200	120
13	0.500	2.00	0.600	120
14	0.255	1.25	0.400	75.0
15	0.500	0.500	0.600	120
16	0.0100	2.00	0.600	120
17	0.0100	2.00	0.600	30.0
18	0.500	2.00	0.200	120
19	0.255	1.25	0.400	75.0
20	0.0100	2.00	0.200	30.0
21	0.255	1.25	0.400	75.0
22	0.255	1.25	0.400	75.0

The experimental work was carried out in a domestic Panasonic® NN-S540 microwave oven equipped with an inverter that allowed for the actual lowering of the power output to a selectable level (e.g., 330 Watts; Varma and Namboodiri 2001). All chemicals were used as received without any further purification. A 5-mL conical vial was charged with 0.20 mmol rhodanine (Aldrich) and the appropriate amounts of sodium acetate trihydrate (Fisher) and glacial acetic acid (Fisher) as shown in Table 1. Using an automatic pipet, the appropriate volume of *o*-chlorobenzaldehyde (Aldrich) was added in one portion to the conical vial (Table 1). The vial was capped and placed in the microwave at 330 Watts for the appropriate time (Table 1). After microwaving, the vial was removed from the oven and placed in an ice-bath. The resulting yellow crystals were isolated via vacuum filtration and washed with 2 × 1-mL cold glacial acetic acid followed by 2 × 1-mL cold deionized water. Upon air drying, the yield was determined, and the melting point and infrared spectrum were obtained.

Results and Discussion

The singular reason why we chose microwave heating was to drastically shorten the reaction time. Clearly, without any statistical analysis, the efficacy of microwave heating was affirmed. However, the recommended 800 Watts of microwave heating (Verekova et al. 1999) was discovered to be somewhat excessive. We found that 330 Watts of microwave heating provided the energy necessary for this reaction.

Another clear result from this set of experiments was the efficacy of the sodium acetate trihydrate catalyst. In general, anhydrous salts are difficult to prepare, hard to handle, and more costly than hydrates. Therefore, the fact that sodium acetate trihydrate proved to be a viable catalyst constitutes another significant improvement in this process.

The intent of the DOE was three-fold: 1) identify underlying relationships between factors, 2) develop a first-generation mathematical model of the process, and 3) provide insight for further development work. In the statistical analysis, the half-normal probability plot revealed that the main effects of all 4 factors as well as several cross-term interactions were significant

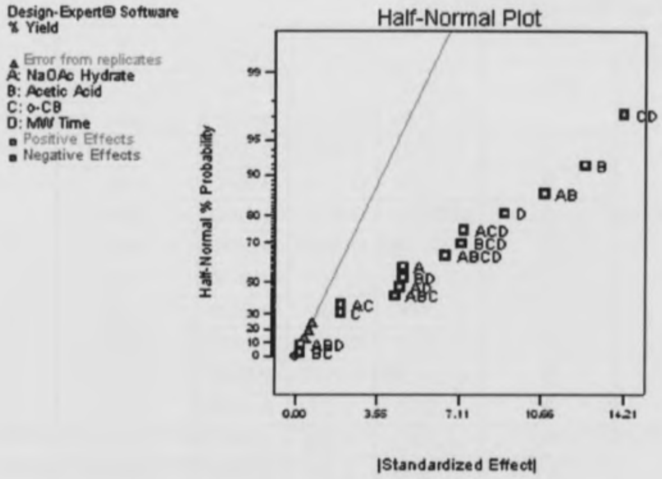


Fig. 1. Half-normal plot showing main and cross-term effects on the yield.

(Fig. 1). Half-normal probability plot is the fundamental method for selecting 2-level effects. In other words, the plot of the ordered values of a sample versus the expected ordered values from the true population will be approximately a straight line. Any terms that turn out to be important for the statistical model show up as outliers.

It should be pointed out that many 2-, 3-, and 4-factor terms appear to be significant, but such appearances can be deceiving. As shown in the ANOVA table (Table 2), effects that are significant in explaining process variability are clearly separated from effects that are not significant on the basis of the magnitude of the p-value. Small p-values (<0.05) suggest that there is model effect (*e. g.*, the term is significant in explaining process variability). This, however, is a screening experiment. Therefore, this experiment only gives us information concerning factors and interactions that may be important in explaining process variability of selected responses (*i. e.*, yield) within the design space. What is clear is that the sodium acetate level, acetic acid volume, *o*-chlorobenzaldehyde level, and the microwave time significantly impacted yield as well as several interaction terms. The expression for the predicted yield is given as Equation 3.

$$\begin{aligned} Yield &= 21.1 + 155.4A + 8.8B - 12.9C - 0.03D - 111.1AB - 362.7AC \\ &- 1.43AD - 11.8BC - 0.05BD + 0.4CD + 206.9ABC \\ &+ 0.8ABD + 4.1ACD - 0.03BCD - 2.0ABCD \end{aligned} \tag{3}$$

Interactions are best understood through 3-dimensional plots. Figure 2 shows three cube plots at the low, medium, and high microwave times (30 sec, 75 sec, and 120 sec), which reveal the behavior of the reaction yield at the extremes of the design space.

The lower right edges of the cube plots reveal a most interesting interaction (the CD interaction shown in Table 2) between *o*-chlorobenzaldehyde level (*C*) and microwave time (*D*). At 30 seconds microwave time, the best yield occurs with 0.50 mol NaOAc, 0.5 mL HOAc, and 0.20 mol (or, one equivalent) *o*-chlorobenzaldehyde. On the other hand, at 120 seconds microwave time, the best yield occurs with 0.50 mol NaOAc, 0.5 mL HOAc, and 0.60 mol (*e. g.*, three equivalents) *o*-chlorobenzaldehyde. Figure 3 shows this *CD* cross-term interaction with two 3-D plots (one at 30 microwave seconds, the other at 120 microwave seconds) of the yield versus NaOAc level and *o*-chlorobenzaldehyde level. Clearly, after 30 seconds at 330 Watts, the optimum yield occured at the lowest level (1 equivalent) of *o*-chlorobenzaldehyde and the highest level of

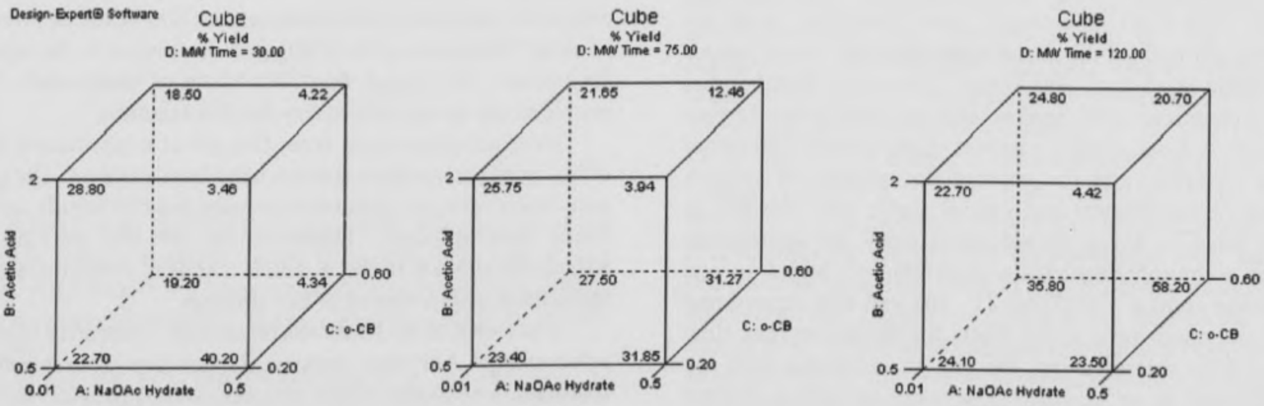


Fig. 2. Cube plots of the yield at the edges of the DOE design space.

able 2. ANOVA Table for the DOE.

Source	Sum of Squares	Degrees of Freedom	Mean Square	F-Value	p-value	Significant?
					Prob > F	Yes/No
Model	3195.584	15	213.0389	113.814	< 0.0001	Yes
A-NaOAc Hydrate	88.1721	1	88.1721	47.10509	0.0010	Yes
B-Acetic Acid	630.5121	1	630.5121	336.845	< 0.0001	Yes
C-o-CB	15.7609	1	15.7609	8.420109	0.0337	Yes
D-MW Time	331.24	1	331.24	176.9618	< 0.0001	Yes
AB	466.9921	1	466.9921	249.486	< 0.0001	Yes
AC	15.7609	1	15.7609	8.420109	0.0337	Yes
AD	82.81	1	82.81	44.24044	0.0012	Yes
BC	0.2025	1	0.2025	0.108184	0.7556	No
BD	87.9844	1	87.9844	47.00482	0.0010	Yes
CD	807.6964	1	807.6964	431.504	< 0.0001	Yes
ABC	74.8225	1	74.8225	39.9732	0.0015	Yes
ABD	0.2304	1	0.2304	0.123089	0.7400	No
ACD	213.7444	1	213.7444	114.1909	0.0001	Yes
BCD	209.0916	1	209.0916	111.7052	0.0001	Yes
ABCD	170.5636	1	170.5636	91.12196	0.0002	Yes
Curvature	1305.867	1	1305.867	697.6468	< 0.0001	Yes
Pure Error	9.359083	5	1.871817			
Cor Total	4510.81	21				

sodium acetate. Alternatively, after 120 seconds at 330 Watts, the optimum yield occured at the highest level (3 equivalents) of *o*-chlorobenzaldehyde and the lowest level of sodium acetate. The explanation for this unexpected result is not clear at this point. However, this *CD* interaction (or, any other interaction for that matter) would have gone undetected in the conventional OVAT method of experimentation.

At least three interactions (*AB, ABC,* and *ABCD*) identified in the DOE (see Table 2) may partially be explained by a combination of the facts that sodium acetate/acetic acid constitute a buffer system and that this modified Perkin reaction is acid catalyzed. Therefore, this suggests the possibility of a subsequent DOE where the levels of sodium acetate and acetic acid may be combined into one buffering pH term.

Conclusions

The DOE clearly identified that all 4 model factors are important in explaining the variability of the yield data. Further, an unexpected, but interesting, cross-term interaction was identified involving *o*-chlorobenzaldehyde and microwave time at 330 Watts (*CD*). Other interactions involving the coupling of sodium acetate and acetic acid suggest that combining these terms in a single buffering pH term may be important in subsequent work. Additionally, it was shown that the anhydrous sodium acetate catalyst could be replaced by the more economical trihydrate and that high wattage microwaves are not required for this process (330 Watts work as well as 800 Watts).

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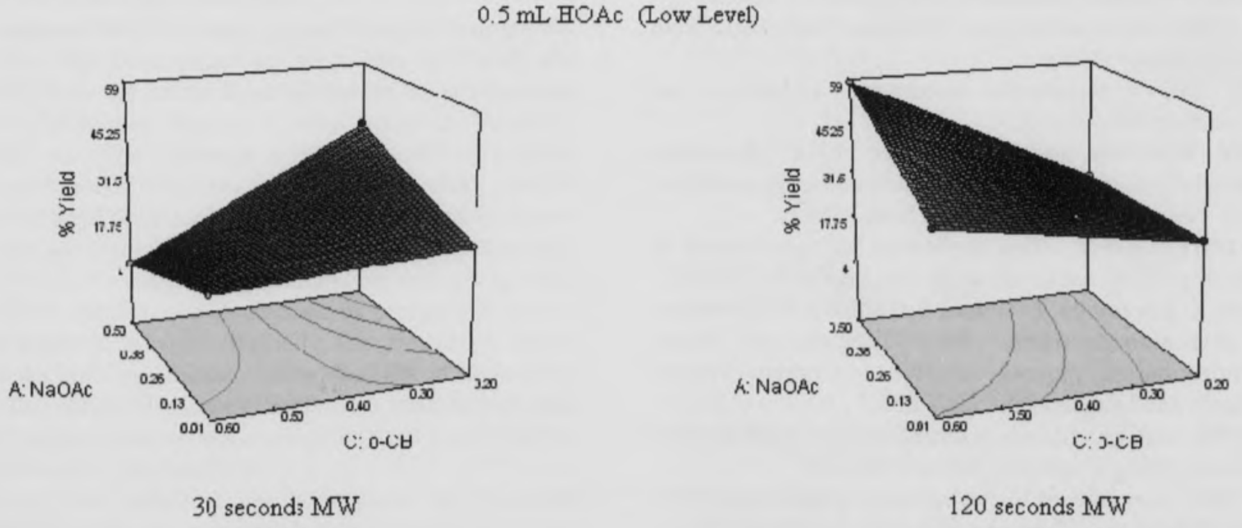
0.5 mL HOAc (Low Level)

30 seconds MW

120 seconds MW

ig. 3. 3-D plots of the yield within the design space.

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Status Survey of the Peppered Shiner, *Notropis perpallidus* Hubbs and Black, in Arkansas and Oklahoma

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Abstract.—The peppered shiner, *Notropis perpallidus* Hubbs and Black, is a small, silvery, upland stream fish found only in southwestern Arkansas and eastern Oklahoma. This fish species was studied from 1999-2001 to determine its distribution, habitat, and conservation status. A total of 81 collections was made during the 2-year study; however, only 17 specimens were collected. The present distribution of the peppered shiner in Arkansas and Oklahoma is described as well as the conservation status of this cyprinid in both states.

Key words:—Peppered shiner, *Notropis perpallidus*, Arkansas, Oklahoma, habitat, distribution, conservation, cyprinid.

Introduction

The peppered shiner, *Notropis perpallidus* Hubbs and Black, is a slender pallid minnow that is restricted to the tributaries of the Red and Ouachita rivers in southeast Oklahoma and southern Arkansas (Snelson et al 1980). It not only occupies large clear streams and rivers of the Ouachita Mountains, but also extends onto the Coastal Plain physiographic region of Arkansas in the Saline River system (Robison and Buchanan 1988). This fish has never been common and appears to have declined over the past 30 years in both Arkansas and Oklahoma. Its conservation status is uncertain, as little data exist on which to make a formal decision as to its need for federal protection. Its current formal status is just whether it is currently protected or not. That can be known.

A 2-year survey of the peppered shiner was initiated to determine the present distribution and abundance of this cyprinid in Arkansas and Oklahoma.

The peppered shiner is a rare cyprinid fish species originally described in 1940 from only two specimens by Hubbs and Black (1940) from the Saline River (Ouachita River Drainage) 8.1 km north of Warren, Bradley County, Arkansas. Actually, this cyprinid was first collected in the Saline River (UMMZ 197684) near Benton, Arkansas, in 1884 by America's premier ichthyologist David Starr Jordan and his student, Charles Henry Gilbert, although it was misidentified as *N. dilectus*. Snelson and Jenkins (1973) later studied the systematics of this rare species, redescribed the species, and established its presently accepted name, the peppered shiner. Originally, this diminutive shiner was called the colorless shiner. Today, the lack of knowledge regarding the peppered shiner's systematic relationships and life history requirements makes it one of the most poorly known cyprinid fishes in North America.

Relatively little attention has been given to this small shiner other than notations regarding locality records or cursory descriptions of ecological requirements (Miller and Robison 1973, Wagner et al. 1987, Robison and Buchanan 1988). In independent studies both Robison (1974) and Buchanan (1974) considered the peppered shiner as "rare" in Arkansas. In Oklahoma, both Robison et al. (1974) and the Rare and Endangered Species of Oklahoma Committee (1975) had also considered this species as "rare." More recently, Warren et al. (2000) reviewed the diversity, distribution, and conservation status of all native freshwater fishes of the southern United States. The peppered shiner was listed with a status of "vulnerable."

The peppered shiner is restricted to the Ouachita and Red rivers in southeastern Oklahoma and southern Arkansas (Snelson and Jenkins 1973, Robison and Buchanan 1988, Fig. 1).

Habitat.—Robison and Buchanan (1988) reported that the peppered shiner inhabits pool regions 0.6 – 1.2 m deep in moderate-sized, warm, clear rivers. They noted this species is

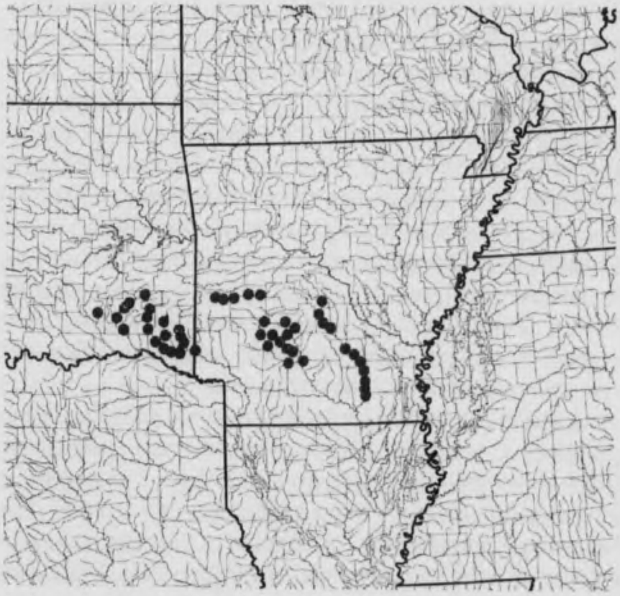

Fig. 1. Distribution of the peppered shiner, *Notropis perpallidus*, in Arkansas and Oklahoma.

rarely found in smaller streams. Typically, it occurs in the lee of islands and other obstructions away from the main current (Snelson and Jenkins 1973). Harris and Douglas (1978) reported the peppered shiner in the mainstream of the Ouachita River in water 0.6 – 1.2 m in depth with slow to moderate current. Most specimens Harris (1977) found were in habitat that included water willow (*Justicia americana*) and a rock and sand substrate. Wagner et al. (1987) found that substrate type was relatively unimportant in determining the microhabitat distribution of *N. perpallidus*, but depth and current were important. They reported that this species tends to occupy water deeper than 0.5 m (20 inches) where current speeds are less than 0.3 cm (.01 feet) per second. Page and Burr (1991) reported the peppered shiner from pools and sluggish areas of small to medium rivers, often in quiet water near vegetation. Moore (1948) collected a single specimen in a silty habitat below a dam on the Mountain Fork River in Oklahoma. In the Ouachita River drainage, it has been found both above and below the Fall Line and is often associated with beds of *Justicia americana* over a variety of habitats.

Species Associates.—The peppered shiner is commonly found in association with the bigeye shiner (*Notropis boops*), carmine shiner (*Notropis percobromus*), steelcolor shiner (*Cyprinella whipplei*), longear sunfish (*Lepomis megalotis*), blackspotted topminnow (*Fundulus olivaceus*), brook silverside (*Labidesthes sicculus*), northern hog sucker (*Hypentelium nigricans*), smallmouth bass (*Micropterus dolomieu*), and greenside darter (*Ethesotoma blennioides*).

Materials and Methods

Field work was conducted from September 1999 to November 2001. Eighty-one collections of fishes were made in Arkansas and Oklahoma during this status survey.

Fishes were collected using standard common sense minnow seines varying in length from 4.6-6 meters and 1.8 meters 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 peppered shiner were preserved from some sites and returned to the laboratory at Southern Arkansas University for further analysis. Associated fishes collected were also transported back to the laboratory where identifications of all species and counts of each were made.

In addition, all known contemporary and historical literature regarding the peppered shiner was reviewed and relevant findings summarized or referenced herein. Data were used from museum collections/known localities of peppered shiners collected in Arkansas and Oklahoma. Coverage includes the University of Michigan Museum of Zoology (UMMZ), Tulane University (TU), the University of Louisiana at Monroe (NLU), Arkansas State University Museum of Zoology (ASUMZ), the University of Arkansas (UA), Oklahoma State University (OSU) and the University of Oklahoma (OU).

Results and Discussion

Habitat.—In this study the peppered shiner was found just off concentrations of water willow beds and to the side of riffles or islands where the current is stronger. There is some indication that juvenile individuals utilize these beds as safe havens. Individuals seem to occupy areas of the stream away from the current in the lee of islands as reported earlier by Snelson and Jenkins (1973). Substrates where specimens were collected ranged from sandy areas and areas with gravel to some larger rocky areas. All specimens were collected from deep water and never in the shallow reaches of moderate-sized river sections. No specimens were taken from side tributaries of main rivers even though seining was done in side tributaries to see if perhaps the peppered shiner retreated into those regions.

This study shows the peppered shiner to be a midwater schooling species that prefers stream sections of clear, upland and lowland, medium to large rivers. It usually occurs over gravel or sand bottoms away from the current. The peppered shiner seems to have an affinity for aquatic vegetation such as *Justicia americana*, which is common within its range.

Distribution.—The peppered shiner is restricted to the tributaries of the Red and Ouachita rivers in southeast Oklahoma and southern Arkansas (Miller and Robison 1973, Snelson and Jenkins 1973, Snelson et al. 1980, Robison and Buchanan 1988, Fig. 1). The following is a presentation of the distribution of the peppered shiner by river system or main river area. Comments are made concerning its historical presence, plus the findings of this survey.

Ouachita River Mainstem, Arkansas.—Harris (1977) collected 74 specimens of the peppered shiner from 4 localities in the upper Ouachita River mainstem. The four localities were (1) Polk County: Ouachita River at McGuire Public Access, approximately 2.4 km (1.5 mi) south of St. Hwy. 88 (Sec. 23, T2S, R29W); (2) Ouachita River and Mill Creek at bridge, 1.1 km (0.7 mi) south of Cherry Hill (Sec. 9, T2S, R28W); (3) Montgomery County: Ouachita River at U. S. Hwy. 270 bridge at Rocky Shoals Recreation Area (Sec. 32, T1S, R25W); and (4) Ouachita River at Chasewood Landing, approximately 1.6 km (mi) east of St. Hwy. 298 (Sec. 28, T1S, R25W). In a subsequent study of the fishes of the upper Ouachita River, Herrock (1986) did not collect a single specimen of the peppered shiner in his survey of 31 stations during 1985-1986.

In this study, only 6 specimens of the peppered shiner were collected from the upper Ouachita River mainstem from only 2 localities of the 15 sampled (Table 1). The 2 localities were (1 Polk County: Ouachita River at McQuire Access Area, 2.4 km (1.5 mi) south of St. Hwy. 88 (Sec. 23, T2S, R29W) and (2) Polk County; Ouachita River, 1.1 km (0.7 mi) south of Cherry Hil able 1. Collecting localities, numbers of collections, and numbers of peppered shiners collected in Arkansas in 1999-2001.

Locality (River System)	No. of Collections	No. Sites w/Peppered Shiners/No. Collected
1. Ouachita River (mainstem)	15	2/6
2. Caddo River	10	0/0
3. Little Missouri River	11	0/0
4. Saline River	15	3/11
5. Kiamichi River	15	0/0
6. Mountain Fork River	10	0/0
TOTAL	76	5/17

(Sec. 9, T2S, R28W).

Raymond (1975) surveyed the lower Ouachita River below Remmel Dam to the AR/LA state line. He collected 42,246 specimens distributed among 111 species in 62 collections from 25 locations. In all these collections, only a single specimen of the peppered shiner was taken from the Ouachita River at the mouth of the Little Missouri River north of Chidester, AR.

Caddo River System, Arkansas.—Fruge (1971) originally surveyed the fishes of the Caddo River and reported 62 specimens of the peppered shiner out of 37,273 individual fishes collected at 4 different localities. The 4 localities were (1) Clark County: Caddo River at St. Hwy. 182, 2.7 km (1.7 mi) north of Amity (Sec. 22, T5S, R23W); (2) Caddo River, approximately 0.5 km (1/3 mi) below DeGray Dam (Sec. 14 and 23, T6S, R20W); (3) Caddo River just below control dam spillway, approximately 3.2 km (2 mi) west of Caddo Valley (Sec. 35 and 36, T6S, R20W); and (4) Caddo River at Chasewood Landing, approximately 1.6 km (1 mi) east of St. Hwy. 298 (Sec. 28, T1S, R25W). Five additional specimens of the peppered shiner were taken by NLU students from below the DeGray Lake dam in 1972. Lisa Herrock (1986) did not collect a single specimen of the peppered shiner in her subsequent 1985-1986 survey of the fishes of the Caddo River, although she collected 37,109 individual fish specimens in 67 collections from 21 localities.

In this study no specimens of the peppered shiner were taken in 10 collections made from the Caddo River system (Table 1).

Little Missouri River System, Arkansas.—Myers (1977) irst surveyed the fishes of the Little Missouri River from 1976-977 and reported 21 specimens of the peppered shiner from 3 ifferent locations out of 58 collections from 20 localities and total of 23,852 individual fishes. The 3 localities where the eppered shiner was collected were (1) Pike County: Antoine iver at St. Hwy. 26; (2) Little Missouri River at end of gravel ad (Sec. 3, T11S, R18W); (3) Little Missouri River at the nction with the Ouachita River (Sec. 1, T11S, R18W).

Later, Loe (1983) re-surveyed the Little Missouri River system from 1980-1983 and did not collect the peppered shiner even though he collected 25,039 fishes in 57 collections from 35 localities. Ponder (1983) surveyed the Terre Noire Creek, a large tributary of the Little Missouri River, and reported 87 specimens of the peppered shiner from 6 localities in a boat ditch.

In the present study not a single specimen of the peppered shiner was taken from the Little Missouri River system even though 11 collections were made from 10 localities in the system (Table 1).

Saline River System, Arkansas.—Hubbs and Black (1940) described the peppered shiner from only two specimens collected 8 km (5 mi) north of Warren, Bradley County, Arkansas. Reynolds (1971) first surveyed the fishes of the Saline River system, a Ouachita River tributary and collected only 23 specimens of the peppered shiner from 5 localities. The 5 localities in the Saline River system were: (1) Saline County: Saline River, 3.2 km (2 mi) east of St. Hwy. 67 on county road (Sec. 21, T2S, R15W); (2) Grant County: Saline River at St. Hwy. 229 (Sec. 4, T4S, R15W); (3) Saline River at St. Hwy 46, 4 km (2.5 mi) NE of Leola, AR (Sec. 8, T6S, R14W); (4) Drew County: Saline River at end of St. Hwy. 172 (Sec. 14, T14S, R9W); (5) Bradley County: Saline River at St. Hwy 4, 4.8 km (3 mi) east of Warren, AR (Sec. 3, T12S, R9W). The 23 specimens of peppered shiners were out of 36,719 individual fishes taken in 62 collections from 32 total localities throughout the river system.

In a subsequent re-survey of the Saline River system in 1981-1982, Stackhouse (1982) collected 65 specimens of the peppered shiner from 10 different localities. The 10 localities in the Saline River system were (1) Grant County: Saline River at St. Hwy 229, 0.8 km (0.5 mi) south of Saline County line (Sec 4, T4S, R14W); (2) Saline River at U.S. Hwy 270, 0.8 km (0.5 mi) west of Prattville (Sec 10, T5S, R15W); (3) Saline River at St. Hwy 46, 4 km (2.5 mi) NE of Leola, AR (Sec. 8, T6W, R14W); (4) Saline River at gravel road, 6.4 km (4 mi) SW of Herbine (Sec. 6, T10S, R9W); (5) Bradley County: Saline River at St. Hwy 15, 8 km (5 mi) north of Warren (Sec 3, T12S, R9W);

(6) Saline River at St. Hwy 4, 4.8 km (3 mi) east of Warren (Sec. 3, T12S, R9W); (7) Saline River, 3.2 km (2 mi) downstream from St. Hwy. 4 (Sec. 10, T13S, R9W); (8) Saline River at end of county road south of St. Hwy 4 (Sec 15, T13S, R9W); (9) Saline River, 1.6 km (1 mi) east of St. Hwy 189 on gravel road 11.2 km (7 mi) north of Johnsville (Sec. 26, T14S, R9W); and (10) Saline River at end of county road, 9.6 km (6 mi) south of Johnsville (Sec. 26, T16S, R9W). Stackhouse (1982) took 27,836 individual fish specimens in 115 collections from 50 different localities throughout the Saline River system.

In this survey, 11 specimens of the peppered shiner were taken in 15 collections from the Saline River system during 1999-2001 (Table 1). Collections made from 1999-2001 from the Saline River by HWR re-established the peppered shiner as a widespread, although rare fish species in the Saline River. The peppered shiner was found at 3 localities in the system during the present study: (1) Grant County: Saline River at St. Hwy. 229; (2) Drew County: Saline River at end of St. Hwy. 172; and (3) Bradley County: Saline River at end of county road, 9.6 km (6 mi) south of Johnsville. A total of 11 specimens was collected from the Saline River during this most recent survey (Table 1).

Kiamichi River System, Oklahoma.—Pigg and Hill (1974) surveyed the fishes of the Kiamichi River system from 1972-1973 and included 141 sampling stations. In addition, data from other ichthyologists were also used in the study. These collections were made prior to the construction of the Hugo Reservoir on the Kiamichi River. Echelle and Schnell (1976) performed a factor analysis of species associations among fishes of the Kiamichi River, but did not include the peppered shiner in their factor analysis. They did mention that *N. perpallidus* was uncommon and found at only 4 localities.

In this survey, 15 collections were made in the upper Kiamichi River system; however, no specimens of the peppered shiner were taken (Table 1).

Mountain Fork River System, Arkansas and Oklahoma.—George A. Moore, noted ichthyologist from Oklahoma State University, collected the first specimen of the peppered shiner from Oklahoma from the Mountain Fork River below a dam on the river in Beaver's Bend State park (Moore 1973) on 6 June 1947. He was able to collect only a single specimen (Moore 1948).

Reeves (1953) collected 25 specimens from the entire Little River system for his doctoral dissertation. Later, Finnell et al. (1956) reported specimens from 2 localities in their survey of the fishery resources of the Little River system. The 2 localities were (1) Cutoff pool near Mountain Fork mouth (Sec 10, T7S, R26E) and (2) Mountain Fork River near the mouth (Sec. 10, T7S, R26E). The peppered shiner was reported to comprise 0.3 % of the 2,097 specimens collected in the lower reaches of the Mountain Fork River drainage.

Wagner et al. (1987) reported the last collection of the peppered shiner in the Mountain Fork River system in 196, before impoundment of the Broken Bow Reservoir in 196. Historically, the peppered shiner has never been taken above the existing reservoir.

No specimens of the peppered shiner were collected in the Mountain Fork River in the current survey although 10 collections were made in the upper river system (Table 1).

Other Oklahoma Areas Inhabited.—In addition to the river systems discussed above which were all sampled during the present survey, the peppered shiner has been taken from the Glover River (Wagner, et al. 1987) in Oklahoma. The Glover River is part of the Little River drainage. Previously, Taylor and Wade (1972) had not collected the peppered shiner in their pre-impoundment survey of the Glover River.

A single collection of 3 specimens of the peppered shiner was made from McGee Creek, a small upland stream with large pools and very small riffles in the Muddy Boggy River system, located near Lane, Atoka County, Oklahoma by Pigg (1977). The distribution of the peppered shiner was thus extended west of the Kiamichi River system in Oklahoma to the Muddy Boggy River system.

Historic Conservation Status.—The state of Arkansas presently has no official state list of threatened or endangered wildlife or plants. Instead, protection is afforded primarily to federally threatened species by the Arkansas Game and Fish Commission.

Both Buchanan (1974) and Robison (1974) in independent assessments of the threatened fishes of Arkansas included the peppered shiner in their publications. Buchanan (1974) gave the peppered shiner a status of rare as did Robison (1974). Robison and Buchanan (1988) listed *Notropis perpallidus* as "threatened" in their discussion of rare and endangered fishes in Arkansas.

In Oklahoma, Robison et al. (1974) listed the peppered shiner as "rare" with little comment. The Rare and Endangered Species of Oklahoma Committee (1975) pronounced the peppered shiner "rare' in Oklahoma with a "Rare-2" status, which meant the species may be quite abundant where it occurs, but is known from only a few localities or in a restricted habitat within Oklahoma.

Habitat loss is one of the greatest causes of the declines in populations of native fishes in North America (Williams et al 1989). Widespread reservoir construction and declines in water quality have severely altered most of North America's clean free-flowing riverine habitat (Benke 1990). In their review of the tolerances and degradation in water quality and habitat, Jester et al. (1992) considered the peppered shiner as "Intolerant."

Recently, the Southeastern Fishes Council Technical Advisory Committee (SFCTA), consisting of 12 ichthyologists from throughout the South reviewed the diversity, distribution and conservation status of the freshwater fishes of the southern United States, which includes over 600 species of fishes. In the resulting publication, Warren et al. (2000) listed the peppered ner as "Vulnerable" where vulnerable meant "a species or species that may become endangered or threatened by atively minor disturbances to its habitat or that deserve careful nitoring of its distribution and abundance in the continental ters of the United States."

Warren et al. (2000) found that 28 percent of the total fish una of over 600 species in the southern United States (which included both Arkansas and eastern Oklahoma) is jeopardized to some extent, of which 6% are endangered, 7% are threatened, and 15% are regarded as vulnerable. The SFCTA Committee reports that this represents a 75% increase since 1989 in jeopardized southern fishes and an incredible 125% increase in the past 20 years. Disappointingly, the trend for fishes in the southern United States is clear; jeopardized fishes are successively being moved from the vulnerable category to that of imminent threat of extinction (Warren et al. 2000).

Conclusions

Present Conservation Status.—Eighty-one collections of fishes were made during this study within the historical distribution of the peppered shiner in Arkansas and Oklahoma in an attempt to document the present conservation status of the peppered shiner. From these 81 collections in Oklahoma and Arkansas, only 17 specimens of peppered shiners were captured, all in Arkansas (Table 1). No specimens of the peppered shiner were collected in Oklahoma. After careful review of all of the major museum holdings of the peppered shiner available, 2 years of intensive field work collecting peppered shiners, review of all pertinent literature, and discussions with virtually all of the major collectors of peppered shiners in Arkansas, it is apparent that the peppered shiner has declined in abundance throughout its historical range in Arkansas.

Table 2 provides a quick view of the decline in abundance of the peppered shiner in Arkansas and Oklahoma by decade. While certainly not definitive, Table 2 shows the peppered shiner seeming to decline in the decade of the 1980s and continuing into the 1990s to the present study. The large number of peppered shiners collected by workers in the 1970s is illustrative of the "golden decade" of ichthyological collecting in both Arkansas and Oklahoma as hundreds of collections of fishes were made. Many of these collections of fishes were made as part of a variety of Master's theses on numerous Arkansas River systems by graduate students (e.g. Fruge 1971, Harris 1977, J. E. Herrock 986, L. W. Herrock 1986) and Jimmie Pigg's 1977 collections n Oklahoma.

A closer inspection of the 791 museum specimens of the eppered shiner grouped by river system reveals that 40.96 % 24 individuals) of the specimens were collected from a single ver, the Little River (Table 3). The next most abundant river stem in producing peppered shiner was the Saline River stem (15.80 %), followed by the Little Missouri River system 3.65 %) and the upper Ouachita River (10.62%). The Kiamichi River produced the fewest number of specimens (3.29 %) (Table 3).

Table 2. Number of peppered shiners collected by year.

Years	No. Peppered Shiners
1940-1949	5
1950-1959	31
1960-1969	36
1970-1979	346
1980-1989	92
1990-1999	18
2000-2001	17
Totals	545

Overall there seems to be a decline in the populations of the peppered shiner in both Arkansas and Oklahoma based on the data gathered from this study. This apparent overall reduction in range and abundance in Arkansas and Oklahoma necessitates a re-evaluation of the conservation status of the peppered shiner.

Reasons for this decline seem to be multiple and complex. Destruction and modification of habitat from impoundments with concomitant cold water release may be a part of the problem for the peppered shiner. The peppered shiner has disappeared from the lower Caddo, which was impounded as DeGray Lake. Northeast Louisiana University students collected peppered shiners from the Caddo River below the dam in the 1970's prior to the closure of the dam on DeGray Lake. The peppered shiner has never been collected since then in the Caddo River system. Cold water releases impacts areas many kilometers downstream from reservoirs. Reservoirs also effectively eliminate migration by obligate stream fishes from one tributary to another, precluding natural colonization of potential suitable streams. Increases in turbidity and siltation have also occurred in the upland streams inhabited by the peppered shiner as poor land practices such as road building, farming, clearing of land for pasture, clearcutting, destruction of riparian buffer strips and other human perturbations continue in these watersheds. Other possible reasons for decline of the peppered shiner include gravel removal operations in a number of Arkansas streams (Filipek and Oliver 1994) and nutrient enrichment from the enormous increase in poultry and swine operations and human population increases.

One factor that figures into the conservation status of the peppered shiner is the possibility that this cyprinid species may have never been abundant, even in earlier years (see Table 2). Note that from 1940-1969, only 72 specimens of the peppered shiner were collected despite the work of a number of eminent active ichthyologists like G. A. Moore (OSU), C. L. Hubbs and John Black (University of Michigan), and others.

During these 2 years HWR was able to document the continued presence of the peppered shiner in only 2 of the river

Table 3. Collections of the peppered shiner by river system.

River System	No. Peppered Shiners	Percentage of Total Peppered Shiners	Collections
Ouachita River	84	10.62	151
Caddo River	62	7.84	141
Little Missouri River	108	13.65	126
Saline River	125	15.80	192
Little River	324	40.96	37
Glover River	33	4.17	7
Mountain Fork River	29	3.67	15
Kiamichi River	26	3.29	76
Total	791	100.0	745

systems in Arkansas from which it was collected historically; the Ouachita mainstem and the Saline River systems (Table 1). No specimens of the peppered shiner were collected from the Little Missouri River, Caddo River, Kiamichi River, Mountain Fork River, or Glover River, although 52 collections were made in those river systems.

Status Recommendation.—Thus, after carefully reviewing the collection records of the peppered shiner from the University of Louisiana at Monroe, the University of Oklahoma, Oklahoma State University, Tulane University, Arkansas State University, and 2 years of field work, the peppered shiner is not herein recommended for official federally threatened status at this time. Rather, this small, cyprinid species should be accorded a status of "Vulnerable" as used by Warren et al. (2000) and a program be initiated to monitor its continued existence. Small population size and low densities make it imperative that a careful watch be maintained on this species in the future.

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Development of *in vitro* Regeneration Protocols for Arkansas Rice Varieties (*Oryza sativa L.*)

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Abstract.—Efficient regeneration of *in vitro* cultures of *Oryza sativa L.* is essential for successful manipulation of recombinant DNA technologies. Arkansas rice varieties perform better on modified Gamborg (B5) media, than on Murashige and Skoog (MS) or N6 media, which are more frequently reported in the literature. While 2,4-dichlorophenoxyacetic acid (2,4-D) is commonly used for regeneration treatments of rice, Picloram (Pic) provides a possible alternative as a synthetic auxin source. Although regeneration frequency appears low, complete regeneration (whole plantlets) was faster and development of the shoot was superior on picloram treatments as compared to 2,4-D.

Screening 5 Arkansas varieties for regeneration efficiency on 3 treatments, 2 drawn from the literature and 1 based on Pic, shows distinct rankings for successful identification of totipotent established lines. LaGrue, important for both production and breeding purposes, shows the highest ranking for successful regeneration as well as for uniform response of individual seedling lines across regeneration treatments. Other varieties proved unsuitable for transformation work due to lack of regeneration response and/or non-uniform response to regeneration treatments. Identifying these parameters will facilitate the ultimate transfer of recombinant DNA into tropical *Japonica* rice types grown in Arkansas for trait improvement or gene expression studies.

Key words:— in vitro, Oryza sativa L., DNA, Arkansas rice, Gamborg (B5), Murashige and Skoog (MS), N6 media, 2,4-dichlorophenoxyacetic acid, Picloram (Pic), synthetic auxin, *Japonica.*

Introduction

Transformation technologies currently use MS (Murashige and Skoog 1962) or N6 (Chu 1978) media treatments (Rashid et al. *1*996, Toki 1997) with several choices of plant growth regulators (PGRs) such as 6-benzylaminopurine (BA) + α-naphthaleneacetic acid (NAA) or kinetin (Kin) + NAA. While the use of picloram as an auxin is not reported for rice regeneration, it has been successfully used for other monocots (Phillips and Luteyn 1983). Rice varieties previously studied vary widely, and includes *O. sativa* ssp. *Japonica* as well as ssp. *Indica* and *Javonica*. Our interest is with the tropical *Japonica* types important for Arkansas rice production. High regeneration efficiency is necessary for successful transformation of rice (Toki 1997). Identification of highly efficient regenerator varieties could provide uniformity in such systems. While 2,4-D is commonly used for short term culture and regeneration of rice, long term exposure may ultimately suppress regeneration potential and make it difficult to utilize known regenerator lines, thus requiring longer culture time. Picloram, which can be used for other monocots, may provide a suitable substitute for 2,4-D and avoid this problem (Dode et al. 2000). Technology directly applicable to local varieties is critical for ultimate success in improving rice yield in Arkansas.

Material and Methods

Five rice varieties were provided by the University of Arkansas Rice Research and Education Center (RREC) at Stuttgart, Arkansas, courtesy of Dr. Karen Moldenhauer: Drew, Gulfmont, Katy, LaGrue and Mars. All varieties were tested for callus induction. De-hulled rice seed were surface sterilized by exposure to 50% Clorox and then plated onto initiation media as the standard starting material for all experiments (Fig. 1). Cultures were placed in dark incubators at 28°C for initial growth. Calli were incubated in constant light conditions only when transferred to regeneration treatments. Calli 1-60 days old were grown in 60 x 15 mm Petri dishes, all regeneration treatments of larger tissues were grown in 100 x 15 mm Petri dishes. Preliminary experiments were conducted to compare modified Gamborg B5 media (Dunstan and Short 1977) with MS and N6 media as the basal salts and nutrients formulation for rice culture.

Rice calli were grown for 60 days on initiation media using either 2,4-D or Pic (2.2 mg/l and 5 mg/l, respectively). Residual tissues from the seed were removed after 30 days, and calli were transferred at 60 days to regeneration treatments to allow for accumulation of biomass. Regeneration treatments labeled

Fig. 1. Callus formation and development on three different basal salts using Katy: A) N6, B) B5, and C) MS.

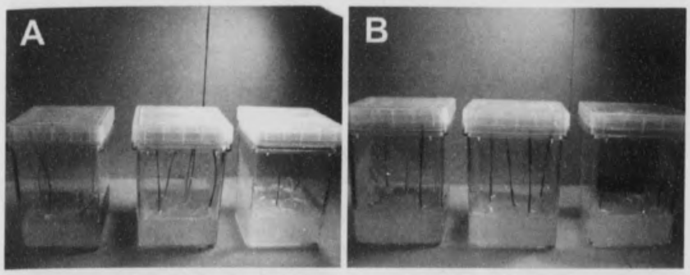

Fig. 2. Seed germination and plant development of two varieties on three different basal salts. A) Katy: MS, B5, and N6 (left to right). B) Mars: MS, B5 and N6 (left to right).

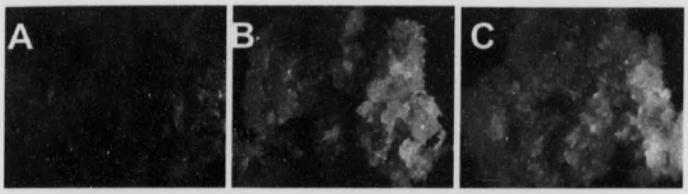

Fig. 3. Callus morphology after 60 days incubation varied on basal salts treatments: A) N6, B) MS, and C) B5.

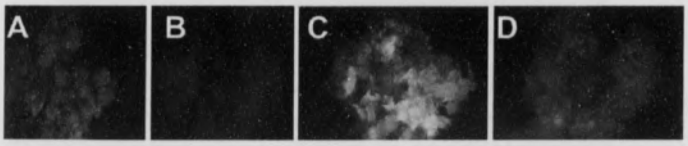

Fig. 4. Callus morphology after 60 days incubation varied on 2,4-D (A, B), and Picloram (C, D) treatments.

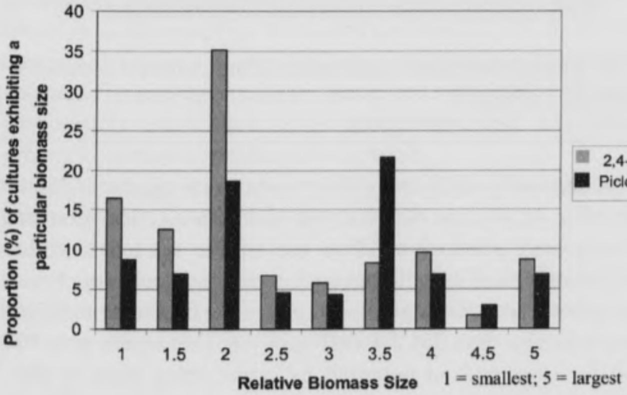

Fig. 5. Biomass comparison using 2,4-D and Picloram. A relative scale using 1 = smallest biomass and 5 = largest biomass was utilized to categorize distribution of cultures within treatments.

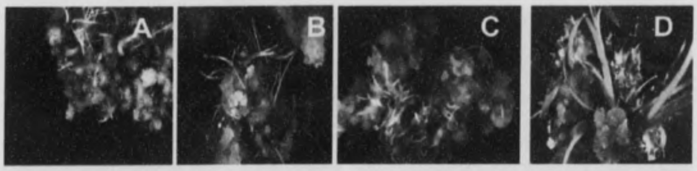

Fig. 6. Somatic embryogenesis response using 2,4-D ranged from embryogenic callus formation with incomplete formation of shoot apices (A, B, C) to whole plant formation (D).

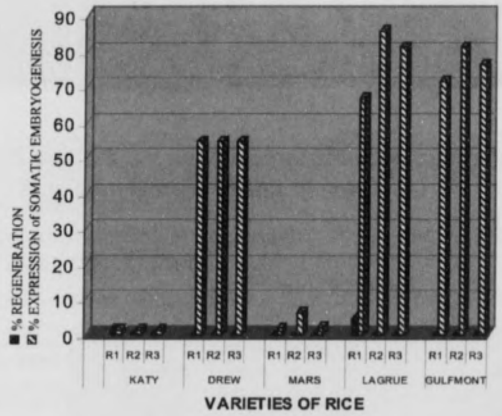

Fig. 7. Induction of somatic embryogenesis and whole plant regeneration of 5 rice varieties on R1, R2, and R3 PGR treatments following callus proliferation on 2,4-D.

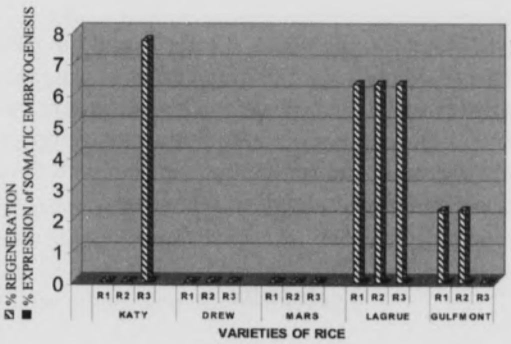

Fig. 8. Induction of somatic embryogenesis and whole plant regeneration of 5 rice varieties on R1, R2, and R3 PGR treatments following callus proliferation on Picloram.

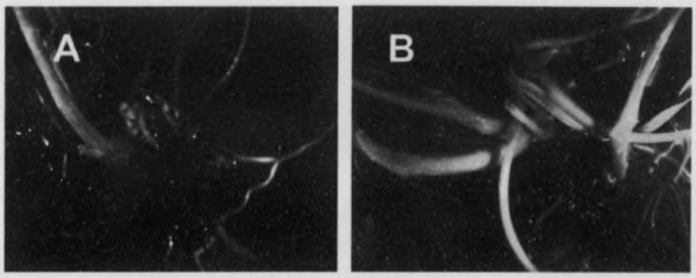

Fig. 9. Somatic embryogenesis response using Picloram leading directly to whole plant formation (A, B).

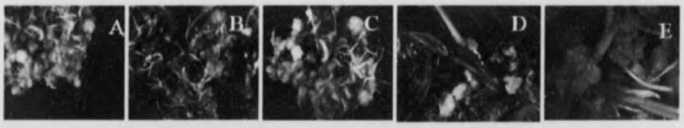

Fig. 10. Embryogenic callus formation on R1, R2, and R3 PGR treatments (A, B, C). Complete plant regeneration on R1 PGR treatment (D, E).

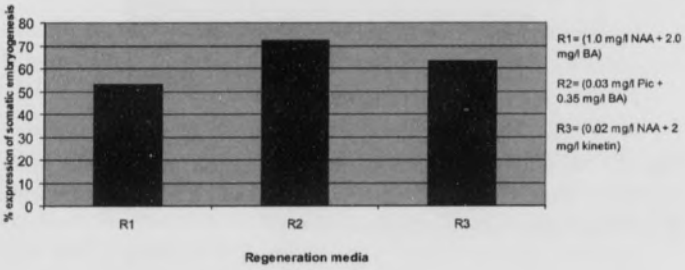

Fig. 11. Comparison of R1, R2, and R3 PGR treatments for expression of somatic embryogenesis.

R1 (1.0 mg/l NAA + 2.0 mg/l BA), R2 (0.03 mg/l Pic + 0.35 mg /l BA), and R3 (0.02 mg/l NAA + 2 mg/l kinetin) were tested for 2 months. Observations were made weekly, data were collected every 30 days. Lines were scored for frequency of induction of somatic embryogenesis and for the frequency of well developed plantlets with both apical and radicle meristems.

Results

Initial studies indicated a benefit of comparing basal salts formulations. Early experiments indicated that growth potential of rice *in vitro* cultures could be optimized by using modified Gamborg B5 media, in comparison to MS and N6 media (Fig. 2).

Fig. 12. Regeneration of albino shoots from Gulfmont (A) and LaGrue (B) cultures.

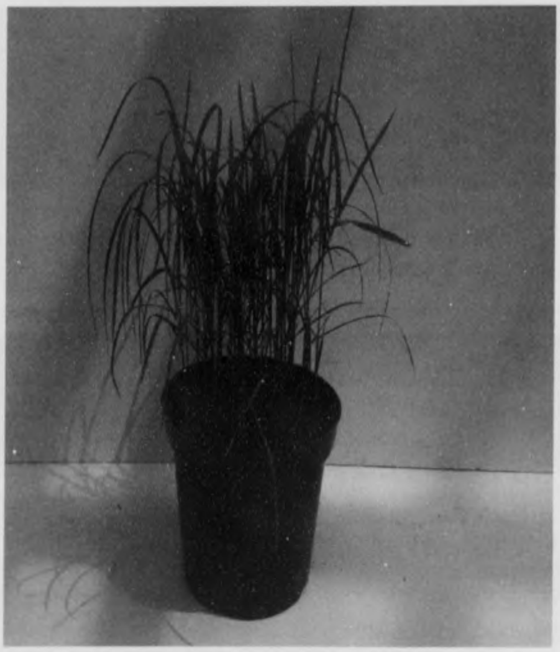

Fig. 13. Established plant regenerated from cultured somatic embryo of LaGrue.

Dramatically different callus types grew on the induction media (Fig. 3). The use of 2,4-D resulted in small, more compact, and nodulated phenotypes. The use of Pic tended to induce prolific rooting and only higher concentrations gave more typical callus phenotypes. Over time, Pic generally produced calli with greater biomass than did 2,4-D (Fig. 4, 5). These calli were then tested for regeneration potential by transferring them to the 3 regeneration treatments.

