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Academy Editors

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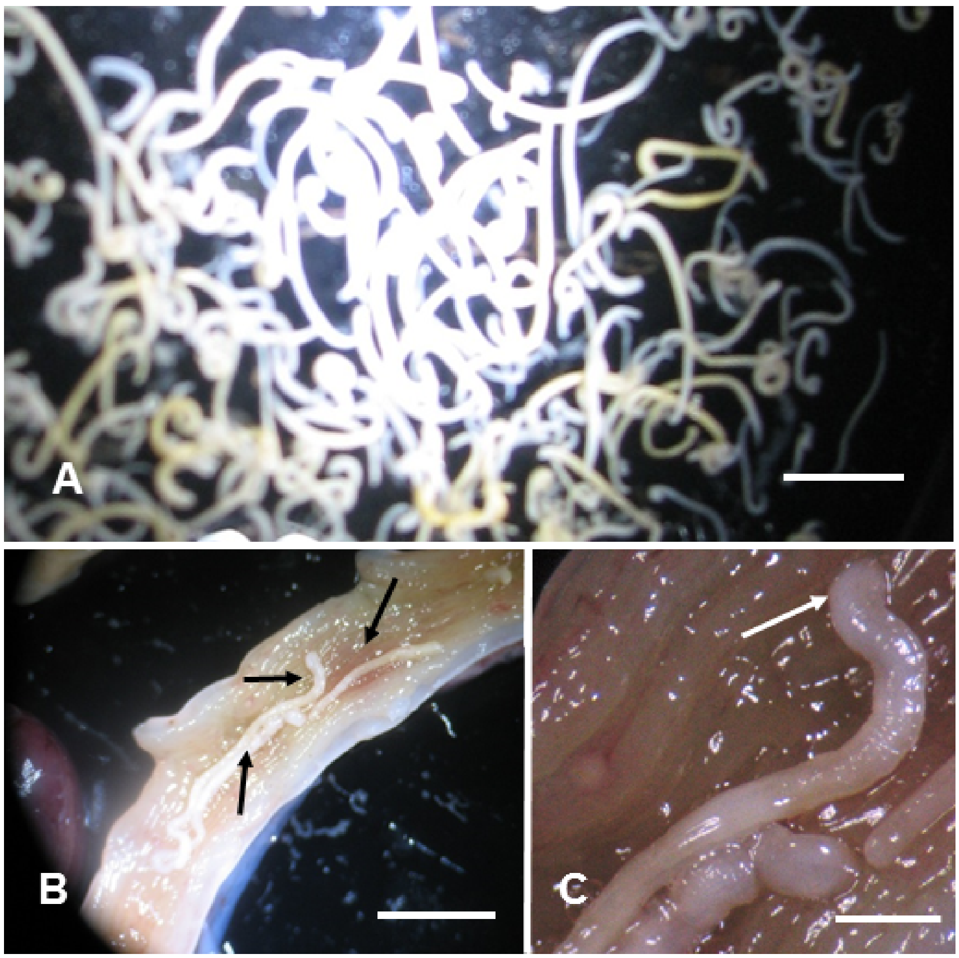

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ARKANSAS TECH UNIVERSITY
DEPARTMENT OF PHYSICAL SCIENCES
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The Arkansas Academy

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OF SCIENCE

Arkansas Academy of Science, Dept. of Physical Sciences, Arkansas Tech University

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L. F. Bailey	1965
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John J. Chapman	1968
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Richard Kluender	1997
James Daly	1998
Rose McConnell	1999
Mostafa Hemmati	2000
Mark Draganjac	2001
John Rickett	2002
Walter E. Godwin	2003
Wayne L.Gray	2004
Betty Crump	2005
Stanley Trauth	2006
David Saugey	2007
Collis Geren	2008
Joyce Hardin	2009
Scott Kirkconnell	2010
Jeff Robertson	2011
Anthony K. Grafton	2012
Marc Seigar	2013
Jeff Robertson	2014

INSTITUTIONAL MEMBERS

The Arkansas Academy of Science recognizes the support of the following institutions through their Institutional Membership in the Academy.

ARKANSAS STATE UNIVERSITY, Jonesboro
ARKANSAS TECH UNIVERSITY, Russellville
JOHN BROWN UNIVERSITY, Siloam Springs
SOUTHERN ARKANSAS UNIVERSITY, Magnolia
UNIVERSITY OF ARKANSAS AT FORT SMITH
UNIVERSITY OF ARKANSAS AT FAYETTEVILLE
UNIVERSITY OF ARKANSAS AT MONTICELLO
UNIVERSITY OF ARKANSAS AT PINE BLUFF
UNIVERSITY OF THE OZARKS, Clarksville
UNIVERSITY OF ARKANSAS FOR MEDICAL SCIENCES, Little Rock

COVER: The acanthocephalan parasite, *Neoechinorhynchus emydis* removed from the intestinal tract of a common map turtle, *Graptemys geographica*. See *Haemogregarina sp.* (Apicomplexa: Haemogregarinidae), *Telorchis attenuata* (Digenea: Telorchiidae) and *Neoechinorhynchus emydis* (Acanthocephala: Neoechinorhynchidae) from Map Turtles (*Graptemys spp.*), in Northcentral Arkansas by C.T. McAllister and colleagues, pp 154-157.

ARKANSAS ACADEMY OF SCIENCE 2014

APRIL 4-5, 2014
98th ANNUAL MEETING

Harding University
Searcy, Arkansas

JOURNAL
ARKANSAS ACADEMY OF SCIENCE

Annual Meeting April 4-5, 2014
Harding University

Secretary's Report
MINUTES OF THE 98th MEETING

ARKANSAS ACADEMY OF SCIENCE
SPRING 2014 BUSINESS MEETING MINUTES
April 5, 2014 – 12:00 pm
Harding University

1. The meeting was called to order by by President Jeff Robertson.

2. Local Arrangements Committee: Ed Wilson
145 people pre-registered for the meeting. There were 80 oral presentations, 55 poster presentations.

3. Secretary's Report: Jeff Robertson
Minutes from the 2013 Fall Executive Committee Meeting in November were reviewed and accepted.
The Academy has 107 members (49 of which are life members).

4. Treasurer's Report: Mostafa Hemmati
An accounting of the AAS "net worth" for 2013 was presented and discussed by the membership. The report was reviewed by an auditing team made of selected members of the Academy and accepted by the membership.

5. Historian's Report: Collis Geren
The 2014 spring meeting of the Arkansas Academy of Science at Harding University in Searcy, Arkansas is the 98th annual meeting of the Academy. This will mark the fourth time that Harding will have hosted the Academy having done so previously in 1955, 1962, and 1971 when Harding was named Harding College. Harding began as a senior college in 1924, when two junior colleges, Arkansas Christian College and Harper College, merged their facilities and assets, adopted the new name of Harding College, and located on the campus of Arkansas Christian in Morrilton, Ark. Harper had been founded in 1915 in Harper, Kan., and Arkansas Christian had been chartered in 1919.
Upon completion of a study begun in May 1978, the board of trustees approved the study's recommended change of Harding to university status, and on Aug. 27, 1979, the name of the institution officially became Harding University.
In 1934 Harding was moved to its present site in Searcy, Ark., on the campus of a former women's institution, Galloway College. The college was named in memory of James A. Harding, co-founder and first president of Nashville Bible School (now David Lipscomb University) in Nashville, Tenn. A preacher, teacher and Christian educator, James A. Harding inspired his co-workers and associates with an enthusiasm for Christian education that remains a significant tradition at Harding University. Harding University is associated with the Churches of Christ. Harding offers 10 undergraduate degrees in more than 100 academic majors, 14 pre-professional programs, and 15 graduate and professional degrees in its colleges of Allied Health, Arts and Humanities, Bible and Ministry, Business Administration, Education, Honors, Nursing, Pharmacy and Sciences. Harding offers 10 undergraduate degrees in more than 100 academic majors, 14 pre-professional programs, and 15 graduate and professional degrees in its colleges of Allied Health, Arts and Humanities, Bible and Ministry, Business Administration, Education, Honors, Nursing, Pharmacy and Sciences. Harding currently enrolls almost 7000 students. While Searcy is the home for the main campus of Harding University, satellite campuses are present in North Little Rock, Rogers, and Memphis as well as in a number of international locations.
The Academy is indebted to Professor Ed Wilson of Harding's Chemistry Department not only for his

organization of this meeting, but also for the many roles he has played in improving science education for all of Arkansas.

6. Journal (JAAS #67) Report:
Editor-In-Chief Mostafa Hemmati

During the spring 2013 semester, 40 manuscripts were submitted for consideration for publication in volume 67 of the *Journal of the Arkansas Academy of Science (JAAS).* Soon after receiving the manuscripts, all manuscripts were sent to three reviewers and Associate Editors. The reviewers sent all manuscripts and their comments back before the end of July 2013.

Reviewers' comments were sent to the authors between July 15, 2013, and July 30, 2013. That process was completed by July 30, 2013. The authors were asked to respond to the reviewers' comments and return their manuscript back by August 31, 2013. That allowed more than a month of time for the authors to respond to the reviewers' comments. In the same letter, the authors were also asked to mail a check for their page charges. August 31, 2013, was also the deadline for receipt of the payment of the page charges.

Seven manuscripts required major revisions, while one was rejected and the rest needed minor revisions. For one of the manuscripts, we did not receive the page charges in a reasonable length of time; therefore, that manuscript will be published in volume 68 of the Journal. Therefore, volume 67 of the *Journa*l will include 38 manuscripts.

Three Associate Editors, Dr. Collis Geren, Dr. Bill Doria and Dr. Frank Hardcastle, helped considerably with locating possible reviewers for the manuscripts or serving as reviewer for more than one manuscript. I am grateful for all three Associate Editors' assistance. All activities relating to the handling of the manuscripts were performed electronically, and on the whole this expedited the review process. Managing editor post was performed by Dr. Ivan H. Still and as usual he did an excellent job. The *Journal* was completed by December 30, 2013. Printing of the *Journal* was completed by February 20, 2014. I used Russellville Printing Company to print the Journal, and their estimate and final price was very reasonable.

Managing Editor Ivan Still

Forty manuscripts were submitted for publication in volume 67 (2013) of the JAAS.

By the beginning of May these manuscripts were checked for style, grammar, format, etc. to ensure compliance with the "Instructions to Authors." Abstracts were sent to potential reviewers mid to late May. Dr. Hemmati handled Physical Science papers and recruited Drs. Collis Geren, Frank Hardcastle, and Bill Doria to serve as Associate Editors, while Biological Science manuscripts were handled by Dr. Still. All manuscripts were sent out electronically for review by the beginning of June. These were returned to the Managing Editor at the end of June/middle of July.

Most authors were contacted by e-mail by the middle of July 2013 and informed if there paper was accepted with the need for minor or major revision while 7 required major revisions. One manuscript was rejected, with reviewers comments passed on the author in hopes that the manuscripts could be improved for resubmission next year. All authors were asked to return their revisions to the Managing Editor electronically by August 31, with page charges being submitted to Dr. Hemmati, Editor-in-Chief. One author failed to send in their page charges, so that manuscript will be published in volume 68.

The total number of manuscripts that will be published this year is 38 (up from 30 the previous year), of which 22 were Articles, 16 were in General Note format. The Journal was completed (238 pages total) and sent for printing in January 2014, with the Table of Contents loaded to the JAAS website in February.

I would like to thank all the reviewers and Assistant/Associate Editors for their help in the preparation of volume 67, and finally the corresponding authors of submitted manuscripts and the reviewers for their efforts in maintaining the quality of the journal.

7. Webmaster: Salomon Itza

On June 06, 2013, AAS purchased an account at the site iPage.com to host the AAS website. The license is for 24 months with backup and restoration. This is the expense detailed (paid by the AAS treasurer):

$1.99/month (billed $47.76 for 24 months), Site backup and restore, $12.95/year (billed for 2 years), TOTAL $73.66, Expiring 2015 June 06.

The main entry portal page is still http://www.arkansasacademyofscience.org.

The webmaster has been updating the page for meetings. Dr. Ed Wilson provided information related to the 98th annual meeting. One issue that came out was the different browsers used by the AAS members and friends, some of them unable to download PDF or MSWord files. The webmaster emailed the files to colleagues requesting the registration and abstract submission files for the meeting.

Also, there has been the suggestion that the AAS website should host online registration. It is possible by creating an alternate account with a password that changes every year. The issue that is developing the HTML code for such a task is not straightforward, however, once developed it should be easy to maintain and update every year so that the hosting institution can download information from the AAS website. Perhaps this is something that should be tried.

9. Committee Reports:

a. Nominations Committee: Mostafa Hemmati

Abdel Bachri inherited presidency of Academy, with Ann Willyard as President-elect. Ed Wilson elected to Vice President.

10. Business Old and New:

In 2015, the 99th annual AAS meeting will be held at Henderson State University. We appear to have an agreement with UA-Fayetteville host the 100th annual meeting in 2016 from Jim Rankin (VPR). The AAS is hoping to make this annual meeting a special event. A host for the future 101st meeting in 2017 is solicited to the community at large at this time.

Newsletter editor Ron Tackett resigned to take a job out of state. R. Panneer Selvam (rps@uark.edu) has volunteered to take on that role for the Academy.

11. Motions and Action Items:

AAS constitution and by-laws revisions were reviewed at Executive Committee Meeting, November 2012 and read for the first time to membership at the spring 2013 meeting. The second reading and vote for adoption occurred at this business meeting of the AAS membership and revisions were approved. The revised AAS constitution and by-laws are available on the Academy website.

Continuation of AAS Undergraduate Research Awards approved.

Academy budget 2014-2015 (outside costs associated with Journal publication) approved:

$2,500	AAS Undergraduate Research Grants (up to 5, up to $500)
$1,050	AAS Annual spring meeting student presentation awards
$100	AAS Secretary, journal mailings (if requested)
$900	AAAS representative travel (if requested)
$2,000	Affiliate student awards Junior Academy, AJSHS, Arkansas Science Fair (if requested)

$6,550	**TOTAL** (outside of costs associated with JAAS publication)

Abdel Bachri inherited presidency of Academy, with Ann Willyard as President-elect, Jeff Robertson as Past-President, and Ed Wilson as Vice President.

Meeting Adjourned

Jeff Robertson, AAS Secretary

Treasurer's Report
ARKANSAS ACADEMY OF SCIENCE
2014 FINANCIAL STATEMENT
December 17, 2014

Balance – November 10, 2014	**$103,764.83**
Balance – November 8, 2013	**$93,603.53**
Net Gain	**$10,161.30**

DISTRIBUTION OF FUNDS

Checking Account Bank of the Ozarks, Russellville, AR, 12/17/2014	**$37,045.72**
Certificate of Deposit Life Membership Endowment, Bank of the Ozarks, Russellville, AR, 12/17/2014 Maturity Date 06/11/15	**$18,554.86**
Dwight Moore Endowment + (Dwight Moore's final balance of $6,002.73+ Short term CD's final balance of $4,157.77+ $9,839.50 from the Bank of the Ozarks checking account = $20,000. 12/17/14 Maturity Date, 06/10/2015	**$20,893.79**

Phoebe and George Harp Endowment **$18,964.57**
($7601 Harp + $6515.15 CD + $3383.85 Checking)=
$17500 CD + Interest Paid. Maturity Date 04/15/2015

Short Term CD **$8,305.89**
Bank of the Ozarks, Russellville, AR, 12/17/2014
New Maturity date 01/27/2015

Combined interest on all accounts as of 12/17, 2014 was $$15.65+$50.97+$22.14+$57.40+$53.69= $199.85

TOTAL **$103,764.83**

INCOME:

1. Transfer from CD to Checking **$0**

2. GIFTS RECEIVED
   a. Ouachita National Forest - Sponsorship -0-
   b. Contribution, Collis Geren $200

$200

3. INTEREST (Interest Earned Year to Date, ~ December 17, 2014)

a. Checking Account, Bank of the Ozarks, …448		$15.65
b. CD1 (Bank of the Ozarks), …..	929	$50.97
c. CD2 (Bank of the Ozarks), …	594	$22.14
d. CD3 (Bank of the Ozarks), …..	583	$57.40
e. CD4 (Bank of the Ozarks)……	396	$53.69

All interest was added to the CDs **$199.85**

4. JOURNAL

a. Page Charges	$6,350
b. 1 Copy of Vol. 67	$50
c. Subscriptions, University of Arkansas	$1,200
d. Journal Subscription EBSCO	$0

$7,600.00

5. MISCELLANEOUS INCOME
   a. Coutts Information Services, Invoice #2014042100 $50

$50

6. MEMBERSHIP

a. Associate	$0
b. Individuals	$2,335
c. Institutional (UAMS)	$100
d. Life (Don Bragg, $500; Ben Rowley, $125, 3rd; Jacobs $125, 3rd; Suresh Kumar, $500; Liner $500)	$1,750
e. Sponsoring	$ 0
f. Sustaining	$35

$4,220.00

7. MEETING INCOME

a. Total Registration and Fees	$6,665
b. Additional Meeting Income	$915

$7580.00

TOTAL INCOME **$19,650.00**

EXPENSES

1. STUDENT AWARDS

1. Charlie Davis	$100
2. Youmna Moufarrei	$100
3. Josh Pennington	$100
4. Amlam Niragire	$100
5. Tyler Files	$100
6. Kaleb Vaughn	$100
7. Ryan Reyes	$100
8. Ryan Rogers	$100
9. Christopher Gillison	$100
10. Jordan Miller	$100
11. Nikisha West	$100
12. Jessica Hartman	$100

$1,200

2. AWARDS (Organizations)

a. Junior Science and Humanities Sym.	$400
b. Arkansas State Science Fair	$400
c. Arkansas Junior Academy of Science	$250
d. Arkansas Science Talent Search	$150

$1,200

3. UNDERGRADUATE RESEARCH AWARDS
   a. Dr. Campbell/Brownmiller, HSU $500

$500

4. JOURNAL

a. Volume 65 Printing Cost	$3,311.42
b. Journal Mailing Cost	$84.90
c. Journal Editorial Cost	$0.00

$3,396.32

5. MISCELLANOUS EXPENSES

1. Partial Reimbursement, Scott Kirkconnell's AAAS	$900
2. Reimbursement Jeff Robertson's Expenses for Website	$76
3. Additional Mailing Cost	$11.98
4. Web Services, Jeff Robertson	$25.96

$1,013.94

6. TRANSFER TO CD from Checking **$0.00**

7. MEETING EXPENSES

1. Meeting Food Expenses	$3,793.92
2. Meeting Program Printing Cost	$606.75
3. Meeting Expenses, Speaker Travel Cost	$463.68
4. Ed Wilson's Out of Pocket	$258.83

$5,123.18

TOTAL EXPENSES **$12,433.44**

ARKANSAS ACADEMY OF SCIENCE
COST OF JOURNAL

VOLUME	COPIES	PAGES	PRINTER CHARGE	TOT. VOL. COST	COST/ COPY	COST/ PAGE
35 (1981)	450	96	$3,694.68	$4,620.99	$10.27	$48.14
36 (1982)	450	110	$5,233.28	$5,291.69	$11.76	$48.11
37 (1983)	450	103	$5,326.91	$5,944.44	$13.21	$57.71
38 (1984)	450	97	$5,562.97	$6,167.72	$13.71	$63.58
39 (1985)	450	150	$7,856.20	$8,463.51	$18.81	$56.42
40 (1986)	450	98	$6,175.20	$6,675.20	$14.23	$68.11
41 (1987)	450	116	$7,122.79	$7,811.25	$17.36	$67.34
42 (1988)	450*	116	$7,210.79	$7,710.15	$17.13	$66.47
43 (1989)	450*	119	$8,057.24	$8,557.24	$19.02	$71.91
44 (1990)	450*	136	$9,298.64	$9,798.64	$21.77	$72.05
45 (1991)	450*	136	$9,397.07	$9,929.32	$22.06	$73.01
46 (1992)	450*	116	$9,478.56	$10,000.56	$22.22	$86.21
47 (1993)	400	160	$12,161.26	$12,861.26	$32.15	$80.38
48 (1994)	450	270	$17,562.46	$18,262.46	$40.58	$67.63
49 (1995)	390	199	$14,725.40	$15,425.40	$39.55	$77.51
50 (1996)	345	158	$11,950.00	$12,640.75	$36.64	$80.00
51 (1997)	350	214	$14,308.01	$15,008.01	$42.88	$70.13
52 (1998)	350	144	$12,490.59	$13,190.59	$37.69	$91.60
53 (1999)	350	160	$13,686.39	$14,386.39	$41.10	$89.91
54 (2000)	350	160	$14,149.07	$14,849.07	$42.43	$92.81
55 (2001)	360	195	$16,677.22	$17,498.22	$48.61	$89.73
56 (2002)	350	257	$18,201.93	$19,001.93	$54.29	$73.94
57 (2003)	230	229	$14,415.12	$15,715.12	$68.33	$68.62
58 (2004)	210	144	$7,875.76	$9,175.76	$43.99	$63.72
59 (2005)	215	226	$16,239.04	$17,835.84	$82.96	$78.92
60 (2006)	220	204	$11,348.06	$12,934.30	$58.79	$63.40
61 (2007)	195	150	$8,196.84	$9,914.69	$50.84	$66.10
62 (2008)	220	166	$2,865.00	$2,967.49	$13.49	$17.88
63 (2009)	213	206	$3,144.08	$3,144.08	$14.76	$15.26
64 (2010)	232	158	$2,713.54	$2,764.30	$11.91	$17.50
65 (2011)	200	194	$2915.12	$2,963.03	$14.82	$15.27
66 (2012)	200	216	$3,087.91	$3,180.29	$15.90	$14.72
67 (2013)	200	238	$3,311.42	$3,396.32	$16.98	$14.27

The Total Volume Cost equals the printer's charge plus the other miscellaneous charges (e.g. Mailing Costs).

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

APPENDIX A

2014 AAS PRESENTATION AWARD WINNERS (underlined)

ORAL PRESENTATION AWARDS

Life Science

"Pleistocene Seed Dispersal of Anachronistic Fruits: Using Elephants to Test Ancient Plant-Animal Interactions" by Charlie N. Davis, Madison J. Boone, and Laura Klasek. Hendrix College.

"Evaluating the Role of Meiotic Genes in DNA Repair using RNAi" by Youmna E. Moufarrej, Sydney L. Haldeman, Emily R. Cariker, and Andrew M. Schurko. Hendrix College.

Chemistry & Biochemistry

"Acetaminophen Increases Styrene Bioactivation to a Toxic Metabolite by CYP2E1" by Jessica H. Hartman, and Grover P. Miller. University of Arkansas for Medical Sciences.

Physics & Engineering

"The Crystal Structure Effect of the Synthesis of Cobalt Oxide Nanoparticles using Multiple Iterations of a Recyclable Precipitation Reaction" by J.S. Pennington, and R. J. Tackett. Arkansas Tech University.

"Software Development for a Diode Laser Spectrometer" by Amlam Niragire, Chih Hao Wu, and Edmond Wilson. Harding University.

POSTERS AWARDS

Life Science

"Sarcophageous Insect Associations and Succession on Pig Carrion During the Summer in Central Arkansas" by Tyler Files, Brianne Baley, and Jess Kelly. Ouachita Baptist University.

"Selection of Fatty Acid Desaturase 7 (fad7-1) Single Mutant Plants in *Arabidopsis thaliana* using SNP-PCR Primer" by Kaleb L. Vaughn[1], Carlos A. Avila[2], and Fiona Goggin[2]. [1]Harding University, [2]University of Arkansas.

Chemistry & Biochemistry

"Molecular Modeling Studies of Phylogenetically Significant Carotenoids of Oxygenic Phototrophs" by Ryan Reyes, and M. Jeffery Taylor. University of Arkansas-Monticello.

"Neutralization and pH Effect of Milk on Aspirin Solutions" by Jordan Miller, Nikisha West, Fontaine Taylor, and Insu 'Frank' Hahn. Philander Smith College.

Physics & Engineering

"X-Ray Fluorescence and Moseley's Laws of Nuclear Radiation Spectra" by T. Ryan Rogers, Hunter P. Ward, Nicholas L. Frederickson, Anthony D. Mitchel, Azida Walker, and Rahul Mehta. University of Central Arkansas.

"Stable Fuzzy Logic Control For Nonlinear Simple Chaotic Maps" by Christopher Gillison, and Juan D. Serna. University of Arkansas at Monticello.

This year, Harding University, and the Academy broke with tradition and did not distinguish between 1st and 2nd Places. We send our congratulations equally to our student presenters.

APPENDIX B
RESOLUTIONS

Arkansas Academy of Science
98th Annual Meeting, 2014 Resolutions

Be it resolved that we, the membership of the Arkansas Academy of Science (AAS) offer our sincere appreciation to Harding University for hosting the 98th annual meeting of the Academy. We thank the local arrangements committee: Edmond Wilson (chair), and committee members Ben Bruner, David Cole, Steve Cooper, Chelsea Essary, Jo Goy, Joseph Goy, Madison Greene, Julie Hixson-Wallace, Burt Hollandsworth, Landry Kamden, Trixie Lee, Victoria McIntosh, Brad Miller, Mike Plummer, Don Sanders, Ryan Stork, Travis Thompson, Lisa Valentine, Cindy White, and Charles Wu.

We sincerely thank the staff of the American Heritage Conference Center for their facilities and service during the meeting.

We especially thank our keynote speaker, Amber Straughn, for her inspiring talk entitled "Beyond Hubble: a new era in astronomy with NASA's James Webb space telescope."

The Academy recognizes the important role of our session chairs: Grover Miller and Joshua Sakon (Cell and Molecular Biology); David Cole and Kevin Stewart (Chemistry and Biochemistry); Joseph Goy and Ryan Stork (Invertebrate Biology); Mostafa Hemmati, Matt Strasser, and Charles Wu (Physics and Engineering), Ann Willyard (Plant Science); Steve Coopers and Cynthia Jacobs (Vertebrate Biology).

Even greater appreciation and sincere gratitude goes out to our judges for the student presentations, including David Cole, Steve Cooper, Lance Gibson, Joseph Goy, Trixie Lee, Nathan Mills, Steve Moore, Mike Plummer, Don Sanders, Kevin Stewart, Cindy White, Taylor Williams, and Charles Wu (Harding); Frank Hardcastle, Mostafa Hemmati, Jim Musser, and Scott Kirkconnell (ATU); Solomon Itza and Befrika Murdianti (Univ Ozarks); Carl Frederickson (UCA); and Jared Gavin (UA Monticello).

We congratulate our faculty and student researchers who presented papers and posters, contributing directly to the future success of the Academy and to the advancement of science in Arkansas.

The Academy recognizes its leadership and offers its thanks to this year's set of executive officers including Jeff Robertson (President), Abdel Bachri (President-Elect), Ann Willyard (Vice President), Mark Siegar (Past-President), Jeff Robertson (Secretary), Mostafa Hemmati (Treasurer), Ivan Still (Journal Managing Editor), Ronald Tackett (Newsletter Editor), Collis Geren (Historian), and Salomon Itza (Webmaster).

Respectfully submitted on this 5th day of April, 2014 by the Resolutions Committee: Ann Willyard (AAS Vice President), Jeff Robertson (AAS Secretary), and Edmond Wilson (Local Arrangements Committee).

2014 MEMBERSHIP

LIFE MEMBERS

FIRST MI.	LAST NAME	INSTITUTION
Edmond J.	Bacon	University of Arkansas-Monticello
Vernon	Bates	Ouachita Mountains
Floyd	Beckford	Lyon College
Don	Bragg	USDA Forest Service
Wilfred J.	Braithwaite	University of Arkansas-Little Rock
Calvin	Cotton	Geographics Silk Screening Co.
Betty	Crump	Ouchita National Forest
James	Daly	UAMS (retired)
Leo	Davis	Southern Arkansas University
Mark	Draganjac	Arkansas State University
Jim	Edson	University of Arkansas-Monticello
Kim	Fifer	UAMS
James H.	Fribourgh	University of Arkansas-Little Rock
Collis	Geren	University of Arkansas
John	Giese	Ark. Dept. of Env. Qual. (ret)
Walter	Godwin	University of Arkansas-Monticello
Anthony	Grafton	Lyon College
Joe M.	Guenter	University of Arkansas-Monticello
Joyce	Hardin	Hendrix College
George	Harp	Arkansas State University
Phoebe	Harp	Arkansas State University
Gary	Heidt	University of Arkansas-Little Rock
Mostafa	Hemmati	Arkansas Tech University
Philip	Hyatt	Retired
Shahidul	Islam	University of Arkansas-Pine Bluff
Cynthia	Jacobs	Arkansas Tech University
Douglas	James	University of Arkansas
Ronald	Javitch	Natural History Rare Book Found.
Art	Johnson	Hendrix College
Cindy	Kane	UAMS
Scott	Kirkconnell	Arkansas Tech University
Roger	Koeppe	University of Arkansas
Suresh	Kumar	University of Arkansas-Fayetteville
Roland	McDaniel	FTN Associates
Grover P.	Miller	UAMS
Herbert	Monoson	ASTA
Mansour	Mortazavi	University of Arkansas-Pine Bluff
James	Peck	University of Arkansas-Little Rock
Michael	Rapp	University of Central Arkansas
Dennis	Richardson	Quinnipiac College
Jeff	Robertson	Arkansas Tech University
Henry	Robison	Southern Arkansas University
Benjamin	Rowley	University of Central Arkansas
David	Saugey	U.S. Forest Service
Ivan	Still	Arkansas Tech University
Suresh	Thallapuranam	University of Arkansas-Fayetteville
Stanley	Trauth	Arkansas State University
Gary	Tucker	FTN Associates
Renn	Tumlison	Henderson State University
Scott	White	Southern Arkansas University
James	Wickliff	University of Arkansas
Robert	Wiley	University of Arkansas-Monticello
Steve	Zimmer	Arkansas Tech University

REGULAR MEMBERS

FIRST MI.	LAST NAME	INSTITUTION
Karen	Abbott	UAMS
Glen	Akridge	Northwest Arkansas Community College
Abdel	Bachri	Southern Arkansas University
Pablo	Bacon	Southern Arkansas University
Brent	Baker	Arkansas Natural Heritage Commission
Coskun	Bayrak	University of Arkansas-Little Rock
Daniel	Berleant	University of Arkansas-Little Rock
Frank	Blume	John Brown University
Anissa	Buckner	University of Arkansas-Pine Bluff
Rosemary	Burk	Arkansas Tech University
Stephen	Chordas III	Ohio State University
Don	Clover	Ft Smith Utilities/Env. Quality
Matthew	Connior	South Arkansas Community College
Magda	El-Shenawee	University of Arkansas-Fayetteville
James	Ettman	Morrilton
Anthony	Fernando	University of Arkansas-Pine Bluff
Karen	Fawley	University of Arkansas-Monticello
Marvin	Fawley	University of Arkansas-Monticello
Robert	Ficklin	University of Arkansas-Monticello
Ingrid	Fritsch	University of Arkansas-Fayetteville
Huaxiang	Fu	University of Arkansas-Fayetteville
Mariusz	Gajewski	Arkansas Tech University
Steve	Gann	Arkansas Tech University
Michael	Garner	Arkansas Tech University
Jared	Gavin	University of Arkansas at Monticello
Gija	Geme	Southern Arkansas University
David	Gilmore	Arkansas State University
Frank	Hahn	Philander Smith College
Franklin	Hardcastle	Arkansas Tech University
Laurence	Hardy	Museum of Life Science
Stewart	Hart	Arkansas Tech University
Alf	Haukenes	University of Arkansas-Pine Bluff
John	Hunt	University of Arkansas at Monticello
Anahita	Izadyar	Arkansas State University
Salomon	Itza	University of the Ozarks
David	Jamieson	Crowder College
Austin	Jones	NorthWest Arkansas Community College
Jess	Kelly	Ouachita Baptist University
Daniel	Kennefick	University of Arkansas-Fayetteville
Shubhalaxmi	Kher	Arkansas State University
Robert	Lambert	Sherwood
Janet	Lanza	University of Arkansas-Little Rock
Brenda	Lauffart	Arkansas Tech University
Ganna	Lyubartseva	Southern Arkansas University
David	Martinez	U.S. Fish and Wildlife Service
Chris	McAllister	Eastern Oklahoma State College-Idabel
Rahul	Mehta	University Central Arkansas
Gerhard	Mensch	American Innovations Academy
Matthew	Moran	Hendrix College
Lloyd	Moyo	Henderson State University
Befrika	Murdianti	University of the Ozarks
Jim	Musser	Arkansas Tech University
Lawrence	Mwasis	University of Arkansas-Pine Bluff
Henry	North	Harding University
Derrick	Oosterhuis	University of Arkansas-Fayetteville
Jeffrey	Padberg	University of Central Arkansas
David	Paul	University of Arkansas-Fayetteville
Forest	Payne	University of Arkansas-Little Rock

REGULAR MEMBERS

FIRST MI.	LAST NAME	INSTITUTION
James	Rippy	Arkansas Otolaryngology Center
Virginie	Rolland	Arkansas State University
Keith	Roper	University of Arkansas-Fayetteville
Joshua	Sakon	University of Arkansas-Fayetteville
Blake	Sasse	Arkansas Game and Fish
Marc	Seigar	University of Arkansas-Little Rock
Panneer	Selvam	University of Arkansas-Fayetteville
Derek	Selvidge	University of Arkansas-Fayetteville
Kimberly	Smith	University of Arkansas
Ryan	Stork	Harding University
Abraham	Tucker	Southern Arkansas University
Timothy	Wakefield	John Brown University
Azida	Walker	University of Central Arkansas
J.D.	Willson	University of Arkansas-Fayetteville
Anne	Willyard	Hendrix College
Ed	Wilson	Harding University
Cathy	Wissehr	University of Arkansas-Fayetteville
Tsunezui	Yamashita	Arkansas Tech University
Yijun	Yu	Philander Smith College

SPONSORING/SUSTAINING MEMBERS

FIRST MI.	LAST NAME	INSTITUTION
David	Cole	Harding University
Philip	Crandall	University of Arkansas-Fayetteville
Shelton	Fitzpatrick	University of Arkansas-Pine Bluff
Tim	Knight	Ouachita Baptist University
Christopher	Marvin	University of Arkansas-Monticello
Cynthia	Sagers	University of Arkansas-Fayetteville
Richard	Standage	USDA Forest Service Ouachita NF.

STUDENT MEMBERS

FIRST MI.	LAST NAME	INSTITUTION
Jessica	Ashcraft	Ouachita Baptist University
Sarah	Bishop	Ouachita Baptist University
Ryan	Evans	Arkansas Tech University
Tyler	Files	Ouachita Baptist University
Preston	Galla	Arkansas Tech University
Sanaa	Jawed	University of Arkansas at Little Rock
Soolaf A.	Kathiar	University of Arkansas at Little Rock
Larkin	McDaniel	Arkansas Tech University
Jordan	Miller	Philander Smith College
Eugene	Nyamugenda	Hendrix College
Dakota	Pouncey	Hendrix College
Jeremiah	Salinger	Arkansas State University
Nikisha	West	Philander Smith College
Whitney	Willis	
Wieyu	Wu	
Qiaozi	Zhao	Harding University

MAJOR INSTITUTIONAL SPONSORS

The Arkansas Academy of Science is an essential component in the science, technology, engineering and math pipeline for Arkansas. As a coalition of Arkansas scientists, it provides a local vehicle for presentation and publication of early scientific accomplishments in Arkansas. By promoting the work of Arkansas students, the Academy increases collaboration among the scientific community and provides a comprehensive network for scientific academics. These endeavors promote a higher standard of education within Arkansas and will encourage and promote a higher quality of life through educational opportunities.

As an integral part of the development and promotion of the Academy's mission, we wish to recognize the commitment and continued support of our Institutional Sponsors, The Arkansas Natural Heritage Commission and the Ouachita National Forest.

ARKANSAS NATURAL HERITAGE COMMISSION

Since 1973, the Arkansas Natural Heritage Commission (ANHC) has been working to conserve Arkansas's natural landscape. ANHC conducts research to determine which elements (species and natural communities) are most in need of protection. Field inventory documents the locations of elements of conservation concern. Information is also gathered from other sources, such herbarium and museum collection records, and scientific publications such as the *Journal of the Arkansas Academy of Science*. ANHC's current strategic planning goals include working to expand the ecological literacy of Arkansans. The Arkansas Academy of Science is a critical partner in helping to address this goal and, in the long term, protect the natural heritage of our state. For more information about the ANHC research, inventory and protection efforts, including the System of Natural Areas around the state, visit the agency website at www.naturalheritage.com. Here is a link to the current enewsletter featuring our support info as well. http://www.naturalheritage.com/enews/archive.aspx?mid=13361.

OUACHITA NATIONAL FOREST

Stretching from near the center of Arkansas to southeast Oklahoma, the pristine 1.8 million acre Ouachita National Forest is the South's oldest national forest, established on December 18, 1907 by President Theodore Roosevelt. Rich in history, the rugged Ouachita Mountains were first explored in 1541, by Hernando DeSoto's party of Spaniards. French explorers followed, flavoring the region with names like Fourche la Fave River. "Ouachita" is the French spelling of the Native American word "Washita" which means "good hunting grounds." The Forest's ecosystem management policy guarantees its management regime as an ecological approach, based upon the most current knowledge and best science, for providing multiple benefits from the Forest and encouraging careful use of the forest for the future. The research local to Arkansas and the Forest published by the Journal of the Arkansas Academy of Science is critical to informing and supporting appropriate management decisions, environmental assessments and biological evaluations. The Ouachita National Forest extends support of the Academy's efforts through this sponsorship.

For more information about the Forest, visit our webpage at: http://www.fs.fed.us/r8/ouachita.

KEYNOTE ADDRESS

Beyond Hubble: A New Era in Astronomy with NASA's James Webb Space Telescope

For over 20 years, the Hubble Space Telescope has been revealing the unknown cosmos; this single scientific instrument has completely revolutionized our understanding of the Universe. In 2009, the complete refurbishment of Hubble gave new life to the telescope and has produced groundbreaking science results, revealing some of the most distant galaxies ever discovered. Despite the remarkable advances in astronomy that Hubble has provided, the new questions that have arisen demand a new space telescope with new technologies and capabilities. I will present the exciting new technology development and science goals of Hubble's 100x-more-powerful successor, NASA's James Webb Space Telescope, which is currently being built and tested and will be launched later this decade.

Dr. Amber Straughn is an Astrophysicist at NASA's Goddard Space Flight Center in Greenbelt, MD and serves as the Deputy Project Scientist for James Webb Space Telescope Communications & Outreach.

Amber grew up in the small farming town of Bee Branch, Arkansas where her fascination with astronomy began under beautifully dark, rural skies. She obtained her B.S. in Physics at the University of Arkansas in Fayetteville in 2002, and completed her M.S. and Ph.D. in Physics at Arizona State University in 2008. She has been involved in NASA Education and Research programs since her undergrad years, beginning with flying an experiment on NASA’s microgravity KC-135 plane (the “vomit comet”) in 2001. During graduate school at Arizona State, Amber received the NASA Space Grant Fellowship for summer studies, and in 2005 was awarded the 3-year NASA Harriett Jenkins Pre-doctoral Fellowship.

Amber’s research focuses on interacting and star-forming galaxies in the context of galaxy assembly, and she has most recently been working on infrared spectroscopic data from the new Wide Field Camera 3 (WFC3) on Hubble Space Telescope. Her broad research interests include galaxy formation and evolution, galaxy mergers and interactions, physical processes induced by galaxy interactions including star formation and black hole growth, emission-line galaxies, and dark energy and its effect on the galaxy merger rate.

In addition to research, Amber’s role with the James Webb Space Telescope project involves working with Communications and Outreach activities. She has participated in extensive public speaking events locally, nationally, and internationally. Amber has also done several live television interviews, media features for NASA (see http://www.nasa.gov/topics/technology/features/webb-faqs.html), and appeared in the Late Night with Jimmy Fallon's "Hubble Gotchu" segment that aired in August 2010, and has interviewed for documentaries. She very much enjoys interacting with the public.

Amber lives in Glenn Dale, MD, with her husband Matt and her two Great Danes and one cat. Outside of her NASA work, she is a yoga teacher and is currently training for her private pilot's license.

SECTION PROGRAMS
ORAL PRESENTATIONS
(Presenter is underlined)

ORAL SESSIONS 1: FRIDAY 1:00-2:30

PHYSICS AND ENGINEERING 207
CHAIR: Mostafa Hemmati

1:00
DETECTION & RANGING SYSTEMS FOR PROXIMITY FLIGHT OF COOPERATING 6U CUBESATS
Mustafa Bayraktar[1], Yupo Chan[1], Po-Hao Adam Huang[2], Edmond W. Wilson, Jr.[3]
[1]University of Arkansas for Medical Sciences, [2]Univ. of Arkansas, Fayetteville, [3]Harding University

1:15
DESIGN OF CCD ARRAY DETECTOR FOR A CZERNY-TURNER SPECTROGRAPH
Brennan Thomason, Tamara Reed, and Edmond Wilson.
Harding University

1:30
SOFTWARE DEVELOPMENT FOR A DIODE LASER SPECTROMETER
Amlam Niragire, Chih Hao Wu, and Edmond Wilson. Harding University

1:45
OPTICAL ABSORPTION PROPERTIES OF GLANCING ANGLE DEPOSITED NANOSTRUCTURE ARRAYS IN DIFFERENT GEOMETRIES
Hilal Cansizoglu[1], Mehmet Cansizoglu[1], Miria M. Finckenor[2], and Tansel Karabacek[1].
[1]University of Arkansas at Little Rock, [2]NASA Marshall Space Flight Center

2:00
TORNADO-TERRAIN INTERACTION EFFECTS ON TORNADO DAMAGE USING GOOGLE EARTH
Nawfal Ahmed, R. Panneer Selvam, University of Arkansas

2:15
WHAT'S REALLY INVOLVED IN BUILDING A 3D PRINTER?
Justin Nesselrotte, and Edmond Wilson. Harding University

CHEMISTRY & BIOCHEMISTRY 208
CHAIR: David Cole

1:00
BOND LENGTH / BOND VALENCE RELATIONSHIPS FOR IRON-IRON, IRON-SULFUR, AND SULFUR-SULFUR BONDS
Welyu (Daniel) Lu, and Franklin D. Hardcastle. Arkansas Tech University

1:15
SYNTHESIS AND CHARACTERIZATION OF TRANSITION METAL COMPLEXES WITH HEXADENTATE HEMI-CAGE LIGAND
Megan Fuller, and Anwar Bhuiyan. Arkansas Tech University

1:45
ATOMIC ORBITAL EXPONENTS FROM VALENCE-LENGTH RELATIONSHIPS
Franklin D. Hardcastle. Arkansas Tech University

2:00
HYPOCHLOROUS ACID AS A METHOD OF SWIMMING POOL SANITATION
Kelton Schleyer, and Dennis Province. Harding University

2:15
CATALYSTS TESTING WITH CARBON PASTE ELECTRODES
Steve Gann. Arkansas Tech University

CELL & MOLECULAR BIOLOGY 209
CHAIR: Joshua Sakon

1:00
EVALUATING THE ROLE OF MEIOTIC GENES IN DNA REPAIR USING RNAi
Youmna E. Moufarrej, Sydney L. Haldeman, Emily R. Cariker, and Andrew M. Schurko. Hendrix College

1:15
COLLAGEN TARGETING MECHANISM OF BACTERIAL COLLAGENASE
Joshua Sakon. University of Arkansas

1:30
ACETAMINOPHEN INCREASES STYRENE BIOACTIVATION TO A TOXIC METABOLITE BY CYP2E1
Jessica H. Hartman, and Grover P. Miller.
University of Arkansas for Medical Sciences

1:45
WARFARIN METABOLITE PROFILES REVEAL THE IMPORTANCE OF FACTORS ON PATIENT DOSE-RESPONSES TO ANTICOAGULANT THERAPY
Dakota Pouncey. Hendrix College

2:00
DISCOVERY AND GENOMIC ANALYSIS OF TWO NOVEL MYCOBACTERIOPHAGES
Alyssa Stubblefield, Nathan Reyna, and Ruth Plymale.
Ouachita Baptist University

2:15
USING MACHINE LEARNING ALGORITHMS TO PREDICT DRUG METABOLISM BY CYP2C ENZYMES
Eugene Nyamugenda[1], Jessica H. Hartman[2], Grover P. Miller[2].
[1]Hendrix College, [2]University of Arkansas for Medical Sciences

VERTEBRATE BIOLOGY 210
CHAIR: Steve Cooper

1:00
URBAN STREAM SYNDROME IN A SMALL TOWN: A COMPARATIVE STUDY OF SAGER AND FLINT CREEKS
T.S. Wakefield. John Brown University

1:15
ONSET OF SCALE FORMATION IN ALLIGATOR GAR
Anthony V. Fernando, and Steve E. Lochmann. Department of Aquaculture and Fisheries, University of Arkansas at Pine Bluff

1:30
DETERMINING INDIVIDUALISTIC VOCAL CHARACTERISTICS OF *PANTHERA TIGRIS* AND IMPLICATIONS FOR ACOUSTIC MONITORING OF IN-SITU POPULATIONS
Courtney Elizabeth Dunn, and Mary Victoria McDonald.
University of Central Arkansas

1:45

SIZE AND AGE RECORDS FOR AN ARKANSAS SPECIMEN OF THE AMERICAN BULLFROG, *LITHOBATES CATESBEIANUS* (ANURA: RANIDAE), FROM NORTHEASTERN ARKANSAS

Stanley E. Trauth, and Timothy A. Welch. Arkansas State University

2:00

DISTRIBUTION OF THE NORTHERN LONG-EARED BAT (*MYOTIS SEPTENTRIONALIS*) IN ARKANSAS

D.B. Sasse[1], M.J. Harvey[2], J.J. Jackson[3], P.R. Moore[4], R.W. Perry[5], T.S. Risch[4], D.A. Saugey[6], and J.D. Wilhide[3].
[1]Arkansas Game and Fish Commission, [2]Tennessee Technological University, [3]Jackson Environmental, [4]Arkansas State University, [5]U.S. Forest Service, [6]Nightwing Consulting

2:15

VARIATION IN THERMALLY INDUCED CROSS-PROTECTION IN CHANNEL CATFISH EXPOSED TO AN AMMONIA CHALLENGE

Sindhu Kaimal, and Alf H. Haukenes. University of Arkansas-Pine Bluff

INVERTEBRATE BIOLOGY Liberty Room
CHAIR: Ryan Stork

1:00

NEW HOST AND DISTRIBUTION RECORDS OF THE LEECH *PLACOBDELLA MULTILINEATA* MOORE, 1953 (HIRUDINIDA: GLOSSIPHONIIDAE)

William E. Moser[1], Dennis J. Richardson[2], Chris T. McAllister[3], J. T. Briggler[4], Charlotte I. Hammond[2], and Stanley E. Trauth[5].
[1]Smithsonian Institution, National Museum of Natural History, [2]Quinnipiac University, [3]Eastern Oklahoma State College, [4]Missouri Department of Conservation, [5] Arkansas State University

1:15

NEW HOST AND LOCALITY RECORDS FOR THE FISH LEECHES *MYZOBDELLA LUGUBRIS* AND *MYZOBDELLA REDUCTA* (HIRUDINIDA: PISCICOLIDAE) FROM ARKANSAS AND OKLAHOMA

Dennis J. Richardson[1], William E. Moser[2], Chris T. McAllister[3], Renn Tumlison[4], Charlotte I. Hammond[1], Henry W. Robison[5], David A. Neely[6]. Quinnipiac University[1], [2]Smithsonian Institution, National Museum of Natural History, [3]Eastern Oklahoma State College, [4]Henderson State University, [5]Southern Arkansas University, [6]Tennessee Aquarium Conservation Institute

1:30

A REVIEW OF SYMBIOTIC RELATIONSHIPS IN STENOPODIDEAN SHRIMPS

Joseph W. Goy. Harding University

1:45

MISCELLANEOUS FISH HELMINTH PARASITE (TREMATODA, CESTOIDEA, NEMATODA, ACANTHOCEPHALA) RECORDS FROM ARKANSAS.

C.T. McAllister[1*], C.R. Bursey[2], H.W. Robison[3], D.A. Neely[4], M.B. Connior[5], and M.A. Barger[6]. [1]Science and Mathematics Division, Eastern Oklahoma State College, Idabel, OK 74745; [2]Department of Biology, Pennsylvania State University, Shenango Campus, Sharon, PA 16146; [3]9717 Wild Mountain Drive, Sherwood, AR 72120; [4]Tennessee Aquarium Conservation Institute, Chattanooga, TN 37402; [5]Health and Natural Sciences, South Arkansas Community College, El Dorado, AR 71730; and [6]Department of Natural Sciences, Peru State College, Peru, NE 68421.

2:00

DISTRIBUTION, HABITAT, AND STATUS OF THE DITCH FENCING CRAYFISH, *FAXONELLA CLYPEATA* (HAY) (DECAPODA: CAMBARIDAE) IN ARKANSAS.

H.W. Robison[1] and C.T. McAllister[2]. [1]9717 Wild Mountain Drive, Sherwood, AR 72120; and [2]Science and Mathematics Division, Eastern Oklahoma State College, Idabel, OK 74745.