When the rice varieties were scored for regeneration potential, it was clear that the PGRs used during callus formation are important. Calli originating on 2,4-D showed high levels of embryogenic induction (Fig. 6, Table 1), up to 89% for LaGrue (Fig. 7).The 5 varieties, ranked from highest to lowest, were LaGrue, Gulfmont, Drew, Mars, and Katy (Table 1). Whole

Table 1. Number of individual seedling-derived rice callus lines responding to different regeneration treatments (R1, R2, or R3 PGR treatments). Sample size of approximately 50 explants per variety per regeneration treatment. R1 (1.0 mg/l NAA + 2.0 mg/l BA), R2 (0.03 mg/l Pic + 0.35 mg /l BA), and R3 (0.02 mg /l NAA + 2 mg/l kinetin).

	--------------------Varieties				
Response to three varying regeneration treatments	**Gulfmont**	**LaGrue**	**Drew**	**Katy**	**Mars**
R1, R2, and R3	**14**	**12**	**4**	**0**	**4**
R1 only	**0**	**0**	**1**	**0**	**1**
R2 only	**1**	**0**	**1**	**2**	**1**
R3 only	**0**	**1**	**0**	**1**	**1**
R1 and R2	**1**	**1**	**0**	**3**	**0**
R2 and R3	**1**	**2**	**3**	**3**	**0**
R1 and R3	**0**	**0**	**0**	**0**	**0**

plant regeneration at 60 days on 2,4-D was limited to LaGrue and Katy, and was low (4.8% for LaGrue and 1.4% for Katy; Fig. 7). Calli induced on Pic showed higher whole plant regeneration (up to 7.7%; Fig. 8). Although whole plant regeneration was faster using Pic (30 days compared to 60 days on 2,4-D), sustained somatic embryogenesis was impossible because the embryogenic callus did not proliferate and callus quality declined (Fig. 9). Complete plant regeneration occurred only on R1 from 2,4-D-derived cultures at 60 days (Fig. 10D). All treatments supported expression of the embryogenic callus; however, R2 gave the highest frequency of somatic embryo development, while R1 showed the lowest frequency of embryogenic callus expression (53.1% on R1, 72.15% on R2, and 63.29% on R3; Fig. 10, 11). Whole plant regeneration within 30-60 days after induction sometimes gave abnormal phenotypes. Albino regenerants were found in Gulfmont (Fig. 12A) and LaGrue (Fig. 12B) and are of concern since such events will negatively impact transformation success. Despite this problem, numerous regenerated plants were successfully established in the greenhouse (Fig. 13) and were grown to maturity, yielding fertile seeds.

In a preliminary experiment we compared regeneration potentials of LaGrue with Nipponbare and Taipei 309, which are the two most often-cited varieties for rice regeneration and transformation (Toki 1997, Dong et al. 1996). Our initial results suggested that LaGrue was comparable to Nipponbare, but Taipei 309 was a better regeneration variety.

Conclusions

B5 basal media gave consistently better responses than either MS or N6.

Picloram produced higher biomass during callus induction events.

Although 2,4-D showed the better induction of somatic embryogenesis, complete regeneration events were fewer than with picloram treatments.

Picloram tended to regenerate whole plants faster (30 days vs. 60 days) than did 2,4-D.

LaGrue and Gulfmont were superior in regeneration response compared to the other varieties (Drew, Katy, and Mars) tested.

Inconsistent regeneration responses among callus lines of some varieties (Katy and Mars) make these varieties undesirable for transformation of unscreened lines.

Future Goals

Optimization of *in vitro* technology for rice for such variables as basal salt formulations, PGRs, and identification of efficient regenerator varieties will facilitate successful transformation protocols. Improving these parameters will facilitate the use of recombinant DNA technology in rice genetics. These improvements will ultimately make these tools useful for studying RNAi genetic manipulation techniques to target inhibition of lipoxygenase gene(s) expressed specifically in the rice grain, which will permit testing whether lipoxygenase enzyme activities lead to grain degradation during storage.

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Evaluation of Cross Pollination of *Zephyranthes* and *Habranthus* Species and Hybrids

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Abstract.—The genus *Zephyranthes* and the related genus *Habranthus* in the family Amaryllidaceae are commonly grown ornamental bulbs having new world homologies. Inter-specific and inter-generic hybridizations are complicated by the fact that some of the species are apomictic, cross incompatible, or have widely variable 2n chromosome numbers. A simple, inexpensive method of pollen storage was tested to evaluate the production of hybrid seed. Intact anthers harvested after pollen release and stored at 4°C were used for pollination. Emasculated flowers were pollinated at noon on the first or second day of anthesis. Hybrid crosses were labeled and seed collected when formed. All available parents were used in hybridizations except when species were known to have apomictic or pseudogamous seed development. Hybridization data were recorded for seedpods having successful seed set as well as those that aborted. Development of 'normal' seedpods filled with seed occurred even though viable embryos were not always formed. F_1 *Z. grandiflora* was successful 55% of the time, and Pink Trihybrid, a rather infertile seed parent, was successful at least 19% of the time. These data suggest that repeating many crosses ultimately produced a few hybrids in problematic crosses. In addition to producing seed of potentially interesting new hybrids, this study helped to identify successful seed and pollen parents for future breeding efforts. Hybrid seed was sown to test viability of the progeny.

Key words.— Zephyranthes, Habranthus, Amaryllidaceae, ornamental bulbs, hybridizations, pollination.

Introduction

The genera *Zephyranthes* and *Habranthus* include many distinct species commonly called rainlilies or surprise lilies due to their habit of episodic flowering after seasonal rainfall. Species belonging to these 2 genera are native to diverse areas of the new world including Argentina, the Caribbean, Mexico and North America. The North American species range from Florida and the Carolinas to the mountains of western New Mexico and comprise perhaps the most cold hardy and drought tolerant members of these genera. The assessment of the family using cladistical analysis indicates that these two genera only have new world homologies (Meerow et al. 1999).

These plants are perennial geophytes that reproduce either asexually (i.e. via offsets or twin scaling) or by seed. Efficient asexual reproduction of most rainlilies makes preservation of selected clones easy. Production of unique phenotypes via seed, however is complicated by the fact that some species are apomictic or pseudogamous (Gupta et al. 1998). Such species reproduce the maternal phenotype faithfully without variation. Chromosome number within *Zephyranthes* range from 2n = 18-96, obviously a concern for crosses among species (Raina and Khoshoo 1971).

Flowers of *Zephyranthes* are usually solitary, with 6

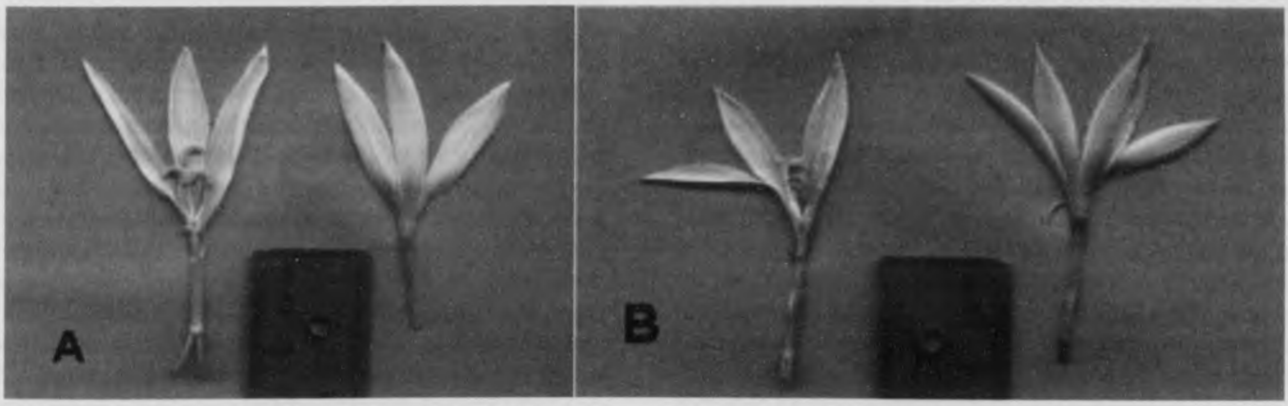

Fig. 1. Dissection of 2 flowers showing arrangement of stigmal surfaces and anthers. A: *Habranthus* flower showing crescent moon shaped anthers and uneven length of filaments typical of the genus. B: *Zephyranthes* flower showing even length of the filaments and more linear anthers. Photos by Joschles H (www.pacificbulbsociety.org/plosesiki/index.php/zephyranthes

Fig. 2. Comparison of seed phenotypes show extreme differences in morphology. A: Seed from *Z. species* with the '*allium*' phenotype. Seed coat is waxy, seed is plump containing lots of endosperm. Typical example would be *Z. citrina.* B: Seed from *Z. species* with the '*lillium*' phenotype. Seed is papery, flat, shriveled and contains only minimal endosperm. Typical example would be *Z. labuffarosea.* C: Hybrid seed often shows an intermediate phenotype. (Big Shot x *Z. citrina*) D: Hybrid seed formation with a robust intermediate phenotype (*Z.grandiflora* x Z. *primulina*) E: Seed from fertile hybrid cross.

petals having colors ranging from white to sulfur yellow to pink. The stamens are often spatially separated with either exserted pistils (i.e. approach herkogamy) or hidden pistils (i.e. reverse herkogamy). This arrangement significantly influences pollination in the wild. Flowers with hidden stigmas are self pollinated unless insect visited. Several species of *Zephyranthes*, such as *Z. jonesii* which have hidden pistils and are mildly fragrant, bloom at night and release pollen at dawn. This arrangement of stamens insures self pollination unless the corolla with attached stamens is excised before pollen release when hand pollinating. *Habranthus* species have similar flowers in colors from white to bronze yellow to pink (some have purple stripes), but the flowers generally remain funnel or trumpet-like not opening fully. *Habranthus* flowers typically have spatially separated stamens and pistils and distinct crescent shaped anthers held on filaments of uneven length (Fig. 1). Seedpods are trilobed capsules and contain D-shaped seed that usually develop rapidly. Species in both genera produce 2 distinct types of seed. Some species produce seed that is flat and papery with a phenotype similar to *Lilium* while other species produce seed that is plump and somewhat waxy similar to *Allium* seed (Fig. 2). Intermediate seed phenotypes are observed in hybrids and such observations are useful in evaluating the result of cross pollinations.

Generally the leaves, bulbs and roots of these plants are toxic. Although the presence of alkaloids has been documented in several species (Kojima et al. 1997), and other species have been evaluated for medicinal value, the primary interest in this breeding program is for the ornamental value of the flowers. Wild collected naturally occurring hybrids such as *Z. sp.* Tenexico have apricot colored flowers and commercially available cultivars now extend the color range from orange to salmon pink and red (Fellers 1996). Although many of the species have small ephemeral flowers some hybrids produce larger flowers that will remain open for up to 3 days. The potential for new cultivars via cross pollination is limited because of some reproductive barriers in these plants. A summary of the barriers to cross pollination among *Zephyranthes* species and hybrids and with the species of *Habranthus* is as follows:

1. Plant structural morphologies: These include length of the floral tube, spatial arrangement of the stamens and length of the pistils.
2. Chromosome number or ploidy level: A wide variety of 2n chromosome numbers is a deterrent to crosses.
3. Pollen production: Certain species and hybrids produce limited amounts of pollen.
4. Self or Cross incompatibility: Apparent sterility might actually be incompatibility.
5. Apomixis and/or Pseudogamy: These species often produce prolific seed which reproduces the maternal phenotype.
6. Flowering season: Some species are once-flowering; others repeat flowering throughout the growing season.
7. Receptivity of the stigma: Some species remain open for more than one day and may actually be receptive on day 2 of anthesis.

While all barriers are noted, the ultimate limiting factors governing pollinations in this study were availability of flowers and pollen during the study period. All crosses were attempted with the exception of apomictic species. Apomictic species were never used as maternal parents; as paternal parents these are good pollen donors which were used freely in crosses. Certain reciprocal crosses are thus impossible. The objectives of this study are as follows:

1. To evaluate a simple pollen storage method that will potentially allow greater flexibility of crossing non-synchronous blooming species.
2. To make as many pollinations as possible and to evaluate the compatibility of inter-specific crosses as well as a few inter-generic crosses.

Table 1. List of species and hybrids used for hybridization.

Species	Color	Hybrids	Color
Zephyranthes candida	white	Apricot Queen	apricot
Z.chlorosolen	white	Benidama	pink
Z. citrina	dark yellow	Best Pink Trihybrid	pink blend
Z.clintae	pink	Big Shot	cream
Z.drummondii	white	Yellow Big Shot	yellow
Z.grandiflora	pink	Pink Big Shot	pink
Z.insularum	white	Orange Big Shot	orange
Z.flavidissumus	yellow	Salmon Big Shot	salmon
Z.jonesii	cream	C C Moon	white
Z.labuffarosea	blush pink	Dark Pink Spider	pink
Z.lindleyana	variable pink	El Cielo	blush pink
Z. longifolia	neon yellow	Fireball	orange/yellow
Z.macrosiphon	pink	Cherry Fireball	cherry red striped
Z.morrisclintae	blush pink	Goliath	pink
Z.primulina	lemon yellow	Ivory Star	white
Z.pulchella	dark yellow	Lemon Pinwheel	light yellow
Z. reginae	light yellow	Norma Pearl	white
Z. simpsonii	white	Orange Citrina	orange
Z.smallii	yellow	Hybrid El Cielo	pink and white
Z.traubii	cream	Prairie Sunset	salmon
Habranthus texensis	bronze yellow	Quad Pink	pink
H. texensis var. roseus	pink w/purple	Quad Orange	light orange
		Sunset Strain	yellow w/red stripes
		Tall Pink	pink
		Tenexico	salmon
		Xzb-H2	white

Material and Methods

A collection of *Zephyranthes* and *Habranthus* species and hybrids is maintained at the Arkansas Biosciences Institute, Arkansas State University, State University, AR 72467. These plants and their seedlings (Table 1) were made available for this project from September to December 2005. It should be noted that maximum flowering of these species normally occurs from April until October, so this project was conducted during the time when flowering normally decreases. The number of crosses attempted strictly depended on the natural blooming capacity of the species and hybrids and was not under the control of the researcher. Successful storage of pollen was critical because flowering flushes normally decrease at the end of the season. The diversity and availability of pollen was expected to be a limiting factor.

Flowers were emasculated using fine tipped forceps at the time of pollination. Entire anthers were collected intact into 25 ml brown plastic sample vials labeled and stored in sealed plastic containers (Rubbermaid ™) in a cold room at 4-6 °C and kept for the duration of the experiment. Details of this storage technique were provided by Mr. John D. Fellers, Rainlily Breeder/Authority, Auburn, Alabama (personal communication 2005). Flowers were pollinated close to noon which is the average time for anthesis and pollen shed. There are extreme exceptions to this; *Z.traubii,* San Carlos and *Z.labuffarosea* which are nocturnal flowering species. Such species are presumed to be insect pollinated and are fragrant. It was not possible within the scope of this project to pollinate at night or dawn. When these plants were used as female parents, emasculation was done the previous day when possible. After pollination the flower stalks were labeled, dated and left for seed formation. Unsuccessful crosses were often noticed within 3-5 days, but some crosses took as long as 1 week or more before aborting. Successful seed formation took approximately 3 weeks depending on the female parent (Fig. 3). El Cielo pods typically took 4-5 weeks to mature properly. Seedpod formation is not always indicative of normal, viable seed. Seed was sown to check for viability. Rainlily seed (of both *Zephyranthes* and *Habranthus*) is generally sown soon after harvest. Seed of most of these species remained viable for only a short time.

Results and Discussion

The results of crosses made in Fall 2005 are given in Table 2 and Fig. 4. In total, 215 crosses were attempted; of

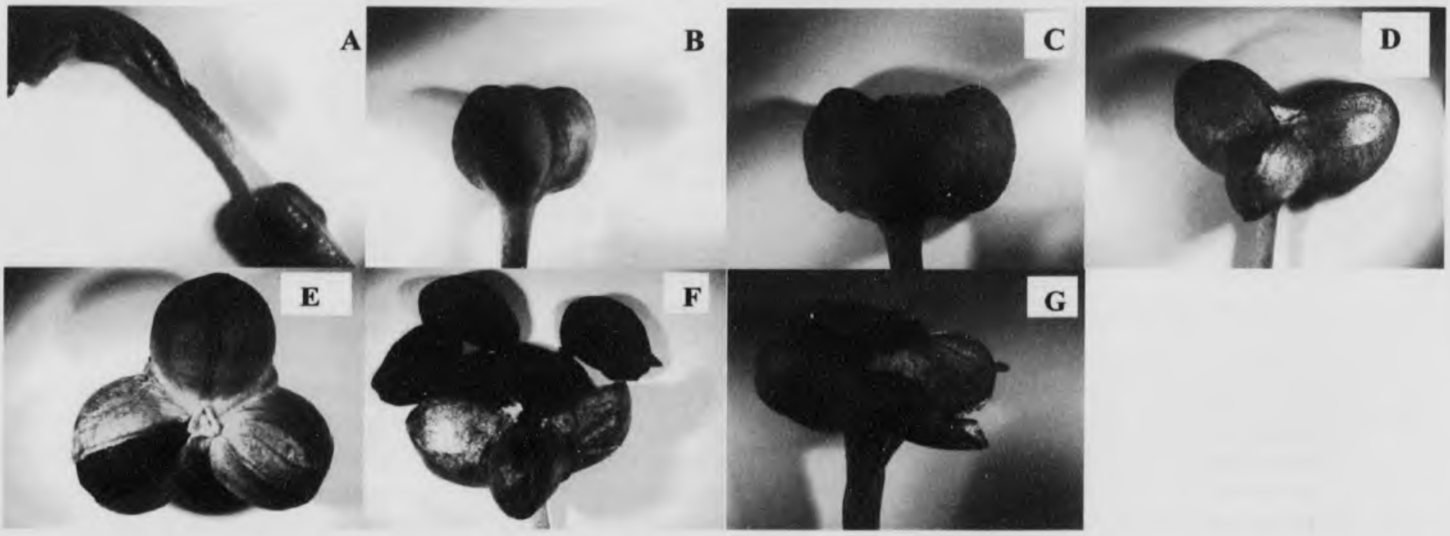

Fig. 3. Development of typical *Zephyranthes* seed from flower to shattering. A: ovary 3 days post pollination; B: developing pod; C: maturing pod two weeks after pollination; D: fully mature pod; E: dehiscence of pod; F: collectable seed; G: empty capsule [all images *Z. citrina*].

these, only 87 or about 41% set seed capsules. These were then harvested. The remainder, about 59% of the crosses aborted. Stored pollen was successful 41% of the time. This provided the breeder with pollen representing a wider variety of genotypes than fresh pollen available on a given day and a greater choice of male parents. Species that are known to be apomictic by experience or via citations in the literature were excluded from use as female parents. These species and/or hybrids include, *Z. primulina, katherinae, macrosiphon, citrina,* Sunset Strain, *jonesii, longifolia,* Ivory Star and *pulchella* (Howard 1996). Such species were all used as pollen parents. Using these parents as females is always successful but never produces hybrid seed.

The choice of female parents other than the apomictic species mentioned above depended strictly on weather and flowering conditions. During this project, episodic flowering events occurred only twice. Some days there were no flowers available, while at least two prolific episodes of synchronous flowering occurred. During these episodes as many as 40-50 flowers were available in a 7 day period. For the remaining bloom season, crosses were accomplished with whatever intermittent flowering occurred. Lack of predictable flowering made experimental design difficult. Considering the relatively small sample sizes, looking at the frequency of successful seed production offers some insight. Successful pollen parents include both species and hybrids; notably, F_1 *Z. grandiflora* and a *Z. traubii* hybrid called Yellow Big Shot (Fig. 4). Pollen sources are always more diverse since all the yellow apomictic parents can be used and are responsible for production of orange and red hued hybrids. When seed parents were evaluated, it is notable that although F_1 *Z. grandiflora* is considered to have an unusual 2n chromosome number, it was among the best of the female parents tested (Fig. 4). Trihybrid female parents [(*Z. candida* x *Z. citrina)* x *Z. macrosiphon*] were used for pollinations. These plants are sister seedlings and have similar phenotypes but are mostly self and cross sterile. The interesting genetics that they represent (cold tolerance of *Z. candida* x deep yellow color of *Z. citrina* x large pink flowers of *Z. macrosiphon*) makes them desirable as parents. Best Pink Trihybrid and Pink Trihybrid (sister seedlings) made fertile seedpods 46% and 19% of the time respectively with multiple pollen parents. This indicated remarkable fertility never seen before in these desirable parents.

For female parents that were attempted 10 or more times, the rate of fertile crosses ranged from 0-60% (Table 2). All of the hybridizations attempted are either inter-specific crosses or crosses with cultivated hybrids which are usually inter-specific crosses themselves when their ancestry is known. These data indicated that unrelated crosses have potential for success.

Among the crosses recorded in Table 2 which were attempted in extremely limited numbers, there are some exceptional responses. F_1 *Z. grandiflora* (which is a seedling code for a fertile *Z. grandiflora* hybrid seedling produced in this breeding program) was crossed with *Habranthus tubispathus* var. *texensis*, which represents an unusual inter-generic cross. The quadhybrids, crossed with Yellow Big Shot, represent at least 6 ancestral species and indicate the potential for making complex hybrids within the genus *Zephyranthes*. At least one female parent, the fertile F_1 *Z. grandiflora* hybrid was successful with 9 different pollen donors. This F_1 parent is derived from a species that is usually self and cross sterile (Kapoor and Tandon 1963). The fact that this plant will set seed with multiple parents is significant because this is the source of the largest flowers

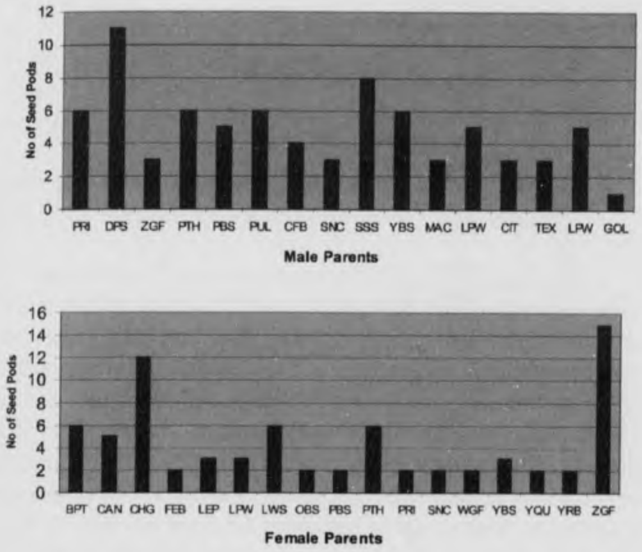

Fig. 4. Relative Fertility of Male and Female Parents. BPT: Best Pink Trihybrid; CAN: *Z. candida*; CFB: Cherry Fireball; FEB: Fireball; LPW: Lemon Pinwheel; LNQ: Lemon Quad; LYP; LWS: Lindlelyana White Star; MAC: *Z. macrosiphon*; OBS: Orange Big Shot; PBS: Pink Big Shot; PTH: Pink Try Hybrid; PRI: *Z. primulina*; DPS: Dark Pink Spider; PUL: Pulchella; SNC: San Carlos; SSS: Sun Set Strain; CIT: *Z. citrina*; LPW: Lemon Pin Wheel; WGF: White Grandiflora; YQU: Yellow Quad; ZGF: *Z. gradiflora*; TEX: *Habranthus tubispsthus* var *texanisis*; GOL: Goliath

within the *Zephyranthes*.

In this study, 44 different female parents were tested. However, only 34 or about 77% of the female parents made at least one fertile cross (Table 2). Testing large numbers of parents gave some insight into the potential for a breeding breakthrough by discovering potential maternal parents that may not be limited by apomixis. In other plant genera there is evidence that apomixis is heritable. In *Zephyranthes* some cultivars, such as Sunset Strain carry this trait. The identification of breeding lines that reliably produce hybrid progeny is necessary to make progress in backcrosses or F_2 crosses. These data identify distinct genetic lines that might be pursued which include 1) Hybrid F_1 *Z. grandiflora* which are fertile, 2) Trihybrid (partially) fertile lines, 3) *Z.traubii* hybrids (these include all of the yellow and pink Big Shot hybrids, 4) Fireball progeny, 5) *Z. labuffarosea* hybrids and finally 6) *Z. lindleyana* lines. Phenotypic traits of interest arising from these crosses include: ruffled petals, bronze foliage, fragrance, and colors of varying intensity (ranging from apricot, salmon, orange, cherry red, orange red and purple) stripes, picotees, bicolors as well as flowers that remain open for as long as 3 days.

Conclusions

The use of stored pollen resulted in seed formation on some recalcitrant crosses (e.g. the trihybrids) and suggests that a high number of pollinations and the use of many pollen parents may help to overcome some barriers to hybridization.

These data identify suitable parents for future crosses. Female parents that produce hybrid progeny predictably (and that are ultimately not apomictic) are needed to advance the generations and, in particular, to permit the evaluation of predicted recombinants in the F_2 and beyond.

Breeding within the *Zephyranthes* and between Z. species and *Habranthus* species may remain problematic due to obvious difficulties expected in making multiple backcrosses and reciprocal crosses. Identifying reliable fertile hybrids may eventually permit breeding for cold hardiness and adaptability to soil types. Disease resistance, fragrance and persistent foliage are also desirable traits for long term breeding efforts.

ACKNOWLEDGMENTS.—The authors would like to thank Dr. Carole Cramer, Executive Director of the Arkansas Biosciences Institute, for her help in preserving this collection of ornamental and medicinal plants. Special thanks to Dean Gregory C. Phillips of Arkansas State University for his support over the years. Ms. Shannon Hill has the remarkable ability to produce flowers from one-year-old seedlings, a major benefit to this breeding program. We thank her for excellent care of the stock plants. Finally thanks to Dr. Cal Shumway for encouraging this research project.

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Table 2. Summary of *Zephyranthes* hybridizations –Fall 2005.

Female Parent	# attempts	# successful	% fertile crosses	# male parents	ID of pollen parents
Apricot Queen	1	1	100	1	SBS
Best Pink Trihybrid	13	6	46	5	DPS,PRI,LIN,GRA
Big Shot	2	0	0	0	none
Candida	10	5	50	5	PTH,PBS,PQU,DPS,DPS
Cherry Fireball	1	1	100	1	DPS
Colorbreak Grand	1	1	100	1	PBS
Dark Fireball	1	1	100	1	SJP
Dark Pink Spider	10	0	0	0	none
El Cielo Pink Hybrid	4	0	0	0	none
Fireball	2	2	100	1	PUL
F1 Grandiflora	36	20	46	10	CIT, DPS,TEX,PBS,SSS,PRI,SNC, PSS, YBS
Goliath	8	3	37	3	PRI, CIT, TEX
Ivory Star	2	2	100	1	SNC
Jonz Pink	2	1	50	1	DPS
Jonz Striped Pink GRF	1	1	100	1	PUL
Labuffarosea	1	1	100	1	SSS
Lemon Pinwheel	11	3	27	2	GOL, PBS
Lemon Quad	3	3	100	1	PTH
Lilypies	1	1	100	1	YRB
Lindleyana	10	6	60	6	PRI,CFB,DPS,YBS,SSS,PTH
Lindleyana 'White Star'	1	1	100	1	PTH
Lt Pink Grandflora	1	1	100	1	PUL
Macrosiphon	2	2	100	2	CIT,LPW
Orange Big Shot	2	2	100	1	MAC
Pink Big Shot	6	3	50	3	SSS, PBS,CFB
Pink Yellow Big Shot	1	0	0	0	none
Pink Quad	1	0	0	0	none
Pink Spider B S	1	1	100	1	DPS
Pink Trihybrid	36	7	19	5	LPW, DPS,YBS, PRI, WSP
Primulina	3	2	66	2	ELS,ZGF
Pulchella	6	0	0	0	none
San Carlos	8	2	25	2	IVS, GOL
Spider Lemon Pinwheel	1	0	0	0	none
Sunset Strain	1	1	100	1	MAC
Veined Pink B S	1	1	100	1	YQU
White Grandiflora	2	2	100	1	YBS
White Star PBS	3	2	66	2	CFB
White Pinwheel	1	0	0	0	none
White Trihybrid	2	1	50	1	PUL
Yellow Big Shot	5	3	60	3	DPS,PQU,YTP
Yellow Pinwheel	1	0	0	0	none
Yellow Quad hyb.	4	2	50	2	DPS,YTP
Yellow Spider BS	1	0	0	0	none
Yellow/Red Striped BS	2	2	100	2	ZGF,LYP
TOTAL	210	93 44%			

DSF: Dark Seedling Fireball; APQ: Apricot Queen; BPT: Best Pink Trihybrid; CAN: *Z. candida*; CFB: Cherry Fireball; FEB: Fireball; IVS: Ivory Star; JLP: Jonz Labuffarosea Pink; JZP: Jonz Pink; JSPG: Jonz Striped Pink Grandiflora; LFR: Labuffarosea: LPW: Lemon Pinwheel; LNQ: Lemon Quad; LYP: Lilypies; LDY: Lindlelyana; LWS: Lindlelyana White Star; MAC: *Z.macrosiphon*; OBS: Orange Big Shot; PBS: Pink Big Shot; PKG: Pink Grandiflora; PSBS: Pink Spider Big Shot; PTH: Pink Try Hybrid; SBS: Salmon Big Shot; PRI: *Z.primulina*; DPS: Dark Pink Spider; PUL: Pulchella; SNC: San Carlos; SSS: Sun Set Strain; CIT: *Z.citrina*; LPW: Lemon Pin Wheel; VBS: Veined Big Shot; WGF: White Grandiflora; WTH: White Trihybrid; ZGF: *Z. grandiflora*; PSS: Prairie Sun Set; TEX: *Habranthus Tubispsthus* var *texansis*; ELS: El Cielo Star; YQU: Yellow Quad; YRB: Yellow/Red Striped Big Shot: WSP: White Star Pink Big Shot; YTP: Yellow Throat Pink Big Shot.

Geographic Distribution and Harvest Dynamics of the Eastern Spotted Skunk in Arkansas

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Abstract.—The eastern spotted skunk (*Spilogale putorius*) is a small carnivore found across much of the central and southeastern United States, and while once common, this species has become rare in most of its range. We used harvest records collected by the Arkansas Game and Fish Commission from 1941-2004 to examine historic and current distribution and long-term harvest dynamics of this species in Arkansas. Eastern spotted skunks have historically been most common in the Ozarks and the Ouachitas though the species appears to have been present, but uncommon, in the Gulf Coastal Plain and in some counties in southeastern Arkansas near the Mississippi River. Annual harvests declined precipitously during the 1940s and 1950s, from >1,800 animals in 1942 to <10% of that number by 1958. During the early 1960s and especially during the late 1970s, there were multi-year increases in the harvest that co-occurred with increases in pelt price. However, across the broader period used for data analyses, pelt price alone was a poor predictor of harvest. Harvest was best predicted by the number of furbuyers in the state, which likely correlates with the number of trappers. By the mid-1980s annual harvests dropped to <50/year, a level at which they have since remained. While harvest levels for spotted skunks in Arkansas were considerably lower than other midwestern states, trends in both annual harvests as well as demand for pelts (as assessed by pelt price) are closely correlated with those in other states.

Key words:— *Spilogale putorius*, harvest, biogeography.

Introduction

The eastern spotted skunk (*Spilogale putorius*) is a small carnivore found in the Great Plains and the southeastern United States ranging up the Appalachian Mountains to Pennsylvania (Kinlaw 1995). At one time the species was a commonly harvested furbearer with annual harvests in some midwestern states measured in tens of thousands (Gompper and Hackett 2005). By the 1940s however, annual harvests of the species began to precipitously decline. Analyses of long-term datasets indicate that capture rates per unit of effort declined sharply and thus that the drop in harvest reflects a real decline in spotted skunk populations although the causes of this decline remain unclear (Gompper and Hackett 2005). These datasets were most complete for states that had high historic harvests of skunks. In some states, however, historic harvests were never greater than a few thousand individuals, and the incomplete nature of the harvest datasets for these states limited insight into the changes in the harvest of the species outside of the central and upper Midwest (e.g. Missouri, Iowa, Nebraska) and especially in states like Arkansas in which most of the harvest is derived from forested habitats.

The first known occurrences of spotted skunks in Arkansas were reported by Black (1936) based on about 20 skins that a dealer who bought furs from Washington and Madison counties had purchased during the first part of the 1934-1935 season. A few years later specimens had been obtained from Washington and Boone counties, and Dellinger and Black (1940) reported that this skunk was common near Hot Springs, which was said to apparently be the eastern limit of the species distribution in the state. However, soon thereafter it was described as primarily a prairie animal that was not found in large numbers anywhere in its range, but that it was more common in counties that make up the Grand Prairie in eastern Arkansas and that its range was expanding westward from those areas (Roberts et al. 1942, Holder 1951). Sealander (1956) stated that spotted skunks were "fairly common" on the prairies of western Arkansas and the Grand Prairie, that they had recently invaded several unnamed eastern Arkansas counties, and that they had been established in parts of the eastern half of the state prior to Dellinger and Black's (1940) project. Based on museum specimens, literature records, and reports from Arkansas Game and Fish Commission personnel, in 1956 the spotted skunk was known from 36 of 75 (48%) counties across most of the state except extreme southwestern Arkansas and the northeastern portion of the Arkansas Delta (Sealander 1956).

Sealander (1979) reported that the spotted skunk was historically found in the Interior Highlands and the counties bordering this region comprising the Ozark and Ouachita Mountains and the intervening Arkansas River Valley. There was an apparent range expansion into most of the Gulf Coastal Plain in the 1950s and 1960s with the possible exception of extreme southwestern Arkansas, although this expansion may have been followed by a range contraction in the 1970s. Additional expansions occurred in the 1970s with the occupation of Clay, Greene, and Craighead counties in northeastern Arkansas.

Simultaneous to these 1970s range shifts was an apparent decline in the population of spotted skunks, which was putatively attributed to rabies, canine distemper, or to land use changes. Trapping was not thought to be a factor in the declines due to low fur prices (Sealander 1979). These Arkansas declines may, however, have well predated the 1970s (Gompper and Hackett 2005).

An updated review of Arkansas mammal distribution in 1990 reported that the spotted skunk occurred statewide, although their status in the Gulf Coastal Plain was questionable due to a lack of sightings and records (Sealander and Heidt 1990). After a reanalysis of museum records and existing range maps, Heidt et al. (1996) agreed that the species was found in the Ozarks and Ouachitas and that it was possibly distributed statewide. Mail surveys of trappers and state wildlife biologists provided evidence that the species was still found in the Gulf Coastal Plain but was absent from the Delta region and for the first time reported their presence in extreme southwestern Arkansas in the Red River bottomlands (Majors et al. 1996).

Thus, based on work carried out throughout the 20th century, Arkansas spotted skunk populations experienced subtly shifting geographic ranges and apparent declines in harvests or population sizes or both. In addition, recent analysis of a partial Arkansas dataset on spotted skunk harvests suggests a decline in the species that mirrors the declines observed in other states (Gompper and Hackett 2005). Therefore, to better understand the current status of spotted skunk populations in Arkansas, and to gain more detailed insights into historic changes in harvest and geographic range we compiled over 6 decades of harvest records so as to gain more detailed insights into historic changes in harvest and geographic range.

Materials and Methods

Spotted skunk harvest and pelt price information was gathered from licensed furdealers by the Arkansas Game and Fish Commission from 1941-2004 using report forms that were required to be submitted to the Commission at the end of each season. For 1943-2004, the data is also subdivided by region (Delta, Gulf Coast Plain, Ouachitas, Ozarks) and county-level information was available for the 1943-1944 1977-1985, and 1995-2004 seasons. Annual harvest for any given year represents the total capture value for a single season that typically runs from about November through January or February and thus incorporates data from two different years. For example, the value for 1962 is derived from captures made during the 62 day season that ran from 20 November 1962 through 20 January 1963.

Annual proportional harvests were calculated to compare the current-year harvest with the previous year harvest ($harvest_t$/$harvest_{t-1}$), and for both raw harvest annual values and proportional values, 3-yr moving averages were calculated. For regionally subdivided data, the percent of the total statewide harvest attributable to each region was calculated. For comparison of Arkansas harvests to those of other states, long-term data sets from Missouri, Iowa, and Nebraska were obtained from the literature (Bennitt and Nagel 1937, Sampson 1980, Novak et al. 1987, Iowa Department of Natural Resources 2002, Gompper and Hackett 2005). Together with the Arkansas data, data sets from these states represent the most complete long-term harvest records available for spotted skunks.

To assess variance in demand for spotted skunk pelts, data were obtained on annual pelt prices in Arkansas and Missouri. A consumer price index inflation calculator (U.S. Dept. of Labor, Bureau of Labor Statistics) was used to adjust the price of pelts for inflation to a 2004 baseline; all pelt price analyses used only the inflation-adjusted values. To partially correct for biases in harvest effort, we also collected data on the length of the trapping season (although data were lacking for this parameter for 1943-1955 as well as for several other shorter periods) to generate harvest per day. Data on the number of trappers operating on the Arkansas landscape were unavailable but as a surrogate of the number of Arkansas trappers, we used annual data on the number of licensed fur-buyers in Arkansas.

Annual harvest levels per region and for the entire state were graphed, and linear regression techniques were used to identify relationships between pelt price (current year and previous year) and annual harvest in Arkansas, as well as the relationship between Arkansas harvest and Arkansas pelt price and those values from other states. We also used multiple regression (stepwise forward and backward) to examine the predictive relationships between number of Missouri trappers, the number of Arkansas fur-dealers, the price paid for pelts in Arkansas, and the annual Arkansas harvest. Given the assumptions inherent in using the Missouri trapper population or the Arkansas fur-dealers population as a surrogate for the Arkansas trapper population and the assumption that much of the spotted skunk harvest may be incidental during efforts to capture other furbearers except during periods of high spotted skunk pelt prices (Gompper and Hackett 2005), we assume independence between the predictor variables and therefore do not include interaction terms in the regressions.

Results

Statewide harvest of spotted skunks peaked at 1,830 animals in 1942 during the second year of data collection, and declined steadily thereafter (Fig. 1). While brief multi-year increases in the harvest occurred in the 1960s and especially in the 1970s, by the late 1980s the annual harvest dropped below 50 animals, a level that has not been exceeded in the ensuing 2 decades. The decline in the harvest was greatest in the 1940s and early 1950s, such that by the late 1950s the 3-year moving average of annual harvest had stabilized at approximately 215 animals (1955-1959 range: 205-222) or 15% of the peak 3-year moving average of 1,445 animals harvested from 1941-1943 (Fig. 1).

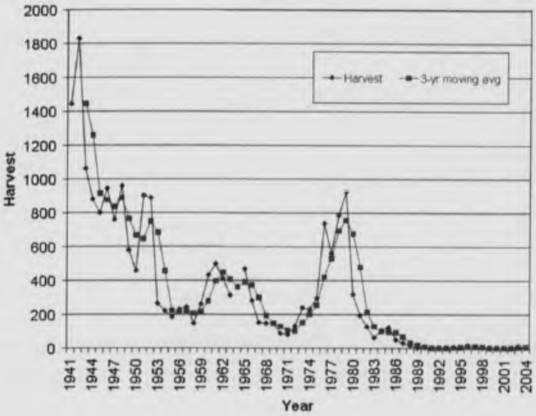

Fig. 1. Annual harvest (diamonds) and 3-year moving average (squares) of annual harvest for spotted skunks in Arkansas.

The decline was not steady, as annual increases over previous year harvests occurred in 7 of 18 years between 1942 and 1959, but when examined as 3-year moving averages, annual change relative to the previous years 3-year average was <1 in 12 of 16 intervals through 1959 (Fig. 2). Harvest increased sharply in the early 1960s relative to the late 1950s, but the peak 1960s harvest (498 animals in 1961) was less than all but one year prior to 1954, and by the late 1960s annual harvest had declined to <150 individuals. A second harvest peak occurred for 4 years in the late 1970s, with annual harvests similar to those observed in the mid-to-late 1950s and early 1960s. Following this brief increase, however, annual harvests steadily declined thereafter; from 1989-2004, annual harvest was <10 animals in all but 2 years (Fig. 1).

During the 1943 and 1944 furbearer seasons, the majority of spotted skunks were taken in the Ozarks and Ouachitas and counties bordering those regions with a small number of animals being reported from 4 counties in the southeastern part of the state and from Bradley County in the Gulf Coastal Plain (Fig. 3a). From 1977-1985, most of the harvest again came from the Ozarks and Ouachitas, however, there were a few harvested from Gulf Coastal Plain and Delta counties from which they were not taken in 1943-1944 (Fig. 3b). From 1995-2004 the small number of spotted skunks taken originated from counties in all ecological regions, though the majority were from the Ozarks (Fig. 3c). The total harvest is dominated by harvests from the Ozark and Ouachita regions with both regions showing temporal trends similar to the broader state harvest trends (Fig. 4). Until 1987, at which point state-wide harvest levels became quite low, harvests from the Ozark, Ouachita, Delta, and Gulf Coastal Plain regions make up on average 76, 17, 5 and 2%, respectively, of the total harvest. Since the 1940s, the relative contribution of the Ozark harvest to the broader statewide harvest has increased significantly ($r = 0.31$; $P = 0.017$).

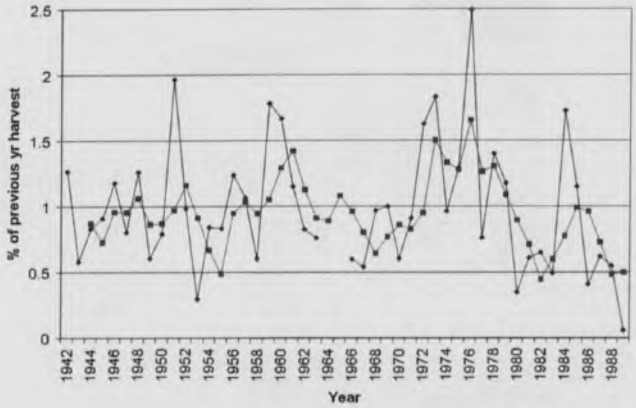

Fig. 2. Year-to-year proportional change in the harvest of spotted skunks throughout Arkansas (1942-1989). A value of 1 indicates no change in harvest, while a value <1 or >1 indicates a decrease or increase, respectively, in the annual harvest. Annual change relative to previous year harvest is shown in diamonds. Annual change in the 3-year moving average of annual harvest relative to previous year's 3-year moving average is shown in squares. Annual harvest is unknown for 1964, and thus 1964-1966 values represent 2-yr moving averages.

Annual harvests in Arkansas closely correlate with harvests in Missouri ($r = 0.78$), Iowa ($r = 0.81$), and Nebraska ($r = 0.70$), suggesting that harvest declines in Arkansas track those observed in other states despite the larger historic harvests from those other states. The Missouri harvest, for example, peaked at >55,000 in 1940 but declined in a similar time frame to Arkansas (Fig. 5). Missouri pelt price was also closely correlated with Arkansas pelt price ($r = 0.88$). Demand for spotted skunk pelts has resulted in considerable fluctuation of price. Arkansas spotted skunk pelts have varied in price from <$1.00 (inflation adjusted) in the early 1990s to over $30.00 in 1978 (Fig 6a). A one-year peak of $62 in 2002 should be considered suspect given the low number of pelts (2) harvested that year. This demand may have driven harvest per day (Fig 6b), although a lack of data on season length throughout most of the 1940s and 1950s hinders further analyses of the relationship.

There was a strong correlation between Arkansas harvests and the total number of fur-dealers ($r^2 = 0.709$; $P < 0.001$). Across all years (1941-2004), there was also no relationship between price and harvest ($P = 0.721$) or between previous-year price and harvest ($P = 0.398$). These analyses were also carried out for data sets limited to 1941-1990 and 1941-1985 and in no cases were a significant relationship identified. For the period 1941-1990 the relationship with previous year price approached significance ($P = 0.057$), but only a small amount of variance in

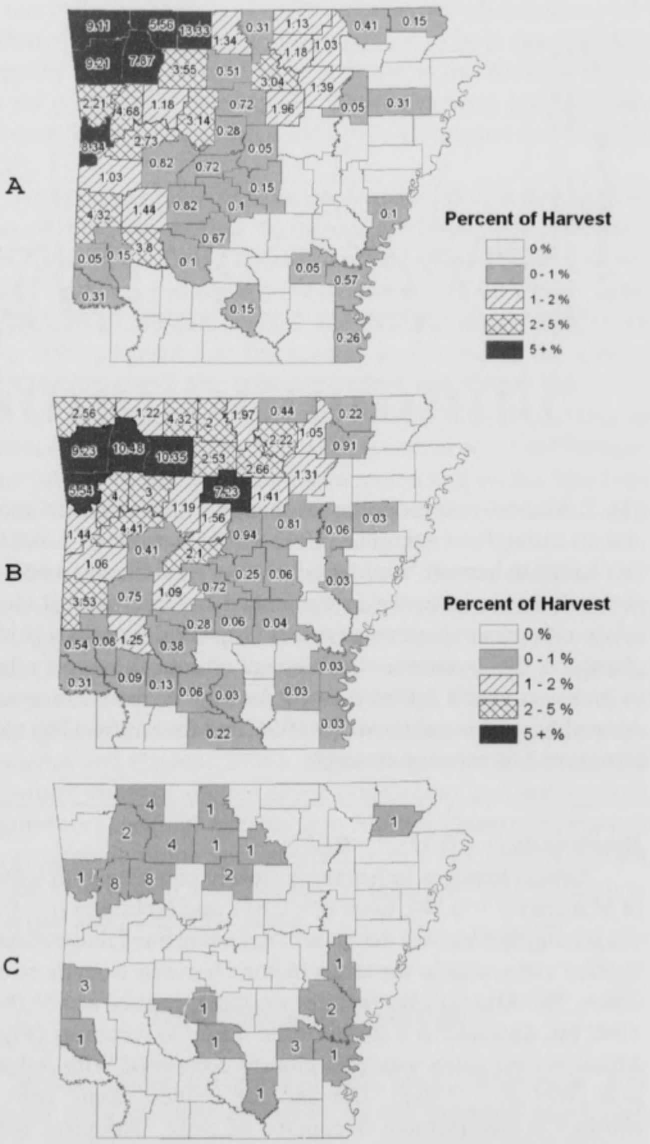

Fig 3. Harvest of spotted skunks in Arkansas, subdivided by county. A) 1943-1944 harvest as a percent of statewide harvest. B) 1977-1985 harvest as a percent of statewide harvest. C) 1999-2004 harvest per county. Given low total harvest, data are not converted to percentage.

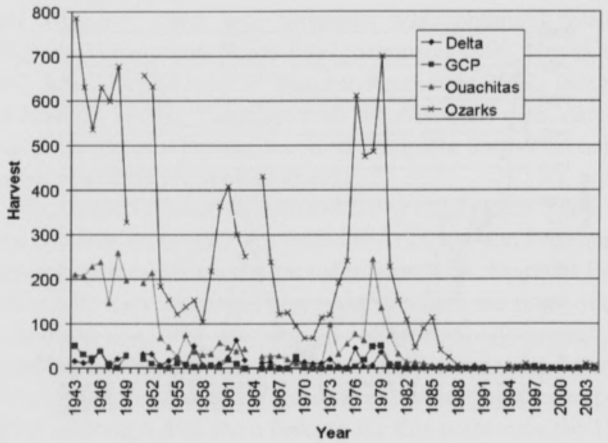

Fig. 4. Regional harvests of Arkansas spotted skunks.

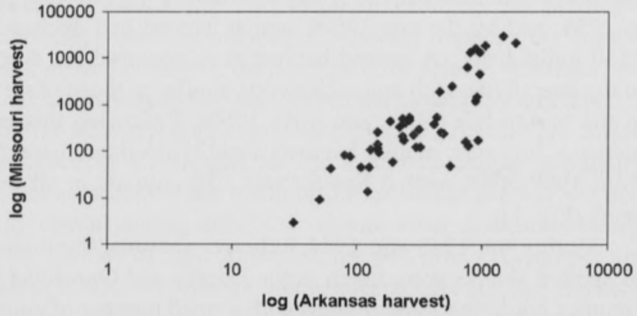

Fig 5. Relationship between Arkansas and Missouri harvests (1941-1987).

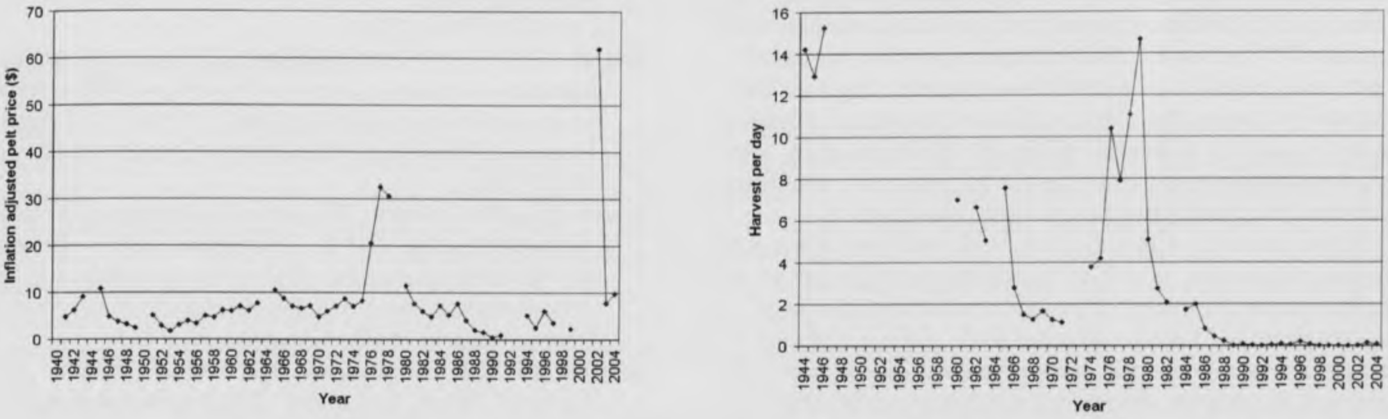

Fig. 6. Annual inflation-adjusted pelt price for spotted skunks in Arkansas (a; 1941-2004) and annual daily harvest (total harvest/season length) of spotted skunks (b; 1944-2004).

harvest was explained ($r^2 = 0.084$) by the relationship. Using all data (1941-2004) and both backwards and forwards stepwise regression, however, harvest was best predicted by the number of fur dealers and the current-year price ($df = 2, 48$; $F = 68.582$; $P < 0.001$) with these two parameters explaining (adj. r^2) 73% of the variance in harvest.

Discussion

General accounts of the distribution of this species in the 1940s and the early 1950s offer contradictory statements on whether the species was confined to the northwest part of the state and was moving east or was a species of the Grand Prairie and was expanding westward (Dellinger and Black 1940, Roberts et al. 1942, Holder 1951). However, records of the 1943 and 1944 furbearer seasons clearly indicate that the spotted skunk was well established in the Ozark and Ouachita mountains and nearby counties though they do not indicate its habitat preferences. And though only a few animals were taken in southeastern Arkansas in this period, it seems likely that the animal was already widely dispersed through southern Arkansas prior to Sealander's (1956) survey of Arkansas Game and Fish Commission personnel.

The continual harvest from areas outside the Ozarks and Ouachitas suggests that despite the decline in spotted skunk harvests and the possible decline in spotted skunk population size in Arkansas, the species remains widely distributed across the state. There has, however, been an increase in the relative portion of skunks harvested from the Ozarks, although the small post-1990 harvest sizes tend to obfuscate this pattern. Even though spotted skunks continue to be found in southern Arkansas they appear to be very rare, if present at all, in similar eco-regions in Mississippi, Louisiana, and Texas (Wolfe 1971, Lowery 1974, Schmidly 2004).

There was a strong decline in the harvest of spotted skunks in Arkansas beginning in the early 1940s – a pattern also seen in other states (Gompper and Hackett 2005). Peak harvest was 1,830 animals in 1942, and 3-year moving averages declined for 12 of 15 years from 1944-1958. Novak et al. (1987) gave harvest values of 2,166, 1,582, and 1,605 for the three years prior to the start of our dataset, but we have excluded these values from our analyses as we were unable to identify the source of the values as well as because values given by Novak et al. (1987) for several other years were seemingly either incorrect or listed for the incorrect season. Nonetheless, it appears that the decline in the spotted skunk harvests began with the 1943-1944 season when harvest was just 58% of the previous season's harvest.

The long-term Arkansas spotted skunk harvest is closely correlated with those of other states with far greater historic harvest levels. Thus, despite the relatively small absolute harvest values from Arkansas, the relative patterns for the state are similar to those for this species from other states. The spotted skunk pelt prices of Missouri and Arkansas also correlate closely. During particular periods, Missouri and Iowa pelt prices closely correlate with the harvest of spotted skunks from those states (Gompper and Hackett 2005), a pattern also observed in Arkansas based on 1965-1983 data (Clark et al.,1985). In Arkansas, increases in harvests did occur during the 1960s and 1970s when pelt prices increased, but over the broader (1941-2004) time frame, few patterns were identified between pelt price and spotted skunk harvest.

Perhaps more important than pelt price are the number of trappers and the length of the season (together giving the number of trapper-days). In Missouri, these variables together with pelt price strongly predict harvest, implying that spotted skunk captures are generally incidental to the capture of other more desirable species (Gompper and Hackett 2005). For Arkansas, data on the number of trappers were not available, and as potential surrogates for this measure, the number of Missouri trappers and the number of Arkansas furbuyers were utilized. The former was of little value in predicting harvest, but the latter parameter alone explained 71% of harvest. In a multiple regression, the addition of pelt price enhanced predictive power slightly. Together these relationships suggest that, like in Missouri, harvest of Arkansas spotted skunks is primarily driven by incidental take, with occasional targeting of the species (or perhaps active avoidance or discard of captured animals) mediated by pelt price.

Given the lack of historic data on the number of trappers per year in Arkansas, as well as limited data on the length of the trapping season for important periods covered in these analyses, the data presented here are insufficient to indicate a decline in spotted skunk populations (versus spotted skunk harvests). However, all patterns observed for Arkansas mirror those of other states where population declines have been documented. Therefore, continued conservation concern for this species is warranted.

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Parallel Algorithms for Multicriteria Shortest Path Problems

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Abstract.—This paper presents two strategies for solving multicriteria shortest path problems with more than two criteria. Given an undirected graph with *n* vertices, *m* edges, and a set of *K* weights associated with each edge, we define a *path* as a sequence of edges from vertex *s* to vertex *t*. We want to find the Pareto-optimal set of paths from *s* to *t*. The solutions proposed herein are based on cluster computing using the Message-Passing Interface (MPI) extensions to the C programming language. We solve problems with 3 and 4 criteria, using up to 8 processors in parallel and using solutions based on two strategies. The first strategy obtains an approximation of the Pareto-optimal set by solving for supported solutions in *bi-criteria sub-problems* using a weighted-sum approach, then merging the solutions. The second strategy applies the weighted-sum algorithm directly to the tri-criteria and quad-criteria problems to find the Pareto-optimal set of supported solutions, with each processor using a range of weights.

Key words.—Multicriteria shortest path problems, Message-Passing Interface (MPI), C programming language, *bi-criteria sub-problems,* weighted-sum algorithm, tri-criteria and quad-criteria problems.

Introduction

The “Shortest Path Problem” has been studied extensively in recent years and numerous algorithmic solutions are available. In its unicriterion version, Dijkstra’s algorithm provides a ready and efficient solution; in the bi-criteria case it has been studied extensively as well, and numerous algorithms have been proposed and tested. Documented research dealing with problem instances involving more than two criteria is also available but not to the same extent as in the bi-criteria case, and research dealing with parallel algorithmic solutions is rare. This paper presents two algorithms for solving multicriteria shortest path problems with more than two criteria. The solutions proposed herein are based on cluster computing using the Message-Passing Interface (MPI) extensions to the C programming language. The proposed algorithms were tested using graphs of various sizes and up to 4 criteria, and performance results are shown.

Multicriteria Shortest Path Problem and Background

Given $G = (V,E)$ is an undirected graph with a set of $|V| = n$ vertices, and $|E| = m$ is a set of edges, and a set of K weights is associated with each edge. We denote by c_{ij}^k the cost of edge (i,j) due to weight function k. A path is defined as a sequence of n arcs, $(s, i_1), (i_1,i_2)\ldots(i_{n-1},t)$ from vertex s to vertex t. We want to find the Pareto-optimal set of paths from node s to node t of G. If $K = 2$, the problem is the bi-criteria shortest path problem, which is defined as follows:

$$\min f_1(x) = \sum_{(i,j)\in E} c_{ij}^1$$

$$f_2(x) = \sum_{(i,j)\in E} c_{ij}^2$$

$s.t.$

$$\sum_{(i,j)\in E} x_{ij} - \sum_{(j,i)\in E} x_{ji} = \begin{cases} 1 & if\ i = s \\ 0 & if\ i \neq s,t \\ -1 & if\ i = t \end{cases} \qquad \textbf{(1)}$$

x_{ij} *binary*

The *nondominated* set of solutions is defined as the set having the following properties. Assume we have a vector of $K \geq 2$ objective functions:

$$\mathbf{f}(\mathbf{x}) = \left[f_1(\mathbf{x}), f_2(\mathbf{x}),\ldots, f_k(\mathbf{x})\right] \qquad \textbf{(2)}$$

to be minimized over a set of criteria. The decision variable is

$$\mathbf{x} = \left[x_1, x_2,\ldots, x_n\right] \qquad \textbf{(3)}$$

where the entire set of solutions is X. We say that $\mathbf{x}^*$ is *nondominated* or *Pareto-optimal* if there is no $\mathbf{x} \in X$ such that

$$\begin{aligned} & f_i(\mathbf{x}) \leq f_i(\mathbf{x}^*),\ i = 1..K, \\ & \text{and } f_j(\mathbf{x}) < f_j(\mathbf{x}^*) \ \text{ for some } j \in \{1..K\}. \end{aligned} \qquad \textbf{(4)}$$

For the bi-criteria problem, a variety of solution techniques based on minimized weighted-sum methods have been presented, such as Mote et. al. (1991), Henig (1985), Coutinho-Rodrigues, et. al. (1999), and Ehrgott (2000). We define weight λ such that $0 \leq \lambda \leq 1$ and we can reduce the bi-criteria problem to a unicriterion problem by using the following objective function:

$$\lambda f_1 + (1-\lambda) f_2 \qquad \textbf{(5)}$$

Finding all supported Pareto-optimal solutions requires solving multiple iterations of the problem using this objective function along with various weights. Various techniques for selecting the weights have been presented and tested. One approach is the parametric analysis. Using this approach, we generate a sequence of weights, $\{\lambda_q\}$, starting with $\lambda_1 = 1$, with each weight resulting in a distinct supported Pareto-optimal solution. (Henig 1985). We begin with the lexicographic shortest path associated with $\lambda_1 = 1$, then let

$$\alpha = \min \frac{-(c_{ij}^1 + f_1(p_j) - f_1(p_i))}{(c_{ij}^2 + f_2(p_j) - f_2(p_i))} \qquad \textbf{(6)}$$

where $f_k(p_j)$ is the cost of path j for criteria k, and the minimum is taken over all arcs in the graph such that the denominator is negative. This ratio is the depreciation in the first objective to the improvement in the second. Based on this value of α, we compute the next iteration based on the value $\lambda_2 = 1/(1 + \alpha)$. We continue in this manner until there are no negative results in the denominator, at which point the process terminates, having resulted in q Pareto-optimal solutions. The complexity of calculating α is $O(mn^2)$.

Another method, also described in Henig (1985), is to generate the sequence starting with $\lambda_1 = 0$ and $\lambda_q = 1$, solve the two lexicographic problems based on these values, resulting in paths p_1 and p_q. Using the ratio

$$\alpha = \frac{f_1(p_q) - f_1(p_1)}{f_2(p_q) - f_2(p_1)}, \qquad \textbf{(7)}$$

we set $\lambda_2 = 1/(1 + \alpha)$ and $\lambda_1 = 1 - \lambda_2$. As long as $\lambda_1 f_1 + \lambda_2 f_2$ results in new solutions, we continue the search of the nondominated front between the recently discovered solutions.

A third, more naïve method, but one which works well in parallel computing, is to generate a sequence of $\{\lambda_q\}$, starting with $\lambda_1 = 0$ and incrementing λ_q by some small α until $\lambda_q = 1$. Each value in the sequence is applied to (5). This method is easily expandable to values of $K > 2$ by employing the following objective function:

$$\lambda_1 f_1 + \lambda_2 f_2 + \ldots + \lambda_K f_K \qquad \textbf{(8)}$$

where the weights are input from file or generated automatically so as to give the desired distribution.

The problem with the first two methods described above is twofold. First, they are not easily expanded to higher values of K. Another problem, and one shared by all of the above approaches, is that they find only the supported Pareto-optimal solutions, defined as those solutions which lie on the convex hull of the feasible region. It has been shown in Ehrgott (2000) that the number of solutions to an MCSP may be exponential, but computational experience shows that this is not always the case. Existing solutions may be neglected due to the fact that they are not on the convex hull. The existence of such solutions can be seen in the following example. Assume that there are only Pareto-optimal supported solutions. We construct an instance of the Bi-criteria Shortest Path Problem (BSPP) in which we seek the Pareto-optimal set of paths from vertex 1 to vertex 5. (See Fig. 1) The solutions, all Pareto-optimal, are shown in Table 1. We construct another instance of the problem by inserting edge (1,5), with a cost of (3.8, 6.8). Now, in addition to the previous solutions, Path 1-5, which we will denote by p_5, with cost vector (3.8, 6.8), is a Pareto-optimal solution. Note that no value of α exists such that p_5 is the shortest path using the weighted objective function $(1-\lambda)f_1 + \lambda f_2$. Such a solution, often referred to as an "unsupported nondominated solution," cannot be found using any weighted-sum method. (See Fig. 2). However, unsupported solutions can be found by pairing a weighted-sum method with a second method designed to search for unsupported solutions, as described in Coutinho-Rodrigues (1999), for example. A complete solution set, including both supported and unsupported solutions, can be found by using labeling algorithms, as in Martins (1984), Mote et. al. (1991), and Brumbaugh-Smith and Shier (1989). Procedures for finding solutions for problems with $K > 2$ have been presented in Martins (1984), Corley and Moon (1985), and Ehrgott (2000); a summary of research in this area is found in Ehrgott and Gandibleux (2002). An *approximation* of a solution set is defined as a solution set obtained by using a heuristic algorithm. It is not guaranteed to be complete, but it provides a reasonable nondominated set of solutions from which to choose. Since the number of solutions

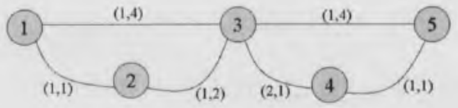

Fig. 1. An Undirected Graph

Table 1. Pareto-optimal Paths From Vertex 1 to Vertex 5 in Fig. 1.

Path	Cost Vector
p_1: 1-2-3-4-5	(5,5)
p_2: 1-3-4-5	(4,6)
p_3: 1-2-3-5	(3,7)
p_4: 1-3-5	(2,8)

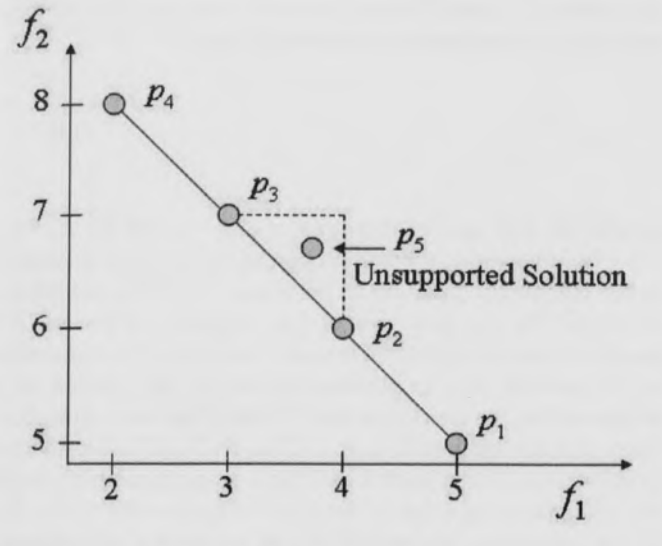

Fig. 2. Unsupported Nondominated Solution

to the multicriteria shortest path problem can be exponential, approximation methods are typically used.

This paper extends previous research by presenting a parallel algorithm for finding an approximation set for problems with $K = 3$ or $K = 4$ based on solving sub-problems involving only two criteria. We compare the performance of this algorithm against a straight-forward, parallel, weighted-sum implementation. The multiobjective version of the problem is the same as that presented in (1), but with the following objective function:

$$\min \sum_{(i,j)\in E} c_{ij}^{k} \quad \text{for } k = 1,\ldots,K \qquad \textbf{(9)}$$

Archival of solutions, which is of special importance in cluster computing, is discussed in Knowles and Corne (2004).

Methods

Decomposition of a K-criteria Problem into Bi-criteria Problems.—Brumbaugh-Smith and Shier (1989) define the *Merge* of any two nondominated sets *A* and *B* as the set of nondominated vectors in the union of sets *A* and *B*:

$$\begin{aligned} Merge(A,B) = A \cup B - \{\mathbf{x} \in A \cup B \mid \mathbf{x}^* \le \mathbf{x} \\ \textit{for some } \mathbf{x}^* \ne \mathbf{x},\ \mathbf{x}^* \in A \cup B\} \end{aligned} \qquad \textbf{(10)}$$

We define a *bi-objective sub-problem* of a *K*-objective problem as a sub-problem obtained by considering only two of the *K* criteria of the original problem. Given $K > 2$ objective functions, an approximation of the nondominated set of solutions can be determined from the merge of the solutions of all $\binom{K}{2}$ bi-objective sub-problems. A *K*-optimal solution is a solution which is optimal for all *K* objective functions.

A *K*-optimal solution discovered in one of the bi-objective sub-problems is discovered for the larger *K*-criteria problem. Assume that $\mathbf{x}^*$ is a *K*-optimal nondominated solution of the *K*-objective problem that is discovered in one of one of the $\binom{K}{2}$ bi-objective sub-problems. Then the following is true for some pair of criteria, *i* and *j*:

$$\begin{aligned} \neg\exists\,\mathbf{x} \text{ s.t. either } f_i(\mathbf{x}) \le f_i(\mathbf{x}^*) \text{ and } f_j(\mathbf{x}) < f_j(\mathbf{x}^*) \\ \text{or } f_j(\mathbf{x}) \le f_j(\mathbf{x}^*) \text{ and } f_i(\mathbf{x}) < f_i(\mathbf{x}^*) \end{aligned} \qquad \textbf{(11)}$$

since $\mathbf{x}^*$ is nondominated in the sub-problem involving criteria *i* and *j*. But since the solution is also *K*-optimal, then by **(4)** the following is true:

$$\begin{aligned} \neg\exists\,\mathbf{x} \text{ s.t. } f_i(\mathbf{x}) \le f_i(\mathbf{x}^*)\ \forall i \\ \text{and } f_j(\mathbf{x}) < f_j(\mathbf{x}^*) \text{ for some } j \end{aligned} \qquad \textbf{(12)}$$

Computational experience shows that a solution discovered in a bi-objective sub-problem is almost certain to be *K*-optimal, but it is not guaranteed that this is the case.

Example 1.—Consider a tri-objective problem in which the complete solution set is

{(1,3,5), (2,4,2), (3,1,7), (4,9,1), (5,2,2)}.

Assume we solve three bi-objective sub-problems: one optimizing k_1 and k_2, one optimizing k_2 and k_3, and one optimizing k_1 and k_3. The solution sets are shown in Table 2. The solution sets only show those objectives under consideration. Note, however, that the entire nondominated solution set can be found in the union of the three sets in Table 2:

Table 2. Bi-criteria Sub-problem Solution Sets.

Problem	**Solution Set**
k_1, k_2	{(1,3), (3,1)}
k_2, k_3	{(1,7), (9,1), (2,2)}
k_1, k_3	{(1,5), (2,2), (4,1)}

{(1,3,5), (3,1,7)} ∪ {(3,1,7), (4,9,1), (5,2,2)} ∪ {(1,3,5), (2,4,2), (4,9,1)}
={(1,3,5), (2,4,2), (3,1,7), (4,9,1), (5,2,2)}.

Based on the observations above, it is possible to find an approximation set for *K*-criteria problems by simultaneously solving bi-objective sub-problems and merging the solutions.

Multicriteria Shortest Path Algorithms.—If we begin with an algorithm that provides a solution set for bi-criteria sub-problems, a reasonable approximation of the nondominated solution set for the tri-criteria ($K = 3$) and quad-criteria ($K = 4$) problems can be found by merging these solution sets. This requires solving 3 and 6 sequences of bi-criteria problems in parallel, respectively. From this discussion we can see at least three strategies exist:

1. Obtain an approximation of the Pareto-optimal set by solving for supported solutions in bi-criteria sub-problems using a weighted-sum approach. Each processor solves a distinct set of bi-criteria sub-problems. Solutions are merged.
2. Apply the weighted-sum algorithm directly to the tri-criteria and quad-criteria problems to find the Pareto-optimal set of supported solutions. Each processor uses a range of weights, and solutions are merged.
3. Obtain an approximation set by solving for all solutions to the bi-criteria sub-problems using label correcting, then merging the solutions.

We provide a computational study of the first two strategies using problem instances with $K = 3$ and 4 and using from 1 to 8 processors.

Complexity and Scalability.—Using the first strategy, which is based on Proposition 1, we obtain an approximation of the Pareto-optimal set by solving for the supported solutions of the bi-criteria sub-problems using a weighted-sum approach. The solutions are merged, according to the definition (10) of "*Merge*()." There are Q Pareto-optimal solutions, $\{1..Q\}$, discovered in parallel in the bi-criteria sub-problems. The discovery of each of the Q solutions requires $O(n^2)$ since the "shortest path" algorithm is based on Dijkstra's Algorithm which is $O(n^2)$. Therefore the discovery of all Q solutions on p processors requires $O(Qn^2/p)$, assuming an even distribution of the solutions across the processors. However, in the worst case, it is possible that one of the processors could discover all Q o the solutions. Recall that we are not using just any arbitrar value for p, but rather the very specific value

$$p = \binom{K}{2}. \tag{13}$$

Concretely, this implies that for $K = 3$, $p = 3$ and for $K = 4$, $p = 6$. In either case, p could be considered as only a constant in the complexity analysis. This leaves us with a complexity of $O(Qn^2)$ for the discovery of the solutions. Although it is possible to create a problem instance in which Q is exponential to the problem size, in practice the size of the solution set is moderated by the restriction that the only solutions allowed are those that are supported in one of the bicritiera sub-problems. Computational experience shows that we can expect in the search for solutions using either of the first two approaches above, that $Q < n$. Assuming a communications constant of κ to transmit a single solution from the processor on which it is discovered to processor P_0 for output, the communication of the solutions requires $O(Q\kappa)$. The time required for both computation of the solutions and communication is, therefore, $O(Qn^2/p + Q\kappa)$. The time required for the merge of solutions on processor P_0 is, worst case, $O(Q^2)$ using a naïve merge algorithm, but using that presented in Brumbauth-Smith and Shier (1989), it is $O(Q)$. Therefore the total time can be expected to be $O(Qn^2/p + Q\kappa)$. For a large problem the first term can be expected to outweigh the second, and the computational time will be driven by the problem size. For a small problem the second term, communication of the results, will outweigh the first term.

Using the second strategy, the number of iterations depends on the value of λ for $K = 2$ and the size of the weight distribution table for $K = 3$ or $K = 4$. Assuming we generalize and denote by Φ the number of iterations, the computational complexity is

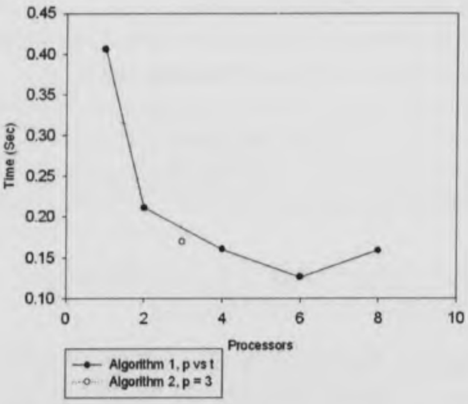

Fig. 3. 500 Vertices, $K = 3$

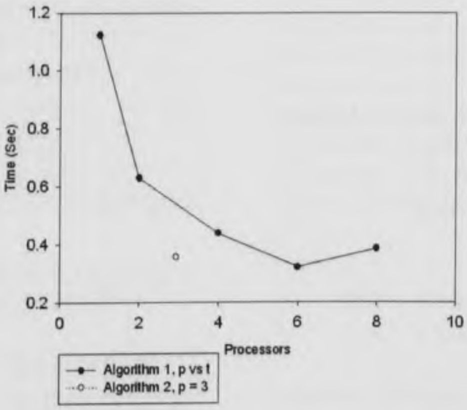

Fig. 4. 1000 Vertices, $K = 3$

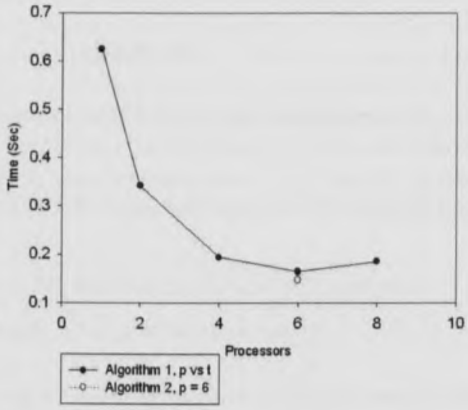

Fig. 5. 500 Vertices, $K = 4$

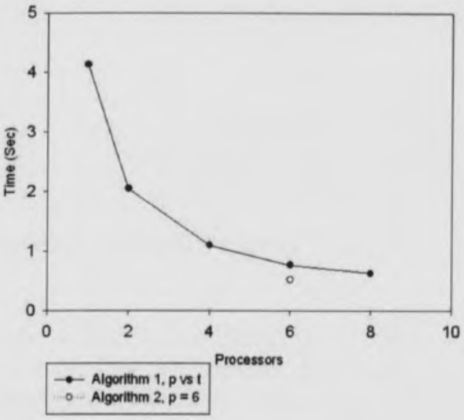

Fig. 6. 1000 Vertices, $K = 4$

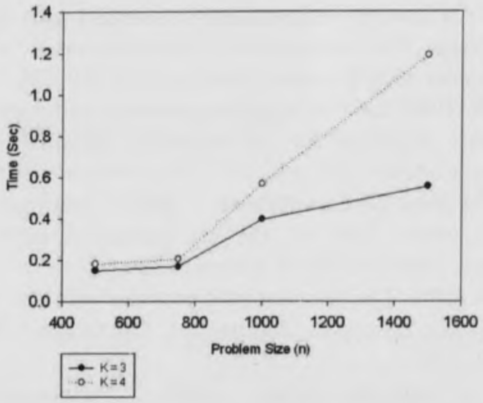

Fig. 7. Algorithm 1, Problem Sizes 500, 750, 1000, 1500 With $K = 3, 4$

$O(\Phi n^2/p + Q\kappa)$. We can therefore expect the performance of the first algorithm to be superior to that of the second in terms of quality of the solution set, but the second algorithm will be superior in terms of scalability in that it can run on a cluster of any size.

Results and Discussion

The graphs used for this study were complete graphs with edge costs generated randomly. The results follow the expectations generated by studying the complexity analysis. Figs. 3 and 4 show the results of applying both algorithms to graphs with 500 vertices and 1000 vertices, respectively, with $K = 3$ criteria. For a problem of either size, we can see that the benefit diminishes beyond the use of 6 processors. With less than 6 processors, performance degrades due to the nature of the first term, specifically its division by p. For more than 6 processors the performance degrades due to increased communication requirements. Figs. 5 and 6 show the results of both algorithms applied to graphs with 500 vertices and 1000 vertices, respectively, with $K = 4$ criteria. It is difficult to draw a direct comparison between Algorithm 1 and Algorithm 2 in terms of speedup because Algorithm 1 requires a very specific decomposition for parallel computing and a specific number of processors. We can, however, see that Algorithm 1 is generally equal or better, regardless of how many processors are used in Algorithm 2. In all cases the quality of the solution obtained by using Algorithm 1 is superior, since it includes all supported nondominated solutions and some unsupported nondominated solutions. Algorithm 2 finds only the supported nondominated solutions. Due to the static nature of the problem decomposition for Algorithm 1, data pertaining to speedup is not available, so studies of its performance with increasing problem size were conducted. Fig. 7 shows an example of the increase in computational time for increasing problem size observed using Algorithm 1.

Conclusions

Either methodology is easily expandable to problem instances involving more than 4 criteria, although in practice it is unlikely that a network or transportation related problem would have more than 4 or 5 criteria. Even so, both procedures can accommodate higher dimensions. For Algorithm 1, we solve a 5-criteria problem by solving 10 bi-criteria sub-problems, and a 6-criteria problem by solving 15 bi-criteria sub-problems. This requires $p = 10$ and $p = 15$ processors, respectively, but for a smaller cluster, multiple solution sets can be found on the same processor. The selection of only the supported solutions to the bi-criteria sub-problems provides a natural filter to the solution set size. As an extension to this research, a search for an approximation set could be implemented by solving for all solutions to the bi-criteria sub-problems using label correcting, then merging the solutions.

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Ichthyofaunal Assemblages in Three Proximate but Ecologically Diverse Streams in Clark County, Arkansas

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Abstract.—Three tributaries to the Ouachita River in eastern Clark County, Arkansas, empty into the river within a collective distance of about 9 km. The streams drain basins derived from the Wilcox formation, partially overlain by terrace and alluvial deposits. Despite their proximity, the streams are very different: L'Eau Frais has a gravel substrate and was recognized by the French as a cool water stream, Tupelo Creek is a bottomland stream from which numerous Water Tupelo (*Nyssa aquatica*) emerge, and Saline Bayou was named due to its marked salinity. We studied the assemblages of fishes in these 3 very different drainages to evaluate the occurrence of species and the degree of similarity of the ichthyofaunas. L'Eau Frais differed from the other streams in a greater occurrence of lampreys (Petromyzontidae), including a disjunct population of *Lampetra appendix*, and suckers (Catostomidae), including the relatively uncommon Blacktail Redhorse (*Moxostoma poecilurum*). Tupelo Creek had an assemblage of species characteristic of a bottomland stream, and Saline Bayou had the least diverse fish fauna.

Key words:—Ichthyofaunas, Ouachita River, Clark County, Arkansas, L'Eau Frais, Tupelo Creek, Water Tupelo, *Nyssa aquatica*, Saline Bayou, Lampreys, *Lampetra appendix*, Blacktail Redhorse, *Moxostoma poecilurum*.

Introduction

A small portion of Clark County, Arkansas, lies east of the Ouachita River. The area is within the West Gulf Coastal Plain just south of the Ouachita Mountains natural division (Foti 1974). Geologically, the region is composed primarily of alluvium deposited during the Holocene by L'Eau Frais Creek, Tupelo Creek, and Saline Bayou. The latter 2 streams drain basins lying almost entirely within alluvial deposits. Alluvium forms the banks of L'Eau Frais, but much of its upper reaches drain exposed strata of the Wilcox group deposited during the Eocene (Haley 1993).

These 3 tributaries enter the Ouachita River within a distance of about 9 km along the river (Fig. 1). Despite their proximity, they are ecologically very different.

L'Eau Frais Creek was named by early French pioneers who noted the "cool (or fresh) water." At about 37 km in length, this stream is the longest of the 3 we studied, and it flows throughout the year. Habitats within the stream include pools and riffles that flow over a substrate of sand and gravel with scattered deposits of detritus and mud.

In contrast, Tupelo Creek represents a bottomland stream of about 14 km in length. It is named for the Water Tupelo (*Nyssa aquatica*) trees that are common along its banks. Pools and riffles occur in the stream, but summertime lows often reduce the flow to a trickle. The substrate is composed of mud and detritus with scattered gravel in riffle areas.

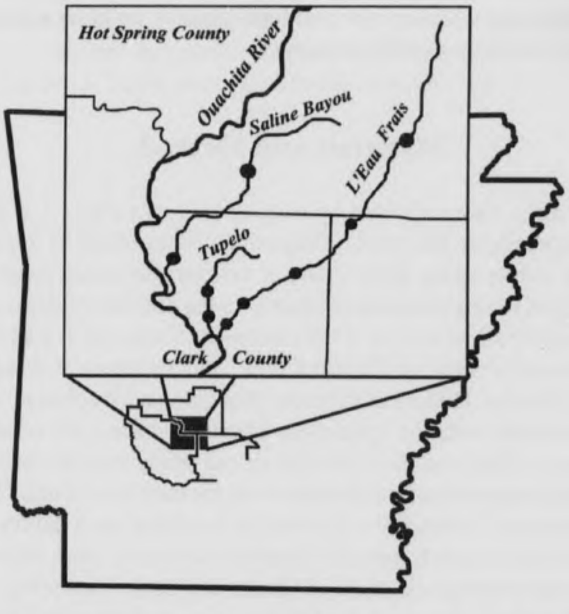

Fig. 1. Location of Saline Bayou, Tupelo Creek, and L'Eau Frais Creek in Clark and Hot Spring Counties, Arkansas. Dots along streams represent locations of sample sites.

Saline Bayou has a substrate of mud, gravel, sand, and detritus. It is about 18 km long and the stream looks somewhat similar to L'Eau Frais except for fewer sand bars. The stream has a long history as a source of salt. In the late 1700s, Louis Badins (2003) referred to the salty stream as *la petite saline*, which later became known as Saline Bayou. In 1804, William Dunbar obtained a sample of saline water after digging in the bed of a dry gully near Arkadelphia. Ten quarts of the water were evaporated, which produced a "saline mass weighing when dry 8 ounces" (Rowland 1930). One of the earliest salt works in Arkansas was established in 1811 at Blakelytown (near modern Arkadelphia) on a site along Saline Bayou where natives had made salt for years.

The substrates of the streams are strongly related to the soils within their basins. Sardis-Guyton-Ouachita soils form the alluvium through which the streams flow. These are flood-plain soils of silty clay loam. Upper slopes of the basins drained by Saline Bayou and Tupelo Creek have Gurdon-Stough-Amy soils, which also are of silty clay loam. Higher slopes of the drainage of L'Eau Frais Creek have Saffell-Sacul-Pikeville soils, which are deep gravelly and sandy loam soils (Hoelscher 1987).

Questions concerning the diversity of fish species in these tributaries arose after the discovery of an isolated population of the American Brook Lamprey (*Lampetra appendix*) in L'Eau Frais Creek (Tumlison and Tumlison 1999). This is a northeastern species formerly known to occur no farther south than the White River in Arkansas (Robison and Buchanan 1988). Cool, clear streams with gravelly substrates are required by *L. appendix*, and those conditions are met only in L'Eau Frais. We began surveys to determine whether other unique species or associations of species occurred in these streams.

Materials and Methods

Fishes were collected by seining (3.2 mm mesh, 1.2 X 3 m net) throughout the year. Collections were made at available access points along Hwy 7 and at bridges for minor roads. We attempted to sample proportionately to the number of access sites and made collections on 25 occasions at 5 sites on L'Eau Frais, 16 times at 2 sites on Tupelo Creek, and 10 times at 2 sites on Saline Bayou. Historical records (Robison and Buchanan 1988) are included with the specimens obtained during the course of this study. Because the objective of our study was to determine species composition, we did not count the numbers of individuals encountered. Instead, we focused on sampling each habitat type encountered in each stream. Voucher specimens were deposited in the vertebrate collections at Henderson State University.

Because some species of fishes (e.g., suckers) tend to enter tributaries of rivers for spawning, we tested water quality during the spring and late summer to determine whether variation among streams was consistent between high and low flow periods. We measured temperature, pH, salinity and conductivity because these parameters are related. Saline waters, expected to occur in Saline Bayou, tend to have a high pH and high conductivity The level of tolerance by fishes to these parameters would likel affect their likelihood of occurrence among the streams.

Species with low tolerance to change best serve as indicator of environmental degradation and pollution. Because most o the species found in our study occur in Oklahoma, we used th classification of Jester et al. (1992) to evaluate the compositio of fish communities found during our study. Chi-square tests were used to compare the frequencies of species within toleranc levels between pairs of streams.

Results and Discussion

A total of 64 species, representing 13 families, was collected during the study (Table 1). The samples included several cosmopolitan species with a mixture of upland (Ouachita Mountains) and lowland (Gulf Coastal Plain) components. L'Eau Frais and Tupelo Creeks each had 47 species, but we found only 36 species in Saline Bayou. L'Eau Frais and Tupelo shared 37 species in common, L'Eau Frais and Saline shared 27 species, and Tupelo and Saline shared 28 species. The lower number of shared species between Saline and the other streams likely reflects the lower number of species found in Saline.

Twenty-six species (40.6%) were shared by all streams, which likely means that those species are tolerant of the range of conditions present in all 3 streams. However, 24 species (37.5%) were unique to 1 of the 3 streams. Of these, 9 species were collected only from L'Eau Frais Creek, 8 species only from Tupelo Creek, and 7 species only from Saline Bayou.

The most important observation was the occurrence of *Lampetra appendix* in L'Eau Frais because it represents a southwestern extralimital population of a northeastern species. Interestingly, 3 of the 4 species of lampreys found in Arkansas were collected from L'Eau Frais. Most species of lampreys require gravel riffles for spawning, and the ammocoete larvae need silty to sandy areas with detritus in which to feed and grow (Robison and Buchanan 1988). Both species of *Ichthyomyzon* also were found in Tupelo Creek, but no species of lamprey was taken from Saline Bayou.

L'Eau Frais also had the richest diversity (4 species) of catostomids, 3 of which were unique to the stream. Tupelo Creek had 2 species of suckers, 1 of which was unique, but no suckers were found in Saline Bayou. Most species of suckers prefer relatively clear streams with gravelly or sandy substrates (Robison and Buchanan 1988), which explains the distribution we observed. We note that the specimens of *Minytrema melanops* and both species of *Moxostoma* were small and likely represent spawn within the streams.

The 2 bottomland species *Elassoma zonatum* and *Aphredoderus sayanus* were taken from all streams. We observed that both species were more common in Tupelo Creek; the most bottomland in nature of the streams sampled. Further, the species *Fundulus dispar*, *Lepomis marginatus*, *Notropis maculatus*, and *teronotropis hubbsi* were found only in Tupelo Creek. These pecies prefer mud and detritus bottoms (Robison and Buchanan 988), which is consistent with the habitat of Tupelo.

The 2 species collected only in Saline Bayou, *Menidia eryllina* and *Lepisosteus osseus*, are of interest because they re tolerant of brackish water and occur in coastal estuarine ituations (Robison and Buchanan 1988).

In L'Eau Frais and Tupelo Creeks, the majority of species 68%) were intolerant to changes in water quality (combining categories of intolerant and moderately intolerant, Table 2). In contrast, the majority of species in Saline Bayou (55.6%) were tolerant of change. Chi-square analysis indicated no significant difference ($P > 0.05$) between L'Eau Frais and Tupelo Creeks in tolerance of the species to changes in water quality, but comparison of Tupelo and Saline, and L'Eau Frais and Saline, indicated that tolerances of species present were significantly different ($P < 0.001$).

In all streams, most species were somewhat intolerant of habitat change, with the species in Saline Bayou averaging most tolerant. Chi-square analysis indicated no difference between species in L'Eau Frais and Tupelo Creeks in tolerance to change in habitat ($P > 0.05$), but comparisons between L'Eau Frais and Saline, and Tupelo and Saline, were significantly different ($P < 0.001$).

Parameters of water quality do seem to explain the differences found among streams. Monthly temperatures of all streams from February through April of 2006 were within 2°C of each other, increasing from about 10°C to 21°C. After diminished flow during summer, temperatures on 16 September 2006 remained about the same for L'Eau Frais (22°C), but had climbed in Tupelo (25°C), and Saline (27°C).

On 24 April 2006, pH of all streams ranged from 6.2-6.5. On 16 September 2006 pH had increased slightly in L'Eau Frais (6.7) and Tupelo (6.9), but markedly in Saline (9.2). With higher flow on 24 April, salinity was 0 in the former two streams and only 0.1 % in Saline Bayou, but on 16 September only Saline Bayou had increased – to a salinity of 1%. Patches of salt were evident at that time along dried sections of the stream.

Conductivity remained under 120 μmhos on 24 April and 16 September for both L'Eau Frais and Tupelo. In contrast, conductivity in Saline Bayou was measured as 180 μmhos on 24 April and increased to 1,900 μmhos on 16 September.

The increase in salt content of water in Saline Bayou explains the basic pH and the high conductivity. The lower diversity of species in that stream likely resulted from conditions that seasonally exceed tolerances of many freshwater species of fishes (Moyle and Cech 2004).

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Table 1. Occurrence (X) of species of fishes in three proximate tributaries to the Ouachita River, Clark County, Arkansas. Tolerance indicate (tolerance to change in water quality, tolerance to change in habitat), where I = intolerant, MI = moderately intolerant, MT = moderately tolerant, and T = tolerant (tolerance data from Jester et al. 1992).

		Tolerance	L'Eau Frais	Tupelo	Saline
Family:	Petromyzontidae				
	Ichthyomyzon castaneus	(MI, I)	X	X	0
	Ichthyomyzon gagei	(I, I)	X	X	0
	Lampetra appendix	(I, I)	X	0	0
Family:	Lepisosteidae				
	Lepisosteus osseus	(T, T)	0	0	X
Family:	Esocidae				
	Esox americanus	(MI, MI)	X	X	X
Family:	Cyprinidae				
	Campostoma anomalum	(MI, MI)	X	X	X
	Luxilus chrysocephalus	(MI, MI)	X	X	X
	Lythrurus umbratilis	(MI, MI)	X	X	X
	Notemigonus crysoleucas	(T, T)	X	0	0
	Notropis boops	(MI, I)	0	X	0
	Notropis maculatus	(MI, I)	0	X	0
	Pteronotropis hubbsi	(I, I)	0	X	0
	Cyprinella venusta	(MT, MT)	X	X	X
	Cyprinella whipplei	(MI, MI)	X	X	X
	Opsopoeodus emiliae	(MI, MI)	X	X	0
	Pimephales notatus	(MT, MT)	X	0	X
	Semotilus atromaculatus	(MI, MI)	X	0	0
Family:	Catostomidae				
	Erimyzon oblongus	(MI, I)	X	0	0
	Hypentilium nigricans	(I, I)	0	X	0
	Minytrema melanops	(MI, I)	X	0	0
	Moxostoma erythrurum	(MI, MI)	X	0	0
	Moxostoma poecilurum	(MI, MI)	X	X	0
Family:	Ictaluridae				
	Ameiurus natalis	(T, MT)	0	0	X
	Noturus eleutherus	(I, I)	X	0	0
	Noturus gyrinus	(MI, I)	0	X	X
	Noturus miurus	(I, I)	X	X	0
	Noturus nocturnus	(MI, MI)	X	X	0
Family:	Aphredoderidae				
	Aphredoderus sayanus	(MT, MI)	X	X	X
Family:	Fundulidae				
	Fundulus catenatus	(MI, I)	0	X	0
	Fundulus chrysotus	(MI, I)	0	0	X
	Fundulus dispar	(T, I)	0	X	0
	Fundulus notatus	(MT, MI)	X	X	X
	Fundulus olivaceous	(MT, MI)	X	X	X

able 1. (cont.)

	Tolerance	L'Eau Frais	Tupelo	Saline
Family: Poeciliidae				
Gambusia affinis	(T, T)	X	X	X
Family: Atherinidae				
Labidesthes sicculus	(MT, MI)	X	X	X
Menidia beryllina	(T, MT)	0	0	X
Family: Centrarchidae				
Centrarchus macropterus	(I, I)	X	0	0
Lepomis cyanellus	(T, T)	X	X	X
Lepomis gulosus	(MT, MT)	X	X	X
Lepomis macrochirus	(MT, MT)	X	X	X
Lepomis marginatus	(MT, MI)	0	X	0
Lepomis megalotis	(MT, MT)	X	X	X
Lepomis microlophus	(MT, MT)	0	0	X
Lepomis punctatus	(MT, I)	X	X	X
Lepomis symmetricus	(I, I)	0	X	X
Micropterus punctulatus	(MI, MI)	X	0	0
Micropterus salmoides	(MT, MT)	X	X	X
Pomoxis annularis	(T, MT)	0	0	X
Pomoxis nigromaculatus	(MT, MT)	0	0	X
Family: Elassomatidae				
Elassoma zonatum	(I, I)	X	X	X
Family: Percidae				
Ammocrypta vivax	(MI, I)	X	X	0
Etheostoma blennioides	(I, I)	X	X	X
Etheostoma chlorosomum	(MI, I)	X	X	X
Etheostoma collettei	(MI, I)	X	X	X
Etheostoma gracile	(MT, I)	X	X	X
Etheostoma parvipinne	(I, I)	X	X	0
Etheostoma proeliare	(MI, I)	X	X	X
Etheostoma radiosum	(MI, MI)	X	X	X
Etheostoma stigmaeum	(MI, MI)	X	X	0
Etheostoma whipplei	(MI, MI)	X	X	X
Etheostoma zonale	(I, I)	X	X	0
Percina copelandi	(MI, I)	0	X	0
Percina maculata	(MI, I)	X	X	0
Percina sciera	(MI, MI)	X	X	X

Table 2. Frequencies of fish species in L'Eau Frais Creek, Tupelo Creek, and Saline Bayou in relation to their tolerance of changes i habitat and water quality. Tolerance assignments were based on Jester et al. (1992). Total numbers of fishes (n) and their distributio among categories are given. The percent of fishes in each category are shown parenthetically. T = tolerant, MT = moderately tolerant MI = moderately intolerant, and I = intolerant.

Tolerance to changes in water quality

	T (%)	MT (%)	MI (%)	I (%)
L'Eau Frais (n = 47)	3 (6.4)	12 (25.5)	23 (48.9)	9 (19.1)
Tupelo (n = 47)	3 (6.4)	12 (25.5)	24 (51.1)	8 (17.0)
Saline (n = 36)	6 (16.7)	14 (38.9)	13 (36.1)	3 (8.3)

Tolerance to changes in habitat

	T (%)	MT (%)	MI (%)	I (%)
L'Eau Frais (n = 47)	3 (6.4)	6 (12.8)	19 (40.4)	19 (40.4)
Tupelo (n = 47)	2 (4.3)	5 (10.6)	17 (36.2)	23 (48.9)
Saline (n = 36)	3 (8.3)	11 (30.6)	12 (33.3)	10 (27.8)

Status and Distribution of the Arkansas Darter (*Etheostoma cragini*) in Arkansas

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Abstract.—The Arkansas darter, *Etheostoma cragini,* has an extremely limited distribution in Arkansas and is designated as a candidate for listing under the Endangered Species Act. It was first discovered in the state in 1979 in Wilson Spring near Fayetteville and has since been found in 4 additional headwater streams in Benton and Washington counties prior to this study. A study in 1997 found the species in 3 of the 5 historic streams, but one stream yielded only a single individual. This study sought to reassess the status of the 5 historically known populations and sample additional spring-run habitats in the Arkansas River basin in these 2 counties. Spring branch habitats were identified using USGS topographic maps and available GIS coverages. Surveys targeting *E. cragini* were conducted at 75 sites providing a broad coverage of the basin and including intense searches in the vicinity of historic sites. *E. cragini* were encountered in 15 stream segments, concentrated in 4 areas within the Illinois River basin. All segments supported numerous *E. cragini* and fell within a 2 km radius of historic sites. Each segment was broadly surveyed to delineate the extent of occupied stream length, which ranged from 10 to 1,645 m. Based on this survey the total occupied stream length for *E. cragini* in Arkansas was determined to be 5,676 m. These segments include three historic locations and 5 disjunct stream reaches. While the presence of *E. cragini* in Arkansas is persisting, rapid urban development in northwest Arkansas raises concern for some populations.

Key words:—Arkansas darter, *Etheostoma cragini,* Endangered Species Act, Fayetteville, Benton County, Washington County, Arkansas River basin, Illinois River basin.

Introduction

The Arkansas darter, *Etheostoma cragini*, was originally described from a site near Garden City, Kansas (Gilbert 1885). It is found in small spring-fed tributaries of the Arkansas River basin in Colorado (Beckman 1970), Kansas (Cross and Collins 1995), Oklahoma (Miller and Robison 2004), Missouri (Pflieger 1997), and Arkansas (Robison and Buchanan 1988). It is rare in Arkansas and is of special concern due to its limited habitat in the state (Robison and Buchanan 1988). It has been designated as a candidate for listing under the Endangered Species Act (USFWS 2005).

E. cragini was first found in the state in 1979 in Wilson Spring near Fayetteville, as reported by Harris and Smith (1985). They subsequently found the species at 5 locations: Healing Spring Run and Little Osage Creek, Benton County; unnamed spring run near Logan community, Benton County (Gailey Hollow area); unnamed spring run near Logan community, Benton County (Lower Palmer Spring); spring run tributary of Wildcat Creek northeast of White Oak Church and cemetery, Washington County (Huffmasters Spring); and Wilson Spring, HWY 112 & 71 bypass, Fayetteville, Washington County (Harris and Smith 1985). The most recent study showed the species to persist at 3 of the 5 historic locations: Gailey Hollow, Healing Spring Run, and Wilson Spring (Hargrave and Johnson 2003).

The objective of this study was to reassess the status of the five known historic locations and sample additional spring-run habitats in the Arkansas River basin of Benton and Washington counties, Arkansas, in order to assess the status and the extent of stream occupation by Arkansas darters in Arkansas. The rapid population growth and associated development in this area make it urgent to document the status of historic populations and identify potential previously undetected locations.

Materials and Methods

Spring branch habitats were identified using USGS topographic maps and available GIS coverages. A semi-uniform distribution of segments was identified for sampling with a minnow seine or dip net to determine presence of *E. cragini* and characterize the associated fish community. These were supplemented with intense searches in the vicinity of historic sites. Local landowner knowledge often exceeded that of current maps and available GIS information, and they occasionally provided access to additional areas likely to support *E. cragini*.

For the purpose of navigation, selected sites and historic sites were plotted on a map of northwest Arkansas, which showed towns, roads, and streams, using ESRI ArcMap™. Coordinates of selected sites were also downloaded to a handheld computer using ESRI ArcPad™, which was combined with a Bluetooth™ enabled wireless GPS unit for real-time mapping and navigation.

Fish were collected at each site using a 1-m, 3/16-inch mesh seine or 1/3-m, 1/8-inch mesh dip-net, as appropriate to habitat. This equipment was effective in sampling the habitat known to be occupied by Arkansas darters, and techniques were similar to the historic efforts (Hargrave and Johnson 2003, Harris and Smith 1985). Voucher specimens were preserved for confirmation and will be deposited in the collections of the University of Arkansas – Fort Smith or the Sam Noble Oklahoma Museum of Natural History. Vouchers of *E. cragini* and *E. microperca* were only taken from possible new sites.

Previous studies of *E. cragini* in Arkansas were focused either on presence/absence (Harris and Smith 1985) or on obtaining mark-recapture population estimates (Hargrave and Johnson 2003). The focus of this study was to determine the species' status in the most time-efficient manner and with minimum negative impact to this rare fish. Population estimates were determined to be too time consuming and detrimental to the fish. As an alternative, we attempted to delineate the extent of occupied habitat by sequential spot sampling along the stream reach at sites where *E. cragini* was encountered. We typically began at an upstream terminus (a spring head) and sampled on approximately 20 m increments downstream until no specimens of *E. cragini* or *E. microperca* were encountered in 2 consecutive samples. We sampled upstream in any tributaries we encountered in the same manner, allowing us to obtain GPS coordinates for endpoints of all occupied segments. Endpoints were defined as spring sources, confluences, dry stream reaches, or empirically determined ends of occupation by the species. Stream segments are defined as the length of stream between two adjacent endpoints. The GPS coordinates (decimal degrees, NAD27) of the endpoints were imported into ArcMap and used to measure the length of each stream segment.

Results

Sampling was completed at 75 total sites (Table 1). While sampling was conducted throughout the Arkansas River tributary basins of northwest Arkansas (Fig. 1), *E. cragini* was only encountered within the Illinois River basin. Fifteen samples included *E. cragini*; all of which were concentrated within 2-km of one of the historic sites.

Occupied Habitat.—We measured occupied habitat at the time of our samples to total 5,676 m of stream length. This habitat was distributed among 4 areas, as follows.

Area 1: Wilson Spring & Clabber Creek

Previous studies documented *E. cragini* throughout Wilson Spring and anecdotal reports from local students indicated presence in the Clabber Creek main stem. Due to active development in the Clabber Creek watershed, efforts were made in 2004 to document the fish community throughout the basin by sampling at 11 sites in the basin. These efforts confirmed the presence of *E. cragini* downstream of Wilson Spring to the beginning of a broad, deep, channelized section of Clabber Creek, in the lower end of a spring run entering from the opposite side of the creek, upstream in the creek at least to the Interstate 530 crossing, and throughout a drainage ditch flowing into the creek upstream of State Highway 112. In ou qualitative samples in this area we encountered 60 *E. cragini* and total occupied stream length was 2,467 m.

Area 2: Near Logan Community

Harris and Smith (1985) found *E. cragini* in 2 spring runs near the Logan community. Hargrave and Johnson (2003 identified these as Lower Palmer Spring and Gailey Hollow (actually a tributary to what USGS topographic maps label as Gailey Hollow). They found no *E. cragini* in Lower Palmer Spring and only one at the site referred to as Gailey Hollow, leading them to abandon attempts to estimate the population at that site. We found what we believe to be Lower Palmer Spring to be dry. In our qualitative samples at the Gailey Hollow site, we encountered 43 *E. cragini* throughout 2 spring branches that merge and flow for 324 m before all flow goes underground, resulting in a dry streambed downstream at the time of sampling. Total occupied stream length was 657 m.

Area 3: Wildcat Creek Basin

Harris and Smith (1985) found *E. cragini* in one spring run tributary of Wildcat Creek. Hargrave and Johnson (2003) identified this as Huffmaster Spring after consultation with Harris. They found no *E. cragini* in this area during their study. We likewise found no *E. cragini* in any spring run that may have been referred to as Huffmaster Spring. Through contact with a local landowner, we did discover another spring run inhabited by *E. cragini*, where our qualitative samples encountered 19 *E. cragini* in 45 m of occupied stream length. Searches of numerous other spring branches in this valley failed to discover any additional specimens of *E. cragini*.