2:15

PREVALENCE OF *CYTAUXZOON FELIS* IN FERAL CATS IN RUSSELLVILLE ARKANSAS

Cynthia H. Jacobs, Rachael E. Urbanek, and Catherine Normand. Arkansas Tech University

ORAL SESSIONS II: FRIDAY 3:00-5:00

PHYSICS AND ENGINEERING 207
CHAIR: Charles Wu

3:00

DYNAMIC EFFECT OF TORNADO FORCES ON CYLINDRICAL STRUCTURES

Majdi Yousef, R. Panneer Selvam, and Piotr Gorecki. University of Arkansas, Fayetteville

3:15

SPEED AND CURRENT RELATION IN ANTI-FORCE BREAKDOWN WAVES

Ryan Evans, and Mostafa Hemmati. Arkansas Tech University

3:30

WAVE PROFILE FOR LOW SPEED CURRENT BEARING WAVES

Preston Galla, and Mostafa Hemmati. Arkansas Tech University

3:45

PHOTOVOLTAIC MATERIALS RESEARCH AT ARKANSAS STATE UNIVERSITY-AN UPDATE

Joshua Vangilder, Dr. Robert Engelken, M. Jason Newell, Maqsood Ali Mughal, Kayla Wood, and Shyam Thapa. Arkansas State University

4:00

GROWTH OF ZnO NANOROD AND NANOFLOWER STRUCTURES BY A FACILE TREATMENT OF Zn FILMS IN HOT DE-IONIZED WATER

Khedir R Khedir, Zubayda S Saifaldeen, Taha Demirkan, Rosure B. Abdulrahman, and Tansel Karabacak. Department of Applied Science University of Arkansas at Little Rock

4:15

SPUTTER DEPOSITED TUNGSTEN CARBIDE (WC) NANORODS FOR CATALYTIC APPLICATIONS AND THIN FILMS WITH COMPLIANT LAYERS FOR WEAR RESISTANT APPLICATIONS

Mehmet F. Cansizoglu, and Tansel Karabacak. Department of Applied Science, University of Arkansas at Little Rock

4:30

EFFICIENCY MEASUREMENT OF WHITE LED DEVICE

Ahmed Zurfi, Dallas Tompkins, and Jing Zhang. Department of Systems Engineering, University of Arkansas at Little Rock

4:45

A COMPARISON OF THE DIFFERENT INDEPENDENT TECHNIQUES FOR MEASURING SUPERMASSIVE BLACK HOLE MASSES

Ismaeel Al-Baidhany[1], Marc Seigar[1], Patrick Treuthardt[1], Amber Sierra[1], Ben Davis[2], Daniel Kennifick[2], Julia Kennefick[2], and Claud Lacy[2]. [1]University of Arkansas at Little Rock. [2]University of Arkansas at Fayetteville

CHEMISTRY & BIOCHEMISTRY 208
CHAIR: Kevin Stewart

3:00

AN OPTICAL NANOSAT DETECTION AND RANGING SYSTEM (SADARS)

Maurisa Orona, Andrew Couch, and Edmond Wilson. Harding University

3:15
APPARATUS FOR CONCENTRATING PLANT AND HUMAN VOLATILES
Maegen Sloan, Brittany Gibson, Ozioma Whittaker, and Edmond Wilson. Harding University

3:30
DESIGNS FOR A RAMAN SPECTROMETER
Maria Medrano, Trevor Drury, and Edmond Wilson. Harding University

3:45
DESIGNING A SAMPLE HOLDER AND OPTICS FOR USE IN RAMAN SPECTROSCOPY
Trevor Drury, Maria Medrano, and Edmond Wilson. Harding University

4:00
OPTIMIZATION OF FUEL GRAIN GEOMETRIES FOR HYBRID ROCKETS USING AN ADDITIVE MANUFACTURING PROCESS
Rachel Beeman, and Edmond Wilson. Harding University

4:15
EXAMINING THE CLAIMS OF NATURAL PRODUCTS: AN EVALUATION OF THE ANTIMICROBIAL AND ANTIOXIDANT PROPERTIES OF AUSTRALIAN TEA TREE OIL
Chelsea Essary, and Dennis Province. Harding University

4:30
SPECTROSCOPY OF COMBUSTION OF HYDROCARBONS FROM 200 NM TO 1650 NM
Maddison Greene, and Edmond Wilson. Harding University

4:45
THE CRYSTAL STRUCTURE EFFECT OF THE SYNTHESIS OF COBALT OXIDE NANOPARTICLES USING MULTIPLE ITERATIONS OF A RECYCLABLE PRECIPITATION REACTION
J.S. Pennington, and R. J. Tackett. Arkansas Tech University

CELL & MOLECULAR BIOLOGY 209
CHAIR: Grover Miller

3:00
PROTECTIVE EFFECTS OF AQUEOUS EXTRACT OF *TERMINALIA ARJUNA* BARK AGAINST DOXORUBICIN-INDUCED CARDIOTOXICITY
Sarah Bishop[1], and Shi Liu[2].
[1]Ouachita Baptist University, Dept. of Pharmaceutical Sciences, College of Pharmacy, [2]University of Arkansas for Medical Sciences

3:15
ASSESSMENT OF TOTAL PHENOLICS AND ANTI-OXIDATION CAPACITIES IN WHEAT SEEDS
Michelle Poe[1] , Andra Bates[1] , Luther Talbert[2], Jamie Sherman[2], and Joseph Onyilagha[1].
[1]University of Arkansas at Pine Bluff, [2]Montana State University

3:30
INTRAVESICAL CHITOSAN/IL-12 IMMUNOTHERAPY INDUCES TUMOR-SPECIFIC SYSTEMIC IMMUNITY AGAINST BLADDER CANCER
Sean Smith, and David Zaharoff. University of Arkansas

3:45
FRESHMAN FIND PHAGE!
Jessica Ashcraft, Ruth Plymale, and Nathan Reyna.
Ouachita Baptist University

4:00
AGING IS A DETERMINANT IN ANOXIA STRESS TOLERANCE IN CAENORHABDITIS ELEGANS
J. M. Goy. Harding University

4:30
INBRE Opportunities

VERTEBRATE BIOLOGY 210
CHAIR: Cynthia Jacobs

3:00
A TEST OF ALTERNATIVE MODELS FOR INCREASED TISSUE NITROGEN ISOTOPE RATIOS DURING FASTING IN HIBERNATING ARCTIC GROUND SQUIRRELS
Trixie Lee[1], C. Loren Buck[2], and Brian M. Barnes[2]
[1]Harding University, [2]University of Alaska Anchorage

3:15
USING eDNA FROM SOIL SAMPLES TO DETECT TERRESTRIAL SPECIES
Subir B. Shakya, Pablo A. Bacon, and Abraham E. Tucker.
Southern Arkansas University

3:30
ECOLOGY OF THE SQUIRREL TREEFROG (HYLA SQUIRELLA) IN SOUTHERN ARKANSAS
M. B. Connior[1*], T. Fulmer[2], C. T. McAllister[3], and C. R. Bursey[4]
[1]Health and Natural Sciences, South Arkansas Community College, El Dorado, AR 71730, [2]1033 Magnolia Drive, El Dorado, AR 71730, [3]Science and Mathematics Division, Eastern Oklahoma State College, Idabel, OK 74745, [4]Department of Biology, Pennsylvania State University-Shenango Campus, Sharon, PN 16146

3:45
PLEISTOCENE SEED DISPERSAL OF ANACHRONISTIC FRUITS: USING ELEPHANTS TO TEST ANCIENT PLANT-ANIMAL INTERACTIONS
Charlie N. Davis, Madison J. Boone, and Laura Klasek. Hendrix College

4:00
LAND USE CHANGES IN THE FAYETTEVILLE SHALE GAS DEVELOPMENT REGION
Alex B. Cox, Rachel L. Wells, and Chloe Benichou. Hendrix College.

4:15
GROWTH AND REPRODUCTION IN THE OUACHITA MADTOM (NOTURUS LACHNERI) AT THE PERIPHERY OF ITS DISTRIBUTION
Renn Tumlison, and James O. Hardage III. Henderson State University

INVERTEBRATE BIOLOGY Liberty Room
CHAIR: Joseph Goy

3:00
PLAGIOPORUS (DIGENEA: OPECOELIDAE) OF ARKANSAS AND MISSOURI
Thomas J. Fayton[1], Chris T. McAllister[2], and Matthew B. Connier[3].
[1]Department of Coastal Sciences, Gulf Coast Research Laboratory, University of Southern Mississippi, [2]Division of Science and Mathematics, Eastern Oklahoma State College, [3]Health and Natural Sciences, South Arkansas Community College

3:15
PROPORTIONALITY OF POPULATION PARAMETERS OF *CLINOSTOMUM METACERCARIE* IN THE OROBRANCHIAL CAVITY OF *MICROPTERUS DOLOMEIU* AND *M. PUNCTULATUS*
James J. Daly, Sr. University of Arkansas for Medical Sciences (retired)

3:30
A COMPARATIVE STUDY OF HELMINTH PARASITES OF THE MANY-RIBBED SALAMANDER, EURYCEA MULTIPLICATA AND OKLAHOMA SALAMANDER, EURYCEA TYNERENSIS (CAUDATA: PLETHODONTIDAE), FROM ARKANSAS AND OKLAHOMA

C.T. McAllister[1*], M.B. Connior[2], C.R. Bursey[3], and H.W. Robison[4].
[1]Division of Science and Mathematics, Eastern Oklahoma State College, Idabel, OK 74745, [2]Health and Natural Sciences, South Arkansas Community College, El Dorado, AR 71730, [3]Department of Biology, Pennsylvania State University, Shenango Campus, Sharon, PA 16146
[4]9717 Wild Mountain Drive, Sherwood, AR 72120

3:45
NOTES ON TARANTULA (*APHONOPELMA HENTZI*) REPRODUCTION IN ARKANSAS
Austin Jones[1], David H. Jamieson[2], and Terry L. Jamieson[2].
[1]NorthWest Arkansas Community College, [2]Crowder College – Cassville Campus

4:00
FIRST RECORD OF THE AMERICAN BURYING BEETLE, NICROPHORUS AMERICANUS, IN CLARK COUNTY, ARKANSAS
Jess Kelly. Ouachita Baptist University

4:15
POPULATION GENOMICS OF THE MICROCRUSTACEAN *DAPHNIA PULEX*
Abraham Tucker, Ph.D.[1], L. Clai Morehead[1], and Matthew Ackerman[2].
[1]Southern Arkansas University, [2]Indiana University

ORAL SESSIONS II: SATURDAY 8:30-10:00

PHYSICS AND ENGINEERING 207
CHAIR: Matt Strasser

8:30
THE DEVELOPMENT OF CRITICAL TECHNOLOGIES FOR NANO-SATELLITE PROXIMITY OPERATIONS AT ARKANSAS
Adam Huang[1], Edmond Wilson[2], and Yupo Chan[3].
[1]University of Arkansas, Fayetteville, [2]Harding University, [3]University of Arkansas, Little Rock

8:45
BRINGING THE LEAST ACTION PRINCIPLE INTO INTRODUCTORY PHYSICS LABS
Juan D. Serna, and Jared Gavin. University of Arkansas at Monticello

9:00
SOFTWARE & HARDWARE DESIGN FOR A HIGH RESOLUTION FIBER-FED VISIBLE/NEAR INFRARED SPECTROGRAPH
Anna Shafer, Joshua Griffith, Brennan Thomason, and Edmond Wilson.
Harding University

9:15
MARS ROVER 3-D CAMERA
Aaron Burns, and Edmond Wilson. Harding University

9:30
REMOTE CONTROL OF MOBILE ROBOTIC VEHICLE VIA WEB-INTERFACE
Douglas Bailey, Justin Nesselrotte, and Edmond Wilson.
Harding University

PHYSICS AND ENGINEERING 209
CHAIR: Charles Wu

8:30
SPECTROSCOPY OF EARTH'S ATMOSPHERE AND SOLAR RADIATION IN THE SPECTRAL RANGE OF 400 nm TO 1000 nm
Stephanie Inabnet, and Edmond Wilson. Harding University

8:45
SHALE GAS DEVELOPMENT AND ITS ASSOCIATED IMPACTS ON ENERGY PATTERN IN THE UNITED STATES
Qiaozi Zhao, and Charles Wu. Harding University

9:00
OPTIMAL EXPERIMENTAL DESIGN IN INVERSION WITH APPLICATION TO SUBSURFACE SENSING
Yijun Yu[1], and Nailong Guo[2]. [1]Philander Smith College, [2]Benedict College

9:15
DESIGNING A HIGH RESOLUTION FIBER-FED SPECTROGRAPH FOR SOLAR OBSERVATIONS
Edmond Wilson, Brennan Thomason, Stephanie Inabnet, and Tamara Reed. Harding University

9:30
HIGH PRESSURE XENON TPC RADIATION BACKGROUND FOR NEXT EXPERIMENT
Abdel Bachri[1], Perry Grant[2], Clayton Martin[1], and Martin Hawron[3]. [1]Southern Arkansas University, [2]University of Arkansas, [3]University of Connecticut

PLANT SCIENCE Liberty room
CHAIR: Ann Willyard

8:30
REDISCOVERY OF PERSEA BORBONIA VAR. BORBONIA (LAURACEAE), PROSOPIS GLANDULOSA VAR. GLANDULOSA (FABACEAE), AND PINUS PALUSTRIS (PINACEAE) IN ARKANSAS, WITH THREE NEW ANGIOSPERM SPECIES FOR ARKANSAS
Tiffany Roeser[1], James H. Peck[2], and Brett E. Serviss[1].
[1]Henderson State University, [2]University of Arkansas at Little Rock

8:45
THE STATUS OF CARDAMINE DISSECTA (BRASSICACEAE) IN ARKANSAS
Karen P. Fawley[1], C. Theo Witsell[2], and Marvin W. Fawley[1].
[1]University of Arkansas at Monticello, [2]Arkansas Natural Heritage Commission

9:00
THE PRESENCE OF ASIATIC SPECIES OF SEAWEED ON THE TEXAS COAST
Randi Lovell, Megan Reed, and Troy L. Bray. Henderson State University

9:15
SERENDIPITOUS DATA FOLLOWING A SEVERE WINDSTORM IN AN OLD-GROWTH PINE STAND
Don C. Bragg[1], and Jess Riddle[2].
[1]USDA Forest Service, [2]University of Arkansas

9:30
ALGAE AND SNAILS INTERACT TO AFFECT LEAF DECOMPOSITION RATES
Ali Mcleod, Steven Polaskey, and Sally Entrekin.
University of Central Arkansas

POSTER PRESENTATION
(Presenter is underlined)

LIFE SCIENCES POSTERS

LS 38 EASTERN BLUEBIRD DIET, BEHAVIOR, AND WATER CONTENT OF PREY ITEMS
Brandi Cansler, and Virginie Rolland.
Arkansas State University Department of Biological Sciences

Meeting Report

LS 39 THE INTRODUCED DIRT-COLORED SEED BUG, *MEGALONOTUS SABULICOLA* (HEMIPTERA: RHYPAROCHROMIDAE) AND WATER BOATMEN, *SIGARA MATHESONI* (HEMIPTERA: CORIXIDAE): NEW FOR ARKANSAS.

S.W. Chordas III[1], C.T. McAllister[2], and H.W. Robison[3].
[1]Center for Life Sciences Education, The Ohio State University, 260 Jennings Hall, 1735 Neil Avenue, Columbus, OH 43210; [2]Science and Mathematics Division, Eastern Oklahoma State College, Idabel, OK 74745; and [3]9717 Wild Mountain Drive, Sherwood, AR 72120.

LS 40 DETERMINING THE EFFECT OF ACTIN DEPOLYMERIZATION OF *DICTYOSTELIUM DISCOIDEUM* MITOCHONDRIA

Jordyn Cleavenger, Greg Berbusse, and Kari Naylor.
University of Central Arkansas

LS 41 NEW RECORDS OF ECTOPARASITES AND OTHER EPIFAUNISTIC ARTHROPODS FROM SCALOPUS AQUATICUS AND BLARINA CAROLINENSIS IN ARKANSAS

M.B. Connior[1*], L.A. Durden[2], and C.T. McAllister[3].
[1]Health and Natural Sciences, South Arkansas Community College, El Dorado, AR 71730, [2]Department of Biology, Georgia Southern University, Statesboro, GA 30458, [3]Science and Mathematics Division, Eastern Oklahoma State College, Idabel, OK 74745

LS 42 NATURAL HISTORY NOTES AND RECORDS OF VERTEBRATES FROM ARKANSAS

M.B. Connior[1*], R. Tumlison[2], H.W. Robison[3], C.T. McAllister[4], and D.A. Neely[5]
[1]Health and Natural Sciences, South Arkansas Community College, El Dorado, AR 71730, [2]Department of Biology, Henderson State University, Arkadelphia, AR 71999, [3]9717 Wild Mountain Drive, Sherwood, AR 72120 [4]Division of Science and Mathematics, Eastern Oklahoma State College, Idabel, OK 74745, [5]Tennessee Aquarium, Chattanooga, TN

LS 43 FIRST RECORD OF RIBBON WORMS (NEMERTEA: TETRASTEMMATIDAE: PROSTOMA) FROM ARKANSAS.

P.G. Davison[1], H.W. Robison[2], and C.T. McAllister[3].
[1]Department of Biology, University of North Alabama, Florence, AL 35632; [2]9717 Wild Mountain Drive, Sherwood, AR 72120; and [3]Science and Mathematics Division, Eastern Oklahoma State College, Idabel, OK 74745.

LS 44 SARCOPHAGEOUS INSECT ASSOCIATIONS AND SUCCESSION ON PIG CARRION DURING THE SUMMER IN CENTRAL ARKANSAS

Tyler Files, Brianne Baley, and Jess Kelly. Ouachita Baptist University

LS 45 PREVALENCE OF SHIGA TOXIN-PRODUCING *ESCHERICHIA COLI*

David F. Gilmore[1], Harneet Kaur[2], Monica Yarbrough[3], and Donald Kennedy[3]. [1]Biological Sciences, [2]Environmental Sciences Program, [3]College of Agriculture, Arkansas State University

LS 46 SAMPLING LOCAL FUNGAL DIVERSITY: A METHOD FOR FUNGAL SPECIES IDENTIFICATION USING DNA BARCODING

A.H. Harrington, A.F. Bigott, B.W. Anderson, M.J. Boone, S.M. Brick, J.F. delSol, C.A. Hotchkiss, R.A. Huddleston, E.H. Kasper, J.J. McGrady, M.L. McKinnie, M.V. Ottenlips, N.E. Skinner, K.C. Spatz, A.J. Steinberg, F. van den Broek, C.N. Wilson, A.M. Wofford and A.M. Willyard.
Hendrix University

LS 47 DETERMINATION OF URSOLIC ACID AND BIOACTIVITY IN ILEX DECIDUA

Cynthia Holland, and Martin Campbell. Henderson State University

LS 48 NEW HOST AND LOCATION RECORDS FOR THE BAT BUG *CIMEX ADJUNCTUS* BARBER 1939, WITH A SUMMARY OF PREVIOUS RECORDS

John Hunt[1], Matthew E. Grilliot[2], and Christopher G. Sims[1].
[1]University of Arkansas at Monticello, [2]Troy University Montgomery

LS 49 LOCALIZATION STUDY OF BETA-HEXOSAMINIDASE IN THE SOCIAL AMOEBA DICTYOSTELIUM DISCOIDEUM

Sanaa Talib Jawed, Azure Yarbrough, and John Bush.
University of Arkansas at Little Rock

LS 50 THE SUGAR AND AMINO ACID CONCENTRATIONS OF EXTRAFLORAL NECTAR IN FIVE COTTON CULTIVARS

Soolaf A. Kathiar, Janet Lanza, and Anindya Ghosh.
University of Arkansas at Little Rock

LS 51 NEW HOST RECORDS FOR MESOCESTOIDES SP. TETRATHYRIDIA (CESTOIDEA: CYCLOPHYLLIDEA) IN AMPHIBIANS (ANURA: BUFONIDAE, RANIDAE) FROM ARKANSAS.

C.T. McAllister[1], M.B. Connior[2], and S.E. Trauth[3]. [1]Science and Mathematics Division, Eastern Oklahoma State College, Idabel, OK 74745; [2]Health and Natural Sciences, South Arkansas Community College, El Dorado, AR 71730; and [3]Department of Biological Sciences, Arkansas State University, State University, AR 72467.

LS 52 *HAEMOGREGARINA SP.* (APICOMPLEXA: HAEMOGREGARINIDAE), *TELORCHIS ATTENUATA* (DIGENEA: TELORCHIIDAE) AND *NEOECHINORHYNCHUS EMYDIS* (ACANTHOCEPHALA: NEOECHINORHYNCHIDAE) FROM MAP TURTLES (GRAPTEMYS SPP.), IN NORTHCENTRAL ARKANSAS.

C.T. McAllister[1*], C.R. Bursey[2], H.W. Robison[3], M.B. Connior[4], and M.A. Barger[5].
[1]Science and Mathematics Division, Eastern Oklahoma State College, Idabel, OK 74745; [2]Department of Biology, Pennsylvania State University, Shenango Campus, Sharon, PA 16146; [3]9717 Wild Mountain Drive, Sherwood, AR 72120; [4]Health and Natural Sciences, South Arkansas Community College, El Dorado, AR 71730; and [5]Department of Natural Sciences, Peru State College, Peru, NE 68421.

LS 53 THE KILLING MECHANISMS OF NOVEL ANTIFUNGAL PEPTIDES

Gabriela Morris, David McNabb, and Yazan Akkam.
University of Arkansas

LS 54 EFFECTS OF MICROGRAVITY AND RADIATION ON ELASTIC MODULUS OF RAT FEMURS USING 3-POINT BENDING

O. Perkins[1], A. Walker[1], R. Mehta[1], M. Dobretsov[2], and P. Chowdhury[3].
[1]Department of Physics and Astronomy, University of Central Arkansas, [2]Department of Anesthesiology, University of Arkansas for Medical Sciences, [3]Department of Biophysics and Physiology, University of Arkansas for Medical Sciences

LS 55 TOAD (ANURA: BUFONIDAE) LIMB ABNORMALITIES FROM AN AQUATIC SITE IN SCOTT, PULASKI COUNTY, ARKANSAS

Christopher S. Thigpen, Dan Beard, and Stanley E. Trauth.
Arkansas State University

LS 56 SELECTION OF FATTY ACID DESATURASE 7 (fad7-1) SINGLE MUTANT PLANTS IN *ARABIDOPSIS THALIANA* USING SNP-PCR PRIMER

Kaleb L. Vaughn[1], Carlos A. Avila[2], and Fiona Goggin[2].
[1]Harding University, [2]University of Arkansas

LS 57 MOLECULAR CLONING TO IMPROVE MITOCHONDRIAL FISSION AND FUSION ASSAYS

Olivia Vogel, Kalyn Holloway, and Kari Naylor.
University of Central Arkansas

LS 58 NATURAL HISTORY NOTES AND NEW COUNTY RECORDS FOR OZARKIAN MILLIPEDS (ARTHROPODA: DIPLOPODA) FROM ARKANSAS, KANSAS AND MISSOURI.

N.W. Youngsteadt[1]and C.T. McAllister[2]. [1]2031 S. Meadowview Avenue, Springfield, MO 65804**;** and [2]Science and Mathematics Division, Eastern Oklahoma State College, Idabel, OK 74745.

CHEMISTRY & BIOCHEMISTRY POSTERS

CBC 15 EXPLORING COORDINATION CHEMISTRY OF TRIS(PYRAZOL-1-YL)METHANE

Morgan Coleman, and Ganna Lyubartseva. Southern Arkansas University

CBC 16 CRITICAL DISSOLVED OXYGEN LEVELS IN TRIBUTARIES OF THE BUFFALO NATIONAL RIVER

John Kincaid[1], Faron Usrey[2], and Sherri Townsend[1].
[1]North Arkansas College, [2]National Park Service

CBC 17 ISOTHERMAL TITRATION CALORIMETRY FOR QUANTIFYING LACTOPEROXIDASE ACTIVITY IN MILK

William Tolleson[1], and Clifton Lewis, Jr.[2] [1]National Center for Toxicological Research, [2]Watson Chapel School District

CBC 18 IRON-CARBON RELATIONSHIP BETWEEN BOND LENGTH AND BOND VALENCE

Larkin McDaniel, and Franklin D. Hardcastle. Arkansas Tech University

CBC 19 NEUTRALIZATION AND pH EFFECT OF MILK ON ASPIRIN SOLUTIONS

Jordan Miller, Nikisha West, Fontaine Taylor, and Insu 'Frank' Hahn. Department of Chemistry, Division of Natural and Physical Sciences, Philander Smith College, Little Rock, AR

CBC 20 WATER QUALITY STUDY AT LAKE COLUMBIA, AR

Kara O'Neal, and Casey O'Hara

CBC 21 EFFECTS OF MICROGRAVITY AND RADIATION ON ELASTIC MODULUS OF RAT FEMURS USING 3-POINT BENDING

O. Perkins[1], A. Walker[1], R. Mehta[1], M. Dobretsov[2], and P. Chowdhury[2], [1]University of Central Arkansas, [2]University of Arkansas for Medical Sciences

CBC 22 MOLECULAR MODELING STUDIES OF PHYLOGENETICALLY SIGNIFICANT CAROTENOIDS OF OXYGENIC PHOTOTROPHS

Ryan Reyes, and M. Jeffery Taylor. University of Arkansas-Monticello

CBC 23 SPICES: TESTING THEIR ANTIMICROBIAL PROPERTIES AND SYNERGISM WITH PENICILLIN ON ESCHERICHIA COLI

Gregory Tyler Rives. Southern Arkansas University

CBC 24 SUPERAMPHIPHOBIC ALUMINUM ALLOY SURFACES

Zubayda S Saifaldeen, Khedir R Khedir, and Tansel Karabacak, Department of Applied Science, UALR

CBC 25 BIOCHEMISTRY OF CHROMIUM

Shelby Sorrells, Silas Brown, and Edmond Wilson. Harding University

CBC 26 MEASURING PAIN WITHDRAWAL THRESHOLD USING A NOVEL DEVICE OPERATING IN "PSEUDO-CONTINUOUS" MODE

Azida Walker[1], Nick Martinez[1], Skipper Thurman[1,] Shelby Burns[1], and Maxim Dobretsov[2]. [1]University of Central Arkansas, [2]University of Arkansas for Medical Sciences

CBC 27 FRESHMAN FIND PHAGE!

Jessica Ashcraft, Ruth Plymale, and Nathan Reyna. Ouachita Baptist University

PHYSICS AND ENGINEERING POSTERS

P&E 22 A STUDY OF THE RELATION BETWEEN THE SPIRAL ARM PITCH ANGLE AND THE KINETIC ENERGY OF RANDOM MOTIONS OF THE HOST SPIRAL GALAXIES

Ismaeel Al-Baidhany[1], Marc Seigar[1], Patrick Treuthardt[1], Amber Sierra[1], Ben Davis[2], Daniel Kennifick[2], Julia Kennefick[2], and Claud Lacy[2]. [1]University of Arkansas at Little Rock, [2]University of Arkansas at Fayetteville

P&E 23 A STUDY OF THE DISCREPANCY BETWEEN DYNAMICAL MASSES AND STELLAR MASSES IN SPIRAL GALAXIES

Ismaeel Al-Baidhany[1], Marc Seigar[1], Patrick Treuthardt[1], Amber Sierra[1], Ben Davis[2], Daniel Kennifick[2], Julia Kennefick[2], and Claud Lacy[2]. [1]University of Arkansas at Little Rock, [2]University of Arkansas at Fayetteville

P&E 24 STRUCTURAL AND OPTICAL PROPERTIES OF ALUMINUM NANORODS FABRICATED BY GLANCING ANGLE DEPOSITION (GLAD)

Rosure B. Abdulrahman, Mehmet F. Cansizoglu, and Tansel Karabacak. University of Arkansas at Little Rock

P&E 2 RUTHERFORD SCATTERING – KINEMATICS AND ANGULAR DEPENDENCE

Fawzi Alzahrani, Cruz Segura, Dawson Long, Forrest McDougal, Lawrence Benzmiller, R. Mehta, and A. Walker. University of Central Arkansas

P&E 26 SYNCHRONIZATION LIMITS OF CHAOTIC CIRCUITS

Christopher M. Church, and Stephen R. Addison. University of Central Arkansas

P&E 27 SUPERHET CONVERTER FOR VLF 60kHz WWVB RADIO SIGNAL

Kody Coleman, Juan D. Serna. University of Arkansas at Monticello

P&E 28 A FRAMEWORK FOR MODELING USABILITY OF TEXT-BASED CAPTCHA

Jasmine DeHart, Jalen Mayfield, and Samar Swaid. Philander Smith College

P&E 29 STABLE FUZZY LOGIC CONTROL FOR NONLINEAR SIMPLE CHAOTIC MAPS

Christopher Gillison, and Juan D. Serna. University of Arkansas at Monticello

P&E 30 GEOMETRICAL VALIDATION OF LINEAR AND NONLINEAR DIOPHANTINE EQUATIONS

Kyra Jerry, and Juan D. Serna. University of Arkansas at Monticello

P&E 31 INVESTIGATION AND COMPARISON OF TWO BASIC AUDIO AMPLIFIER CIRCUITS

Michael Kelley, and Dr. Charles Wu. Harding University

P&E 32 TOTAL EXCITATION SCATTERING CROSS SECTION FOR POSITRONS SCATTERED BY MOLECULAR NITROGEN IN THE ENERGY RANGE (3 – 10 keV)

H.L. Mansour[1] and W.A. Jabbar[2]. [1]Al – Mustansiriyah University, [2]University of Arkansas at Little Rock

P&E 33 DETERMINATION OF THE TOTAL EXCITATION SCATTERING CROSS SECTIONS OF MOLECULAR NITROGEN FOR POSITRONS AT ENERGIES UP TO (10 keV)

H.L. Mansour[1] and W.A. Jabbar[2]. [1]Al–Mustansiriyah University, [2]University of Arkansas at Little Rock

P&E 34 FABRICATION OF CDS NANORODS AND NANOPARTICLES WITH PANI FOR DYE-SENSITIZED SOLAR CELLS

Muatez Zamil Mohammed, and Tar-Pin Chen.
University of Arkansas at Little Rock

P&E 35 APPLYING PROBABILISTIC MATCHING AND POST RESOLUTION CLERICAL REVIEW TECHNIQUES TO IMPROVE ENTITY RESOLUTION RESULTS

Daniel Pullen, Pei Wang, and John Talburt.
University of Arkansas at Little Rock

P&E 36 GAMMA GAMMA COINCIDENCE

Lucus Ratz, Doug Roisen, Jeremy Jacobs, Tanner Feeler, Azida Walker, and Rahul Mehta. Department of Physics and Astronomy, University of Central Arkansas

P&E 37 X-RAY FLUORESCENCE AND MOSELEY'S LAWS OF NUCLEAR RADIATION SPECTRA

T. Ryan Rogers, Hunter P. Ward, Nicholas L. Frederickson, Anthony D. Mitchel, Azida Walker, and Rahul Mehta. University of Central Arkansas

P&E 38 CLASSIFICATION OF PARALLEL VORTEX-BODY INTERACTION

Matthew Strasser, and R. Panneer Selvam. University of Arkansas

P&E 39 NANOSAT SPECTROMETER

Edmond Wilson, Mauris Orona, and Andrew Couch. Harding University

P&E 40 ENERGY LOSS OF ALPHA PARTICLES IN COPPER FOILS

Xavier Redmon, Gerard Munyazikwiye, Ashley Cotnam, Azida Walker, and Rahul Mehta. University of Central Arkansas

Solid State Dye Sensitive Solar Cells Based on ZnO Nanowire as the N-type Semiconductor

S. AbdulAlmohsin

Department of Physics, University of Arkansas, Little Rock, AR 72204, USA

Correspondence: samir_mahdi47@yahoo.com

Running Title: Solid State Dye Sensitive Solar Cells on ZnO Nanowires

Abstract

We fabricated solid state dye-sensitized solar cells with ZnO nanorods as the n-type material and polypyrrole as the p-type material. The ZnO nanorods were grown on indium-tin oxide (ITO) glass by electrochemical methods for one hour. Scanning electron micrographs of the ZnO nanowire (NW) indicated a length of about 1 micrometer and a diameter of approximately 100-200 nm for the nanorods. Polypyrrole deposited on ITO/ZnO NW/dye and the fabricated device of ITO glass/ZnO nanorods/dye/polypyrrole/Ag showed a power conversion efficiency of 1.29%

Introduction

New concepts and devices that are challenging photovoltaic applications based on p-n junctions have been reported. Currently, exciton solar cells such as organic (Chen et al. 2012), hybrid organic-inorganic (AbdulAlmohsin et al. 2012) and dye sensitive solar cells (DSCs)(Kenneth et al. 2013) are promising devices for inexpensive, large scale solar energy conversion. The most commonly used types are TiO_2 nanoparticles and ZnO nanorods as working electrodes with CuSCN as a counter p-type electrode (Umang et al. 2012), however we are using polypyrrole as a counter electrode instead of CuSCN.

Here we report on a solid-state DSC consisting of ZnO nanorods as a wide band gap material, ruthenium dye (N719) and polypyrrole (PPY) doped by Li^+ as a hole transport conductor (Fig. 1). The mechanism of the device is that incoming light is absorbed in the dye monolayer and excites electrons from the highest occupied molecular orbital (HOMO) to the lowest unoccupied molecular orbital (LUMO). The excited electrons are then injected into the conduction band of the ZnO by taking an electron from the HOMO level of the polypyrrole.

Methods

Glass coated with indium-tin oxide (ITO) was supplied by SPI supplies (West Chester, PA, U.S.A). The vertically aligned ZnO nanowire arrays were fabricated on ITO glass substrates (SPI Supplies) by a low temperature electrochemical method (Chen et al. 2011). The ZnO nanorod electrode was immersed in an 3mM ethanolic solution of ruthenium dye (N719) (Sigma-Aldrich) overnight, washed with ethanol and then introduced to deposit polypyrrole (PPY) by electrochemical polymerization using platinum wire as the counter electrode and 0.1 M of pyrrole monomer dissolved in acetonitrile and 0.1 M LiI salt with 2 volts as the applied voltage between counter and working electrode for 3 min. The PPY thin film deposited directly on the ZnO NW/dye. The electrode was then placed on top of the PPY using silver paste.

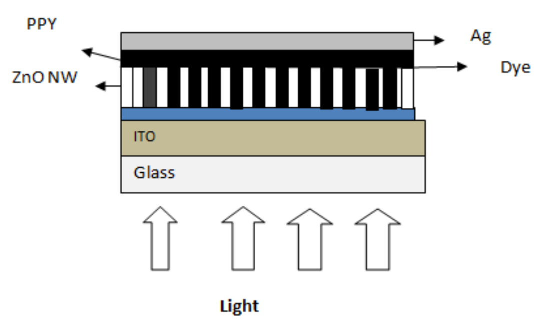

Figure1. Schematic of the ZnO/dye/PPY/Ag solar cells

Results and Discussion

Figure 2 shows a scanning electron microscope (SEM) top view image of a typical ZnO NW array grown by the electrochemical process. The nanowires had an average diameter of 100-200 nm and are vertically aligned on the substrate. The surface of the nanowire array is clean and free of particles and the roots of the wires separated from each other - both are important factors that affect the performance of the device of solar cells

In order to investigate the effects of dye on the optical properties of ZnO NWs, the UV-Vis absorption spectra were measured for ZnO NWs with and without dye N719 (Fig. 3). The ZnO NW/dye has strong absorption peaks around 434 nm, 522 nm, and 673 nm, the latter two being in the visible range. The modified ZnO NW with Dye N719 exhibit a strong increase in absorbance over the entire measured range relative to pristine ZnO NWs .

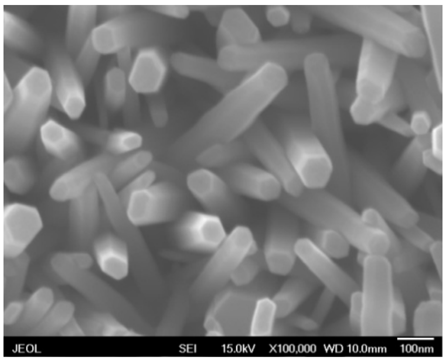

Figure 2. SEM cross-section image of ZnO nanowire arrays.

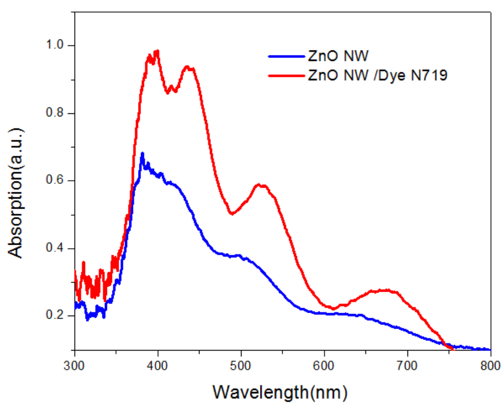

Figure 3. UV-Vis absorption spectra of ZnO NW and ZnO NW/Dye

The current density versus voltage curve shown in Fig. 4 indicates that the ZnO NW with Dye shows typical photovoltaic characteristics under illumination. An open-circuit voltage (V_{oc}) of 0.36 V and short-circuit current density of 7.3 mA/cm^2 was observed with a fill factor of 0.49. The overall power conversion efficiency of this solar cell is 1.29 % (Fig. 4). The high short circuit current leads to the higher power conversion efficiency due to the conductive polymer Polypyrrole acts as the hole-transport material in the solid State dye solar cells with N-type ZnO NWs.

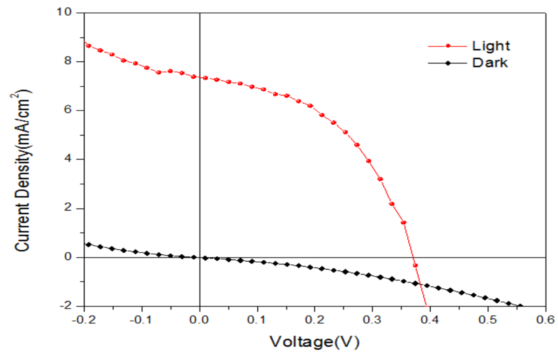

Figure 4. Current density – voltage curves measured on PPy/Dye/ ZnO nanowire

Conclusion

A solid-state dye sensitive solar cell has been fabricated using a vertically aligned ZnO nanowire array as the working electrode and polypyrrole (Li^+) as the hole-transport materials. The solid state dye cells give current densities of 7.3 mA/cm^2 and a voltage open-circuit voltage (V_{oc}) of 0.36 V. The power conversion efficiency is 1.29%, which is promising for a solar cell application.

Literature Cited

AbdulAlmohsin SM and **JB Cui**. 2012. Graphene-enriched P3HT and porphyrin -Modified ZnO nanowire arrays for Hybrid Solar Cells. The Journal of Physical Chemistry C 116:9433-9438.

Chen KS, **HL Yip**, **CW Shlenker**, **DS Ginger** and **AK Jen**. 2012. Halogen-free solvent processing for sustainable development of high efficiency organic solar cells. Organic Electronics 13(12):2870-2878.

Chen H, **W Li**, **Q Hou**, **H Liu** and **L Zhu**. 2011. Growth of three-dimensional ZnO nanorods by Electrochemical method for quantum dot-sensitized solar cells. Electrochimica Acta 56(24): 8358-8364.

Kenneth H, **DJ Wilger**, **ST Jones**, **DP Harrison**, **SE Bettis**, **H Lno** and **T Meyer**. 2013. Electron transfer of peptide-derivatized Ru polypyridyl complexes on nanocrystalline metal oxide film. Peptide Science 100(1):25-37.

Umang VD, **C Xu**, **J Wu** and **D Gao**. 2012. Solid -state dye -sensitized solar cells based on ordered ZnO nanowire arrays. Nanotechnology 23:205401-205406.

A Study of the Relation between the Spiral Arm Pitch Angle and the Kinetic Energy of Random Motions of the Host Spiral Galaxies

I. Al-Baidhany[1,3], M. Seigar[1], P. Treuthardt[1], A. Sierra[1], B. Davis[2], D. Kennefick[2], J. Kennefick[2], C. Lacy[2], Z.A. Toma[3], and W. Jabbar[3]

[1]*Department of Physics and Astronomy, University of Arkansas at Little Rock, AR 72204*
[2]*University of Arkansas at Fayetteville*
[3]*Al- Mustansiriyah University, Baghdad, IRAQ*

Correspondence: iaakhlite@ualr.edu

Running Title: A Relation between Pitch Angle and Kinetic Energy in Spiral Galaxies

Abstract

In this work, we report a relation between the kinetic energy of random motions of the corresponding host galaxies and spiral arm pitch angles ($M_{dyn}\sigma^2$ - P), ($M_*\sigma^2$ - P) where M_{dyn} is the bulge dynamical mass, M_* is bulge stellar mass, and σ is the velocity dispersion of the host galaxy bulge. We measured the spiral arm pitch angle (P) for a sample of Spitzer/IRAC 3.6-μm images of 54 spiral galaxies, estimated by using a 2D Fast Fourier Transform decomposition technique (2DFFT). We selected a sample of nearly face-on spiral galaxies and used IRAF ellipse to determine the ellipticity and major-axis position angle in order to deproject the images to face-on, and using a 2D Fast Fourier Transform decomposition technique, we determined the spiral arm pitch angles. We estimated the kinetic energy of random motions of the corresponding host galaxies ($M_{dyn}\sigma^2$, $M_*\sigma^2$) by using M_{dyn}, M_*, and σ, where the stellar velocity dispersion (σ) of the bulge was taken from the literature. We determined the bulge dynamical mass (M_{dyn}) using the virial theorem, and the bulge stellar mass (M_*) was estimated by using the bulge 3.6-μm luminosity with the appropriate stellar mass-to-light ratio (M/L).

Introduction

It is becoming apparent that the energy output from supermassive black holes (BH) at galaxy centers plays a important role within the formation and evolution of galaxies (Pastorini et al. 2007). Over the past 15 years, one of the most important advances and the most fascinating discoveries was that galaxies typically contain supermassive black holes at their centers, on the order of millions to billions of solar masses (Heckman and Kauffmann 2011).

SMBH mass is an important parameter for us to understand nuclear energy mechanics and the formation and evolution of SMBHs and their host galaxies (Rees 1984, Tremaine et al. 2002). Nowadays astrophysicists believe that the energy released by growing SMBHs plays an important role in shaping the properties of the structure of galaxies (Benson and Bower 2010, Fabian 2012). The co-evolution of galaxies and SMBHs is now widely accepted although many details on how this coexistence works are still understudied (Heckman et al. 2004). Therefore, we cannot understand how galaxies formed and evolved without understanding the co-evolution of galaxies and SMBHs.

In light of the increasing evidence derived from scientific research that indicates that the mass of SMBHs are tightly related to the properties of their host galaxy bulges, it seems obvious that SMBHs play an important role in galaxy formation.

Most galaxy bulges contain a central supermassive black hole whose mass strongly correlates with stellar velocity dispersion (σ^*) within the effective radius (r_e) (Ferrarese and Merritt 2000, Gebhardt et al. 2000, Tremaine et al. 2002) with the bulge luminosity or spheroid luminosity of the galaxy (L_{bul}) (Kormendy and Richstone 1995, Magorrian et al. 1998, Marconi & Hunt 2003, Häring and Rix 2004, Gültekin et al. 2009), with the bulge mass (M_{bulge}) (Magorrian et al. 1998, MH03, Häring and Rix 2004, hereafter HR04), and circular velocity (Ferrarese 2002), with the galaxy light concentration (Graham et al. 2001), the dark matter halo (Ferrarese 2002), with the effective radius (Marconi and Hunt 2003), the Sersic index (Graham and Driver 2007), with the gravitational binding energy and gravitational potential (Aller and Richstone 2007), combination of bulge velocity dispersion, effective radius and/or intensity (Aller and Richstone 2007), with the radio core length (Cao and Jiang 2002), and the inner core radius (Lauer et al. 2007a). Using more sophisticated techniques of measuring the bulge luminosity or dynamical modeling of the host galaxy such as two-dimensional image decompositions (e.g., McLure and Dunlop 2001, Wandel 2002, Häring and Rix 2004, Hu 2009, Sani et al. 2011), produces a tighter correlation between SMBHs and the host galaxy.

The results of Hopkins et al. (2007) and Marulli et al. (2008) provide evidence for a hypothesis that bulge of galaxy and SMBHs do not form and evolve independently. Furthermore, Feoli and Mancini (2009) explained the relation M_{bul}- σ^2 by using a plausible physical interpretation that resembles the H–R diagram, where they indicate that certain properties of SMBHs at the centers of galaxies, such as entropy, can increase with time or at most remain the same, but do not decrease. Therefore M_{BH} depends on the age of the galaxy.

Several previous studies have tested the M_{BH}–$M_{bul}\sigma_2$ relation using several independent galaxy samples, with clear positive results, and therefore the M_{BH}–$M_{bul}\sigma 2$ relation can be used as an indirect measurement of the SMBH mass in the center of galaxies (Feoli and Mele 2005,2007, Feoli and Mancini 2009, Mancini and Feoli 2012).

Previous work has found that central SMBH mass is strongly related with spiral arm pitch angle of its host galaxy (Seigar et al. 2008, Davis et al. 2012, Berrier et al. 2013). Pitch angle is the angle between a line tangent to the arm in a spiral galaxy at a given radius and a line tangent to a circle at the same radius. The degree of twist of the spiral arms is a characterization of the pitch angle, where the galaxies with small and large pitch angles have tightly wound spiral arms and open arms respectively (Kennicutt 1981, Ma 2001, Savchenko and Reshetnikov 2011). The measurement of spiral arm pitch angle gives a measure of how tightly the spiral arms of a galaxy are wound. Since the creation of a morphological classification scheme of galaxies by Hubble (1926), authors have competed to investigate the wide correlation of the spiral and morphological type of the observed galaxies (e.g., Kennicutt 1981).

Seigar et al. (2006) and Davis et al. (2012) concluded that pitch angle does not depend measurably on the waveband of the image. Instead, they found consistency between pitch angles of the same galaxy measured both in the B-band and in a near-IR waveband by using a 2D fast Fourier transform (2DFFT) analysis and assuming logarithmic spirals.

The objective of this work is to analyze the cited scaling relationships that involve bulge properties (M_{BH} –$M_{bul}\sigma^2$, M_{BH} –M_{bul}, $M_{bul}\sigma^2$– P and M_{bul}– P) in images of 41 spiral galaxies observed using the Spitzer Space Telescope at 3.6-μm.

Materials and Methods

Sample

Our sample in this research consists of a total of 41 spiral galaxies observed with the Spitzer Space Telescope at 3.6μm. The main requirement to estimate the kinetic energy of random motions of the corresponding host galaxies ($M_d\sigma^2$ & $M_*\sigma^2$) is an estimate of the bulge mass and the stellar velocity dispersion. We have measured both the bulge dynamical mass and the bulge stellar by applying the isothermal model (Hu 2009, Sani et al. 2011) and the calibration by Oh et al. (2008) respectively. The central velocity dispersion of the galaxy hosts were obtained from the literature (see Table 1 at the end of this manuscript).

Our sample consists of Hubble types ranging from Sa to Sc for which it is possible to measure pitch angle for each galaxy. We derived an inclination (ranging from 25 to 65 degrees) by using ellipticity values of the outer 3.6-μm isophotes, which were determined with ELLIPSE in IRAF[1]. Seigar et al. (2005, 2008) noted that the largest source of error in estimating P presumably comes from this determination of radial range, although P can also have a variance as large as 10% for galaxies with large inclinations (>60°) (Block et al. 1999)

In this paper, some of the galaxies had spiral arm pitch angles which had been previously determined by our research group using B- and K- band images (Seigar et al 2006, Davis et al 2012). The remaining spiral arm pitch angles were measured using Spitzer/IRAC 3.6-μm images of 41 galaxies using a two-dimensional fast Fourier transformation (Schröder et al. 1994), assuming logarithmic spirals. In this study, we have considered a consistent sample of 41 spiral galaxies, which consists of 27 barred galaxies, 14 non-barred galaxies, 31 AGN-host galaxies, 10 non-AGN galaxies, 10 galaxies with classical bulges, and 31 galaxies with pseudo-bulges.

[1] [1]IRAF is distributed by the National Optical Astronomy Observatories, which is operated by the Associated Universities for Research in Astronomy, Inc., under cooperative agreement with the National Science Foundation.

Measurement of the dynamical bulge mass:

The bulge dynamical mass M_{dyn} is estimated using the virial theorem, i.e., the virial bulge mass (Hu 2009, Marconi and Hunt 2003, Sani et al. 2011) given by:

$$M_{dyn} = kR_e\sigma^2/G \quad \ldots\ldots\ldots\ldots\ldots\ldots(1)$$

Where k is in general a function of the Sérsic index n (Sani et al. 2011, Jun and Im 2008), we follow the method of Cappellari et al (2006) and use k=5 and this can then be used to estimate an accurate value of M_{dyn}, where σ, and R_e are the host-galaxy bulge velocity dispersion and the bulge effective radius respectively, and G is the gravitational constant.

Measurement the stellar mass (M_*) from the 3.6 µm M/L ratio:

Bell and de Jong (2001) estimated the stellar mass-to-light (M/L or γ) ratio of disk galaxies by using relation between optical colors (e.g., B−R, B−V) and the near-infrared

Previous studies of optical colors of the disk of galaxies do not provide the γ values for the Spitzer/IRAC bands, so we cannot use them here. Therefore we will use a new relation to obtain γ in the 3.6-µm Spitzer/IRAC. This relationship is between γ^K and γ in the 3.6-µm waveband was reported by Oh et al. (2008):

$$\gamma^{3.6} = B^{3.6} \times \gamma^K + A^{3.6} \quad \ldots\ldots\ldots(2)$$

Where $A^{3.6} = -0.05$ and $B^{3.6} = 0.92$

And a relation between the (γK) and optical colors:

$$\log_{10}(\gamma^K) = b^K \times \text{Optical Color} + a^K \quad ..(3)$$

Where a^K and b^K are coefficients for the relation between γ^K and optical colors given in Bell and de Jong (2001).