Area 4: Healing Springs Area

Harris and Smith (1985) found *E. cragini* in Healing Spring Run, a tributary of Little Osage Creek. Hargrave and Johnson (2003) included a portion of this run in their study and estimated populations of *E. cragini* and *E. microperca*. We found *E. cragini* in Little Osage Creek at the mouth of the spring run, throughout the course of the spring run upstream to the source, and in the lower 230 m of a tributary spring branch (above this point the tributary spring became higher gradient and rockier, becoming less suitable habitat). We failed to keep count of the *E. cragini* encountered in this area, but the total occupied stream length for Healing Spring Run was 1,252 m.

E. cragini was discovered in 2 parallel spring runs on the east side of Little Osage Creek, which had not been reported by previous studies. We encountered *E. cragini* in 198 m of the easternmost of these springs and 161 m of the western, but did not keep accurate count in either case. We also found 9 *E. cragini* in an isolated, 10-m, roadside ditch, which historically would have been seasonally connected to the eastern of these springs (an intervening pond has isolated this population).

We also discovered *E. cragini* in another spring tributary of ble 1. Species collected by site. Collections highlighted in gray include *E. cragini.*

Collection #	Stream	Collection Date	*Campostoma anomalum*	*Luxilus cardinalis*	*Nocomis asper*	*Notemigonus crysoleucas*	*Notropis nubilus*	*Phoxinus erythrogaster*	*Pimephales notatus*	*Semotilus atromaculatus*	*Ameiurus melas*	*Noturus exilis*	*Fundulus olivaceus*	*Gambusia affinis*	*Cottus carolinae*	*Lepomis cyanellus*	*Lepomis macrochirus*	*Lepomis megalotis*	*Lepomis microlophus*	*Micropterus salmoides*	*Etheostoma cragini*	*Etheostoma flabellare*	*Etheostoma microperca*	*Etheostoma punctulatum*	*Etheostoma spectabile*
BKW2004-004	Clabber Creek	08-Apr-04				X					X		X	X	X	X	X	X		X				X	X
BKW2004-005	Clabber Creek	08-Apr-04							X				X	X		X		X							
BKW2004-006	Clabber Creek	08-Apr-04							X				X	X		X	X	X	X						
BKW2004-007	Clabber Creek	08-Apr-04									X		X	X		X	X	X	X	X					
BKW2004-008	Clabber Creek	09-Apr-04											X	X		X					X				
BKW2004-009	Wilson Spring	09-Apr-04														X					X				
BKW2004-010	Spring Run	09-Apr-04												X							X				
BKW2004-011	Marshy seep	09-Apr-04												X							X				
BKW2004-012	Clabber Creek	09-Apr-04									X		X	X		X	X								
BKW2004-013	Clabber Creek	09-Apr-04									X			X		X					X				
BKW2004-014	Ditch by old drive-in	09-Apr-04									X			X							X				
BKW2005-014	Healing Springs	04-May-05	X					X						X							X		X	X	X
BKW2005-015	Spring branch (no fish)	05-May-05																							
BKW2005-016	Ozark National Forest spring	05-May-05						X		X					X							X			X
BKW2005-017	Logan Spring	05-May-05								X					X										
BKW2005-018	Spring Run	10-May-05						X							X										
BKW2005-019	Shinn Spring	10-May-19													X										
BKW2005-020	Spring Run	10-May-05	X					X						X											X
BKW2005-021	Near Bennie Robison Road	10-May-05	X					X		X						X									X
BKW2008-022	Chambers Spring	11-May-05												X	X	X									
BKW2005-023	Unnamed private spring	11-May-05						X							X										X
BKW2005-024	Golf Course nr. Walnut Grove-	11-May-05	X					X						X											X
BKW2005-025	Shepard Spring	18-May-05						X		X															X
BKW2005-028	Spring run	25-May-28						X																	
BKW2005-029	Spavinaw Creek	25-May-05		X	X					X					X	X						X			X
BKW2005-030	Mikissic Spring	26-May-05	X					X																	X
BKW2005-031	Butler Creek	26-May-05		X				X		X				X	X	X						X		X	X
BKW2005-032	Trib. to Chalybeate Creek	26-May-05	X	X				X		X		X										X			X
BKW2005-033	Honey Creek	26-May-05		X				X				X			X										X
BKW2005-035	Spring run (no fish)	25-May-05																							
BKW2005-036	Chaney Spring	07-Jun-05	X	X			X	X		X				X	X	X					X	X		X	X
BKW2005-037	Spring in Bella Vista (no fish)	07-Jun-05																							
BKW2005-038	Blowing Spring	07-Jun-05								X															X
BKW2005-046	Spring run	21-Sep-05	X																						
BKW2005-047	Spring run trib	21-Sep-05						X						X		X								X	X
BKW2005-048	Unnamed spring seep	21-Sep-05						X						X		X									
BKW2005-049	Spring trib to Hamstring Creek	22-Sep-05	X					X						X											X
BKW2005-050	Spring run	22-Sep-05																					X		
BKW2005-051	Spring trib to Wildcat Creek	22-Sep-05						X						X							X				X

Table 1. continued

Collection #	Stream	Collection Date	*Campostoma anomalum*	*Luxilus cardinalis*	*Nocomis asper*	*Notemigonus crysoleucas*	*Notropis nubilus*	*Phoxinus erythrogaster*	*Pimephales notatus*	*Semotilus atromaculatus*	*Ameiurus melas*	*Noturus exilis*	*Fundulus olivaceus*	*Gambusia affinis*	*Cottus carolinae*	*Lepomis cyanellus*	*Lepomis macrochirus*	*Lepomis megalotis*	*Lepomis microlophus*	*Micropterus salmoides*	*Etheostoma cragini*	*Etheostoma flabellare*	*Etheostoma microperca*	*Etheostoma punctulatum*	*Etheostoma spectabile*
BKW2005-052	Spring trib to Wildcat Creek	22-Sep-05	X					X						X								X		X	X
BKW2005-053	Trib. to Wildcat Creek	22-Sep-53						X		X					X							X			
BKW2005-054	Spring run	23-Sep-05						X							X										
BKW2005-056	Little Osage Creek	05-Oct-05	X	X				X		X				X	X						X		X	X	X
BKW2005-057	Still House Spring run	05-Oct-05						X														X			X
BKW2005-058	Bluff Spring	05-Oct-05						X							X							X			X
BKW2005-059	Spring run	05-Oct-05	X					X		X	X			X	X			X			X		X	X	X
BKW2005-060	Spring run	06-Oct-05	X					X						X	X										
BKW2005-061	Spring trib. to Little Osage Creek (no fish)	06-Oct-05																							
BKW2005-062	Spring run	06-Oct-05												X											
BKW2005-063	Trib. of Dancing Rabbit Creek	06-Oct-05													X										
BKW2005-064	Huffmaster Spring	07-Oct-05										X		X		X						X		X	X
BKW2005-065	Ezell spring	22-Oct-05						X																X	X
BKW2005-067	Roaring Spring	22-Oct-05						X							X	X									X
BKW2005-068	Winton Spring	26-Oct-05													X										
BKW2005-069	Spring trib. to Spanker	26-Oct-05	X					X														X			
BKW2005-070	Griggs Spring	27-Oct-05																			X		X	X	X
BKW2005-071	Spring run	27-Oct-05						X							X									X	
BKW2005-072	Healing Spring Run	27-Oct-72						X						X							X	X	X		
BKW2005-073	Spring run	28-Oct-05																			X		X		
BKW2005-074	Spring run	28-Oct-05																			X				
BKW2005-076	Spring trib. to Barren Fork	14-Nov-05												X											
BKW2005-077	Spring run to Osage Creek	15-Nov-05						X																	
BKW2005-078	Big Spring trib. to Flint Creek	15-Nov-05						X							X										
BKW2005-079	Spring run	15-Nov-05						X																	
BKW2005-080	Spring run	15-Nov-05						X		X					X							X			
BKW2005-081	Little Osage Creek	16-Nov-05												X									X		X
BKW2005-082	Big Muddy Spring	16-Nov-05						X																X	
BKW2005-083	Rocky Spring	15-Nov-05						X						X	X									X	X
BKW2005-084	Spring trib. of Big Sugar Creek	17-Nov-05						X							X							X			
BKW2005-087	Spring run (no fish)	06-Dec-05																							
BKW2005-088	Elkhorn Springs	06-Dec-05						X						X											
BKW2005-089	Spring run	06-Dec-05	X					X							X									X	X
BKW2005-090	Spring trib. to Little Wildcat	07-Dec-05						X						X											
BKW2005-091	Little Wildcat	07-Dec-05							X													X		X	X
BKW2005-092	Dot Springs	22-Sep-53												X	X										X

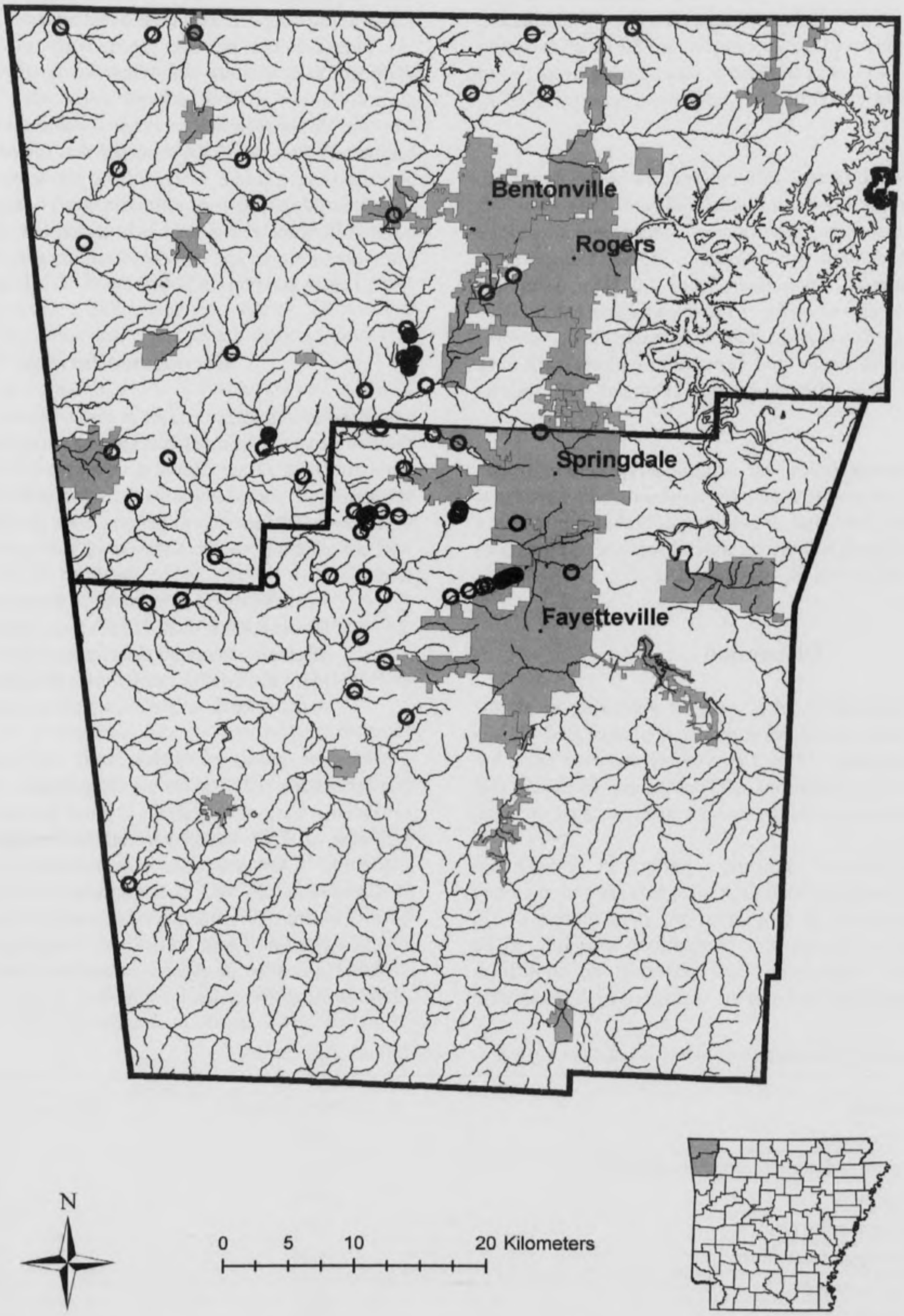

Fig. 1. Map of all sample locations in Benton and Washington counties, Arkansas. Solid circles indicate sites where *E. cragini* was encountered; opens circles indicate sites without *E. cragini*. Gray shading represents urban areas.

Little Osage Creek, which joins the creek 1 kilometer north of the source of any of these other springs. During our qualitative samples, we encountered 28 *E. cragini* in 886 m of occupied stream length. Searches of numerous other spring branches in this valley failed to encounter any additional specimens of *E. cragini.*

Species Associations.—Fifteen species were found to occur with *E. cragini*: *Gambusia affinis* (73% of sites), *Phoxinus erythrogaster, Etheostoma microperca, Etheostoma spectabile* (each 40%), *Etheostoma punctulatum* (33%), *Campostoma anomalum, Lepomis cyanellus* (each 27%), *Semotilus atromaculatus, Ameiurus melas, Cottus carolinae* (each 20%), *Luxilus cardinalis, Etheostoma flabellare* (each 13%), *Notropis nubilus, Fundulus olivaceus,* and *Lepomis megalotis* (each 7%). The diversity at sites with *E. cragini* averaged 4.87 species (range 1 to 12).

Habitat Characteristics.—*E. cragini* is reported to inhabit clear, spring-fed streams with aquatic vegetation, silt substrates, open canopy, slow flow, and of small size. Table 2 provides a comparison of selected habitat variable observations from sites with *E. cragini* and across all sites sampled.

Discussion

We found populations of *E. cragini* persisting in all the general areas where it had been reported historically. It has possibly been extirpated from 2 previously reported sites, but we also discovered populations at 5 additional locations that appear to be separated from the historic sites, at least for most of the year.

Surveys in Missouri indicate a pattern of downstream dispersal and presence at particular sites to vary among years (Missouri Department of Conservation, pers. comm.). In contrast to this, we found that populations appeared to be concentrated near springheads and observed no significant downstream dispersal away from the springs. This is consistent with dependence on stable thermal regimes, as reflected by survival rates reported by Labbe and Fausch (2000).

This does not, however, agree with our observation at the Wildcat Creek tributary site where occupation of habitat ended abruptly above shallow, pooled areas. Our initial hypothesis was that this was linked to either predation or thermal regime. Labbe and Fausch (2000) reported that introduced *Esox lucius* appeared to exclude *E. cragini* from pools, but native *Ameiurus melas* and *Lepomis cyanellus* did not (T. Labbe, unpublished data). Likewise, *E. cragini* has been found to be very tolerant of high temperatures and low dissolved oxygen (Smith and Fausch 1997, Labbe and Fausch 2000).

Recommendations

Populations of *E. cragini* in northwest Arkansas represent the southeastern limit of the species' range and appear to be persisting at this time. Rapid development in this area is already impinging on some habitats of this fish and should be managed in a way to maintain key habitats. We feel that the following will contribute to the persistence of *E. cragini* in the Arkansas fauna:

- Avoid filling in of small headwater spring branches.
- Divert contaminant-laden storm-water runoff.
- Maintain vegetated riparian buffers.
- Maintain open canopy in riparian areas.
- Avoid excessive livestock access and nutrient input.

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Table 2. Comparison of habitat characteristics at sites with *E. cragini* and all sites sampled.

Habitat Characteristic	At All Sites Sampled	At Sites with *E. cragini*
Described as "very clear"	98%	100%
Multiple types of rooted aquatic vegetation noted	31%	78%
Watercress only present	60%	14%
Mud/silt as dominant substrate	23%	78%
Sand/gravel as dominant substrate	61%	11%
<25% of shoreline wooded	63%	100%
Stream width < 10 m	96%	100%
Flow described as "slow"	52%	67%
Flow described as "moderate"	47%	33%

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The Vascular Flora of the South Fork Native Plant Preserve, Van Buren County, Arkansas

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Abstract.—The South Fork Native Plant Preserve, a property owned and managed by the Gates Rogers Foundation, Inc., is located along the South Fork of the Little Red River (now Greers Ferry Lake) in Van Buren County, Arkansas. We conducted a floristic inventory of a 61.3 ha study area consisting of the preserve, adjacent lakefront property owned by the U.S. Army Corps of Engineers, and a riparian corridor on neighboring land from February through November 2005. A total of 582 vascular plant taxa, representing 118 familes and 336 genera, was documented including six species (*Carex lupuliformis* Sartwell ex L. H. Dewey, *Carex swanii* (Fernald) Mack., *Claytonia caroliniana* Michx., *Nemastylis nuttallii* Pichering ex R. C. Foster, *Phemeranthus rugospermus* (Holz.) Kiger, and *Viola canadensis* L. var. *canadensis*) tracked by the Arkansas Natural Heritage Commission as species of conservation concern in Arkansas. Fifty-five taxa (9.5% of the total) are considered introduced to the state. The largest plant families represented were Asteraceae (69 taxa), Poaceae (62 taxa), Cyperaceae (51 taxa), and Fabaceae (39 taxa). Fourteen distinct communities (habitat types) were delineated in the study area. Among these, sandstone glades, a bedrock-bottom stream, bluffs, and an upland depression wetland were found to contribute significantly to the species richness of the study area. The depression wetland contained three species (*Vaccinium fuscatum* Ait., *Acer rubrum* L. var. *drummondii* (Hook. & Arn. ex Nutt.) Sarg., and *Carex lupuliformis* Sartwell ex L. H. Dewey) more typical of the lowlands of the Gulf Coastal Plain and the Mississippi Alluvial Plain.

Key words.—South Fork Native Plant Preserve, Gates Rogers Foundation, Inc., Little Red River, Greers Ferry Lake, Van Buren County, Arkansas, U.S. Army Corps of Engineers, vascular plant taxa, *Carex lupuliformis, Carex swanii*, *Claytonia caroliniana*, *Nemastylis nuttallii*, *Phemeranthus rugospermus*, *Viola canadensis* L. var. *canadensis,* Arkansas Natural Heritage Commission, Asteraceae, Poaceae, Cyperaceae, Fabaceae, *Vaccinium fuscatum*, *Acer rubrum* L. var. *drummondii*, and *Carex lupuliformis.*

Introduction

The South Fork Native Plant Preserve, owned by the Gates Rogers Foundation, Inc. is located on the South Fork of the Little Red River (Greers Ferry Lake), in Van Buren County, Arkansas (Fig. 1). We conducted an inventory of the preserve, adjacent shoreline property, and an adjacent tract of land (hereafter collectively refered to as "the study area") to document the botanical biodiversity of the site. This included documenting plant species richness as well as the location, type and condition of all significant terrestrial and aquatic plant communities on the site. Field work was conducted from February to November 2005.

Materials and Methods

Aerial infrared digital orthophotos and U.S. Geological Survey 7.5' topographic data (available at www.geostor.arkansas.gov) of the study area were analyzed to classify and map plant communities using ArcView GIS software. Ground truthing of these communities was conducted, new communities were added, and boundaries were redrawn and adjusted based on repeated site visits and GPS points taken throughout 2005 and in March of 2006.

Thorough species inventories were conducted in each community, by visiting each community in the study area a minimum of once every 2 weeks during the growing season. Each plant species encountered was recorded (by community) and a master plant species list was compiled. Habitat codes were assigned for each species in the study area, and a relative abundance value was assigned at the conclusion of the field surveys. Non-native and non-native invasive species were identified and labeled as such according to Arkansas Vascular Flora Committee (2006).

Voucher specimens were collected for each species encountered in identifiable condition (typically flowering or fruiting specimens of herbaceous species, and mature specimens of woody plants), with the exception of those species occurring in especially small populations, which we believed might be harmed by collecting. Specimens were pressed and dried according to standard methods and were mounted onto archival specimen paper for storage in the herbarium of the South Fork Native Plant Preserve, housed at the office of the Gates Rogers Foundation in Clinton, Arkansas.

Populations of plant species of conservation concern (ANHC 2005) were mapped using GPS/GIS technology and detailed data on habitat and associate species were collected. These data

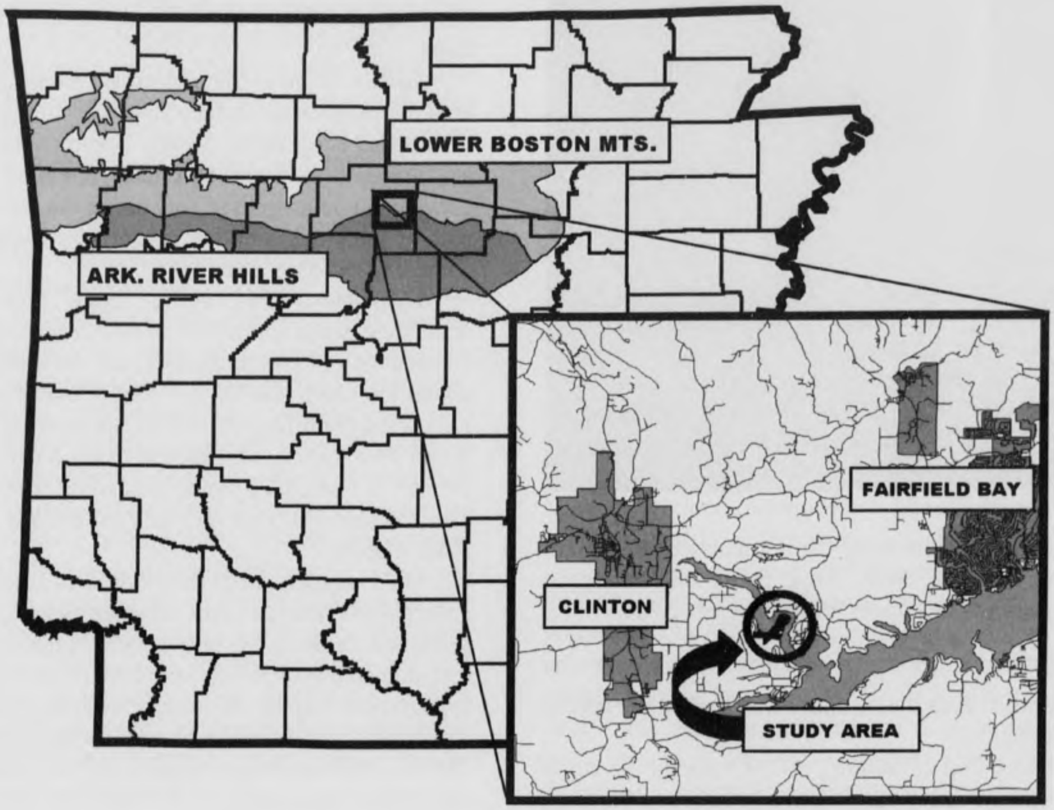

Fig. 1. Map showing the location of the South Fork Native Plant Preserve Study Area, Van Buren County, Arkansas.

will be provided to the Arkansas Natural Heritage Commission for inclusion into their statewide database of elements of special conservation concern.

Description of the Study Area

Location.—The South Fork Native Plant Preserve is located in Van Buren County, Arkansas, approximately 8 km southeast of Clinton, 5.5 km northeast of Choctaw, 13 km south-southwest of Shirley, and 16 km southwest of Fairfield Bay. The study area is larger than the existing preserve. It includes the existing preserve (owned by the Gates Rogers Foundation), land lying between the existing preserve and Greers Ferry Lake (owned by the U.S. Army Corps of Engineers), and a parcel of private land to the south of the Foundation land that includes a high quality example of a bedrock-bottom stream with springs, riparian habitat, and adjacent sandstone glades. The study area encompasses approximately 61.3 ha and falls within the SE ¼ of the SE ¼ of Section 29, the NW ¼ of Section 33, and that part of Section 28 that is south and west of Greers Ferry Lake, all within Township 11 North, Range 13 West (T11N, R13W). The site is prominent and easily located on a map or aerial photograph by the large peninsula projecting into the lake from the south shore (Figs. 1 and 2).

Ecoregional Position.—Woods et al. (2004) show the study area located at the boundary between the Lower Boston Mountains Ecoregion (Level IV Ecoregion 38b) of the Ozark Plateau and the Arkansas Valley Hills Ecoregion (Level IV Ecoregion 37c).

Elevation.—Elevation in the study area ranges from approximately 174 m above sea level at the highest point (near the entrance gate at the corner of Sections 28, 29, 32, and 33), to 140.5 m above sea level at the normal pool elevation of Greers Ferry Lake. The lowest elevation rises and falls with the lake level and the amount of dry land available to terrestrial plants expands and contracts accordingly based on lake level response to seasonal precipitation and withdraws. At the lowest lake level of 2005, the study area extended down to 137.5 m above sea level (USACOE 2006). This exposed approximately 10.3 ha

Fig. 2. Aerial photograph showing the boundary of the South Fork Native Plant Preserve Study Area. Note that boundary shown in lake represents low water level in October 2005.

of dry land in the study area that is under water when the lake is at normal pool elevation.

Geology.—Haley and co-authors (1976) mapped the study area as the Bloyd Shale and Prairie Grove Member of the Hale Formation and showed an area of the Atoka Sandstone to the southwest of the study area (Bald Mountain). Based on the lithologic descriptions of McFarland (2004), it seems possible that the coarse scale of the map (1:500,000) introduced some error and that the upper geologic units in the study area are potentially Atoka sandstone underlain by the older Bloyd Shale. The upper elevations, bedrock-bottom stream, and sandstone glades all have tan, brown, or gray sandstone bedrock, which is consistent with the Atoka Formation (McFarland 2004). This is clearly underlain by an older gray shale unit that is visible on the lower part of the north-facing slope and the area below a waterfall (which is located at the boundary between the sandstone and shale units near the southeastern edge of the preserve where a stream enters Greers Ferry Lake).

Anecdotal information about the ecological preferences and distribution of certain plants (e.g. the calciphile *Asplenium rhizophyllum* on a shale outcrop) indicates that the shale unit contains some calcareous material. Similarly, the abundance of a number of acid-loving plants over the sandstone portions of the study area (e.g. *Pinus echinata*, *Vaccinium* spp., and *Quercus falcata*) indicates that this rock does not contain significant amounts of calcareous material. It is possible, however, that this sandstone is part of the Trace Creek Member of the Bloyd Formation which has significant amounts of sandstone and is known to occur as far east as the study area (McFarland 2004). McFarland (2004) also mentions that many modern workers consider the Trace Creek Member to be a part of the Atoka Formation. It is unclear if the Hale Formation outcrops at all in the study area above the normal pool elevation of Greers Ferry Lake.

Soils.—Soils in the highest elevation uplands in the study area are mapped as Enders-Steprock complex, 8 to 20% slopes (Townsend et al. 1986). Gently sloping areas at lower elevation are mapped as Steprock-Linker complex, 3 to 8% slopes. Lowlands and a riparian area along the stream in the southern part of the study area are mapped as Steprock-Mountainburg complex, 3 to 8% slopes.

Hydrology.—The entire study area lies within the watershed of the South Fork of the Little Red River. Prior to the construction of Greers Ferry Lake, the study area was situated above the south side of the South Fork at a bend in the river. Following the filling of the lake, the portion of the valley of the South Fork adjacent to the study area was flooded to a depth of 10.5 to 13.5 m above the normal water level of that river. This also inundated a large area of bottomlands, river terraces, and lower slopes.

Only one stream in the study area is large enough to have a developed riparian plant community associated with it. This unnamed stream is the only stream in the study area that shows up (indicated by a broken blue line) on the 1:24,000 scale USGS topographic map for the area. The stream flows from southwest to northeast and drains a watershed with an area of approximately 168 ha. This watershed ranges from 290 m above sea level at the top of Bald Mountain to 140.5 m at the normal pool elevation of Greers Ferry Lake (137.5 m in December 2005). Land use in the watershed of this stream is predominantly low-intensity forestry (selective cut and natural regeneration) with three pastures covering a total of 16.6 ha on top of Bald Mountain in the uppermost reaches of the west end of the watershed. As of 2000, there was a single residence in the watershed, also on top of Bald Mountain.

Human History of the Study Area.—While we are unaware of any documented evidence of Native American occupation within the study area proper, archaeological evidence of Native American presence in the Ozark Highlands dates back to approximately 12,500 years before present, with evidence indicating widespread occupation by about 10,000 years ago (Nelson 2005). Native American influence on the environment can be attributed to two principle patterns: ecosystem-wide effects from widespread burning of the landscape and local effects resulting from agriculture, hunting, and settlement (Nelson 2005). At least the former of these effects may have played an important role in the shaping of the plant communities in and around the study area prior to the removal of Native Americans and arrival of European settlers.

The earliest European settlement in the 93.2 sq. km (36 sq. mile) township containing the study area (T11N, R13W) occurred in areas along the South Fork of the Little Red River, beginning at least as early as 1844 according to Government Land Office records (Risener 2006). The more rugged and ocky upland areas in the vicinity were not settled until the late 300s and early 1900s (Risener 2006). The first land patent eeded from the government in the four sections surrounding e study area was for a 16.2 ha (40 acre) tract along the South ork deeded from the government in 1844 (Risener 2006). The rst deeded parcel of land that included part of the study area as deeded in 1860 (Risener 2006). Other parcels containing e study area were deeded in 1877, 1882, and 1894 (Risener 006).

Construction on the Greers Ferry Dam began in 1959 and was completed in 1962. The dam impounds the Little Red River and several of its tributaries, inundating an area of approximately 164 sq. km creating Greers Ferry Lake (Greers Ferry Chamber of Commerce 2006). Greers Ferry Lake partially surrounds the study area and influences some of the plant communities, particularly the shoreline and lowland forest communities.

While the study area is almost entirely forested today, an historical aerial photo provided by the Gates Rogers Foundation reveals that a large portion was cleared for fields or pasture as recently as the 1950s. More recently, these fields have converted to young stands of *Pinus echinata* and *Juniperus virginiana* var. *virginiana* in the uplands and to stands of *Liquidambar styraciflua* and *Quercus phellos* in the lowlands at the north end of the peninsula. Several old abandoned roads are still evident in the study area, as well as other signs of past human use including an old home site, an dry-stacked stone wall, and a large, apparently human-dug hole, presumably a well or prospecting pit (Witsell and Baker pers. obs.).

Evidence of past logging can be found in the mixed pine-hardwood forests of the study area. Some of the persisting old stumps are tall enough to be consistent with those left by crosscut saw logging. There is no evidence of mining in the study area (Witsell and Baker pers. obs.).

Climate.—The nearest known site to the study area for which precipitation data are officially recorded is the home of Mary Alice Beer at Fairfield Bay. The yearly precipitation total at this site for 2005 was 89.79 cm, a near record low. This was 38.46 cm below the normal amount of 128.25 cm, averaged over several years at the same site. May, October, and December were particularly dry months with 1.14, 2.34, and 0.74 cm of precipitation respectively. The low May total in particular likely had an adverse impact on some plant species, occurring at the height of the growing season. Total precipitation in 2004 (an above-average year) was 144.04 cm.

While this low rainfall may have had an adverse impact on some plant species, it was responsible for a lowering of Greers Ferry Lake, which exposed a larger-than-normal area of exposed shoreline (Fig. 2), benefiting plant species that utilize this habitat. In 2005 the water level in the lake dropped to 137.48 m above sea level, 3.0 m below the normal pool elevation of 140.5 m (USACOE 2006). The maximum pool elevation of Greers Ferry Lake in January was 142.13 m, giving a total drop of 4.65 m for the year. Monthly mean lake levels show a more or less steady (with the exception of a small rise in April) drop in the lake level throughout 2005.

Summary of Floristic Diversity

A total of 582 taxa of vascular plants representing 118 families and 336 genera was found in the 61.3 ha study area. Of these 582 taxa, 6 taxa are tracked by the Arkansas Natural Heritage Commission as species of conservation concern in Arkansas (ANHC 2005, NatureServe 2005). Fifty-five taxa are considered to be introduced, either intentionally or accidentally, to the United States from elsewhere (AVFC 2006). The largest families are Asteraceae (69 taxa), Poaceae (63 taxa), Cyperaceae (51 taxa), and Fabaceae (39 taxa). The largest genera are *Carex* (Cyperaceae - 28 taxa), *Dichanthelium* (Poaceae - 11 taxa), *Quercus* (Fagaceae - 9 taxa), *Juncus* (Juncaceae - 9 taxa), *Desmodium* (Fabaceae – 8 taxa), *Lespedeza* (Fabaceae – 8 taxa), *Cyperus* (Cyperaceae – 8 taxa), *Solidago* (Asteraceae – 8 taxa), *Symphyotrichum* (Asteraceae – 8 taxa), *Hypericum* (Clusiaceae – 8 taxa), and *Viola* (Violaceae – 7 taxa).

It should be noted that this total of 582 taxa represents 20.04 % of the total number of vascular plant taxa known from Arkansas (AVFC 2006). According to Smith (1988), these are more taxa than have been documented from 36 of the 75 counties in the state. As far as we know, this inventory of the study area has documented the highest number of vascular plant taxa from any area of comparable size in the history of Arkansas's botanical exploration. This is due in part to the significant habitat diversity of the study area and in part to the thoroughness of the inventory.

As of 1988, Van Buren County was ranked 65th (out of 75 counties) in terms of documented plant diversity with 443 taxa documented by voucher specimens (Smith 1988). This was increased to a total of 943 taxa based on collection activities of Mary Alice Beer from 1988-1993 (Beer pers. comm.). Babb (1973) reported 984 taxa from adjacent Cleburne County.

Of the 582 taxa documented, voucher specimens for 537 taxa were collected, leaving 45 species that were observed in the study area, but were not collected. Some of these species were seen only once (perhaps just as leaves in the early spring) and were never relocated. Others were seen only in sterile (non-flowering/non-fruiting) condition, probably due to lack of sufficient light in many cases. Many other plants aborted flowering, died, or went dormant in the drought periods of the spring and summer.

Descriptions of Plant Communities in the Study Area

The following plant communities (Fig. 3) were identified, mapped, and qualitatively described in the study area:

1) Lowland Forest.—This community occurs in flat to nearly flat lowlands at the northeastern end of the peninsula and

Fig. 3. Map of Plant Communities at South Fork Native Plant Preserve Study Area.

occupies 4.8 ha within the study area. The canopy is dominated by *Quercus nigra*, *Q. phellos*, and *Liquidambar styraciflua. Diospyros virginiana*, *Fraxinus pennsylvanica*, *Acer rubrum* var. *rubrum*, and *A. negundo* are common in localized areas. The understory is dominated by *Smilax rotundifolia* and *Smilax bona-nox* with *Smilax glauca* locally common in some areas. Other woody vines include *Campsis radicans*, *Trachelospermum difforme*, and *Brunnichia ovata*. The herbaceous layer is typically sparse and of low species richness. Where present, it consists primarily of sedges, particularly *Carex lupulina*, *C. tribuloides*, and *C. typhina*, and a few species of forbs including *Triadenum tubulosum*, *T. walteri*, *Botrychium biternatum*, *B. dissectum* f. *obliquum*, *Eryngium prostratum*, *Pluchea camphorata*, *Stachys tenuifolia* var. *tenuifolia*, and *Helenium flexuosum*.

According to the historic aerial photo provided by the Gates Rogers Foundation this community occurs primarily in low areas that were fields as recently as the late 1950s. A dense understory of greenbriers makes this community impenetrable to walk through in many areas. Much of this community is in an area that is flooded by Greers Ferry Lake for short periods of time when water levels are high. Water levels were low during 2005, but high-water levels can be determined by the location of an obvious debris line deposited by the water.

2) ***Shoreline.***—The shoreline community extends from the normal pool level of Greers Ferry Lake to the low-water mark in a given year. The amount of area of this habitat exposed depends on the annual rainfall and the amount of water lost from the reservoir through evaporation and discharge through the dam. In 2005 the reservoir level dropped to 137.5 m above sea level, 3 m lower than the normal pool elevation (USACOE 2006). This exposed approximately 10.3 ha of shoreline habitat with 2.82 km of water edge within the study area.

By September of 2005, field surveys found that the shoreline community could be divided into 3 distinct concentric "zones" from the edge of the lowland forest to the water's edge. Vegetation patterns in these zones were clearly related to the length of time each zone had been exposed. In addition to these zones, a sandy grassland dominated by perennial grasses, sedges, and forbs was identified in a small area on the east side of the peninsula. The flora of this grassland was distinct enough and the community persistent enough that we felt that it deserved recognition as a formal community. It should be noted that each of these zones grades into the next, but that they are extensive and uniform enough to be considered distinct by even casual observer. However, in a wetter year not all of the zones re likely to be exposed. It can be assumed that diversity of this ommunity is highest during drought years (because more soil exposed) and was thus well-expressed in 2005.

The following associations were observed on the east side f the peninsula where elevation change was the most gradual.

A) Green Ash/Silver Maple Zone: This community ccurs in a thin, linear strip at the edge of the normal pool levation of Greers Ferry Lake. Only species tolerant of hort-term inundation can occur here. Scattered trees of *Acer accharinum* and *Fraxinus pennsylvanica* occur with a shrub layer of *Cephalanthus occidentalis.* Woody vines are common, particularly *Smilax* spp., *Campsis radicans*, *Trachelopsermum difforme*, and *Brunnichia ovata.* Ground layer vegetation includes *Lespedeza cuneata*, *Smilax bona-nox*, *S. glauca*, *Dichanthelium dichotomum* subsp. *dichotomum*, and seedlings of *Diospyros virginiana* and *Liquidambar styraciflua.*

B) Buttonbush Zone: This zone is evidently too wet for woody plants other than buttonbush and for most perennial herbaceous plants. *Cephalanthus occidentalis* occupies the upper stratum and may occur in thickets or as scattered individual shrubs. An herbaceous layer, when the ground is exposed due to low lake levels, consists mostly of weedy annuals, but includes the emergent perennial *Justicia americana* and scattered clumps of *Panicum rigidulum* subsp. *rigidulum.* Common annuals include *Eragrostis hypnoides*, *Oldenlandia boscii*, *Fimbristylis vahlii*, *F. autumnalis*, *Cyperus squarrosus*, *Eryngium prostratum*, *Lindernia dubia*, *Rotala ramosior*, *Lipocarpha micrantha*, and *Eleocharis acicularis.*

C) Recently Exposed Sandy Shoreline: This zone is occupied by a diversity of annual grasses, sedges, and forbs. The only perennial species in this zone appeared to have germinated during the study year. Dominant species include *Eragrostis hypnoides*, *Cyperus squarrosus*, *Lipocarpha micrantha*, *Fimbristylis vahlii*, *Fimbristylis autumnalis*, and *Eleocharis acicularis.* Other commonly encountered species include *Ammania* X *coccinea*, *Rotala ramosior*, *Oldenlandia boscii*, *Persicaria pensylvanica*, *Persicaria punctata*, *Persicaria lapathifolia*, *Echinochloa muricata* var. *microstachya*, *Ludwigia decurrens*, *Panicum dichotomiflorum* subsp. *dichotomiflorum*, *Ipomoea lacunosa*, *Eryngium prostratum*, *Bidens frondosa* var. *frondosa*, and *B. aristosa.*

3) Perennial Shoreline Grassland.—This community occurs in a small area of sandy soil on the eastern side of the peninsula just above the normal pool elevation of the lake. It is essentially treeless, though scattered *Liquidambar styraciflua* and *Diospyros virginiana* saplings and *Cephalanthus occidentalis* shrubs occur. *Hibiscus moscheutos* subsp. *lasiocarpos* is common in this community. The herbaceous layer differs markedly from the other open shoreline habitat in that it is dominated by perennial species. This community is very wet during periods of high lake levels and very dry in times of low lake levels. As such, it contains a mix of wetland species and species characteristic of xeric, sandy habitats. It can be considered analogous to sandbar communities along infrequently flooded river terraces. Total area occupied by this community within the study area was 0.14 ha.

The herbaceous layer is dominated by *Panicum rigidulum* subsp. *rigidulum*, *Axonopus furcatus*, *Cyperus pseudovegetus*, and *Helenium flexuosum.* Perennial sedges include *Carex lupulina* and *Carex typhina.* Other species commonly encountered include *Diodia teres*, *Paspalum laeve*, *Eryngium prostratum*, *Trachelospermum difforme*, *Boltonia diffusa*, *Pluchea camphorata*, *Polypremum procumbens*, *Hypericum gentianoides*, *H. drummondii*, *H. mutilum*, *Triadenum walteri*, *Chamaecrista nictitans* var. *nictitans*, and *Linum striatum.* *Shoenoplectus pungens*, *Rhexia mariana* var. *mariana*, *R. virginica*, *Axonopus furcatus*, *Crotalaria sagittalis*, and *Ludwigia glandulosa* are apparently restricted to this habitat within the study area. Annual grasses and sedges typical of the shoreline community are also common in this community, especially at the lower, eastern end and in areas where perennial species are sparse.

This community is easily discernible on both the 1950s and 2000 aerial photos. As such, it appears that this community is the result, at least in part, of edaphic factors (perhaps related to the sandy, nutrient-poor soil) and is not entirely the result of periodic flood-related disturbance from the lake.

4) Upland Depression Wetland.—This unique community is represented by a single example within the study area, occupying a total area of approximately 0.32 ha. This wetland is naturally occurring and is situated in a shallow circular depression in an otherwise upland setting. Drainage is impeded by a low ridge at the south end of the wetland. Shallow, standing water is common during the winter and spring months and in periods of higher rainfall, but the wetland became dry during the summer and in periods of low rainfall.

The wetland is forested and is dominated by *Liquidambar styraciflua* and *Quercus nigra.* The shrub layer is sparse but includes species more commonly found in the Gulf Coastal Plain including *Vaccinium fuscatum* and *Acer rubrum* var. *drummondii. Ilex decidua* var. *decidua* and *Lyonia ligustrina* also occur in the shrub layer. *Smilax bona-nox* and *S. rotundifolia* are common components of the understory. Plants in the herbaceous layer are typically clump-forming and are concentrated around the edge of the wetland and on raised hummocks within it. *Sphagnum* spp. and other mosses are common, forming dense mats in the wetter areas.

The herbaceous layer is dominated by a diversity of sedges including *Carex albolutescens*, *C. complanata*, *C. glaucodea*, *C. vulpinoidea*, *C. swanii*, *C. tribuloides*, *C. lupuliformis*, *Eleocharis tenuis* var. *verrucosa*, *Rhynchospora recognita*, and *Scirpus georgianus.* Rushes and grasses are also common, including *Juncus effusus*, *Agrostis perennans*, and *Leersia virginica.* Common forbs include *Packera obovata*, *Lycopus rubellus*, *Pycnanthemum tenuifolium*, *Claytonia virginica*, *Thalictrum thalictroides*, *Hedyotis caerulea*, and *Mitchella repens.* In addition to 2 sedges of conservation concern (*Carex* *swanii, C. lupuliformis*), several species of orchids are found in and around this wetland including *Liparis liliifolia, Malaxis unifolia*, and *Spiranthes cernua*. There is no sign of human alteration of this wetland and it appears to be forested (and wet) in the 1950s aerial photo.

5) Sandstone Glades and Woodlands.—Several small glades, all with a sandstone substrate, are found within the study area. These typically occur on south- and west-facing slopes above bluffs and along both sides of a broad, shallow bedrock-bottom stream. These glades grade into open woodlands, which occur on deeper soil surrounding the glades. The total area of all sandstone glade and associated woodland habitat in the study area is approximately 3.7 ha.

Woodlands surrounding the glades are dominated by *Quercus stellata, Q. marilandica, Pinus echinata, Carya texana*, and *Juniperus virginiana* var. *virginiana*. *Fraxinus americana, Chionanthus virginicus, Vaccinium arboreum, Sideroxylon lanuginosum, Ulmus alata,* and *Celtis tenuifolia* are common small trees which occur scattered in and around the edge of the glades. Common shrubs include *Rhus aromatica* var. *aromatica, R. copallina, Symphoricarpos orbiculatus*, and *Hypericum prolificum*.

Open areas were dominated by a diversity of warm season grasses including *Aristida dichotoma* var. *curtissii, Aristida purpurascens, Sporobolus clandestinus, Dichanthelium depauperatum, D. linearifolium, Tridens flavus* var. *flavus, Schizachyrium scoparium* var. *scoparium, Andropogon ternarius* var. *ternarius*, and *A. virginicus*. Common forbs include *Hypoxis hirsuta, Hypericum drummondii, H. gentianoides, Viola pedata, V. sagittata, Allium canadense* var. *mobilense, Nothoscordum bivalve, Croton wildenowii, C. monanthogynus, Euphorbia corollata, Pycnanthemum tenuifolium, Scutellaria parvula* var. *australis, Oenothera linifolia, Delphinium carolinianum* subsp. *carolinianum, Saxifraga palmeri*, and *Glandularia canadensis*.

Uncommon species restricted to this habitat include *Acalypha monococca, Astragalus distortus* var. *engelmannii, Claytonia caroliniana, Polygonum tenue, Camassia scilloides*, and *Nemastylis nuttallii*.

6) Sandstone Bluffs.—Sandstone bluffs occur in areas where the South Fork of the Little Red River and the unnamed stream at the south end of the study area cut into the hillsides. Within the study area these occur primarily on the north-facing slope, on the west side of the peninsula, and, to a lesser degree, on the east side of the peninsula.

The flora of these bluffs depends on local exposure and hydrology. North- and east-facing bluffs support a more mesophytic flora, while south- and west-facing bluffs are drier and support more xerophytic species. More mesic bluffs in the study area typically support *Quercus muhlenbergii, Styrax grandifolius, Ulmus americana, Acer rubrum* var. *rubrum, Carpinus caroliniana, Lonicera flava, Euonymus americanus, Rhododendron prinophyllum, Hydrangea arborescens, Philadelphus pubescens*, and *Toxicodendron radicans*. Herbaceous plants include *Mitchella repens, Heuchera americana* var. *americana, H. americana* var. *hirsuticaulis, Saxifraga palmeri, Parietaria pensylvanica, Hybanthus concolor, Carex albicans, C. oligocarpa, C. umbellata, Dioscorea villosa, Poa sylvestris, Sphenopholis intermedia, S. obtusata, Asplenium platyneuron, A. trichomanes* subsp. *trichomanes, Pleopeltis polypodioides* var. *michauxiana*, and *Thaspium trifoliatum* var. *flavum*.

Drier bluffs support *Pinus echinata, Quercus muhlenbergii, Q. stellata, Chionanthus virginicus, Amelanchier arborea, Ulmus alata, Juniperus virginiana* var. *virginiana, Rhus aromatica* var. *aromatica, Toxicodendron radicans, Lyonia ligustrina*, and *Vaccinium arboreum*. Herbaceous plants include *Solidago buckleyi, S. ulmifolia* var. *palmeri, Symphyotrichum anomalum, Paronychia fastigiata, Tephrosia virginiana, Cunila origanoides, Penstemon arkansanus, Viola pedata, Luzula echinata, Dichanthelium linearifolium*, and *Woodsia obtusa*.

7) Shale Bluffs and Outcrops.—Shale bluffs are rare within the study area and are small. The flora of these outcrops is similar to that of the north-facing slope. A small outcrop occurs on the north facing slope, on a small bluff mostly of sandstone. It is the only site within the study area for walking fern (*Asplenium rhizophyllum*), a species typically found on moist limestone rocks.

8) Springs and Spring Runs.—Several small intermittent springs and associated groundwater-fed stream segments (spring runs) occur in the study area. These are characterized by lush beds of *Sphagnum* sp., sedges, rushes, and other wetland plants. All of the springs are associated with sandstone bedrock near the surface of the ground which acts as an impermeable barrier to groundwater and channels it to the point of emergence.

Characteristic species include *Lycopus rubellus, Scutellaria parvula* var. *australis, Spigelia marilandica, Eleocharis tenuis* var. *verrucosa, Clematis reticulata, Saxifraga palmeri, Mitchella repens, Carex oklahomensis, Juncus debilis, Spiranthes cernua, Agrostis perennans, Chasmanthium latifolium, Leersia virginica, Isoetes melanopoda, Selaginella eclipes, Amsonia tabernaemontana, Cardamine pensylvanica, Hypericum mutilum*, and *Vicia minutiflora*. Plants restricted to this community within the study area include *Callitriche heterophylla* subsp. *heterophylla, Selaginella eclipes*, and *Isoetes melanopoda*.

9) Roadsides.—There are a total of 2.7 ha of roads and roadside habitat in the study area. These areas support a mix of weedy species (both native and exotic) and conservative native species dependent on open habitat. This latter suite of species cannot survive, or at least cannot flower, in the closed canopy forests that dominate the study area. This habitat, due to suppression of woody plants by mowing, is dominated by herbaceous species with a significant graminoid component. Common graminoid species include *Schedonorus arundinaceus, Tridens flavus* var. *flavus, Andropogon virginicus, Agrostis hyemalis, Bromus* spp., *Panicum anceps, Dichanthelium* spp., *Carex cephalophora, C. leavenworthii, C. hrisutella*, and *C. muehlenbergii* var. *enervis*. A wet area on the west side of chelor Road supports a diversity of wetland associated sedges d rushes including *Carex vulpinoidea*, *Scirpus cyperinus*, *eocharis lanceolata*, and *Juncus effusus*.

Shrubs are common along the edge of the roadsides and clude *Acer rubrum* var. *rubrum*, *Liquidambar styraciflua*, *sculus pavia*, *Callicarpa americana*, *Hypericum prolificum*, *iospyros virginiana*, *Aralia spinosa*, *Rhus aromatica* var. *omatica*, *Rhus copallina*, and *Rhus glabra*.

Forbs in the roadside habitat can be divided into two groups: 1) weedy native and exotic species and 2) conservative native species dependent on open habitat. Weedy species (exotic species noted by an "*") include *Chaerophyllum tainturieri* var. *tainturieri*, *Daucus carota* *, *D. pusillus*, *Ambrosia artemisiifolia*, *A. bidentata*, *Bidens aristosa*, *Conyza canadensis*, *Erigeron strigosus*, *Eupatorium serotinum*, *Gamochaeta purpurea*, *Helenium amarum*, *Pseudognaphalium obtusifolium* subsp. *obtusifolium*, *Rudbeckia hirta* var. *pulcherrima*, *Solidago nemoralis*, *Draba brachycarpa*, *Lonicera japonica* *, *Cerastium* spp., *Chamaesyce maculata*, *Kummerowia striata* *, *Trifolium* spp.*, *Vicia sativa* *, *Hedeoma hispida*, *Salvia lyrata*, *Plantago* spp., *Rubus* spp., *Sherardia arvensis* *, *Veronica arvensis* *, *Nuttallanthus texanus*, *Valerianella radiata*, and *Viola bicolor*. Conservative, light-dependent native species found in roadsides in the study area include *Asclepias tuberosa* subsp. *interior*, *Liatris squarrulosa*, *Brickellia eupatorioides* var. *texana*, *Vernonia baldwinii*, *V. missurica*, *Trifolium reflexum*, *Lespedeza hirta* var. *hirta*, *L. virginica*, *Scutellaria ovata*, *Senna marilandica*, *Stylosanthes biflora*, *Tephrosia virginiana*, *Sabatia angularis*, *Pcynanthemum albescens*, *P. tenuifolium*, *Linum medium* var. *texanum*, *Delphinium carolinianum* subsp. *carolinianum*, *Ceanothus americana*, *Penstemon digitalis*, *P. tubaeflorus*, *Ruellia humilis* var. *humilis*, *Monarda bradburiana*, *M. fistulosa*, and *Phlox pilosa* subsp. *pilosa*.