By combining Equation (2) with Equation (3), adopting 20% solar metallicity (Miller and Hodge 1996), optical colors given in Bell and de Jong (2001) and a scaled Salpeter IMF[2] cutting off the stars less massive than ~0.35$M_\odot$ (Bell and de Jong 2001), we calculated the 3.6 µm M/L ratio.

[2] The initial stellar mass function

Measurement the bulge luminosity (L_{bulge}):

The method to measure the bulge luminosity in this work is based on a two-dimensional (bulge - bar - disk) decomposition program (Laurikainen et al 2005), which we used to decompose Spitzer/IRAC 3.6-µm images of spiral galaxies into a bulge and disk model. From the resulting bulge model, we determined bulge luminosity at 3.6-µm for the sample of 41 spiral galaxies. In this method, we used an exponential function to describe the disk:

$$Id(r) = I_{od}\exp[-(r/h_r)],$$

Where I_{od} is the central surface density of the disk, h_r is the exponential scalength of the disk, and r is distance from the galaxy center. The bulge is described by a Sersic function:

$$I_b(r_b) = I_{0b}\exp[-(r_b/h_b)^\beta],$$

Where I_{ob} is the central surface density of the bulge, h_b is the scale parameter of the bulge, and β=1/n. The half-light radius (effective radius), r_e, of the bulge is obtained by converting h_b,

$$r_e = (b_n)^n h_b$$

Where the value of b_n is a proportionality constant defined such that Γ(2n) = 2γ(2n,b_n). Γ and γ are the complete and incomplete gamma functions, respectively. We use the approximation $b_n \approx 2.17n_b - 0.355$ (Fisher and Drory 2010).

The bars and ovals (when present) are estimated by using a Ferrers or a Sersic function:

$$I_{bar}(r_{bar}) = I_{0bar}(1-(r_{bar}/a_{bar})^2)^{n_{bar}+0.5} \qquad , r_{bar} < a_{bar}$$

$$I_{bar}(r_{bar}) = 0 \qquad , r_{bar} > a_{bar}$$

Where I_{0bar} is the central surface brightness of the b_{ar}, a_{bar} is the bar major axis, and n_{bar} is the exponent of the bar model defining the shape of the bar radial profile.

The orientation parameters were estimated using Spitzer/IRAC 3.6-µm images of 53 galaxies with M_{BH} estimates. These images were used to measure the minor-to-major axis ratio (q = b/a), effective radii (Re), the radial profiles of the isophotal major-axis position angles (φ), and the estimated inclinations of the disk using the mean values in the outer parts of the disks (Laurikainen et al. 2005). We first removed foreground stars and masked out all point sources from the Spitzer

3.6-µm images by using SExtractor (Bertin and Arnouts 1996), then the surface brightness profiles were derived using the ELLIPSE routine in IRAF (Jedrzejewski 1987, Laurikainen et al. 2005).

Results and Discussion

Table 1 (see end of manuscript) lists the bulge stellar mass, spiral arm pitch angle, the SMBH masses, bulge dynamical mass, bulge stellar mass, and the kinetic energy of random motions of the dynamical and stellar bulge respectively.

From the virial theorem and the stellar mass-to-light ratios, we derived the dynamical bulge mass and stellar bulge mass respectively. Also, from the flux density, we have determined model-based bulge luminosities. Absolute magnitudes were calculated from apparent magnitudes using the distance moduli, and known redshifts.

In this paper, the relations that we studied can be written in the following forms:

$$\log_{10} M_{BH} = b + m\log_{10} x \quad (5)$$

$$\log_{10} M_{bul}\sigma^2 = b + m\log_{10} x \quad (6)$$

$$\log_{10} M_{bul} = b + m\log_{10} x \quad (7)$$

Where b and m are the intercept and the slope of the relation, x is a parameter of the bulge or spiral arm pitch angle.

Equations (5, 6, 7) can be used to predict the values of M_{BH}, $M_{bul}\sigma^2$, M_{bul} in other galaxies once we know the value of x. We have to perform an ordinary linear regression of M_{BH}, $M_{bul}\sigma^2$, M_{bul}, on x for the considered galaxies, for which we already know both the quantities.

Figures 1 and 2 show the SMBH masses as a function of $M_{dyn}\sigma^2$ and $M_*\sigma^2$, for 41 galaxies respectively. We found that the Pearson's linear correlation coefficients for a correlation between M_{BH}-$M_{dyn}\sigma_2$ and M_{BH}-$M_*\sigma^2$ relationship are 0.79, and 0.80 respectively, whereas the slopes of these relationships are 0.59, and 0.58 respectively. Thus, there is no significant difference between the M_{BH}-$M_{bul}\sigma^2$ relation and the M_{BH}-$M_{bul}\sigma^2$ relation.

The fitting results of M_{BH}-$M_{bul}\sigma^2$ correlations are presented in Table 3. Our work in this part, has confirmed the results of Feoli and Mele (2005,2007), Feoli and Mancini (2009), and Mancini and Feoli (2012) who also suggested the existence of a strong relationship between the masses of the SMBHs and the kinetic energy of random motions of its host spiral galaxies.

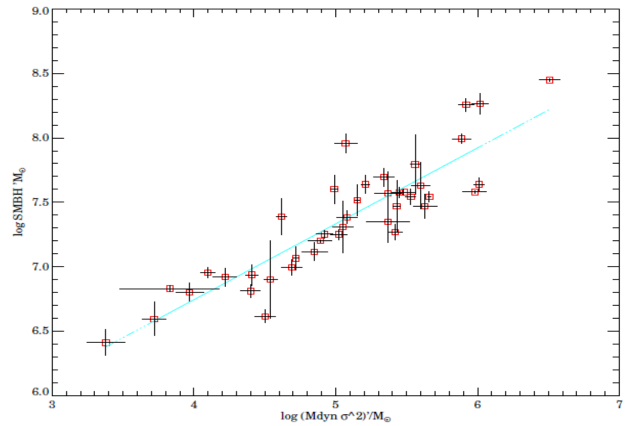

Figure 1. SMBH masses from (M_{BH}-σ) relation as a function of the $M_{dyn}\sigma^2$. The cyan solid line is the fit to all spiral galaxies.

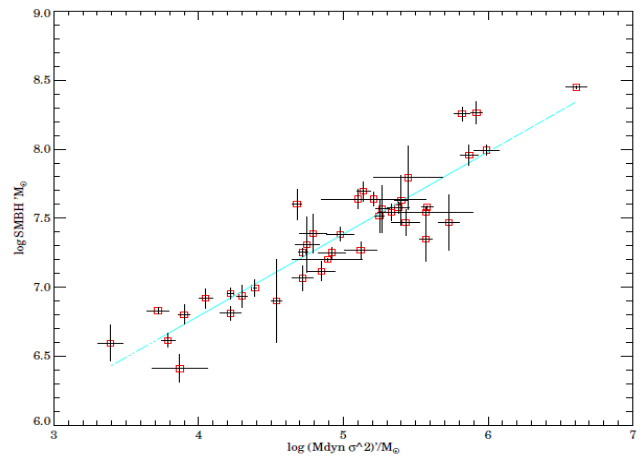

Figure 2. SMBH masses from (M_{BH}-σ) relation as a function of the $M_s\sigma^2$. The cyan solid line is the fit to all spiral galaxies.

Figure 3 presents the M_{BH}-P relation, where P is obtained by using a 2D Fast Fourier Transform decomposition technique (2DFFT). Using the M_{BH}-P relation to study SMBH masses, we can be fairly confident that for galaxies with bulges the pitch angle of the spiral arms should correlate well to the SMBH mass at center of the galaxies. The fitting result of M_{BH}-P correlation is presented in Table 3.

This relation is consistent with that presented in Seigar et al. (2008) and virtually identical in slope:

$$\mathrm{Log}_{10} M_{BH}=(8.44 \pm 0.1) - (0.07 \pm 0.005) P$$

We also compared our results with the previous work. Our correlation is consistent with that given by

Seigar et al (2008) for 41 spiral galaxies, but is larger than Berrier et al. (2013). It may be reflective of differences in the data used by Seigar et al. (2008) and Berrier et al. (2013). However, our results confirm the existence of a relationship between spiral arm pitch angle and SMBH mass as originally presented by Seigar et al. (2008) and Berrier et al. (2013).

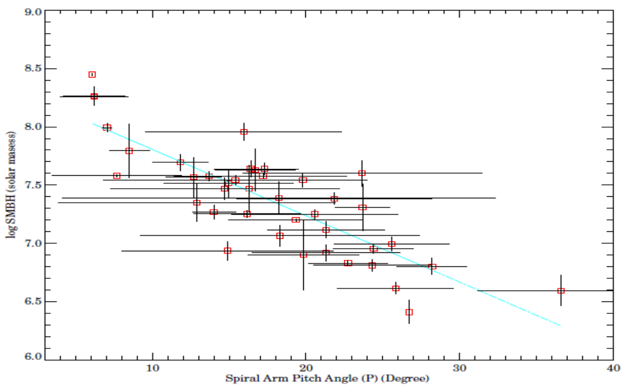

Figure 3. The SMBH mass from (M_{BH}-σ) relation as a function of the pitch angle of spiral arm (P). The cyan solid line is the fit to all spiral galaxies.

Figures 4 and 5 show the SMBH masses as a function of M_{dyn} and M_s for all of our spiral galaxy bulges, where the masses were obtained by using equations (1) and (2). The fitting results of M_{BH}-M_{bul} correlations are presented in Table 3.

From Figures 4 and 5, we can draw two conclusions: the best fitting line for M_{BH} - M_s and M_{BH} - M_d relations, which are shown in Tables 2 and 3. In these figures, containing data on galaxies with both classical bulges and pseudo-bulges, we note that galaxies with both types of bulges follow independent relations although some of the galaxies do harbor an intermediate bulge type, located between the relations of two type of bulge, and this reflects the mixed nature of their bulge properties. The different black hole-bulge relations obeyed by the two types of bulge are emphasized in Figures 4 and 5.

We found Pearson's linear correlation coefficients for a correlation between SMBH and M_{dyn}, M_* are 0.79, and 0.80 respectively, whereas the slope of the M_{BH} - M_d and M_{BH} − M_s relation are 0.76 and 1.01 respectively, which means there is a slight difference between values from both relations, because the difference in M_*/M_d ratio may be related to the mass contribution from the dark matter (Lauer et al. 2007b). In this work, we assumed that dynamical mass of bulges is dominated by the stellar mass, with a negligible contribution of dark matter and gas (Drory et al. 2004, Padmanabhan et al. 2004).

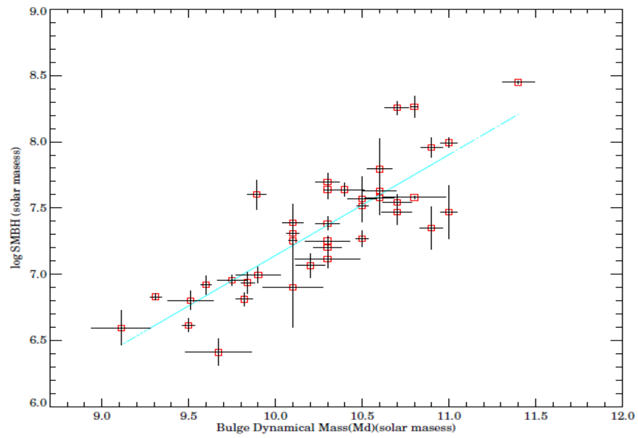

Figure 4. The SMBH mass from (M_{BH}-σ) relation as a function of the bulge dynamical mass. The cyan solid line is the fit to all spiral galaxies.

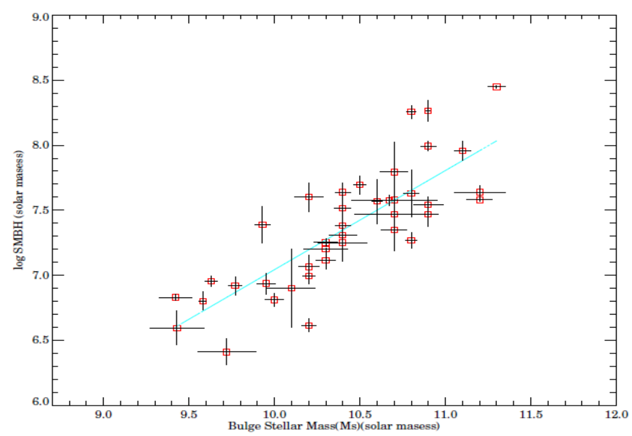

Figure 5. The SMBH mass from (M_{BH}-σ) relation as a function of the bulge stellar mass. The solid line is the fit to all spiral galaxies.

The fitting results are plotted in Figures 6 and 7, where we present the M_{dyn} - P and M_* - P relations for 41 spiral galaxies respectively. We found that M_{dyn} and M_* correlate well with P (we find a correlation coefficient of 0.74, and 0.77 with a significance of 99.99%, and 98.4% respectively).

This is a moderate correlation. The fitting results of M_{bul}– P correlations are presented in Table 3.

Recent studies have begun to discover the importance of the SMBHs in the evolution, or co-evolution, of their host galaxies (e.g., Magorrian et al. 1998, Gebhardt et al. 2000, Marconi and Hunt 2003, Springel et al. 2005, Hopkins et al. 2007, Rosario et al. 2010, Treuthardt et al. 2012).

Also, a recently discovered important relation between the spiral arm pitch angle of a galaxy and the SMBH mass, the M–P relation was presented by Seigar et al. (2008), whereas Feoli and Mancini (2009) found the relation between $M_{bul}\sigma^2$ and SMBH mass.

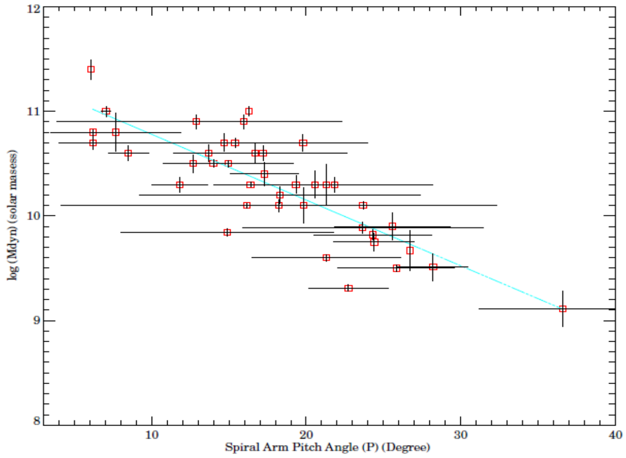

Figure 6. The bulge dynamical masses as a function the spiral arm pitch angle. The solid line is the fit to all spiral galaxies

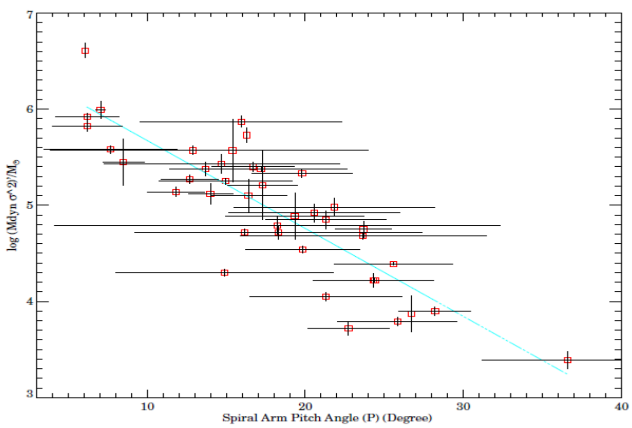

Figure 7. The bulge stellar masses as a function the spiral arm pitch angle. The solid line is the fit to all spiral galaxies.

In Figures 8 and 9 we show the bulge (dynamical and stellar) kinetic energy of random motions as a function of the spiral arm pitch angle for 41 spiral galaxies. $M_{dyn}\sigma^2$ and $M_*\sigma^2$ correlate with P (we find a correlation coefficient of 0.74, and 0.79 with a significance of 99.9%, and 99.7% respectively). It is evident that there is a moderate correlation relating $M_{bul}\sigma^2$ with P. The fitting results of M_{BH}-$M_{bul}\sigma^2$ correlations are presented in Table 3. In Table 4, we compare the fits of our relationship with the previous studies.

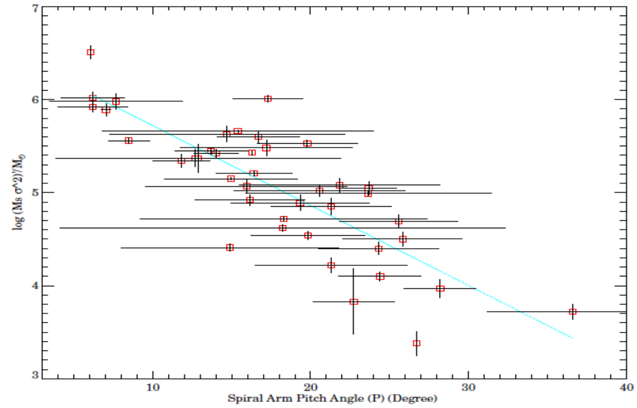

Figure 8. The kinetic energy of random motions for bulge dynamical mass as a function the spiral arm pitch angle. The solid line is the fit to all spiral galaxies

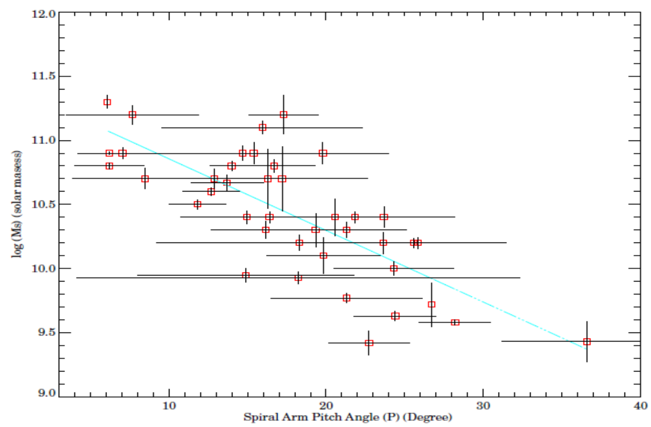

Figure 9. The kinetic energy of random motions for bulge dynamical mass as a function the spiral arm pitch angle. The solid line is the fit to all spiral galaxies.

Conclusion

In this study, we presented the bulge dynamical and stellar masses in 35 spiral galaxies, estimated by applying the isothermal model and the calibration by Oh et al. (2008) respectively. Furthermore, we found the kinetic energy of random motions of the corresponding host galaxies using $M_{dyn}\sigma^2$ and $M_{s*}\sigma^2$.

We have obtained the best-fit lines of four scaling relations. Among them, we found that M_{dyn} - P, M_* - P, $M_{dyn}\sigma^2$ - P, and $M_*\sigma^2$– P have a linear correlation coefficient 8.23, 7.56, 7.78, and 7.29 respectively. In other words, both the stellar and dynamical masses of bulges correlate well with spiral arm pitch angle. Furthermore, the kinetic energies of random motions in the bulge (whether determined from stellar or dynamical mass) correlates well with pitch angle too.

Hence, pitch angle is a good instrument to determine indirect measurements of the dynamical bulge mass, stellar bulge mass, and the kinetic energy (dynamical and stellar) of random motions in bulges.

Table 1. Estimated Galaxy Parameters.

Name (1)	Leda Type (2)	σ (km/sec) (3)	P (deg.) (4)	SMBH (M_{BH}-σ) (6)	$M_{dyn}(M_\odot)$ (7)	$M_s(M_\odot)$ (8)	$M_{dyn}\sigma^2$ (9)	$M_s\sigma^2$ (10)
Circinus	Sb	75 (1)	26.7	6.418±0.1	9.67±0.190	9.72±0.17	3.87±0.19	3.883±0.13
IC 2560	SBb	137 (1)	16.3	7.469±0.2	11±0.047	10.7±0.23	5.732±0.075	5.433±0.023
NGC 224	Sb	160±8 (2)	8.5±1.3	7.794±0.23	10.6±0.071	10.7±0.08	5.458±0.238	5.568±0.035
NGC 613	Sbc	125.3±18.9 (3)	23.68±1.77	7.309±0.2	10.1±0.035	10.4±0.08	4.755±0.083	5.055±0.071
NGC 1022	SBa	99 (4)	19.83±3.6	6.902±0.3	10.1±0.170	10.1±0.14	4.541±0.036	4.541±0.047
NGC 1068	Sb	151±7 (5)	17.3±2.2	7.639±0.05	10.4±0.11	11.2±0.15	5.217±0.36	6.017±0.035
NGC 1097	SBb	150 (6)	16.7±2.62	7.627±0.18	10.6±0.094	10.8±0.047	5.402±0.048	5.602±0.065
NGC 1300	Sbc	218±10 (7)	12.7±1.8	7.568±0.17	10.5±0.085	10.6±0.031	5.272±0.047	5.372±0.094
NGC 1350	Sab	120.91±2.08 (8)*	20.57±5.38	7.251±0.04	10.3±0.13	10.4±0.142	4.924±0.094	5.024±0.058
NGC 1353	Sb	83 (9)	36.6±5.4	6.594±0.13	9.11±0.73	9.43±0.057	3.394±0.085	3.728±0.083
NGC 1357	Sab	121±14 (10)	16.16±3.48	7.252±0.03	10.1±0.023	10.3±0.067	4.726±0.032	4.925±0.059
NGC 1365	Sb	151±20 (11)	15.4±2.4	7.639±0.07	10.3±0.025	10.4±0.045	5.105±0.17	5.217±0.027
NGC 1398	SBab	216±20 (12)	6.2±2	8.264±0.08	10.8±0.023	10.9±0.013	5.928±0.037	6.028±0.058
NGC 1433	SBab	84±9 (13)	25.82±3.79	6.615±0.05	9.5±0.034	10.2±0.043	3.798±0.046	4.508±0.07
NGC 1566	SABb	100±10 (14)	21.31±4.78	6.919±0.07	9.6±0.032	9.77±0.037	4.056±0.048	4.221±0.083
NGC 1672	Sb	130.8±2.09 (8)*	18.2±14.07	7.388±0.14	10.1±0.057	9.93±0.046	4.793±0.094	4.623±0.036
NGC 1808	Sa	148 (9)	23.65±7.77	7.601±0.11	9.89±0.053	10.2±0.083	4.23±0.035	4.991±0.025
NGC 2442	Sbc	140.74±2.18 (8)*	14.95±4.2	7.516±0.12	10.5±0.032	10.4±0.048	5.256±0.032	5.153±0.015
NGC 3031	Sab	143±7 (7)	15.4±8.6	7.544±0.04	10.7±0.046	10.9±0.085	5.576±0.328	5.664±0.01
NGC 3227	SABa	128±13 (7)	12.9±9	7.35±0.16	10.9±0.065	10.7±0.074	5.574±0.043	5.3744±0.15
NGC 3368	SABa	122±28 (7)	14±1.4	7.267±0.06	10.5±0.037	10.8±0.034	5.122±0.11	5.422±0.047
NGC 3511	SABc	93.56±2.04 (8)*	28.21±2.27	6.803±0.07	9.51±0.13	9.58±0.019	3.902±0.042	3.972±0.096
NGC 3521	SABb	130.5±7.1 (15)	21.86±6.34	7.384±0.05	10.3±0.071	10.4±0.045	4.981±0.094	5.081±0.073
NGC 3673	Sb	117.45±2.07 (8)*	19.34±4.38	7.2±0.011	10.3±0.083	10.3±0.13	4.899±0.240	4.899±0.084
NGC 3783	SBab	95±10 (16)	22.73±2.58	6.83±0.021	9.31±0.032	9.42±0.094	3.725±0.075	3.835±0.35
NGC 3887	Sbc	102.01±2.05 (8)*	24.4±2.6	6.954±0.04	9.75±0.084	9.63±0.038	4.227±0.023	4.107±0.051
NGC 4030	Sbc	122.43±2.1 (8)*	19.8±3.2	7.544±0.06	10.7±0.082	10.9±0.084	5.335±0.046	5.535±0.037
NGC 4151	SABa	156±8 (7)	11.8±1.8	7.696±0.07	10.3±0.071	10.5±0.036	5.146±0.048	5.346±0.072
NGC 4258	SABb	146±15 (7)	7.7±4.2	7.58±0.012	10.8±0.18	11.2±0.074	5.588±0.041	5.988±0.084

Table 1. Estimated Galaxy Parameters. *continued*

NGC 4462	SBab	146±8 (17)	17.2±5.42	7.579±0.02	10.6±0.074	10.7±0.25	5.388±0.026	5.485±0.081
NGC 4594	Sa	240±12 (7)	6.1	8.448±0.01	11.4±0.092	11.3±0,049	6.6104±0.07	6.5104±0.07
NGC 4699	SABb	215±10 (18)	6.2±2.2(1)	8.256±0.05	10.7±0.067	10.8±0.024	5.824±0.053	5.924±0.053
NGC 5054	Sbc	104.48±2.05 (8)*	25.57±3.73	6.996±0.06	9.9±0.13	10.2±0.036	4.398±0.012	4.698±0.071
NGC 5055	Sbc	101±5 (15)	14.9±6.9	6.937±0.08	9.84±0.037	9.95±0.054	4.308±0.036	4.418±0.043
NGC 6300	SBb	94±5 (3)	24.3±3.8	6.811±0.05	9.82±0.046	10±0.053	4.226±0.073	4.406±0.068
NGC 6744	SABb	112±25 (19)	21.28±3.8	7.117±0.07	10.3±0.19	10.3±0.059	4.858±0.093	4.858±0.091
NGC 6902	SBab	145.86±2.1 (8)*	13.71±2.3	7.578±0.04	10.6±0.084	10.67±0.05	5.387±0.073	5.457±0.035
NGC 7213	Sa	185±20 (17)	7.05±0.28	7.993±0.03	11±0.048	10.9±0.046	5.994±0.087	5.894±0.064
NGC 7531	SABb	108.7±5.6 (9)	18.31±9.09	7.065±0.09	10.2±0.083	10.2±0.059	4.722±0.072	4.752±0.021
NGC 7582	SBab	137±20 (7)	14.7±7.44	7.469±0.09	10.7±0.086	10.9±0.057	5.433±0.097	5.613±0.082
NGC 7727	SABa	181±10 (20)	15.94±6.39	7.955±0.07	10.9±0.064	11.1±0.049	5.875±0.058	6.075±0.079

Columns: **(1)** galaxy name. **(2)** Hubble type taken from the Hyper-Leda catalogue. **(3)** Velocity dispersion in km/s, Velocity dispersion references: (1) Hu 2009 (2) Lucey et al. 1997 (3) Beifior et al. 2009 (4) Garcia-Burillo et al. 2003 (5) Gültekin et al. 2009 (6) Davies 2009 (7) Sani 2011 (8) Ferrarese 2002 (9) Douglas 1995 (10) Lauer 2007 (11) Oliva 1995 (12) Whitmore 1985 (13) Buta 2011 (14) Nelson 1995 (15) Ho et al. 2009 (16) Greene et al. 2006 (17) Idiart et al. 1996 (18) Bower et al. 1993 (19) Benttoni et al. 1997 (20) Lake 1986. **(5)** Spiral arm pitch angle (P). Most of (P) taken from Berrier et al. (2013), and Davis et al. (2012). The spiral arm pitch angle given for M31, MW, and NGC 4945 are taken from Braun (1991), and Levine et al. (2006) respectively. **(6)** $\log(M_{BH}/M_{\odot})$ calculated by using M_{BH}-σ relation. (**7**) dynamical bulge mass. **(8)** Stellar bulge mass. **(9)** The kinetic energy for dynamical bulge mass ($M_{dyn}\sigma^2$). **(10)** the kinetic energy for stellar bulge mass ($M_{dyn}\sigma^2$).

Table 2. Regression results for log $M_\bullet = b + m \log x$ with the sample consisting of 41 spiral galaxies

Relation	b	m	r
M_{BH} - M_d	-0.46 ± 0.04	0.76 ±0.06	0.84, 100%
M_{BH} – M_s	-0.57 ± 0.07	0.76 ±0.09	0.81, 100%
M_{BH} - P	8.37 ± 0.65	-0.05 ± 0.004	-0.82, 99.25%
M_{BH} – $M_d\,\sigma^2$	4.41 ± 0.03	0.59 ± 0.05	0.87, 100%
M_{BH} – $M_s\sigma^2$	4.38 ± 0.04	0.58± 0.03	0.85, 100%
M_d - P	11.4 ± 0.15	-0.06 ± 0.005	-0.82, 99.24%
M_s - P	11.41 ± 0.32	-0.05 ± 0.002	0.75, 98.95%
$M_d\,\sigma^2$- P	6.59 ± 0.43	-0.09 ± 0.005	-0.77, 99.06
$M_s\,\sigma^2$-P	6.58 ± 0.049	-0.08 ± 0.007	0.72, 98.79%

Table 3. Scaling relation for log $M_{\bullet} = b + m \log x$ with the sample of 41 spiral galaxies

Relation	
M_{BH} - M_d	$\log_{10} M_{BH} = (-0.46\pm0.04) + (0.76 \pm 0.06) \log_{10} (M_{dyn})$
M_{BH} – M_s	$\log_{10} M_{BH} = (-0.57\pm0.07) + (0.76\pm0.09) \log_{10} (M_s)$
M_{BH} - P	$\log_{10} M_{BH} = (8.37\pm0.65) – (0.05 \pm 0.004)$ P
M_{BH} – $M_d\ \sigma^2$	$\log_{10} M_{BH} = (4.41\pm0.03) + (0.59 \pm0.05) \log_{10} (M_{dyn}\sigma^2)$
M_{BH} – $M_s\ \sigma^2$	$\log_{10} M_{BH} = (4.38\pm0.04) + (0.58\pm0.03) \log_{10} (M_s\ \sigma^2)$
M_d - P	$\log_{10} M_d = (11.4\pm0.15) – (0.06\pm0.005)$ P
M_s - P	$\log_{10} M_s = (11.41\pm0.32) – (0.05\pm0.002)$ P
$M_d\ \sigma^2$- P	$\log_{10} M_{dyn}\sigma^2 = (6.58\pm0.43) – (0.09\pm0.005)$ P
$M_s\ \sigma^2$-P	$\log_{10} M_s\sigma^2 = (16.13\pm0.43) – (0.08\pm0.007)$ P

Table 4. Comparisons with previous studies

Relation	a	b	r	References
M_{BH} - M_d	-1.64 ± 2.55 -9.01 ± 1.96 -1.05 ±2.00	0.87 ±0.25 1.58 ± 0.10 0.81 ± 0.2	0.68	Benedetto et al. 2013
M_{BH} - P	8.21 ± 0.16 8.44 ± 0.10	-0.062 ± 0.009 -0.076 ± 0.005	-0.81, 99.7% -0.91, 99.99%	Berrier et al. 2013 Seigar et al. 2008
M_{BH} – $M_d\ \sigma^2$	4.55 ± 0.8 2.36 ± 0.62 4.88 ± 0.56	0.75 ± 0.22 1.37 ± 0.17 0.66 ± 0.16	0.68	Benedetto et al. 2013

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Serendipitous Data Following a Severe Windstorm in an Old-Growth Pine Stand

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Running Title: Serendipitous Data Following a Severe Windstorm in an Old-Growth Pine Stand

Abstract

Reliable dimensional data for old-growth pine-dominated forests in the Gulf Coastal Plain of Arkansas are hard to find, but sometimes unfortunate circumstances provide good opportunities to acquire this information. On July 11, 2013, a severe thunderstorm with high winds struck the Levi Wilcoxon Demonstration Forest (LWDF) near Hamburg, Arkansas. This storm uprooted or snapped dozens of large pines and hardwoods and provided an opportunity to more closely inspect these rare specimens. For instance, the largest tree killed in this event, a loblolly pine (*Pinus taeda*), was 105 cm in diameter at breast height, 39.3 m tall, and if the tree had been sound would have yielded 3,803 board feet (Doyle log rule) of lumber. Gross board foot volume yield was also estimated from two other recently toppled large pines, an 85-cm-DBH loblolly and an 86-cm-DBH shortleaf pine (*Pinus echinata*), which tallied 2,430 and 2,312 board feet Doyle, respectively. A number of the other wind thrown pines on the LWDF were sound enough to count their rings for a reasonable (± 2-5 years) estimate of their ages. The stump of the fallen national champion shortleaf pine had 168 rings, and counts from other pines toppled by this storm had from 68 to 198 rings. We also searched for a new champion shortleaf pine using a LiDAR canopy height model of the LWDF to narrow our search. This preliminary assessment produced a number of targets that exceeded 40 m in height; further field checking of the tallest of these trees found that these were loblolly pines up to about 44 m. We eventually found shortleaf pines between 37 and 41 m tall, with diameters of up to 85 cm, indicating that the LWDF could still contain the Arkansas state champion.

Introduction

The scarcity of forests that can be considered representative of "virgin" timber limits our ability to get many desirable kinds of quantitative data, such as stand density, maximum tree size, age class distributions, and species composition. Hence, evidence adapted from old sources is an important supplement for researchers interested in restoring stands using historical forests as a guide. However, historical documentation presents a number of challenges to its application, many of which have been described elsewhere (e.g., Egan and Howell 2001, Bragg 2004b), including the difficulty of confirming the validity of the data. For example, it was common practice for people to write the board foot lumber volume of felled trees or logs on old photographs (Figure 1). Using the men in this picture for scale can help evaluate the lumber volume written on this photograph, but it is not possible to confirm the value given because of insufficient information on the length and diameter of this log.

Figure 1. A pine log from Ashley County, Arkansas, with the quantity of lumber estimated to be sawn from this log written on the photograph (1,684 board feet). Copy of a historical postcard courtesy of the Crossett Public Library.

While it is unlikely researchers will be able to unequivocally prove the claims of most of these unscientific documents, it may be possible to find contemporary trees that could confirm or refute the information presented. It is therefore critical to take advantage of every opportunity to collect such data in modern-day forests, especially given the rapid degradation of the resource due to management practices and biological processes. One such opportunity arose recently at the Levi Wilcoxon Demonstration Forest (LWDF) in Ashley County, Arkansas. The LWDF is a small remnant stand of pine-dominated old-growth that has been studied in recent years, both before and after a recent restoration thinning conducted by the current landowner (Bragg 2004a, 2006, 2010). On July 11, 2013, a severe thunderstorm with high winds struck the LWDF, uprooting or snapping dozens of large pines and hardwoods, including the national champion shortleaf pine (*Pinus echinata*). Though the loss of these big trees was unfortunate, it allowed us to more closely inspect these unique specimens.

Methods

Site description

The ~60 ha LWDF is located ~6 km south of Hamburg, Arkansas. This stand has been described in detail in previous research (e.g., Bragg 2004a, 2006, 2010), so only a brief description will be included in this paper. Following a restoration harvest in 2009-2010, the LWDF's overstory basal area is now over 83% pine, primarily loblolly (*Pinus taeda*), with a prominent shortleaf pine component (Bragg 2010). The LWDF is dissected by a number of small ephemeral streams. The gently (<2% slopes) rolling Calloway and Grenada silt loam (Glossic Fragiudalfs) soils found on the LWDF are seasonably wet. Locally, the annual precipitation averages about 140 cm and there are 200 to 225 frost-free days (Gill et al. 1979). The LWDF was protected as an informal "natural area" by the Crossett Lumber Company in 1939 (Anonymous 1948). Over the intervening decades, the only consistent management treatments conducted in this stand have been the occasional salvage of dead or dying pines (Bragg 2004a, 2006).

The windstorm that damaged the LWDF in July 2013 was a small, localized event that primarily affected the southeastern portion of this stand, with some additional damage near the parking lot and picnic tables just north of the juncture of Highways 425 and 52. We did not attempt to document all felled trees from this event; rather, we identified a non-random subset of the toppled pines for further description (see next sections for details).

Board foot lumber estimation

One goal of this effort was to determine if the lumber estimates found in historical photographs are reasonable approximations or gross exaggerations. In the days following the storm, a field crew from the U.S. Forest Service visited the LWDF to scale the board foot lumber volume of three very large pines (two loblolly, one shortleaf) that had fallen to the ground. Starting at "stump height" (approximately 30 cm above the former ground line), we measured outside-bark diameter (DOB) every 1.22 m along the merchantable portion of the bole across two axes using a large set of calipers—these values were then averaged to produce a mean DOB for that segment. We also cut into the bark at each location to estimate its thickness at that point, which was then subtracted to produce the inside-bark diameter (DIB).[1]

The fallen pines were then apportioned into 3.7 m to 4.9 m sawlogs[2] until their stems got too branchy for utilization (historically, lumber operations did not utilize the entire tree, but only took sawlogs to the point that removing the limbs with hand tools became too time consuming or unwieldy). Log volume estimates were adapted from Table 7 in Mesavage and Girard (1946, pgs. 15-16) for 4.9-m-long logs, using the smallest DIB from the two cut ends of the log. This table applies the Doyle log rule, which was one of the most commonly applied log scaling rules for this part of the United States well into the 20th Century (Freese 1974). Because lumber yield (English units) has no direct conversion to metric cubic volume measures (e.g., Fonseca 2005), log volume estimates in this paper have been reported in terms of board feet (Doyle log rule).

Pine age estimation

Following salvage operations (which commenced within weeks of the storm event), we returned to the LWDF to count the rings on any pine stumps that were sufficiently sound. Rings were tallied for two different radii of each stump; the values were then averaged and rounded off to the nearest ring. Although loblolly and shortleaf pine have prominent annual growth rings, they may have false or missing rings that can affect aging of trees and must be corrected with cross-dating to produce a date of origin. However, we did not cross-date the rings; therefore, these estimates are probably within 2 to 5 years of true tree age.

Champion shortleaf pine search

In addition to the volume and age samples, we searched the LWDF to see if a replacement champion

Figure 2. A pre-storm aerial photograph of the LWDF showing the forest structure and extent of the search area (light gray line).

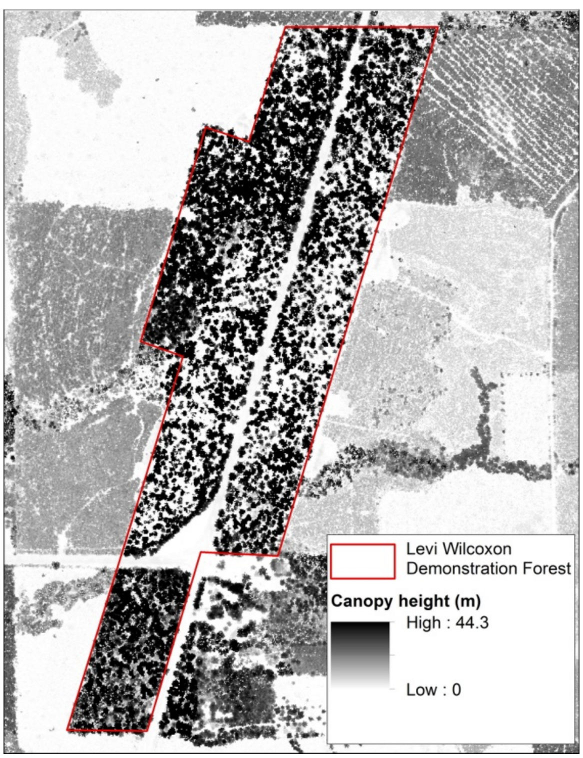

Figure 3. A LiDAR canopy height model of the LWDF with all heights shown.

shortleaf pine could be found. The search for a new champion was no small task—the forest area of the LWDF covers over 50 hectares with plenty of tall pines, including scores of shortleaf pine (Figure 2). To facilitate our search for a new champion, we obtained LiDAR data flown during winter 2011-2012 with average point spacing of 1.0 m through the USGS Earth Explorer (http://earthexplorer.usgs.gov/).

We then used Fusion software (McGaughey 2014) to produce a canopy height model (Figure 3) with 2-m pixels of LWDF and adjacent lands; this produced a map of the LWDF that could then be used as a guide to concentrate on areas with a higher probability of finding very tall trees. Previous experience in the LWDF suggested that shortleaf pine >38 m were present; we thus used this height threshold to classify favorable search locations (Figure 4).

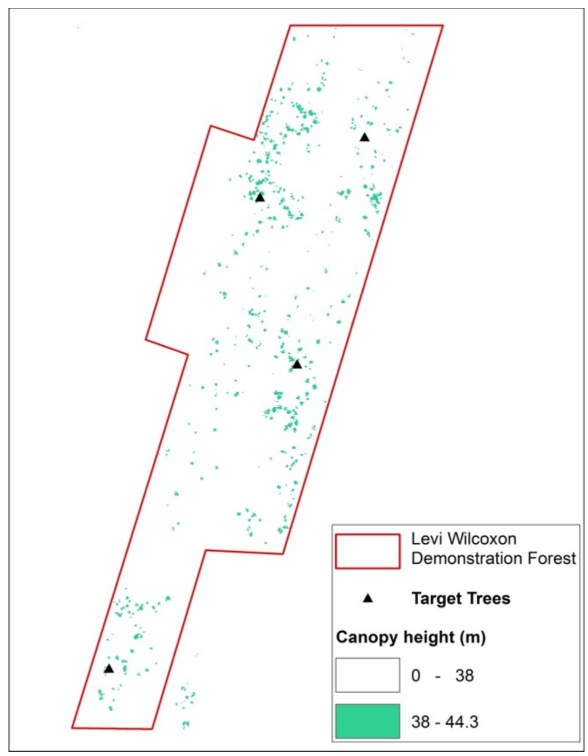

Figure 4. The LWDF LiDAR canopy height model with only heights greater than 38 m highlighted and the four target trees identified.

While it helped focus our search, LiDAR alone was insufficient for identifying champion trees for several reasons. First, LiDAR measures canopy height rather than tree height; these measures may differ where the ground is sloped and may not strike the highest point on the individual crowns (Kelly et al.

2010). Second, LiDAR does not provide any independent taxonomic information—if there is no clear stratification of the canopy by taxa, the remotely sensed data cannot distinguish tree species.

Third, in addition to total tree height (*HT*, in feet) the index used to determine champions (*AFBI*, American Forests 2014):

$$AFBI = CBH + HT + \frac{1}{4}\, CS \quad (1)$$

also incorporates crown spread (*CS*, in feet) and stem circumference at breast height (*CBH*, in inches), the latter of which cannot be measured with the requisite accuracy from remotely sensed data.

We transferred the spatial coordinates of the four tallest trees identified by the model to a Garmin eTrex GPS. In the field, we located these highest LiDAR hits and searched the surrounding areas for large shortleaf pines. We used either a TruPulse 200 (with built-in clinometer) or a Nikon Prostaff 440 laser rangefinder (with a separate Suunto clinometer) to measure heights of potential champion trees with the sine method (Bragg 2008). Diameter at breast height (DBH) was measured at 1.37 m above ground and then converted to circumference for CBH. Because of time constraints, we only measured crown spread (the average of the widest and narrowest spread of the live tree crown) for the five largest shortleaf pines and one post oak (*Quercus stellata*).

Results and Discussion

Evaluation of lumber volume

We examined the largest tree killed in the July 2013 windstorm, a loblolly pine 105 cm in DBH and 39.3 m tall, for its lumber yield. This specimen had a gross sawtimber yield of 3,803 board feet (Doyle log rule) of lumber in four 4.9-m-long sawlogs (which tallied 1,050, 961, 942, and 900 board feet, respectively). For perspective, a typical 38-cm-DBH pine with three 4.9-m sawlogs (more consistent with trees produced by modern-day plantations) would yield 121 board feet. It is important to note that the 3,803 board feet assumed the pine was sound (i.e., it did not lose volume due to decay and defect). This particular loblolly pine did have extensive butt rot, so its net yield would have been significantly lower (we did not determine net yield). The low bole taper of this loblolly pine is also apparent from the modest decrease in board foot volume in each sawlog—the smallest log is only about 14% less than the biggest.

The gross Doyle log scale results for the other two pines were noticeably lower but followed similar patterns. The 85-cm-DBH loblolly pine was 38.7 m tall before it fell; this specimen was estimated to yield 2,430 board feet from four 4.9-m and a single 3.7-m sawlog (610, 571, 511, 467, and 271 board feet, respectively). The 86-cm-DBH shortleaf pine was 39.6 m tall, and had an estimated 2,312 board feet in five 4.9-m sawlogs (655, 566, 441, 361, and 289 board feet, respectively).

All of these pines had additional log volume that was not included in this assessment because they would have had too many branches to have been utilized in historical lumbering operations. Though our results cannot confirm the accuracy of the stated tree volumes on any historical photographs (e.g., Figure 1), they do suggest that these claims are plausible. Sawtimber yields of the largest pines from the Upper West Gulf Coastal Plain of Arkansas, Louisiana, and Texas have been given in the historical literature between 7,000 and 11,000 board feet Doyle (e.g., Record 1910, Morbeck 1915, Chapman 1942, Bragg 2002). A sign on the 142-cm-DBH Morris Pine, the oldest and largest living loblolly pine in the LWDF, reports a volume of 5,000 board feet (Bragg 2002).

Although these values are substantially higher than our estimates, they also came from pines with much bigger boles that probably had more sawlogs. Loblolly pines exceeding 150 cm in DBH and over 45 m tall have been documented in this region and shortleaf pine greater than 100 cm in DBH and over 40 m tall are also possible (e.g., Mohr and Roth 1897, Chapman 1942, Bragg 2002); it is almost certain that these species probably exceeded even these values. Very large, columnar, branch-free boles helped to accentuate the sawtimber volume yield of the virgin timber. As an example, one such loblolly pine from central Louisiana that scaled over 10,000 board feet was 137 cm at DBH and 102 cm in diameter at 29.3 m above the stump (Chapman 1942).

Pine age estimates

The extensive basal bole decay (butt rot) found in the LWDF limited the number of pines that could have their age estimated via ring counts. However, enough sound trees were found to show a poor (but positive) relationship between stump diameter and estimated pine age (Table 1). The youngest pine (a loblolly) examined had 68 rings; the oldest (a shortleaf) yielded 198 rings, and the former national champion shortleaf was estimated to be 168 years old when it was killed in this storm (Table 1). The former national champion shortleaf pine happened to grow on a favorable site by

Table 1. Stump ring counts for pines killed by the July 2013 windstorm at the LWDF.

	------ Average stump ------	
Species	diameter (m)	ring count
Shortleaf pine	1.00	198
Shortleaf pine	1.12	168*
Shortleaf pine	0.72	160
Shortleaf pine	0.81	148
Shortleaf pine	0.79	147
Shortleaf pine	0.87	144
Shortleaf pine	0.62	139
Shortleaf pine	0.56	133
Shortleaf pine	0.77	126
Shortleaf pine	0.77	108
Shortleaf pine	0.65	89
Shortleaf pine	0.52	81
Loblolly pine	0.70	186
Loblolly pine	1.16	160
Loblolly pine	0.72	134
Loblolly pine	0.68	116
Loblolly pine	0.52	68

* Former national champion shortleaf pine.

a small ephemeral stream, which probably accounts for its larger size and relatively fast growth.

The limited age data available for the LWDF from past research (e.g., Bragg 2004a, Bragg 2006, Bragg 2010) found similar spans of ring counts—between 50 and 170 for dominant and codominant pines. Bragg (2004a) suggested that some of the standing live loblolly and shortleaf pines that either yielded incomplete cores or were too decayed to even attempt to core were 200 years of age, and that the oldest loblolly pine on the LWDF, the Morris Pine, probably exceeded 300 years. The presence of a 186-ring loblolly and 198-ring shortleaf pine in the current sample (Table 1) support these assertions. We did not examine any of the windthrown hardwoods following this storm event for their ages; it is expected from earlier work (Bragg 2010) that the larger hardwoods in the LWDF are about as old as the dominant pines.

It is important to note that none of these samples were randomly chosen and, hence, these should not be construed as representative of the LWDF's actual age class structure. However, the limited information available continues to suggest that the lack of discrete age cohorts and the wide span of the ring counts support the hypothesis that the virgin pine forests in this part of the Arkansas Gulf Coastal Plain were largely uneven-aged, with the notable exception of areas struck by catastrophic disturbances such as fires or tornadoes (Chapman 1912, Forbes and Stuart 1930, Turner 1935, Bragg 2002). Severe wind events such as the July 2013 storm and a similar May 2003 storm that occurred in a different part of the LWDF (Bragg 2004a) impact relatively small patches and often leave individual pines or small groups of pines largely unscathed. Such heterogeneity helped to structure the virgin pine forests of the region (Chapman 1912, Bragg 2002), particularly when coupled with other natural processes such as fire and insect-related pine mortality.

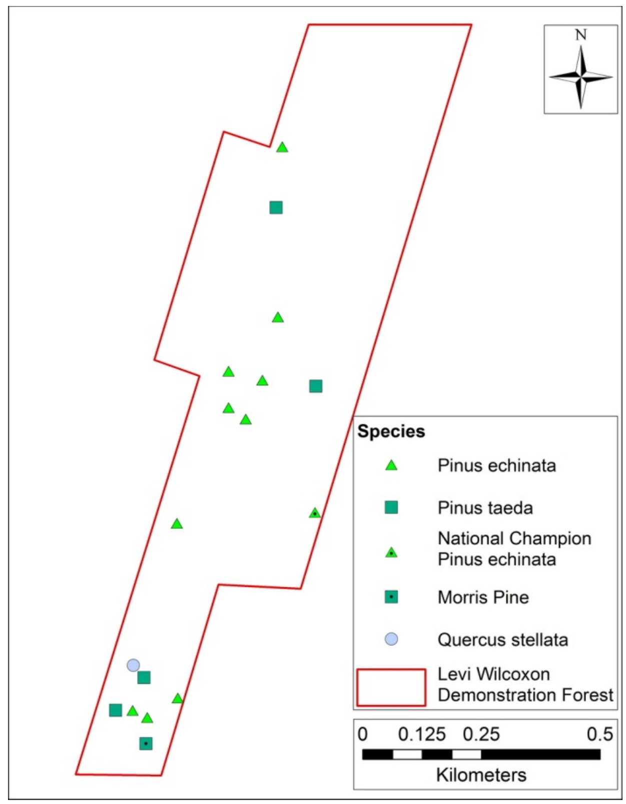

Figure 5. Locations of the field-measured tall trees from the LWDF reported in Table 2. This map includes the locations of the Morris Pine and the former national champion shortleaf pine.

LiDAR search for a new champion shortleaf pine

According to the LiDAR canopy height model, much of the LWDF has trees that exceed 38 m in height (Figure 3). Hence, our search for a new shortleaf pine champion concentrated on distinct parts of the stand surrounding four target trees, three in areas of generally high canopy and an isolated tall specimen tree (Figure 4). All four target trees proved to be loblolly pines, which we measured on-site using laser

Table 2. Tree size measurements taken at the LWDF while searching for a new champion shortleaf pine; columns with English units provided because this is how the AFBI is calculated.

Common name	Height (m)	DBH (cm)	Crown spread (m)	Height (ft)	CBH (in)	Crown spread (ft)	AFBI[1]
Loblolly pine	44.1	98.4	--[2]	144.7	121.7	--[2]	--[2]
Loblolly pine	43.1	101.5	--	141.5	125.5	--	--
Loblolly pine	43.0	91.3	--	141.0	112.9	--	--
Loblolly pine	42.5	94.3	--	139.3	116.6	--	--
Shortleaf pine	40.9	68.5	--	134.1	84.7	--	--
Shortleaf pine	40.5	76.7	12.9	133.0	94.9	42.2	238
Shortleaf pine	40.2	78.3	11.4	132.0	96.8	37.5	238
Shortleaf pine	39.9	77.9	11.8	131.0	96.4	38.8	237
Shortleaf pine	39.6	67.2	--	130.0	83.1	--	--
Shortleaf pine	39.6	78.0	12.2	130.0	96.5	40.1	236
Shortleaf pine	39.3	69.7	--	129.0	86.2	--	--
Shortleaf pine	37.8	85.3	13.4	124.0	105.5	44.1	241
Shortleaf pine	37.8	75.2	--	124.0	93.0	--	--
Shortleaf pine	36.7	80.2	--	120.3	99.2	--	--
Post oak	34.2	68.7	11.8	112.3	85.0	38.6	207

[1] AFBI = American Forests bigness index (American Forests 2014) = total tree height (in feet) + stem circumference (in inches) at 1.37 m above groundline (CBH) + ¼ crown spread (in feet).

[2] Crown spread was measured only on the 5 biggest shortleaf pines and the post oak; AFBI is therefore only calculated for these 6 trees.

rangefinders as 42.5 to 44.1 m tall (Table 2). Under most circumstances, loblolly is larger in girth and taller than shortleaf pine (Baker and Langdon 1990, Lawson 1990), so this result was not surprising. Loblolly pines over 42 m are exceptional for upland sites in southern Arkansas, but not nearly the tallest recorded; this species has been documented to exceed 52 m on large river bottomlands in the eastern part of its range (Native Tree Society 2009).

After confirming that the tallest LiDAR returns were all loblolly pines, we then searched other parts of the stand for big shortleaf pines. The removal of most of the hardwood midstory during 2009-2010 greatly facilitated our field-based search by making crowns more visible and easier to measure. Dozens of shortleaf pines were examined for their potential champion status; Table 2 provides the 10 most notable specimens (these, as well as the four large loblolly pine targets and the large post oak, can be found in Figure 5). These shortleaf ranged in height from 36.7 to 40.9 m; DBHs ranged from 68.5 to 85.3 cm; and crown spreads ranged from 11.4 to 13.4 m. Under national (and most state) champion lists using AFBI points, trees within five points of each other qualify as co-champions, and the five largest shortleaf pines fell within the 236 to 241 point range. Though impressive, none of these shortleaf reached the stature of the former champion, which measured 91.4 cm DBH (or 287 cm [113 inches] CBH), 41.5 m (136 ft) tall, with a 15.2 m (50 ft) crown spread and produced a AFBI score of 262 points when nominated in 2006 (American Forests 2014).

Even though most of the overstory hardwoods at the LWDF were removed in a restoration harvest conducted several years ago (Bragg 2010), a number were retained throughout the stand. These include some of considerable size, including one post oak we measured at 34.2 m tall and 68.7 cm DBH, with an average crown spread of 11.8 m (a total of 207 AFBI points; Table 2). The currently listed Arkansas state champion post oak is 31.1 m tall, with a 147.1 cm DBH and an average crown spread of 31.7 m (310 AFBI points). Large forest-grown specimens such as the post oak measured on the LWDF often fail to make champion lists because they tend to be tall but with less bole girth and (typically) much narrower crowns than trees growing in the open.

Conclusions

Our results indicate that many historical sources of tree dimensions in the pine-dominated forests of southern Arkansas are reasonable in their claims. For example, based on his observations of the virgin forest, Mattoon (1915) had placed the maximum height threshold for old shortleaf pine at just under 40 m with diameters of 60 to 90 cm and ages of 200 to 300 years as being "common"; the evidence from the LWDF suggests that these are acceptable restoration targets for most sites in southern Arkansas. This is encouraging because we are rapidly running out of examples of very large trees in today's highly modified landscapes. The loss of mature, pine-dominated forests of natural origin across the southeastern United States is a major conservation concern. In particular, the decline of shortleaf pine across the coastal plain, including that in southern Arkansas, presents a challenge for our understanding of the mechanism(s) behind this change, as well as reasonable measures for successful restoration efforts.

We believe the outcomes reported in this paper speak to the need for researchers to closely monitor any remnant tracts of old-growth timber for similar opportunities to quantify the structural and composition attributes of these stands. Many of these remnants are understandably protected to a degree that limits the ability of scientists to gather certain types of information—their scarcity supports extra caution to minimize any threats to their health and integrity. The deaths of these dwindling examples of large, old loblolly and shortleaf pines in the Upper West Gulf Coastal Plain is an unfortunate loss that can be somewhat offset by capturing whatever information we can from these trees before it is lost to decay or salvage.

Acknowledgments

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Endnotes

[1] Bark thicknesses ranged from 0.25 to 1.8 cm, depending on the location on the bole. Bark is thicker nearer the lowest portion of the bole, and thinner further up the stem.

[2] Sawlog lengths are another unique attribute of historic lumber information; hence, the rather curious metric lengths for some logs. For instance, a 3.7-m log is 12 feet long, and a 4.9-m log is 16 feet long.

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Synchronization Limits of Chaotic Circuits

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Running Title: Synchronized Limits of Chaotic Circuits

Abstract

Through system modeling with electronic circuits, two circuits were constructed that exhibit chaos over a wide ranges of initial conditions. The two circuits were one that modeled an algebraically simple "jerk" function and a resistor-inductor-diode (RLD) circuit where the diode was reverse-biased on the positive voltage cycle of the alternating current source. Using simulation data from other experiments, the waveforms, bifurcation plots, and phase space plots of the concrete circuit were verified. Identical circuits were then built containing variable components and coupled to their original, matching circuits. The variable components were used to observe a wide range of conditions to establish the desynchronization parameters and the range of synchronization.

Introduction

History

Ever since the conception of chaos became a subject to be studied, the list of systems that can be modeled by chaotic equations has been growing. Many of these systems are of great importance, featuring such scientific irritations as the weather, noise, and the precise movement of fluids. While chaos cannot be defined by specific sets of equations as waves can be, the study of chaos has revealed several characteristics of chaotic systems that help to define the term and show its potential for engineering applications.

The most obvious trait of chaos is its aperiodic behavior, never repeating a solution. With his computer-made weather model and simulator based on iterative mapping in the 1960's, Edward Lorenz stumbled upon another important feature of chaos, *sensitive dependence on initial conditions.* Due to computer rounding, Lorenz found that different initial conditions that vary by only a slight decimal difference will result in drastically different outcomes after only a few iterations. Lorenz also discovered that for some initial conditions the solutions never repeated (indicating chaos) but they did tend to be similar taking advantage of the universe that lies between 1 and 0. That is to say, for some initial conditions, the solutions would be incredibly close to each other but off by miniscule decimal places when evaluated quantitatively. When the solutions were plotted, they displayed a shape that became known as the Lorenz attractor. Since then, many other systems have been found with unique attractors of a variety of shapes (reviewed in Gleick 1987). Later in the 1970's, Mitchell Feigenbaum studied iterative mapping of several non-linear equations using a wide range of initial conditions (reviewed in Gleick 1987). He found initial conditions that when mapped converged on a single solution, and that when the value of the initial conditions are increased to a certain point, the mapping converges to two solutions. As the initial conditions are increased the number of solutions continues to double (now called *period-doubling,* or *bifurcation*) until the solutions diverge in chaos. The period-doubling alone is remarkably useful for identifying chaos, but Feigenbaum also discovered a chaotically, universal constant (reviewed in Gleick 1987). Through the study of numerous non-linear systems, Feigenbaum found that the range of initial values that yield a specific number of solutions compared to the range of the next period-doubling converges on the number 4.67. The mathematical statement for this idea is,

$$\frac{f_2 - f_1}{f_3 - f_2} = 4.67 \qquad (1)$$

Where f is a bifurcation point. This constant holds true for all systems that approach chaos through period-doubling. The universality of this number allows for predictions of period-doublings and another way to verify chaos within a system (reviewed in Gleick 1987).

More recently, J.C. Sprott (2011) discovered several simple functions (simple meaning they contain few terms) that still exhibit chaos. Equations containing a third order differential (*Jerk functions*) can achieve chaos with only one non-linear term. Having only one non-linear term allows a function to be easily modeled with electrical components where the signal can be viewed on an oscilloscope so that measurements such as chaotic and periodic ranges can be made. Sprott analyzed several of these functions by modeling them with circuits, however, we now report the analysis of a physical circuit that Sprott has only measured through computer simulation (Sprott 2011).

Theory

The simplicities of the Sprott circuit and the resistor-inductor-diode (RLD) circuit are useful for producing the same exact signal in two nearly identical circuits, but trying to create the exact chaotic signal in two separate circuits is a rather difficult task due to the sensitive dependence on initial conditions described earlier. It is difficult to control every possible initial condition in a real world system, but through synchronization, exact replication of chaotic signal is possible. *Synchronization* is the process of allowing one circuit to drive another circuit through circuit coupling, and when used with chaotic waveforms, the use of synchronization is very powerful. In synchronization, the secondary circuit is driven so that the exact signal in the primary circuit appears in the secondary. Even with differing initial conditions, the primary and secondary circuits can still exhibit identical behavior provided that the initial conditions are similar. The nonspecific term “similar” is used because the question addressed in this manuscript is to define how "similar" the two circuits must be.

The driving force behind the design of our experiment is the use of synchronization for noise cancellation. From a practical sense, the noise appearing in a machine will not be the same every time it is used, and more realistically will vary depending on the settings of the machine and how it is used. To meet the demands of a wide range of chaotic possibilities, the cancellation circuit needs to be robust, requiring a wide range of parameters over which it is chaotic. “Jerk” functions have been shown numerically to be very robust, and these third order differentials can exhibit chaos with minimal terms making them algebraically simple. For instance, the following functions achieve chaos with only two non-linear terms and four total terms,

$$\dddot{x} + Ax\ddot{x} - (\dot{x})^2 + x = 0 \qquad (2)$$

$$\dddot{x} + Ax\ddot{x} - x\dot{x} + x = 0 \qquad (3)$$

The simplistic functions allow for better predictions of what the waveform might do and are easy to model with electrical components in a circuit. Simulations done have shown that chaotic jerk functions are very robust (Sprott 2011). In this experiment, a circuit was constructed to model the equation,

$$\dddot{x} + A\ddot{x} + x + (\dot{x})^2 = 0 \qquad (4)$$

The value of the parameter A changes the initial conditions allowing for bifurcations and chaos to be observed and evaluated. It will be shown later that A can be controlled with a potentiometer (Sprott 2011). The equation of an RLD circuit also contains a bifurcation parameter but this time it is controlled by varying the amplitude of the voltage source. The amplitude parameter can easily be seen in the following equation found with Kirchoff’s voltage loop rule (Hammill 1993).

$$\mathrm{A}e^{j(\omega t-\varphi)} - i(t)R + L\frac{di}{dt} - nV_T\ln\left(\frac{i(t)}{Is} + 1\right) = 0 \qquad (5)$$

Here $Ae^{j(\omega t-\varphi)}$ represents the oscillating voltage source with a controllable amplitude, $L\frac{di}{dt}$ is the voltage drop across the inductor, $i(t)R$ is the voltage drop across the resistor as the current changes with the voltage, and $nV_T ln(\frac{I}{Is} + 1)$ is the voltage drop across the diode according to Shockley’s theorem. However, Shockley’s theorem does not include the capacitive effects of the diode at high frequencies. V_T is the thermal voltage characteristic of the diode, I_s is the reverse bias saturation current, and n is another characteristic of the diode called the ideality factor. This is a much more complex equation but the circuit is much simpler and easier to construct. Easier construction reduces build time and simplifies the synchronization process (Hammill 1993).

Synchronization

Mathematically, synchronized systems can be defined as a situation where one system determines the behavior of another.

$$f(x) = x + \dot{x} \qquad (6)$$

$$g(x,y) = (\mu x + (1-\mu)y) + (\mu\dot{x} + (1-\mu)\dot{y}) \qquad (7)$$

Here, μ represents the amount of the output that is governed by x and $1 - \mu$ is the remainder that is governed by y. Thus $0 \leq \mu \leq 1$ as it is a fraction of the whole. When $\mu = 0$, $g(x,y)$ is no longer dependent on $f(x)$ making the two uncoupled with a loss in synchronization.

Materials and Methods

A jerk circuit was constructed to model Equation (4) using three integrating sub-circuits to produce the third order differential, and an inverting sub-circuit to bring about a positive first order term. The circuit schematic is shown below in Figure 1. The diode was responsible for the non-linear term. The circuit was constructed using 5% tolerance 1 kΩ resistors and 1.0 µF capacitors, OP27 amplifiers, and a 1N4001 silicon rectifier diode on a standard prototyping board (breadboard as they're colloquially known). The waveforms were observed in an uncoupled jerk circuit using an Agilent Technologies DSO1002A digital oscilloscope, and the bifurcations were measured by substituting R^* with a 10 kΩ potentiometer. The bifurcations were measured by steadily increasing the potentiometer while watching the oscilloscope for period doubling at the positive first differential due to the clarity of the doublings at this point. The potentiometer was then switched to a 1 MΩ potentiometer to examine the higher values of the parameter with decreased accuracy.

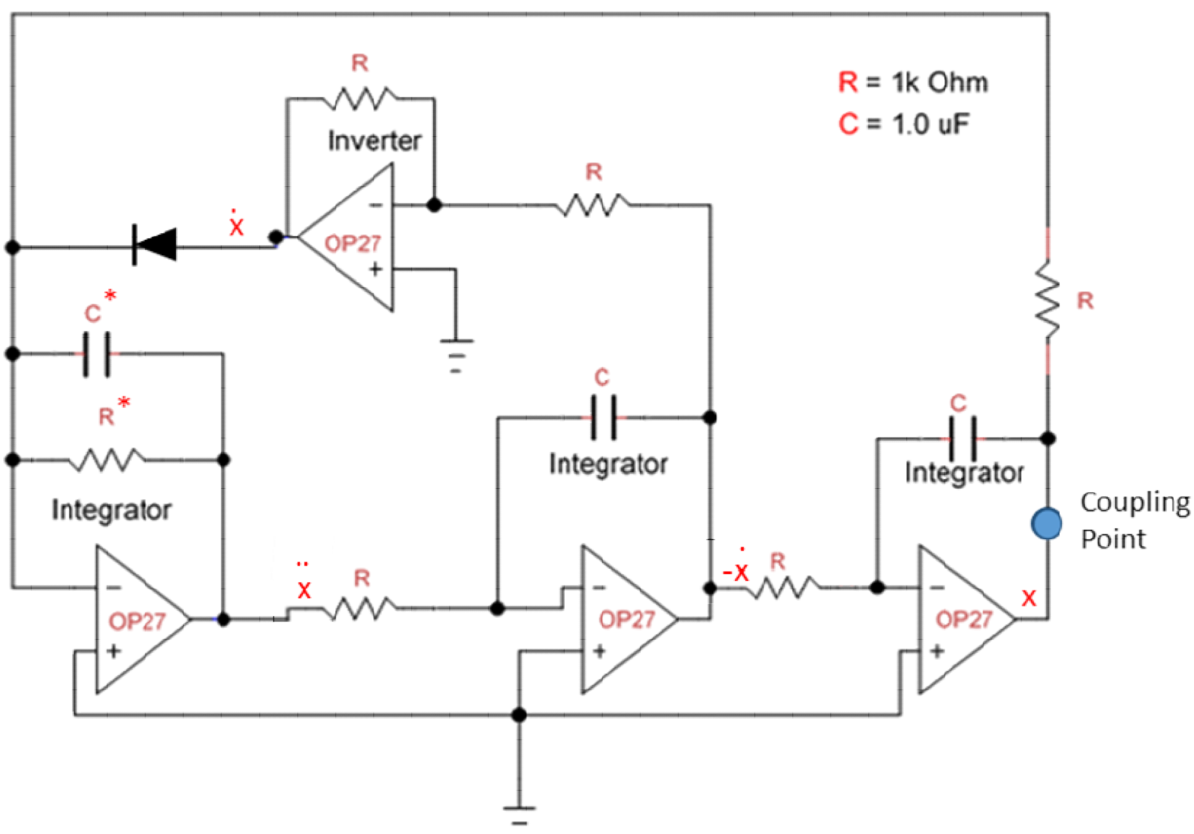

Figure 1. Chaotic circuit modeling a jerk function. It contains three integrating sub-circuits (to create the "jerk" term) and an inverting sub-circuit.

A fixed-value RLD circuit is shown in Figure 2 and was constructed and hardwired to a circuit board with a 5% tolerance 1 Ω resistor, a 1N4001 diode, and a BK Precision 2 MHz signal generator. The inductor for the fixed value circuit was a single 1 mH inductor. In this circuit, bifurcations were found by increasing the voltage output of the signal generators. It is also worth noting that the resistance of the signal generator is 50 ohms and the internal resistance of the inductor is likely and order of magnitude higher than the resistor.

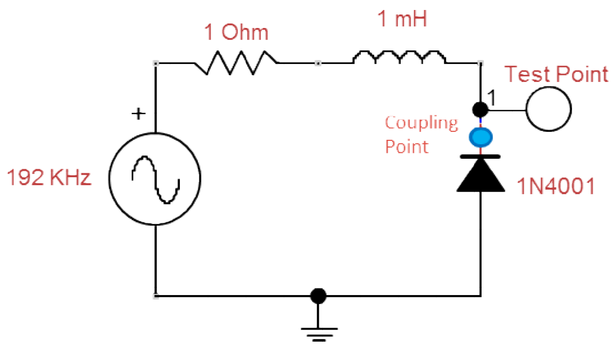

Figure 2. RLD circuit that exhibits chaos when the diode is reverse-biased

After bifurcations were recorded and waveforms observed for each circuit, similar circuits with variable components were built for synchronization purposes. Another Sprott jerk circuit was built with all fixed components except for a 100 kΩ potentiometer at $R*$. The two jerk circuits were then coupled with an ordinary jumper wire at the x signal points of each circuit, and the second differential waveform of each circuit was probed by the digital oscilloscope. The oscilloscope displayed the waveforms from each circuit as well as the waveform produced by subtracting the fixed circuit waveform from the variable circuit waveform.

The synchronized waveform in Figure 3 has a line in the middle that is the subtraction voltage of the variable circuit voltage from the fixed circuit voltage. The subtraction waveform is not shown in the desynchronized figure because its presence makes the figure very confusing. However, the subtraction waveform was still used when the circuit was desynchronized. The subtraction waveform allowed for desynchronization to be observed easily (Figure 3) due to the abruptness of desynchronization. The desynchronization of two circuits is a rapid event that occurs in a matter of a couple of ohms making it possible to record the synchronization limits with little uncertainty (±5 ohms) when watching an oscilloscope. The 100 kΩ potentiometer was positioned around 1 kΩ and gently increased until either desynchronization or loss of chaos occurred. The value of the potentiometer was then measured using a Fluke multi-meter. The same was done for decreasing the potentiometer from 1 kΩ. The potentiometer again was set to 1 kΩ and the value of $C*$ was increased by adding more capacitors in parallel with the initial 1.0 μF capacitor including a variable capacitor that allowed for more precise measurements of the desynchronization or loss of chaos parameters. The lower limit was found by adding capacitors in series.

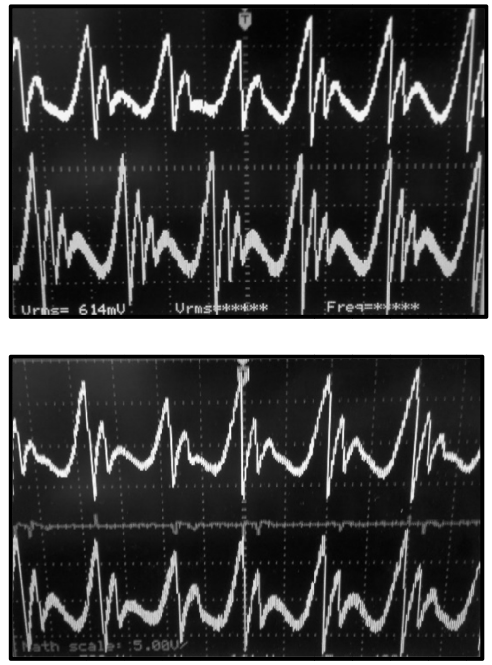

Figure 3. Observed jerk circuit waveforms of desynchronized (above) and synchronized (below)

Next, a similar RLD circuit was built with variable components including, a 10k potentiometer. The inductor for the variable circuit was an array of 8.0 mH inductors added in parallel to achieve 0.998 mH. The oscilloscope probe was attached just before the diode for measurements, and the signal generators were both set to 192 kHz to more accurately match the waveforms seen in previous works.

To synchronize the RLD circuits, the two circuits were coupled by a jumper wire at the same points where the probes were connected. Examples of the synchronized and desynchronized waveforms are shown in Figure 4. The voltages of the generators were varied with respect to one another to find upper and lower synchronization limits and a potentiometer in the variable circuit was used to find the synchronization limits when the resistance is varied. The array of inductors was also varied.

Results

Both the RLD waveforms and the jerk circuit waveforms shown in Figures 5 and 6 are appropriately scaled to match the simulation data on both time and

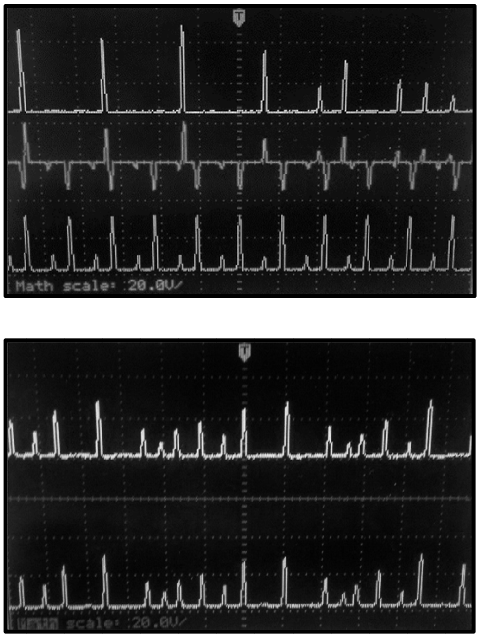

Figure 4. Observed RLD waveforms and subtraction (middle line) of synchronized (top) and desynchronized (bottom) circuits

voltage. The observed waveforms are very similar to those seen in previous works (Sprott 2011, Hammill 1993). While bifurcations were easily observed, there was some uncertainty when defining the instant that a bifurcation occurred. Bifurcations were recorded for the parameter value when the new waveform dominated with little to none of the previous waveform being visible. The sparse amount of data points for the jerk circuit was due to the circuit achieving chaos after three bifurcations. The RLD also produced only three data points due to the bifurcations becoming too minute to observe. It is still useful to see that the ratios from equation (1) are within the range of the Feigenbaum constant with the uncertainty accounted for. The bifurcations and their ratios are in Table 1 below.

The attractor for the Sprott circuit was observed on an analog oscilloscope (Figure 7) by putting the original signal on the x axis and the first derivative on the y axis because it had more time divisions allowing for a better view of the attractor. This observed attractor is very similar to the one found by Sprott (Sprott 2011). The attractor for the RLD circuit was found using the digital oscilloscope (Figure 7) with the signal from coupling point put on the x axis and signal from the signal generator placed on the y axis and appears to provide further evidence of chaos based on the very similar patterns that never repeat.

The desynchronization parameters in Table 1 indicate the high and low values at which the variable components were too far from the fixed value components in the other circuit and caused desynchronization. The desynchronization values show a wide range of conditions where synchronization can occur. The window of capacitance is on the order of a few microFarads. The window for the resistor is on the order of a few thousand ohms for both circuits and the input amplitude difference between the two RLD circuits is around ten volts before synchronization is

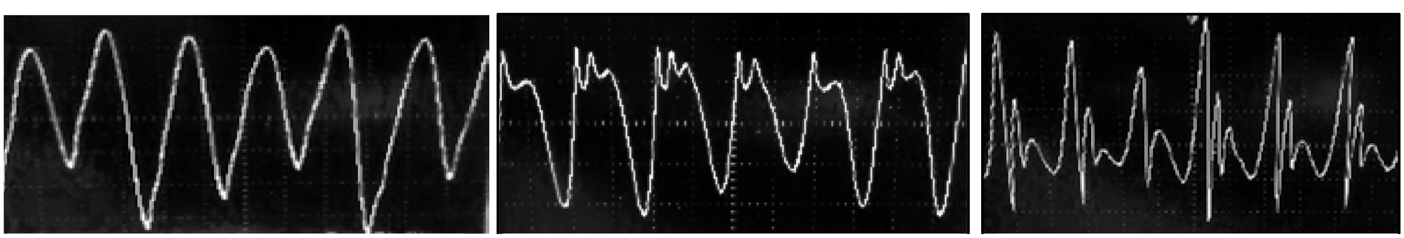

Figure 5. Waveforms from the Sprott circuit observed in this experiment. R* for these waveforms was 1k ohm and C* was 1.0 microFarads.

Figure 6. Observed waveform for the RLD circuit

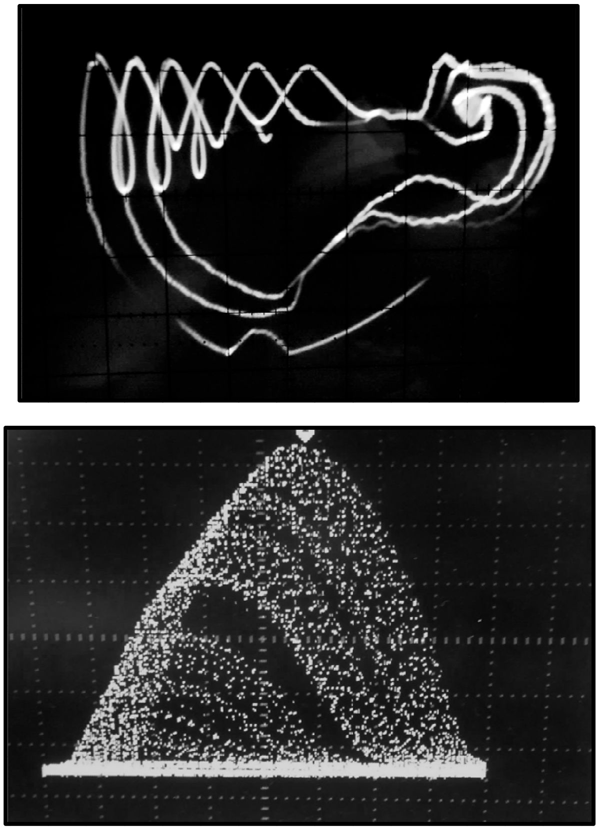

Figure 7. Observed attractors for the Sprott circuit (top) and the RLD circuit (bottom)

lost. Reducing the potentiometer completely did not cause desynchronization which is probably due to the resistances still in the circuit due to the inductors and signal generators. Interestingly, varying the inductance of the RLD circuit did not affect the synchronization.

Discussion

While the desynchronization parameters were able to be determined, the numbers could have been more accurate by using other measuring methods. For instance, running the circuit through LabVIEW would allow for precise, quantitative observation of the difference between the two signals. No anomalies from capacitance inside the prototyping board were observed and the 60 Hz signal from the lights and other external sources appeared to be minimal compared to the actual signal. Hardwiring the RLD circuit after examining its behavior on a prototyping board did not appear to improve the performance of the circuit by much, thus it is believed that leaving the jerk circuit on the prototyping board for measurements had little to no impact on the results. The chaotic range of the RLD circuit could not be fully established because the signal generator could not reach any higher in amplitude than around 10V. However, the range that could be verified is large enough that the RLD circuit is considered robust.

Conclusion

The waveforms observed on the oscilloscope verify that the signals seen in each circuit in this experiment are chaotic and are the same as those in previous works (Hammill 1993, Sprott 2011). The bifurcation points indicate that both circuits approach chaos through period-doubling in accordance with the Feigenbaum constant. The wide range of chaos in each circuit suggested that synchronized chaos would be

Table 1: Bifurcations and desynchronization parameters for each circuit

Circuits	Bifurcations	Bifurcation Ratios	Chaotic Range	Desynchronization Values		
				C (μF)	R (Ω)	V_{source} Difference
Jerk	R (±0.1 kΩ) 0.519 0.960 1.038[c]	5.65	1.038 – 3.940 kΩ	Upper: 4.522 Lower: 0.290	Upper: 3,860 Lower: 48	N/A
RLD	V_{source} (±0.10V) 1.63 3.48 3.88	4.63	5.8V-	N/A	Upper: 2,016 Lower: N/A	Upper: 5.98 Lower: -6.08

c: Chaos occurred at bifurcation point

maintained for largely varying circuits, and the suggestion was verified by the measured ranges of chaotic synchronization. While there are means for achieving more accurate numbers, the large range of variation allowed is undeniable. These findings are the first ones needed to begin an examination of synchronized chaos as a means of cancelling noise. The possibility of noise cancellation is exciting and experimental applications of chaotic noise cancellation through synchronization with these circuits can now be examined (Hammill 1993, Sprott 2011).

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Ecology of the Squirrel Treefrog (*Hyla squirella*) in Southern Arkansas

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Running Title: Ecology of the Squirrel Treefrog

Abstract

We conducted an ecological study of the Squirrel Treefrog, *Hyla squirella* near El Dorado, Union Co., Arkansas from May-Oct. 2013. We extended the known distribution by ~2 km and documented the first breeding occurring on 28 May and the first transformation of juveniles on 27 Aug. Three endoparasites were documented: *Opalina* sp., *Nyctotherus cordiformis*, and *Physaloptera* sp. larvae. We also provide information on endoparasites of Florida *H. squirella* as well as a summary of helminths of this frog.

Introduction

Hyla squirella Bosc, the Squirrel Treefrog, is a small hylid frog found throughout the southeastern United States (Conant and Collins 1998; Fig. 1). It was only recently discovered to occur in Arkansas, with the first record on 21 May 2013 (Fulmer and Connior 2013). This frog is distributed throughout Louisiana (Dundee and Rossman 1989), and, in fact, is known to occur ~80 km from the Arkansas location in nearby Ouachita Parish, Louisiana (Dundee and Rossman 1989, Fulmer and Connior 2013). Since this species was just detected in Arkansas in 2013, this study was conducted to elucidate the ecology of this species within Arkansas, specifically in regards to habitat, reproduction, and parasites. In addition, we provide information on some endoparasites of Florida *H. squirella* as well as a summation of the helminths of this frog.

Materials and Methods

During May-Jul. 2013, potential locales within ~5 km were searched near the discovery of the initial population (Site 1; 33.2327°N; 92.6287°W) within Union Co. Individuals of *H. squirella* were collected by hand, measured for snout-vent length (SVL), and necropsied for parasite infection and reproductive status.

Additional ecological characteristics, such as number observed, numbers of males calling, reproductive activity, and other anuran species observed were noted as well. Specimens were placed in individual bags on ice and within 48 hr frogs were overdosed with a 10% v/v ethanol solution (HACC 2004). A mid-ventral incision from mouth to cloaca was made to expose the gastrointestinal tract. Specimens were examined for select protists, including the gall bladder for myxozoans and the rectum for opalinids and ciliates following McAllister (1987, 1991). Protists were processed for scanning electron microscopy following standard techniques used on other frogs (see McAllister et al. 2013) or were stained with Gomori trichrome for light microscopy. Nematodes were fixed in hot 70% v/v ethanol and placed on a glass slide in a drop of undiluted glycerol for identification. Voucher specimens of parasites were deposited in the United States National Parasite Collection (USNPC), Beltsville, Maryland. Host voucher specimens were deposited in the Henderson State University Herpetological Collection (HSU 1712-1719), Arkadelphia.

Reproductive status of females was noted by the presence of ovarian eggs. When females were gravid, eggs were counted. We recorded egg counts or estimates of egg counts for five individuals. For the first female, we counted every egg within the abdominal egg mass and we estimated the egg counts for the remaining four. We determined the number of eggs in a volume that displaced 0.5 mL of water. Then, we estimated the total number of eggs by the volume of water displaced by the total egg mass. Final estimates were calculated by multiplying egg number in 0.5 mL of water by the total volume displaced.

Figure 1. *Hyla squirella* captured from Union County, Arkansas. Top (adult male); Bottom (recently metamorphosed juvenile).

Figure 2. *Hyla squirella* breeding site.

Results

One additional site was found (Site 2; 33.2142°N; 92.6315°W); a large breeding site ~2 km S of the original site near the Junction of US 63 and US 167 Bypass (Fig. 2). Individuals of *H. squirella* were collected by hand (four collected on 28 May 2013 [from Site 1] and three on 9 June 2013 [from Site 2]) and necropsied for parasite infection. In addition, 34 adult individuals were collected on 26 July 2013 for size and reproductive data, in addition to 12 recently metamorphosed juveniles collected from 28 August – 2 September 2013 from Site 2.

We documented numerous males calling (see Table 1) as well as pairs in amplexus (Fig. 3). We observed the following additional anurans calling from the same area: *Acris blanchardi*, *Anaxyrus fowlerii*, *Hyla cinerea*, *H. chrysoscelis*, *Gastrophryne carolinensis*, and *Lithobates sphenocephalus*. On 26 July 2013, we collected and measured 34 adults with a mean SVL of 31.8 mm (range = 29-35 mm) for 25 males and 33.8 mm (range = 30-37 mm) for 9 females. Of the 9 females, 7 were gravid. Total mean egg count estimates for the four individuals was 1324 eggs (range = 701-1635 eggs) per female. On 31 October 2013, after a heavy rain, we did not hear any *H. squirella* calling or observe any individuals.

We also observed that the younger tadpoles of *H. squirella* had golden dorsolateral stripes and older tadpoles were brown with golden flecks. Tail fins were clear, except for some dark mottling. Older tadpoles had white pigmentation on their throats. The mean SVL for 12 recently metamorphosed juveniles was 12.5 mm (range = 11-55 mm), with the first individual being observed on 16 August 2013 from Site 2.

Figure 3. *Hyla squirella* exhibiting amplexus.

Three species of endoparasites were found in *H. squirella*: *Opalina* sp., (USNPC 107672.02; Fig. 4a), *Nyctotherus cordiformis* (USNPC 107672.01; Fig. 4b), and three third-stage larval *Physaloptera* sp. (USNPC 107935); all in one of seven (14%) *H. squirella*. Each represents a new host record. We provide a summation of all previously reported helminths in *H. squirella* as well as previously unpublished records collected in Florida by the senior author in Table 2.

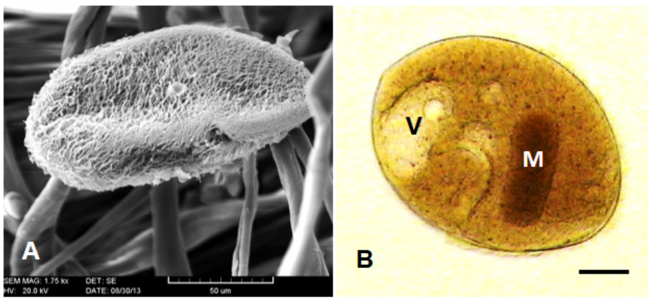

Figure 4. Protists from *Hyla squirella.* A. Scanning electron micrograph of *Opalina* sp. B. *Nyctotherus cordiformis* (unstained). Note macronucleus (M) and vacuole (V). Scale bar 25µm.

Discussion

Hyla squirella primarily reproduce during late spring and summer with calling choruses being recorded from Louisiana from March to November, and with calling individuals even being recorded in December (Dundee and Rossman 1989). We documented successful breeding of adults and metamorphosis of larvae for the first time in Arkansas (Fig. 1). Our reported breeding from May to July coincides with the typical breeding season reported in the literature. Our egg count of 989 is similar to two egg counts of 972 and 942 from Georgia (Wright 1932). Wright (1932) reported hatching within two days and an estimated larval period of 40-50 days, which coincides with the development at our site. Dundee and Rossman (1989) reported the only newly transformed individuals during their research from 25 September in Louisiana.

Both *Opalina* sp. and *N. cordiformis* were reported from every hylid host that inhabits Arkansas (Muzzall and Sonntag 2012, McAllister et al. 2013). Both of these parasites are ubiquitous in amphibians. *Physaloptera* sp. nematode larvae was recently reported in Arkansas for the first time from the Cajun chorus frog, *Pseudacris fouquettei*, in Union County (McAllister et. al. 2013). *Physaloptera* has been reported previously from the hylid frogs *Hyla versicolor* in Virginia and *Pseudacris crucifer* in North Carolina (Goldberg et al. 2009); both of these hylid species also occur in Arkansas. All of these endoparasites are cosmopolitan in their ranges. This small hylid is now host of three trematodes, one cestode, and five nematodes (Table 1). Although its parasite fauna is depauperate (Aho 1990), additional collections from this site and elsewhere will likely increase the number of helminths known from this host.

This study extended the known distribution of *H. squirella* within Arkansas; however it was only extended by about 2 km to the south. We suspect further systematic distributional surveys may produce additional breeding populations. Future surveys should include areas from southwestern to southeastern Arkansas, focusing on the southernmost tier of counties bordering Louisiana. As evidenced by the large breeding site, *H. squirella* can be quite numerous during breeding events in rainy weather, while going undetected during the rest of the year.

Acknowledgments

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Table 1. Ecological Notes of *Hyla squirella* from Union Co., Arkansas.

Locality	Date	Time (hrs)	Temp (°C)	Weather (rain)	Notes
Site 1	1 Jun 2013	2320	21.1	cloudy (8.9cm)	3 males calling 2 amplectant pairs
	2 Jun 2013	2130	22.2	clear	5 males calling
	18 Jun 2013	2120	25.6	cloudy (1.3cm)	one male calling
	23 Jul 2013	2300	25.6	cloudy (1.9cm)	6 males calling
	26 Jul 2013	2330	21.1	cloudy (6.3cm)	13 males calling
Site 2	7 Jun 2013	2230	20.0	clear	12 males calling
	8 Jun 2013	2115	23.3	clear	3 males calling
	9 Jun 2013	2100	23.9	clear	5 males calling
	26 Jul 2013	2330	21.1	cloudy (6.3cm)	>50 males calling >10 amplectant pairs
	27 Jul 2013	2300	20.6	clear	>50 males calling
	20 Sept 2013	2215	21.7	cloudy (8.9cm)	>25 males calling

Table 2. Summary of helminth parasites from *Hyla squirella*.

Helminth	Locality	Prevalence*	Reference
Trematoda			
Lechriorchis tygartii[1,2]	Florida	not given	Sears et al. (2012)
Polystoma nearcticum	Florida[4]	4/14 (29%)	This report
	North Carolina	not given	Price (1939)
Renifer aniarum[1,2]	Florida	not given	Sears et al. (2012)
Cestoidea			
Cylindrotaenia americana	Florida[4]	3/14 (21%)	This report
	Texas	1/11 (9%)	Harwood (1932)
Nematoda			
Acuariidae gen sp.[3, 5]	Florida[4]	1/14 (7%)	This report
Cosmocercoides variabilis	Florida[4]	4/14 (29%)	This report
	Texas	2/11 (18%)	Harwood (1932)
Gyrinicola batrachiensis[2]	Florida	1/1 (100%)	Pryor and Greiner (2004)
Physaloptera sp.[3, 6]	Arkansas	1/7 (14%)	This report
	Florida[4]	1/14 (7%)	This report
Rhabdias ranae	Florida	not given	Walton (1938)

*Number infected/number examined (percent).
[1]Experimental infection.
[2]Tadpoles only.
[3]New host record.
[4]Previously unpublished records from Topsail Hill Preserve State Park, Walton Co., FL collected on 28 Mar. 2014 by MB Connior. Host vouchers (SVL = 26.6 ± 4.2 mm, range = 21-33 mm, *n* = 14) deposited in Arkansas State University Museum of Zoology (ASUMZ 33216-33226). Parasite vouchers deposited in USNPC.
[5]Larvae in cysts.
[6]Third-stage larvae.

Proportionality of Population Descriptors of Metacercariae of *Clinostomum marginatum* in the Orobranchial Cavity of Black Bass (*Micropterus* spp.) from Arkansas Ozark and Ouachita Streams

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Running title: Proportionality of Population Descriptors of *Clinostomum marginatum* in Black Bass.

Abstract.

In a previous study of *Clinostomum marginatum* metacercariae in *Micropterus dolomieu*, I reported that the population parameters of mean abundance, standard deviation, maximum abundance, mean intensity and mean intensity standard deviation were proportional between the total population and the orobranchial numbers for 16 locations in Arkansas Ozark and Ouachita streams. This allowed an assessment of the parasite populations by only examining the mouth and gill areas without sacrificing a valued sports fish. The present study examined the same orobranchial parameters utilizing correlation and descriptive statistics to determine if proportionality also existed between the different localities. I have now included an analysis of skewness and kurtosis (drift and shape) of the populations' curves. Proportionality of regression values was highly significant in terms of R^2 and *P* between all parameters except prevalence, which showed much weaker correlations with the other parameters. The interpretation of these results is that the distribution of infections in the different bass populations are density independent i.e., although the numbers of parasites change from location to location, the pattern of distribution in the host populations remains similar. This may best be explained by a spatiality of distance from the infection source (snails) and nonrandom distribution of hosts (bass) producing aggregation near the snails and a negative binomial distribution throughout the the population.

Introduction

Daly et al. (2007) reported that there was proportionality of the population descriptors between *Clinostomum marginatum* metacercariae in the orobronchial cavity relative to the total population numbers in the entire body of *Micropterus dolomieu* (smallmouth bass) hosts. The importance of this finding was that by just counting the metacercariae in the gills and mouth of the bass hosts one could get a reasonably close estimate of total population parameters of the parasite without harming a valuable sport fish, which could then be returned to its habitat. It would seem that if such proportionality existed between anatomical sites in the fish hosts then proportionality of population parameters might also exist between the different geographical locales and a single anatomical site (orobranchial). The present study uses similar techniques, correlation and descriptive statistics, from the raw orobronchial data from 17 different locales to test this hypothesis. If such is the case then the parasite distribution in the hosts would appear to follow a pattern, most likely a spatial and stochastic one, independent of the different densities of *Clinostomum* seen in the different populations.

Methods and Materials

Smallmouth and Kentucky bass (KY), *Micropterus dolomieu* and *M. punctulatus* (from 1 locale) hosts were collected from 1 Ozark (Crooked Creek) and 3 Ouachita Mountain (Caddo, Ouachita, and Saline) streams and consisted of 17 different locales on 4 streams in Arkansas with one exception: WR 88 and WR 90 on Crooked Creek, whose values were so greatly different they were treated as separate locales. The total number of hosts collected from May to October 1988-1990 was 579. Details of the collecting locales and necropsy techniques are found in Daly et al. (2002, 2007) and Daly Jr. et al. (2002). Metacercariae (or yellow grubs) were collected from the orobranchial cavity (visible in/on mouth and gill surfaces) of the hosts and counted. The values of population parameters for this site were calculated from these counts using Microsoft Excel (2010). First, descriptive statistics for each locale were obtained and these results were then used to determine correlation coefficients between the data from the different locales. To test for significance, an F test was applied to determine equal or unequal variances and then an appropriate T Test was used to determine *P* values. Definitions of population parameters followed that of Bush et al. (1997) and, with abbreviations used, are as follows: Mean abundance, MA (average number of parasites per host); Mean abundance standard deviation (MASD); Maximum abundance, Max (largest number of parasites in a single host); Prevalence (percent of infected hosts); Mean intensity, MI (average number of parasites in infected hosts only); Mean intensity standard deviation (MISD); Variance (S); S/MA (Variance to mean ratio, a relative measurement of aggregation – reflected as a negative binomial population distribution where the heaviest infections are in only a few hosts); SD/MA (a simpler measurement of aggregation eliminating a power function ($S = SD^2$) for linear arithmetic comparisons); Skewness, Skew (measure of shift in population curves) and Kurtosis, Kurt (measure of shape of the population curve). One is dealing not only with 17 different populations but also with two different population parameters of MA and MI within those 17 populations. The earliest study in this series of yellow grub and black bass examined length and maximum circumference of the bass hosts and found no correlation with parasite density measurements (Daly et al. 1987) therefore this is not an issue in this study.

Results

Descriptive statistics for *Clinostomum marginatum* metacercariae in smallmouth (*Micropterus dolomieu*) and Kentucky bass (*M. punctulatus*) orobranchial cavities collected from 17 different locales can be found in Table 1 (mean abundance and derivations) and Table 2 (mean intensity and derivations). For mean abundance the average per host ranged from 0.1 to 3.8 giving a low to high spread that provides excellent correlation analysis as opposed to population means being close together. The WR 90 locale is not included in the regression analyses because its values are very high relative to other locales, (the highest ever recorded for yellow grub in fish hosts – Daly et al. (1991) and gives highly skewed but significant results. Although using such a large outlier does give higher R^2 and *P* values the predictive values obtained for the other locales with smaller means, using the applied correlation coefficients of slope and intercept, gives overestimations of mean abundance values. Excluding WR 90 gives very close predictive values for mean abundance (and other derivations). This skewing of values by WR 90 indicates a break in the linearity required for accurate correlation analysis, therefore the exclusion. However, using the coefficients from the other 16 populations gives a reasonable estimate of the WR90 values, e.g. actual MASD = 97.2 and the predictive value of MASD with MA as the independent variable = 81.3.

Variance to mean ratios.

Discussion of results requires an understanding of the basic population distribution of helminth parasitic infections. First, SDs being larger than the mean (Table 1) indicates a negative binomial distribution (Crofton 1971, Pennycuick 1971) that requires non-parametric statistics for population comparisons as opposed to SDs being smaller than the mean which would assume a normal distribution necessary for parametric statistics. The negative binomial distribution is due to aggregation or dispersion where a few hosts have the majority of the parasites. Mean intensity eliminates zeros and increases the mean and reduces SD. That this reduction of zero values to produce normal distributions does not always work can be seen when the mean intensity SD is still greater than the mean (Table 2). The variance (S or SD^2) to mean ratio reflects the degree of dispersion and is interpreted as less than one = normal distribution, one = Poisson distribution, and greater than one is a negative binomial. For mean abundance (Table 1) the dispersion ratio is 1 or less for only 2 populations and for smaller mean values. For mean intensity dispersion deviates from negative binomials for 6 populations and primarily at the lower mean values. The ratio of SD/MA (or SD/MI) as suggested by Poulin (2007) is preferred here because the variance to mean ratio of S/MA (or SD^2/MI) has a power function, which is not arithmetically linear that is required for parametric statistics. However, the latter is useful for determining relative strength of aggregation. The average for the ratio of mean abundance SD/MA (Mean±SD, confidence limit) for 16 populations is 2.1 ±0.7, 0.36 and for mean intensity is 0.84±0.5, 0.23 (WR90 is included since the value is a ratio). A Student T test for unequal variances gave a *p* value of 1.9E-06 indicating a significantly high probability of the two populations being unequal. This is expected since the MI is a redacted form of MA. A major finding is that the ratios are proportionally the same and a rounded factor of 2 or 0.9 (derived from the averages of both factors) can be used as a rough but reasonable predictive estimate of SD/MA, SD/MI.

Proportionality of population descriptors.

Table 3 shows the results of regression analysis of the various descriptors of mean abundances, mean abundance standard deviations (SD), maximum abundances, mean intensities, mean intensity standard deviations (SD), and prevalence.