Fifteen of the exotic species that occur in the study area were identified as known or potentially invasive species. All of these occur in this habitat. These species are *Carduus nutans*, *Daucus carota*, *Leucanthemum vulgare*, *Lonicera japonica*, *Lespedeza bicolor*, *Lespedeza cuneata*, *Perilla frutescens*, *Melia azedarach*, *Ligustrum sinense*, *Bromus commutatus*, *Bromus hordeaceus* ssp. *hordeaceus*, *Bromus racemosus*, *Dactylis glomerata*, *Schedonorus arundinaceus*, and *Ailanthus altissima*.

10) Upland Pine-Hardwood Forest.—This is the matrix habitat in the upland portions of the study area and occupies a total of approximately 25.8 ha. It is dominated in most areas by mixed species of *Quercus* and *Pinus echinata* with *Carya tomentosa*, *C. texana*, *Prunus serotina*, *Liquidambar styraciflua*, *Fraxinus americana*, and *Nyssa sylvatica* occurring as locally important species. The density of *P. echinata* varies and is likely as much the result of past timber management activities as it is a function of geomorphology, hydrology, or other abiotic factors. However, *P. echinata* is generally more common on south- and west-facing aspects in areas where past timber management is not a deciding factor in distribution and relative abundance.

Patterns of oak distribution in the community are largely a function of slope, aspect, and associated hydrology. *Quercus alba* is the dominant oak over most of the area with *Q. velutina* dominant to co-dominant in some areas. *Quercus falcata* is common to co-dominant, often in areas of *P. echinata*. Drier ridgetops and south- and west-facing slopes are dominated by *Q. stellata* and *Q. marilandica*. *Quercus muhlenbergii* may be locally common but is generally widely scattered in the study area. *Quercus rubra* may be locally common, particularly on north- and east-facing aspects, but is more common in the mesic north slope hardwood forest.

Common understory species include *Ostrya virginiana*, *Ulmus alata*, *Cornus florida*, *Frangula caroliniana*, *Amelanchier arborea*, *Chionanthus virginicus*, *Aesculus pavia*, *Vaccinium pallidum*, *Rhus aromatica* var. *aromatica*, *Diospyros virginiana*, and *Juniperus virginiana* var. *virginiana*. The herbaceous layer is fairly diverse but is sparse, due to dense shade under the closed canopy and a thick layer of leaf litter on the forest floor.

11) Shortleaf Pine Forest.—This community occupies approximately 8.6 ha and occurs in areas that were fields as recently as the 1950s. It is dominated by *Pinus echinata* with *Juniperus virginiana* var. *virginiana* important to occasionally co-dominant. *Quercus falcata* occurs occasionally in the canopy but more often in the understory. Scattered *Nyssa sylvatica*, *Liquidambar styraciflua*, *Acer rubrum* var. *rubrum*, *Prunus serotina*, *Quercus alba*, and *Q. velutina* occur as understory trees. Stands of this community type do not appear to have been planted, but rather to have arisen from natural regeneration following seedfall from surrounding forest remnants. The canopy in this community is dense and continuous. Woody plants in the understory are typically thick, and the herbaceous layer is sparse.

12) Clearcut.—This community is restricted in the study area to a 0.81 ha area near the southern boundary. It is situated on a gentle south-facing slope. Scattered saplings and shrubs dominate a grassy, early-successional shrubland. A rich diversity of herbaceous species occurs in this community of abundant sunlight. Common species include *Andropogon virginicus*, *Carex hirsutella*, *Dichanthelium* spp., *Lespedeza* spp., *Erechtites hieraciifolius* var. *hieraciifolius*, *Parthenium integrifolium*, *Liatris squarrulosa*, *Cirsium altissimum*, *Conyza canadensis*, *Monarda bradburiana*, *Eupatorium serotinum*, *Solidago odora* subsp. *odora*, *Solidago nemoralis*, *Rudbeckia hirta* var. *pulcherimma*, *Asclepias quadrifolia*, and *Verbascum thapsus*.

13) Riparian Forest.—This community is restricted to the narrow floodplain, adjacent terraces, and valley bottom along the stream near the southern boundary of the study area. The western portion of the stream within the study area has a wide, shallow bedrock-bottom with sandstone glades and springs along both sides. This area has a different flora than the middle part of the stream within the study area, which is wooded with boulders, cobbles, and gravel in the bed. The eastern part of the stream, just before it flows into Greers Ferry Lake, becomes open again with streamside seeps and drops down a 3 m waterfall into the lake.

The forest along the stream is a mix of hardwood species with some *Pinus echinata*. Important hardwood trees include *Acer rubrum* var. *rubrum*, *Liquidambar styraciflua*, *Quercus alba*, *Q. muhlenbergii*, *Q. nigra*, *Fraxinus americana*, and *Platanus occidentalis*. A diverse understory includes *Chionanthus virginicus* (concentrated along the stream banks), *Ilex decidua* var. *decidua*, *Carpinus caroliniana*, *Ostrya virginiana*, *Euonymus americanus*, and *Hypericum hypericoides* subsp. *hypericoides*. The rocky channel of the creek is occupied by *Platanus occidentalis*, *Cornus obliqua*, *Salix caroliniana*, *S. nigra*, and *Amorpha nitens*.

The herbaceous layer is rich and varied along the stream. Important species along the banks include *Coreopsis tripteris*, *Pedicularis canadensis*, *Chasmanthium latifolium*, *Conoclinium coelestinum*, *Krigia biflora* var. *biflora*, *Verbesina helianthoides*, *V. virginica*, *Lobelia cardinalis*, *L. puberula* var. *mineolana*, *Spiranthes cernua*, and *Lycopus rubellus*. Wooded terraces are occupied by *Polystichum acrostichoides*, *Botrychium virginianum*, *Elephantopus carolinianus*, *Podophyllum peltatum*, *Lysimachia lanceolata*, *Carex blanda*, *C. planispicata*, *Bromus pubescens*, and *Dichanthelium* spp. The channel proper is occupied by *Justicia americana*, *Mecardonia acuminata*, *Gratiola virginiana* var. *virginiana*, *Boehmeria cylindrica*, and *Scirpus pendulus*.

14) Mesic North Slope Hardwood Forest.—This community occurs on north-facing slopes and is dominated by *Quercus alba* and *Q. rubra* with *Carya tomentosa*, *Fraxinus americana*, *Acer saccharum* var. *saccharum*, *Acer rubrum* var. *rubrum*, *Morus rubra*, *Prunus serotina*, and *Tilia americana* var. *americana* important, at least locally. *Carya cordiformis* and *C. laciniosa* are restricted to this community in the study area. The understory is diverse and includes *Cornus florida*, *Asimina triloba*, *Staphylea trifolia*, *Carpinus caroliniana*, *Ostrya virginiana*, *Viburnum rufidulum*, *Euonymus americanus*, *Lyonia ligustrina*, *Rhododendron prinophyllum*, *Vaccinium pallidum*, *V. stamineum*, and *Aesculus pavia*.

The herbaceous layer is especially rich and includes a large number of species that are restricted to this habitat type within the study area. Common species are *Phlox divaricata* subsp. *laphamii*, *Polystichum acrostichoides*, *Adiantum pedatum* var. *pedatum*, *Erythronium albidum*, *Cardamine concatenata*, *Geranium maculatum*, and *Trillium recurvatum*. *Uvularia sessilifolia*, *Polygonatum biflorum*, *Cimicifuga racemosa*, *Carex jamesii*, *C. rosea*, *Erythronium rostratum*, *Cystopteris protrusa*, *Osmorhiza longistylis*, *Ageratina altissima* var. *altissima*, *Solidago caesia*, *Campanulastrum americanum*, *Silene stellata*, *Monotropa hypopithys*, *Sanguinaria canadensis*, *Delphinium tricorne*, *Viola pubescens*, and *V. canadensis* var. *canadensis* are among those herbaceous species restricted to this community in the study area.

This community has been reduced in size by the construction of Greers Ferry Lake. The species composition becomes more rich and mesophytic downslope and presumably was even more rich and mesophytic near the bottom of the slope, which is below the present day lake.

Plant Species of Conservation Concern in the Study Area.—Six state species of conservation concern were found in the study area during the 2005 field season. Each of these rare species is actively monitored by the Arkansas Natural Heritage Commission (2005). They are listed below followed by their conservation status ranks (ANHC 2005).

1. *Phemeranthus rugospermus* (rough-seeded fame-flower) G3G4S1 – This species was previously known in the state only from high-quality sand barrens in the Gulf Coastal Plain of southwestern Arkansas. Twelve plants were found growing in a small sandstone glade in the study area. This occurrence represents the first record from the Interior Highlands and is a significant range extension for the species.

2. *Viola canadensis* var. *canadensis* (Canada white violet) G5S2 – Twelve individuals of this species were found growing on the north-facing slope above the lake in a small depression associated with a tip-up mound.

3. *Carex lupuliformis* (false hop sedge) G4S1S2 – This species of is typically found in low, rich, bottomland forests along major rivers in the eastern and southern parts of the state. A single clump of about 20 stems was found growing in the depression wetland. Several smaller clumps were also found in the lowland forest on USACOE land at the northern tip of the peninsula. This occurrence represents the first record from the Interior Highlands and is a significant range extension for the species.

4. *Nemastylis nuttallii* (Nuttall's pleatleaf) G4S2 – This uncommon member of the iris family occurs only in glades and prairies and is limited in distribution to the Interior Highlands (Zollner et al. 2005). There is a population of at least 50 plants in a small, seasonally wet sandstone glade on the north side of the bedrock-bottom stream.

5. *Claytonia caroliniana* (Carolina spring beauty) G5S2S3 – This species is restricted to 2 areas on the property, both shaded seasonally moist areas within sandstone glades and associated woodlands. Fewer than 20 individuals were observed.

6. *Carex swanii* (Swan's sedge) G5S3 – This species is locally common in the depression wetland with more than 100 clumps. It is also found in a small spring-fed drainage in a thin-soiled area and is thinly scattered in the lowland forest.

Other plant species of note -- While the following species are not monitored as elements of special concern by state or federal conservation agencies, their occurrence within the study area is notable because they occur as significantly disjunct populations, edge-of-range occurrences, or are otherwise significant.

1. *Acer rubrum* var. *drummondii* (Drummond's red maple, swamp red maple) – This variety of red maple is typically found in lowland forests and swamp margins in the Mississippi Alluvial Plain and Gulf Coastal Plain ecoregions nith 1988, Witsell pers. obs.). It is found in the study area the upland depression wetland community where it occurs th *Vaccinium fuscatum*, another unusual shrub for the Interior ghlands.

2. *Axonopus furcatus* (big carpetgrass) – This species is pically found in the Gulf Coastal Plain in southern Arkansas here it grows in wet pine flatwoods and open seasonally wet eas. To our knowledge, this is the first documentation of this native grass from the Interior Highlands (Smith 1988, Witsell pers. obs.). This species was locally common in the perennial shoreline grassland community in the study area.

3. *Carex hirsutella* X *Carex swanii* (a hybrid sedge) – Several clumps of this hybrid sedge were found in the pine forest in the study area and were identified by Tyler Smith of the University of Ontario, an expert on *Carex* Sect. *Porocystis*, to which both parent species belong. Recent correspondence with Smith reveals that the results of molecular genetic analysis support the preliminary determination (based on morphological characters) that these plants are a hybrid between the two species. Because the hybrid involves a parent species that is of conservation concern, *Carex swanii*, it too may be of conservation concern.

4. *Isolepis pseudosetacea* (a bulrush) – This species, listed in Smith (1988) as *Scirpus molestus* M. C. Johnst., is apparently uncommon in Arkansas. It is documented by specimens from wet depressions in sandstone glades in Independence, Izard, Stone, and Logan counties and from wet depressions in igneous glades in Saline County. Smith (1988) lists it as reported (without a voucher specimen) from Drew County.

5. *Vaccinium fuscatum* (highbush blueberry) – This species of native blueberry is known in Arkansas almost exclusively from the Gulf Coastal Plain with a few stations in the southern Ouachita Mountains near the boundary with the Gulf Coastal Plain in Garland and Montgomery Counties where it is associated with wooded seepage wetlands (Smith 1988, Marsico 2005) and in Saline County (Witsell pers. obs.). It was also recently found in similar habitat (an upland depression wetland) in Cleburne County at Big Creek Natural Area (Witsell pers. obs.). Together, these two stations represent the only known sites in the Ozarks/Arkansas Valley and represent small populations that are disjunct from the main range of the species. It is interesting to note that upland depression wetlands in the Ozarks and Ouachitas provide habitat for disjunct populations of a number of other species more typical of the Coastal Plain.

Annotated Checklist

The following is a list of all vascular plant taxa documented within the study area. Taxa are arranged alphabetically by family within the four major groups of Pteridophytes, Gymnosperms, Angiosperms (Dicots), and Angiosperms (Monocots). Nomenclature follows the *Checklist of the Vascular Plants of Arkansas* (AVFC 2006). Introduced taxa are indicated by an asterisk (*). Taxa monitored by the Arkansas Natural Heritage Commission as species of conservation concern are listed in **bold** type. Codes for the communities in which taxa were found are listed and correspond to the communities as follows:

CC = clearcut
DW = upland depression wetland
LF = lowland forest
NS = mesic north slope hardwood forest
PF = shortleaf pine forest
PHF = pine-hardwood forest (upland)
PSG = perennial shoreline grassland
RD = roadsides
RI = riparian forest along creek
SG = sandstone glade and associated open woodland
SHB = shale bluffs (calcareous)
SL = shoreline area of Greers Ferry lake
SP = springs and spring runs
SSB = sandstone bluffs

Those taxa with a slash (/) between two codes occur in the transition zone between the two communities. Following the community codes is a number which corresponds to the relative abundance of the taxon within the entire study area, classified as follows:

5 = abundant
4 = common
3 = occasional
2 = infrequent
1 = rare

In many cases, more than one specimen of a taxon was collected by an author or both authors, with each specimen given a separate collection number. Only one voucher number is listed for each taxon except where both authors collected specimens of the same taxa, in which case, one collection number is listed for each author. For taxa that were observed but not collected, the words "not collected" are listed in parentheses. Voucher specimens are deposited in the Herbarium of the South Fork Native Plant Preserve, owned and managed by the Gates Rogers Foundation, Inc.

Pteridophytes

Aspleniaceae

Asplenium platyneuron (L.) Britton, Stearns, & Poggenb.; DW, NS, PF, RD, SG, SSB; 5 (Baker GRF-175)

Asplenium rhizophyllum L.; SHB/NS; 1 (Witsell 05-27, Baker GRF-007)

Asplenium trichomanes L. subsp. *trichomanes*; SSB; 1 (Witsell 05-248)

Dennstaedtiaceae

Pteridium aquilinum (L.) Kuhn in Decken; PHF, RD; 2 (Baker GRF-232)

Dryopteridaceae

Cystopteris protrusa (Weath.) Blasdell; NS; 1 (Baker GRF-176)

Polystichum acrostichoides (Michx.) Schott; NS, PHF, RI; 4 (Baker GRF-144)

Woodsia obtusa (Spreng.) Torr.; NS, RD, SG, SSB; 3 (Baker GRF-177)

Isoetaceae

Isoetes melanopoda Gay & Durieu; RI, SP; 1 (Witsell 05-85)

Ophioglossaceae

Botrychium biternatum (Savigny) Underw.; LF; 3 (Witsell 05-1312)

Botrychium dissectum Spreng. f. *obliquum* (Muhl.) Fernald; LF; 3 (Baker GRF-339)

Botrychium virginianum (L.) Sw.; NS, PHF, RI; 3 (Baker GRF-045)

Ophioglossum pycnostichum (Fernald) Á. Löve & D. Löve; SG/SP; 1 (Witsell 05-244)

Polypodiaceae

Pleopeltis polypodioides (L.) E. G. Andrews & Windham in Windham var. *michauxiana* (Weath.) E. G. Andrews & Windham in Windham; NS, SSB; 3 (Witsell 05-28)

Pteridaceae

Adiantum pedatum L. var. *pedatum*; NS; 2 (Baker GRF-167)

Cheilanthes lanosa (Michx.) D. C. Eaton in Emory; SG; 2 (Baker GRF-019)

Selaginellaceae

Selaginella eclipes W. R. Buck; SP; 1 (Witsell 05-270, Baker GRF-148)

Gymnosperms

Cupressaceae

Juniperus virginiana L. var. *virginiana*; NS, PF, PHF, RD, RI, SG, SSB; 5 (Baker GRF-153)

Pinaceae

Pinus echinata Mill.; CC, PF, PHF, RD, RI, SG, SSB; 5 (Witsell 05-791)

Angiosperms (Dicots)

Acanthaceae

Justicia americana (L.) Vahl; RI, SL; 3 (Witsell 05-528)

Ruellia humilis Nutt. var. *humilis*; RD, SG/RI; 2 (Baker GRF-248)

Ruellia pedunculata Torr. ex A. Gray subsp. *pedunculata*; PHF, SG; 3 (Baker GRF-111)

Aceraceae

Acer negundo L.; LF, RD; 2 (Witsell 05-534)

Acer rubrum L. var. *drummondii* (Hook. & Arn. ex Nutt.) Sarg.; DW; 1 (Baker GRF-068)

Acer rubrum L. var. *rubrum*; CC, LF, NS, PF, RD, RI, SSB; 4 (Baker GRF-234)

Acer saccharinum L.; SL; 2 (Baker GRF-160)

Acer saccharum Marshall var. *saccharum*; NS; 3 (Baker GRF-161)

Altingiaceae

Liquidambar styraciflua L.; DW, LF, NS, PF, PHF, PSG, RD, RI, SL; 4 (Witsell 05-747)

Anacardiaceae

Rhus aromatica Aiton var. *aromatica*; PHF, SG, SSB; 4 (Witsell 05-24, Baker GRF-004)

Rhus copallina L.; CC, RD, SG; 3 (Baker GRF-235)

Rhus glabra L.; RD; 3 (Witsell 05-636)

Toxicodendron radicans (L.) Kuntze; CC, LF, NS, PF, PHF, RD, RI, SSB; 5 (Baker GRF-076)

Annonaceae

Asimina triloba (L.) Dunal; NS; 2 (Baker GRF-086)

Apiaceae

Ammoselinum butleri (Engelm. ex S. Watson) J. M. Coult. & Rose; RD; 1 (Witsell 05-223)

Chaerophyllum tainturieri Hook. var. *tainturieri*; RD; 3 (Baker GRF-037)

**Daucus carota* L.; RD; 1 (Baker GRF-224)

Daucus pusillus Michx.; RD; 2 (Baker GRF-115)

Eryngium prostratum Nutt. ex DC.; LF, PSG, SL; 4 (Witsell 05-749, Baker GRF-186)

Osmorhiza longistylis (Torr.) DC.; NS; 1 (not collected)

Ptilimnium nuttallii (DC.) Britton; RD, SG; 4 (Witsell 05-630, Baker GRF-198)

Sanicula canadensis L.; NS, PHF, RI; 3 (Baker GRF-170)

Spermolepis divaricata (Walter) Raf. ex Ser.; RD; 1 (Witsell 05-523, Baker GRF-211)

Thaspium trifoliatum (L.) A. Gray var. *flavum* Blake; RI, SSB/NS; 2 (Witsell 05-386, Baker GRF-083)

Trepocarpus aethusae Nutt.; LF, RD, SG, SSB; 3 (Baker GRF-182)

Zizia aurea (L.) W. D. J. Koch; PHF, SG; 1 (not collected)

Apocynaceae

Amsonia tabernaemontana Walter; NS, SG/SP; 1 (Baker GRF-046)

Apocynum cannabinum L.; RD; 1 (Baker GRF-098)

Trachelospermum difforme (Walter) A. Gray; LF, PSG, RI, SL; 4 (Baker GRF-187)

Aquifoliaceae

Ilex decidua Walter var. *decidua*; DW, LF, RI; 4 (Witsell 05-267, Baker GRF-222)

Araliaceae

Aralia spinosa L.; NS, RD; 2 (Witsell 05-1304)

Aristolochiaceae

Aristolochia serpentaria L.; NS, PHF; 3 (Witsell 05-254, Baker GRF-074)

Asclepiadaceae

Asclepias quadrifolia Jacq.; CC, NS, PHF; 2 (Witsell 05-275)

Ascelpia tuberosa L. subsp. *interior* Woodson; RD; 1 (not collected)

Asclepias variegata L.; PF, PHF, RD; 2 (Witsell 05-539, Baker GRF-137)

Matelea decipiens (Alexander) Woodson; LF, PHF, RI, SG;

3 (Baker GRF-185)

Asteraceae

Achillea millefolium L.; RD; 1 (Baker GRF-097)
Ageratina altissima (L.) R. M. King & H. Rob. var. *altissima*; NS; 2 (Witsell 05-1367, Baker-331)
Ambrosia artemisiifolia L.; RD, SL; 4 (Witsell 05-1190)
Ambrosia bidentata Michx.; RD, SG, SL, SSB; 4 (Witsell 05-1294)
Antennaria parlinii Fernald subsp. *fallax* (Greene) R. J. Bayer & Stebbins; PHF, RI, SG; 4 (Witsell 05-244, Baker-009)
Bidens aristosa (Michx.) Britton; LF, RD, SL; 3 (Baker GRF-328)
Bidens frondosa L. var. *frondosa*; RI, SL; 2 (Witsell 05-1332)
Boltonia diffusa Elliott; LF, PSG, RI, SG, SL, SSB; 3 (Witsell 05-1199)
Bradburia pilosa (Nutt.) Semple; RD; 2 (Baker GRF-290)
Brickellia eupatorioides (L.) Shinners var. *texana* (Shinners) Shinners; RD; 1 (Witsell 05-1167)
**Carduus nutans* L.; CC, RD; 2 (Baker GRF-163)
Cirsium altissimum (L.) Hill; CC, RI; 2 (Baker GRF-321)
Conoclinum coelestinum (L.) DC.; RI; 1 (not collected)
**Conyza canadensis* (L.) Cronquist; CC, RD; 3 (Baker GRF-285)
Coreopsis grandiflora Hogg ex Sweet; RD, SG, SSB; 3 (Witsell 05-519, Baker GRF-095)
Coreopsis tripteris L.; RI; 2 (Baker GRF-253)
Echinacea purpurea (L.) Moench; RD; 1 (Baker GRF-209)
Eclipta prostrata (L.) L.; RI, SL; 4 (Witsell 05-1187)
Elephantopus carolinianus Raeusch.; LF, PF, RI; 3 (Witsell 05-1162)
Elephantopus tomentosus L.; LF, PHF, RD; 2 (Baker GRF-293)
Erechtites hieraciifolius (L.) Raf. ex DC. var. *hieraciifolius*; CC, RD, SL; 3 (Witsell 05-1193)
Erigeron pulchellus Michx. var. *pulchellus*; PHF; 1 (not collected)
Erigeron strigosus Muhl. ex Willd.; RD, SG/RI; 3 (Witsell 05-507, Baker GRF-112)
Eupatorium serotinum Michx.; CC, RD, RI, SL; 3 (Witsell 05-1168, Baker GRF-351)
Eurybia hemispherica (Alexander) G. L. Nesom; SG/RI; 1 (Baker GRF-319)
**Facelis retusa* (Lam.) Sch. Bip.; CC, RD; 2 (Witsell 05-269)
**Gamochaeta purpurea* (L.) Cabrera; RD; 3 (Baker GRF-032)
Helenium amarum (Raf.) H. Rock var. *amarum*; RD; 2 (Baker GRF-099)
Helenium campestre Small; RD, SG; 3 (Witsell 05-524, Baker GRF-139)
Helenium flexuosum Raf.; LF, PSG; 4 (Baker GRF-219)
Helianthus divaricatus L.; PHF, SG, SSB; 3 (Baker GRF-229)
Hieracium gronovii L.; PHF, RD, SG; 3 (Witsell 05-1328, Baker GRF-353)
Krigia biflora (Walter) S. F. Blake var. *biflora*; NS, RI; 2 (Witsell 05-240)
Krigia cespitosa (Raf.) K. L. Chambers var. *cespitosa*; RD; 3 (Baker GRF-042)
Krigia dandelion (L.) Nutt.; DW, PHF, RD; 3 (Witsell 05-280)
Krigia virginica (L.) Willd.; RD; 3 (Baker GRF-043)
Lactuca sp.; RI; 1 (not collected)
**Leucanthemum vulgare* Lam.; RD, SSB; 1 (Baker GRF-093)
Liatris squarrulosa Michx.; CC, PHF, RD; 1 (Witsell 05-1172)
Packera obovata (Muhl. ex Willd.) W. A. Weber & Á. Löve; DW, PHF, RI, SG, SP; 4 (Baker GRF-050, Witsell 05-259)
Packera tomentosa (Michx.) C. Jeffrey; SG/RI, SG/SP; 1 (Witsell 05-96)
Parthenium integrifolium L.; CC, PHF, RD; 2 (Witsell 05-1293)
Pluchea camphorata (L.) DC.; LF, PSG, SL; 3 (Baker GRF-308)
Prenanthes altissima L.; NS, RI; 2 (not collected)
Pseudognaphalium obtusifolium (L.) Hilliard & B. L. Burtt subsp. *obtusifolium*; CC, RD, SG; 3 (Baker GRF-294)
Rudbeckia hirta L. var. *pulcherrima* Farw.; CC, RD, SSB; 3 (Baker GRF-199)
Solidago buckleyi Torr. & A. Gray; SSB/PHF; 1 (Witsell 05-1176)
Solidago caesia L.; NS; 3 (Baker GRF-329)
Solidago flexicaulis L. vel. aff.; NS, SG/PHF; 1 (Witsell 05-1175)
Solidago hispida Muhl. ex Willd.; NS/RI; 1 (not collected)
Solidago nemoralis Aiton; CC, PHF, RD, SG; 4 (Witsell 05-1311, Baker GRF-352)
Solidago odora Aiton subsp. *odora*; CC, PHF; 2 (Witsell 05-1161)
Solidago sp.; SSB/PHF; 1 (Baker GRF-307)
Solidago ulmifolia Muhl. ex Willd. var. *palmeri* Cronquist; PHF, SSB; 3 (Baker GRF-313)
**Sonchus asper* (L.) Hill; CC, RD; 2 (Baker GRF-162)
Symphyotrichum anomalum (Engelm.) G. L. Nesom; PHF, SSB; 3 (Baker GRF-333)
Symphyotrichum drummondii (Lindl. in Hook.) G. L. Nesom; RD; 2 (Baker GRF-354)
Symphyotrichum dumosum (L.) G. L. Nesom; LF, RI; 3 Baker GRF-338)
Symphyotrichum lateriflorum (L.) Á. Löve & D. Löve; NS, RD; 3 (Witsell 05-1308, Baker GRF-355)
Symphyotrichum patens (Aiton) G. L. Nesom var. *patentissimum* (Lindl. ex DC.) G. L. Nesom; PHF, RD,

SG, SSB; 4 (Baker GRF-330)

Symphyotrichum pilosum (Willd.) G. L. Nesom; RD; 2 (Witsell 05-1330)

Symphyotrichum sagittifolium (Wedemeyer) G. L. Nesom vel. aff.; NS; 1 (Witsell 05-1366)

Symphyotrichum turbinellum (Lindl.) G. L. Nesom; PHF/CC; 1 (Witsell 05-1307)

**Taraxacum officinale* Weber ex F. H. Wigg.; RD; 3 (Baker GRF-041)

Verbesina helianthoides Michx.; RI; 2 (Witsell 05-517)

Verbesina virginica L.; DW, RD, RI, SG/RI; 3 (Witsell 05-1166)

Vernonia baldwinii Torr.; RD, SG; 2 (Baker GRF-246)

Vernonia missurica Raf.; RD, RI/SG; 2 (Baker GRF-318)

Xanthium strumarium L.; SL; 4 (Witsell 05-1191)

Balsaminaceae

Impatiens capensis Meerb.; LF; 1 (not collected)

Berberidaceae

Podophyllum peltatum L.; NS, PHF, RI, SP/SG; 3 (Baker GRF-058)

Betulaceae

Betula nigra L.; LF, SL; 2 (Witsell 05-752)

Carpinus caroliniana Walter; NS, PHF, RI, SSB; 4 (Witsell 05-276, Baker GRF-065)

Ostrya virginiana (Mill.) K. Koch; LF, NS, PHF, RI; 4 (Witsell 05-676)

Bignoniaceae

Bignonia capreolata L.; NS, PHF, RI; 2 (Witsell 05-382)

Campsis radicans (L.) Seem.; LF, RD, RI, SL; 3 (not collected)

Boraginaceae

Cynoglossum virginianum L.; NS, PHF, RI; 3 (Baker GRF-053)

**Heliotropium indicum* L.; SL; 3 (Baker GRF-272)

Myosotis macrosperma Engelm.; RD, SG; 3 (Baker GRF-055)

Brassicaceae

Boechera canadensis (L.) Al-Shehbaz; PHR, SG; 2 (Baker GRF-073)

Boechera laevigata (Muhl. ex Willd.) Al-Shehbaz; SG; 1 (Witsell 05-109)

Cardamine concatenata (Michx.) O. Schwarz; NS, PHF, RI, SG, SP; 4 (Baker GRF-003)

**Cardamine hirsuta* L.; RD, RI; 4 (Baker GRF-001)

Cardamine parviflora L. var. *arenicola* (Britton) O. E. Schultz; SG; 1 (Witsell 05-20)

Cardamine pensylvanica Muhl. ex Willd.; SP; 1 (Baker GRF-015)

Draba brachycarpa Nutt. ex Torr. & A. Gray; RD, SG; 3 (Witsell 05-29, Baker GRF-010)

Lepidium virginicum L. var. *virginicum*; RD, SG; 3 (Baker GRF-108)

Buddlejacaceae

Polypremum procumbens L.; PSG, SL; 3 (Baker GRF-218)

Cactaceae

Opuntia humifusa (Raf.) Raf. var. *humifusa*; SG; 1 (Witsell 05-1391)

Callitrichaceae

Callitriche heterophylla Pursh subsp. *heterophylla*; SP; 1 (Witsell 05-258)

Campanulaceae

Campanulastrum americanum (L.) Small; NS; 1 (Baker GRF-281)

Lobelia cardinalis L.; RI; 2 (Baker GRF-317)

Lobelia inflata L.; LF; 1 (Witsell 05-736)

Lobelia puberula Michx. var. *mineolana* E. Wimm.; RI; 2 (Witsell 05-1194)

Lobelia spicata Lam.; NS, PHF, RD, SP; 3 (Witsell 05-626, Baker GRF-233)

Triodanis perfoliata (L.) Nieuwl. var. *biflora* (Ruiz & Pav.) T. R. Bradley; RD; 2 (Baker GRF-100)

Triodanis perfoliata (L.) Nieuwl. var. *perfoliata*; RD; 2 (Baker GRF-081)

Caprifoliaceae

Lonicera flava Sims.; NS, SSB; 2 (Baker GRF-159)

**Lonicera japonica* Thunb.; LF, NS, PHF, RD, SSB; 3 (Baker GRF-135)

Sambucus nigra L. subsp. *canadensis* (L.) Bolli; NS; 1 (not collected)

Symphoricarpos orbiculatus Moench; NS, PHF, SG; 2 (not collected)

Viburnum rufidulum Raf.; NS; 2 (Baker GRF-228)

Caryophyllaceae

**Cerastium brachypetalum* Pers.; RD, SG; 2 (Witsell 05-88)

Cerastium brachypodum (Engelm. ex A. Gray) B. L. Rob.; RD, SG; 3 (Baker GRF-025)

**Cerastium glomeratum* Thuill.; RD; 3 (Baker GRF-029)

**Cerastium pumilum* Curtis; RD; 3 (Baker GRF-013)

**Dianthus armeria* L. subsp. *armeria*; RD; 2 (Baker GRF-110)

Paronychia fastigiata (Raf.) Fernald; SSB; 1 (Witsell 05-1185, Baker GRF-273)

Sagina decumbens (Elliott) Torr. & A. Gray subsp. *decumbens*; RD; 2 (Baker GRF-024)

Silene stellata (L.) W. T. Aiton in W. Aiton & W. T. Aiton; NS; 2 (Baker GRF-280)

Silene virginica L.; PHF, RI, SG, SSB; 2 (Baker GRF-059)

**Stellaria media* (L.) Vill.; NS, PHF, RD, RI, SP, SSB; 3 (Witsell 05-110)

Celastraceae

Euonymus americanus L.; NS, RI, SSB; 2 (Baker GRF-196)

Celtidaceae

Celtis tenuifolia Nutt.; LF, SG; 3 (Witsell 05-230, Baker GRF-060)

Cistaceae

Lechea tenuifolia Michx.; PHF, RD, SG; 2 (Witsell 05-

1204)

Clusiaceae

Hypercium drummondii (Grev. & Hook.) Torr. & A. Gray; PSG, SG; 2 (Witsell 05-808)

Hypericum gentianoides (L.) Britton, Stearns, & Poggenb.; PSG, SG; 2 (Witsell 05-809)

Hypericum hypericoides (L.) Crantz subsp. *hypericoides*; RI; 3 (Witsell 05-811, Baker GRF-279)

Hypericum hypericoides (L.) Crantz subsp. *multicaule* (Michx. ex Willd.) N. Robson; PHR, RD; 2 (Baker GRF-286)

Hypericum mutilum L.; PSG, RI, SL, SP; 3 (Baker GRF-249)

Hypericum prolificum L.; RD, RI, SG, SP, SSB; 4 (Baker GRF-238)

Hypericum pseudomaculatum Bush in Britton; LF, RD, SG, SP, SSB; 3 (Baker GRF-181)

Hypericum punctatum Lam.; RD; 2 (Baker GRF-237)

Triadenum tubulosum (Walter) Gleason; LF, SL; 2 (Witsell 05-1319)

Triadenum walteri (J. F. Gmel.) Gleason; LF, PSG, RI; 3 (Witsell 05-1317, Baker GRF-314)

Convolvulaceae

Ipomoea pandurata (L.) G. F. W. Mey.; RD, RI; 2 (Baker GRF-284)

Ipomoea lacunosa L.; RI, SL; 3 (Witsell 05-1188, Baker GRF-326)

Cornaceae

Cornus florida L.; NS, PHF, RD; 3 (Witsell 05-105)

Cornus obliqua Raf.; RI; 3 (Witsell 05-533, Baker GRF-201)

Cuscutaceae

Cuscuta compacta Juss. ex Choisy; LF, SL; 2 (Baker GRF-336)

Ebenaceae

Diospyros virginiana L.; LF, PHF, PSG, RD, RI, SL, SSB; 4 (Witsell 05-535, Baker GRF-107)

Ericaceae

Lyonia ligustrina (L.) DC.; DW, NS, SSB; 2 (Baker GRF-089)

Rhododendron prinophyllum (Small) Millais; NS, RI/NS, SSB; 3 (Witsell 05-271)

Vaccinium arboreum Marshall; PHF, RI, SG, SSB; 5 (Witsell 05-509, Baker GRF-104)

Vaccinium fuscatum Aiton; DW; 1 (Baker GRF-063)

Vaccinium pallidum Aiton; NS, PHF, RI; 4 (Baker GRF-017)

Vaccinium stamineum L.; NS, PHF; 4 (Baker GRF-062)

Euphorbiaceae

Acalypha monococca (Engelm. ex A. Gray) Lill. W. Mill. & Gandhi; SG; 1 (Witsell 05-1323)

Acalypha virginica L.; PF, PHF, SL, SSB; 3 (Witsell 05-738, Baker GRF-255)

Chamaesyce maculata (L.) Small; RD, SL; 3 (Baker GRF-244)

**Chamaesyce nutans* (Lag.) Small; RI, SL; 2 (Witsell 05-1196, Baker GRF-315)

Croton glandulosus L. var. *septentrionalis* Müll.-Arg.; RD; 1 (Baker GRF-291)

Croton monanthogynus Michx.; SG/RI; 2 (Baker GRF-258)

Croton willdenowii G. L. Webster; RD, SG, SL; 3 (Baker GRF-216)

Euphorbia commutata Engelm.; RI; 1 (not collected)

Euphorbia corollata L.; SG; 2 (Baker GRF-305)

Phyllanthus caroliniensis Walter var. *caroliniensis*; RI, SL; 2 (Baker GRF-271)

Fabaceae

Amorpha nitens F. E. Boynton; NS, RI, SP/SG, SP; 3 (Baker GRF-078)

Amphicarpaea bracteata (L.) Fernald; NS, RI; 1 (not collected)

Apios americana Medik.; LF, RI, SL; 3 (Baker GRF-269)

Astragalus distortus Torr. & A. Gray var. *engelmannii* (E. Sheld.) M. E. Jones; SG; 1 (Witsell 05-237)

Baptisia bracteata Muhl. ex Willd. var. *leucophaea* (Nutt.) Kartesz & Gandhi; PHF, RI; 2 (Witsell 05-279)

Cercis canadensis L. var. *canadensis*; RD; 2 (Witsell 05-22)

Chamaecrista nictitans (L.) Moench var. *nictitans*; PSG, RD, SL; 3 (Witsell 05-1186, Baker GRF-262)

Clitoria mariana L.; PHF; 3 (Baker GRF-302)

Crotalaria sagittalis L.; PSG; 1 (Witsell 05-627)

Desmodium laevigatum (Nutt.) DC.; NS/SL, RD; 3 (Witsell 05-1182)

Desmodium marilandicum (L.) DC.; RD; 3 (Witsell 05-1373, Baker-301)

Desmodium nudiflorum (L.) DC.; NS, PF, PHF; 3 (not collected)

Desmodium paniculatum (L.) DC.; LF, PHF; 2 (not collected)

Desmodium rotundifolium DC.; PHF; 2 (Witsell 05-1300)

Desmodium viridiflorum (L.) DC.; RD; 2 (Baker GRF-322)

Galactia regularis (L.) Britton, Stearns, & Poggenb.; PHF, SG; 1 (not collected)

Gleditsia triacanthos L.; LF; 2 (Witsell 05-1305)

**Kummerowia striata* (Thunb.) Schindl.; RD; 3 (Baker GRF-275)

**Lespedeza bicolor* Turcz.; RD; 1 (not collected)

**Lespedeza cuneata* (Dum. Cours.) G. Don; LF, RD, RI, SG, SL; 4 (Baker GRF-295)

Lespedeza hirta (L.) Hornem. var. *hirta*; PHF, RD, SG; 3 (Baker GRF-334)

Lespedeza intermedia (S. Watson ex A. Gray) Britton; PHF, SG; 1 (Witsell 05-1327)

Lespedeza procumbens Michx.; CC, PHF, SG; 3 (Witsell 05-1195)

Lespedeza repens (L.) W. P. C. Barton; CC, PHF, RD; 3

(Baker GRF-261)

Lespedeza stuevei Nutt.; RD; 2 (Baker GRF-300)

Lespedeza virginica (L.) Britton; RD, SG; 2 (Witsell 05-1180, Baker GRF-320)

Mimosa quadrivalvis L. var. *nuttallii* (DC.) Beard ex Barneby; SG; 1 (Witsell 05-366)

Orbexilum pedunculatum (Mill.) Rydb. var. *pedunculatum*; RI, SG; 1 (Witsell 05-550)

Rhynchosia latifolia Nutt. ex Torr. & A. Gray; PHF; 1 (not collected)

Senna marilandica (L.) Link; RD, RI; 1 (not collected)

Strophostyles umbellata (Muhl. ex Willd.) Britton; RD; 3 (Baker GRF-292)

Stylosanthes biflora (L.) Britton, Stearns, & Poggenb.; RD, SG; 2 (Baker GRF-114)

Tephrosia virginiana (L.) Pers.; PHF, RD, SSB; 3 (Baker GRF-171)

**Trifolium campestre* Schreb.; RD; 2 (Baker GRF-033)

**Trifolium dubium* Sibth.; RD; 2 (Witsell 05-227)

Trifolium reflexum L.; RD; 1 (Baker GRF-082)

**Trifolium repens* L.; RD; 2 (Baker GRF-101)

Vicia minutiflora F. Dietr.; RI, SG, SP; 2 (Witsell 05-111)

**Vicia sativa* L.; RD; 2 (Witsell 05-98, Baker GRF-094)

Fagaceae

Quercus alba L.; NS, PF, PHF, RI; 5 (Witsell 05-1376)

Quercus falcata Michx.; CC, NS, PF, PHF; 4 (Witsell 05-1377)

Quercus marilandica Münchh.; PHF, SG; 4 (Witsell 05-792)

Quercus muhlenbergii Engelm.; PHF, RD, RI, SSB; 3 (Witsell 05-1184)

Quercus nigra L.; DW, LF, RI; 4 (Witsell 05-745)

Quercus phellos L.; LF; 3 (Witsell 05-755)

Quercus rubra L.; NS, PHF; 4 (Witsell 05-793)

Quercus stellata Wangenh.; PHF, SG, SSB; 4 (Witsell 05-794)

Quercus velutina Lam. in Lam. et al.; CC, PF, PHF; 4 (not collected)

Gentianaceae

Sabatia angularis (L.) Pursh; LF, RD, RI, SG; 2 (Witsell 05-740)

Geraniaceae

Geranium carolinianum L. var. *carolinianum*; RD; 2 (Baker GRF-088)

Geranium maculatum L.; NS; 2 (Witsell 05-238)

Hippocastanaceae

Aesculus pavia L.; NS, PHF, RD, SP; 4 (Witsell 05-101, Baker GRF-040)

Hydrangeaceae

Hydrangea arborescens L.; NS/SSB; 1 (not collected)

Philadelphus pubescens Loisel.; NS/SSB; 1 (Witsell 05-634)

Hydrophyllaceae

Phacelia hirsuta Nutt.; RI/SG; 2 (Witsell 05-234)

Juglandaceae

Carya cordiformis (Wangenh.) K. Koch; NS; 2 (Baker GRF-358)

Carya laciniosa (F. Michx.) Loudon; NS/SL; 2 (Baker GRF-288)

Carya texana Buckley; PHF, RD, SG; 4 (Baker GRF-287)

Carya tomentosa (Poir.) Nutt.; NS, PHF; 4 (Baker GRF-344)

Juglans nigra L.; NS, RD, RI; 2 (Witsell 05-651)

Lamiaceae

Cunila origanoides (L.) Britton; CC, NS, PHF, SSB; 3 (Baker GRF-304)

Hedeoma hispida Pursh; RD; 3 (Witsell 05-393, Baker GRF-113)

Lycopus rubellus Moench.; DW, RI, SP; 3 (Witsell 05-1177, Baker GRF-316)

Monarda bradburiana L. C. Beck; CC, PHF, RD; 3 (Witsell 05-273, Baker GRF-087)

Monarda fistulosa L. var. *fistulosa*; NS, PHF, RD; 3 (Baker GRF-230)

**Perilla frutescens* (L.) Britton; LF, RD, RI, SL; 3 (Witsell 05-1170)

Prunella vulgaris L. subsp. *lanceolata* (W. P. C. Barton) Hultén; RD, RI, SP; 3 (Witsell 05-406, Baker GRF-116)

Pycnanthemum albescens Torr. & A. Gray; PHF, RD, RI; 3 (Baker GRF-240)

Pycnanthemum tenuifolium Schrad.; CC, DW, RD, RI, SG; 4 (Witsell 05-682, Baker GRF-231)

Salvia lyrata L.; RD, RI, SP; 3 (Baker GRF-035)

Scutellaria elliptica Muhl. ex Spreng. var. *elliptica*; PHF, RI; 3 (Witsell 05-540, Baker GRF-145)

Scutellaria ovata Hill; NS, PHF, RD, RI; 3 (Baker GRF-180)

Scutellaria parvula Michx. var. *australis* Fassett; SG, SP; 3 (Witsell 05-368, Baker-051)

Stachys tenuifolia Willd. var. *tenuifolia*; LF; 2 (Baker GRF-215)

Teucrium canadense L. var. *canadense*; RD; 1 (Witsell 05-680, Baker GRF-225)

Trichostema dichotomum L.; SG, SL, SSB; 2 (Baker GRF-325)

Lauraceae

Sassafras albidum (Nutt.) Nees; PHF, RD; 3 (Baker GRF-221)

Linaceae

Linum medium (Planch.) Britton var. *texanum* (Planch.) Fernald; RD, SG; 2 (Witsell 05-633)

Linum striatum Walter; LF, PSG; 2 (Witsell 05-628, Baker GRF-217)

Loganiaceae

Spigelia marilandica L.; RI, SP; 2 (Witsell 05-514, Baker GRF-141)

Lythraceae

Ammannia Xcoccinea Rottb.; SL; 3 (Witsell 05-802)
Rotala ramosior (L.) Koehne; SL; 4 (Witsell 05-737, Baker GRF-306)
Malvaceae
Hibiscus moscheutos L. subsp. *lasiocarpos* (Cav.) O. J. Blanch.; PSG, SL; 3 (Witsell 05-805, Baker GRF-278)
Melastomataceae
Rhexia mariana L. var. *mariana*; PSG; 1 (Witsell 05-743)
Rhexia virginica L.; PSG; 1 (Witsell 05-733)
Meliaceae
**Melia azedarach* L.; CC, RD; 1 (Witsell 05-547)
Menispermaceae
Cocculus carolinus (L.) DC.; RD, RI, SG; 3 (not collected)
Molluginaceae
Mollugo verticillata L.; RI/SL; 2 (Baker GRF-270)
Monotropaceae
Monotropa hypopithys L.; NS; 1 (Witsell 05-654)
Moraceae
Morus rubra L.; LF, NS; 2 (Witsell 05-758)
Nyssaceae
Nyssa sylvatica Marshall; PF, PHF; 4 (Witsell 05-757)
Oleaceae
Chionanthus virginicus L.; PHF, RI, SG, SSB; 4 (Baker GRF-066)
Fraxinus americana L.; NS, PHF, RI, SG; 3 (Baker GRF-184)
Fraxinus pennsylvanica Marshall; LF, SL; 3 (Witsell 05-228, Baker GRF-064)
**Ligustrum sinense* Lour.; LF, RD, RI; 2 (Witsell 05-526, Baker GRF-152)
Onagraceae
Gaura longiflora Spach; NS/SL; 1 (Witsell 05-1372)
Ludwigia alternifolia L.; RI, SL; 2 (not collected)
Ludwigia decurrens Walter; SL; 3 (Witsell 05-1324)
Ludwigia glandulosa Walter; PSG; 1 (Witsell 05-734)
Oenothera biennis L.; SL; 1 (not collected)
Oenothera fruticosa L.; SG; 2 (Witsell 05-365)
Oenothera laciniata Hill; RD; 2 (Baker GRF-117)
Oenothera linifolia Nutt.; SG; 2 (Witsell 05-363, Baker GRF-142)
Oxalidaceae
Oxalis dillenii Jacq.; RD; 2 (Baker GRF-048)
Oxalis violacea L.; PHF, SG, SSB; 3 (Witsell 05-94)
Papaveraceae
Sanguinaria canadensis L.; NS; 1 (Witsell 05-103)
Passifloraceae
Passiflora incarnata L.; RD, RI; 1 (Witsell 05-1375)
Passiflora lutea L.; DW, LF, RI; 3 (Witsell 05-1192)
Phytolaccaceae
Phytolacca americana L. var. *americana*; PHF, SL; 2 (Baker GRF-327)
Plantaginaceae
Plantago aristata Michx.; RD; 3 (Baker GRF-207)
Plantago elongata Pursh; RD; 3 (Baker GRF-027)
**Plantago lanceolata* L.; RD; 2 (Baker GRF-118)
Plantago virginica L.; RD, SG; 3 (Baker GRF-028)
Platanaceae
Platanus occidentalis L.; NS, RI, SL; 3 (Witsell 05-384)
Polemoniaceae
Phlox divaricata L. subsp. *laphamii* (A. W. Wood) Wherry; NS; 2 (Witsell 05-104)
Phlox pilosa L. subsp. *pilosa*; PHF, RD; 2 (Witsell 05-274)
Polygalaceae
Polygala sanguinea L.; SG; 1 (Witsell 05-629)
Polygonaceae
Brunnichia ovata (Walter) Shinners; LF, SL, SSB; 3 (Witsell 05-751)
Fallopia scandens (L.) Holub; LF; 1 (not collected)
Persicaria hydropiperoides (Michx.) Small; LF, RD, SL; 2 (Witsell 05-1314, Baker-283)
Persicaria lapathifolia (L.) A. Gray; RD, RI, SL; 2 (Witsell 05-1302)
**Persicaria longiseta* (Bruijn) Kitagawa; SL; 2 (Baker GRF-347)
Persicaria pensylvanica (L.) M. Gómez; SL; 2 (Witsell 05-1315)
Persicaria punctata (Elliott) Small; SL; 2 (Witsell 05-1181)
Polygonum tenue Michx.; SG; 1 (Witsell 05-1325)
Rumex crispus L.; RD; 2 (Baker GRF-103)
Rumex hastatulus Baldwin; SG; 2 (Witsell 05-268, Baker GRF-147)
Portulacaceae
***Claytonia caroliniana* Michx.**; SG; 1 (Witsell 05-113)
Claytonia virginica L. ; DW, PHF, RD, RI, SG; 4 (Witsell 05-91, Baker GRF-014)
***Phemeranthus rugospermus* (Holz.) Kiger**; SG; 1 (Witsell 05-1183)
Primulaceae
Lysimachia lanceolata Walter; RI; 2 (Baker GRF-202)
Ranunculaceae
Anemone virginiana L. var. *virginiana*; PHF, SG; 3 (not collected)
Cimicifuga racemosa (L.) Nutt.; NS; 2 (not collected)
Clematis reticulata Walter; NS, SG/SP; 2 (Witsell 05-635)
Delphinium carolinianum Walter subsp. *carolinianum*; RD, SG; 2 (Baker GRF-140)
Delphinium tricorne Michx.; NS; 2 (Witsell 05-231)
Ranunculus abortivus L.; RI; 1 (Witsell 05-226)
Ranunculus fascicularis Muhl. ex J. M. Bigelow; PHF, SG; 2 (Baker GRF-057)
Ranunculus harveyi (A. Gray) Britton var. *harveyi*; NS; 2 (Witsell 05-84, Baker-021)
Ranunculus micranthus Nutt. in Torr. & A. Gray; NS, PHF; 2 (Witsell 05-239)
**Ranunculus sardous* Crantz; RI; 1 (Witsell 05-265)
Thalictrum thalictroides (L.) A. J. Eames & B. Biovin; DW, NS, PHF, RI, SG; 5 (Baker GRF-006)