All regressions showed significant correlations. Log-log transformations were required to obtain more significant correlations for all prevalence comparisons Other log-log transformations gave better correlations than arithmetic comparisons, increasing the R^2 and P values slightly. Slope and intercept values can be applied to the independent variables and these give close estimates of the dependent variables. This proportionality can be seen in Figure 1 (A, B, C, D) as examples where the dependent variable was estimated using the correlation coefficients. Maximum abundance (largest number of parasites in a single

Table 1. Population parameters of *Clinostomum marginatum* metacercariae in the orobranchial cavity of *Micropterus dolomieu* and *M. punctulatus* (O KY) from locales in Ozark and Ouachita streams in Arkansas. Locations are identified in Methods. N = host number, Prev. = prevalence, Max = maximum number of parasites in one host, MA = mean abundance, SD = standard deviation, S = variance, Disp. = dispersion, Skew = Skewness, Kurt = kurtosis. Values are for Mean abundance.

Location	N	Prev.	Max	MA	MASD	S	Disp.	MA/SD	Skew	Kurt	Kurt/Skew
HU	10	10	1	0.10	0.30	0.09	0.9	3.0	3.2	NA	NA
H1	38	11	2	0.13	0.41	0.17	1.3	3.2	3.4	11.8	3.5
H3	37	32	3	0.41	0.69	0.48	1.2	1.7	2.5	6.2	2.5
CG	29	11	2	0.18	0.55	0.30	1.7	3.1	3.0	8.2	2.7
GL	23	20	2	0.29	0.63	0.40	1.4	2.2	2.3	4.7	2.0
H2	45	16	2	0.18	0.43	0.18	1.0	2.4	2.0	4.7	2.0
S	20	20	8	0.64	1.73	2.99	4.7	2.7	4.8	4.8	1.0
BS	20	47	5	1.00	1.49	2.22	2.2	1.5	1.5	2.2	1.5
O	37	44	10	1.07	2.07	4.28	4.0	1.9	1.0	1.8	1.8
Y	44	49	11	1.14	1.90	3.61	3.2	1.7	3.5	16.2	4.6
CC	42	49	17	1.84	3.25	10.56	5.7	1.8	2.9	10.4	3.6
P	27	59	10	1.85	2.24	5.02	2.7	1.2	2.4	6.1	2.6
G	30	67	13	2.70	3.40	11.56	4.3	1.3	2.3	2.9	1.2
O KY	19	37	26	3.60	7.40	54.76	15.2	2.1	1.9	8.3	4.4
T	105	64	25	3.70	4.90	24.01	6.5	1.3	1.9	3.5	1.9
WR88	36	53	67	3.80	11.04	122	32.1	2.9	3.3	11.3	3.5
WR90	17	65	400	42.50	97.20	9447	222.3	2.3	5.4	31.0	5.7

Table 2. Population parameters of *Clinostomum marginatum* metacercariae in the orobranchial cavity of *Micropterus dolomieu* and *M. punctulatus* (O KY) from locales in Ozark and Ouachita streams in Arkansas. Locations are identified in Methods. MI= mean intensity, SD = standard deviation, S = variance, Disp. = dispersion, Skew = Skewness, Kurt = kurtosis. Values are for Mean intensity (all infected hosts only – prevalence = 100 %).

Locale	MI	MISD	MI S	Disp	SD/MI	Skew	Kurt	Kurt/Skew
HU	1	NA	NA	NA	NA	NA	NA	-----
H1	1.25	0.43	0.18	0.10	0.34	2.4	6.0	2.4
H2	1.14	0.35	0.12	0.11	0.31	2.6	6.2	2.4
H3	1.25	0.50	0.25	0.20	0.40	2.0	4.0	2.0
CG	1.67	1.58	2.50	1.50	0.95	-1.7	NA	-----
GL	1.25	0.52	0.27	0.22	0.42	2.0	4.0	2.0
S	3.20	3.77	14.21	4.44	1.18	0.5	NA	NA
BS	2.29	1.38	1.90	0.83	0.60	1.3	1.5	1.2
O	2.40	2.57	6.60	2.75	1.07	3.2	6.5	2.0
Y	1.80	1.05	1.10	0.61	0.58	3.2	12.1	3.8
CC	3.86	3.80	14.44	3.74	0.98	2.2	6.0	2.7
P	3.13	2.40	5.80	1.84	0.77	1.9	3.6	1.9
G	4.05	3.50	12.25	3.02	0.86	1.6	1.5	0.9
T	5.70	5.10	26.01	5.60	0.89	1.6	2.2	1.4
O KY	9.70	9.70	94.09	9.68	1.00	1.7	2.4	1.4
WR88	7.26	14.40	240	30.26	1.95	2.9	15.6	5.4
WR90	65.6	120	14634	213	1.83	2.6	7.0	2.7

host), is related to the degree of aggregation since its value produces a right shift of population distribution. Maximum abundance also shows excellent correlation with other variables.

All of the correlations show proportionality between the parameters of both MA and MI populations.

Skewness and Kurtosis.

Regression analyses were performed on the skewness and kurtosis values from 14 populations (including WR90) that had available values for analysis as seen in Table 4. Correlations were significant for skewness and kurtosis within the parameters of MA and MI but not between the two parameters with P = <0.05. The descriptor values change but the ratios and the shape of the curves do not. A major difference is the effect of the elimination of zero values from MI which is clearly shown by the shift of the mean or mode (skewness) of the population distribution from the MA values (Tables 1 and 2).

In summary, these results overall show highly significant proportionality amongst the population descriptors. Population curves also have similar shapes proportionally but differ in skewness when the population values are changed (redacted zeros). SD/MA and SD/MI ratios have a common factor for each of the population sets of about 2.2 and 0.9 respectively and are also significantly different between the two sets of populations.

Table 3. Regression analysis of population parameters of *Clinostomum marginatum* metacercariae in the orobranchial area of *Micropterus dolomieu* and *M. punctulatus* (O KY) hosts from 16 stream locales in the Arkansas Ozark and Ouachita highland areas. Variables used are from Tables 1 and 2. SD = Standard deviation.

Independent Variable	Dependent variable	R^2	Intercept	Slope	*P*
Mean abundance	Mean abundance SD	0.81	-0.12	2.0	2.0E-06
Log_{10} mean abundance	Log_{10} maximum abundance	0.91	0.93	0.9	1.2E-08
Mean abundance	Maximum number	0.67	-1.37	10.0	1.1E-04
Mean abundance	Mean intensity	0.83	0.81	1.7	7.7E-07
Mean abundance	Mean intensity SD	0.70	-0.19	2.4	4.9E-05
Mean abundance	Mean abundance variance	0.56	-9.60	17.5	9.0E-04
Log_{10} mean abundance	Log_{10} MA Variance	0.94	0.57	1.7	4.1E-10
Mean abundance SD	Mean intensity	0.82	1.16	0.8	1.4E-06
Mean abundance SD	Mean intensity SD	0.96	-0.20	1.3	8.6E-11
Mean abundance SD	Maximum abundance	0.94	-1.62	5.4	9.8E-10
Mean abundance SD	Mean abundance variance	0.91	-12.10	10.3	1.0E-08
Mean intensity	Mean intensity SD	0.81	-1.30	1.4	1.0E-06
Mean intensity SD	Maximum abundance	0.90	-0.40	4.1	6.9E-08
Log_{10} mean intensity	Log_{10} MI variance	0.96	-0.69	2.8	3.7E-10
Prevalence	Mean abundance	0.58	-0.49	0.05	5.8E-04
Log_{10} Prevalence	Log_{10} mean abundance	0.85	2.70	1.7	4.4E-07
Prevalence	Mean abundance SD	0.29	-0.29	0.08	0.03
Log_{10} Prevalence	Log_{10} mean abundance SD	0.67	-1.70	1.30	1.0E-04
Prevalence	Mean intensity	0.25	0.90	0.06	0.05
Log_{10} prevalence	Log_{10} mean intensity SD	0.45	0.60	0.70	0.004
Prevalence	Maximum abundance	0.26	-2.73	0.42	0.05
Log_{10} Prevalence	Log_{10} maximum abundance	0.67	-1.30	1.43	1.2E-04

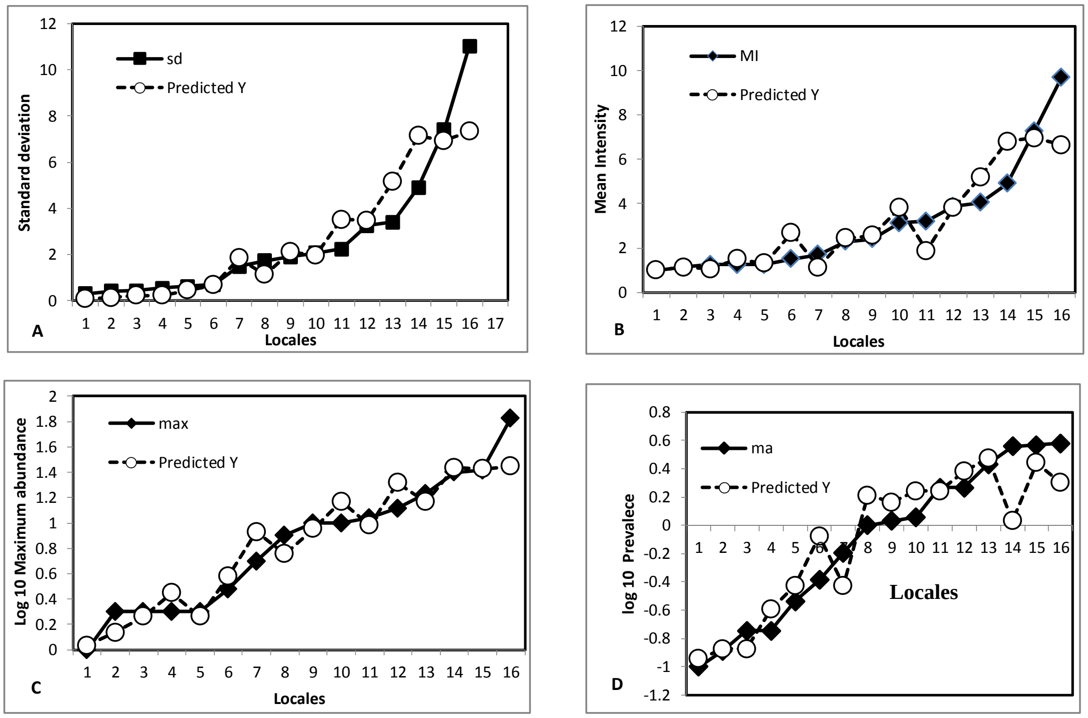

Figure 1. Examples of regressions of descriptive parameters from 16 populations of *Clinostomum marginatum* from the orobranchial cavity of stream black bass (*Micropterus dolomeiu and M. punctulatus)* from Ozark and Ouachita streams in Arkansas. A, B, C regressions are with mean abundance as the independent variable with the graphs showing the dependent variable and the predicted values of the dependent values using the regression coefficients from Table 3. A. Standard deviation of mean abundance, B. Mean intensity, C. Maximum abundance and D. Prevalence as the independent variable and mean abundance as the dependent variable showing the predicted mean abundance from the prevalence values.

Table 4. Regression analysis of skewness and kurtosis of mean abundance and mean intensity of population curves of *Clinostomum marginatum* in black bass orobranchial cavities.

Independent variable	Dependent variable	R^2	Intercept	Slope	*P*
Mean abundance skewness	Mean abundance kurtosis	0.89	-8.8	6.7	3.4E-07
Mean intensity skewness	Mean intensity kurtosis	0.70	-6.6	5.5	2.1E-04
Mean abundance skewness	Mean intensity skewness	0.12	1.7	0.2	0.23*
Mean abundance kurtosis	Mean intensity kurtosis	0.21	3.6	0.2	0.10*
Mean abundance skewness	Mean intensity kurtosis	0.23	1.1	1.7	0.08*
Mean intensity skewness	Mean abundance kurtosis	0.15	1.0	3.2	0.17*

* Not significant with $P = > 0.05$

Discussion

Population parameters of helminth parasites have traditionally been used to compare differences and/or similarities between parasite populations (Dobson and Beveridge 1987, Poulin 2007). At least 62 reports in just the last 10 years of the Journal of Parasitology have utilized these parameters for comparisons and predictive power. This is the first study to show close correlations between all the commonly used population parameters of prevalence, mean abundance, mean abundance standard deviation, mean intensity, mean intensity, mean intensity standard deviation, and of variance to mean ratios. The 17 populations examined herein are useful for these parameter analyses because they are from geographically different locales spread along a single stream, or even from different streams, and therefore represent a range of parasite densities in the hosts that can give a reasonable spread for representative correlations. The correlation coefficients of R^2 and P are so significant that the relationships do not require more advanced statistics to demonstrate their existence. The positive correlations between MA and MI parameters were not unexpected, since one population (MA) was derived from the other (MI) and they are different only due to the redacted zeros.

The relationship between prevalence and mean abundance have been most popularly examined as a predictor of population density and/or intensity, but prevalence has not always been shown to be a reliable predictor of abundance or intensity of infection. Janovy et al. (1997) and recently Shostak (2014) resorted to log transformations to show such a relationship and in the latter case the curve obtained was asymptotic at the top end. In this present study, prevalence without log conversions was shown to have the weakest correlations of all the combinations of parameters between mean abundance or mean intensity. Prevalence can be a problem for estimations of population numbers at higher values because at 100% or values near 100% they cannot recognize a further increase in the number of parasites. Conversely, Shostak (2014) found that abundance actually leveled off at 90% prevalence, but this is not necessarily the case, as with the total population values for *Clinostomum* in smallmouth bass (Daly et al. 2007) which increased. There is an interesting relationship that can be detected between prevalence and the two density measurements. If one knows the prevalence and the value of one of the two means, then simply by multiplying a factor determined by dividing 100/prevalence or prevalence/100, the other mean can be closely estimated (e.g., Prevalence = 50 %, then MA multiplied by 2 = MI or MI multiplied by 0.5 = MA).

This is the first report that reveals the close relationships that exist between population curves for skewness and kurtosis for the same population parameter (mean abundance or mean intensity). The correlations between skewness and kurtosis showed that the population curves are similar within MA and MI parameters, but are statistically dissimilar between the curves of the two groups as shown by a T test which did not show a difference between the average skewness' within MA and MI but did with correlations which paired the values between MA and MI. This is to be expected since the two population curves are obviously different because of redacted zeros.

The negative binomial distribution of most helminth parasite populations has been a problem for comparisons of populations requiring transformations such as log values (or nonparametric techniques) to obtain a normal distribution required for parametric statistics. Mean intensity removes zeros which raises the mean value and lowers the standard deviation, but even this does not necessarily produce normal distributions for parametric comparisons. This redaction of zeros (only) is somewhat subjective and questionable methodology since removing zero values defies the logic of population sampling. Despite this, in the current study, close correlations were found between the two populations' MA and MI parameters. This indicates that removal of zeros did not affect the basic relationships except for geometry of the curves.

One caveat is the exceptionally large parasite density in WR 90 that shifts the R^2 and P values to greater significance, but lowers the predictability for smaller density populations. This suggests that the relationships between the variables may not be totally linear but are probably polynomial when including very high values (outliers?). This does not diminish or exclude the use of correlations since the predictability of the independent variable is still high for most parameters (Table 3; Figure 1).

Degree of aggregation has traditionally been the use of S/mean or a calculated K value (Esch et al. 1977, Poulin 2007). The use of SD/MA removes a power function (SD^2) that can make linear correlations questionable. A strong similarity between means and standard deviations can be seen in the ratios obtained for the different populations, which can be rendered to a factor of approximately 2. Standard deviation/mean can also be a comparative measure of degree of aggregation, although values such as assigned to S/mean (>1 = aggregation) have not yet been generally accepted. Aggregation is not dramatically different amongst the populations, as seen herein with the ratios, but has a relativity; i.e., the SD/MA, that unites the populations' distributions. Maximum abundance is also a useful tool to indicate aggregation and it is also proportional although the high value distorts the population curve from a random, normal distribution.

In conclusion, the mean and standard deviation relationships show populations with a strong stochastic characteristic with the interaction of black bass and yellow grub in an aquatic environment. This is evidence for the presence of a relatively stationary snail host producing cercariae into a setting where the bass hosts are not randomly distributed (Etnier and Starnes 1993). The proportionality of parameters is further evidence for the non-randomness in that it also implies very structured populations. A simple way to look at the proportionalities of the population parameters is to consider the different populations similar to a nesting set of Russian dolls that are alike in appearance but different in geometric proportions and size. Empirical evidence is necessary to further substantiate the theory that these relationships are due, at least in this case, to the spreading cercariae encountering a non-random host distribution of smallmouth bass producing a stochastic aggregated population, but where the parameters remain mathematically proportional.

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Sampling Local Fungal Diversity in an Undergraduate Laboratory Using DNA Barcoding

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Running title: Sampling Local Fungal Diversity using DNA Barcoding

Abstract

Traditional methods for fungal species identification require diagnostic morphological characters and are often limited by the availability of fresh fruiting bodies and local identification resources. DNA barcoding offers an additional method of species identification and is rapidly developing as a critical tool in fungal taxonomy. As an exercise in an undergraduate biology course, we identified 9 specimens collected from the Hendrix College campus in Conway, Arkansas, USA to the genus or species level using morphology. We report that DNA barcoding targeting the internal transcribed spacer (*ITS*) region supported several of our taxonomic determinations and we were able to contribute 5 *ITS* sequences to GenBank that were supported by vouchered collection information. We suggest that small-scale barcoding projects are possible and that they have value for documenting fungal diversity.

Introduction

At present, 70,000 species of fungi have been published (Blackwell 2011); however, estimates suggest there may be as many as 1.5 million extant fungal species, leaving the majority of species to be described (Hawksworth and Rossman 1997). Traditional taxonomy requires diagnosable morphological and/or anatomical characters, but variation within some fungal species and subtle variation between related taxa makes it difficult to rely solely on these characters when identifying an unknown fungal specimen (Hyde et al. 2013). Fungal identifications are greatly complicated because many species do not produce, inconsistently produce, or briefly and seasonally produce the macroscopic reproductive structures (sporocarps) that are used to distinguish between sister taxa, e.g. the gills or pores of mushrooms. Environmental samples, where fungal hyphae are collected without fruiting bodies, are especially challenging to identify using morphological characters as they are likely to represent a mixture of different species (Nilsson et al. 2012). In addition to subterranean fungi, endophytic fungi that grow between living plant cells are very poorly understood taxonomically but are of great interest as a source of antimicrobial metabolites for use in agriculture and human health (Schulz et al. 2002). Small geographic areas have the potential to harbor very high levels of fungal biodiversity that will require extensive surveying, but identifying the fungal specimens from these surveys continues to be a challenge even if sporocarps are available. In order to identify a vascular plant in North America, we could refer to a flora, which lists the known species of each geographic region and provides identification guides and habitat information. Vascular plant flora are incomplete or outdated for many regions of the world. In contrast, a comparable mycoflora is completely absent, even for fungi in North America with large sporocarps. For Arkansas, there is no checklist of fungal species that have been documented to grow here. Promising efforts are underway to create a mycoflora (Bruns 2012) that will rely on well-documented collections with voucher specimens. These comprehensive guides, still in the early stages of compilation, will also integrate genetic relatedness using similarity of DNA sequences (DNA barcoding).

Using Polymerase Chain Reaction (PCR) to amplify a targeted genetic marker and DNA sequencing to determine the content of that sequence, DNA barcoding allows for the comparison of an informative sequence from an unknown fungal sample against a database of identified sequences. This technique will eventually allow us to describe and identify the asexual species and can provide an alternative method of identification for sexual species when sporocarps are not available. When used in conjunction with morphological identification, DNA barcoding is predicted to provide a more reliable means of species identification and will contribute to the understanding of cryptic species that cannot be reliably distinguished using morphological characters. Although the databases that can be used for reference are limited in several important ways, active efforts are underway to remedy these challenges (Nilsson et al. 2012, Schoch et al. 2014). First, many misidentifications remain that are difficult to correct because the DNA sequence is not paired with a vouchered specimen in a herbarium with adequate geographic collection data. Second, the databases are incomplete. Third, active curation is needed in order to coordinate taxonomic assignments.

A critical shortage of trained mycologists has been noted (Bruns 2012). Given the limited resources that are currently available to train novices on morphological fungal identification and the high likelihood that fungal taxonomy would benefit from local surveys, how might a college undergraduate laboratory best be structured? Novices are unlikely to make substantial contributions to the taxonomic issues. However, our premise was that a small local effort, conducted in a classroom setting, could make use of DNA barcoding to confirm morphological identifications and simultaneously contribute vouchered DNA sequences to a reference database that might help experts document the geographic range of fungal species. These student researchers could apply inquiry-based methods to learn and their results would help others evaluate the effectiveness of DNA barcoding and whether this type of small-scale study can play a role by strengthening geographic distribution information.

We used a protocol for fungal DNA isolation and barcoding (Schoch et al. 2012) that could be adapted by other student researchers or by natural resource managers. Using a limited set of collections, one pair of PCR primers, and one pass of PCR and nucleotide sequencing for each specimen, we evaluated how reliably DNA barcoding can presently be utilized to identify local fungi. In order to accomplish this, we collected fungal fruiting bodies from the Hendrix College campus. We sequenced the *ITS* region because it is taxonomically informative (Schoch et al. 2012) and widely represented in fungal DNA barcoding databases such as GenBank (Benson et al. 2014). DNA barcoding results were compared to our morphological identification to make species determinations. Inconsistencies between molecular and morphological identifications reflect fundamental challenges with the DNA barcoding process, but we suggest that our DNA barcoding project, conducted in an undergraduate laboratory course, has increased the number of vouchered collections in central Arkansas and will support mycologists in their efforts to document our fungi. With improving curation of barcoding databases and mycofloras, we expect these efforts to become increasingly useful in the near future.

Methods

We collected 9 specimens on October 7, 2013 on the Hendrix College campus in Conway, Arkansas (Faulkner County). An edible mushroom (J-M Farms Inc., Miami, OK, USA), purchased from a grocery store, served as a positive control. We photographed each specimen prior to collection, documented the substrate and surrounding environment, and noted morphological characters (Table 1). We removed the fruiting bodies from their substrates close to the base using a knife. We collected spore prints by placing the specimens on herbarium paper with their gills or pores facing the surface of the paper and storing them overnight in a cabinet with limited airflow. Specimens were dried at 28°C for one week then stored in zip-lock bags. Dried specimens along with their spore print and herbarium labels were permanently stored in the Hendrix College Herbarium (HXC) as voucher specimens. We identified each fungal specimen using dichotomous keys and other resources (Arora 1986, Gilbertson and Ryvarden 1988, Lincoff and Nehring 1981, Lincoff and Giovanni 1982, Roody 2003, Jay Justice *pers. comm.*).

To reduce contamination prior to DNA isolation, we scraped off the surface layer of each fruiting body with a razor blade and chopped a 15-20 mg section of the fungal fruiting body into small pieces. We placed these pieces into FastPrep Tubes (MP Biomedicals, Santa Ana, CA, USA) with a ceramic bead, garnet sand, 400 µL AP1 Buffer and 4 µL RNase A from a DNeasy kit (Qiagen, Germantown, MD, USA). We submerged the tubes for 2 minute intervals in alternating dry ice/ethanol and boiling water baths for 6 cycles in order to compromise the fungal cell walls. We processed each sample in a FastPrep homogenizer (MP Biomedicals) using 3 runs of 20 seconds at 6 m/s. We transferred the resulting lysate to a Qiagen DNeasy membrane tube and followed the DNA isolation protocol for the DNeasy kit but eluted DNA

Table 1. Ten fungal samples used in this study.

Sample	Identification	Morphological Characters	Substrate	Latitude and Longitude	Herbarium Accession	GenBank Accession	Consensus Sequence Length
A	*Bjerkandera* sp.	Bracket shape, spongy and fibrous, grey cap, dark grey pores, beige spores, no distinct scent	Stump of unknown hardwood tree species	35.100° N -92.441° W	HXC5819	N/A	N/A
B	*Inonotus dryadeus* (Pers.) Murrill	Indeterminate shape, dimples on cap, spore color unknown, flesh turned black in presence of KOH	Exposed roots of *Quercus* sp.	35.098° N -92.443° W	HXC5812	N/A	N/A
C	*Trichaptum biforme* (Fr.) Ryvarden	Bracket shape, stipe absent, tough and leathery skin, banded green and gray coloration, spore color unknown	Trunk of living *Q. shumardii*	35.098° N -92.443° W	HXC5818	KF986264	720 bps
D	*Russula* sp.	Convex and red cap, white gills, white stipe, brittle stipe and gills, off-white spores, apple-like scent when fresh	Grassy soil near *Q. pagoda* and *Q. phellos*	35.101° N -92.442° W	HXC5817	N/A	N/A
E	*Bondarzewia berkeleyi* (Fr.) Bondartsev & Singer	Overlapping caps with wavy margins, cap upper surface rough and yellow-brown, white flesh, diminished stipe, pore surface decurrent and white, spore color unknown	Trunk of living *Q. phellos* at soil surface	35.099° N -92.441° W	HXC5816	KF986266	444 bps
F	*Ganoderma* sp. (lucidum complex)	Bracket shape, sessile cap, blood-red color, porous surface, tan to brown spores	Trunk of living *Q. palustris*	35.099° N -92.441° W	HXC5820	N/A	N/A
G	*Amanita jacksonii* Pomerl.	Smooth red cap, free gills, peach colored stipe, white and sac-like volva, white spores	Moist soil near roots of *Q. phellos*	35.100° N -92.440° W	HXC5814	KF986265	643 bps
H	*Amanita* sp. (section *Lepidella)*	Dome shape, white with brown scales, brown gills, smooth stipe, partial veil, light brown spores	Mossy, moist soil under *Q. phellos*	35.100° N -92.440° W	HXC5811	N/A	N/A
I	*Boletus bicolor* Raddi	Short with convex brown cap, yellow stipe, olive brown ellipsoidal spores, and no bluing reaction when cap was removed	Grassy soil	35.101° N -92.441° W	HXC5813	KF986268	557 bps
J	*Agaricus bisporus* (J.E. Lange) Imbach	Dome shaped off-white caps, white flesh and stipe, brown gills, brown spores	Commercially cultivated	N/A	HXC5815	KF986267	670 bps

to a final volume of 40 µl. The concentration of the isolated DNA was measured via absorbance using a NanoPhotometer P-Class (Implen, West Lake Village, CA, USA).

We used PCR to amplify about 700 bp of the *ITS* (*ITS1*, *5.8S*, *ITS2*). Primers (IDTdna, Coralville, IA, USA) were designed to be fungal-specific for *18S* (ITS1-F: CTT GCT CAT TTA GAG GAA GTA A; Gardes and Bruns 1993) and *25S* regions (ITS4: TCC TCC GCT TAT TGA TAT GC; White et al. 1990). Each 50 µl PCR reaction contained 1X Bullseye Red Taq DNA Polymerase buffer (Midsci, Valley Park, MO, USA), 0.5mM each primer, and from 30 ng to 150 ng of DNA. The thermocycler settings were as follows: denaturation at 95ºC for 3 minutes; 35 cycles of denaturing at 94ºC for 30 seconds, annealing at 55ºC for 40 seconds, and extension at 72ºC for 50 seconds; and a final extension at 72ºC for 7 minutes.

We ran 3 µl of each PCR product and a ladder designed for approximate quantification (GeneRuler 100 bp, Thermo Fisher Scientific, Pittsburgh, PA, USA) with SYBR green loading dye (1:1000; Life Technologies, Carlsbad, CA, USA) on a 1% agarose gel in sodium borate buffer at 200V for 35 minutes. We photographed gels under UV light to confirm the success of PCR, to estimate amplicon length, and to estimate the quantity of PCR product. We purified PCR products using the QIAquick PCR purification kit (Qiagen) according to the manufacturer's instructions but eluted to a final volume of 32 µl. For one sample with 2 bands, the brightest band was cut from the gel using a razor blade on a UV light table. DNA from the gel slice was purified using the QIAquick Gel Extraction kit (Qiagen). About 20 ng of purified PCR product and 20 pmol of primer were submitted to the DNA Core Facility at the University of Arkansas for Medical Sciences (Little Rock, AR, USA). Each sample was Sanger sequenced twice, once with the forward and once with the reverse PCR primer.

We edited the trace files using Geneious Pro software (vers. 6.1.7; Biomatters Ltd, Auckland, NZ) by trimming low quality ends of the forward and reverse sequences and aligning them to create a consensus sequence of double-stranded, confident reads. A few ambiguous base calls were manually edited to 'N' or the more appropriate base. We used each consensus sequence to search GenBank using MegaBLAST (NCBI 2014) with default parameters. The results of each MegaBLAST search were visualized using a Distance Tree of Results with the default options (Fast Minimum Evolution; Maximum Sequence Difference = 0.75). Trees were downloaded in Nexus format and nodes were collapsed and relabeled using FigTree (vers. 1.4.0; http://tree.bio.ed.ac.uk/software/figtree/). Consensus sequences were submitted to GenBank (Table 1).

Results

We were able to identify 6 collections to the species level (B, C, E, G, I, J) and 4 specimens to the genus level (A, D, F, H) based on morphology (Table 1). DNA isolation failed for sample F when a tube split during homogenization, but PCR was successful for the other 9 samples. Based on agarose gels, amplicons ranged from 550 to 1000 bp and were single-banded except for a faint second band in sample H. We did not submit sample H for sequencing because DNA was not recovered from the gel isolation. The trace files for 3 of 8 samples (A, B, D) showed non-specific amplification that did not support creation of a consensus sequence. Nucleotide sequencing was successful for the remaining 5 specimens (C, E, G, I, J; Table 1), and we created consensus sequences ranging from 444 to 720 bps in length that contained a maximum of 3 unknown base calls per sample.

Pairwise *ITS* similarities supported some of our morphological identifications and raised uncertainty for others. Specimen C, identified morphologically as *Trichaptum biforme*, was confirmed using a query length of 720 bps with 98% identity (95% query cover) to *T. biforme* (AM269815) and the 7 other most similar sequences (90 to 96% identity and 90 to 97% query cover) were also *T. biforme* (Fig. 1A). Specimen E was identified based on morphology as *Bondarzewia berkeleyi* (Arora 1986, Gilbertson and Ryvarden 1988). In contrast, the identity search using a query length of 444 bps found 14 entries from widespread taxonomic groups to all have 89% sequence identity (95 to 100% query cover), none of which were *B. berkeleyi* (Fig. 1B). This consensus sequence was shorter than our others, but only had 0.06% unclear base calls (3 in 444 bps). The morphological, ecological, and distributional features of specimen G suggested *Amanita jacksonii* (Arora 1986). The search using 643 bps found 3 GenBank accessions with 94% sequence identity and 89% query cover - *A. arkansana* H.R. Rosen (JX844674) and 2 species known only from the west coast of the U.S. (*A. calyptroderma* G.F. Atk. & V.G. Ballen (JX844696) and *A. vernicoccora* Bojantchev & R.M. Davis (JX844746)). In addition, there was an incomplete 299 bp *ITS* sequence with 99% sequence identity, *Amanita sp-AR01* (JX844754). Although this match yielded only 67% query cover to our search

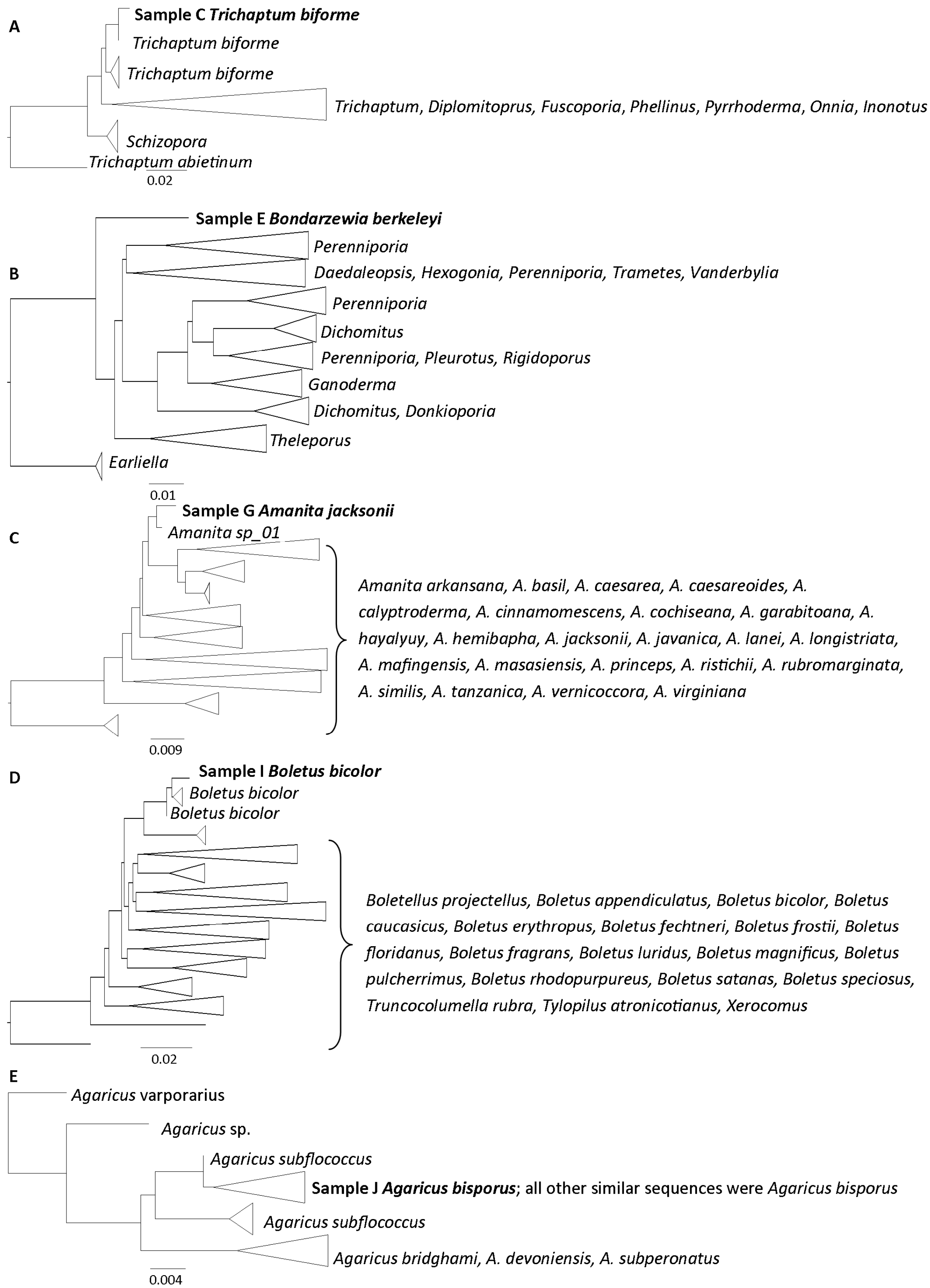

Figure 1. Distance trees showing relationship of each *ITS* nucleotide sequence to similar sequences in GenBank. A, *Trichaptum biforme*; B, *Bondarzewia berkeleyi*; C, *Amanita jacksonii*; D, *Boletus bicolor*; E, *Agaricus bisporus*.

sequence, it covered all of the 299 bp target sequence and our sample was sister to this unpublished taxon in the distance tree (Fig. 1C). Based on morphology, there were 2 candidate species for specimen I with geographic distributions that include central Arkansas - *Boletus bicolor* and *B. luridiformis* Rostk (*B. erythropus* Krombh.). Both species can have some of the colors observed in our specimen, although our sample did not turn blue when the cap was removed (a feature of *B. luridiformis*). The pairwise identity search showed 99% sequence identity (100% query cover) to *Boletus bicolor* (GQ166877) and the distance tree supported the *B. bicolor* accessions as sisters to our specimen (Fig. 1D). For the cultivated mushroom, pairwise identity was in agreement with morphology. For a query length of 670 bps, there was 99% sequence identity (99% query cover) to *Agaricus bisporus*, and this was consistent for the top ten matches.

Discussion

Our method was successfully executed by novice experimenters, suggesting that these are useful DNA isolation, PCR, purification, sequencing, and similarity search techniques. A major limitation in our study was that our experimental design did not budget for repeating failed samples. With only one attempt, 5 out of 10 samples (C, E, G, I, and J) produced nucleotide sequences that we were able to use for DNA barcoding (Table 1). One DNA isolation failure and one gel isolation failure would likely have been overcome by an experimental design that planned for repeating about 20% of the samples. Five of the 8 samples submitted for sequencing yielded a consensus sequence. Failure of nucleotide sequencing in 3 samples that exhibited apparently single-banded PCR products (A, B, D) may be attributed to intra-individual variation in the *ITS* region (Lindner et al. 2013). Because we did not clone our samples, length variation within the individual could create the symptoms we observed in our trace files. Better PCR amplification and sequencing of *ITS* in *Amanita* may be achieved with primer NS7 paired with ITS4 (Lim and Jung 1998). Primers AML1 and ITS4-B tend to produce better amplification in some other taxa (Gardes and Bruns 1993). Alternative DNA barcode markers might also be useful, e.g. the *28S* nuclear ribosomal large subunit rRNA gene (Schoch et al. 2012). We plan for future undergraduate laboratories to repeat 5 of our samples and to test other PCR primer combinations and/or marker combinations for those that fail in a second round.

The high similarity matches to the 5 samples that we obtained indicate that a robust set of *ITS* sequences are presently available in GenBank and that *ITS* currently allows identification to the species level in some cases (Schoch et al. 2012). However, interpretation of these similarity results is not always straightforward. In cases where highly similar database records suggest different species (or even genera), tools such as Distance Trees help visualize the results. We show a distance tree for each of our samples to allow comparison between those with clear results (Fig. 1A, 1D, 1E) with more complicated patterns (Fig. 1B, 1C). For the control sample (Fig. 1E), the assignment to *Agaricus bisporus* was trivial, as all of the most similar nucleotide sequences were assigned to this species. Our morphological determinations for *Trichaptum biforme* (Fig. 1A) and *Boletus bicolor* (Fig. 1D) were also strongly supported by DNA barcoding similarity. However, 2 of our collections raise intriguing questions regarding species identity and also beg the question of what taxonomic determination should be submitted to GenBank. The *ITS* similarities for sample G suggest that we have added another documented location for a putative new species of *Amanita*. Although distance tree relationships (Fig. 1C) and additional analysis of morphological features supported the affinity of our collection for this new species (Tulloss, *pers. comm.*), we used the *Amanita jacksonii* determination for our GenBank submission pending publication of the new species. The polypore (sample E) results are perplexing. Despite 89% identity to 30 samples in GenBank with 95-100% query coverage, our DNA barcode was not grouped within any clade. The morphologically determined *Bondarzewia* was not one of the 11 potential sister genera based on *ITS* similarity. We confirmed that there were *ITS* accessions in GenBank that have been identified as *B. berkeleyi*, but they were not highly similar to ours. Each of the *ITS*-similar genera appear to be highly unlikely candidates based on known geographic distribution or differing diagnostic characters. For example, the sister genus *Theleporus* (Fig. 1B) has only been reported from China; *Daedaleopsis confragosa* in known from Arkansas, but differs in color, spore surface, and substrate of dead wood; *Perenniporia tenuis* var. *pulchella* was reported from Arkansas on an oak, but is described as being bright yellow; *P. robinophila* is reported in Arkansas but mainly grows on dead *Robinia* sp. or on Moraceae stumps (Gilbertson and Ryvarden 1986). A new search in the UNITE database (Koljacg et al. 2013) during review of this paper did not offer any new reference sequences or new species hypotheses that explain this dilemma. The taxonomy of the polypores is unsettled (Riley et al. 2014), allowing several possible explanations for our results. The morphological characters that we used may not be definitive or may even be misleading, although it is possible that anatomical characters used in the polypores (Gilbertson and Ryvarden 1986) may be helpful. A third possibility is that there are cryptic polypore genera in Arkansas or that our southeastern U.S. species have been assigned to incorrect genera based on their morphology and anatomy. Although not supported by our molecular evidence, we contributed our *ITS* sequence to GenBank with a determination of *Bondarzewia berkelei*. We note that our precise geographic collection location and our voucher provide a means for mycologists to re-examine the specimen.

Broader use of DNA barcoding in fungal identification will be an important tool for the expansion and improvement of quality nucleotide sequence databases (Begerow et al. 2010). Based on the success rate of our study as well as a similar success rate reported by Osmundson et al. (2013) in a larger study, experiments that budgeted for re-runs, included alternative primers for *ITS*, and sequenced additional loci would likely show a successful DNA barcode identity for a majority of the specimens (Schoch et al. 2012). A central assumption is that these databases are already robust and reliable, but Nisson et al. (2006) concluded that not only do public databases not contain representatives of many fungal groups, roughly 20% of accessions are poorly annotated or misidentified. Several competing projects are underway that seek to remedy these problems by actively curating existing sequences and by including DNA barcodes for the type specimens for each fungal taxon (Koljacg et al. 2013, Schoch et al. 2014). Future similarity searches that use UNITE (Koljacg et al. 2013) and RefSeq (Schoch et al. 2014) can expect increasingly reliable DNA barcoding for fungi. However, concurrent morphological identification for fungi with sporocarps is needed in order to expand barcoding databases to include better geographic distributions. It is also important to note that while some literature is currently available for identifying fungi, (e.g. dichotomous keys and field guides), many of these do not include all local species or even all local genera (Arora 1986). Support for continuing development of a North American mycoflora (Bruns 2012) is also vital. Together, well-curated DNA barcode databases and better morphological documentation will facilitate the future identification of environmental samples lacking sporocarps. This process will be useful to analyze the true diversity of fungal species, particularly those that may never produce a fruiting body. Because our vouchered specimens are available in the Hendrix College Herbarium to support the 5 *ITS* sequences we have submitted to GenBank, we suggest that we have made a small contribution to documenting the fungi of central Arkansas and that small scale barcoding projects such as this one are feasible, even in an undergraduate setting.

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Low Speed Current Bearing Anti-force Waves

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Running Title: Low Speed Current Bearing Anti-force Waves

Abstract

For theoretical investigation of electrical breakdown of a gas, we apply a one-dimensional, steady profile, constant velocity, three-component (electrons, ions and neutral particles) fluid model. Our fluid model consists of the equations of conservation of mass, momentum and energy, coupled with the Poison's equation. The set of equations is referred to as the electron fluid dynamical equations (EFD). This investigation involves breakdown waves with a substantial current behind the wave front, and waves for which the electric field force on electrons is in the opposite direction of the wave propagation (anti-force waves – lightning return stroke). Therefore, the set of electron fluid dynamical equations need to be modified. For a low wave speed, we intend to find current values, and also the maximum current, for which solutions for our set of electron fluid dynamical equations become possible.

Introduction and Model

For anti-force waves the electric field force on electrons is in the opposite direction of the wave propagation; however, the electron gas pressure is considered to be large enough to provide the driving force. The leading edge of the wave is treated as a shock front followed by a thin dynamical transition region, referred to as the sheath region of the wave. Following the sheath region of the wave is a relatively thicker region, in which the electron gas cools down through further ionization of the heavy particles. This region is referred to as the quasi-neutral-region of the wave. In the sheath region, the electric field starting with its maximum value at the shock front reduces to zero at the trailing edge of the wave; and the electrons, starting with an initial speed at the wave front, slow down to speeds comparable to those of ions and neutral particles.

To analyze breakdown waves, we use the set of equations which were developed by Fowler et al. (1984). This set of equations describes pro-force waves and has proven to be successful (1984). The set of equations consists of the equations of conservation of mass, momentum, and energy plus the Poisson's equation, and they respectively are

$$\frac{d(n\upsilon)}{dx} = n\beta, \tag{1}$$

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

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

$$\frac{dE}{dx} = \frac{e}{\varepsilon_0}n(\frac{v}{V} - 1). \tag{4}$$

where n, v, T_e, e and m represent the electron number density, velocity, temperature, charge, and mass, respectively, and M, E, E_0, V, k, K, x, β and ϕ represent the 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.

To reduce the set of electron fluid dynamical equations to a non-dimensional form, Fowler et al. (1984), introduced the following set of dimensionless variables:

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

in which η, ν, ψ, θ, μ and ξ represent the dimensionless net electric field of the applied field plus the space charge field, electron number density, electron velocity, electron gas temperature, ionization rate, and position within the sheath region, while α and κ

represent wave parameters. Substituting these dimensionless variables in equations 1-4 yields

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

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

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

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

To solve for anti-force problems, we will use the set of non-dimensional variables developed by Hemmati (1999), in which all quantities including κ are positive and ξ is positive backward. The set of non-dimensional variables for anti-force waves are

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

For breakdown waves with a significant current behind the shock front, in addition to the Poisson's equation and equation of conservation of energy, the boundary condition on electron temperature at the shock front needs to be modified as well. For theoretical investigation of anti-force waves with a significant current behind the shock front, we will use Hemmati et. al's (2011) modified set of electron fluid dynamical equations.

$$\frac{d}{d\xi}[\nu\psi] = \kappa\mu\nu, \qquad (9)$$

$$\frac{d}{d\xi}[\nu\psi(\psi-1)+\alpha\nu\theta] = \nu\eta - \kappa\nu(\psi-1), \qquad (10)$$

$$\frac{d}{d\xi}[\nu\psi(\psi-1)^2 + \alpha\nu\theta(5\psi-2) + \alpha\nu\psi - \frac{5\alpha^2\nu\theta}{\kappa}\frac{d\theta}{d\xi} + \alpha\eta^2] = 2\eta\kappa\iota\alpha - \omega\kappa\nu[3\alpha\theta + (\psi-1)^2], \qquad (11)$$

$$\frac{d\eta}{d\xi} = \kappa\iota - \frac{\nu}{\alpha}(\psi-1). \qquad (12)$$

Where, with I_1 representing the current behind the shock front,

$$\iota = \frac{I_1}{\varepsilon_0 K E_0}, \qquad (13)$$

is the dimensionless current behind the wave front.

Results and Discussion

Uman and McLain (1970) derived an expression to calculate the current for stepped leader (pro-force waves) in lightning. Their calculated values for current were in the range of 800 to 5000 amperes. With optical observations and measuring currents at the lightning channel base, Rakov et al. (1998) reported a stepped leader current value of 5 kA and return stroke (anti-force) peak current value of 10 kA. In their study of lightning attachment processes in rocket-triggered lightning strokes, for return-strokes, Wang et al. (1999) reported a current peak value of about $12 \approx 21kA$. Determining K from experimental curves (McDaniel 1964), at a temperature of 10^5 K, K will be $2.4x10^9$ for helium and $9x10^9$ for nitrogen. In our formulas E_0, K, and β are scaled with electron pressure, P, and the applied fields are of the order of 10^5 V/m. For $I_1 = 10kA$, using the values of I_1, ε_0, E_0, and K, one can estimate the value of the dimensionless current, ι, which is on the order of one. Using helium-filled discharge tubes with different diameters, Asinovsky et al. (1994) measured breakdown wave speeds ranging from 10^7 m/s to $6x10^7$ m/s.

We use a trial and error method to integrate equations (9-12). For a given wave speed, α, at the wave front a set of values of wave constant, κ, electron velocity, ψ_1, and electron number density, ν_1, were selected and equations (9-12) were integrated with that set. The values of κ, ψ_1, and ν_1 were changed repeatedly in integrating equations (9-12), until the process leads to a conclusion in agreement with the expected conditions at the trailing edge of the wave.

Using Hemmati et al.'s (2011) modified electron temperature at the shock front,

$$\theta_1 = \frac{\psi_1(1-\psi_1)}{\alpha} - \frac{\kappa\iota}{\nu_1}, \qquad (14)$$

for several current values and for a relatively low wave speed, we have been able to integrate equations (9-12) through the sheath region of the wave. Our solutions meet the expected physical conditions at the trailing edge of the wave. However, for low wave speeds, integration of the set of equations became possible for lower current values only. For wave speed value of 5.93x10^6 m/s (α = 0.25), successful solutions required the following boundary values

$$\iota = 0.0, \kappa = 0.3883, \psi_1 = 0.96, \nu_1 = 0.985$$

$$\iota = 0.25, \kappa = 0.36115, \psi_1 = 0.9205, \nu_1 = 0.91405$$

$$\iota = 0.7, \kappa = 0.332, \psi_1 = 0.87, \nu_1 = 0.8342$$

$$\iota = 1.5, \kappa = 0.308, \psi_1 = 0.7805, \nu_1 = 0.723$$

Figure 1 is a graph of the electric field as a function of electron velocity. As the graph shows, dimensionless current value of 0.7 seems to be the maximum value for which solutions for the set of electron fluid dynamical equations come to a successful conclusion $(\eta_2 \rightarrow 0, \psi_2 \rightarrow 1)$.

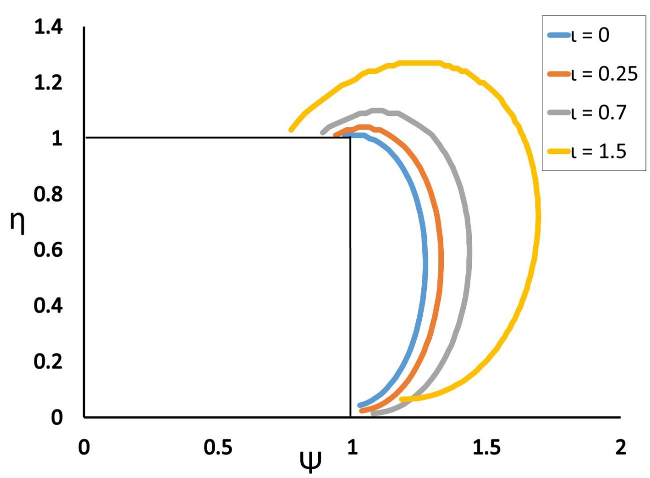

Figure 1. Electric field, η, as a function of electron velocity, Ψ, within the sheath region of current bearing anti-force waves for a wave speed value of α=0.25 and for current values 0, 0.25, 0.7 and 1.5.