Rhamnaceae
Berchemia scandens (Hill) K. Koch; LF, RI, SSB; 2 (Baker GRF-200)
Ceanothus americanus L.; RD, RI, SG; 2 (Witsell 05-537)
Frangula caroliniana (Walter) A. Gray; NS, PHF, RD; 3 (Witsell 05-546, Baker-105)
Rosaceae
Agrimonia rostellata Wallr.; NS, RI; 3 (not collected)
Amelanchier arborea (F. Michx.) Fernald; PHF, SSB; 3 (Witsell 05-21)
Crataegus intricata Lange; PHF; 2 (Baker GRF-061)
Crataegus uniflora Münchh.; PHF, SG; 3 (Witsell 05-253, Baker GRF-214)
Geum canadense Jacq.; NS; 3 (not collected)
Potentilla simplex Michx.; RD, RI, SG; 3 (Baker GRF-047)
Prunus americana Marshall; PHF; 2 (Witsell 05-23, Baker GRF-020)
Prunus mexicana S. Watson; RI; 1 (Witsell 05-518, Baker GRF-154)
Prunus serotina Ehrh.; CC, NS, PF, PHF, SG, SSB; 4 (Witsell 05-1374)
Rosa carolina L.; SG; 1 (Witsell 05-530, Baker GRF-136)
Rubus argutus Link; RD; 3 (Baker GRF-085)
Rubus flagellaris Willd.; RD; 3 (Baker GRF-054)
Rubiaceae
Cephalanthus occidentalis L.; PSG, RI, SL: 4 (Witsell 05-677)
**Cruciata pedemontana* (Bell.) Ehrend.; RD; 2 (Witsell 05-247)
Diodia teres Walter; PSG, RD, SG, SL; 3 (Baker GRF-256)
Diodia virginiana L.; PSG, RI, SL; 3 (Baker GRF-190)
Galium aparine L.; NS, RD, RI; 2 (Baker GRF-052)
Galium circaezans Michx.; NS; 3 (Witsell 05-538, Baker GRF-169)
Galium concinnum Torr. & A. Gray; NS; 3 (Witsell 05-631, Baker GRF-213)
Galium pilosum Aiton; NS, PHF, RD; 3 (Baker GRF-236)
Hedyotis australis Lewis & Moore; RD; 1 (Witsell 05-92)
Hedyotis caerulea (L.) Hook.; DW, RD, RI, SG; 3 (Witsell 05-99)
Hedyotis crassifolia Raf.; RD; 3 (Baker GRF-011)
Hedyotis longifolia (Gaertn.) Hook. var. *longifolia*; PHF, SG; 3 (Witsell 05-242)
Hedyotis purpurea (L.) Torr. & A. Gray; RI; 2 (Witsell 05-512, Baker GRF-143)
Mitchella repens L.; DW, NS, RI, SSB, SP; 3 (Baker GRF-151)
Oldenlandia boscii (DC.) Chapm.; SL; 4 (Witsell 05-735, Baker GRF-340)
**Sherardia arvensis* L.; RD, RI; 2 (Baker GRF-023)
Salicaceae
Salix caroliniana Michx.; RI; 3 (Witsell 05-226, Baker GRF-077)
Salix nigra Marshall; RI/RD, SL; 1 (Baker GRF-226)
Santalaceae
Comandra umbellata (L.) Nutt. subsp. *umbellata*; PHF; 2 (Witsell 05-232)
Sapotaceae
Sideroxylon lanuginosum Michx.; NS, PHF, SG; 3 (Baker GRF-164)
Saxifragaceae
Heuchera americana L. var. *americana*; NS, SG, SSB; 3 (Baker GRF-056)
Heuchera americana L. var. *hirsuticaulis* (Wheelock) Rosend., Butters, & Lakela; NS/SSB; 2 (Baker GRF-172)
Saxifraga palmeri Bush; SG, SP, SSB; 3 (Witsell 05-19, Baker GRF-008)
Scrophulariaceae
Agalinis tenuifolia (Vahl) Raf.; SG, SL/SSB; 2 (Witsell 05-1179, Baker GRF-323)
Aureolaria flava (L.) Farw.; PHF, RI; 2 (not collected)
Gratiola virginiana L. var. *virginiana*; RI; 2 (Witsell 05-364)
Lindernia dubia (L.) Pennell; RI, SL; 2 (Baker GRF-250)
Mecardonia acuminata (Walter) Small; PSG, RI, RI/SG; SSB/SL; 3 (Witsell 05-810)
Nuttallanthus texanus (Scheele) D. A. Sutton; RD; 2 (Witsell 05-235)
Pedicularis canadensis L.; RI; 2 (Witsell 05-95)
Penstemon arkansanus Pennell; SG/RI, SSB; 2 (Baker GRF-080)
Penstemon digitalis Nutt. ex Sims; RD; 2 (Witsell 05-549)
Penstemon tubaeflorus Nutt.; RD, SG/RI; 3 (Baker GRF-138)
**Verbascum thapsus* L.; CC, RD, SL; 2 (Baker GRF-239)
**Veronica arvensis* L.; RD; 3 (Baker GRF-030)
Veronica peregrina L. subsp. *peregrina*; RD; 3 (Baker GRF-026)
Simaroubaceae
**Ailanthus altissima* (Mill.) Swingle; CC, RD; 1 (Witsell 05-536)
Solanaceae
Physalis angulata L.; SL; 2 (Witsell 05-1371)
Physalis heterophylla Nees; RD; 1 (Witsell 05-222)
Physalis pubescens L.; RI, SL; 3 (Witsell 05-1369, Bake GRF-266)
Solanum carolinense L.; RD; 3 (Witsell 05-541, Bake GRF-096)
Staphyleaceae
Staphylea trifolia L.; NS; 2 (Witsell 05-653)
Styracaceae
Styrax grandifolius Aiton; SSB/PHF; 2 (Witsell 05-278, Baker GRF-195)
Thymelaeaceae
Dirca palustris L.; PHF; 1 (Witsell 05-107)

Tiliaceae

Tilia americana L. var. *americana*; NS; 3 (Witsell 05-637, Baker GRF-165)

Ulmaceae

Ulmus alata Michx.; CC, RD, PHF, SG, SSB; 4 (Baker GRF-106)

Ulmus americana L.; SSB; 2 (Baker GRF-223)

Urticaceae

Boehmeria cylindrica (L.) Sw.; RI, SL; 3 (Baker GRF-252)

Parietaria pensylvanica Muhl. ex Willd.; NS/SSB; 2 (Baker GRF-173)

Valerianaceae

Valerianella radiata (L.) Dufr.; RD, RI, SG; 3 (Baker GRF-031)

Verbenaceae

Callicarpa americana L.; PHF, RD; 3 (Witsell 05-1378, Baker GRF-357)

Glandularia canadensis (L.) Nutt.; RD, SG; 2 (Witsell 05-26)

Verbena urticifolia L.; RD, RI; 2 (Witsell 05-681, Baker GRF-247)

Violaceae

Hybanthus concolor (T. F. Forst.) Spreng.; NS/SSB; 1 (Witsell 05-245)

Viola bicolor Pursh; RD; 3 (Baker GRF-012)

Viola canadensis* L. var. *canadensis; NS; 1 (Baker GRF-084)

Viola palmata L.; PHF, RD; 3 (Witsell 05-241)

Viola pedata L.; PHF, SG, SSB; 3 (Baker GRF-016)

Viola pubescens Aiton; NS; 1 (Witsell 05-102)

Viola sagittata Aiton; RI, SG; 2 (Witsell 05-100)

Viola sororia Willd.; NS, RD, RI; 3 (Witsell 05-87)

Vitaceae

Parthenocissus quinquefolia (L.) Planch.; NS, PHF, RD, RI; 4 (not collected)

Vitis aestivalis Michx.; NS, RD, RI; 3 (Witsell 05-796)

Vitis rotundifolia Michx.; CC, NS, PHF, RD, RI; 3 (Baker GRF-183Witsell 05-1309,)

Angiosperms (Monocots)

Agavaceae

Camassia scilloides (Raf.) Cory; SG; 1 (Baker GRF-049)

Manfreda virginica (L.) Salisb. ex Rose; SG/RI; 1 (Witsell 05-1198)

Alliaceae

Allium canadense L. var. *canadense*; RI; 1 (Baker GRF-146)

Allium canadense L. var. *mobilense* (Regel) Ownbey; RI/SG; 1 (Witsell 05-529)

**Allium vineale* L.; LF, RD; 2 (Baker GRF-109)

Nothoscordum bivalve (L.) Britton in Britton & A. Br.; SG; 3 (Witsell 05-89, Baker GRF-022)

Amaryllidaceae

**Narcissus Xodorus* L.; RD/NS; 1 (Witsell 05-25)

**Narcissus pseudonarcissus* L.; RD/NS; 1 (Baker GRF-359)

Araceae

Arisaema dracontium (L.) Schott in Schott & Endl.; LF, RI, SP/SG; 1 (Baker GRF-166)

Colchicaceae

Uvularia sessilifolia L.; NS; 1 (not collected)

Commelinaceae

**Commelina communis* L.; RI, SL; 2 (Baker GRF-267)

Tradescantia ohiensis Raf.; SG; 2 (not collected)

Cyperaceae

Carex albicans Willd. ex Spreng.; NS, PHF, SSB; 3 (Witsell 05-218)

Carex albolutescens Schwein.; DW; 1 (Witsell 05-375)

Carex blanda L. H. Dewey; NS, RD, RI; 4 (Witsell 05-216, Baker GRF-069)

Carex bushii Mack.; SG/RI; 2 (Witsell 05-385, Baker GRF-092)

Carex cephalophora Muhl. ex Willd.; NS, PHF, RD; 3 (Witsell 05-370, Baker GRF-044)

Carex complanata Torr. & Hook.; DW; 2 (Witsell 05-377)

Carex digitalis Willd.; LF; 1 (Witsell 05-367)

Carex festucacea Schkuhr ex Willd.; DW; 2 (Witsell 05-396)

Carex glaucodea Tuck. ex Olney; DW, NS, RD, RI; 3 (Witsell 05-380, Baker GRF-036)

Carex hirsutella Mack.; CC, DW, LF, PHF, RD, RI, SG; 5 (Baker GRF-120)

Carex hirsutella Mack. X *swanii* (Fernald) Mack.; PF; 1 (Witsell 05-548) [Det. by T. Smith of U. of Ontario]

Carex intumescens Rudge; LF; 2 (Witsell 05-217)

Carex jamesii Schwein.; NS; 1 (Witsell 05-374)

Carex leavenworthii L. H. Dewey; NS, RD, SG; 3 (Witsell 05-371)

***Carex lupuliformis* Sartwell ex L. H. Dewey**; DW, LF; 2 (Witsell 05-372)

Carex lupulina Willd.; LF, PSG; 3 (Witsell 05-744, Baker GRF-191)

Carex muehlenbergii Schkuhr ex Willd. var. *enervis* Boott; PF, PHF, RD, RI; 3 (Witsell 05-255, Baker GRF-034)

Carex oklahomensis Mack.; RI/SG/SP; 1 (Witsell 05-510, Baker GRF-210)

Carex oligocarpa Willd.; NS, PHF, RD, SSB; 3 (Witsell 05-252)

Carex oxylepis Torr. & Hook.; PF; 1 (Witsell 05-373)

Carex planispicata Naczi; NS, PHF, RD, RI; 3 (Witsell 05-221)

Carex rosea Schkuhr ex Willd.; NS; 2 (Witsell 05-369, Baker GRF-178)

Carex molestiformis Reznicek & P. E. Rothrock; RD; 1 (Baker GRF-121)

***Carex swanii* (Fernald) Mack.**; DW, PF, PHF; 2 (Witsell 05-250)

Carex tribuloides Walenb.; DW, LF; 3 (Witsell 05-543)
Carex typhina Michx.; LF, PSG; 2 (Witsell 05-644, Baker GRF-192)
Carex umbellata Schkuhr ex Willd.; PHF, SSB; 2 (Witsell 05-220)
Carex vulpinoidea Michx.; DW, RD, RI; 2 (Witsell 05-389)
Cyperus echinatus (L.) A. W. Wood; RD; 2 (Witsell 05-1163, Baker GRF-212)
Cyperus erythrorhizos Muhl.; SL; 2 (Witsell 05-1333)
**Cyperus esculentus* L.; SL; 3 (Witsell 05-1316, Baker GRF-337)
Cyperus pseudovegetus Steud.; PSG; 3 (Baker GRF-188)
Cyperus retroflexus Buckley; RD; 1 (Witsell 05-1174)
Cyperus retrorsus Chapm.; NS, SL; 2 (Witsell 05-1368)
Cyperus squarrosus L.; PSG, SL; 3 (Witsell 05-746, Baker GRF-310)
Cyperus strigosus L.; SG/RI; 2 (Baker GRF-265)
Eleocharis acicularis (L.) Roem. & Schult. in Roem. et al.; SL; 4 (Witsell 05-801)
Eleocharis lanceolata Fernald; RD; 3 (Witsell 05-648, Baker GRF-208)
Eleocharis palustris (L.) Roem. & Schult. in Roem. et al.; RI/SG; 1 (Witsell 05-645)
Eleocharis tenuis (Willd.) Schult. var. *verrucosa* (Svenson) Svenson; DW, RI/SG, SP; 3 (Witsell 05-647)
Fimbristylis autumnalis (L.) Roem. & Schult. in Roem. et al.; PSG, SL; 3 (Witsell 05-803, Baker GRF-311)
Fimbristylis vahlii (Lam.) Link; SL; 4 (Witsell 05-741, Baker GRF-309B)
Isolepis pseudosetacea (Daveau) Gand.; RI/SG; 2 (Witsell 05-86, Baker GRF-070)
Lipocarpha micrantha (Vahl) G. C. Tucker; PSG, SL; 4 (Witsell 05-739, Baker GRF-309A)
Rhynchospora capitellata (Michx.) Vahl; SG/RI; 2 (Baker GRF-277)
Rhynchospora recognita (Gale) Kral; DW, RI/SG; 3 (Witsell 05-639, Baker GRF-150)
Schoenoplectus pungens (Vahl) Palla; PSG; 1 (Witsell 05-798)
Scirpus cyperinus (L.) Kunth; RD; 1 (Baker GRF-263)
Scirpus georgianus R. M. Harper; DW, RI; 2 (Witsell 05-642)
Scirpus pendulus Muhl.; RI; 2 (Witsell 05-511)
Scleria oligantha Michx.; DW, NS, PF, PHF, SG; 4 (Baker GRF-158)
Dioscoreaceae
**Dioscorea polystachya* Turcz.; RI; 3 (Witsell 05-797)
Dioscorea villosa L.; DW, NS, PHF, SSB; 3 (Witsell 05-679, Baker GRF-168)
Hemerocallidaceae
**Hemerocallis fulva* (L.) L.; RD; 1 (not collected)
Hyacinthaceae
**Ornithogalum umbellatum* L.; SG/SP; 1 (not collected)
Hypoxidaceae
Hypoxis hirsuta (L.) Coville; RI/SG, SG; 4 (Witsell 05-9)
Iridaceae
**Belamcanda chinensis* (L.) DC. in Redouté; PHF; 1 (not collected)
Iris cristata Sol. ex Aiton; NS, RD; 3 (Witsell 05-236)
***Nemastylis nuttallii* Pichering ex R. C. Foster**; RI/SG; 1 (Witsell 05-632)
Sisyrinchium angustifolium Mill.; PHF, RD, RI; 2 (Witsell 05-233)
Sisyrinchium langloisii Greene; PHF, RD, SG; 2 (Witsell 05-277)
Juncaceae
Juncus acuminatus Michx.; RI; 2 (Baker GRF-206)
Juncus brachycarpus Engelm. in A. Gray; RI; 2 (not collected)
Juncus debilis A. Gray; DW, RI, SP; 2 (Baker GRF-149)
Juncus dudleyi Wiegand; RI; 2 (Baker GRF-203)
Juncus effusus L.; DW, RD, RI; 3 (Witsell 05-649, Baker GRF-251)
Juncus marginatus Rostk.; RI; 1 (Witsell 05-640)
Juncus secundus P. Beauv. ex Poir. in Lam. et al.; RI; 3 (Witsell 05-641)
Juncus tenuis Willd.; RD; 3 (Baker GRF-134)
Juncus validus Coville var. *validus*; SL; 1 (Witsell 05-754)
Luzula echinata (Small) F. J. Herm.; NS, SSB; 4 (Baker GRF-018)
Liliaceae
Erythronium albidum Nutt.; NS, PHF, SG, SP; 4 (Baker GRF-002)
Erythronium rostratum W. Wolf; NS; 1 (Baker GRF-005)
Melanthiaceae
Trillium recurvatum Beck; NS, PHF, SP/SG; 3 (Witsell 05-106)
Orchidaceae
Liparis liliifolia (L.) Rich. ex Lindl.; DW; 1 (Baker GRF-071)
Malaxis unifolia Michx.; DW; 1 (Baker GRF-072)
Spiranthes cernua (L.) Rich.; DW, RI, SP; 3 (Baker GRF-345)
Spiranthes tuberosa Raf.; PHF, RI/SG; 2 (Witsell 05-1178 Baker GRF-303)
Tipularia discolor (Pursh) Nutt.; PHF, SG; 2 (Witsell 05-243)
Poaceae
Agrostis elliottiana Schult.; RI/SG; 1 (Witsell 05-407)
Agrostis hyemalis (Walter) Britton, Stearns, & Poggenb. RD, RI, SG; 4 (Witsell 05-399, Baker GRF-132)
Agrostis perennans (Walter) Tuck.; DW, RI, SP; 2 (Witsel 05-1189)
**Aira elegans* Willd. ex Kunth; RD; 4 (Witsell 05-398 Baker GRF-102)
Andropogon ternarius Michx. var. *ternarius*; RD, SG; 2 (Witsell 05-1306, Baker GRF-356)

Andropogon virginicus L.; CC, LF, PHF, RD, SG; 4 (Witsell 05-1303)

Aristida dichotoma Michx. var. *curtissii* A. Gray; SG; 2 (Witsell 05-1301)

Aristida purpurascens Poir.; SG; 2 (Witsell 05-1197)

Axonopus furcatus (Flüggé) Hitchc.; PSG; 3 (Baker GRF-342)

**Bromus commutatus* Schrad; RD; 3 (Witsell 05-522)

**Bromus hordeaceus* L. subsp. *hordeaceus*; RD; 4 (Witsell 05-403, Baker GRF-128)

Bromus pubescens Muhl. ex Willd.; NS, RD, RI; 4 (Witsell 05-655, Baker GRF-125)

**Bromus racemosus* L.; RD; 3 (Witsell 05-281)

Chasmanthium latifolium (Michx.) H. O. Yates; RI, SP; 4 (Witsell 05-795, Baker GRF-276)

Cinna arundinacea L.; RD; 1 (not collected)

**Dactylis glomerata* L.; CC, RD; 3 (Witsell 05-264)

Danthonia spicata (L.) P. Beauv. ex Roem. & Schult.; CC, PHF, SG; 3 (Witsell 05-391)

Dichanthelium acuminatum (Sw.) Gould & C. A. Clark subsp. *acuminatum*; SG; 2 (Witsell 05-516)

Dichanthelium boscii (Poir.) Gould & C. A. Clark; LF, NS, PHF, RD, RI; 4 (Witsell 05-394, Baker GRF-127)

Dichanthelium commutatum (Schult.) Gould; NS, PHF, RI; 4 (Witsell 05-395, Baker GRF-075)

Dichanthelium depauperatum (Muhl.) Gould; PHF, RD, SG; 3 (Witsell 05-388, Baker GRF-122)

Dichanthelium dichotomum (L.) Gould subsp. *dichotomum*; LF, PHF, RD, SG, SL; 4 (Witsell 05-646, Baker GRF-133)

Dichanthelium dichotomum (L.) Gould subsp. *microcarpon* (Muhl. ex Elliott) Freckmann & Lelong; RI; 2 (Baker GRF-204)

Dichanthelium dichotomum (L.) Gould subsp. *nitidum* (Lam.) Freckmann & Lelong; RI; 1 (Witsell 05-542)

Dichanthelium laxiflorum (Lam.) Gould; PHF, RD, SG; 4 (Witsell 05-387, Baker GRF-039)

Dichanthelium linearifolium (Scribn.) Gould; PHF, RD, RI, SG, SHB, SSB; 4 (Witsell 05-392, Baker GRF-038)

Dichanthelium polyanthes (Schult.) Mohlenbr.; RD, RI, SG; 4 (Witsell 05-515, Baker GRF-119)

Dichanthelium sphaerocarpon (Elliott) Gould; LF, RI, SG; 4 (Witsell 05-521, Baker GRF-189)

**Digitaria ciliaris* (Retz.) Koeler var. *ciliaris*; RD; 3 (Witsell 05-1202, Baker GRF-243)

**Digitaria ischaemum* (Schreb.) Muhl.; RD; 3 (Witsell 05-1201)

Echinochloa muricata (P. Beauv.) Fernald var. *microstachya* Wiegand; SL; 2 (Witsell 05-1331, Baker GRF-341)

Elymus canadensis L.; RI; 2 (Witsell 05-789, Baker GRF-260)

Elymus virginicus L.; LF, PHF, RI; 2 (Witsell 05-790)

Eragrostis hypnoides (Lam.) Britton, Stearns, & Poggenb.; SL; 4 (Witsell 05-804, Baker GRF-312)

Eragrostis spectabilis (Pursh) Steud.; CC, RD; 3 (Witsell 05-1173, Baker GRF-298)

Festuca paradoxa Desv.; SG; 1 (Witsell 05-652)

Festuca subverticellata (Pers.) E. B. Alexeev; NS, RI; 2 (not collected)

**Hordeum pusillum* Nutt.; RD; 3 (Witsell 05-400, Baker GRF-130)

Leersia virginica Willd.; DW, LF, NS, RI, SP; 3 (Witsell 05-1169)

Melica mutica Walter; NS, RI, SG; 4 (Witsell 05-229)

Muhlenbergia sobolifera (Muhl. ex Willd.) Trin.; PHF, RD, SG; 3 (Witsell 05-1203)

Muhlenbergia sylvatica (Torr.) Torr. ex A. Gray; RI; 3 (Witsell 05-1200, Baker GRF-348)

Panicum anceps Michx. subsp. *anceps*; PF, PHF, RD, SG, SP; 4 (Witsell 05-1165, Baker GRF-299)

Panicum dichotomiflorum Michx. subsp. *dichotomiflorum*; RD, SL; 3 (Witsell 05-1171)

Panicum flexile (Gatt.) Scribn.; RD; 2 (Baker GRF-332)

Panicum rigidulum Bosc ex Nees subsp. *rigidulum*; LF, PSG, SL; 3 (Witsell 05-800, Baker GRF-343)

Paspalum laeve Michx.; PSG, RI; 3 (Witsell 05-1298, Baker GRF-264)

**Poa annua* L.; RD; 3 (Witsell 05-246)

Poa sylvestris A. Gray; RI, SSB; 2 (Witsell 05-249)

Saccharum alopecuroideum (L.) Nutt.; PF, PHF, RD; 2 (Witsell 05-1296)

**Schedonorus arundinaceus* Roem. & Schult. (nom. cons.); RD, RI, SG; 3 (Witsell 05-508, Baker GRF-155)

Schizachyrium scoparium (Michx.) Nash var. *scoparium*; PHF, SG; 1 (not collected)

Setaria parviflora (Poir.) Kerguélen; SL; 2 (Witsell 05-1365)

Setaria pumila (Poir.) Roem. & Schult. subsp. *pumila*; RD; 2 (Baker GRF-242)

Sphenopholis intermedia (Rydb.) Rydb.; NS, PHF, SSB; 2 (Witsell 05-225)

Sphenopholis nitida (Biehler) Scribn.; NS; 2 (Witsell 05-390, Baker GRF-179)

Sphenopholis obtusata (Michx.) Scribn.; SSB; 2 (Baker GRF-124)

Sporobolus clandestinus (Biehler) Hitchc.; SG; 2 (Witsell 05-1295)

Steinchisma hians (Elliott) Nash; RI/SG; 2 (Witsell 05-678)

Tridens flavus (L.) Hitchc. var. *flavus*; RD, SG; 3 (Witsell 05-1164, Baker GRF-296)

**Vulpia myuros* (L.) C. C. Gmel.; RD; 3 (Witsell 05-397, Baker GRF-126)

Vulpia octoflora (Walter) Rydb. var. *octoflora*; RD, SG; 3 (Witsell 05-401, Baker GRF-129)

Ruscaceae

Maianthemum racemosum (L.) Link subsp. *racemosum*; NS; 1 (not collected)

Polygonatum biflorum (Walter) Elliott; NS; 1 (not collected)

Smilacaceae

Smilax bona-nox L.; DW, LF, RD, SL; 4 (Witsell 05-753)
Smilax glauca Walter; LF, SL; 3 (Witsell 05-756)
Smilax rotundifolia L.; DW, LF, RI; 4 (Baker GRF-067)
Smilax tamnoides L.; NS; 1 (not collected)

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Safety of a Red Diode Laser Source for Fetal Retinal Stimulation Studies

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Light sources are now being used in clinical research to study development of the fetal visual system (Eswaran et al. 2004). These studies are making use of a relatively new technique called magnetoencephalography (Eswaran et al. 2002) to study fetal development. The technique calls for placing a light wand (Fig. 1) on the mother's abdomen and recording the magnetic fields associated with optically-induced nerve signals from the fetus. Current methods use a wand of 5.0 cm x 9.0 cm with a total output power of 20 mW at 630 nm from an array of 200 high intensity light emitting diode sources. Plans are underway to develop a more intense source to enhance the physiological measurements. The irradiance associated with the next-generation light simulator needs to be quite high because the transmission of light through the tissue which separates the outside world from the fetal retina is very low. The high irradiance suggests the need for a hazard analysis to determine if the source presents an optical danger to the mother or the research staff. Sliney and Wolbarsht (1982) give an overview of the hazards to the eye and skin from lasers and high intensity light sources. The following analysis addresses the question of potential danger to the eye and skin of both mother and research staff due to the high intensity light simulator.

The 630 nm red light from a high power (500 mW) diode laser will be launched into a fiber optic cable with an exit port fitted with a diffusing disc. For the purposes of this analysis the disc is assumed to be an ideal diffuse transmitter so that the red light from the fiber end will be taken as 500 mW emitting in a pure Lambertian geometry, i.e. its intensity varies as the cosine of the angle relative to the emitting surface normal and its radiance is independent of angle. An emitting surface of 1.0 cm^2 is assumed. It can be shown (Williams and Becklund 1972) that for a Lambertian source, the total emitted radiant flux Φ (in units of Watts or W) is related to the radiant intensity normal to the surface I(0), which has units of Watts/steradian (W-sr^{-1}) by the following expression:

$$\Phi = \pi\, I(0).$$

Since Φ is 500 mW in our case we can write

$$I(0) = (500\ \text{mW})/(\pi) = 160\ \text{mW-sr}^{-1}.$$

For a Lambertian surface the radiance L, which is independent of angle and is defined as the radiant flux per unit area of emitter (1.0 cm^2 in our case) per unit steradian, is given by

$$L = 160\ \text{mW-cm}^{-2}\ \text{sr}^{-1}.$$

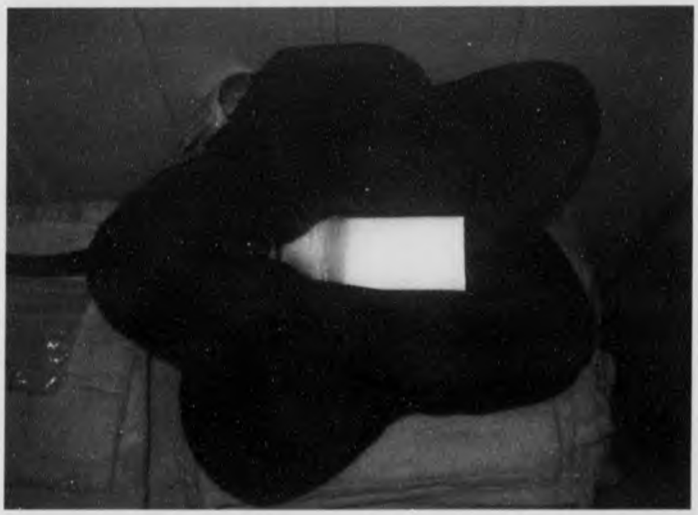

Fig. 1. A photograph of an optical wand used in a clinical study of light-evoked retinal response. The side facing the camera and lighted would be placed in direct contact with the mother's skin in this application.

The primary concern with most optical sources is the potential hazards to the human eye. The standards for safe viewing of laser beams (Laser Institute of America 2000) are separated into two primary viewing situations: the first is a direct viewing of a collimated laser beam and the second is the viewing of a beam that is relatively large and is considered to be an extended source. The former situation (small-source viewing) assumes that the laser acts as a point source with perfect collimation, and the consequent imaging by the eye will not be resolvable into a geometric image, rather a diffraction-limited spot on the retina. For the extended source, the radiation can be resolved by the eye into an image of finite size.

Since the size of the source relative to the viewing distance will distinguish the two viewing situations, a calculation of the source angular subtense is necessary. This value must then be compared to the limiting angular subtense (α_{min}), the apparent visual angle in the safety standards that divides small-source viewing from extended-source viewing. If the source angle exceeds α_{min}, then extended source viewing is presumed, otherwise the limits for small-source viewing are applied. For wavelengths between 400 and 1400 nm α_{min} = 1.5 mrad. The angular subtense for a 1.0 cm^2 source at a viewing distance of 50.0 cm (a value representing a reasonable distance from the mother's stomach to her eyes) is α = 1.0/50.0 = .020 radians = 20.0 mrad. Since α exceeds α_{min}, the appropriate limits for extended source viewing must be applied.

The safety standards for extended source ocular exposure depend on wavelength and exposure duration (LIA-2000, Table 5b). For a wavelength of 630 nm and an angular subtense of 20.0 mrad the maximum permissible exposure (MPE) for a duration of 0.7 to 15.4 s (T_2 in Table 5b of ANSI) is given in terms of an integrated irradiance:

$$\text{MPE} = 1.8\, C_E\, t^{0.75} \times 10^{-3}\ \text{J-cm}^{-2}, \qquad (1)$$

where t is the exposure time in seconds and C_E is the ratio of source angular subtense to α_{min}. In our case C_E = 20/1.5 or 13.3. For a 1 second viewing, MPE in terms of irradiance would be 24 mW-cm^{-2}. For continuous viewing, the maximum permissible irradiance (LIA-2000, Table 5b) is given by:

$$\text{MPE} = 1.8\, C_E\, T_2^{-0.25} \times 10^{-3}\ \text{W-cm}^{-2}. \qquad (2)$$

For the conditions in our case this latter equation yields a value of 12 mW-cm^{-2}. Extended sources that produce an irradiance less than 12 mW-cm^2 are within safety limits for continuous viewing.

A hazard analysis for the laser light simulator can now be done. Given a 1.0 cm^2 Lambertian source with a radiance of 160 mW cm^{-2} sr^1, the resulting irradiance at a viewing distance of 50.0 cm would be LΩ, where Ω is the solid angle subtended at the eye by the source, i.e. 1/50^2 or 0.0004 sr. Thus the irradiance for the mother would be, at most, 0.064 mW-cm^{-2}. The calculations above yield a 1.0 second viewing limit of 24 mW-cm^{-2} and a long-term continuous viewing limit of 12 mW-cm^2, well above the actual value of 0.064 mW-cm^{-2}. The wand should present no hazard to the mother's eyes even under continuous long-term viewing from a distance of 50.0 cm. In order to achieve an exposure of 12 mW-cm^{-2}, the separation distance between diffusing disc and eye would have to be reduced to 3.7 cm.

One question that may arise is whether multiple 1.0 cm^2 sources, each of 500 mW/cm^2, placed side-by-side would constitute a hazard if viewed by the mother. For purposes of this analysis a 7.0 cm x 7.0 cm source (approximately 50 cm^2) with an emittance of 500 mW-cm^2 is presumed. The radiance of the source will not change; it will still be 160 mW/cm^2 sr. The angular subtense α will now increase to 7/50 or 140 mrad. The value of C_E in (2) becomes 131. T_2 is now 100 s (LIA-2000, Table 6). Application of (2) yields an MPE of 75 mW-cm^{-2}. With the larger source the exposure will be greater due to the larger value of Ω, the solid angle formed by the source at the eye. With this larger value of Ω (0.02) the irradiance at the eye becomes 3. mW-cm^2, still well below the 75 mW-cm^{-2} limit. For the smaller 1.0 cm^2 source and a viewing distance of 50 cm, exposure is 0. % of the limit; for the larger 50 cm^2 source exposure is 4.2% of the safe limit.

While optical radiation hazards for the skin are generally considered secondary to those for the eye, it is important to limit the exposure of the skin to high levels of optical radiation in order to prevent harmful thermal or photochemical effects. In our application the light simulator surface will be in contact with the mother's skin, thus maximizing maternal exposure to the optical radiation. The standards (LIA-2000, Table 7) for the safe use of lasers specify the maximum permissible exposure (MPE) for skin to a laser beam. This standard will be used to assess the safety hazard to the mother's skin and to set a limiting-exposure condition to guarantee a safe condition for the mother.

While the safety standards do give limits for various time exposures the most conservative case will be used in this analysis, i.e. the limits for continuous exposure will be determined and used to limit the operating parameters for the wand. For continuous exposure in the visible region of the spectrum to small areas (less than 100 cm^2) the limit is 200 mW-cm^{-2}. For a conservative design with a margin of safety of 2 the maximum allowed irradiance is 100 mW-cm^{-2}. Since we assume the output of the simulator is 500 mW-cm^{-2} and it is assumed to be in direct contact with the mother's skin, it is clear that continuous illumination is not a safe condition for the skin.

In order to ensure that a continuous limiting value of 100 mW-cm^{-2} is achieved, a duty cycle of 20% would be necessary. Thus a 1.0 s on-time followed by a 4.0 second off-time would yield an effective continuous irradiance of 100 mW-cm^{-2}. Further reductions in on-time to full cycle period ratio will enhance the safety margin. As a reference check, the continuous level of 100 mW-cm^{-2} is of the same order of magnitude as the maximum irradiance of the noonday sun on the skin for someone in Arkansas. Moreover, solar radiation contains a full spectrum of energies with a significant absorption component, whereas the red 630 nm radiation is absorbed less and should provide no photochemical threat to the skin tissue.

Here we consider only the effect of the optical radiation on the skin. In terms of the complete safety hazard analysis, we are assuming that there is negligible light absorption at the exterior surface of the simulator and that its temperature will not exceed body temperature even when in continuous contact with the mother's skin.

These calculations show that continuous viewing of the 630 nm light simulator wand with a radiance of 160 mW cm^{-2} sr at a distance of 50.0 cm will not present a hazard to the eyes of the mother or the research staff. Also, if the source is pulsed with a 20% duty cycle the wand placed in direct contact with skin will not constitute a hazard to the skin.

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An Unusual Prey Item Found in an Eastern Bluebird Nest

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Adult Eastern Bluebirds consume small fleshy fruits in the winter but switch to terrestrial arthropods during the breeding season (Gowaty and Plissner 1998). Prey delivered to chicks also consists mostly of small arthropods (Gowaty and Plissner 1998). In Michigan, Lepidopterous larvae (moths and butterflies) made up 32.4% of the chick diet and included individuals from the families Noctuidae, Arctiidae, Pieridae, Geometridae, Notodontidae, Pyralidae, and Sphingidae. Orthopterans (grasshoppers, crickets, and katydids) made up 25.6% of the chick diet and included individuals from the families Acrididae, Tetrigidae, Tettigoniidae, Gryllidae, and Mantidae. Arachnids (spiders) made up 11.3% of the chick diet, Coleopterans (beetles and weevils) made up 11.0%, and several other insect orders made up very small percentages of the chick diet (Pinkowski 1978). Other studies have found similar results (Pitts 1978, reviewed by Gowaty and Plissner 1998).

From March 2002 to July 2006, graduate students at Arkansas State University have monitored approximately 200 nest boxes. The boxes are located in Brookland, Arkansas in a mixed habitat of open pastures and woodlots. Nest-monitoring methods follow those outlined by Robinson (2005). In brief, nest boxes were checked weekly for nest building activity, then every third day for egg laying, and daily until the last egg was laid. Chicks were visited every third day after hatch.

On 8 May 2005, a dead midwest worm snake (*Carphophis amoenus helenae*), approximately 8 cm in length, was found among chicks in the cup of an active Eastern Bluebird nest box. The snake was intact and well preserved, which facilitated identification. The nest box was located in fairly open habitat, the nest box pole was covered with axle grease, and there were no overhanging branches nearby, making it unlikely for this fossorial snake (Trauth et al. 2004) to have climbed into the box. It was presumably delivered as prey to chicks by an adult bluebird. The chicks were 9 days old at the time and all four chicks successfully fledged from the nest.

There are few records of Eastern Bluebirds taking vertebrates as prey; an unknown species of snake (Flanigan 1971); a shrew (*Sorex* sp.) (Pinkowski 1974); lizards (Gowaty and Plissner 1998); and tree frogs (*Hyla* sp.) (Bent 1949). We are aware of only 1 record of parent bluebirds feeding a vertebrate (*Eumeces* sp.) to their chicks (Pitts 1978). Our record appears to be the second to document delivery of vertebrate prey by an Eastern Bluebird to its chicks, and the first to identify a species of snake as a prey item.

ACKNOWLEDGMENTS.—We owe thanks to Stan Trauth o Arkansas State University for identification of the snake specie and to Jonathan Stanley for helping us to include the correc nomenclature in our manuscript.

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New Evidence for a Maternity Colony of the Indiana Bat (*Myotis sodalis*) in the Delta of Arkansas

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The migratory Indiana bat (*Myotis sodalis*) hibernates in caves and mines. Upon emergence from hibernation, females will segregate from males and migrate typically to riparian and/or bottomland forests where they will form maternity colonies where the young are born and raised (Hall 1962, Carter 2006). Arkansas contains appropriate caves for hibernation and lowland riparian forests that are seemingly suitable habitats for Indiana bat maternity colonies. Although Indiana bats are well known from several hibernacula in Arkansas (e.g., Harvey 2002), direct evidence of maternity colonies is lacking. However, historical information suggests that Indiana bats may bred the Arkansas Delta. Notably, in July 1898 2 juvenile Indiana bats were collected in the extreme northeast portion of Arkansas near the town of Greenway in Clay County (Hall 1962) and these specimens reside in the Field Museum of Chicago. Since this capture occurred in July, the young had likely only recently become volant, which suggests that they were born in a nearby maternity colony.

In this paper we describe the capture of a lactating Indiana bat in Clay County, Arkansas. This capture is perhaps the strongest evidence to date of breeding activity of Indiana bats within the borders of Arkansas.

The Black River Wildlife Management Area (BRWMA), located in Clay County, Arkansas, is a large bottomland hardwood forest approximately 5.8 km^2 in size. The forest is comprised primarily of mature hardwoods and cypress with a closed canopy. Many areas are annually inundated with water. The capture of the Indiana bat occurred in 1 of 3 small openings in the forest along the Black River. These openings are used as flight corridors by the bats to fly between the river and a dirt road. In 2005, netting for bats was conducted for 6 nights at this site. Four mist nets 6 to 12 m in length were placed perpendicular to the corridors. Nets were opened at sunset, checked every 15 to 20 min, and then closed after 5 hrs. Once removed from the net, bats were identified to species and assessed for reproductive status. Identification as juvenile or adult was based on the degree of ossification of the epiphyseal-diaphyseal (Edythe 1988).

The capture of the adult Indiana bat occurred at 9:45 on 9 July, after 2 previous nights of netting at that location. The captured Indiana bat was identified as such based on the combined characteristics of a keeled calcar, tricolor fur, and short sparse hairs on the toes (Thomson 1982, Sealander and Heidt 1990). The bat was confirmed to be reproductively active as lactation was induced by palpation of the mammary glands. The weight (7.5 gram) and size (forearm 40.19 mm) were consistent with measurements reported for Indiana bats (Thomson 1982, Sealander and Heidt 1990).

It is our conviction that the capture of a lactating Indiana bat in June provides evidence of a nearby maternity colony. Our conjecture is supported by proximity of our capture location to known Indiana bat hibernacula, and the suitability of the habitat. The closest known hibernating colonies of Indiana bats occur in Missouri and Arkansas within 150 km, which is well within the known migration distance for the species. Migrating Indiana bats from maternity colonies in Michigan migrated an average distance of 460 km to various hibernacula (Kurta and Murray 2002). The habitat represented in our site of capture is fairly typical of the large forest block of the BRWMA. Hydric habitats with large living and dead hardwood trees, the habitat typically associated with maternity sites (Carter 2006), are abundant in the area. It is interesting to note that the 2 juvenile Indiana bats captured in 1898 were reported from the town of Greenway, AR, which is located approximately 7.2 km away from our capture site (Hall 1962). Considerable effort has been put forward by local agencies in studying and surveying Arkansas's hibernating populations of Indiana bats, but conservation efforts in the Delta have been lacking since maternity colonies were unknown. Based on this capture, we plan to perform extensive mist-netting surveys of the BRWMA in conjunction with radio-telemetry to gather further evidence of Indiana bat maternity colonies within Arkansas.

Acknowledgments.—Conversations with Blake Sasse alerted us the presence of the historical data. Funding was provided by the Arkansas Game and Fish Commission. This work benefited from the reassignment time granted to TSR from the Environmental Sciences Program at ASU. This study was done under Fedeal Permit TE075912-0 and State Permit 081620041. Thanks to Tanja McKay for comments on a previous draft of this manuscript.

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Hyperinfection with the Bass Tapeworm, *Proteocephalus ambloplites* (Cestoda), in the Black Basses *Micropterus punctulatus* and *M. dolomieui* from Certain Arkansas Reservoir Lakes

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Proteocephalus ambloplites is a tapeworm that lives as an adult in the intestinal tract of black bass (*Micropterus* spp.). The life cycle is complex and hosts include first a microcrustacean followed by a small fish as a second intermediate host. In the small fish the parasite transforms into a metacestode larval stage, the plerocercoid, which migrates through the gut into the coelom where it awaits ingestion by the final host. The plerocercoid form of *P. ambloplites* can be found in a number of freshwater fishes that can act as intermediate hosts (Hoffman 1999). When a bass eats the infected small fish, the plerocercoids penetrate the gut and invade parenteric organs, most importantly, the gonads which can cause reduced fertility in the host fish. Certain plerocercoids under the stimulus of a water temperature change will reinvade the gut to become tapeworm adults. (Esch and Huffines 1969, Fischer and Freeman 1969, Fischer 1973). The major importance of this parasite is the effect on bass reproduction when the bass tapeworm plerocercoids are in large numbers in gonadal tissue.

All three major *Micropterus* species (*M.salmoides, M. dolomieui*, and *M. punctulatus*) are known to be infected with both the adult tapeworms and their plerocercoid larvae. Preliminary data from our necropsies of bass for infections with *Clinostomum marginatum* metacercaria indicated that *P. ambloplites* infections were heavier in Arkansas than previously reported. This study was initiated to determine the extent of these infections in black bass from Arkansas lakes. Bass were collected from tournament fishermen in 1988-89 from selected Arkansas reservoir lakes, transported to the University of Arkansas for Medical Sciences and necropsied for the presence of plerocercoids of *P. ambloplites*. Definition of population characteristics followed that of Bush et al. (1997) and were as follows: Prevalence (percent of fish infected), maximum abundance (largest number of parasites in a single host), mean abundance (average number of parasites per fish) and its standard deviation. The findings of this study are summarized in Table 1. Mean intensity (average number of parasites for infected fish only) and its standard deviation are not included since this report was to compare the extent of the plerocercoid infections in the Arkansas reservoirs with older literature and in those studies these values are not given. The variance of a mean intensity reported with 100% prevalence, as seen in Table 1, is equivalent to the variance of a mean abundance of 100% prevalence.

Very few (<0.1 percent) adult tapeworms were found in any of the collections. Also, the plerocercoid numbers in the present study may be a result of seasonal effects (late spring, summer) because the plerocercoid larvae are generally not found in bass during the winter in northern climes, but reoccur in large numbers again in the spring (Esch and Huffines 1973, Fischer 1973). However, Eure (1976) found that plerocercoids could be found year-round in a reservoir in South Carolina. Studies in Arkansas have found plerocercoids in Arkansas black bass in the winter (Cloutman 1975, Kilambi and Becker 1977), but any temporal relationship between adult tapeworms and plerocercoids in the State's reservoirs is still to be definitively determined.

Table 1 shows data from this study, data from other Arkansas waters, and other selected locales from North America that have the highest reported abundances of plerocercoid infection. The current data for spotted bass (*M. punctulatus*) and largemouth data (*M. salmoides*) are both from Lake Maumelle in Pulaski County. The infected smallmouth (*M. dolomieui*) were collected from Bull Shoals Lake. All of the data in Table 1 represent the largest mean abundances for individual collections in each study but collections of other bass hosts were also made from the same bodies of water, but with smaller abundances being reported in the other bass populations. An exception is the Gull Lake, Michigan data which included all bass collections that were made in that locale for a given year.

Table 1 shows that the infrapopulations of *P. ambloplites* plerocercoids in spotted bass from Lake Maumelle are hyperinfections and among the highest reported for bass anywhere and are the highest ever for spotted bass. The intensity of the smallmouth infection from Bull Shoals Lake, although not as high, can also be considered a hyperinfection based on comparative literature reports. Largemouth bass in Lake Maumelle have many fewer plerocercoids than seen in the other two bass. This discrepancy between largemouth and other bass has also been noted before in the Gull Lake study (Gilliland and Muzzall 2004). They found that largemouth plerocercoid prevalence was 100% with a mean intensity of 18.1 (±12.9 SD). This agrees with the degree of infection seen in the largemouth bass from Lake Maumelle in Arkansas. It might seem that the largemouth may have a natural resistance to tapeworm infections, relative to the other 2 species. However, Szalai and Dick (1990) found infected largemouth in a reservoir in Saskatchewan, Canada to have plerocercoid numbers equivalent

Table 1. Population parameters of selected plerocercoid infections of the bass tapeworm, *Proteocephalus ambloplites*, in Arkansa and North America. LM = largemouth, SM = smallmouth, and SP = spotted bass. Only the parameters of prevalence, maximu abundance, and mean abundance (standard deviation) are listed. Daly et al. refers to the present study. ---- are unrecorded data.

Investigators	N	Locale	Bass	Prevalence	Max. Abundance	Abundance
Daly et al. this study	17	Arkansas	SM	100	117	35.5 (± 34)
Daly et al. this study	55	Arkansas	SP	100	200	66.3 (± 43)
Daly et al. this study	8	Arkansas	LM	75	52	12.4 (± 17)
Becker et al., 1978	347	Arkansas	SP	91.5	----	12.3 (----)
Cloutman, 1975	89	Arkansas	LM	----	----	7.4 (----)
Kilambi and Becker, 1977	12	Arkansas	SM	4	7	0.3 (± 2.02)
Gilliland and Muzzall, 2004	54	Michigan	SM	100	200	72.5 (± 44.8)**
Szalai and Dick, 1990	8*	Saskatchewan	LM	100	----	99.0 (± 116)**

*Largest bass hosts only
**Mean intensities (SD)

to those found in Arkansas and Michigan smallmouth and Arkansas spotted black bass (Table 1). The reasons for these geographical variations between the black bass species are unknown and could be due to a number of regional factors such as habitat preference or forage fish diet and perhaps even an inherent strain immunity (The Saskatchewan largemouth were hatchery-raised and introduced).

The most important pathology produced by these tapeworm larvae is destruction of the host gonadal tissue, but in Arkansas reservoirs the overall effect of such large numbers of plerocercoids on bass fecundity is yet to be ascertained.

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Understanding Global Positioning System Limitations: A Case Study of Mapping and Survey Grade GPS

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The Global Positioning System (GPS) is used to locate positions on Earth. The accuracy of a position derived by GPS depends on the GPS-receiver design, the satellite configurations at the time of data collection, local conditions which may interfere with signal reception, length of time spent on data collection, and the method of signal interpretation. GPS receivers are grouped into 3 general categories based on the previous criteria and additional data-recording capabilities. GPS receivers with low accuracy and relatively few features are considered recreational grade. Increased accuracy and data management features designed for advanced data collection characterize mapping grade. Survey-grade GPS receivers have the highest accuracies.

"The accuracy of a position computed by a GPS receiver is a function of the geometry of the GPS constellation visible at that moment in time" (Trimble 2001). The geometry of the satellite configuration is reported as the Position Dilution of Precision (PDOP) and is considered a measure of accuracy of the GPS data. This data may be represented as a horizontal precision.

This study compares mapping-grade GPS data to survey-grade GPS data to assess the accuracy of mapping-grade data. In addition, the reported horizontal precision is used to assess the quality of reported locations when the sample locations are near the reported horizontal accuracy of the data collector.

The study was conducted at Pine Tree Branch Station located in St. Francis County, Arkansas. The test site was an open field of approximately 92,000 m^2. The test plot was constructed on a 3.6 m grid with 480 staked corners. Initial grid construction was with tape measure. Two Trimble Model 4700 GPS receivers with Trimble Microcentered L1/L2 GPS antennas with ground planes (part number 33429-00) were positioned for data collection, one at 6 meters, the second at 29 meters away from the grid. A TopCon GTS-603AF Total Station survey instrument was positioned 7.6 meters away from the grid (Fig. 1). The Trimble GPS-data recorders recorded carrier-phase GPS data for approximately 6 hours. This data was post-processed for differential correction using the Online Positioning User Service (OPUS). Three base stations were used for differential correction: Memphis 2 CORS, 78,075 m from the site; Bloomfield CORS, 216,825 m from the site; and Memphis WAAS CORS, 88,851 m from the site.

A TopCon Total Station survey instrument was used to record relative positions of the 2 survey-grade GPS recorders and all stake positions (TopCon 2002). The total station data was imported into a Geographic Information System (GIS) and the two survey-grade GPS positions were used to assign coordinates to all stake positions.

Each stake location was also captured by 1 of 2 Trimble GeoExplorer 3 GPS-data recorders by 1 of 2 operators. Data masks were set within the GPS-data recorder to minimize positional errors. The settings used for this study were Position Dilution of Precision (PDOP): < 4; Signal to Noise Ratio (SNR): 6; and satellite elevation mask: 15°. Data was collected in the Universal Transverse Mercator (UTM) projection, Zone 15, North American Datum 1983 (NAD83). Positions were collected at one-second intervals with a goal of reaching at least 30 positions per stake. The positions were averaged together into a single location for each stake. All stake locations were captured in a single day.

GPS location data from the GeoExplorer 3s were post-processed in GPS Pathfinder Office for differential correction. The base station used for differential correction was a Trimble 12-Channel Community Base Station located at the Ground Water Institute, Memphis, TN.

GPS Pathfinder Office produces a horizontal precision based on a user-selectable confidence level. The precision is a function of the GPS receiver type, method used to collect point data (number of positions averaged), distance to the base station used for differential correction, the PDOP, and other reference variances (Trimble 2001). The horizontal precisions were calculated at the 68% confidence level, the default value for the software. Corrected data were imported into a GIS for analysis.

The data were analyzed for positional agreement between the mapping- and survey-grade GPS. The mapping-grade data were also analyzed using reported 68% horizontal precision circles. The precision circles were tested for intersection with the correct survey-grade location, overlap with adjacent survey-grade locations, and overlap with adjacent mapping-grade locations. The horizontal precision estimates were also tested against the RMS-positional error in SPSS statistical software to determine the best-fit model.

The RMS error from the survey-grade GPS positions as reported by the OPUS solution was 0.025 m. The distance between the Total Station position and the furthest survey-grade GPS position was 118.5 m. Reported accuracy for the GTS-603AF Total Station is ±2 mm + 2 ppm (Topcon 2004). Therefore, expected accuracy for the stake locations was 0.027 m. The final grid measurements between stake locations were a mean distance of 3.689 m with a standard deviation of 0.179 m.