Figure 2 is a graph of the electric field as a function of position within the sheath region of the wave. As the graphs show, for larger current values the sheath thickness becomes larger as well.

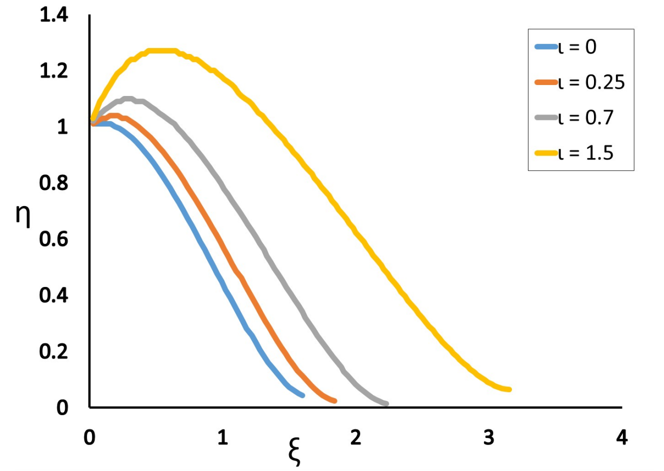

Figure 2. Electric field, η, as a function of position, ξ, within the sheath region of current bearing anti-force waves for a wave speed value of α=0.25 and for current values 0, 0.25, 0.7 and 1.5.

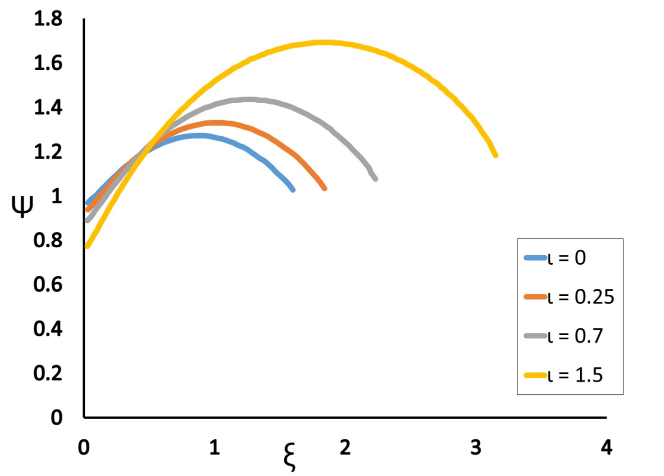

Figure 3. Electron velocity, Ψ, as a function of position, ξ, within the sheath region of current bearing anti-force waves for a wave speed value of α=0.25 and for current values 0, 0.25, 0.7 and 1.5.

Figure 3 is a graph of the dimensionless electron velocity as a function of dimensionless position within the sheath region of the wave. Figure 4 is a graph of the dimensionless electron number density as a function of dimensionless position within the sheath region of the wave. For current values for which solutions to the set of EFD equations become possible, 0.8 seems to be the average dimensionless electron number density within the sheath region of the wave. Dimensionless electron number density of 0.8 is equivalent to 8.85x10^{15} $elc\ /\ m^3$. In his fluid model simulations of a 13.56-MHz rf discharge, David Graves (1987) reports electron number density values between 5x10^{15} $/m^3$ and 2x10^{16} $/m^3$.

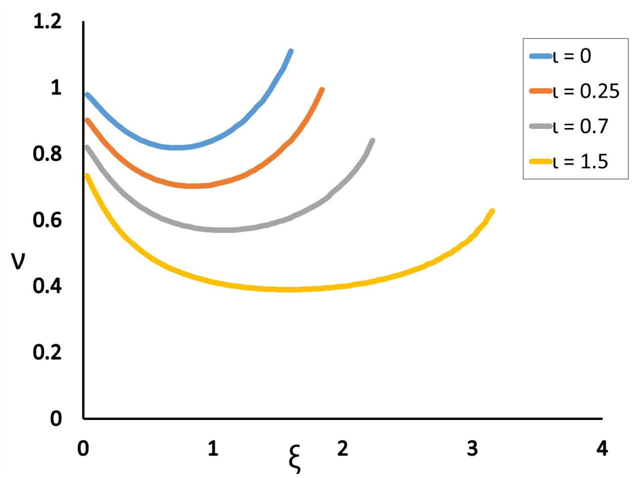

Figure 4. Electron number density, ν, as a function of position, ξ, within the sheath region of current bearing anti-force waves for a wave speed value of α=0.25 and for current values, 0, 0.25, 0.7 and 1.5.

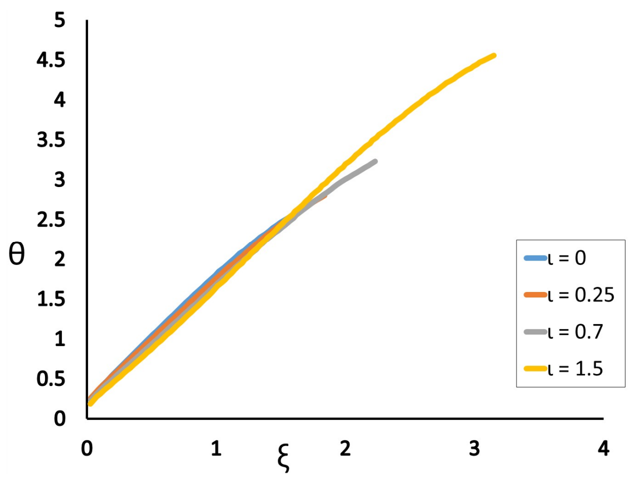

Figure 5. Electron temperature, θ, as a function of position, ξ, within the sheath region of current bearing anti-force waves for a wave speed value of α=0.25 and for current values 0, 0.25, 0.7 and 1.5.

Figure 5 shows a graph of the dimensionless electron temperature as a function of dimensionless position within the sheath region of the wave. Our average dimensionless electron temperature value of 2 is equivalent to an approximate temperature of $1.44x10^6$ K. For ionizing waves propagating counter to strong electric fields (anti-force waves), Sanmann and Fowler (1975) reported that the electron temperature increases rapidly away from the wave front until it reaches a peak value of around $3.17x10^7$ K.

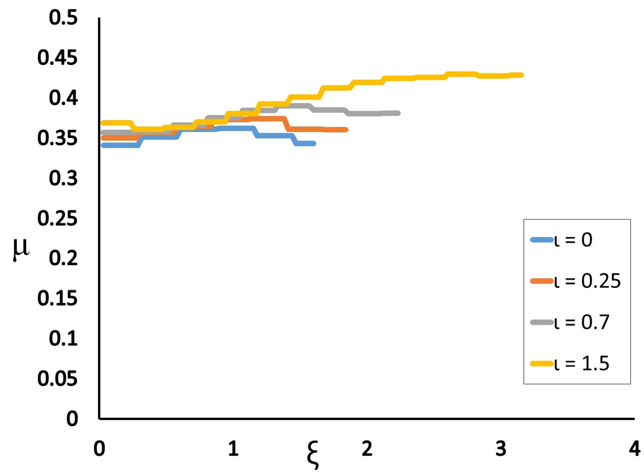

Figure 6. Ionization rate, μ, as a function of position, ξ, within the sheath region of current bearing anti-force waves for a wave speed value of α=0.25 and for current values 0, 0.25, 0.7 and 1.5.

Figure 6 is a graph of the dimensionless ionization rate as a function of dimensionless position with the sheath region of the wave. Earlier studies considered the ionization rate to be a function of temperature only; however, in our numerical integration of the set of electron fluid dynamical equations, the ionization rate was calculated considering both random and directed motion of the electrons (Fowler 1983). The graphs indicate that the ionization rate remains almost constant at the beginning of the sheath; however, it varies slightly as we traverse through the sheath.

Conclusions

For low wave speeds, solutions for the set of electron fluid dynamical equations became possible for smaller current values only. For low wave speeds, integration of the set of electron fluid dynamical equations becomes very time consuming and difficult. However, for larger current values, the sheath thickness becomes larger and makes the integration of the set of equations even harder. However, for wave speed value of $5.93x10^6$ m/s ($\alpha = 0.25$), dimensionless current value of 0.7 ($I_1 \leq 10kA$) seems to be the cut-off point for current. For fast moving waves, $\nu = 10^8$ *m/s*, we have been able to find solutions for dimensionless current values as high as 10. Our results are in good agreement with the results reported by other investigators; this is another confirmation of the validity of our set of electron fluid dynamical equations and boundary conditions.

Acknowledgement

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Miscellaneous Fish Helminth Parasite (Trematoda, Cestoidea, Nematoda, Acanthocephala) Records from Arkansas

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Running Title: Fish Helminth Parasite Records

Abstract

Between June 2012 and January 2014, 147 fishes (10 species) within five families were collected from watersheds in 8 counties of Arkansas and examined for helminth parasites. Almost every fish species examined harbored at least one or more helminth parasite, including 5 trematodes (*Alloglossidium* sp., *Plagioporus* sp., *Crepidostomum* sp., *Clinostomum marginatum* and unknown metacercaria), 2 cestodes (unknown cyclophyllidean cysticerci and *Corallotaenia parva*), 3 nematodes (*Spiroxys* sp., *Capillaria catostomi*, and *Eustrongylides* sp.), and 3 acanthocephalans (unknown cystacanths, *Neoechinorhynchus* sp., and *Leptorhynchoides* sp.). We document 16 new host and 2 new distributional records for these helminths. In addition, this is the first time any helminth has been reported from the Blackspot Shiner, *Notropis atrocaudalis* and Caddo Madtom, *Noturus taylori.*

Introduction

Reports on helminth parasites of non-game fishes are mostly lacking in North America (Scholz and Choudbury 2014), with an obvious paucity of reports from Arkansas. Of the approximately 200 non-game fishes reported from the state (Robison and Buchanan 1988), we are aware of only 6 species that have been surveyed for helminth parasites in general (Cloutman 1976, Fiorello et al. 1999, McAllister et al. 2014a, b). Although there are fragmentary studies reporting monogenes from Speckled and Rainbow Darters, *Etheostoma* spp. (Wellborn 1967, Wellborn and Rogers 1967), a comparative study on stonerollers, *Campostoma* spp. (Cloutman 1976), one of white grub in a minnow (Mitchell et al. 1982), descriptions of monogenes from shiners (see Cloutman 1994, 1995, 2011), reports of acanthocephalans and tapeworm in Pirate Perches, *Aphredoderus sayanus* (McAllister and Amin 2008, McAllister et al. 2012, respectively), a study of black-spot disease in various fishes (McAllister et al. 2013), and helminths of Banded Sculpins, *Cottus carolinae* and madtoms, *Noturus* spp. (McAllister et al. 2014, 2015, respectively), studies on freshwater fish parasites in Arkansas are lacking. Here we report some new host and distributional records for helminth parasites of select fishes of the state.

Materials and Methods

Between June 2012 and January 2014 the following 147 fishes were collected from watersheds in eight counties of Arkansas (Fig. 1) and examined for helminth parasites (sample sizes in parentheses): **APHREDODERIDAE**: *A. sayanus* (21); **CYPRINIDAE**: *N. atrocaudalis* (10); **ICTALURIDAE**: Black Bullhead, *Ameiurus melas* (11), Yellow Bullhead, *Ameiurus natalis* (31), Ozark Madtom, *Noturus albater* (6), Tadpole Madtom, *Noturus gyrinus* (7), Ouachita Madtom, *Noturus lachneri* (20), *N. taylori* (16); **ELASSOMATIDAE**: Pygmy Sunfish, *Elassoma zonatum* (5); **COTTIDAE**: Knobfin sculpin, *Cottus immaculatus* (20). Fishes were collected with backpack electrofishers, dipnets or seines. They were placed in habitat water and necropsied within 24 h. We followed accepted guidelines for the use of fish in research (AFS 2004) and specimens were overdosed with a concentrated Chloretone solution, measured for total length (TL) and a mid–ventral incision from anus to stomach was made to expose the gastrointestinal tract and other internal viscera (including gallbladder) which was removed and placed in a Petri dish containing 0.6% w/v saline. Their gills/gill filaments were not examined for monogenes. Trematodes and cestodes were stained with acetocarmine and mounted in Canada balsam or Kleermount®. Nematodes and Acanthocephalans were placed on a slide with glycerol and studied as temporary mounts. Voucher specimens of parasites were deposited in the United States National Parasite Collection (USNPC), Beltsville, Maryland or Harold W. Manter Collection (HWML), University of Nebraska, Lincoln. Host voucher specimens preserved in 10% v/v formalin, transferred to 40% v/v ethanol, and deposited in the Henderson State University Museum (HSU), Arkadelphia, Arkansas as HSU 3540-3543, 3545, 3555-3556. Prevalence, mean intensity, and range of infection are provided and are in accordance with terminology given in Bush et al. (1997).

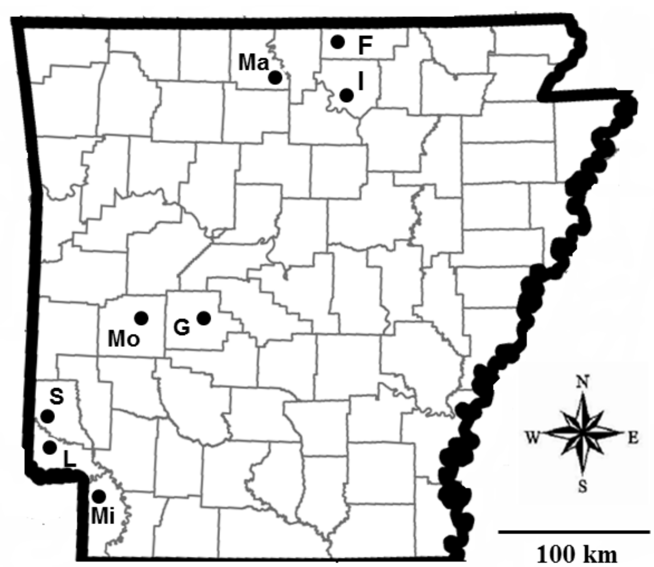

Figure 1. Localities (dots) in eight counties of the state where fish were collected. Abbreviations: Fulton (F), Garland (G), Izard (I), Little River (L), Marion (Ma), Miller (Mi), Montgomery (Mo), Sevier (S).

Results and Discussion

Every fish species examined, except 20 *C. immaculatus* (Spring River, Fulton County), harbored at least one or more helminth parasite. Seventy-three of 141 (52%) of the fishes examined were infected. The following is an annotated list of data as follows: host and total length (TL), prevalence, intensity, total length of host, collection site, collection date, USNPC accession number.

PLATYHELMINTHES: TREMATODA
Digenea: Plagiorchiida: Macroderoididae
***Alloglossidium* sp. (Fig. 2)**

Noturus gyrinus, 60 mm TL, 1/7 (14%), 5 worms, Sevier Co., Rolling Fork River, 24 Oct. 2013.

Ameiurus natalis, 103 mm TL, 1/6 (17%) 2 worms, Sevier Co., Rolling Fork River, 24 Oct. 2013.

Figure 2. Gravid *Alloglossidium* sp. (unstained) with expelled ova from *Noturus gyrinus*; inset shows single ovum with operculum (arrow).

Six (35%) of the 17 recognized species of *Alloglossidium* have been reported from fishes (mainly ictalurids), including *Ameiurus* spp., *Ictalurus* spp., and *Noturus* spp. (see Smythe and Font 2001, Tkach and Mills 2011, Tkach et al. 2013). In addition, Kasl et al. (2014) recently reported *A. floridense* from *N. gyrinus* from Florida.

Species of *Alloglossidium* from fishes have been reported from Arkansas, California, Colorado, Florida, Georgia, Kansas, Kentucky, Idaho, Illinois, Indiana, Louisiana, Maine, Massachusetts, Michigan, Minnesota, Mississippi, Missouri, Nebraska, New York, North Dakota, Ohio, Oklahoma, North Dakota, Texas, Virginia, and Wisconsin and Ontario, Canada (Hoffman 1999; Tkach and Mills 2011, Kasl et al. 2014, McAllister et al. 2015).

Alloglossidium corti (Lamont, 1921) Van Cleave and Mueller, 1932 has been reported in *A. natalis*, channel catfish, *Ictalurus punctatus* (Becker and Houghton 1969) and *N. lachneri* (Fiorillo et al. 1999), with metacercaria in the antennal gland of some crayfishes from the state (McAllister et al. 2011). As yet, there are no additional species of *Alloglossidium* reported from Arkansas. Our specimens do not fit any previous description of *Alloglossidium* and is most likely a new species that we are investigating in an on-going morphological and molecular study (V.V. Tkach, *pers. comm*).

Opecoelidae, *Plagioporus* sp. (Fig. 3)

Noturus lachneri, 60.9 ± 11.2, 43-82 mm TL, 17/20 (85%), 2.0 ± 0.9, range 1-4 worms, Garland Co., Hot Springs, Middle Branch Gulpha Creek off E Grand Avenue (34.5092°N, 93.009039°W). 26 Oct. 2013.

Figure 3. *Plagioporus* sp. (unstained) from *Noturus lachneri*; abbreviations: testes (T), ovary (O).

McAllister et al. (2014, 2015) recently reported *Plagioporus* sp. from *C. carolinae* and Slender Madtoms, *Noturus exilis* from Arkansas, respectively. Interestingly, except for the report by Harms (1959) of *Plagioporus* sp. in Black Bullheads, *Ameiurus melas* from Kansas, our specimens reported herein from *N. lachneri* and those of McAllister et al. (2015) from *N. exilis* are the only other specimens of the genus reported from North American ictalurids and are likely a new species; DNA and morphological analyses are ongoing (T.J. Fayton, *pers. comm.*).

Allocreadiidae, *Crepidostomum* sp. (Fig. 4)

Aphredoderus sayanus, 59.6 ± 5.2, range 53-67 mm TL, 5/9 (56%), 3.4 ± 1.5, range 1-5 worms, Sevier Co., Rolling Fork River (34.064667°N, 94.380023°W), 24 Oct. 2013.

Figure 4. *Crepidostomum* sp. from *Aphredoderus sayanus*. Abbreviations: oral sucker (O), ventral sucker (V).

To date, 3 species of *Crepidostomum* have been reported from *A. sayanus*: *C. farionis* (Müller, 1784) Nicoll, 1909, *C. isostomum* Hopkins, 1931, and an unidentified immature *Crepidostomum* sp. (see

Hoffman 1999). In addition, *Crepidostomum* sp. has been reported from *A. natalis* and *I. punctatus* from Lake Fort Smith, Arkansas (Becker and Houghton 1969). Further, at least 25 other genera of fishes have been reported as hosts of this digene from Alabama, Arkansas, Georgia, Illinois, Kentucky, Louisiana, Maine, Massachusetts, Michigan, Mississippi, New York, North Carolina, North Dakota, Oklahoma, Ohio, Pennsylvania, Oregon, Tennessee, Texas and Wisconsin and Ontario and Quebec, Canada (Hoffman 1999, Muzzall and Whelan 2011, summarized by McAllister et al. 2014). McAllister et al. (2014) recently reported *C. cooperi* from *C. carolinae* from Arkansas. Our specimens will be identified following DNA analyses (V.V. Tkach, *pers. comm*.).

Strigeidida: Clinostomidae
***Clinostomum marginatum* Rudolphi, 1819 (metacercaria) (Fig. 5)**

Noturus albater, 74 mm TL, 1/6 (17%), 1 worm, Marion Co., Crooked Creek at Kelly's Slab (36.245207°N, 92.715611°W), 26 Jul. 2013.

Noturus taylori, 67 and 74 mm TL, 2/16 (13%), 2 and 6 worms, Montgomery Co., Caddo River, 26 Oct. 2013. USNPC 107670.

Clinostomum marginatum is a very common trematode that is cosmopolitan in distribution and, according to Hoffman (1999), it is "likely capable of infecting any species of freshwater fish." Indeed, yellow grub has been commonly reported primarily from Arkansas game fishes (*Micropterus* spp.), including those from Crooked Creek (Daly et al. 2002) and the Caddo River (Daly et al. 1999). However, this is the first time *C. marginatum* metacercaria have been reported from *N. albater* and *N. taylori.*

Unknown digene metacercaria (Fig. 6)

Aphredoderus sayanus, 57 mm TL, 1/9 (11%), Sevier Co., Rolling Fork River (34.064667°N, 94.380023°W), 24 Oct. 2013.

Unknown metacercaria of a digene trematode was found in the mesenteries of *A. sayanus*. Numerous metacercaria were found as spheroidal to ovoidal cysts (Fig. 6). This is the first time metacercaria have been reported from *A. sayanus*.

Figure 5. Metacercaria of *Clinostomum marginatum* from *Noturus taylori*. (A) Lateral side of madtom showing two encysted metacercaria (arrows) in dermis; scale bar = 10 mm. (B) Unstained metacercaria extracted from cyst showing typical morphology; scale bar = 1 mm.

CESTOIDEA

Cyclophyllidea: unidentified cysticerci (Fig. 7)

Ameiurus melas, 55 mm TL, 1/11 (9%), too numerous to count, Little River Co., Little River oxbow (33.908447°N, 94.396119°W), 24 Oct. 2013.

Noturus lachneri, 48, 59, and 64 mm TL, too numerous to count, 3/20 (15%), Garland Co., Hot Springs, Middle Branch Gulpha Creek off E Grand Avenue (34.5092°N, 93.009039°W), 26 Oct. 2013. USNPC 107690.

Noturus taylori, 71 mm TL, 1/16 (6%), 6 worms, Montgomery Co., Caddo River (34.455676°N, 93.714543°W), 26 Oct. 2013.

Notropis atrocaudalis, 55, 59, 66 mm TL, 3/10 (30%), too numerous to count, Miller Co., Nix Creek at Texarkana (33.444116°N, 94.016049°W), 3 Jan. 2014.

Elassoma zonatum, 5/5 (100%), too numerous to count, Miller Co., Nix Creek at Texarkana (33.444116°N, 94.016049°W), 3 Jan. 2014.

Cyclophyllidean tapeworms have not been previously reported from any of these hosts (above); therefore, we document 5 new host records for these cyclophyllidean cysticerci.

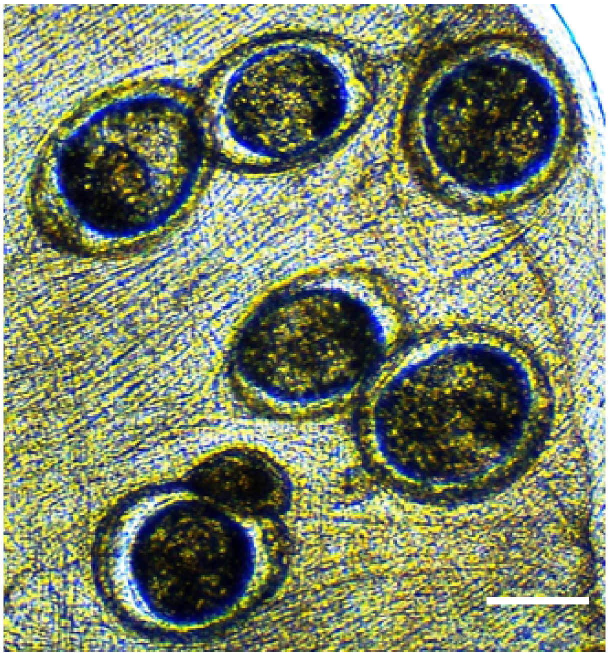

Figure 6. Unknown digene metacercaria from *Aphredoderus sayanus*; scale bar = 50 µm.

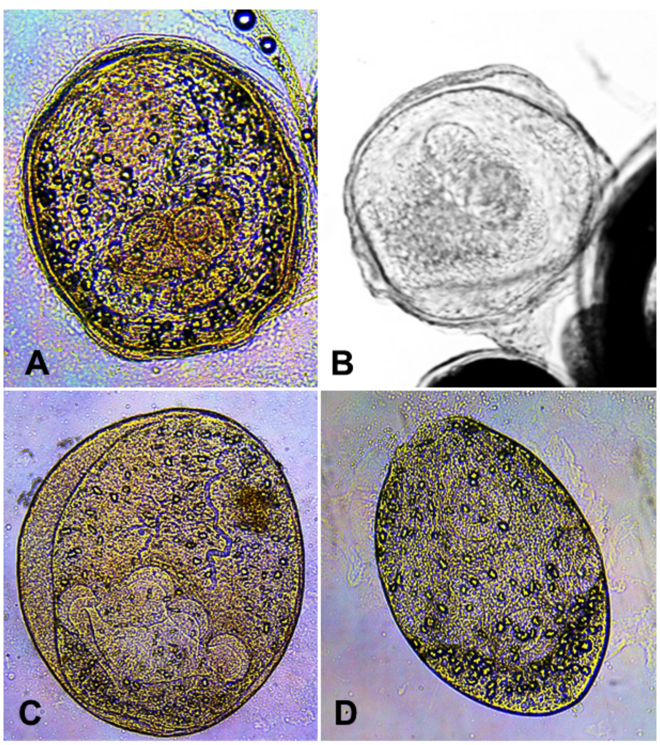

Figure 7. Tapeworm cysticerci from Arkansas fishes. (A) Specimen from *Noturus lachneri*. (B) Specimen from *Noturus taylori*. (C) Specimen from *Notropis atrocaudalis*. (D) Specimen from *Elassoma zonatum*.

Proteocephalidea: Proteocephalidae
***Corallotaenia parva* (Larsh, 1941) Frese, 1965**

Ameiurus melas, 58, 58 and 62 mm TL, 3/11 (27%), Little River Co., Little River oxbow (33.908447°N, 94.396119°W), 24 Oct. 2013, USNPC 107688-107689.

McAllister and Bursey (2011) recently reported *C. parva* from *A. melas* from Oklahoma. Other hosts include the brown bullhead, *Ameiurus nebulosus* and channel catfish, *Ictalurus punctatus* (Hoffman 1999). The range includes Colorado, Illinois, Maine, Michigan and Oklahoma (Hoffman 1999, McAllister and Bursey 2011), and now Arkansas (this report). We document a new distributional record for *C. parva* in the state.

NEMATODA

Spirurida: Gnathostomatidae, *Spiroxys* sp. (larvae) (Fig. 8)

Aphredoderus sayanus, 67 mm TL, 1/9 (11%), Sevier Co., Rolling Fork River (34.064667°N, 94.380023°W), 24 Oct. 2013. USNPC 107687.

Ameiurus natalis, 41.9 ± 14.8, range 30-75 mm TL, 13/23 (56%), Miller Co., Nix Creek at Texarkana (33.444116°N, 94.016049°W), 29 Jun. 2012. 6 Jul. 2012, 29 Sept. 2012.

Noturus lachneri, 59 mm TL, 1/20 (5%) with five worms, Garland Co., Hot Springs, Middle Branch Gulpha Creek off E Grand Avenue (34.5092°N, 93.009039°W), 26 Oct. 2013.

Noturus taylori, 55 mm TL, 1/16 (6%) Montgomery Co., Caddo River (34.455676°N, 93.714543°W), 26 Oct. 2013, USNPC 107671.

Species of *Spiroxys* from North American fishes have been reported from Arkansas (McAllister et al. 2014a), California, New York, North Dakota, Pennsylvania, West Virginia, Wisconsin, and Wyoming (see Hoffman 1999). The larval *Spiroxys* sp. that we found in intestinal mesenteries possessed the distinctly trilobed lips and triangular appearing anterior end (see Hedrick 1935, his figs. 2-6, our fig. 8B). Hoffman (1999) noted that *Spiroxys* sp. to be "very common in pond-reared fishes" (unidentified species) in Arkansas. In the experimental life cycle, the first intermediate host of *Spiroxys* was reported to be the crustacean, *Cyclops* sp. (Hedrick 1935). Larval *Spiroxys* sp. has been previously reported from *A.*

natalis (Hoffman 1999); however, we document 3 new host records and the first nematode, to our knowledge, ever reported from *A. sayanus*.

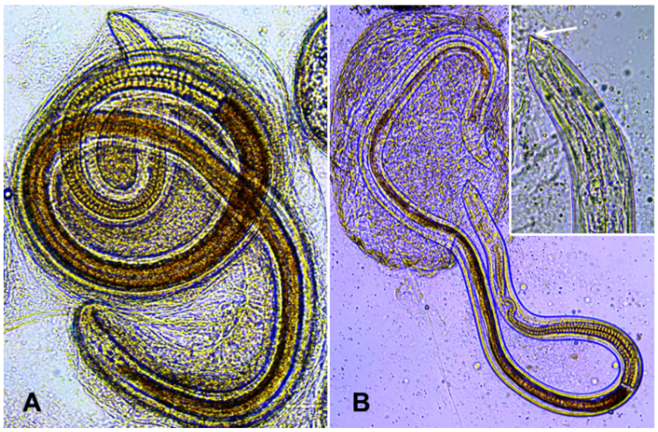

Figure 8. *Spiroxys* sp. larvae from intestinal mesenteries of madtoms. (A) Whole specimen from *Noturus lachneri*. (B) Whole specimen from *Noturus taylori* and anterior end of worm showing lips (inset, arrow).

Trichuroidea: Capillaridae, *Capillaria catostomi* Pearse, 1924

Aphredoderus sayanus, 78 mm TL, 1/3 (33%), 1 worm, Little River Co., Little River oxbow (33.908447°N, 94.396119°W). USNPC 107685.

Capillaria catostomi has been reported from suckers (*Catostomus* spp.) and carp *Cyprinus carpio*) from Florida and Wisconsin and Ontario, Canada (Hoffman 1999). In addition, Hoffman (1999) further noted "from 1962 to 1985, I saw *C. catostomi* from *Ctenopharyngodon idella*, *Lepomis cyanellus, L. macrochirus*, *Notemigonus chrysoleucus* and *Pimephales promelas* from West Virginia and Arkansas." However, we document a new host and the first genuine voucher of *C. catostomi* from Arkansas.

Dioctophymatoidea: Dioctophymatidae
***Eustrongylides* sp.**

Aphredoderus sayanus, 67 mm TL, 1/9 (11%), 1 worm, Sevier Co., Rolling Fork River (34.064667°N, 94.380023°W), 24 Oct. 2013, USNPC 107686.

Nematodes of the genus *Eustrongylides* are found as adults in the proventriculus of piscivorous birds with larvae encysted in the body cavity and musculature of fishes (Hoffman 1999). Early larval development occurs in oligochaetes (Lichtenfels and Stroup 1985). Specific identification of *Eustrongylides* requires rearing larvae in an avian host and our study did not include this experimental transmission. However, we document a new host record for this nematode and the first report of the genus from Arkansas.

ACANTHOCEPHALA

Unidentified cystacanth (Fig. 9)

Notropis atrocaudalis, 56, 60 mm TL, 2/10 (20%), too numerous to count, Miller Co., Nix Creek at Texarkana (33.444116°N, 94.016049°W), 3 Jan. 2014.

Ameiurus natalis, 46.1 ± 20.6, range 30-75 mm TL, 6/23 (26%), too numerous to count, Miller Co., Nix Creek at Texarkana (33.444116°N, 94.016049°W), 29 Jun. 2012, 6 Jul. 2012, 29 Sept. 2012.

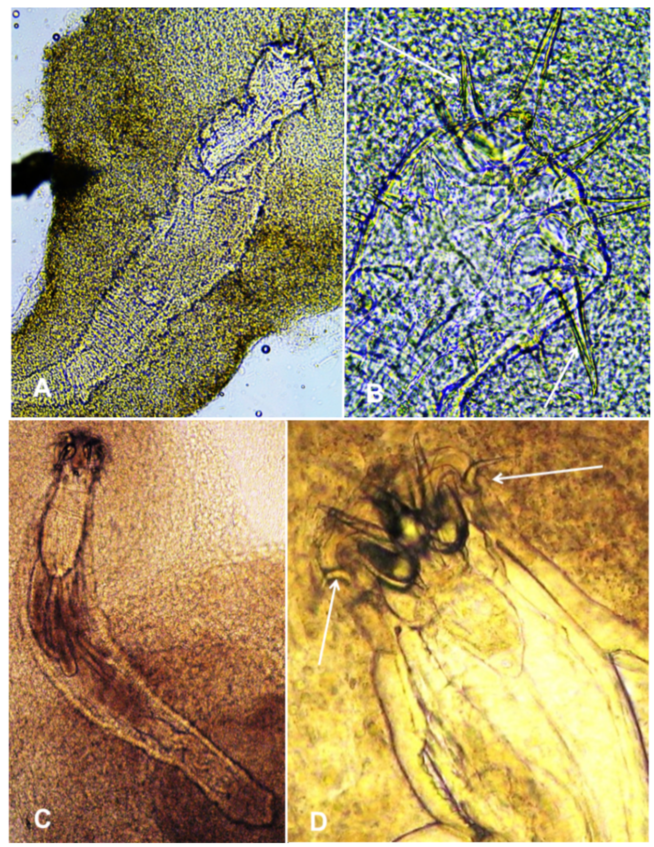

Figure 9. Acanthocephalan cystacanths from intestinal mesenteries of *Notropis atrocaudalis* and *Ameiurus natalis*. (A) View showing entire specimen from *N. atrocaudalis*. (B) Higher magnification of same showing proboscis and hook arrangement. (C) Entire specimen from *A. natalis*. (D) Higher magnification of same showing proboscis and hook arrangement (arrows).

The black-spot shiner and yellow bullhead are new hosts of acanthocephalan cystacanths and more importantly, we document the first helminth, to our knowledge, ever reported from *N. atrocaudalis.* Several genera of acanthocephalans have been previously reported from *A. natalis*, including *Leptorhynchoides*, *Neoechinorhynchus*, *Pilum*, and *Pomphorhynchus* (Hoffman 1999). Unfortunately, it is not possible to place our cystacanths within any genera.

Eoacanthocephala: Neoechinorhynchida: Neoechinorhynchidae, *Neoechinorhynchus* sp.

Aphredoderus sayanus, 4 worms, 61 and 67 mm TL; 2/9 (22%) Sevier Co., Rolling Fork River, (34.064667°N, 94.380023°W), 24 Oct. 2013, HWML 49932.

Two male and 2 immature female *Neoechinorhynchus* sp. were found in the intestinal tract of *A. sayanus*. There are no previous reports of this genus from Pirate Perches although *Neoechinorhynchus cylindratus* (Van Cleave, 1913) Van Cleave, 1919 has been reported from other Arkansas fishes (Hoffman 1999). Additional specimens are needed to determine a species identity. However, we document a new host record for the genus *Neoechinorhynchus*.

Palaeacanthocephala: Echinorhynchida: Rhadinorhynchidae, *Leptorhynchoides* sp.

Aphredoderus sayanus, 60 mm TL, 4 worms, 1/9 (11%), Sevier Co., Rolling Fork River (34.064667°N, 94.380023°W), 24 Oct. 2013, HWML 49931.

Leptorhynchoides aphredoderi was described by Buckner and Buckner (1976) from *A. sayanus* from Louisiana. In addition, *Leptorhynchoides thecatus* (Linton, 1891) Kostylew, 1924 was reported from basses (*Micropterus* spp.) from Arkansas (Becker et al. 1966). The lemnisci of *L. aphredoderi* is described as being "short, equal" (see Amin et al. 2013, their Table 1), a view consistent with our specimens. However, because we lack enough mature specimens, we cannot make a confident species diagnosis at this time. Therefore, additional specimens are needed to determine a species identity.

McAllister and Amin (2008) reported the acanthocephalans *Pomphyrhynchus lucyi* and *Aspersentis* sp. from *A. sayanus* from the Caddo River, Arkansas. We report 2 new host records for 2 acanthocephalans from *A. sayanus*.

In summary, we document 16 new host and 2 new distributional records for some helminth parasites of non-game fishes of the state. Most importantly, we have only begun to realize the diversity of fish helminths in Arkansas and future studies will undoubtedly report additional records, including descriptions of new taxa.

Acknowledgments

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A Comparative Study of Helminth Parasites of the Many-Ribbed Salamander, *Eurycea multiplicata* and Oklahoma Salamander, *Eurycea tynerensis* (Caudata: Plethodontidae), from Arkansas and Oklahoma

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Running Head: Helminths of *Eurycea* spp.

Abstract

Ninety many-ribbed salamanders, *Eurycea multiplicata* and 135 Oklahoma salamanders, *Eurycea tynerensis* were collected between April 2010 and April 2014 from 14 counties of Arkansas and McCurtain County, Oklahoma (*E. multiplicata* only) and examined for helminth parasites. Twelve (13%) *E. multiplicata* were infected, including two (2%) each with *Brachycoelium salamandrae*, *Bothriocephalus rarus*, *Batracholandros magnavulvaris*, *Cosmocercoides variabilis*, and *Omeia papillocauda*, and one (1%) each with an oligacanthorhynchid cystacanth and *Fessisentis vancleavei*. Forty-one (30%) of the *E. tynerensis* were infected, including seven (5%) with *Gorgoderina tenua*, two (1%) each with *Phyllodistomum solidum* and cyclophyllidean tapeworm cysticerci, one (0.7%) with *Cylindrotaenia americana*, six (3%) with *B. rarus*, eight (12%) with *Desmognathinema nantahalaensis*, 10 (7%) with *O. papillocauda*, two (1%) with *Amphibiocapillaria tritonipunctata* and six (4%) with *F. vancleavei*. We document 13 new host and two new distributional records for helminths of these salamanders. In addition, a summary of the endoparasites of *E. multiplicata* and *E. tynerensis* is provided.

Introduction

The many-ribbed salamander, *Eurycea multiplicata* ranges south of the Arkansas River and throughout the Ouachita Mountains of west-central Arkansas and adjacent southeastern Oklahoma (Trauth et al. 2004, Sievert and Sievert 2011). It is a metamorphic surface-dwelling plethodontid that frequents aquatic sites including abandoned mine shafts and spring seeps and can also be found under damp rocks and logs in deciduous forest. Although much has been published on the ecology of *E. multiplicata* (Dundee 1965, Trauth and Dundee 2005), little is published about its helminth parasites. McAllister and Bursey (2010) examined 66 *E. multiplicata* from Arkansas and Oklahoma and reported three species of nematodes.

The Oklahoma salamander, *Eurycea tynerensis* (Ozark gray-belly salamanders, *Eurycea multiplicata griseogaster* Moore and Hughes 1941 = *E. tynerensis* [sensu Bonett and Chippendale 2004]) ranges north of the Arkansas River in the state throughout the Ozark Highlands and westward to northeastern Oklahoma where it is found in cool springs, spring-fed creeks with cherty gravel bottoms and cave streams (Trauth et al. 2004). Likewise, a great deal has been published on the biology of this salamander (Dundee 1965, Ireland 1976, Cline et al. 1989, 1997, 2001, Tumlison et al. 1990a, b, Tumlison and Cline 2003, Bonett 2005, Emel and Bonett 2011, Martin et al. 2012, Connior et al. 2014) but less is available on its helminths. However, most studies are of a fragmentary nature including: Hughes and Moore (1943a,b) who described an acanthocephalan (*Fessisentis vancleavei*) and a monogenean (*Sphyranura euryceae*) from *E. tynerensis* from Cherokee County, Oklahoma; Malewitz (1956) reported *F. vancleavei* from specimens from Cherokee County; Buckner and Nickol (1978) provided a redescription of *Fessisentis vancleavei* (Acanthocephala) from *E. tynerensis* from Oklahoma; McAllister et al. (1991) reported *S. euryceae* from *E. tynerensis* from Arkansas; Bonett et al. (2011) reported on *Clinostomum marginatum* in *E. tynerensis* from Oklahoma; and McAllister et al. (2011) provided a study of *S. euryceae* from *E. tynerensis* from northeastern Oklahoma. In the most thorough survey to date, McAllister et al. (1995b) reported trematode, nematode, and acanthocephalan parasites from *E. tynerensis* (=*E. m. griseogaster*) from seven counties of the Arkansas River Valley. There have also been several unpublished theses on parasites of this salamander from Arkansas and Oklahoma, and while not mentioned specifically herein, they are referenced in McAllister et al. (1995b, 2011).

Here, we provide 13 new host and two new distribution records for some helminth parasites of *E. multiplicata* and *E. tynerensis*. In addition, a summary of their endoparasites is reported.

Materials and Methods

Between April 2010 and April 2014, 90 larval and adult *E. multiplicata* (mean ± snout-vent length [SVL] = 35.2 ± 6.6, 19-48 mm) were collected by hand or aquatic dip-net from (sample sizes in parentheses) Clark (4), Conway (21), Garland (1), Montgomery (21), Polk (1) and Saline (40) counties, Arkansas, and McCurtain (2) County, Oklahoma; 135 larval, paedomorphic and adult *E. tynerensis* ([SVL] = 41.5 ± 5.3, 23-53 mm) were collected in the same manner from Benton (3), Carroll (5), Cleburne (8), Franklin (5), Johnson (9), Marion (36), Searcy (68) and Washington (1) counties (Fig. 1).

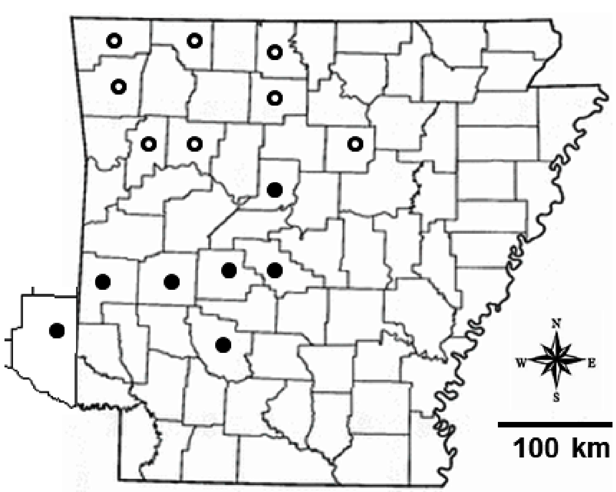

Figure 1. Arkansas and se corner of Oklahoma. Solid dots mark counties where *Eurycea multiplicata* were collected on the Ouachita Plateau/Arkansas River Valley; open dots mark counties where *Eurycea tynerensis* were collected on the Ozark Plateau.

Specimens were placed in habitat water on ice and taken to the laboratory for necropsy. Salamanders were killed by prolonged immersion in a dilute chloretone® (chlorobutanol) solution. If gills were present, they were examined for monogeneans under a stereomicroscope. A mid-ventral incision was made to expose the viscera and the entire gastrointestinal tract, liver, gall bladder, spleen, urinary bladder and gonads were examined for helminths. Trematodes and cestodes were stained with acetocarmine and mounted in Canada balsam, and nematodes and acanthocephalans were placed on a glass slide in a drop of undiluted glycerol for identification. Prevalence, mean intensity, and range of infection are provided and are in accordance with terminology given in Bush et al. (1997). Helminth voucher specimens were deposited in the United States National Parasite Collection (USNPC), Beltsville, Maryland. Host voucher specimens were deposited in the Arkansas State University Museum of Zoology, Herpetological Collection (ASUMZ) as ASUMZ 32605-32611 and 32616.

Results and Discussion

We found 13 helminths, including three trematodes, three cestodes, five nematodes, and two acanthocephalans. Six helminths were found in *E. multiplicata* and 10 helminths were harbored by *E. tynerensis*; four helminths were shared by both species. Twelve (13%) of the *E. multiplicata* and 41 (30%) of the *E. tynerensis* harbored at least one helminth. A detailed annotated list of the helminths recovered from *E. multiplicata* and *E. tynerensis* is presented below, with a Table summarizing all helminths reported from these two hosts.

TREMATODA

Digenea: Brachycoeliidae

Brachycoelium cf. *salamandrae* (Frölich, 1789) Dujardin, 1845. – We tentatively document *B. salamandrae* (Fig. 2) in three (3%) *E. multiplicata*. A larval specimen (35 mm SVL) from Cox Spring off St. Hwy. 8, Montgomery County (34.456421°N, 93.845254°W) had six worms and two adults (34, 38 mm SVL) from Shannon Hills, Saline County (34.60996°N, 92.43227°W) possessed two and three worms in their small intestine, respectively. McAllister et al. (1995) reported *B. salamandrae* from *E. tynerensis* from Conway County. Other hosts from Arkansas include six salamanders, four frogs/toads and a skink (McAllister 2013c, 2014). Interestingly, McAllister et al. (2014) recently noted they had serious doubts about Old World and New World *B. salamandrae* being conspecific (see summary by Bursey et al. 2012), and suggested caution with their former conclusions (McAllister et al. 2013d) and until a molecular approach was completed (V.V. Tkach, *pers. comm.*). However, regardless what species is eventually verified, this is a new host record for the genus *Brachycoelium*.

Figure 2. *Brachycoelium* cf. *salamandrae* from *Eurycea multiplicata*, Saline County, Arkansas. Note uterus (U) with ova; scale bar = 200 µm. Abbreviations: O (ovary); T (testes). Inset: two ova; scale bar = 25 µm.

Gorgoderidae

Phyllodistomum solidum Rankin, 1937. –Two (3%) *E. tynerensis* from 3 km S of Mull off Ramblewood Trail, Searcy County (36.059975°N, 92.59847°W) were infected with three and one *P. solidum*, respectively. Interestingly, the specimens (USNPC 108057) came from the intestinal tract of these salamanders, not the urinary bladder. This digenean has been previously reported from northern dusky salamander, *Desmognathus fuscus* from Illinois (Dyer 1986), New York (Goodchild 1943 [experimental infection]), North Carolina (Rankin 1937a, b) and Ohio (Groves 1945) and northern two-lined salamander, *Eurycea bislineata* from Ohio (Groves 1945). The life cycle involves fingernail clams (*Pisidium* sp.) as first intermediate hosts and dragonfly nymphs as second intermediate hosts (Goodchild 1943). Thus, we document a new host and a significant new geographic record for *P. solidum*.

Gorgoderina tenua Rankin, 1937 – Seven *E. tynerensis* (42.9 ± 3.3, 36-46 mm SVL) from 3 km S of Mull off Ramblewood Trail, Searcy County (36.059975°N, 92.59847°W) were infected with one to four (mean intensity = 1.4 ± 1.1) *G. tenua*. Rankin (1937a) described *G. tenua* from three-lined salamander (*Eurycea guttolineata*) from North Carolina. There are currently at least 52 recognized species of *Gorgoderina* Looss, 1902 with five from North American salamanders (*Ambystoma, Eurycea, Desmognathus, Pseudotriton, Necturus, Notophthalamus* spp.) (Mata-López et al. 2005). Rosen and Manis (1976) reported *Gorgoderina attenuata* (Stafford, 1902) Stafford 1905 and *Gorgoderina schistorchis* Steelman, 1938 from American bullfrog (*Lithobates catesbeianus*) and Red River mudpuppy (*Necturus maculosus louisianensis*) from Arkansas, respectively. This is only the second report of *G. tenua* since the original description and we document a new host and geographic record. Molecular analysis of the ITS2/28S region is ongoing (T.J. Fayton, *pers. comm.*).

CESTOIDEA

Cyclophyllidea

Two *E. tynerensis* (3%) from Panther Creek at Mull, Marion County (36.082643°N, 92.594726°W) harbored unknown cyclophyllidean tapeworm cysticerci in the mesenteries. Cysticerci were spheroidal to ovoidal and possessed calcareous corpuscles (USNPC 107940, Fig. 3). This is the first time cyclophyllidean tapeworm cysticerci have been reported from this salamander.

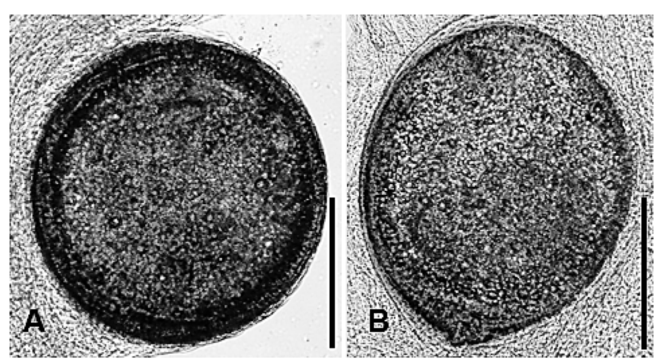

Figure 3. Unknown tapeworm cysticerci from *Eurycea tynerensis*, Marion County, Arkansas. (A) Spheroidal shape (B) Ovoidal shape. Scale bars = 25 µm.

Cyclophyllidea: Cylindrotaeniidae

Cylindrotaenia americana Jewell, 1916. – One (2%) *E. tynerensis* (40 mm SVL) from a spring seep S of Oark off St. Hwy. 103, Johnson County (35.5929°N, 93.584011°W) had nine immature cyclophyllidean tapeworms (USNPC 107961) in the small intestine that match the description of *C. americana* (Jewell 1916). Previous hosts from Arkansas include Ouachita dusky salamander, *Desmognathus brimleyorum*, western slimy salamander, *Plethodon albagula*, Ozark zig-zag salamander, *Plethodon angusticlavius* and bird-voiced treefrog, *Hyla avivoca* (see McAllister et al. 2013c). There are many other amphibian hosts of *C. americana* and its geographic range stretches north to south from Alberta, Canada to Uruguay, including 18 U.S. states, two provinces of Canada, Trinidad, Costa Rica, Mexico, and seven South American countries (see McAllister et al. 2013c). We document a new host record and the first report of this tapeworm in salamanders of the genus *Eurycea*.

Pseudophyllidea: Bothriocephalidae

Bothriocephalus rarus Thomas, 1937. – Two (2%) *E. multiplicata* (37, 38 mm SVL) collected from Shannon Hills, Saline County (34.60996°N, 92.43227°W) each harbored one worm, and six (4%) *E. tynerensis*, one (30 mm SVL) from Spavinaw Creek, Benton County (36.353059°N, 94.552347°W) and five (35.1 ± 4.5, 29-45 mm SVL) from 3 km S of Mull, Searcy County (36.059975°N, 92.59847°W) were infected with *B. rarus* (USNPC 107958, 107960) (Fig. 4) in their small intestines. Intensity of infection was 1.5 ± 0.9, 1-3 worms. This tapeworm has been previously reported from the dwarf salamander, *Eurycea quadridigitata* and dark-sided salamander, *Eurycea longicauda melanopleura* from Arkansas (McAllister and Bursey 2003, 2004) as well as several other salamanders from California, Kentucky, Michigan, Missouri, New Hampshire, Ohio, Pennsylvania, Tennessee, and West Virginia (see McAllister et al. 2013b). We document two new host records for *B. rarus*.

NEMATODA

Seuratoidea: Quimperiidae

Desmognathinema nantahalaensis Baker, Goater, and Esch, 1987. – Eight (12%) *E. tynerensis* (39.2 ± 5.6, range 28-47 mm SVL) harbored a total of 17 (mean intensity = 2.2 ± 1.9, range 1−6) *D. nantahalaensis* (USNPC 107937, 107941) in their small intestines. One salamander came from S of Oark off St. Hwy. 103, Johnson County (35.5929°N, 93.584011°W), one was collected from 3.2 km S of Cass off St. Hwy. 23, Franklin County (35.646329°N, 93.839612°W), one came from a wellhouse off St. Hwy. 59 N of Gentry, Benton County (36.299061°N, 94.450533°W), three were collected from Panther Creek at Mull, Marion County (36.082643°N, 92.594726°W) and two were taken 3 km S Mull, Searcy County (36.059975°N, 92.59847°W). The Oklahoma salamander (as *E. m. griseogaster*) and cave salamander, *Eurycea lucifuga* from Arkansas have previously been reported as hosts of this nematode (McAllister and Bursey 2004, McAllister et al. 1995a). In addition, *E. multiplicata* from Oklahoma is a host (McAllister and Bursey 2010) as well as *Desmognathus quadramaculatus* (type host) and *Desmognathus monticula* from North Carolina (Baker et al. 1987). Interestingly, the disjunct range of *D. nanthalaensis* includes only three states, Arkansas, North Carolina, and Oklahoma.

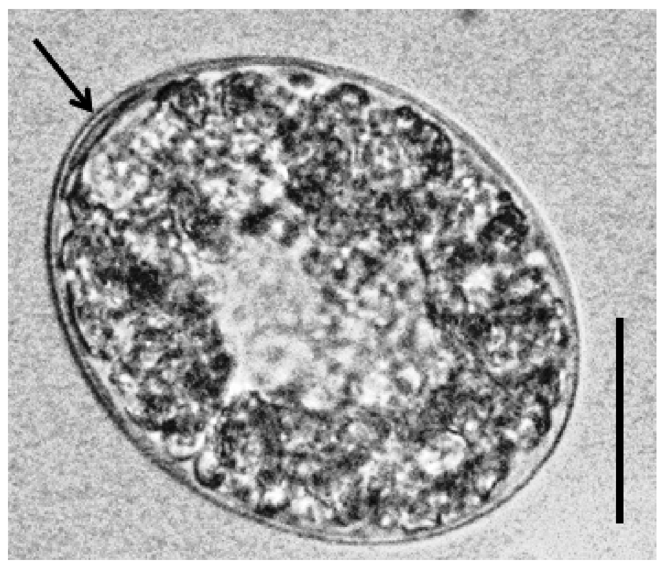

Figure 4. Ovum of *Bothriocephalus rarus* from *Eurycea multiplicata*, Saline County, Arkansas. Note operculum (arrow). Scale bar = 50 µm.

Omeia papillocauda Rankin, 1937. – Two (2%) *E. multiplicata* were infected, a single (35 mm SVL) specimen from Tanyard Springs, Conway County (35.115908°N, 92.916619°W) had one larval worm and one (32 mm SVL) salamander from 4 km NW of Caddo Valley and 1 km W of St. Hwy. 7, Clark County (34.215204°N, 93.095655°W) had one larval and two male *O. papillocauda*. In addition, 10 (7%) *E. tynerensis*, three (41-43 mm SVL) from Panther Creek at Mull, Marion County (36.082643°N, 92.594726°W) and seven (40.6 ± 7.2, 34-52 mm SVL) from 3 km S

Mull, Searcy County (36.059975°N, 92.59847°W) possessed a total of 26 (2.6 ± 3,1, 1-11) *O. papillocauda* (Fig. 5, USNPC 107938) in their stomachs. Many-ribbed salamanders from Arkansas have been previously reported as hosts of *O. papillocauda* (McAllister and Bursey 2010). It has also been reported from *D. brimleyorum* (McAllister et al. 1995d) and *D. monticola* from Arkansas (Connior et al. 2013). This nematode has also been reported from several other members of the genus *Eurycea* as well as *Desmognathus* and *Gyrinophilus* from Alabama, North Carolina, Ohio, and Tennessee (see McAllister and Bursey 2010). We document a new host record for *O. papillocauda* in *E. tynerensis*.

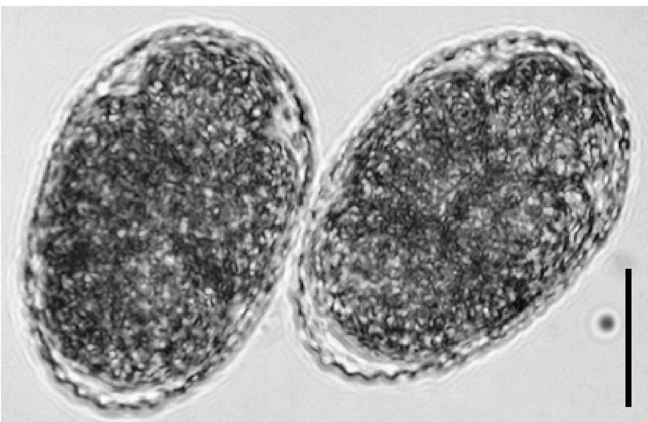

Figure 5. Embryonated ova of *Omeia papillocauda* from *Eurycea tynerensis*, Marion County, Arkansas. Note outer rugose shell. Scale bar = 25 µm.

Oxyurida: Pharyngodonidae

Batracholandros magnavulvaris (Rankin, 1937) Petter and Quentin, 1976. – Two (2%) *E. multiplicata* (43, 45 mm SVL) from Petit Jean State Park, Conway County (35.114642°N, 92.943574°W) each harbored one female pinworm (USNPC 107942) in its rectum. In addition, a single (3%) *E. tynerensis* (30 mm SVL) from Spavinaw Creek, Benton County (36.353059°N, 94.552347°W) had three female *B. magnavulvaris* (USNPC 107959) in the rectum. McAllister et al. (2013c) recently summarized records of *B. magnavulvaris* in caudate amphibians, including seven species of salamanders from Arkansas. There are four other members of the genus *Eurycea* reported as hosts of this oxyurid from Alabama, Michigan, North Carolina and Tennessee (see McAllister et al. 2013c). The many-ribbed and Oklahoma salamander are new hosts of *B. magnavulvaris*, which exhibits a direct life cycle (Anderson 2000).

Enoplida: Capillaridae

Amphibiocapillaria tritonispunctati (Diesing, 1851) Moravec, 1982. – Two (1%) *E. tynerensis*, one (43 mm SVL) from Lake Leatherwood, Carroll County (36.442033°N, 93.756562°W) and the other (30 mm larvae) from Savoy Cave, Washington County (36.109846ºN, 94.340588ºW) possessed six and one *A. tritonispunctati* (USNPC 107936) in their small intestine (Fig. 6). This nematode was previously reported from Arkansas in *E. spelaea* (McAllister et al. 2006) and *P. angusticlavius* (McAllister et al. 2013c). In addition, McAllister et al. (2013c) provided a summation of records of *A. tritonispunctati* in Nearctic and Palearctic amphibians of the world; only two members of the genus *Eurycea* have been previously reported as hosts of this worm and we add one more.

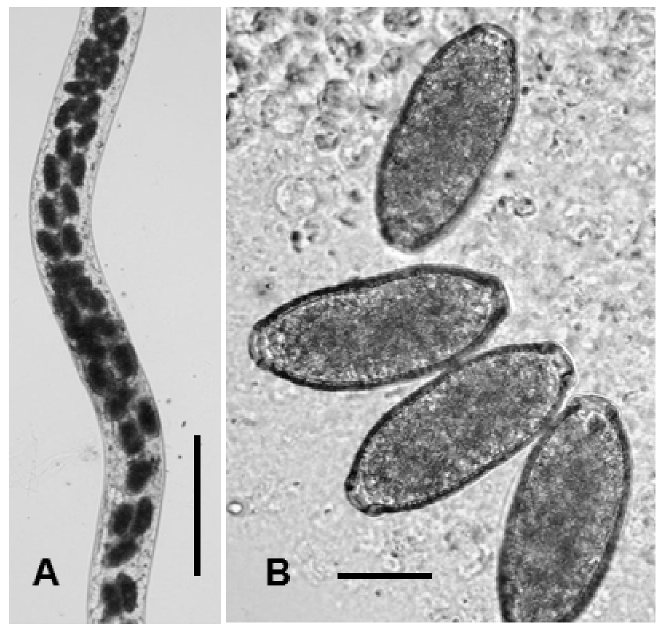

Figure 6. *Amphibiocapillaria tritonispunctati* from *Eurycea tynerensis*, Carroll County, Arkansas. (A) Gravid female showing numerous ova; scale bar = 100 µm. (B) Higher magnification of four individual ova from same showing typical capillariid morphology; scale bar = 25 µm.

Ascarida: Cosmocercidae

Cosmocercoides variabilis (Harwood, 1930) Travassos, 1931. – Two (2%) *E. multiplicata* (34, 37 mm SVL) from Shannon Hills, Saline County (34.60996°N, 92.43227°W) harbored one female and two male *C. variabilis* (USNPC 107939) in the large intestine, respectively. Previously, only female *Cosmocercoides* sp. was reported from this same host species and same population by McAllister and Bursey (2010); since only females were found it was not possible to assign their specimens to species. In

Arkansas, *C. variabilis* has been reported from ringed salamander, *Ambystoma annulatum*, *E. l. melanopleura*, *E. lucifuga*, Caddo Mountain salamander, *Plethodon caddoensis*, Rich Mountain salamander, *Plethodon ouachitae*, pickerel frog, *Lithobates palustris* and Cajun chorus frog, *Pseudacris fouquettei* (see McAllister et al. 2013a). Nematodes of this genus/species are common in both amphibians and reptiles and its range includes at least 24 U.S. states, four provinces of Canada, Mexico, Costa Rica and Panama (summarized by Bursey et al. 2012, McAllister et al. 2013d). We document a new host record for *C. variabilis*.

ACANTHOCEPHALA

Oligacanthorhynchidae (cystacanth)

An unknown oligacanthorhynchid cystacanth (USNPC 107944) was found in one (1%) *E. multiplicata* (39 mm SVL) from Petit Jean State Park, Conway County (35.114642°N, 92.943574°W). There is only one previous report of this parasite from an Arkansas salamander, the western slimy salamander, *Plethodon albagula* (McAllister et al. 1993). Juvenile stages of oligacanthorhynchid acanthocephalans have been found in other amphibians (Moore 1946, McAlpine 1996), reptiles (Elkins and Nickol 1983) and mammals (Radomski 1991). However, Elkins and Nickol (1983) and Bolette (1997) consider reptiles in these instances to be paratenic hosts and we believe salamanders are accidental or transport hosts acting as a trophic bridge between intermediate and definitive hosts. For those acanthocephalans parasitic in terrestrial animals, the intermediate hosts are usually insects (Nickol 1985). Salamanders are known to eat insects (Trauth et al. 2004) and thus, might be expected to become infected. We document a new host record.

Echinorhynchida: Fessisentidae

Fessisentis vancleavei (Hughes and Moore, 1943) Nickol, 1972 – Six (4%) *E. tynerensis* (47.3 ± 5.9, 40-55 mm SVL), one collected from S of Oark off St. Hwy. 103, Johnson County (35.5929°N, 93.584011°W), another from 3.2 km S of Cass off St. Hwy. 23, Franklin County (35.646329°N, 93.839612°W) and four from JFK Park, Little Red River, Cleburne County (35.512919ºN, 91.997125ºW) was found to harbor a total of 18 (3.1 ± 2.4, 1-7) *F. vancleavei* (USNPC 107962). In addition, one of two (50% [overall prevalence = 1%]) larval *E. multiplicata* (30 mm SVL) from Beavers Bend State Park off St. Hwy. 259A, McCurtain County, Oklahoma (34.113292°N, 94.708729°W) had a single acanthocephalan (USNPC 107943). *Fessisentis vancleavei* has been previously reported from *E. tynerensis* in Arkansas (Buckner and Nickol 1978, McAllister et al. 1995b) and Oklahoma (Hughes and Moore 1943a, Malewitz 1956). The life cycle of *Fessientis* spp. involves aquatic isopods as intermediate hosts (Buckner and Nickol 1979). *Eurycea multiplicata* is a new host of *F. vancleavei*.

This paper represents the second report of endoparasites of *E. multiplicata* and only the second thorough survey of *E. tynerensis* for helminths. We document 13 new host records for *E. multiplicata* and *E. tynerensis* and new distributional records for *P. solidum* and *G. tenua*. As noted by McAllister and Bursey (2010), the number of parasite species in *E. multiplicata* should increase with further study, and they did here by 50%, from three to six with our additional survey. Where comparisons are made, helminths shared by both salamanders include a trematode (*B. salamandrae*), a tapeworm (*B. rarus*), three nematodes (*B. magnavulvaris*, *D. nantahalaensis*, *O. papillocauda*) and an acanthocephalan (*F. vancleavei*), with most exhibiting prevalences < 5% (Table 1). Therefore, studies on these salamanders lend support to Aho's (1990) contention that caudate species are among the most depauperate hosts of all vertebrates. We suggest that future studies should include a larger sample size of *E. multiplicata* and *E. tynerensis* from a variety of localities in Oklahoma. In addition, if samples of the recently described and related Ouachita streambed salamander, *Eurycea subfluvicola* (Steffen et al. 2014) from Arkansas become available for study, it will be interesting to see if its helminth parasites are shared with *E. multiplicata* and *E. tynerensis*, particularly in areas of sympatry with the former.

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Table 1. Summary of helminth parasites of *Eurycea multiplicata* and *Eurycea tynerensis*.

Helminth	State	Prevalence*	Reference
		Eurycea multiplicata	
Trematoda			
Brachycoelium cf. *salamandrae*†	Arkansas	2/88 (2%)	This study
Cestoidea			
Bothriocephalus rarus†	Arkansas	2/88 (2%)	This study
Nematoda			
Batracholandros magnavulvaris†	Arkansas	2/88 (2%)	This study
Cosmocercoides sp.‡	Arkansas	3/61 (5%)	McAllister and Bursey (2010)
Cosmocercoides variabilis†	Arkansas	2/88 (2%)	This study
Desmognathinema nantahalaensis	Oklahoma	3/5 (60%)	McAllister and Bursey (2010)
Omeia papillocauda	Arkansas	1/61 (2%)	McAllister and Bursey (2010)
		2/88 (2%)	This study
Acanthocephala			
Fessisentis vancleavei†	Oklahoma	1/2 (50%)	This study
Oligacanthorhynchid cystacanth†	Arkansas	1/88 (1%)	This study
		Eurycea tynerensis	
Trematoda			
Brachycoelium cf. *salamandrae*	Arkansas	1/50 (2%)	McAllister et al. (1995)
Clinostomum marginatum	Oklahoma	9/74 (12%)	Bonett et al. (2011)
Phyllodistomum solidum†	Arkansas‖	2/135 (1%)	This study
Gorgoderina tenua†	Arkansas‖	7/135 (5%)	This study
Sphyranura euryceae	Arkansas	10/10 (100%)	McAllister et al. (1991)
		37/74 (50%)	McAllister et al. (2011)
	Oklahoma	45/90 (50%)	Moore and Hughes (1943b)
Cestoidea			
Bothriocephalus rarus†	Arkansas	6/135 (4%)	This study
Cylindrotaenia americana†	Arkansas	1/135 (0.7%)	This study
Unknown cysticerci	Arkansas	2/135 (1%)	This study
Nematoda			
Amphibiocapillaria tritonipunctati†	Arkansas	2/135 (1%)	This study
Batracholandros magnavulvaris†	Arkansas	1/135 (0.7%)	This study
Desmognathinema nantahalaensis	Arkansas	3/50 (6%)	McAllister et al. (1995)
		8/135 (6%)	This study
Omeia papillocauda†	Arkansas	10/135 (7%)	This study
Acanthocephala			
Fessisentis vancleavei	Arkansas	not given	Buckner and Nickol (1978)
		2/50 (4%)	McAllister et al. (1995)
		6/135 (4%)	This study
	Oklahoma	10/73 (14%)	Moore and Hughes (1943a)
		8/19 (42%)	Malewitz (1956)

*Prevalence = number infected/number examined (%). †New host record. ‡Only females; specific identity not possible. ‖New distributional record.

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A Binary Star Light Curve and Model of TYC 3670-588-1 From Professional-Amateur Collaboration

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Running Title: A Binary Star Light Curve and Model of TYC 3670-588-1

Abstract

We present the orbital light curve and model system parameters of a newly discovered eclipsing binary star in the constellation of Perseus.

Our professional-amateur astronomy collaboration between Arkansas Tech University (ATU), the Central Arkansas Astronomy Society (CAAS) and Whispering Pine Observatory, produced photometry in two wavelengths (Johnson V and R) in order to model the system for fundamental parameters with a binary modeling code.

We determined that this binary system contains two F-type stars orbiting each other with a short orbital period and having the following characteristics for the two components: mass ratio (q ~ 0.92), temperatures (T_1 ~ 7170 K, T_2 ~ 7350 K), sizes (R_1 ~ 1.7 R_{sun}, R_2 ~ 2.4 R_{sun}), orbital inclination (i ~ 77^o), and stellar separation (a ~ 8.3 R_{sun}).

The ephemeris for physical observations and primary mid-eclipse orbital phasing from the period in days is (HJD 2456488.885 + 1.292467 x N).

Introduction

This star, TYC 3670-588-1 (=NSVS 1748687, =AAVSO 000-BKF-917, =2MASS J01323010+ 5414055, =SDSS J013230.11+541405.5), had observations from the Northern Sky Variability Survey, NSVS (Wozniak et al 2004), the Sloan Digital Sky Survey, and the 2MASS survey, but was otherwise unremarkable within their errors and temporal sampling. It was discovered as having deep eclipses by Bruce McMath at the CAAS River Ridge Observatory after an unsuccessful attempt to use it as a constant comparison for the variable star IS Perseus that is found nearby.

The goal of this research was to photometrically observe this newly discovered binary at multiple wavelengths and model the system in order to obtain the fundamental stellar parameters for each component star. Observations of binary stars still represent the most dependable and accurate way of determining individual stellar characteristics of stars such as temperature, size and mass.

Observations and Methods

Photometric observations, carried out at both the CAAS River Ridge Observatory (RRO) near Bigelow, and at Whispering Pine Observatory (WPO) near Jasper, are listed in Table 1. The CAAS-RRO system consists of an 0.3m SCT and an SBIG ST-10 CCD camera with standard Johnson V and R filters. The ATU-WPO system consists of an 0.3m SCT with f6.3 focal reducer and an SBIG ST-9 CCD camera with standard Sloan g and r filters.

All image frames were calibrated in a standard way (Warner 2006) utilizing dark frames for removing CCD thermal noise, digitizing and readout noise errors and dome flat fields for removing CCD pixel sensitivity variances. Aperture photometry was then done on the calibrated star field images to extract differential magnitudes using a set of non-variable comparison stars using standard astronomy algorithms incorporated into Cmuniwin (Hroch 1998, Motl 2014). This software in short 1) converts images to FITS if necessary, 2) flat-fields and dark subtracts if desired, 3) processes to find stellar targets and photometrically measures them utilizing algorithms of DAOPHOT (Stetson 1987), 4) target lists pattern matched to identify stars in each image via the algorithm of Groth (1986), and 5) variable, comparison and check stars selected to generate differential photometry and light curves.

Table 1. Observation log for TYC3670-588-1 photometry.

Civil Date	JD-2450000.	Observatory	Filter
2013-AUG-27/28	6532	ATU-WPO	g
2013-AUG-28/29	6533	CAAS-RRO ATU-WPO	V r
2013-AUG-29/30	6534	ATU-WPO	g
2013-AUG-30/31	6535	ATU-WPO	r
2013-AUG-31/01	6536	CAAS-RRO ATU-WPO	V g
2013-SEP-02/03	6538	ATU-WPO	r
2013-SEP-03/04	6539	ATU-WPO	g
2013-SEP-04/05	6540	ATU-WPO	r
2013-SEP-05/06	6541	CAAS-RRO ATU-WPO	V g
2013-SEP-06/07	6542	ATU-WPO	r
2013-SEP-07/08	6543	CAAS-RRO ATU-WPO	V g
2013-SEP-08/09	6544	CAAS-RRO	V
2013-SEP-09/10	6545	CAAS-RRO ATU-WPO	V r
2013-SEP-10/11	6546	CAAS-RRO ATU-WPO	V g
2013-SEP-11/12	6547	CAAS-RRO ATU-WPO	V r
2013-SEP-12/13	6548		V
2013-SEP-13/14	6549	CAAS-RRO ATU-WPO	V g
2013-SEP-14/15	6550	ATU-WPO	r
2013-SEP-16/17	6552	CAAS-RRO	V
2013-SEP-17/18	6553	CAAS-RRO	V
2013-SEP-21/22	6557	CAAS-RRO	V
2013-OCT-06/07	6572	CAAS-RRO	V
2013-NOV-18/19	6615	CAAS-RRO	R
2013-NOV-19/20	6616	CAAS-RRO	R
2013-NOV-28/29	6625	CAAS-RRO	R

Results and Discussion

The resulting light curves, phased on the ephemeris, are shown in Figure 1. In general, the error bars are small, on the order of the size of the data points in the plot (~0.005-0.01 mag). There were some marginal nights with cirrus clouds, where the scatter is larger (i.e. V filter data near phase 0.6, R filter data near phase 0.9), but not used for fitting. Any well sampled eclipses that were captured, were analyzed to obtain the times of minima using the tried and true method of Kwee and van Woerden (1956). Four primary and five secondary eclipses were observed at several different wavelengths. The mid-eclipse times of minima are listed in Table 2.

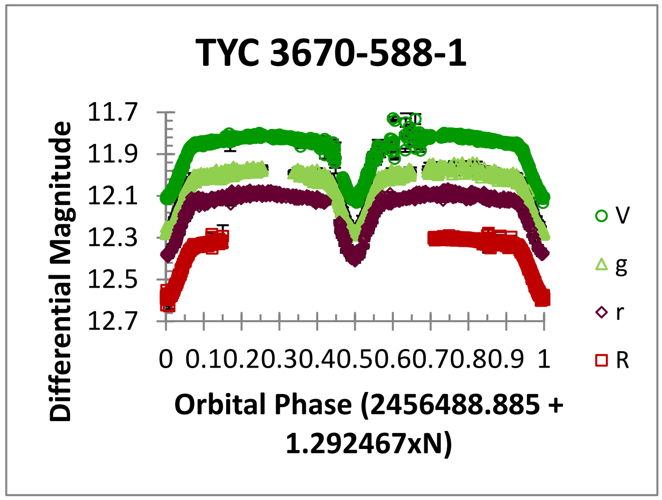

Figure 1. Orbital light curve at four wavelengths for TYC 3670-588-1 from CAAS-RRO and ATU-WPO. Observations in each filter have been arbitrarily offset in the vertical axis for clarity.

Table 2. Times of Primary and Secondary Mid-eclipse Minima.

HJD	Error	Cycle	Filter
2456536.707104	0.000403	37	g
2456536.708618	0.000301	37	V
2456541.876961	0.000123	41	V
2456541.877568	0.000209	41	g
2456550.923729	0.000236	48	r
2456572.896862	0.000210	65	V
2456534.766400	0.000190	35.5	g
2456539.935631	0.000230	39.5	g
2456543.815484	0.000205	42.5	g
2456547.691140	0.000151	45.5	r
2456552.861351	0.000174	49.5	V

The light curves were then examined to determine rough initial parameters for the system to facilitate full scale modeling. Using the colors both inside and out of eclipses as well as the eclipse depths and durations, suggested two similar F-type dwarf stars with a moderate orbital inclination.

We then took the initial parameters for such a binary system as a starting point and utilized a standard binary star code (Wilson and Devinney 1971) to compute models using the interface and scripting software PHOEBE (Prsa and Zwitter 2005) as a front end that can facilitate simultaneous fitting of up to 30 separate system parameters to the multicolor light curve data (see http://phoebe-project.org). Since PHOEBE does not yet recognize the newer Sloan filter band-passes, only the Johnson V and R data were used for the model fit.

The model parameters for the best fit solution are found in Table 3. The model parameters that were allowed to vary and then solved for by least squares are indicated in the table with their errors in (). The other physical parameters for the system were either set as fixed or calculated from the final best fit solution.

Table 3. Model Parameters for TYC3670-58801. An "*" indicates parameters that were allowed to vary and solved for in the least squares fitting process.

T0	Epoch (HJD-2456488.)	2456488.885
P	Orbital period (days)	1.292467
q	*Mass ratio (M_2/M_1)	0.92 (0.02)
i	*Orbital inclination (degrees)	77 (1)
a	*Orbital separation (R_{sun})	8.3 (1.2)
T1	*Star 1 effective temperature (K)	7175 (40)
T2	*Star 2 effective temperature (K)	7350 (40)
R1	*Star 1 radius (R_{sun})	1.72 (0.2)
R2	*Star 2 radius (R_{sun})	2.41 (0.3)
M1	Star 1 mass (M_{sun})	2.44
M2	Star 2 mass (M_{sun})	2.25
γ	*Center of Mass Velocity (km/s)	73 (17)
Ω1	*Star 1 surface potential	5.75 (0.6)
Ω2	*Star 2 surface potential	4.29 (0.55)
g1	Star 1 gravity brightening	0.32
g2	Star 2 gravity brightening	0.32
x1V	*Star 1 limb darkening (V filter)	0.487 (.015)
x2V	*Star 2 limb darkening (V filter)	0.489 (.015)
x1R	*Star 1 limb darkening (R filter)	0.389 (.015)
x2R	*Star 2 limb darkening (R filter)	0.389 (.015)
log g_1	Star 1 surface gravity	4.35
log g_2	Star 2 surface gravity	4.02

Figure 2 displays an example of the model goodness of fit versus the V filter light curve data. Data points in the figure are the observations (flux versus phase) and the WD-PHOEBE solution is the solid line. Also, in the bottom panels, are representations of the binary system model as seen from outside the binary orbit (Earth viewpoint) during the corresponding orbital phases.

These system model parameters are consistent with a binary containing two F-type stars, with spectral types of roughly F0 V + F0 V. Spectroscopic observations would help solidify this classification as well as tie down the individual masses via observation of radial velocities obtained from spectra lines of one or both stars.

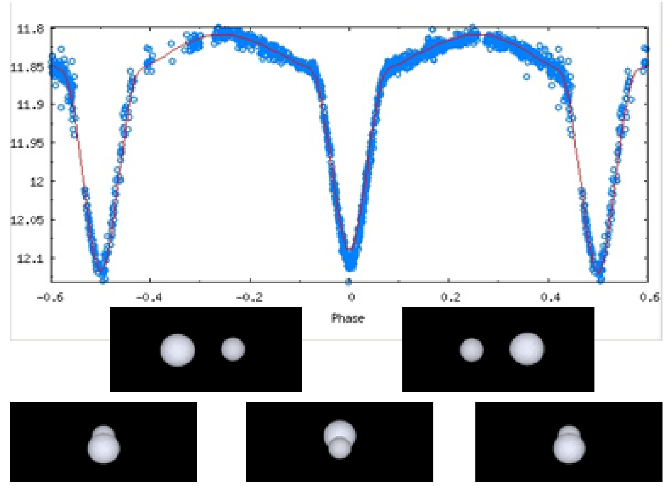

Figure 2. Observed V filter light curve data (points) and best fit model (line) with corresponding binary system model in panels displayed at various orbital phases (i.e. 0.0, 0.25, 0.5, 0.75).

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Distribution, Habitat Preference, and Status of the Ditch Fencing Crayfish, *Faxonella clypeata* (Hay) (Decapoda: Cambaridae), in Arkansas

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Running Title: *Faxonella clypeata* in Arkansas

Abstract

The ditch fencing crayfish, *Faxonella clypeata* (Hay), is a common and widespread crayfish that inhabits roadside ditches, intermittent first-order streams, shallow sloughs with heavy vegetation, and edges of swamps in Arkansas. Between 1997-2012, we made 55 collections of *F. clypeata* in 34 counties throughout eastern Arkansas, including 23 counties where *F. clypeata* had not been previously documented. At most of these locations within the West Gulf Coastal and Mississippi Alluvial Plain provinces, *F. clypeata* was found to be a locally abundant crayfish. With regard to conservation status, *F. clypeata* should be considered as "Currently Stable" due to its widespread distribution and general abundance throughout its range in the state.

Introduction

Arkansas is home to approximately 53 currently described species of crayfishes (Bouchard and Robison 1980, HWR *unpubl.*). Among these many crayfishes is the ditch fencing crayfish, *Faxonella clypeata* (Hay). Hay (1899) originally described *F. clypeata* as *Cambarus clypeatus* from near Bay St. Louis, Hancock Co., Mississippi. This crayfish occurs from southeastern Texas across the southern states to northern Florida and to South Carolina, ranging north to southeastern Missouri (Walls 2009). Recent studies of Arkansas crayfishes have improved our knowledge of several species (Robison and McAllister 2006, 2008, 2010, Robison et al. 2009, 2014, McAllister and Robison 2010, 2012, Wagner et al. 2010a, b, McAllister et al. 2011) but no investigation has involved *F. clypeata* in the state. *Faxonella clypeata* is a commonly encountered state crayfish species; however, we know little of its precise distribution and habitat in Arkansas. In an unpublished thesis, Reimer (1963) provided a cursory look of the distribution of this species in Arkansas. Fitzpatrick (1963) studied geographic variation in this species and elevated it to the genus *Faxonella* from a subgenus of *Orconectes*. Smith (1953) investigated the life history of this crayfish in Louisiana. Oklahoma crayfishes were surveyed by Reimer (1969) who provided locations of *F. clypeata* and some habitat information. Pflieger (1996) included this crayfish as a member of the Missouri crayfish fauna, and Walls (2009) surveyed the Louisiana crayfish fauna and included *F. clypeata* as a state member. More recently, Morehouse and Tobler (2013) reported that *F. clypeata* was found in three counties of southeastern Oklahoma.

The purpose of this present study was to attempt to accurately describe the habitat and distribution of *F. clypeata* in Arkansas. Specific objectives of the study were: (1) to determine the distribution of *F. clypeata*; (2) to document the habitat of *F. clypeata*; and (3) examine the current conservation status of this crayfish in the state.

Materials and Methods

Fieldwork was conducted from March 1997 through April 2012. The majority of collections was made during the months of March, April, and May. *Faxonella clypeata* was collected by hand, aquatic dipnets, baited and unbaited crayfish traps, and by digging burrows with shovels. Notes on habitat type were made at each of the 55 collection sites and later summarized for presentation in the text. Collection efforts were centered in southern and eastern Arkansas within the Mississippi Alluvial Plain (Delta) and West Gulf Coastal Plain (Fig. 1). Fifty-five collections of *F. clypeata* were made in 34 counties throughout eastern Arkansas (Appendix). Select voucher specimens were preserved in 60% v/v isopropanol and deposited in the Southern Arkansas University (SAU) Invertebrate Collection, and the Smithsonian National Museum of Natural History (USNM) Invertebrate Zoology

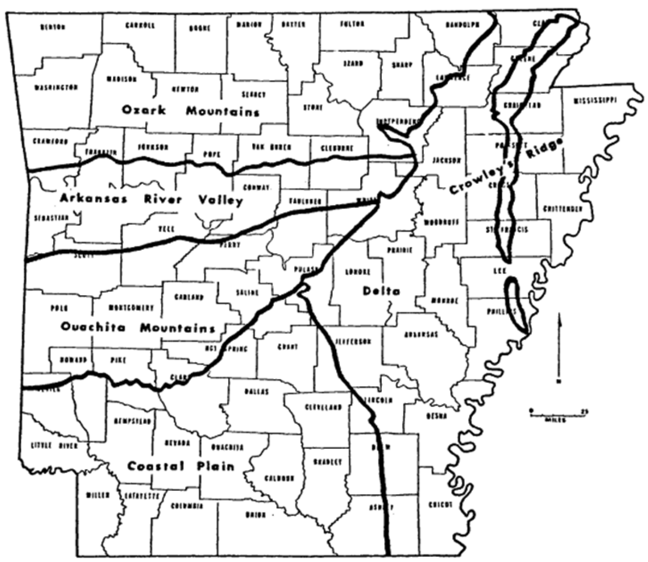

Figure 1. Physiographic regions of Arkansas. Coastal Plain (=West Gulf Coastal Plain), Delta (=Mississippi Alluvial Plain).

Collection in Washington, D.C. In addition to our field collections, crayfish collections housed at SAU were examined for specimens of *F. clypeata*, and a search of the online computerized database of crayfishes at the National Museum of Natural History, Smithsonian Institution (USNM 2014) and Illinois Natural History Survey Crustacean Collection (INHS 2014) was also performed.

Diagnosis of *Faxonella clypeata*:

Faxonella clypeata is a small crayfish (≤5 cm in total length) with a short, broad, turned down rostrum lacking marginal spines. The areola is short and wide and the cervical spine is absent with chelae sexually dimorphic. Male gonopods of Form I specimens possess only a central projection and a mesial process. The central projection of the gonopod is three times longer than the mesial projection. The mesial process is short, stout, and extending at most a quarter length of the central projection, never overlapping the other mesial process. Tips of the central projection overlap in normal position like crossed sabers when viewed from below. Copulatory hooks are found only on leg 3. See Pflieger (1996, plate 9) and Walls (2009, p. 141) for morphological characters.

Other *Faxonella* spp. in the state:

In Arkansas, two *Faxonella* species (*F. clypeata* and *F. blairi*) have been documented to occur (Robison et al. 2014) with a third, the Ouachita fencing crayfish (*F. creaseri*), possibly occurring in the state. Differences between the three species are as follows (see also Walls 2009): In male *F. clypeata* the mesial process of the gonopod is much shorter than in *F. creaseri* and the central projection is a bit thicker and less attenuated at the tip. Male *F. blairi* can be distinguished from all other *Faxonella* species by the much straighter central projection of *F. blairi*, which reaches to the coxae of the first pereiopod. In *F. creaseri*, the central projection reaches basically to the same level, but the distal half of the ramus is bent more mesially. In *F. clypeata*, the mesial process is much shorter. Hayes and Reimer (1977) described the distinguishing characters of *F. blairi*, including the annulus ventralis of the *F. blairi* female, which is much more firmly embedded in the sternum, much more than in other species of *Faxonella* and the sinus is simpler in sculpture.

Genetics:

Robison et al. (2014) recently provided information on the genetics of *F. blairi* and *F. clypeata*. Phylogenetic analyses (see their Fig. 2) clearly showed that these two crayfish species form reciprocally monophyletic groups and are genetically differentiated from one another and from species in other genera.

Results and Discussion

Our 15 years of collecting this species in 34 counties in Arkansas has established *F. clypeata* as an inhabitant of roadside ditches, intermittent first-order streams, shallow sloughs with heavy vegetation, and edges of swamps. Because this species is a secondary burrower, individuals construct simple burrows 10-30 cm deep topped by small turrets of tiny round pellets when water levels recede. Like Pflieger (1996), we found *F. clypeata* sequestered in these burrows for most of the year. We rarely found *F. clypeata* in permanent lentic situations. Generally, we found that this crayfish inhabited waters that dried up during the summer when they then took refuge in burrows dug into the ditch bottom or sides.

Reimer (1963) documented *F. clypeata* from 11 counties in Arkansas (Ashley, Calhoun, Cleveland, Columbia, Grant, Greene, Hempstead, Lincoln, Little River, Phillips, and St. Francis). Our studies amassed a total of 55 specific localities for *F. clypeata* (n =1,198 specimens), which are listed in the Appendix and plotted as counties in Fig. 2. *Faxonella clypeata* was documented from 34 counties throughout the Coastal Plain of Arkansas, 23 (68%) of them new county records. This crayfish was collected most frequently in southern Arkansas and was less abundant in the

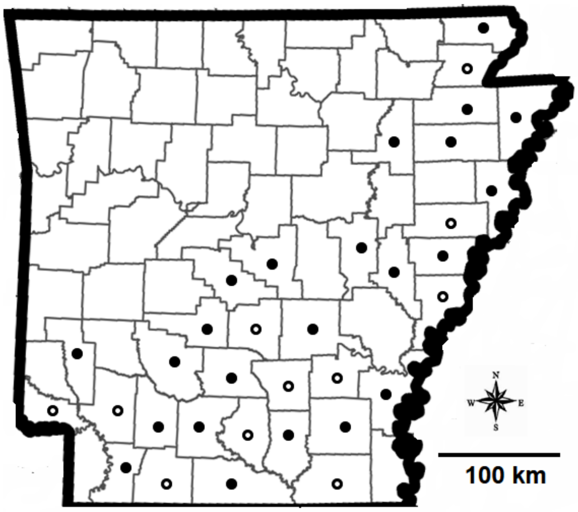

Figure 2. County distribution of *F. clypeata* in Arkansas. Open dots = previous records (Reimer 1963); dots (new records).

northeast Arkansas counties. Our sites in southwestern and southcentral Arkansas are well within the limits of the range of *F. clypeata* shown on the ecological niche model map of Morehouse and Tobler (2013, Fig. 44).

The highest number of specimens collected at one time was 208 individuals (USNM 208547) collected on 16 April 1983 by one of us (HWR) and D. Koym from a roadside ditch in Dallas County. Even though the crayfish was collected throughout the Coastal Plain physiographic province (Fig. 2), most often *F. clypeata* was found associated with pine woodlands or areas where trees were located rather than open alluvial farming areas. This finding mirrors what Walls (2009) found in Louisiana where he collected *F. clypeata* mostly in the pinelands, and not in the alluvial soils of the Mississippi and Atchafalaya basins.

Reimer (1969) collected *F. clypeata* in Oklahoma in roadside ditches, creeks, ponds, and burrows, while Morehouse and Tobler (2013) reported *F. clypeata* inhabited swamps and standing pools of water in roadside ditches in the state. In Louisiana, Walls (2009) found that *F. clypeata* was seldom in permanent waters deep enough for predatory fish, but preferred shallow ditches, sloughs and ponds with permanent vegetation. When this habitat dried, Walls (2009) reported the crayfish burrowed into the sides and bottom in individual mud cells. Interestingly, in Missouri, Pflieger (1969) collected this crayfish from small intermittent creeks and the shallows of seasonally flooded sloughs and swamps.

Collections of crayfishes have been made in all 75 Arkansas counties by HWR during the past 25 yrs. These collection records showed an absence of *F. clypeata* from the Ozark and Ouachita Mountains physiographic regions as well as the Arkansas River Valley and Crowley's Ridge. Rather, *F. clypeata* occupies the West Gulf Coastal Plain Province becoming less abundant in northeastern and extreme southwestern Arkansas. Intensive searches throughout all 75 counties of Arkansas revealed the presence of *F. clypeata* in only 34 (45%) counties (Fig. 2, Appendix). At most of these locations (n = 55) *F. clypeata* was found to be a locally abundant crayfish.

During the study period, six additional species of crayfish associates found sympatrically were collected while searching for *F. clypeata.* These included the digger crayfish (*Fallicambarus fodiens*), painted devil crayfish (*Cambarus ludovicianus*), Cajun dwarf crayfish (*Cambarellus puer*), twin crayfish (*Procambarus geminus*), White River crayfish (*Procambarus acutus*), and giant bearded crayfish (*Procambarus tulanei*).

Taylor et al. (2007) provided the most current conservation estimate of status of all native crayfishes in the United States and Canada. They reported 1.2% of the crayfish fauna of the two countries was endangered, while 14.3% was threatened, and 14.9% was considered vulnerable. In addition, 52% or 189 of the 363 native crayfishes were considered stable while 48% or 173 species were in need of some conservation status. Under American Fisheries Society status guidelines, *F. clypeata* was listed by Taylor et al. (2007) as CS (currently stable) with a Nature Conservancy/NatureServe heritage rank of G5 (demonstrably widespread, abundant and secure). In addition, the species is listed as Least Concern on the IUCN Red List (Crandall 2010). After extensive collecting in Arkansas, we agree with Taylor et al. (2007) and Crandall (2010) and feel *F. clypeata* should be considered as currently stable due to its widespread abundance throughout its range in Arkansas.

In summary, *F. clypeata* inhabits the West Gulf Coastal Plain and Mississippi Alluvial Plain physiographic provinces of Arkansas. Our research indicates this species is widespread and common in the state. Within Arkansas, the distributional range includes 34 counties located primarily in these physiographic provinces where *F. clypeata* was locally abundant.

Acknowledgments

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Appendix. County locations of 1,198 specimens of *F. clypeata* from Arkansas (locality, latitude/longitude in decimal degrees or township, section, and range [if known], date of collection, collector, museum collection, and number of specimens). HWR = Henry W. Robison.

Arkansas (*n* = 1,198)

Ashley County (*n* = 201)

(1) Ditch, 1.3 km S of Crossett Experimental Forest on St. Hwy. 133. 16 March 1967. J. Cooper & M. Cooper. USNM 118469. (40)

(2) Ditch, 8.0 km SW of Hamburg on US 82. 16 March 1967. J. Cooper & M. Cooper. USNM 118470. (115)

(3) South Fork of Fountain Creek at St. Hwy. 81. 16 March 1967. J. Cooper & M. Cooper. USNM 118472. (37)

(4) South Fork of Fountain Creek at St. Hwy. 81 in ditch from woodland stream. 16 March 1967. J. Cooper & M. Cooper. USNM 118473. (5)

(5) North Fork of Fountain Creek at St. Hwy. 81 S of Fountain Hill. 16 March 1967. J. Cooper & M. Cooper. USNM 118474. (2)

(6) Roadside ditch ca. 1.6 km S of Fountain Hill on St. Hwy. 81. 18 April 1986. HWR. USNM 218913. (2)

Bradley County (*n* = 31)

(1) Roadside ditch, 4.5 km E of Banks on St. Hwy. 275/4. 18 April 1986. HWR. USNM 218922. (31)

Calhoun County (*n* = 15)

(1) Roadside ditch, 4.7 km N of jct. of US 167 and St. Hwy. 272 on US 167. 18 April 1986. HWR. USNM 218908. (15)

Clark County (*n* = 14)

(1) Roadside ditch, 3.7 km S of Gurdon on St. Hwy. 53 (Sec. 10, R20W, T10S). 2 May 2002. HWR. SAU. (14)

Clay County (*n* = 1)

(1) Burrow, 4.3 km E of Corning on US 62 (Sec. 3, R5E, T20E). 15 March 1997. HWR. SAU. (1)

Cleveland County (*n* = 20)

(1) Backwater area ca. 3 mi. S of Rison on US 79 (Sec. 27, R11W, T9S). 4 May 2002. HWR. SAU. (20)

Columbia County (*n* = 168)

(1) Roadside ditch and burrows at jct. of co. rd. and US 82 (Sec. 23, R20W, T17S). 6 March 1982. HWR. USNM 177931. (17)

(2) Ditch and burrows 3.2 km E of jct. of St. Hwy. 98 and St. Hwy. 82 (Sec. 23, R19W, T17S). 6 March 1981. HWR. USNM 177939. (12)

(3) Roadside ditch, 18.3 km E of Magnolia on US 82. 30 April 2007. HWR. SAU. (133)

(4) Trib. to Little Cornie Creek, off US 82, vic. Calhoun jct. 13 April 2012. C. T. McAllister & M. B. Connior. Uncatalogued. (3)

(5) Off US 82 at co. rd. 36, Columbia/Union Co. line. 13 April 2012. C. T. McAllister & M. B. Connior. Uncatalogued. (3)

Craighead County (*n* = 5)

(1) Roadside ditch, 4.9 km S of Jonesboro on St. Hwy 1 (Sec. 9, R4E, T13N). 10 April 1992. HWR. SAU. (4)

Crittenden County (*n* = 3)

(1) Roadside ditch, 1.9 km S of Norvell on St. Hwy. 149 (Sec. 9, T7N, R6E). 18 April 1994. HWR. SAU. (3)

Dallas County (*n* = 204)

(1) No specific locality data. USNM 206946 (1).

(2) Roadside ditch, 0.6 km N of Ouachita-Dallas Co. line on St. Hwy. 7. 16 April 1983. HWR & D. Koym. USNM 208547. (203)

Desha County (*n* = 1)

(1) Backwater slough 1.6 km W of Dumas on St. Hwy. 54 (Sec. 29, R4W, T9S). 3 June 1999. HWR. SAU. (1)

Drew County (*n* = 77)

(1) N of Hamburg on St. Hwy. 81, creek beyond county line. 16 March 1967. J. Cooper & M. Cooper. USNM 118471. (28)

(2) Roadside ditch, 0.2 km E of Bradley Co. line on St. Hwy. 4. 18 April 1986. HWR. USNM 218910. (48)

(3) Roadside ditch, just E of Cut-Off Creek at St. Hwy. 35. 18 April 1986. HWR. USNM 218921. (1)

Grant County (*n* = 45)

(1) Creek, 12.9 km S of Sheridan on US 167. 18 March 1997. HWR. SAU. (29)

(2) Burrows, 4.0 km SW of Sheridan on St. Hwy. 35

(Sec. 19, R13W, T4S). 18 March 1997. HWR. SAU (16)
Green County (*n* = 1)
(1) Roadside ditch, 3.1 km S of Clay Co. line on St. Hwy. 135. 12 April 1985. HWR. USNM 218921. (1)
Hempstead County (*n* = 16)
(1) Roadside ditch in Blevins. 20 May 1983. E. Laird. USNM 208517. (8)
(2) Vicinity of Collins Bayou, outside of Blevins. 20 May 1983. S. Hill & B. Hill. USNM 208556. (1)
(3) Roadside ditch 6.4 km S of Blevins on St. Hwy. 24 (Sec. 19, R23W, T10S). 16 April 2001. HWR. SAU. (7)
Hot Spring County (*n* = 15)
(1) Roadside ditch, 3.2 km W of Grant Co. line on US 270. 30 April 1976. H. H. Hobbs, Jr. & Kearny. USNM 147220. (5)
(2) Roadside ditch, 5.6 km W of Poyen, ca. 8.0 km E jct. of US 167 and St. Hwy. 27 on US 270. 17 March 1980. HWR. USNM 177213. (10)
Howard County (*n* =11)
(1) Burrows, 3.9 km SE of Mineral Spring on St. Hwy. 355 (Sec. 33, R27W, T10S). 9 May 2006. HWR. SAU. (11)
Jackson County (*n* = 12)
(1) Roadside ditch and culvert on St. Hwy. 17, 0.5 km N of Auvergne. 4 April 1973. S. Pelt. USNM 144587. (12)
Jefferson County (*n* = 12)
(1) Roadside ditch at Beth Lovorn's residence at Hardin on W. Holland Rd. 25 April 1982. B. Lovorn. USNM 208650. (2)
(2) Roadside ditch, 5.0 km S of St. Hwy. 54 on US 79. 18 March 1987. HWR. USNM 219235. (10)
Lafayette County (*n* = 3)
(1) Roadside ditch, 1.9 km N of Lewisville on St. Hwy. 82. 26 April 1976. H.H. Hobbs, Jr. & Kearny. USNM 147182. (1)
(2) Roadside ditch at US 82, 10.0 km E of Red River. 25 April 1975. R. W. Bouchard. USNM 176773. (2)
Lee County (*n* = 6)
(1) Backwater ditch 1.6 km SE of Marianna on St. Hwy. 185 (Sec. 35, R3E, T2N). April 28 2003. HWR. SAU. (6)
Lincoln County (*n* = 74)
(1) Roadside ditch, 5.0 km NW of Yorktown. 17 April 1983. HWR & D. Koym. USNM 208543. (14)
(2) Roadside ditch, 6.6 km S of jct. of St. Hwy. 11 and 293 on St. Hwy. 293. 17 April 1983. HWR & D. Koym. USNM 208544. (32)
(3) Roadside ditch, W off Holland Rd. at Hardin. 17 April 1983. HWR & D. Koym. USNM 208551. (2)
(4) Roadside ditch, 8.4 km N of Star City. 25 April 1986. HWR. USNM 218938. (28)
Little River County (*n* = 17)
(1) Roadside ditch, 3.2 km W of Ashdown on St. Hwy. 32. 27 April 1976. H. H. Hobbs, Jr. & Kearny. USNM 147190. (17)
Mississippi County (*n* = 9)
(1) Roadside ditch in Manila on St. Hwy. 18 (Sec. 36, R8E, T14N). 21 March 1999. HWR. SAU. (9)
Monroe County (*n* = 4)
(1) Roadside ditch, 2.4 km N of Arkansas Co. line on St. Hwy. 33. 16 April 1985. H. H. Hobbs, Jr. & R. Gilpin. USNM 219019. (3)
(2) Roadside ditch, 6.4 km S of jct. of US. 79 and 49 on US 49. 17 April 1985. H. H. Hobbs, Jr. & R. Gilpin. USNM 219020. (1)
Nevada County (*n* = 2)
(1) Roadside ditch on gravel rd. ca. 9.0 km W of jct. with US 67. 28 February 1981. HWR. USNM 177941. (2)
Ouachita County (*n* = 11)
(1) Unnamed trib. of Two Bayou Creek between St. Hwy. 4 and St. Hwy. 24. 30 March 1975. S. Pelt. USNM 146675. (8)
(2) N of Stephens off St. Hwy. 57. 13 April 2012. C. T. McAllister & M. B. Connior. Uncatalogued. (3).
Phillips County (*n* = 3)
(1) Roadside ditch, 0.8 km SE of Marvell on St. Hwy. 316. D. Jones. 2 April 1982. USNM 208649. (3)
Poinsett County (*n* = 15)
(1) 0.8 km N of Fisher (35.5047°N, 90.9651°W). 5 May 2006. B. Wagner. INHS 10825. (15)
Prairie County (*n* = 3)
(1) Roadside ditch and burrows ca. 4.0 km NW of DeValls, Bluff (Sec. 7, T2N, R4W). 23 March 1995. HWR. SAU. (3)
Pulaski County (*n* = 1)
(1) Camp Pike. 1918. No other data. USNM 218623 (1)
Saline County (*n* = 9)
(1) 3.2 km SE of Shannon Hills (34.6113°N, 92.3644°W). 13 April 2006. B. Wagner. INHS 10558. (1)
(2) 1.6 km N of Lakeside, Woodson Lateral Rd. (34.5478°N, 92.2575°W). 13 April 2006. B. Wagner. INHS 10559. (8)
St. Francis County (*n* = 1)
(1) Roadside ditch, 3.2 km S of Forrest City on St. Hwy. 1. 13 April 1983. HWR. USNM 218697. (1)
Union County (*n* = 188)
(1) No locality data. 30 October 1991. J. Stanley. USNM 260204. (2)
(2) Roadside ditch, ca. 9.7 km W of El Dorado on US 82. 2 April 2000. HWR. SAU. (79)
(3) Roadside ditch at Marysville on US 82. 20 April 2006. HWR. SAU. (106)
(4) Roadside ditch, ca. 3.2 km NW of Mount Holly on St. Hwy. 57. 20 April 2006. HWR. SAU. (1).