Stake locations collected with the GeoExplorer 3 GPS recorders were reported in 100ths of a meter from the origin

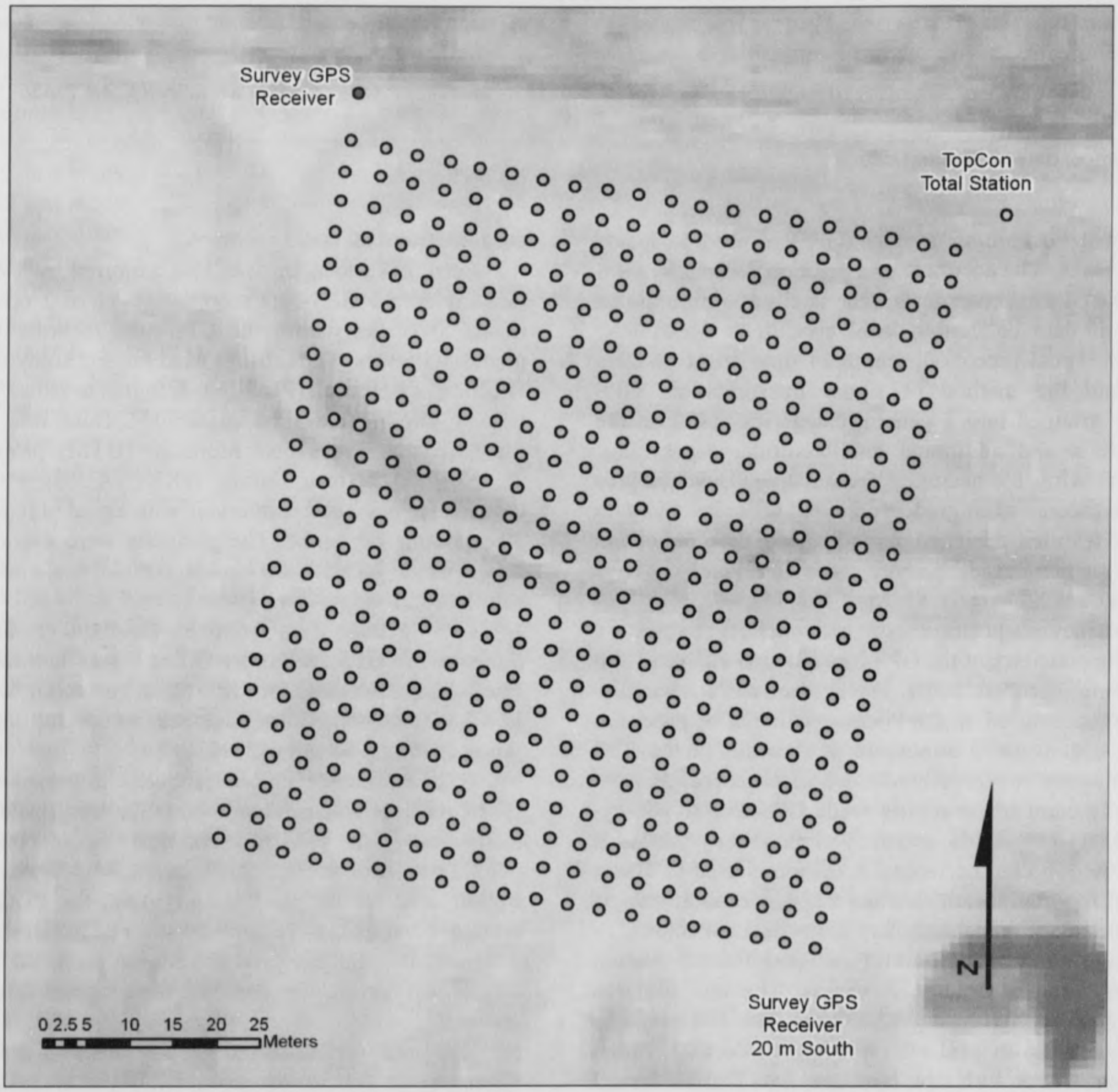

Fig. 1. Relative positions of survey GPS receivers, TopCon Total Station, and stake locations.

of the projection (UTM Zone 15, NAD83). The 68% horizontal precisions averaged 2.48 m with a standard deviation of 0.53 m. The RMS error between the mapping- and survey-grade locations averaged 0.80 m with a standard deviation of 0.53 m. The straight-line distances between the mapping and survey grade locations averaged 0.99 m with a standard deviation of 0.76 m.

The intersection between the mapping-grade GPS-precision circles and nearby GPS locations resulted in an average of 1.41 GPS locations observed in each precision circle with a standard deviation of 0.74 and a maximum of 8. The intersection between the mapping-grade GPS-precision circles and the survey locations resulted in an average of 1.4 survey-grade locations in each precision circle with a standard deviation of 0.66 and a maximum of 5. No statistical relationship was found between the 68% horizontal-precision and RMS-positional error.

The results show that the accuracy of the GeoExplorer 3 mapping-grade GPS recorder can be much better than specification. Average RMS-positional error in this study was below 1 m. However, no relationship was found between the 68% horizontal-precision estimates generated by GPS Pathfinder Office and the RMS error between the mapping-grade and survey-grade locations. This shows that while the reported GPS horizontal precision may be high (a low value), the accuracy of that position is unknown (Fig. 2).

The spacing of the test grid was within the accuracy

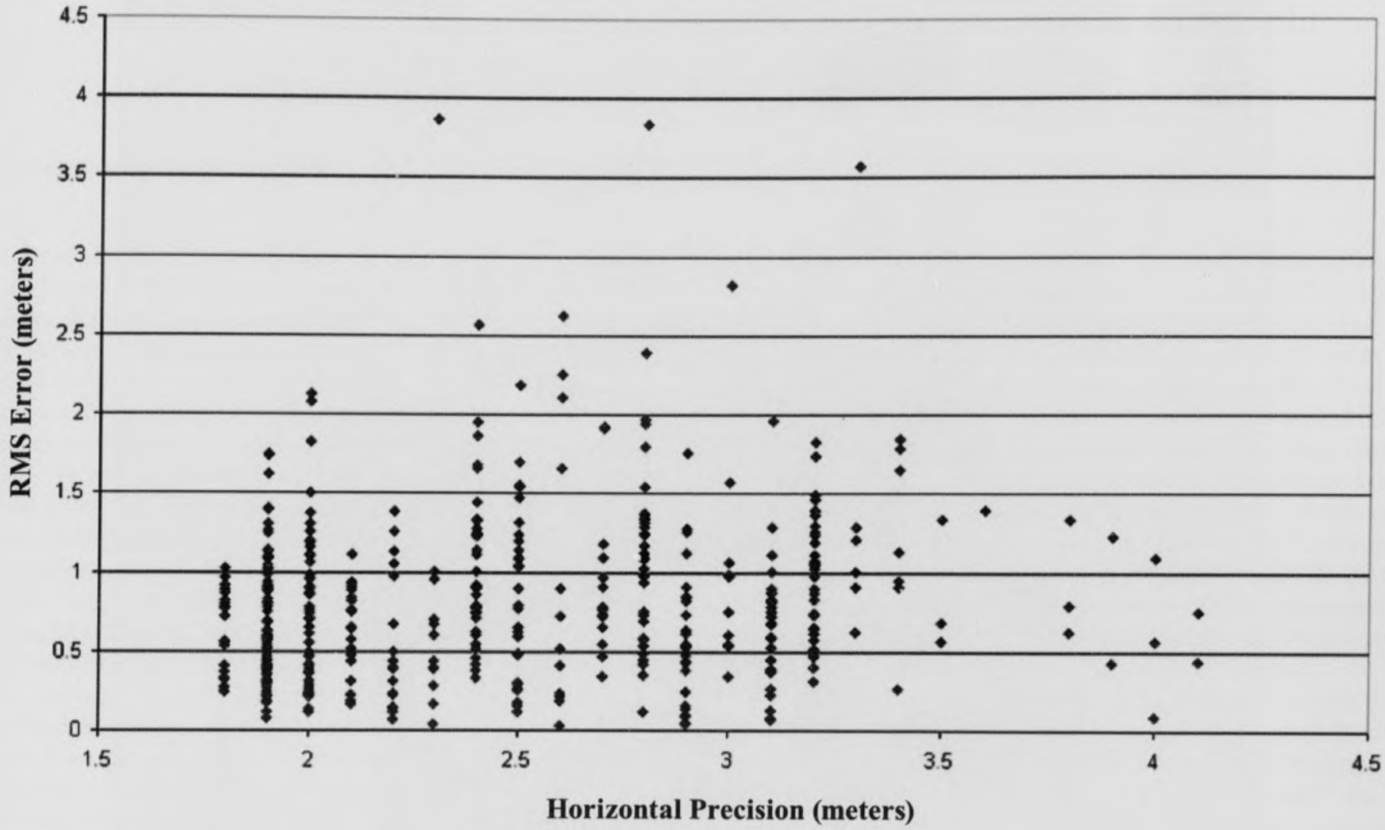

Fig. 2. Sixty-eight percent horizontal precision circle radius versus RMS error between survey location and mapping grade GPS location for stakes.

specifications for the GeoExplorer 3 GPS recorder. The precision circles captured the true location (survey grade data) in 66.46% of the data. However, more than one true location was captured in the precision circle in 32.92% of the data. In 3 of the mapping-grade GPS locations, the true location was not captured in the precision circle (Fig. 3).

It is useful to compare the precision circles to the reported locations when the grid spacing is within the horizontal accuracy of the GPS equipment. The horizontal-precision circles captured more than 1 GPS location 31.46% of the time. In all of the data, there was only one location where the precision circle did not overlap one or more precision circles for nearby locations. Because the precision circles represent the 68% probability of the location being within the circle, overlapping precision circles suggests that the locations with overlapping precision circles are indistinguishable.

This work demonstrates the high accuracy of mapping-grade GPS-data recorders when they are used in ideal conditions with proper procedures. However, it should be noted that the reported horizontal precisions represent a probability of a location being within the precision circle, and there is no relationship between the reported precision and the RMS error between the mapping-grade GPS location and the location derived by survey techniques. Using the reported precision as a measure of accuracy should be done with caution. This work also demonstrates the importance of horizontal spacing in experimental design when GPS is to be used to locate objects. To prevent uncertainty in feature location, the distance between adjacent features should be at least twice the maximum expected accuracy of the equipment used to identify locations.

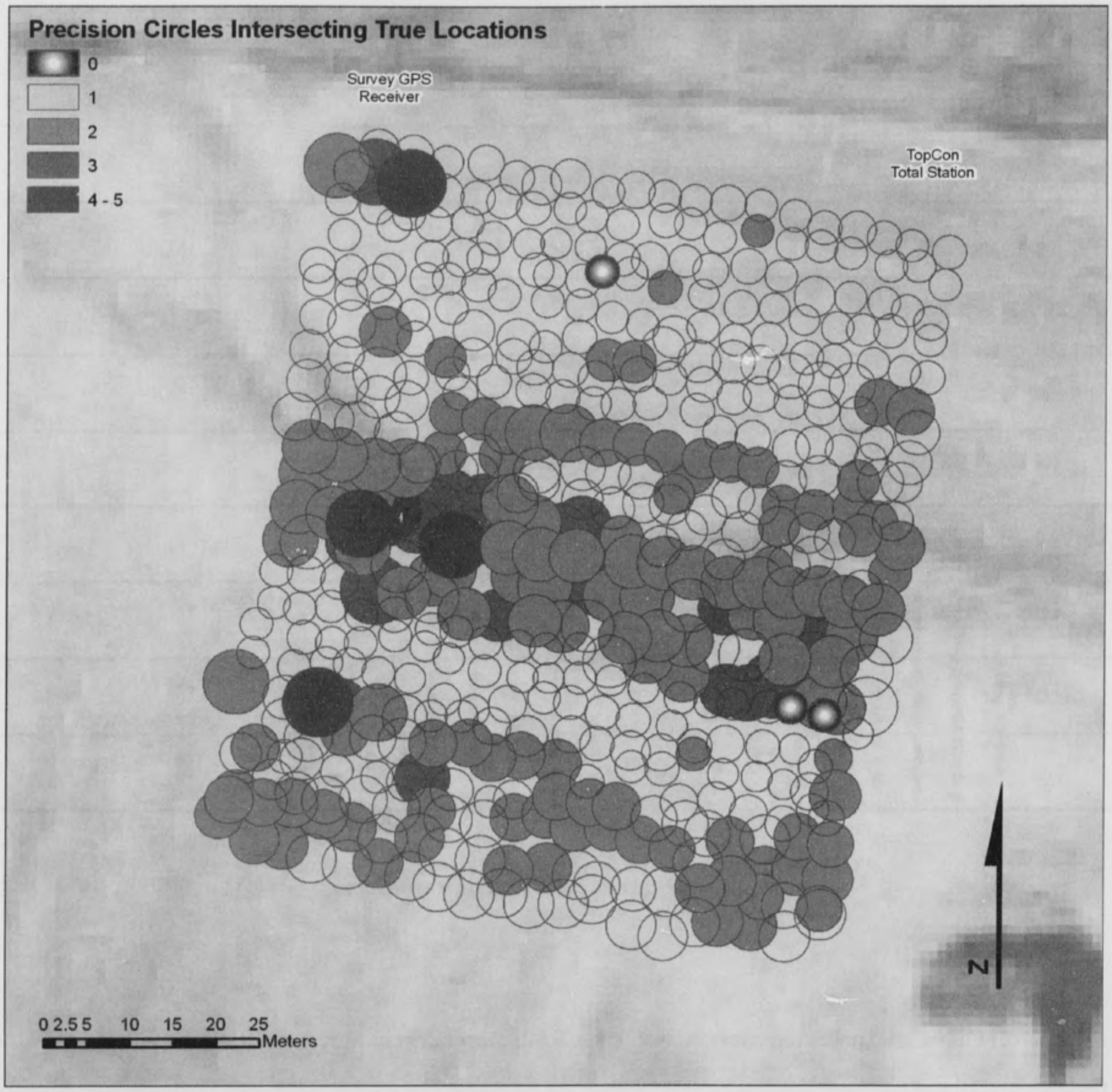

Fig. 3. Intersection of 68% precision circles and survey location. Hollow circles indicate precision circles which intersect survey location. Bullseye shading indicates circles which did not intersect any survey location. Gray circles indicate circles which intersect more than 1 survey location.

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Documentation of the First Known Occurrence of the Broad-billed Hummingbird (*Cynanthus latirostris*) in Arkansas

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Broad-billed Hummingbirds (*Cynanthus latirostris*) occur as residents in Mexico from Sonora and Chihuahua south to Chiapas in the west and in the east to parts of western Tamaulipas and Veracruz (American Ornithologist' Union 1998). In the United States this species is migratory, typically wintering in Mexico and occurring during the breeding season in southeastern Arizona and southwestern New Mexico (Powers and Wethington 1999). Herein we report on the occurrence of a first-year male Broad-billed Hummingbird found during winter 2005-2006 in White Hall, Arkansas (Jefferson Co.). This represents the first documented occurrence of this species in the state.

In late November 2005 an apparent over-wintering hummingbird was detected visiting a nectar feeder at a residence in White Hall (34.2528° N, 92.1059° W [NAD 27]) (M. Branch, pers. comm.). On 28 December 2005 this bird was initially identified as a Broad-billed Hummingbird by Becky Wheeler, Lance Peacock, and Leslie Peacock (pers. comm.). A general description of the bird, based on both field observation and inspection of photographs by RHD, included: long, needle-like bill widened at the base; lower mandible bright orange-red with dark tip and with upper mandible mostly dark but orange-red near posterior end; head and nape iridescent emerald to coppery-green; gorget iridescent blue; eyes dark with slightly-elongated white postocular spot; back, wing coverts, and uppertail coverts iridescent coppery-green; remiges dark gray-black, extending to tip of tail when perched; breast and belly iridescent emerald green; undertail coverts light gray; tail deeply forked; rectrices (r) dark blackish-blue with gray tips on r1 (right r4 with sheath visible at base suggestive of recent molt). The lack of extensive red on the upper mandible, elongated postocular spot, and lack of gray on tips of outer rectrices signify this individual was a first-year male Broad-billed Hummingbird (Moore 1939, Pyle 1997, Powers and Wethington 1999). Partial anterior and posterior images of the bird are presented in Fig. 1.

Vocalizations from the bird were delivered at rest and in flight. Sounds were a chatter consisting of a rapid, repetitive sequence of notes ("*chit*"). These notes are thought to be given when the bird is in mild alarm (Powers and Wethington 1999). This Broad-billed Hummingbird chatter call is similar in sound to the call produced by Ruby-crowned Kinglets (*Regulus calendula*).

The bird continued to visit the nectar feeder, and on 5 January 2006 it was trapped, measured, banded by EPF, and released unharmed. Measurements of the bird were as follows: wing chord = 53.5 mm, tail length = 31.0 mm, exposed culmen = 21.0 mm, mass = 3.42 g. In addition to plumage characteristics, these measurements help to distinguish this bird from most North American hummingbirds and identify it as a Broad-billed Hummingbird (Pyle 1997). Slight bill corrugations were noted, further indicating that the bird was likely in its first year (Ortiz-Crespo 1972). As additional documentation of this record, the third rectrix (r3) on the right side of the tail was collected as a preserved partial specimen and is archived at the University of Arkansas – Little Rock (accession number UALR-H053, W. Baltosser, pers. comm.). This Broad-billed Hummingbird continued to feed at the same nectar feeder until it was last detected on 30 March 2006 and was presumed to have dispersed.

This occurrence of a Broad-billed Hummingbird is the first for Arkansas and one of the most distant winter records for the species, approximately 1400 km from its usual winter range in southern Tamaulipas, Mexico. Most previous extralimital winter records of this species have occurred in areas of the northern coast of the Gulf of Mexico, principally in coastal areas of Louisiana, in central and west Texas, and in southern California (Howell 2002, Williamson 2001). A few recent additional extralimital observations of wintering Broad-billed Hummingbirds from eastern and central North America have been recorded from Alabama (Cooley 2002, 2003), Georgia (Davis 2002), Illinois (Brock 1997), Kansas (Grzybowski and Silcock 2005), North Carolina (LeGrand et al. 2002), and Oklahoma (Arterburn 2004). While the number of vagrant occurrences of non-breeding Broad-billed Hummingbirds has increased across North America, much of this may be due to an increase in the number of observers and their increased skill at field identification. Additional factors, such as climate change, weather patterns, and residential plantings may also be contributing factors to the increased number of reported extralimital occurrences of this species throughout North America.

ACKNOWLEDGMENTS.—We appreciate the hosts, Chester and Maxine Branch, who graciously allowed many visitors to view Arkansas's first Broad-billed Hummingbird at their home. Max Parker trapped the bird for EPF to subsequently measure, band, and obtain a feather specimen. This manuscript was improved by the comments of Gary Schnell and Terry Maxwell.

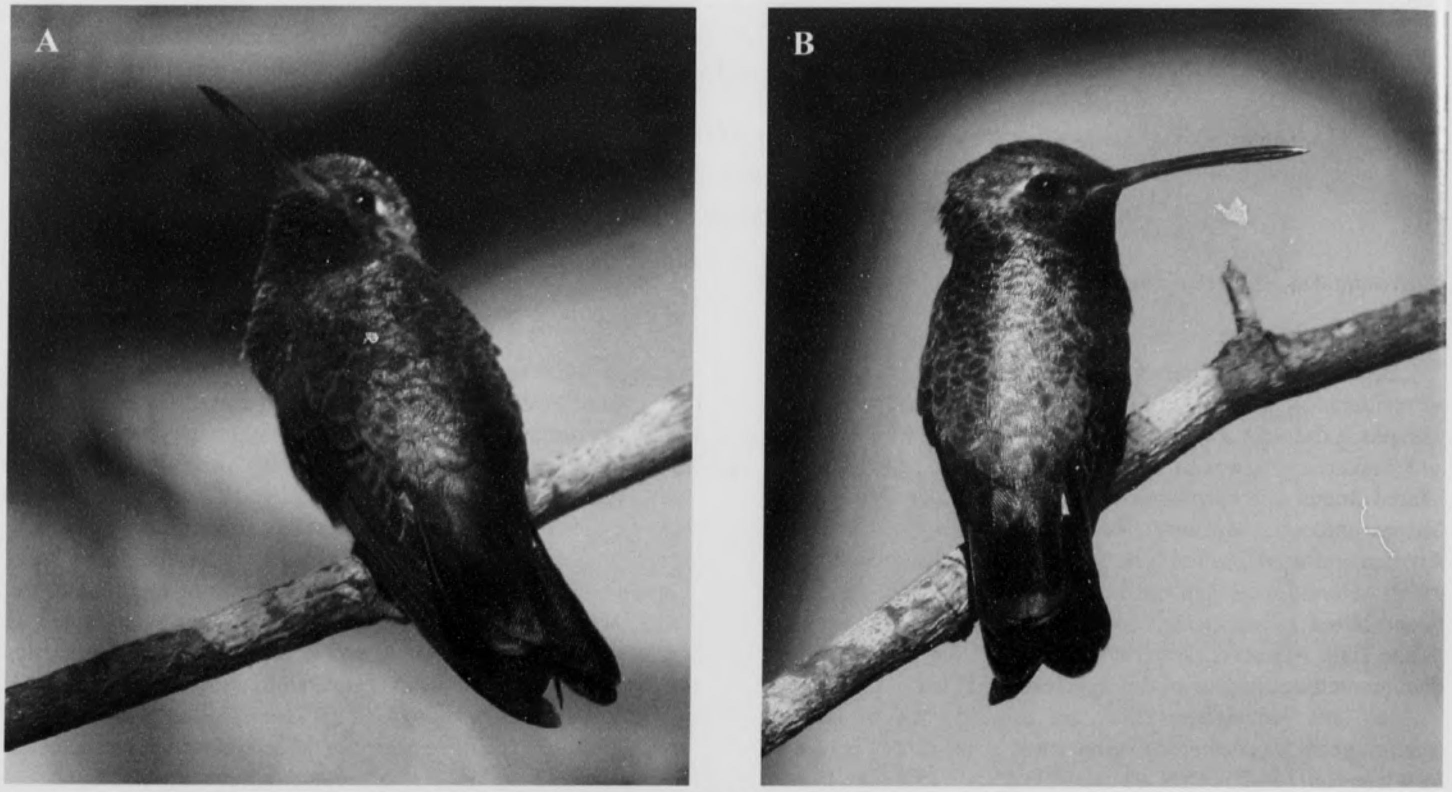

Fig. 1. Partial anterior (A) and posterior (B) views of a Broad-billed Hummingbird found in White Hall, Arkansas, during the 2005-2006 winter. Images taken 29 December 2005.

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Density Functional Studies of Structure and Bonding in Piano Stool Carbene Complexes

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Recently we reported the results of approximate molecular orbital calculations used to explain the structural preferences and other properties of the chelating carbene complex $\mathrm{Cp'(CO)Mn\{C(OEt)CH_2PPh_2\}}$ (**1**) and related complexes (Graham 2003). Since that time we have reexamined the electronic structure of the complex **1** using more quantitative density functional calculations. These calculations, unlike the previously published Fenske-Hall calculations, are well suited to determination of total energies and optimal geometries for molecular systems. The results of these new calculations are briefly described in this note.

Density functional calculations were used to optimize the geometries of the complexes $\mathrm{Cp'(CO)Mn\{C(OEt)CH_2PPh_2\}}$ (**1**) and $\mathrm{Cp(CO)(PPh_3)Mn\{C(OMe)Et\}}$ (**2**). Optimized geometries were also determined for the related model complexes $\mathrm{Cp(CO)Mn\{C(OMe)CH_2PH_2\}}$ (**1a**) and $\mathrm{Cp(CO)(PH_3)Mn(C(OMe)CH_2CH_3)}$ (**2a**). All calculations were carried out using the Amsterdam Density Functional program, version ADF 2004.01 (ADF 2004, Velde et. al. 2001, Guerra et. al. 1998). The Local Density Approximation and a triple ζ basis set with polarization functions (LDA/TZP) were used. Ball and stick representations of the optimized structure of each complex are given in Fig. 1. It is observed that the optimized model complexes are very similar to the optimized structures for the full complexes in important structural features (such as metal ligand distances and angles). The optimized structures are also very close to the experimentally determined structures for the true complexes (Lugan, pers. comm.). These observations provide support for the earlier use of model complexes to describe the electronic structures of **1** and **2**.

Analysis of the molecular orbitals of complexes **1** and **2** shows picture of bonding similar to that previously derived from approximate molecular orbital calculations. In the chelating complex **1**, the largely metal-dπ based highest occupied molecular orbital (HOMO) receives little stabilization due to the lack of a π-acceptor orbital of suitable symmetry on either the carbene or carbonyl ligands. Consequently only two of the three dπ orbitals on the metal center are strongly stabilized resulting in an essentially non-bonding HOMO. The orbital energies of complex **2** indicate significant stabilization of all three dπ orbitals on the metal center, as the orientation of the ligands allows each of the dπ orbitals to back-donate to either the carbene or carbonyl ligands. The ADF calculated energies of the frontier dπ orbitals for complexes **1** and **2** are given in Table 1. The energies from Fenske-Hall calculations for model complexes **1a** and **2a** (Graham 2003) are also included for

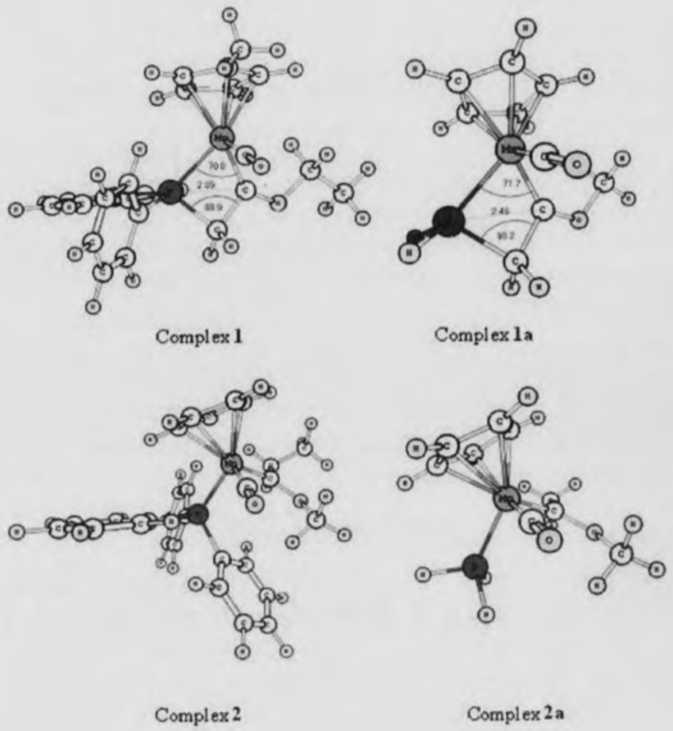

Fig. 1. Optimized Geometries for Complexes **1**, **1a**, **2**, and **2a**.

Table 1. Calculated ADF and Fenske Hall (Graham 2003) Frontier Orbital Energies (SHOMO = Second Highest Occupied Molecular Orbital, THOMO = Third Highest Occupied Molecular Orbital).

	HOMO	SHOMO	THOMO
Complex 1 (ADF–LDA/TZP)	-3.47	-4.12	-4.74
Complex 2 (ADF-LDA/TZP)	-4.24	-4.59	-4.60
Model for Complex 1a (Fenske-Hall)	-4.26	-5.37	-6.69
Model for Complex 2a (Fenske-Hall)	-5.35	-5.85	-6.12

comparison. Although the energies of the orbitals obtained from ADF and Fenske-Hall calculations are quite different (as would be expected considering the very different levels of theory), the orbital energies for each complex/model complex follow similar patterns as described above.

For the non-chelating model complex **2a** two energy minima and two transition states were identified using DFT geometry optimizations. As would be expected, the energy minima occur at the two orientations of the carbene ligand that are parallel to the CO ligand. The transitions states occur at the two orientations where the carbene ligand is perpendicular to the CO. The variation in total energy (relative to atomic fragments) with angle is illustrated in Fig. 2. An estimate of 45 kJ/mole was calculated for the rotational barrier for the carbene ligand in the model complex.

The above analysis supports the proposal that the observed differences in properties of complexes **1** and **2** arise largely from the orientation of the carbene ligand relative to the carbonyl ligand. Further work incorporating exchange and correlation corrections is being carried out to calculate structures at higher levels of theory.

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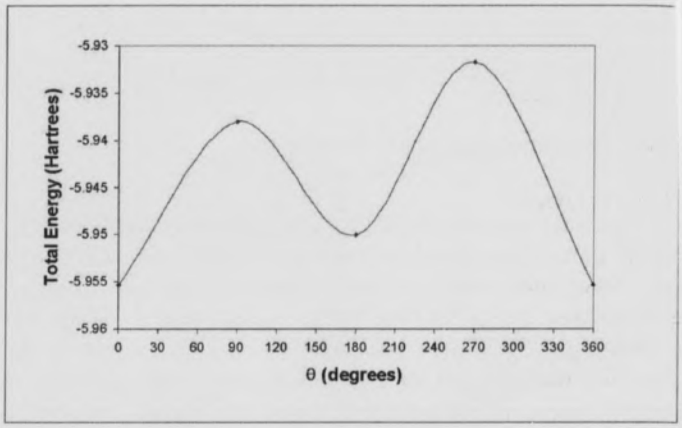

Fig. 2: Relative Total Energy Variation with Carbene – CO Angle for Complex **2a**.

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A Complex Stand on the White River National Wildlife Refuge: Implications for Bottomland Hardwood Old Growth

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With the possible re-discovery of the ivory-billed woodpecker (*Campephilus principalis*), interest has increased in the habitat requirements for the species and the current state of these habitats (Fitzpatrick et al. 2005). Tanner (1942) indicated that the ivory-billed woodpecker needs large, decadent trees for foraging. Trees in such decline provide habitat for wood-boring beetles, whose grubs are a primary food-source for the ivory-billed woodpecker. Old trees in various states of decline are an integral part of old-growth forests (Davis 1996, Oliver and Larson 1996). Unfortunately, we have little information on the species composition and structure of bottomland hardwood old-growth forests, especially in the Lower Mississippi Alluvial Valley. Therefore, the objective of this study was to determine the species composition of a potential old-growth bottomland hardwood stand located in east-central Arkansas near the possible sightings and recordings of the ivory-billed woodpecker.

Location.—The study site is located on about 50 ha within the 280-ha Sugarberry Research Natural Area in the White River National Wildlife Refuge in Desha County, AR within the unprotected lands along Scrubgrass Bayou and the White River in the Lower Mississippi Alluvial Valley (34°06’ north, 91°05’ west). The site is characterized by ridge and swale topography due to channel migration of the White River (Mitsch and Gosselink 1986). Soils vary but are primarily composed of Commerce silt loam (fine-silty, mixed, superactive, nonacid, thermic Fluvaquentic Endoaquepts) and Robinsonville very fine sandy loam (coarse-loamy, mixed, superactive, nonacid, thermic Typic Udifluvents) on the ridge tops to Sharkey clay (very-fine, smectitic, thermic Chromic Epiaquerts) in the swales. Climatically, the site has hot, humid summers and mild winters (SCS 1972). The average monthly high temperature is 24.5° C and peaks in July and August (34.9° C) and the average monthly low temperature is 11.1° C with the low occurring in January (1.6° C, SCS 1972). Precipitation averages 1,321 mm per year with the greatest monthly average in March (144 mm) and the lowest monthly average in October (68 mm) (SCS 1972). Past activities in the stand may have included light cutting for firewood around 1900 when paddle boats used Scrubgrass Bayou for traveling from the White River to the Mississippi and Arkansas rivers.

Measurements and Analyses.—Twenty north-south transects were installed on the eastern and southern portions of the Sugarberry Research Natural Area beginning 50 m from the edge of Scrubgrass Bayou. Transects were located 100 m apart and points were established at 50 m intervals along each transect. Most transects contained only four or fewer points before reaching a large beaver pond. No points were taken in the water impounded area. Twenty points were randomly selected from the 93 total to establish 0.1-ha circular tree plots. All trees greater than 10 cm DBH (diameter at breast height, 1.4 m above the ground) were tallied by species, DBH, and crown class (dominant, codominant, intermediate, or suppressed; Smith et al. 1997). Importance values, the sum of relative density and relative dominance, were calculated for each plot and averaged across all plots (Curtis and McIntosh 1991, Skeen 1973).

Six-hundred and twenty three trees greater than 10 cm DBH was tallied from 19 species in this study. The two most prominent species were sugarberry (*Celtis laevigata* Willd.) and sweet pecan (*Carya illinoensis* (Wang.) K. Koch; Table 1). Other important species included overcup oak (*Quercus lyrata* Walt.), Nuttall oak (*Q. nuttallii* Palmer), and green ash (*Fraxinus pennsylvanica* Marsh.), all with importance values greater than 10 (Table 1).

A key characteristic of most bottomland hardwood old-growth stands is large tree diameters (Lynch 1996). Noteworthy in the Sugarberry Research Natural Area were 3 trees greater than 100 cm DBH – a 143 cm American sycamore (*Platanus occidentalis* L.), a 119 cm Nuttall oak and a 102 cm overcup oak. Three additional trees not located on the tree plots but measured were a 185 cm eastern cottonwood (*Populos deltoides* Bartr. ex Marsh.), a 145 cm American elm (*Ulmus americana* L.), and a 76 cm common persimmon (*Diospyros virginiana* L.; a large DBH for this species).

Mean number of trees per hectare was 311.5 (standard error = 28.4) and mean basal area was 30.4 $m^2\ ha^{-1}$ (standard error = 2.5). The trees-per-hectare value is within the range of old-growth attributes as described by Meadows and Nowacki (1996) for eastern riverfront forests. The basal area value is low relative to Meadows and Nowacki (1996), probably due to the number of canopy gaps located throughout the Sugarberry Research Natural Area, but is within the values reported for other bottomland hardwood old-growth forests (Jackson 1969, Phillippe and Ebinger 1973, Ramp 1990, Devall and Ramp 1992, Roovers and Shifley 1997).

Nineteen percent of the sampled trees in the study area were classified in dominant or codominant crown classes, that is, with a majority of their crowns in the upper canopy (Table 2). Species with at least 25 percent of their crowns classed as dominant or codominant included American sycamore, green ash, honey-locust (*Gleditsia triacanthos* L.), cedar elm (*U.*

Table 1. Relative density, relative dominance, and importance values for trees located on the Sugarberry Research Natural Area, Desha County, AR.

Species	Relative Density	Relative Dominance	Importance Value
boxelder (*Acer negundo* L.)	0.48	0.20	0.68
silver maple (*Acer saccharinum* L.)	0.32	0.24	0.56
water hickory (*Carya aquatica* (Michx. f.) Nutt.)	1.28	1.15	2.43
sweet pecan (*Carya illinoensis* (Wang.) K. Koch)	20.55	19.19	39.74
sugarberry (*Celtis laevigata* Willd.)	30.39	33.82	64.21
hawthorn (*Crataegus* spp.)	0.16	0.01	0.17
common persimmon (*Diospyros virginiana* L.)	4.17	2.58	6.75
swamp privet (*Forestiera acuminata* (Michx.) Poir)	1.28	0.15	1.43
green ash (*Fraxinus pennsylvanica* Marsh.)	5.46	9.98	15.44
honey-locust (*Gleditsia triacanthos* L.)	0.48	0.44	0.92
deciduous holly (*Ilex decidua* Walt.)	1.12	0.11	1.23
red mulberry (*Morus rubra* L.)	1.12	0.13	1.25
American sycamore (*Platanus occidentalis* L.)	0.16	2.64	2.80
overcup oak (*Quercus lyrata* Walt.)	14.45	17.36	31.81
Nuttall oak (*Quercus nuttallii* Palmer)	11.24	6.88	18.12
winged elm (*Ulmus alata* Michx.)	0.16	0.09	0.25
American elm (*Ulmus americana* L.)	4.94	3.99	8.93
cedar elm (*Ulmus crassifolia* Nutt.)	1.12	0.85	1.97
slippery elm (*Ulmus rubra* Muhl.)	1.12	0.19	1.31

crassifolia Nutt.), water hickory (*C. aquatica* (Michx. f.) Nutt.), and overcup oak. Species which were completely overtopped by the overstory included boxelder (*Acer negunda* L.), hawthorns (*Crataegous* spp.), deciduous holly (*Ilex decidua* Walt.), red mulberry (*Morus rubra* L.), and slippery elm (*U. rubra* Muhl.), all shade-tolerant, understory species.

Oliver (1981) described four stages of stand development following a major disturbance. The stand initiation stage immediately follows the disturbance when regeneration of the site begins. The stem exclusion stage begins when regeneration can no longer become established due to the intense competition among trees for available growing space. The understory reinitiation stage begins following a relatively long period of growth and mortality during the stem exclusion stage. Mortality of a few overstory trees releases growing space, thereby allowing tree regeneration to become established in the understory. The final stage of stand development, old growth, occurs when continued mortality in the overstory allows regeneration to eventually grow into the overstory canopy. These stages of stand development progress from an even-aged stand structure to an uneven-aged stand structure with trees of various ages and diameters occupying different canopy strata. Old-growth stands are further characterized as containing canopy gaps of different ages and sizes, depending on the number of trees that have died or fallen in a disturbance. The old-growth stage of stand development has also been called the steady-state stage of ecosystem development where total stand biomass and nutrient cycling fluctuate around a consistent mean (Bormann and Likens 1979). Oliver (1981), Meadows (1994), and Oliver and Larson (1996) state that the old-growth stage of stand development is rarely achieved due to the long time period necessary to reach these stand structures and the likelihood that a major disturbance will set the stand back to an earlier stage of development.

We hypothesize that the stand at the Sugarberry Research Natural Area is in the old-growth stage of stand development. In addition to the large tree diameters, many snags occur throughout the stand and large coarse woody debris exists on the forest floor. Snags and coarse woody debris are important structural components in many old-growth forests (Maser and Trappe 1983, Spetich et al. 1999, Fan et al. 2003). Further study is needed to quantify these structural characteristics and relate them to other studies of old-growth in eastern hardwood forests.

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Table 2. Percent of trees by crown class within each species located on the Sugarberry Research Natural Area, Desha County, AR.

Species	n	Dominant	Co-Dominant	Intermediate	Overtopped
		percent			
boxelder	3	0	0	0	100
silver maple	2	0	0	50	50
water hickory	8	25	0	13	62
sweet pecan	128	13	9	12	66
sugarberry	192	7	11	32	50
hawthorn	1	0	0	0	100
common persimmon	26	8	4	31	57
swamp privet	8	0	0	13	87
green ash	34	21	26	41	12
honey-locust	3	33	0	0	67
deciduous holly	7	0	0	0	100
red mulberry	7	0	0	0	100
American sycamore	1	100	0	0	0
overcup oak	90	11	14	26	49
Nuttall oak	70	1	7	30	62
winged elm	1	0	0	100	0
American elm	28	7	0	18	75
cedar elm	7	14	14	0	72
slippery elm	7	0	0	0	100

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Noteworthy Geographic Distribution Records for the Golden Topminnow, *Fundulus chrysotus* (Cyprinodontiformes: Fundulidae), from Arkansas

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The golden topminnow, *Fundulus chrysotus* (Günther), is a small killifish that is distributed in the Coastal Plain from the Santee River drainage of South Carolina through Georgia, Florida, Alabama, Mississippi, and Louisiana, and northeastward from the Trinity River drainage of Texas and Oklahoma through eastern Arkansas up the Mississippi River Embayment to Tennessee, Kentucky and Missouri (Shute 1980, Etnier and Starnes 1993). In Arkansas, this fish is widely scattered in all major drainages of the Gulf Coastal Plain lowlands of the southern and eastern parts of the state (Robison and Buchanan 1988). It inhabits oxbows, sluggish creeks, swamps near rivers, and shallow and quiet water in or near aquatic vegetation.

During the period from 1960 to 1987, Robison and Buchanan (1988) reported 33 localities in the state for the species; however, prior to 1960, only 3 localities were known. In addition, Buchanan et al. (2003), Buchanan (2005), and Robison (2005) reported this fish from the Red River drainage (rarely), 11 of 66 Arkansas reservoirs (1,380 specimens), and the Pine Bluff Arsenal (Jefferson County, 7 specimens), respectively. Additional fieldwork in Arkansas has revealed further distributional records in 27 counties for *F. chrysotus*, and we document 98 new locales herein.

Between August 1996 and September 2005, golden topminnows were collected with standard nylon seines (6 x 1.5 m and 9 x 1.5 m of 3.2 mm mesh) or dipnets. Fishes were preserved in 10% formalin and later transferred to 45% isopropanol. Specimens were identified in the field, verified in the laboratory, and vouchers were deposited in the collections at Southern Arkansas University, Magnolia (SAU), the University of Louisiana-Monroe Museum of Natural History (NLU), the University of Arkansas-Fort Smith (UA-FS), Arkansas State University Museum of Zoology (ASUMZ), Henderson State University (HSU) and the North Carolina State Museum of Natural Sciences-Raleigh (NCSM).

We document the collection of 3,619 *F. chrysotus* from 27 of 75 counties (36%) of Arkansas (Ashley, Bradley, Calhoun, Clark, Columbia, Crawford, Crittenden, Dallas, Desha, Drew, Hempstead, Hot Spring, Howard, Jackson, Jefferson, Lafayette, Lincoln, Little River, Lonoke, Miller, Ouachita, Poinsett, Prairie, Sebastian, Sevier, St. Francis, Union). Of those, 3,278 (90.5%) specimens were taken on 3 dates from a Red River oxbow (Fifty-one Cutoff Lake) in Hempstead County (see below). Detailed collection data on the 98 new sites are as follows: [county, specific locality (township, section, and range when available), date, collector (coauthors initials HWR and TMB), museum accession number (if known), and number of specimens in parentheses].

ASHLEY COUNTY: Hank's Creek at St. Hwy. 52, 1.3 km. W jct. U.S. 82. 26 March 1988. Hutchins et al. SAU (1); Hank's Creek, 0.8 km. W St. Hwy. 81 on U.S. 82. 8 May 1991. J. Pennington and J. McLead. NLU 65400 (1); Hank's Creek, 0.4 km. W St. Hwy. 81 on U.S. 82. 9 May 1991. J. Pennington and J. McLead. NLU 65385 (1); Lucas Lake, Crossett (Sec. 30, T18S, R8W). June 1995. R. Tumlison. HSU 1116 (30); Ouachita River, 10.5 km. W Crossett on U.S. 82 (Sec. 14, T18S, R10W). 11 April 1997. J. Thompson. HSU 2003 (5); Wilson Brake Lake (Bayou Bartholomew oxbow). 22 July 1998. TMB. UA-FS (3).

BRADLEY COUNTY: Snake Creek at Broad (Sec. 30, T16S, R9W). 15 April 1999. HWR. SAU (1); Moro Creek at Moro Bay State Park (Sec. 21, T16S, R12W). 10 June 2005. HWR. SAU (3); L'Aigle Creek off co. rd. 2, 14.5 km. S Hermitage (Sec.18, T16S, R10W). 10 June 2005. HWR. SAU (1).

CALHOUN COUNTY: Champagnolle Creek (Sec. 27, T15S, R14W). 6 July 2001. HWR. SAU (1); Locust Bayou at St. Hwy. 4 (Sec. 30, T13S, R15W). 7 May 2005. HWR. SAU (2). Moro Creek at St. Hwy. 160 (Sec. 9, T16S, R12W). 10 June 2005. HWR. SAU (2).

COLUMBIA COUNTY: Dorcheat Bayou off co. rd. 15, 4.8 km. SW Philadelphia (Sec. 16, T18S, R22W). 4 September 1993. HWR. SAU (1); Cypress Creek at St. Hwy. 19, 16.1 km. SW Macedonia (Sec. 7, T20S, R21W). 6 June 2003. HWR. SAU (1); Dorcheat Bayou at St. Hwy. 160, ca. 6.4 km. E Taylor (Sec. 9, T19S, R22W). 19 May 2004. HWR. SAU (3); Horsehead Creek at U.S. 79, 9.7 km. E Magnolia (Sec. 17, T18S, R20W). 14 May 2005. HWR. SAU (1).

CLARK COUNTY: Tupelo Creek at St. Hwy. 51, 3.2 km. E Arkadelphia. 13 March 1996. TMB. UA-FS (6); McNeeley Creek, 6.4 km. S Beirne off St. Hwy 51 (Sec. 31, T10S, R20W). 20 April 1997. R. Tumlison. HSU 2155 (4); Gurdon Lake. 17 August 2000. TMB. UA-FS (1).

CRAWFORD COUNTY: Arkansas River, 0.8 km. below Lock and Dam No. 13 at Ozark Pool (Trimble). 2 October 1998. TMB. UA-FS (1); Arkansas River backwater along access rd. to Haroldton access, just above Lock and Dam No. 13, Pool 13 (Trimble). 13 and 15 April 2005. TMB. UA-FS (2). These specimens document the first records of *F. chrysotus* from Ozark Pool and Pool 13, respectively, and, as such, represents the northwesternmost distribution in the state for this species.

CRITTENDEN COUNTY: Horseshoe Lake (Mississippi River oxbow) along St. Hwy. 147, 22.5 km. SW West Memphis. 6 August 1997. TMB. UA-FS (4)

DALLAS COUNTY: Moro Creek (specific locality not known). 21 April 2005. L. Self. HSU 3080 (1).

DESHA COUNTY: Arkansas River backwater at RM 22.8 above Pendleton Bridge. 30 July 2001. TMB. UA-FS (14); Morgan Point Bendway, old Arkansas River channel below Lock and Dam No. 2. 30 July 2002. TMB. UA-FS (4).

DREW COUNTY: Cut-Off Creek at St. Hwy. 35, 11.3 km. E Collins (Sec. 31, T13S, R4W). 26 March 1998. Hutchins et al. SAU (3); Bayou Bartholomew at St. Hwy. 35, 6.4 km. E Collins (Sec. 28, T13S, R4W). 28 September 1999. HWR. SAU (1); Hazel Creek at St. Hwy. 8 (Sec. 1, T14S, R9W). 10 June 2000. HWR. SAU (1).

HEMPSTEAD COUNTY: Millwood Lake at Saratoga Landing, 0.8 km. W Saratoga (Sec. 6, T12S, R27W). 10 August 1990. HWR. SAU (1); Bois D'Arc Creek at U.S. 67, 11.3 km. SW Hope (Sec. 8, T13S, R25W). 15 June 1992. HWR. SAU (1); Fifty-one Cutoff Lake (Red River oxbow), 6.4 km. SE Fulton. 14 August 1996, 17 August 1998, and 4-5 August 2000. TMB. UA-FS (3,278); Grassy Lake 4.8 km. E Beard's Lake use area (Sec. 22, T12S, R27W). 11 August 1997. TMB. UA-FS (1); Beard's Lake below Millwood Dam. 23 July 1999. TMB. UA-FS (2); Red Lake (Red River oxbow), 1.6 km. below Millwood Dam, 12-13 July 2001. TMB. UA-FS (8). These specimens fill a distributional gap in southwestern Arkansas.

HOT SPRING COUNTY: Saline Bayou, 4.0 km. S Friendship (Sec. 23 T6S R19W). 14 February 1997. R. Tumlison. HSU 2721 (2).

HOWARD COUNTY: Plum Creek at St. Hwy. 355, ca. 3.2 km. S Tollette (Sec. 20, T11S, R29W). 11 August 1990. HWR. SAU (1); Mine Creek off co. rd. 2, 1.6 km. W Tollette (Sec. 6, T11S, R29W). 11 August 1990. HWR. SAU (2); Saline River off co. rd. 2, 3.2 km. W Schaal (Sec. 10, T11S, R28W). 12 June 2000. HWR. SAU (1); Plum Creek, 0.8 km. above Millwood Lake. 14 July 2004. TMB. UA-FS (3). The collection of these specimens help extend the range westward from previous sites in southcentral Arkansas.

JACKSON COUNTY: Unnamed tributary, 8.0 km. NW Swifton (Sec. 15, T14N, R2W). 4 January 1994. A. Holt. ASUMZ 12918 (16). These specimens represent a northwestward extension of their range in Arkansas.

JEFFERSON COUNTY: Bayou Meto off St. Hwy. 114, confluence with Arkansas River. 27 September 2002. TMB. UA-FS (2).

LAFAYETTE COUNTY: Bodcau Creek, 6.4 km. N Lewisville, AR (Sec. 14, T15S, R24W). 5 July 1992. HWR. SAU (2); Mays Old River Lake (Red River oxbow), 7.2 km. N U.S. 82. 16 August 1997. TMB. UA-FS (3); Wallace Lake (Red River oxbow), 3.2 km. N U.S. 82. 20 August 1998. TMB. UA-FS (7); Unnamed Red River oxbow (Sec. 6, T19S, R26W). 13 July 2001. TMB. UA-FS (7); Lake Erling at St. Hwy. 160 (Sec. 31, T19S, R23W). 14 June 2002. HWR. SAU (1); Swan Lake (Red River oxbow) (Sec 22, T17S, R25W). 9 July 2002. TMB. UA-FS (14); Jones Lake (Red River oxbow) (Sec. 34, T19S, R26W). 10 July 2003. TMB. UA-FS (11); Bodcau Creek at St. Hwy. 360 (Sec. 1, T16S, R24W). 15 July 2004 and 11 July 2005. TMB. UA-FS (4).

LINCOLN COUNTY: Bayou Bartholomew at Garrett Bridge (Sec. 6, T10S, R5W). 3 June 1989. HWR. SAU (2); Bayou Bartholomew, 6.4 km. N Star City (Sec. 21, T8S, R7W). 7 July 2003. HWR. SAU (2); Bayou Bartholomew at St. Hwy. 293 (Sec. 15, T9S, R6W). 7 July 2003. HWR. SAU (1).

LITTLE RIVER COUNTY: Little River relief at St. Hwy. 41, 6.6 km. SW Horatio (Sec. 10, T10S, R32W). 31 May 2003. W. Starnes et al. NCSM 37414 (2); Caney Creek at St. Hwy. 41, 4.0 km. Billingsley's Corner (Sec. 28, T10S, R32W). 6 June 1989. HWR. SAU (1); Cypress Creek at St. Hwy. 234 in Winthrop (Sec. 7, T11S, R31W). 6 June 1989. HWR. SAU (1); Flat Creek at St. Hwy. 234, 3.2 km. E Winthrop (Sec. 21, T11S, R31W). 10 July 1990. HWR. SAU (1); spring-fed backwater of Red River, 2.4 km. below St. Hwy. 41. 25 July 1995. TMB. UA-FS (7); Allen Creek on Richmond Road, 4.0 km. SW Ashdown. 13 August 1996. TMB. UA-FS (2); Choctaw Lake (Red River oxbow), 3.2 km W St. Hwy. 41. 19 August 1998. TMB. UA-FS (2); Grassy Lake (Red River oxbow) (Sec. 29, T13S, R29W). 11 July 2001. TMB. UA-FS (50); Little River backwater at U.S. Hwy. 71, 3.2 km. N Wilton (Sec. 24, T11S, R29W). 5 October 2001. HWR. SAU (4); Red River, 3.2 km. S St. Hwy. 108 near Oklahoma state line. 7 July 2003. TMB. UA-FS (1); Beard's Lake below Millwood Dam at U.S. 71 access. 8 July 2003. TMB. UA-FS (2). These 72 specimens provide additional records for the species in far southwestern Arkansas. Interestingly, one county westward in adjacent Oklahoma, *F. chrysotus* is restricted to Little River and Red River tributaries of McCurtain County (Miller and Robison 2004).

LONOKE COUNTY: Bayou Meto at St. Hwy. 13 bridge. 2 October 2002. TMB. UA-FS (1).

MILLER COUNTY: Kelly Bayou, 6.4 km. SW Doddridge (Sec. 9, T20S, R27W). 3 September 1990. HWR. SAU (1); West Fork Kelly Bayou, 1.6 km. S Brightstar (Sec. 28, T18S, R28W). 3 September 1990. HWR. SAU (1); Turkey Creek at St. Hwy. 237, 6.4 km. N Brightstar (Sec. 28, T18S, R28W). 14 November 1993. HWR. SAU (1); Sulphur River, 4.8 km. above mouth in spring-fed backwater. 12 August 1996. TMB. UA-FS (4); Second Old River Lake (Red River oxbow) (Secs. 1-2, T15S, R26W). 20 July 1999. TMB. UA-FS (2); Red River backwater, 12.8 km. N Fulton (Sec. 22, T14S, R26W). 12 August 1996 and 17 August 1998. TMB. UA-FS (4); Adams Cutoff Lake (Red River oxbow), 3.2 km. SW Fulton. 19 August 1997. TMB. UA-FS (32); Kuykendall Lake (Red River oxbow), 4.8 km. SW Fulton. 9 July 2001. TMB. UA-FS (15); Borrow pit near Red River levee (Sec. 13, T15S, R26W). 10 July 2001. TMB. UA-FS (2); Cypress City Lake (Red River oxbow) (Secs. 6-8, T16S, R25W). 9 July 2002. TMB. UA-FS (15); Caswell Lake (Red River oxbow) (Secs. 27-28, T18S, R26W). 11 July 2002. TMB. UA-FS (4); unnamed slough off Palmer Lake (Sec. 6, T20S, R26W). 12 July 2002. TMB. UA-FS (1); Palmer Lake (Red River oxbow) (Sec. 6, T20S, R26W). 12 July 2002. TMB. UA-FS (7); Willow Bend Lake (Red River oxbow) (Sec. 7, T14S, R26W). 16 July 2004. TMB. UA-FS (7).

MONROE COUNTY: Caney Slash, tributary of Bayou DeView at RR right-of-way, 3.2 km. W Brinkley. 4 June 1988. TMB. UA-FS (1); Boat Gunwale Slash at St. Hwy. 17, 6.4 km. S Holly Grove. 10 November 1989. TMB. UA-FS (3).

OUACHITA COUNTY: En Core Fabre Bayou, ca. 1.6 km. N Camden (Sec. 10, T13S, R17W). 22 August 1994. HWR. SAU (2); Bragg Lake 2.1 km. SE Bragg City, on St. Hwy. 24 (Sec. 33 T12S R18W). 12 April 1997. J. Thompson. HSU 2050 (1); Ben Davis Lake at Snow Hill (Sec. 28, T15S, R15W). 20 October 2004. HWR. SAU (1).

POINSETT COUNTY : L'Anguille River, 6 km. NW Harrisburg (Sec. 4, T11N, R3E). 15 November 1987. J. McLean. ASUMZ 11913 (1); L'Anguille River, 8 km. SW Harrisburg off St. Hwy. 214 (Sec. 20, T10N, R3E). 15 November 1987. J. McLean. ASUMZ 11889 (1). These specimens represent a northwestward extension of the range of this topminnow from previous sites in northeastern Arkansas.

PRAIRIE COUNTY: White River at DeValls Bluff boat ramp, DeValls Bluff (Sec. 17, T2N, R4W). 11 April 1998. N. H. Douglas and J. J. Hoover. SAU (2).

SEVIER COUNTY: Rolling Fork River at St. Hwy. 24, 4.8 km. W Horatio (Sec. 28, T9S, R32W). 8 October 1989. HWR. SAU (1); Millwood Lake at Paraloma Landing at end of St. Hwy. 234 (Sec. 29, T11S, R28W). 18 June 2002. HWR. SAU (1); Little River at end of St. Hwy. 317, 4.8 km. S Brownstown (Sec. 25, T11S, R29W). 19 June 2002. HWR. SAU (2); Cossatot River, 2.4 km. above confluence of Millwood Lake. 13 July 2004. TMB. UA-FS (3). These specimens extend the range of *F. chrysotus* to the north from previous records in southwestern Arkansas.

ST. FRANCIS COUNTY: L'Anguille River off St. Hwy. 261, 4.0 km. W Caldwell (Sec. 3, T5N, R2E). 21 February 1988. J. McLean. ASUMZ 11691 (2).

UNION COUNTY: Grand Marais boat ramp on Ouachita River (Sec. 37, T19S, R10W). 30 January 1999. A. Rainwater. HSU 2253 (2); Cornie Bayou at U.S. 167, 1.6 km. N Junction City (Sec. 36, T19S, R16W). 10 March 2000. HWR. SAU (1); Lapoile Creek at U.S. 82, 16.1 km. NE Strong (Sec. 18, T18S, R10W). 18 May 2001. HWR. SAU (1); LaPere Creek at St. Hwy. 129, 3.2 km. SW of Huttig (Sec. 35, T19S, R11W). 11 June 2002. HWR. SAU (1); Calion Lake at Calion (Sec. 22, T16S, R14W). 17 May 2003. HWR. SAU (3); Big Cornie Creek at St. Hwy. 15 (Sec. 35, T19S, R18W). 6 October 2003. HWR. SAU (2).

This new geographic data provides a much needed update on the distribution of *F. chrysotus* in the state prior to the much-anticipated second edition of Robison and Buchanan's, *Fishes of Arkansas*. From our numerous collections it is evident that this topminnow is common in many of the oxbows of southern Arkansas and its overall range in the state is greater than previously known. Interestingly, the golden topminnow is a species of "special concern" in Alabama (Boschung et al. 2004) and some other coastal states (except Florida) where it is confined to coastal streams. In addition, *F. chrysotus* is of "special concern" in states near the northern extent of its range, Kentucky, Missouri, and Tennessee (Burr and Warren 1986, Etnier and Starnes 1993, Pflieger 1997). Results of our study indicate that Arkansas most likely has some of the most secure populations of this topminnow and the third largest geographic area of occurrence of any state in its range, after Florida and Louisiana (Douglas 1974, Burgess et al. 1977, Loftus and Kushlan 1987).

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Distribution of Arkansas's Bottomland Bats

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The lower Mississippi River valley originally was the site of 10 million hectares of bottomland hardwood forests (Tiner 1984), but today less than half remain (Hefner and Brown 1985). Annual palustrine wetland loss has been estimated at 178,000 ha as of 1970, the majority of which (87%) is from conversion to agriculture (Tiner 1984).

These forested floodplains are important roosting and foraging sites for bottomland bats, including Rafinesque's big-eared bat (*Corynorhinus rafinesquii*; Gooding and Langford 2004) and the southeastern myotis (*Myotis austroriparius*; Mirowsky and Horner 1997), and their loss could be detrimental to populations of these poorly known (Sealander and Heidt 1990, Menzel et al. 2001) and rare bats (Harvey and Redman 2002). Yet only limited knowledge of the bats' distributions exist for the southeast (Mirowsky and Horner 1997, Fokidis et al. 2005).

To obtain current and detailed state distributions for these species, we conducted a mist-net survey of the forested bottomlands within their projected range in the Gulf Coastal Plain and Mississippi River Delta regions of Arkansas. We targeted primarily public lands such as Arkansas Game and Fish Commission Wildlife Management Areas (WMA), National Wildlife Refuges (NWR), U.S. Army Corps of Engineers' Parks, and Arkansas State Parks.

Our protocol mandated a minimum of 1 night and a maximum of 3 nights of netting in at least 1 location in each bottomland county that was lacking a documented record for either the southeastern myotis or Rafinesque's big-eared bat. Mist-netting was conducted from 6 May to 11 July 2005. Mist-nets were normally 2.6 x 6-12 m in size, although occasional use of high nets (two 2.6 x 6m nets united and placed one above the other) were made. Nets, supported by metal conduit poles, were set perpendicular to the expected flight path and rose from ground or water surface level upwards. Standard netting procedures were followed regarding net placement; and efforts were concentrated on potential bat foraging and gleaning areas including forest corridors, such as logging roads and access trails, and areas of standing water, such as ponds, ditches, and sloughs. The number of nets placed each night ranged from 2 to 9 depending upon terrain. Each net setup was maintained for a minimum of 5 hours beginning at dusk. Nets were checked for bats every 15 minutes. Species, gender, reproductive status, mass, forearm length, and presence/absence of external parasites were recorded for each bat captured.

We surveyed 22 sites in 16 counties in the Gulf Coastal Plain and the Mississippi River Delta regions of Arkansas during 2005. Trapping efforts totaled 56 nights or 384 net-nights, with a more intensive continuous trapping effort put forth for 30 of those nights (31 May to June 29) by three investigators who netted three separate areas simultaneously. A net-night is defined as the sum of the number of nets open for 1 night of trapping. The 2005 season resulted in the capture of 401 individual bats and 6 recaptures and provided the 24 new county records detailed below.

Corynorhinus rafinesquii

1) Ashley Co. – Felsenthal NWR, 33°05' N 92°06' W, 18 May 2005. Logging road just outside boat launch parking lot. Two females, one pregnant and one non-reproductive (NR).

2) Chicot Co. – Lake Boggy Bayou, 33°56' N 91°45' W, 11 July 2005. Pond adjacent to road. NR male.

3) Crittenden Co. – Wapanocca NWR, 35°05' N 90°12' W, 20 June 2005. Pond on edge of farm field. NR Male.

4) Hempstead Co. – Bois d'Arc WMA, 33°35' N 93°44' W, 05 July 2005. Pond in forest interior. NR male.

5) Lincoln Co. – Cane Creek boat dock, AGFC, 33°56' N 91°45' W, 09 July 2005. Stream and stream access from boat dock. NR male.

6) Pike Co. – Saline Creek, 33°58' N 93°34' W, 07 July 2005. Stream near Highway 301 Bridge. NR male.

Eptesicus fuscus

7) Lincoln Co. – Cane Creek boat dock, AGFC, 33°56' N 91°45' W, 09 July 2005. Stream and stream access from boat dock. NR male.

8) Ashley Co. – Felsenthal NWR, 33°05' N 92°06' W, 18 May 2005. Logging road. Scrotal male and NR male.

Lasiurus borealis

9) Chicot Co. – Lake Boggy Bayou, 33°29' N 91°14' W, 11 July 2005. Pond adjacent to road. Two NR females.

10) Lincoln Co. – Cane Creek boat dock, AGFC, 33°56' N 91°45' W, 09 July 2005. Stream and stream access from boat dock. Two NR males.

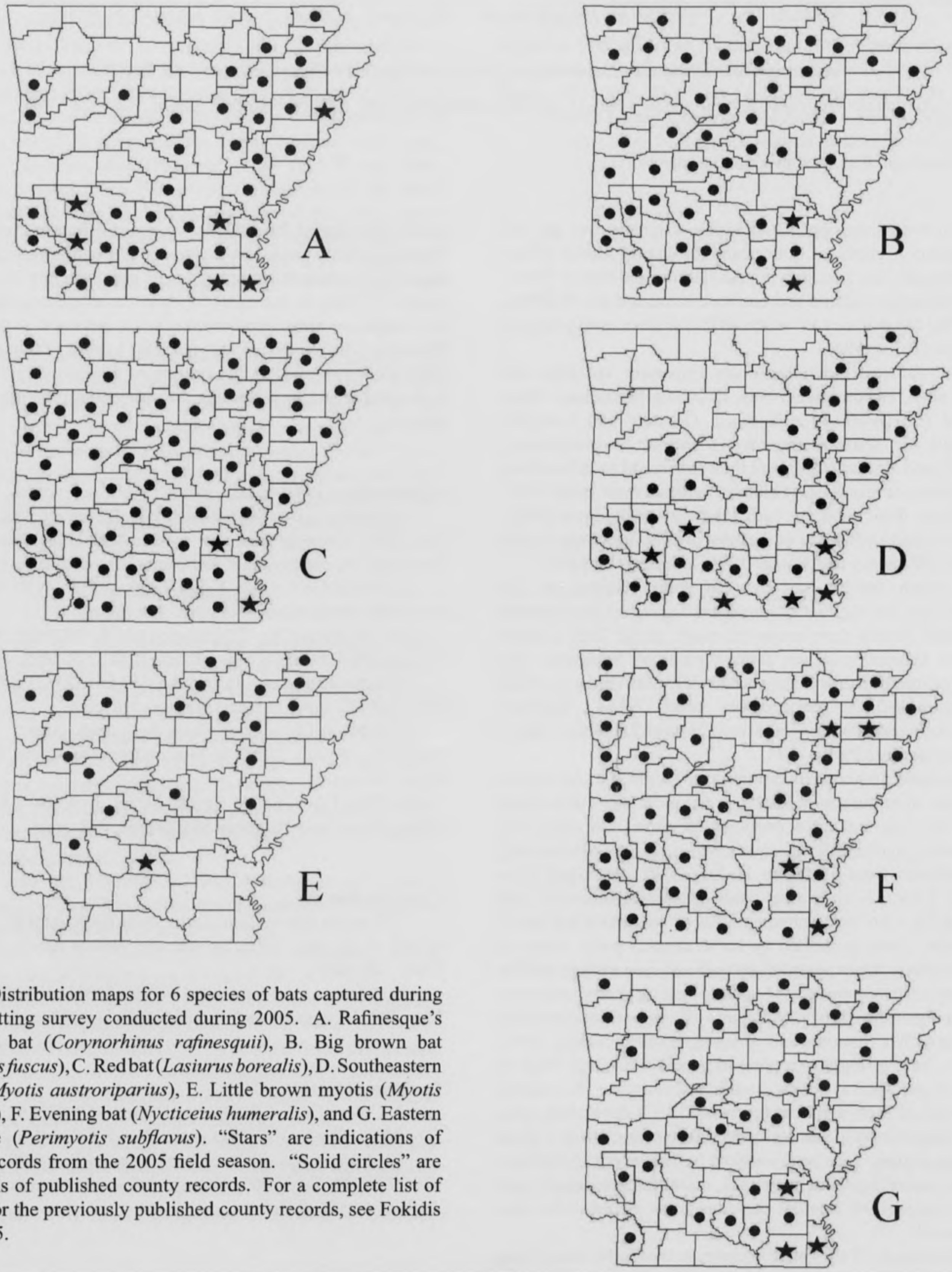

Fig. 1. Distribution maps for 6 species of bats captured during a mist-netting survey conducted during 2005. A. Rafinesque's big-eared bat (*Corynorhinus rafinesquii*), B. Big brown bat (*Eptesicus fuscus*), C. Red bat (*Lasiurus borealis*), D. Southeastern myotis (*Myotis austroriparius*), E. Little brown myotis (*Myotis lucifugus*), F. Evening bat (*Nycticeius humeralis*), and G. Eastern pipistrelle (*Perimyotis subflavus*). "Stars" are indications of county records from the 2005 field season. "Solid circles" are indications of published county records. For a complete list of sources for the previously published county records, see Fokidis et al. 2005.

Myotis austroriparius

11) Ashley Co. – Casey Jones WMA, 33°20' N 91°58' W, 17 May 2005. Unnamed logging road. Two NR males.

12) Chicot Co. – Lake Boggy Bayou, 33°29' N 91°14' W, 11 July 2005. Pond adjacent to road. One NR male.

13) Clark Co. – Big Timber WMA, 33°49' N 93°09' W, 12 May 2005. Crossing of creek and gravel road. Two NR males.

14) Desha Co. – Sutton Bayou, 33°53' N 91°11' W, 10 July 2005. Slough near bridge. One NR male.

15) Hempstead Co. – Bois d'Arc WMA, 33°35' N 93°44' W, 05 July 2005. Pond in forest interior. One lactating female and 1 NR male.

16) Union Co. – Felsenthal NWR, 33°11' N 92°44' W, 21 May 2005. Slough off of access trail. Lactating female.

Myotis lucifugus

17) Dallas Co. – Private land, 33°54' N 92°53' W, 08 July 2005. Gravel road. NR female.

Nycticeius humeralis

18) Chicot Co. – Lake Boggy Bayou, 33°29' N 91°14' W, 11 July 2005. Pond adjacent to road. NR female, NR male.

19) Lincoln Co. – Bayou Bartholomew, 33°56' N 91°46' W, 28 May 2005. Backwater from lake. Pregnant female.

20) Poinsett Co. – Bayou de View, 35°36" N 90°57" W, 31 May 2005. Handicap access trail. Scrotal male.

21) Jackson Co. – Cache River NWR, 35°32" N 91°08" W, 2 June 2005. River access road. NR male.

Perimyotis subflavus

22) Ashley Co. – Casey Jones WMA, 33°21' N 91°58' W, 17 July 2005. Logging road near bayou. Pregnant female.

23) Chicot Co. – Lake Boggy Bayou, 33°21' N 91°14' W, 11 July 2005. Pond adjacent to road. NR female.

24) Lincoln Co. – Cane Creek boat dock, AGFC, 33°56' N 91°45' W, 09 July 2005. Stream and stream access from boat dock. NR male.

We collected bottomland bat species presence / absence data across the Gulf Coastal Plain and Mississippi River Delta regions of Arkansas resulting in range expansion over published records for 7 species (Fig. 1). The results of this survey indicate that *M. austroriparius* and *C. rafinesquii* have a broad distribution in Arkansas and suggest that they may not be as rare as previously believed. We are presently working on a more in-depth analysis of these species populations in northeast Arkansas.

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The Cajun Dwarf Crawfish (*Cambarellus shufeldtii*): An Intermediate Host for *Southwellina dimorpha* (Acanthocephala)

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Southwellina dimorpha Schmidt, 1973 (Polymorphidae: Acanthocephala) was originally described from juveniles and adults from the white ibis (*Eudocimus albus*) in Florida and cystacanths from cultured red crawfish, *Procambrus clarki*, from Pecan Island, Vermilion Parish, Louisiana (Schmidt (1973). The specific epithet refers to the fact that the female has 1 field of trunk spines whereas the male has 2.

Southwellina dimorpha has also been reported from *P. clarki* from St. James Parish, Louisiana (Lantz 1974). *Southwellina dimorpha* is known only from the 2 species of birds the white ibis and the whooping crane, *Grus americana* (United States National Parasite Collection USNPC 090666), from Florida. Although infrequently reported, *S. dimorpha* may occur in high enough prevalence in crawfish intermediate hosts to have a commercial impact. The survey of Lantz (1974) of *P. clarki* in 2 ponds in St. James Parish, Louisiana, was initiated when crawfish buyers in New Orleans reported the presence of a parasite which appeared as "a pinkish grain of rice near the junction of the abdominal muscle and the cephalothorax," in stock purchased from St. James Parish. Examination of 385 *P. clarki* revealed a prevalence of infection of 46% with infection intensity ranging from 1 to 79 cystacanths per crawfish, usually 1-11. Based on anecdotal evidence, Lantz (1974) suggested that complete dewatering of pond soils for a minimum of 2 months each summer may reduce the incidence of *S. dimorpha* infections.

On 3 March 2005, cystacanths of *S. dimorpha* were collected from hemocoels of 2 of 44 (4.5%) Cajun dwarf crawfish, *Cambarellus shufeldtii*, collected at Head of Island, Ascension Parish, Louisiana (30°15.769'N 90°43.325'W). One specimen, a 24 mm long male, was infected with 2 cystacanths, and another specimen, a 23 mm long male, was infected with 1 cystacanth. Cystacanths were placed in tap water to allow the proboscides to become everted, stained with acetocarmine, mounted in Canada balsam and examined by light microscopy. Voucher specimens were deposited in the Harold W. Manter Laboratory, University of Nebraska State Museum, Lincoln, Nebraska (HWML48464). No infections were found in an additional 298 individuals of *C. shufeldtii* collected from the same site on 14 June 2005. None of 417 *C. shufeldtii* collected on 16 June 2005 from the Honey Island Swamp region of St. Tammany Parish, Louisiana, were infected.

Only 4 other species of acanthocephalans have been reported from crawfish (Evans et al. 2002), although various species within the family Polymorphidae utilize crabs as intermediate hosts (Schmidt and Kuntz 1967, Schmidt 1985). *Polymorphus biziurae* was reported from *Cherax destructor* in South Australia (Johnson and Edmonds 1948, O'Donoghue et al. 1990), *Polymorphus minutus*, which normally utilizes an amphipod intermediate host, has been reported from European crawfish by Siebold (1835), and *Cambarus affinis* (Golvan 1961, Schmidt 1985) and *Polymorphus formosus* were reported from freshwater crawfish, *Macrobrachium* sp., in Taiwan (Schmidt and Kuntz 1967). In North America, the only other report of acanthocephalans occurring in crawfish was by Merritt and Pratt (1964) who found cystacanths of *Neoechinorhynchus rutili* in 3 of 154 (2%) crawfish, *Pacifastacus trowbridgi*. *Neoechinorhynchus rutili* normally utilizes an ostracod intermediate host, leading Merritt and Pratt (1964) to suggest that crawfish were serving as paratenic hosts for this parasite, becoming infected by ingestion of infected ostracods. The viability of acanthocephalan cystacanths from crawfish has never been tested.

This report extends the known intermediate host range of *S. dimorpha* to include a second species of crawfish *C. shufeldtii* and constitutes the first report of an acanthocephalan from a crawfish of the genus *Cambarellus*.

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New Distributional Records of Lampreys from Arkansas

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Lampreys (Petromyzontidae) are members of an ancient group of fishes which are unique among the vertebrates in being jawless. Four species of lampreys occur in Arkansas; the least brook lamprey, *Lampetra aepyptera* (Abbott); the American brook lamprey, *L. appendix* (DeKay); the chestnut lamprey, *Ichthyomyzon castaneus* Girard, and the southern brook lamprey, *I. gagei* Hubbs and Trautman. Lampreys are generally difficult to collect unless during spawning and therefore are not represented in museum collections as often as other species of fishes. Recent collecting in Arkansas since the publication of Robison and Buchanan (1988) has revealed a number of new locality records for each of the 4 Arkansas lampreys. This effort consolidates and updates records of lampreys collected since 1988 and adds additional natural history data on Arkansas lampreys.

Museum numbers are provided where appropriate as are locality, date, collector, number of individuals, and any other pertinent aspects dealing with the biology of the species. Museums where collections are housed and their respective abbreviations are listed as follows: Henderson State University (HSU), University of Louisiana - Monroe (NLU), and Southern Arkansas University (SAU).

***Lampetra aepyptera* (Abbott). Least brook lamprey.** Robison and Buchanan (1988) reported only 10 localities in Arkansas for *L. aepyptera*, only one of which was in the White River system. Harp and Matthews (1975) reported the first White River system records of *L. aepyptera* from Piney Creek in Fulton County, Arkansas. On 8 May 2003 2 specimens of *L. aepyptera* were collected from Mill Branch near Mt. Hershey, Newton County, Arkansas (Table 1); (Petersen and Justis 2005). These represent the first record of this species in the Buffalo River system and only the second record in the entire White River system in Arkansas. In addition, 2 records are documented from Spring River (HSU 2047) and Piney Creek (HSU 2423), respectively (Table 1). The new Piney Creek collection (HSU 2423) consisted of 3 adult specimens and represents only the second collection from Piney Creek and the fourth collection made from the White River system.

***Lampetra appendix* (DeKay). American brook lamprey.** Only 7 records of *L. appendix* were reported by Robison and Buchanan (1988) for Arkansas. All *L. appendix* records were from the White River system until Tumlison and Tumlison (1999) discovered this species in the Ouachita River system in Clark County in southern Arkansas. Adults are also known from L'Eau Frais Creek in Clark and Hot Spring counties. A total of 15 specimens collected since 1988 taken in 8 additional collections are documented in Table 1. Twelve of the 15 specimens were collected from L'Eau Frais Creek, whereas 3 specimens (HSU 2798) were taken from Piney Creek (White River system) in northern Arkansas (Table 1).

***Ichthyomyzon castaneus* Girard. Chestnut lamprey.** The chestnut lamprey is the most widely distributed lamprey species in Arkansas. Robison and Buchanan (1988) reported 54 localities for this parasitic lamprey within the state. We document an additional 22 localities for this species and 33 individuals in Arkansas including 12 specimens, which were spawning, collected from the lower Caddo River at the I-30 bridge at Caddo Valley, Arkansas (Sec. 31, T6S, R19W) (SAU collection) in Clark County on 16 April 1996 which were spawning. These adults were spawning in an excavated gravel nest approximately 1 m from shore in 45.7 cm of water. The water temperature was 16.7 °C. Chestnut lampreys were also observed (JCP) spawning in the Buffalo River near Hasty, Arkansas on 24 April 2003.

***Ichthyomyzon gagei* Hubbs and Trautman. Southern brook lamprey.** Robison and Buchanan (1988) reported 50 localities for this non-parasitic lamprey in Arkansas. Since 1988, 12 additional adult specimens and 3 identified ammocoetes were collected from 11 Arkansas localities in Clark, Franklin, Johnson, Hot Spring, Saline, and Stone counties (Table 1).

Unidentified Ammocoetes. Collections of unidentified ammocoetes taken since 1988 are provided in Table 1 from Clark, Carroll, Fulton, Madison, Marion, Newton, and Searcy counties to account for known records of lampreys at specific sites.

Table 1. Lamprey collections in Arkansas, 1988-2005.

Date	Collector	Locality, Section, Township, Range, Sample Size (parentheses), HSU Museum Number
***Lampetra aepyptera* Abbott**		
8 May 2003	J. C. Petersen	Newton Co.: Mill Branch downstream from Mitch Hill Spring near Mt. Hershey (SE ¼ of NW1/4 of Sec. 25, T16, R19 (n = 2).
18 Apr 1997	J. Thompson	Fulton Co.: Spring River, 6 mi. E of Mammoth Spring @ Hwy 63 (Sec. 16, T21N, R5W) (n = 1) HSU 2047.
20 Feb 1999	B. Fluker	Izard Co.: Piney Creek @ access road (Sec. 5, 16N, R10W) (n = 3) HSU 2423.
***Lampetra appendix* (DeKay)**		
24 Feb 1996	R. Tumlison	Clark Co.: L'Eau Frais Creek, 6 mi. SE of Arkadelphia, Hwy 7 (Sec. 1, T8S, R19W) (n = 2) HSU 1398.
31 Jan 1997	R. Tumlison	Clark Co.: L'eau Frais Creek 9 km SE of Arkadelphia, Hwy 7 (Sec. 1, T8S, R19W) (n = 2) HSU 2189.
2 Mar 1996	R. Tumlison	Clark Co.: L'Eau Frais Creek 1 mi. SE of Joan (Sec. 22, T7S, R18W) (n = 2) HSU 1401.
25 Feb 1995	R. Tumlison	Clark Co.: L'Eau Frais Creek, 6 mi SE of Arkadelphia at St. Hwy. 7 (Sec. 1, T8S, R19W) (n = 1) HSU 1400.
19 Feb 1994	J. Rutherford	Clark Co.: L'Eau Frais Creek at St. Hwy 7 near Manchester (Sec. 1, T8S, R18W) (n = 1) HSU 672.
2 Mar 1996	R. Tumlison	Clark Co.: L'Eau Frais Creek 6 mi SE Arkadelphia at St. Hwy. 7 (Sec. 1, T8S, R19W) (n = 1) HSU 1399.
22 Feb 1997	R. Tumlison	Hot Spring Co.: L'Eau Frais Creek 8 km E Donaldson at St. Hwy. 222 (Sec. 15, T6S, R17W) (n = 3) HSU 2191.
20 Feb 1999	D. Fendley and B. Fluker	Izard Co.: Piney Creek (Sec. 5, T16N, R10W) (n = 3) HSU 2798.
***Ichthyomyzon castaneus* Girard**		
16 Apr 1996	H. W. Robison	Clark Co.: Caddo River at I-30 bridge at Caddo Valley, AR (Sec. 31, T6S, R19W) (n = 12) SAU Collection.
4 May 1999	R. Tumlison D. Fendley J. Hardage	Clark Co.: Tupelo Creek at St. Hwy. 7 (Sec.35, T7S, R19W) (n = 1) HSU 2752.
6 Apr 1994	J. Nix	Clark Co.: Caddo River at St. Hwy. 84 (Sec.24,T5S, R23W) (n = 1) HSU 978.
Apr 1991	R. Tumlison B. Hesington	Clark Co.: Lower dam DeGray Lake, Caddo River (Sec. 35, T6S, R20W) (n = 1) HSU 295.
19 Apr 1994	K. Bailey	Clark Co.: Brushy Creek at DeGray Lake (Sec.8, T6S, R21W) (n = 1) HSU 856.
20 Apr 1994	M. Major C. Petty	Clark Co.: L'Eau Frais Creek, 4 mi from Manchester, Hwy 7 at 2nd bridge (NE ¼, Sec.1, T8S, R19W) (n = 1) HSU 412.
25 Mar 1994	J. Russell	Clark Co.: L'Eau Frais Creek on Hwy 7, 5.6 mi. S jct Hwys 51 & 7 (Sec.1, T8S, R19W) (n = 1) HSU 542.
4 May 1994	M. Withers	Clark Co.: L'Eau Frais Creek near Manchester (Sec. 1, T8S, R18W) (n =1) HSU 842.
17 Feb 1996	R. Tumlison	Clark Co.: L'Eau Frais Creek 9 km SE Arkadelphia, at St.Hwy. 7 (Sec.1, T8S, R19W) (n = 2) HSU 2188.
22 May 1997	G. W. Gaddis	Clark Co.: Caddo River (Sec.26, T5S, R22W) (n = 1) HSU 2135.
6 Mar 1997	D. Rowland	Garland Co.: Lake Hamilton (Sec.18, T2S, R20W) (n = 1) HSU 2139.
20 Mar 1999	D. Dyer	Garland Co.: Lake Hamilton at St. Hwy. 192 (T3S, R20W) (n = 1) HSU 2276.
4 Mar 1997	J. Thompson	Hot Spring Co. L'Eau Frais Creek 6 mi. E Donaldson at St. Hwy. 222 (Sec. 10, T6S, R17W) (n = 1) HSU 1605.
20 Feb 1999	D. Fendley	Izard Co.: Piney Creek at access road (Sec.5, T16N, R10W) (n = 1) HSU 2414.
3 Apr 1999	B. Crump	Pike Co.: Caddo River at Glenwood (Sec.10, T5S, R24W) (n = 1) HSU 2799.
25 Mar 1997	W. Daggett	Newton Co.: Buffalo River (Sec. 19, T16N, R20W) (n = 1) HSU 1924.
12 Apr 1997	D. Dunlap	Pike Co.: Caddo River at U.S. Hwy 70 (Sec.10, T5S, R24W) (n = 1) HSU 1704.

Table 1. Lamprey collections in Arkansas, 1988-2005. (cont.)

Date	Collector	Locality, Section, Township, Range, Sample Size (parentheses), HSU Museum Number
***Ichthyomyzon castaneus* Girard (cont.)**		
12 Apr 1994	R. Fisher	Stone Co.: White River at St. Hwy. 9 (Sec.12, T15N, R11W) (n = 1) HSU 836.
16 Jul 2002	J.C. Petersen	Searcy Co.: Buffalo River near Woolum (Sec. 4, T15N, R18W) (n = 1).
24 Apr 2003	J.C. Petersen	Newton Co.: Buffalo River near Hasty (Sec. 34, T16N, R20W) (n = 1).
19 Mar 2003	J.C. Petersen	Marion Co.: Buffalo River near mouth (Sec. 36, T18N, R14W) (n = 1).
5 Apr 2005	D. Thompson	Clark Co.: DeGray Lake near Brushy Landing (Sec. 8, T6S, R21W) (n = 1) HSU 3154.
***Ichthyomyzon gagei* Hubbs and Trautman**		
30 Apr 2003	H. Runyan	Clark Co.: Caddo River (Sec.7, T7S, R19W) (n = 1) HSU 2888.
4 May 1999	J. Patterson	Clark Co.: L'Eau Frais Creek at St. Hwy 7 (Sec.1, T8S, R19W) (n = 1) HSU 2361 (ammocoete).
11 Feb 1999	J. Hardage	Clark Co.: Bradshaw Creek at St. Hwy. 51 (Sec.1, T8S, R21W) (n = 1) HSU 2503.
4 May 1999	D. Fendley and J. Hardage	Clark Co. Tupelo Creek at St. Hwy. 7 (Sec. 35, T7S, R19W) (n = 1) HSU 2753.
22 Mar 1997	R. Tumlison	Hot Spring Co.: L'Eau Frais Creek, 7.5 mi. E. Donaldson (Sec. 1, T6S, R17W) (n = 4) HSU 1489 (1 adult, 3 ammocoetes).
27 Feb 1999	S. Ryders	Saline Co.: Caney Creek at U. S. Hwy. 70 near I-30 bridge (Sec. 20, T2S, R16W) (n = 1) HSU 2462.
8 Mar 2002	H. W. Robison	Stone Co.: East Livingston Creek (Sylamore Creek Dr.) (Sec. 23, T16N, R11W) (n = 2) SAU Fish Collection.
20 Mar 1995	G. Leeds	Franklin Co.: Mulberry River (Sec. 34, T12N, R27W) (n = 1) SAU Fish Collection.
20 Feb 1999	H. W. Robison	Johnson Co.: Little Piney Creek (Sec. 26, T11N, R22W) (n = 1) SAU Fish Collection.
31 Oct 1996	R. Tumlison	Clark Co.: Caddo River at St. Hwy 7 (Sec. 31, T6S, R19W) (n = 1) HSU 1386.
20 Apr 1994	M. Major and C. Petty	Clark Co.: L'Eau Frais Creek, 4 mi. from Manchester at Hwy. 7 (NE ¼, Sec. 1, T8S, R19W) (n = 1) HSU 3155.
Unidentified Ammocoetes		
31 Jul 2001	J.C. Petersen	Newton Co.: Buffalo River near Boxley (Sec. 22, T15N, R23W) (n = 1).
27 Jun 2002	J.C. Petersen	Newton Co.: Buffalo River near Pruitt (Sec. 7, T16N, R20W) (n = 1).
31 Jul 2001	J.C. Petersen	Newton Co.: Buffalo River near Hasty (Sec. 34, T16N, R20W) (n=1).
18 Jul 2001	J.C. Petersen	Searcy Co.: Buffalo River near Woolum (Sec. 4, T15N, R18W) (n=1).
18 Jul 2001	J.C. Petersen	Searcy Co.: Buffalo River at Shine Eye near Gilbert (Sec. 36, T16N, R17W) (n=1).
13 Sep 2002	J.C. Petersen	Marion Co.: Buffalo River at Highway 14 near Harriet (Sec. 34, T17N, R15W) (n=1).
2 Aug 2001	J.C. Petersen	Newton Co: Little Buffalo River near mouth near Pruitt (Sec. 20, T16N, R20W) (n=1).
14 Aug 2001	J.C. Petersen	Madison Co.: Kings River near Kingston (Sec. 29, T16N, R24W) (n=1).
20 Aug 2002	J.C. Petersen	Carroll Co.: Osage Creek near Berryville (Sec. 12, T19N, R25W) (n=1).
10 Sep 2002	J.C. Petersen	Carroll Co.: Long Creek near Denver (Sec. 16, T20N, R22W) (n=1).
17 Sep 2002	J.C. Petersen	Marion Co.: Hampton Creek near Eros (Sec. 21, T18N, R17W) (n=1).
8 May 1999	A. Westin	Clark Co.: L'Eau Frais Creek at St. Hwy. 7 (Sec. 1, T8S, R19W) (n = 1) HSU 2332.
3 Mar 2005	S. Townsend	Clark Co.: L'Eau Frais Creek at St. 28 (Sec. 22, T7S, R18W) (n = 1) HSU 3022.
10 Apr 2005	S. Townsend	Fulton Co.: Spring River (Sec. 21, T21N, R5W) (n = 1) HSU 3066.

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Chinese Flame Tree (*Koelreuteria bipinnata* Franch.) (Sapindaceae) New to the Arkansas Flora

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Non–native plant species are continuously being introduced into the United States by both accidental and intentional means. Many of these species never become established in the flora, or if establishment occurs, many do not seriously threaten native plant species (Williamson 1996). However, numerous non–native plant species have become invasive subsequent to establishment and naturalization. Invasive species can alter native habitats and ecosystems and often seem to reduce native biodiversity (D' Antonia and Vitousek 1992; Daehler and Strong 1994; Wilcove et al. 1998). Some of the worst invasive plants are ornamental trees and shrubs. A few examples of woody ornamentals that have established in the Arkansas flora and subsequently become invasive include nandina (*Nandina domestica* Thunb.), Chinese privet (*Ligustrum sinense* Lour.), glossy privet (*Ligustrum lucidum* Ait.), and Chinese tallow tree [*Sapium sebiferum* (L.) Roxb.]. About 23% of the Arkansas flora consists of non–native species (Arkansas Vascular Flora Committee 2006). *Koelreuteria bipinnata* Franch. (Chinese flame tree), another non–native ornamental species, is here reported as spontaneous in the Arkansas flora. This species is a small to medium–sized tree that is native to southwest China (Krüssmann 1977). It is occasionally cultivated in the southern US for ornamental purposes because of its showy flowers and fruits and its tolerance of a wide variety of soil types (Bailey and Bailey 1976, Krüssmann 1977, Griffiths 1994). While it is too early to determine whether or not *K. bipinnata* (Fig. 1) will become invasive or not in Arkansas, it has shown the ability to reproduce successfully, escape cultivation, and establish in the Arkansas flora.

Forty–two spontaneous plants of *K. bipinnata* were discovered growing in two places on the Ouachita Baptist University (OBU) campus in Clark County, Arkansas. Voucher specimens of *K. bipinnata* were deposited in the HSU herbarium (Serviss 7026, Serviss and Melancen 7028). Four of the 42 individuals documented were reproductive, being found with mature or nearly mature fruits. Reproductive individuals ranged in dbh from 10.1–12.0 cm. All 42 individuals were much smaller than the maximum height that can be attained for the species (about 20 m) and ranged in size from 7.2 m to seedlings only a few centimeters tall (Table 1). The 42 spontaneous

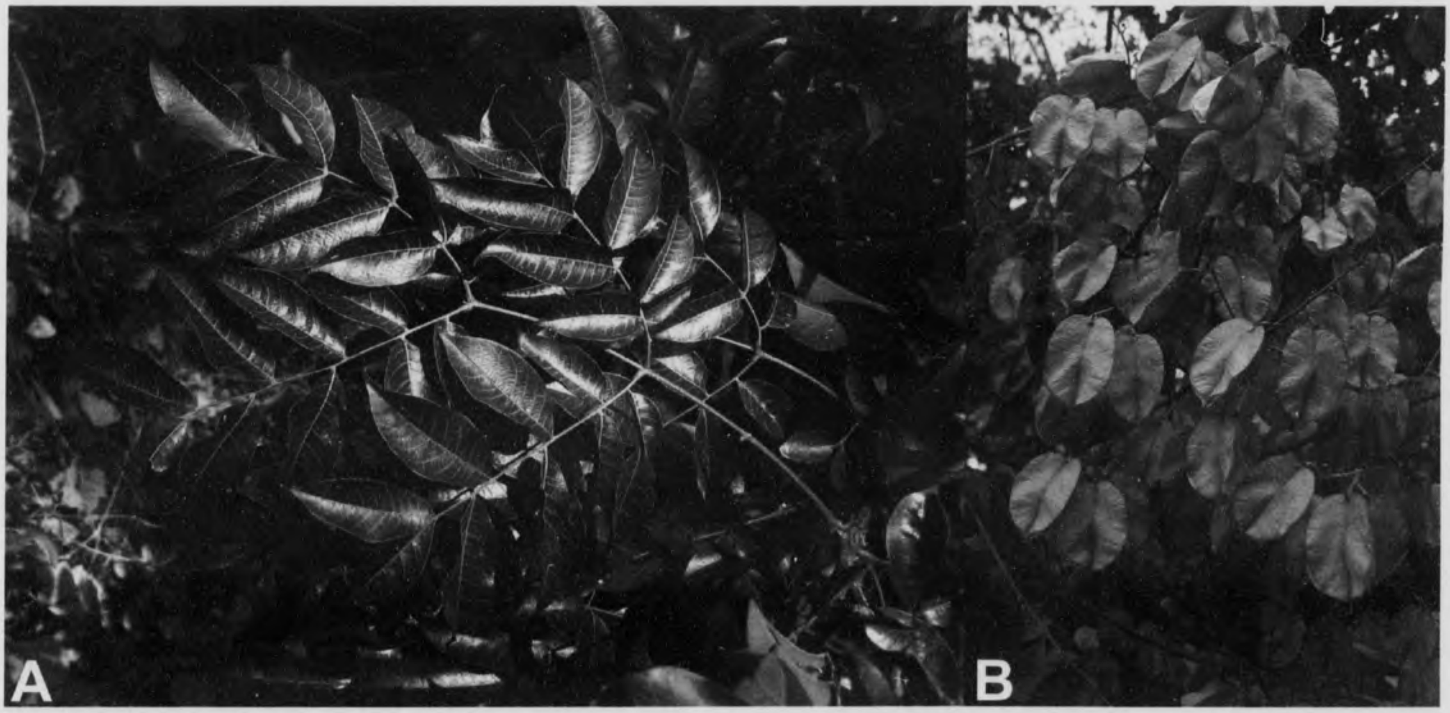

Fig. 1. Photos of *Koelreuteria bipinnata*. A. Twice pinnately compound leaf. B. Mature fruits.

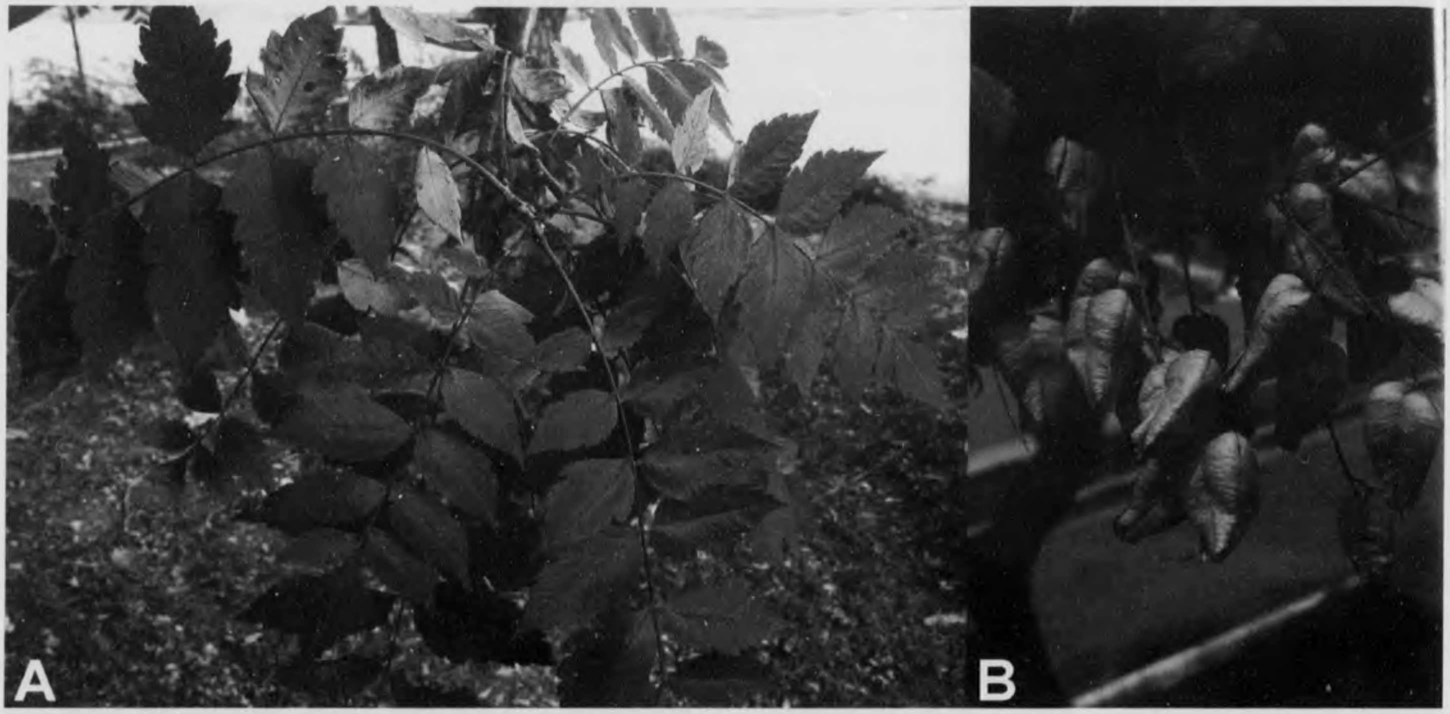

Fig. 2. Photos of *Koelreuteria paniculata* (for comparison with *K. bipinnata*). A. Once pinnately compound leaves. B. Mature fruits.

plants are distributed within two populations that occur at two widely separated areas on the OBU campus. We speculate that both populations were originally founded from seeds that were produced and then subsequently dispersed to both sites from a single, cultivated *K. bipinnata* tree on the campus. The fruits of *Koelreuteria* spp. are wind dispersed and thus the seeds can be transported great distances from the parent plant. The larger of the two populations (population one), consisting of 38 individuals, is located within a small portion of a natural area that extends through much of the campus. Much of this area consists of riparian habitat with varying degrees of disturbance. There are also several small areas in it that are without canopy cover or with only a minimal canopy present. In population one, individual plants range in development from reproductive age individuals to seedlings, and plants were found growing in areas with and without canopy cover. All plants, regardless of size, displayed only minimal branching and crown development, which is probably because of young age. The smaller population (population two), consisting of four individuals, is adjacent to the putative parent plant, which is located near the southwest corner of the campus and several hundred meters from population one. Three of the four *K. bipinnata* plants in population two were found growing in a yard, which was unkempt and overgrown with a mixture of trees, shrubs, and other vegetation. The remaining *K. bipinnata* plant occurred a short distance away from the previously mentioned yard area and was present in a highly disturbed area at the edge of a parking lot.

It is probable that at least the original spontaneous plants in population one were likely descended from the cultivated tree on the OBU campus, but because several of these individuals are reproductive, at least some of the smaller juveniles in population one may have been produced from the original, spontaneous, now reproductive age trees. It is important, however, to note that seeds may also continue to be introduced into population one from the cultivated *K. bipinnata* tree. Although *K. bipinnata* can reach a mature size of up to 20 m (Bailey and Bailey 1976), it is capable of sexual reproduction at a much smaller size and presumably younger age. Additionally, species of *Koelreuteria* can produce hundreds of seeds per plant, grow rapidly when young, are tolerant of a wide range of soil conditions, and tolerate a variety of light regimes (observed with *K. bipinnata*). Additionally, seeds collected from *K. bipinnata* in Arkansas show high rates of germination and seedling emergence. The combination of these traits seemingly increases the likelihood that *K. bipinnata* will become firmly established and even possibly invasive in Arkansas.

Koelreuteria bipinnata is not the only species of *Koelreuteria* to be documented in the Arkansas flora. *Koelreuteria paniculat*[illegible] Laxm. (golden–rain tree; Fig. 2) is also spontaneous in Arkans[illegible] We observed two, small, spontaneous populations (composed essentially of seedlings) beneath and in proximity to two, large, presumably cultivated trees of *K. paniculata* on the Hendrix College campus in Conway, Arkansas, in 2005 and in Hot Springs, Arkansas, in 2006 (one tree at each location), indicating that the non–native *K. paniculata* is spontaneous, does reproduce successfully, and could potentially establish in Arkansas. These two species of *Koelreuteria* are somewhat similar, but can easily be distinguished by using the following key.

Key to the spontaneous or naturalized species of *Koelreuteria* in Arkansas:

1. Mature leaves twice pinnately compound; margins of leaflets of mature leaves with small teeth or entire (leaflet margins of juvenile plants are coarsely toothed to lobed); fruits cream colored to greenish–yellow, but usually tinged or flushed with pink to pinkish–red coloration during development, and widest in the middle (elliptic) .... *K. bipinnata*

1. Mature leaves mostly to wholly once pinnately compound (a few leaves may be imperfectly bipinnately compound); mature leaflets with coarse, crenate teeth, sometimes also lobed or incised; fruits green to greenish–yellow during development, and widest at the base, slightly past point of attachment of the pedicel (conical)... *K. paniculata*

Acknowledgments.—We would like to sincerely thank Ouachita Baptist University for allowing us to conduct research and collect numerous plant specimens on their campus. We would also like to thank the Biology Department at Henderson State University (HSU) for supporting this research. Additionally, we thank Drs. Renn Tumlison (HSU) and James Peck (UALR), along with 2 anonymous reviewers, for reviewing this paper and providing many helpful comments and suggestions.

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Table 1. List of spontaneous *Koelreuteria bipinnata* individuals documented in Clark County, Arkansas, including reproductive status, height, and specific location in habitat.

Reproductive	Height in cm	Location in habitat
	Population 1 (38 individuals)	
yes	726.4	edge; in canopy
yes	639.4	edge; in canopy
yes	607.0	edge; in canopy
no	513.0	edge; in canopy
no	509.2	edge; in canopy
no	452.1	edge; in canopy
no	416.5	edge; in canopy
yes	403.8	edge; in canopy
no	401.3	edge; in canopy
no	391.6	edge; in canopy
no	231.1	edge
no	180.3	wooded; in understory
no	179.7	wooded; in understory
no	177.8	edge; in understory
no	176.5	edge; in understory
no	166.3	edge; in understory
no	149.2	open; no canopy
no	140.3	open; no canopy
no	125.7	wooded; in understory
no	124.4	edge; in understory
no	94.6	edge
no	87.6	open; no canopy
no	69.8	open; no canopy
no	55.8	edge
no	42.5	wooded; in understory
no	40.6	open; no canopy
no	35.5	wooded; in understory
no	34.2	wooded; in understory
no	33.0	wooded; in understroy
no	33.0	open; no canopy
no	30.4	open; no canopy
no	25.4	wooded; in understory
no	25.4	wooded; in understroy
no	24.1	open; no canopy
no	19.6	open; no canopy
no	15.8	wooded; in understory
no	15.8	wooded edge
no	13.3	wooded edge
	Population 2 (4 individuals)	
no	26.6	understory
no	21.5	open; no canopy
no	13.9	understory
no	12.7	understory

List of Associate Editors and Reviewers for the Journal of the Arkansas Academy of Science, Volume 60, 2006

The Arkansas Academy of Science gratefully acknowledges the following individuals who served as Associate Editors and outside reviewers of manuscripts for volume 60 of the *Journal* during 2006. 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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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 the Manuscript

1. Use Microsoft Word 6.0 or better for preparation of the document.
2. Save figures as tiff or jpeg files.
3. Double space the manuscript and all associated text including the Literature Cited on 8 ½ 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.
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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 type-written pages. A *JOURNAL* printed page is equal to approximately 3 ½ 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. A note should have the title at the top of 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.
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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.

Standard Journal Article

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(5085):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 111:232-7.

Form of Citation: (Zheng and Luh 1989)

Electronic Journal Articles and Electronic Books 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: www.usfw.gov/ozarkstreams. Accessed 2004 Nov 29.

Books, Pamphlets, and Brochures

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)

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Form of Citation: (Engelberger 1989)

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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 285-96.

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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 122-30.

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Stevens, WB. 2004. An ecotoxilogical analysis of stream water in Arkansas [dissertation]. State University (AR): Arkansas State University. 159 p.

<u>**Scientific and Technical Reports**</u> – 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)

<u>**Published Conference Proceedings**</u> – 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., postpartum) 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
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Mathematical Reviews
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