Toad (Anura: Bufonidae) Limb Abnormalities from an Aquatic Site in Scott, Pulaski County, Arkansas

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Running Title: Toad (Anura: Bufonidae) Limb Abnormalities from an Aquatic Site in Scott, Pulaski County, Arkansas

Abstract

We collected and examined 16 Fowler's toads, *Anaxyrus fowleri*, and one dwarf American toad, *Anaxyrus americanus charlesmithi*, in central Arkansas in 2011. Collection was initiated by observation of abnormal toads. Toads were euthanized, measured, photographed, and deposited in the Arkansas State University herpetological collection. Several toads were radiographed. We found various abnormalities in both forelimbs and hindlimbs and on both sides of the body. The causes of the abnormalities remain unknown and will require further studies to determine if the environment is imperiled.

Introduction

Studies on amphibian limb abnormalities have recently become common in the field of amphibian conservation. These abnormalities can vary from limb malformations to complete limb absence. Many stressors can cause similar responses and a singular cause that links the array of abnormalities is not known. Instead the several known causes of these abnormalities appear to have variable effects.

Although research has led to a better understanding of the abnormalities, there is a lack of knowledge of the causes and implications of the deformities (Blaustein and Johnson 2003). Lannoo (2008) believes many sources, both natural and artificial, ranging from fish excrement to overcrowding, are significant when pinpointing causes of the abnormalities.

Herein, we present an observational investigation of abnormalities in Fowler's toads (*Anaxyrus fowleri*) and dwarf American toads (*Anaxyrus americanus charlesmithi*) from a site in south central Arkansas. This is intended to describe the abnormalities and allow for future studies to determine potential causes.

Materials and Methods

Seventeen toads of varying sizes were collected by hand in Scott, Arkansas (Pulaski County) (34˚38"03.19"N, 92˚07'58.95"W) and sent to Arkansas State University for processing. The random collection was opportunistic and non-exhaustive. The site (Fig. 1) was around an in ground swimming pool in a residential neighborhood, bordered by several lakes with forest areas and many agricultural plots. The site was not chosen, and the collection was initiated by the property owner (D. Beard) when abnormal toads were observed. The toads were euthanized in a dilute chlorobutanol solution. Fixation was done in a 10% v/v neutral buffered formalin solution. All toads were then photographed, given museum tags, and placed in 70% v/v ethanol.

Figure 1. Map of collection site near Scott (Pulaski Co., Arkansas).

Results

Thirteen toads of the 17 collected exhibited some abnormality. Most (92.3%) abnormalities affected the limbs. A list of common abnormalities of the limbs adapted from Lannoo (2008) was used as a basis for identification of the abnormalities (Table 1). Limb abnormalities varied among the toads and were found in both adult and sub adult toads (maximum SVL was 75 mm; minimum SVL was 27 mm).

Table 1. Common amphibian limb abnormalities from Lannoo (2008).

Abnormality	Description
Amelia	Missing limb
Ectromelia	Missing limb segments
Polydactyly	Extra digits
Ectrodactyly	Complete absence of digit including metatarsal bone
Skin Webbing	Band of skin crossing a joint

Table 2 documents all abnormalities observed and gives a short description of location and affected area.

One of the 5 toads that did not exhibit any limb abnormalities did appear to have a slightly above average curvature of the sacral hump (seen in Fig. 1a), but it was not radiographed or considered suspicious. The other 4 toads that were collected did not exhibit any noteworthy malformation or abnormality and are not presented in any of the following tables or figures.

Photographs of toads can be seen in Figure 2. Figure 3 shows radiographs and photographs of some of the abnormal toads.

Discussion

The abnormalities presented do not have a known cause as of yet. We can speculate, however, to the causes of some of the abnormalities.

The trematode parasite *Ribeiroia ondatrae* is frequently associated with limb abnormalities in anurans, but usually affects the hind limbs. Johnson et al. (2001) found that infections of *R. ondatrae* in *Bufo* (*Anaxyrus*) *boreas* tadpoles induced severe limb malformations ranging from supernumerary limbs to complete limb absence. In 2002, Johnson et al. linked *R. ondatrae* infection to amphibian malformations in the western United States. In 9 species of amphibians from 4 states, they found that the parasite caused both forelimb and hind limb malformations of varying degrees. Although many believe the parasite is the culprit behind most abnormalities, others disagree.

In a study of wood frogs, Eaton et al. (2004) found abnormalities associated with *R. ondatrae,* but found no evidence, i.e. cysts, linking the parasite with the abnormalities and instead suggested that sublethal predation was a primary cause for the abnormalities. Because limbs develop outside the body, abnormalities in hind limbs of anurans ranging from missing digits to missing limbs may be linked to sublethal predation (Ballengée and Sessions 2009, Bowerman et al. 2010). Ballengée and Sessions (2009) attribute the abnormalities to dragonfly nymphs and Bowerman et al. (2010) attribute abnormalities to sticklebacks. Others believe pollution is to blame.

Table 2. Observed abnormality and description

Abnormality	Description
Superimposition	Third toe on left foot perpendicular to 2nd toe
Ectromelia	Right forelimb and hindlimb missing distal elements
Oligodactyly	Left hand missing 4th toe and distal portion of 3rd toe
Oligodactyly and skin webbing	Right foot missing 3 medial toes and two lateral toes fused
Ectromelia	Elements below proximal half of right radioulna absent
Adactyly	All toes on right foot absent
Ectrodactyly	All metatarsals and phalanges on left hand absent
Skin webbing	Medial toes on right hand fused
Skin webbing and oligodactyly	Second, third, and fourth toe fused on right hand and 2nd and 3rd toe missing distal phalanges
Amelia	Entire right forelimb missing
Bilateral Ectromelia	Distal portions of both forelimbs missing
Bilateral Ectromelia	Elements distal to proximal portion of humeri missing
Kyphosis	Above average convex curvature of the sacral hump

Pesticides, herbicides and other chemical compounds used for agriculture have been suggested as the direct or indirect cause of some amphibian malformations (Ouellet et al. 1997, McCallum 1999, Taylor et al. 2005). Reeves et al. (2008) found that skeletal abnormalities increased with proximity to roads in wood frogs in Alaska and suggested that multiple factors from vehicles, pollution, and predator community shifts may cause the increase in abnormalities. Reeves et al. (2010) discussed multiple stressors that may increase likelihood of abnormalities such as radiation, pollution, and predation. The cutaneous fusion observed in some of the toads may be linked to the trematode parasite *Ribeiroia* that has been found by Johnson et al. (2001) to cause skin webbing in western toads. Missing digits and kyphosis could be caused by injury from sublethal predation. Other abnormalities such as partial limbs or ectromelia of the hind limbs may also be caused by sublethal predation. Ouellet et al. (1997) and Lannoo (2008) believe hindlimb deformities can be attributed to agricultural chemicals such as pesticides and fertilizers.

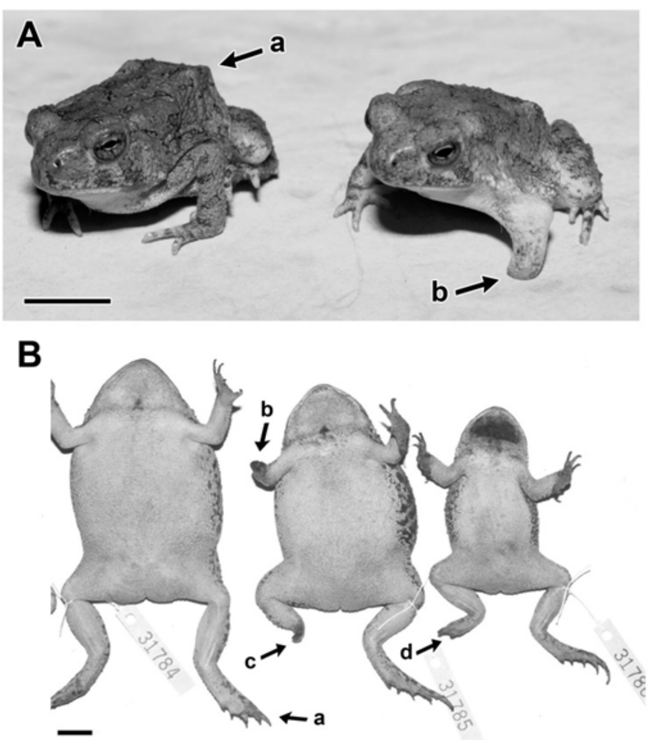

Figure 2. Toads in A with abnormal curvature of sacral hump (a) and ectrodactyly of right hand (b). Toads in B with superimposed toe (a), ectromelia of right forelimb (b) and right hindlimb (c), and oligodactyly (d)

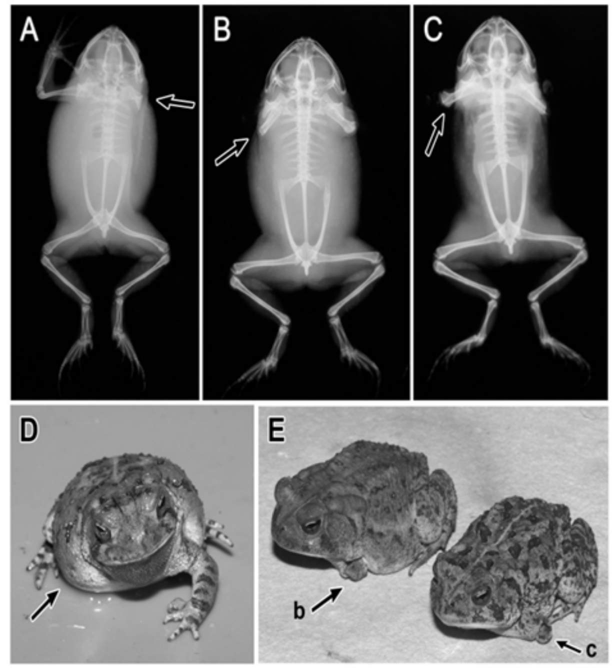

Figure 3. Radiographs showing amelia (A), and bilateral ectromelia (B and C). Photographs D and E are the corresponding toads for radiographs A,B, and C. Missing and partial limbs shown in photographs D and E (b and c).

Acknowledgments

Authorization of toad collection was granted by a collecting permit from the Arkansas Game and Fish Commission. We would like to thank Mike Lannoo for his contribution in determination of abnormalities and production of radiographs. This study was conducted under established protocols set by the IACUC at Arkansas State University. All specimens were deposited into the Arkansas State University Herpetological Collection and are labeled using museum coding numbers (31724, 31725, 31782-31786, 31794, 31800-31808).

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Growth and Reproduction in the Ouachita Madtom (*Noturus lachneri*) at the Periphery of its Distribution

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Running title: Growth and Reproduction in the Ouachita Madtom

Abstract

The Ouachita madtom (*Noturus lachneri*) occurs primarily in drainages of the upper Saline River and in a few small tributaries to the Ouachita River in Arkansas, USA. We collected specimens by hand and by use of aquarium dipnets on 29 occasions from 20 October 1999 through 25 July 2000 in Cooper Creek, presently a feeder creek into Lake Catherine on the Ouachita River. Total length was measured, reproductive attributes were noted, and individuals were released at the capture site (with exception of 3 gravid females retained to assess fecundity). We recognized 2 age (size) classes during most of the year based on a plot of length-frequency distributions. Regression of total length against time indicated a mean growth rate of 0.14 mm/day for the population, and 0.20 mm/day for juveniles during warmer months. Hatchlings were found from 27 June through 4 November.

Introduction

The Ouachita madtom (*Noturus lachneri*) is a small catfish endemic to the upper Saline and Ouachita River drainages in central Arkansas (Taylor 1969, Robison and Buchanan 1988). It has a uniform dorsal coloration ranging from brown to gray to pinkish, and prefers small to medium-sized, high-gradient streams with cobble, gravel, or softer-substrate bottoms (Robison and Buchanan 1988). Most of the historic records for *N. lachneri* are from the Saline River drainage, with 1 record previously known from the Ouachita River drainage (Robison and Buchanan 1988).

Distribution, habitat, and foods of *Noturus lachneri* were described by Robison and Harp (1985), metapopulation dynamics by Gagen et al., (1998), food habits by Patton and Zornes (1991), helminth parasites by Fiorillo et al., (1999), and reproduction by Stoeckel and Gagen (2011).

The most southwestern record, and the second for the Ouachita drainage, is located in Cooper Creek – a small tributary to Lake Catherine on the Ouachita River (Tumlison and Tumlison 1996). It is a shallow, high-gradient creek with a largely non-embedded substrate that includes gravel, cobble, and boulders. Ouachita madtoms are common in Cooper Creek, which provided an opportunity to study this species considered by Buchanan (1974) to be endangered and by Robison and Harp (1985) to be threatened due to its small population size and vulnerability to environmental degradation caused by gravelling and road construction. Presently, the Arkansas Game and Fish Commission designates this species as being one of special concern.

Growth rates based on periodical collection for this madtom are not known. Herein we report growth rates, longevity, and reproduction of *N. lachneri* at the periphery of its range in the Ouachita drainage system.

Methods

Four sites were selected along Cooper Creek in Garland County, Arkansas based on accessibility and presence of madtoms (for a map of sites see Tumlison and Tumlison 1996). A total of 29 sampling trips was made between October and July, 1999-2000. During each visit to each site, we spent about 2 hrs to thoroughly search all microhabitats in stream sections of about 40 m length, turning stones to reveal madtoms. Pools, riffles, and runs were represented in each sample site, and all of these were searched from bank to bank during each sampling period. Depth of the pools reached 75 cm, although most successful sampling was done at depths < 30 cm and especially in riffle areas where stones protruded above the surface of the water. Also, we established a fixed $1m^2$ plot at each site and lifted all stones within a $1m^2$ wooden quadrat frame to estimate density, per visit.

During each trip, individuals located by lifting stones were caught by use of aquarium dipnets, then transferred to a holding bucket. Seining had proven to be a less efficient sampling technique and electroshockers were not available. Madtoms placed in plastic bags for measurement were in motion constantly, so we decanted water to a minimum prior to measuring specimens. To reduce stress on individuals prior to release, we then measured total length (TL) rather than standard length (SL) on most live specimens. However, to allow comparisons with other studies, we measured both TL and SL on a subset of specimens (those collected 17 May and 25 July: N = 73). Reproductive data were noted when evident (females with eggs, breeding males identified by enlarged cephalic muscles). Subsequently, specimens were released at the site of capture, with the exception of three gravid females retained to obtain eggs counts (vouchered HSU 3589).

Ages of madtoms have been estimated by use of various bony structures (Clugston and Cooper 1960), which may require the sacrifice of specimens. To estimate age distributions without the need to sacrifice specimens, a plot of the frequency distribution of lengths was made for the collection dates of 17 May through 25 July, when sample sizes were larger and young-of-the-year (YOY) were appearing in the population. A linear regression analysis was performed on the data collected over the entire 284-day sampling period to determine the slope of the regression equation, which reflects the mean rate of growth in the population in mm/day. A second regression analysis concerning only hatchlings was made using data from 17 May – 25 July, when sample size permitted distinction of hatchling specimens based on bimodal distribution of sizes. This analysis was used to evaluate the expected higher growth rate for hatchlings during the warmer months (Mayden and Burr 1981, Mayden and Walsh 1984). For the regression analyses, dates were converted to days from day 1 (14 October) to day 284 (27 July).

Results and Discussion

We made 609 captures over the 29 sampling dates between October and July. Hatchlings appeared in July, and 3 age (size) classes could be discerned at that time.

Densities of *N. lachneri* have been reported to be low (Robison and Harp 1985). Gagen et al., (1998) indicated higher than expected densities obtained by electrofishing, averaging 95/100 m^2 (range 17.2-204/100 m^2). We found usually 0-2, but a maximum of 8, *N. lachneri* per m^2 at the 4 plots over 15 dates (60 samples - mean 80/100 m^2). Densities differed at the 2 most productive sites, from May-July averaging 28.1/100 m^2 and 84.8/100 m^2.

At the latter site, a separate density estimate of 106/100 m^2 was calculated based on the largest sample of 33 individuals at the site of 31 m^2 area on 27 June. This should be a low estimate of density because it is unlikely that we caught all individuals at the site. However, these figures support the contention of Gagen et al., (1998) that densities of *N. lachneri* can be much higher at some locations than previously thought.

The mean ratio of SL/TL was 0.854 (85.4%, miminum 81.2%, maximum 90.0%, SE 0.26%). Similarly, a regression of SL against TL for *Noturus insignis* revealed a slope of 0.851 (Clugston and Cooper 1960). On average, the caudal fin comprises 14.6% of the total length.

The maximum length we found was 88 mm TL (73 mm SL), from a male collected 25 July. Previously reported maximum lengths were 69.5 mm SL (Robison 1980), a male 83.1 mm SL collected 1 August (Robison and Harp 1985), and 94 mm TL (we estimate 80.3 mm SL) (Gagen et al.1998). Adults usually range from 23-66 mm SL (Robison 1980), and the largest specimens collected in February by Fiorillo et al., (1999) were 69-70 mm (SL). Our largest specimens were male and were collected in July-August, thus it is likely that the maximum length is attained by males that die before the next spring (because none of that size was found entering the breeding season, and larger males appeared to be senescent).

The frequency distribution of lengths prior to the new hatch indicated 2 size (age) classes (Figure 1). With the hatch beginning in July, 3 age classes exist. However, older (larger) specimens were disappearing, and none were present in the previous October samples, suggesting longevity appears to be just over 2 years in the Cooper Creek population. Fiorillo et al., (1999) reported 3 size classes in a February sample from the Saline River, based on a plot of standard length versus body mass. Their larger specimens were 69-70 mm SL in the third size class. Specimens of this size did not appear in the Cooper Creek samples until May. If the sudden appearance (Figure 2) of these sizes does not indicate movement from other locations to the sample areas for breeding, it appears that longevity and adult sizes for *N. lachneri* may differ among localities, with maximum longevity just entering a third year. Based on our findings, very few individuals survived long into a third age class.

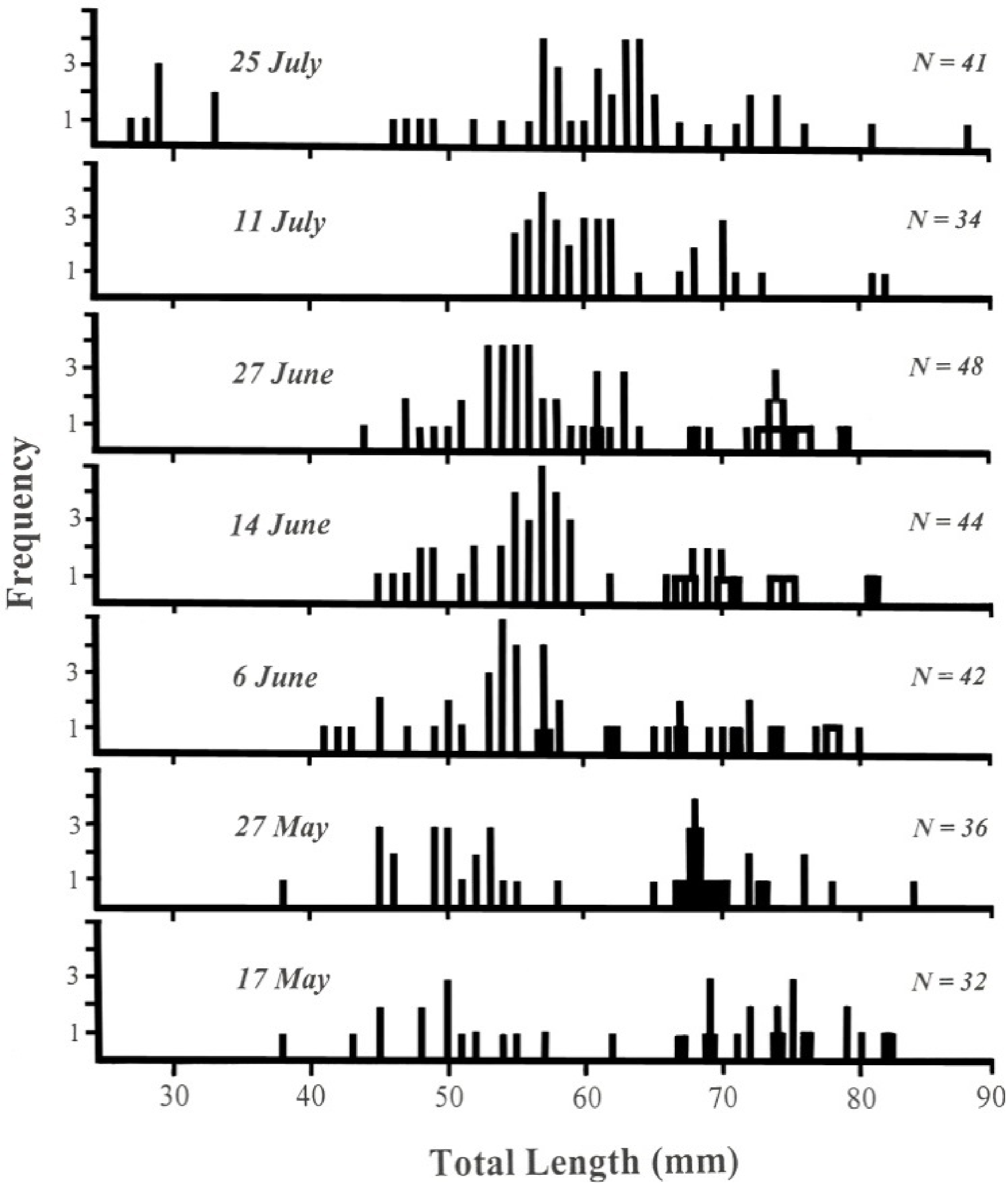

Figure 1. Frequency distribution of total lengths (mm) of *Noturus lachneri* for May-July, 1999-2000, collections in Cooper Creek, Garland Co., AR. Gravid females are indicated by blackened rectangles and breeding males by open rectangles. Earlier hatchlings were beginning to exceed 30 mm TL on 25 July.

Similarly, a study of *N. miurus* (Burr and Mayden, 1982a) found that < 0.5% of the population was $\geq$ 2 years old, and a study of *N. placidus* demonstrated that few individuals survived to age 2 (Bulger & Edds 2001).

Fiorillo et al., (1999) noted the small number of *N. lachneri* specimens in their third size class and reported that heavily parasitized individuals may experience greater mortality. We noted a battered

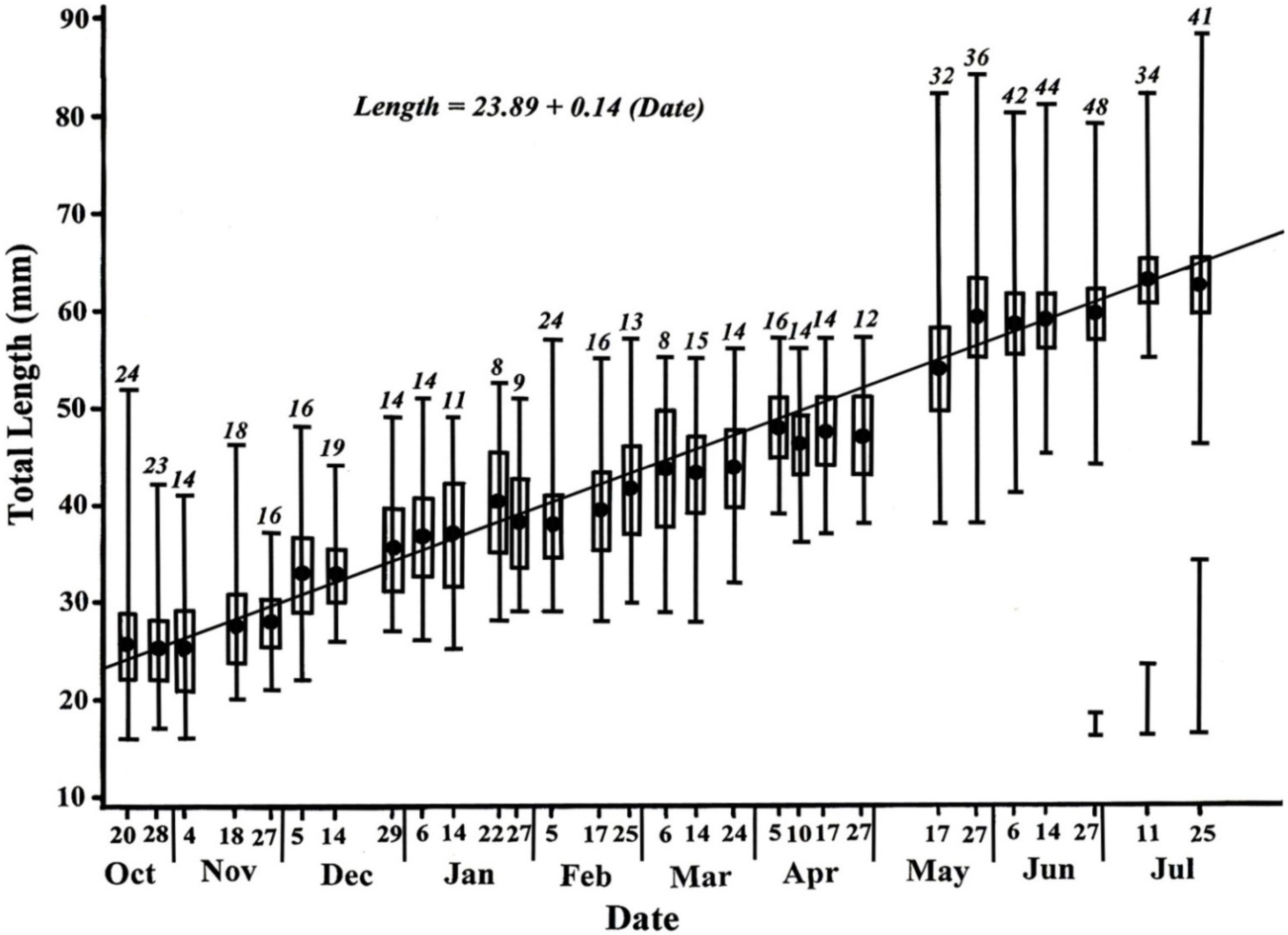

Figure 2. Linear regression of total length (mm) against time for *Noturus lachneri* in Cooper Creek, 1999-2000. Mean (black dot), +2 SE (open rectangles), and range (bars) for each sample date are shown. Sample sizes appear above the maximum length for each date. Additional range bars in July reflect hatchlings (not used in calculation of the regression). The slope of the regression equation indicates a mean population growth rate of 0.14 mm/day, $R^2 = 0.69$.

appearance on many larger male specimens, which could indicate that costs of reproduction may contribute to mortality of larger (breeding) individuals.

The mean length of *N. lachneri* increased in a linear pattern (Figure 2). The slope of the regression equation indicated a mean growth rate of 0.14 mm/day ($R^2 = 0.69$, $P < 0.0001$), but the growth rate of hatchlings during the warmer months was estimated to be 0.20 mm/day ($R^2 = 0.39$, $P < 0.0001$). The regression likely appeared to be linear, even with 2 age classes present, because most of the juvenile cohort (the hatchlings from the previous year) already had accomplished its most rapid summer growth by the start of the study in October. Burr and Mayden (1982a) reported that one half of annual growth in length of *N. miurus* was attained within 2 months, and evaluated growth rate with a curvilinear regression model. In *N. insignis*, Clugston and Cooper (1960) noted a rapid increase in TL during the first 2 summers and thereafter only in late summer following reproduction.

Variation in the percent of each age class comprising successive samples could alter the calculated growth rate, thus the percent of each sample representing juveniles was calculated to evaluate bias. From October to early May, juveniles comprised an average of 80.1% of the samples (range 56-88%, with 10 of 12 dates ≥79%), meaning that about 80% of the sampled population during that portion of the year was from the most recent hatch. During the part of the reproductive season from mid-May through July, the frequency of the juvenile class (not including new hatchlings) decreased to an average of 67.9% (range 47-80%). At this time, more and larger adults were collected, perhaps because of spawning activity.

Smallest hatchlings were measured at 15 mm TL, and juveniles (N = 32) collected on 25 July (the same date when hatchlings became most common) averaged 61.4 mm TL (estimated 52.4 mm SL). Average size of adults (N = 10) on 27 June (when hatchlings were first observed) was 73.4 mm TL (estimated 62.7 mm SL).

Populations of madtoms can be especially susceptible to environmental damage if individuals are semelparous (Simonson and Neves 1992, Fuselier and Edds 1994). Presently, reproductive data for *N. lachneri* are limited (Robison and Buchanan 1988, Stoeckel et al. 2011). In our study, 5 of 18 adults (identified when TL > 65 mm, see Figure 1) were identifiable as females with developing eggs on 17 May (water temperature 19.0°C). Gravid females were noted on each collection date through 27 June (Figure 2, blackened rectangles). The frequency distribution of sizes indicated that on 6 June and 27 June, 2 females presumably of the juvenile class (based on TL of 57 and 61 mm) were gravid with eggs visible through the abdomen. Stoeckel et al. (2011) also noted oocyte development in some smaller *N. lachneri* and believed that females mature between 58 – 66 mm TL in Saline River drainage populations. Most reproductive females in our study were estimated to be 2 years old (Figure 1). Mature ova in subadults are also known in *N. exilis* (Mayden and Burr 1981) and *N. miurus* (Burr and Mayden 1982a).

Counts of eggs were made on 3 preserved gravid females. Two females collected 17 May (TL = 82 mm) and 27 June (TL = 79 mm) contained 25 and 22 eggs, respectively, of 2-3 mm diameter. A third female collected 17 May (TL = 69 mm) contained 37 eggs of 1.5-2.5 mm diameter. The only nest found, on 6 June, contained 23 eggs. A mean number of 35 oocytes was found in *N. lachneri* during a Saline River drainage study, and the mature size was estimated to be >3 mm (Stoeckel et al. 2011).

Schooling or 'communal activity' was not observed by Robison and Harp (1985). No schooling was observed in our study, however on 27 May, 4 individuals were revealed by lifting a single stone of 150 mm diameter. This proximity may be due to the onset of the reproductive season, although these specimens were not caught to determine their sex. Previously, pairs of unsexed madtoms were seen in Cooper Creek by lifting single stones in July (Tumlison and Tumlison 1996).

Males of many species of madtoms are known to guard the nest (Taylor 1969, Robison and Buchanan 1988). As in many other species of madtoms, breeding males of *N. lachneri* can be identified by the enlarged cephalic epaxial muscles, and such males were found from 6 June through 27 June (open rectangles, Figure 1). On 6 June, at a water temperature of 17.0°C, a male 78 mm TL was found guarding a nest. The clutch consisted of 23 eggs of yellowish color adhering to one another in a mass. The brooding site was a depressed area under a flat stone 190 x 180 mm wide and 30 - 40 mm thick, at a water depth of 200 mm. The site was mid-stream about 3.5 m from each bank, and in a pool area just upstream from the nearest riffle. Stoeckel et al. (2011) reported nests of *N. lachneri* from mid-June through July at water temperatures of 19-27°C and with characteristics consistent with this single observation. These habitat characteristics also are consistent with other descriptions of nests of *Noturus* (Mayden et al. 1980, Mayden and Burr 1981, Burr and Mayden 1982a).

As a behavioral caveat, we noted this male remained on site even when the eggs were dipped into a net, allowing the individual to be captured. Non-brooding madtoms moved to cover under adjacent stones when a stone was lifted exposing them. The male and the egg mass were placed in the same bucket, and the male ingested the eggs. Similarly, Burr and Mayden (1982a) noted that a disturbed guardian male *N. miurus* picked up a clutch of eggs and shook it vigorously. By 27 June, breeding males tended to be thin and battered in appearance, and were less vigorous in attempts to escape capture, presumably as a result of the energy spent guarding the eggs and the concomitant lack of opportunity to forage. We found no evidence of males breeding in their first year.

Robison and Buchanan (1988) reported hatchlings (16-25 mm SL) collected 1 August, and Tumlison and Tumlison (1996) found hatchlings (20 mm SL) as early as 15 July but more commonly on 29 July (15 mm SL). In the present study, hatchlings (2, each 17 mm TL) were first captured on 27 June (water temperature 21.5°C). Eight hatchlings, 20-23 mm TL, were seen on 11 July. By 25 July, hatchlings of different sizes were found: 9 recently hatched at 15 mm TL and 22 older hatchlings at 27-33 mm TL. Although most hatching appears to occur in late July and August, recent hatchlings still were found as late as 20 October (18 mm), 28 October (17 mm), and 4 November (16 mm). From these data, the length of the hatching season appears to be from late June through early November. It is not known whether the long hatching season is extended via multiple spawning as suggested by Mayden and Burr (1981) and Vives (1987) for *N. exilis*.

Finding hatchlings in a stream smaller than was considered to be typical habitat, Robison and Harp (1985) suggested that *N. lachneri* may move to spawn in smaller tributaries. In contrast, Vives (1987) found no significant difference in depths occupied by smaller versus larger *N. exilis*. Some species of *Noturus* move to riffles to spawn (Mayden and Burr 1981), and others move to pools (Burr and Mayden 1982b, Starnes and Starnes 1985). We seldom found hatchlings of *N. lachneri* in mainstream sections of the sample sites, but did discover many of them while searching smaller and shallower reaches of the stream. If most spawning did not occur at these fine-grained sites, at least the young moved there soon after hatching.

Searches for hatchlings of *N. lachneri* revealed them in shallower riffle areas where cobble protruded above the surface of the water and where particle size of most of the substrate was smaller (gravel versus cobble). Tumlison and Tumlison (1996) previously noted that smaller YOY tended to be found in finer-grained microhabitat. Only 1 nest was found during this study, but Stoeckel et al. (2011) also found nests in similar conditions in Saline River populations. If these observations do represent typical spawning habitat, hatchlings soon move into the smaller gravel of the main stream but away from the larger individuals until some growth has occurred.

This madtom is considered to be a species of special concern largely due to its very limited range and lack of life history information. Our results reveal that the species is short-lived and most individuals likely breed only once, therefore it is important that habitats be protected against sedimentation and other degradation to the limited habitat.

Acknowledgments

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Urban Stream Syndrome in a Small Town: A Comparative Study of Sager and Flint Creeks

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Running Title: Urban Stream Syndrome in a Small Town

Abstract

Utilizing rapid bioassessment procedures and aquatic physiochemical techniques, a three-year investigation of Sager and Flint creeks was completed. Bioassessment indices and physiochemical parameters of the 2 streams were compared and the effects of urbanization on both watersheds were assessed. Correlating data concerning land usage in both watersheds and alterations of both streams' geomorphology were also utilized to conclude that Sager Creek shows a higher degree of urban stream syndrome than Flint Creek.

Key words:--- Aquatic insects, macroinvertebrates, urban stream syndrome, water quality

Introduction

Urban stream syndrome (Meyer et al. 2005, Walsh et al. 2005, Korminkova 2012) is a condition used to describe the effects of urbanization on stream ecosystems. Symptoms of the syndrome include elevated levels of contaminants and nutrients, altered channel morphology, more frequent occurrences of flood events, and a reduction in biotic richness with a corresponding increase in pollution tolerant species (Paul and Meyer 2001, Meyer et al. 2005).

From 1999-2004, the United States Geological Survey (USGS) conducted a comprehensive study of urban stream syndrome in 9 metropolitan areas around the country. One of the primary objectives of this study was to determine the response of chemical, biological and physical processes to increasing urbanization (USGS 2013). Since temporal studies of increasing urbanization were impossible, similarly sized watersheds, within the same geographic area, were selected to represent a gradient of urbanization. This gradient, called the urban intensity index ranked watersheds from 0 to 100 (low to high) according to the level of urbanization (Falcone et al. 2007). In theory, all continental United States watersheds would fall within the urban intensity index ranking dependent upon each watershed's level of urbanization.

The urban intensity index was computed by analyzing approximately 300 geographic information system (GIS) variables for each watershed (Falcone et al. 2007). This level of analysis would not be possible for all watersheds, thus a precise urban intensity index ranking of many streams may be impossible. However, Steuer (2010) computed a much simpler disturbance metric based off the GIS derived landcover characteristics of watersheds, **{** % impervious surface + (0.15 x (% agriculture + grasslands))**}**, and correlated this to invertebrate diversity to produce a regression curve. According to Steuer (2010), invertebrate diversity sharply declines with increased impervious surfaces and agriculture and grassland cover, changes that are indicative of increased urbanization.

The 40 km^2 Sager Creek watershed is located in an Ozark Highlands Ecoregion of Northwest Arkansas (Omernick 1987). Pastures for grazing or hay production dominate this watershed (55%). The main channel of Sager Creek flows through the city of Siloam Springs and the downtown area is built around it. An estimated 30.5% of the watershed is occupied by urbanized land. Only a small fraction (11%) of the watershed remains forested (AWIS 2006a). An application of the Steuer formula (2010) on the Sager Creek watershed produces a disturbance metric of 38.7.

The somewhat larger Flint Creek watershed (74 km^2) lies adjacent to the Sager Creek watershed on its northern border. Pastures for hay production and grazing also dominate this watershed (53%). The small city of Gentry lies within the Flint Creek watershed, however, the main channel of the stream does not flow through the city limits, and only 7% of the watershed is occupied by urbanized land. Unlike Sager Creek, 35% the Flint Creek watershed is still forested (AWIS 2006b). Based on the Steuer formula (2010), Flint Creek would have a disturbance metric of 15.1.

In a previous publication, the author indicated that wastewater treatment effluent had compromised the integrity of one reach of Sager Creek compared to other portions of the stream (Wakefield 2013). However, there is reason to suspect that the entire Sager Creek watershed may be affected, at some level, by urban stream syndrome as indicated by the Steuer formula. Because of its geographic location and similar land usage, but contrasting reduced amount of urban influence, Flint Creek serves as a reference stream for comparison to the ostensibly more urbanized Sager Creek (ADEQ 1987).

If the Steuer formulation is accurate then Sager Creek should show a higher degree of urban stream syndrome than Flint Creek. The purpose of this study was to utilize physiochemical testing of stream water as well as stream macroinvertebrate populations to test this hypothesis.

Materials and Methods

Both Sager and Flint creeks are relatively small 1-3 order streams (Vannote et al. 1980). Three sampling reaches on each stream, {Honeycutt (Hon), John Brown University (JBU), and Waste Water (WW) for Sager Creek; Ozark Academy (OA), Siloam Springs City Lake (Lake) & North (Nor) for Flint Creek}, were chosen based on accessibility and geomorphic conditions (Fig. 1). Each sampling reach was further divided into 8 riffle-dominated sampling sites, labeled A-H with A being the most downstream site. Sampling of Sager and Flint creeks began in August of 2010 and continued until April of 2013. A total of 16 samples were collected from each reach over the 32 month period.

Macroinvertebrate samples were collected using a 500-µm D-net. At each sample site, the net was placed downstream of the water-flow, and an approximate 0.30 m^2 area in front of the net was kicked for 30 seconds to dislodge organisms. This process was repeated at a different location in the site to insure an adequate collection of organisms. A 0.5 cm^2 mesh rock screen was used to catch large rocks and debris as the net contents were transferred to a bucket. Accumulated rocks, algae or other debris collected in the rock screen, were inspected and observed clinging organisms were removed and placed in the bucket. All clinging organisms found in the net were also placed in the bucket. A 500 µm screen was used to eliminate excess water from the bucket before the final sample was transferred to a collection container and preserved with 95% v/v ethyl alcohol. This process was repeated for all sites, A-H, within each reach. However, due to limited time and assistance during the summer months, collections in June and July were made at only 4 of the 8 sampling sites.

In the laboratory, samples were emptied into a gridded counting tray. A random number generator was used to determine a starting grid and then a 100-organism subsample was separated, identified to the family level (Needham and Needham 1962, Voshell 2002), and recorded. Using a method created by Hilsenhoff (1988) a family-level biotic index (FBI) was generated from each subsample. Sixty-six insect families, in 8 different orders, as well as 2 crustacean groups, (Isopoda and Amphipoda), could be utilized in the production of a FBI. The FBI represented the presence of higher levels of organic pollution with higher numeric values on a scale of 0 to 10. However, the FBI was developed utilizing arthropods native to Wisconsin. To more accurately reflect the sensitivity of the arthropods found in Sager and Flint creeks, organic pollution tolerance values were assigned according to a database provided by the Missouri Department of Natural Resources. These values also ranged from 0 to 10, on a low to high pollution tolerance scale (Sarver 2005).

Utilizing the same subsample from each site, a family-level Simpson's Index of Diversity (SID) was also calculated from each subsample (Simpson 1949). SID indicates the probability of 2 repeated samples being different. In other words, on a scale of 0 to 1, as

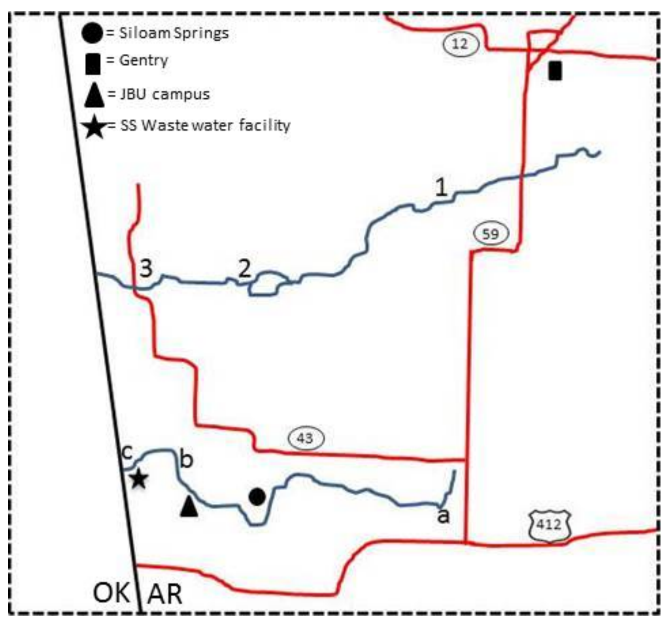

Fig. 1. Map of the study areas in Benton County, AR. Flint Creek study sites; 1=OA, 2=Lake, 3=Nor. Sager Creek study sites; a=Hon, b=JBU, c=WW. Both streams flow east to west.

the diversity within a stream increases the probability that a second sample will be different from the first also increases.

Utilizing all 8 of the individual site FBI and SID, a mean FBI and mean SID was calculated for each reach per sample day. All June and July mean FBI and mean SID were calculated utilizing the 4 individual site's data. Both the mean FBI and mean SID were pooled in 2 different manners for statistical analysis. An overall stream-specific mean FBI (Overall Index) and overall stream-specific SID (Overall Diversity) were calculated by pooling all 48 mean FBI and mean SID values collected over the 3years of study. Also a stream reach-specific mean FBI (Reach Index) and stream reach-specific mean SID (Reach Diversity) were produced utilizing the 16 individual mean FBI and mean SID for each reach.

The mean number of individuals of each recorded family per reach (M/R) was also generated from the total number of individuals identified in each 100 organism subsample. These values were utilized to compare the overall diversity of pollution tolerant versus pollution intolerant species within each stream.

Environmental Protection Agency (EPA) standard procedures were used to calculate stream water flow for both Sager and Flint creeks (USEPA 2004). Physiochemical data was collected using several different methods. A Hanna Instruments HI 991300 Multiparameter Water Quality Meter was used to record stream temperature, pH, electrical conductivity (EC) and total dissolved solids (TDS). Utilizing EPA standard procedures (USEPA 2004), approximately 120 ml of water was collected from each of these sites for additional physiochemical testing. Tests for dissolved oxygen (O_2), (HRDO method 8166), nitrate (NO_3^-), (cadmium reduction method 8039), and phosphate (PO_4^{3-}), (USEPA method 365.2), concentrations were performed on unfiltered water using a Hach™ colorimeter (model DR/850). All tests were performed three times from randomly selected sites within each reach. A mean value for each parameter was then calculated and recorded. Mean values for each parameter were then pooled in the same manner as mean FBI and mean SID to produce an overall stream-specific mean (Stream Mean) and a stream reach-specific mean (Reach Mean) for each parameter.

Overall Index, Overall Diversity, M/R and Stream Mean data for Sager Creek versus Flint Creek were all compared using paired t-tests with an α=0.05. Reach Index, Reach Diversity, and Reach Mean values for each parameter were first tested with an ANOVA (α=0.05). Then, for each parameter, paired t-tests were performed between each Sager Creek reach compared to each Flint Creek reach. To avoid a Type I error, the Bonferroni Correction was applied to all stream reach-specific comparisons (α = 0.016) (Triola and Triola 2006).

Results

Table 1. The overall stream-specific mean FBI (Overall Index), overall stream-specific mean SID (Overall Diversity) and overall mean for each physiochemical parameter (Stream Mean), n=48. Diff= t-test results; ppm= parts per million; μS= microsiemen

Parameter	Sager $\bar{X}$±SE	Flint $\bar{X}$±SE	Diff
Overall Index	4.97±0.059	4.84±0.074	n.d.
Overall Diversity	0.723±0.017	0.805±0.015	*p=1.12E-04*
Stream Mean Waterflow (m^3/s)	0.479±0.058	0.956±0.108	*p=1.91E-06*
Stream Mean TDS (ppm)	164.39±9.46	122.86±2.08	*p=1.43E-05*
Stream Mean EC (μS)	327.96±19.16	247.50±3.95	*p=3.13E-05*
Stream Mean Temp. (^oC)	17.71±0.691	17.17±1.27	n.d.
Stream Mean pH	7.66±0.079	7.80±0.065	*p=3.04E-02*
Stream Mean NO_3^- (ppm)	2.92±0.155	3.05±0.168	n.d.
Stream Mean PO_4^{3-} (ppm)	0.512±0.098	0.171±0.012	*p=8.00E-04*
Stream Mean O_2 (ppm)	10.55±0.244	10.07±0.267	*p=2.48E-02*

Biotic Index.--- All 8 of the insect orders and both crustacean groups utilized by Hilsenhoff (1988) were collected in this study. However, only 30 of the 66 distinct families were collected. There was no difference in the Overall Index between Sager Creek and Flint Creek (Table 1). These values fell within the "good" ranking on the Hilsenhoff (1988) FBI scale, and would suggest that both streams are showing some level of organic pollution. However, the ANOVA of the Reach Index from all 6 reaches did indicate a statistical difference and the subsequent t-test analysis revealed that although both streams have some reaches with organic pollution, Sager Creek seems to have higher levels (Table 2 and Fig. 2).

Table 2. The stream reach-specific mean FBI (Reach Index), stream reach-specific mean SID (Reach Diversity) and stream reach-specific mean (Reach Mean) physiochemical comparisons, n=16. Significant differences in ANOVA values are indicated for comparisons on all six reaches. Comparisons of individual Sager Creek reaches (SCR) and Flint Creek reaches (FCR) are indicated in each row. P-values in grey boxes with bold type indicate that the SCR had the larger mean value. ppm= parts per million; μS/cm= microsiemen per centimeter.

Parameter	ANOVA	SCR	FCR	Difference
Reach Index	*p=9.94E-11*	Hon	OA	***p=1.25E-05***
			Lake	*p=9.73E-05*
			Nor	nod.
		JBU	OA	***p=4.46E-03***
			Lake	*p=2.18E-05*
			Nor	nod.
		WW	OA	***p=2.55E-09***
			Lake	nod.
			Nor	***p=1.22E-05***
Reach Mean Waterflow (m^3/S)	*p=4.09E-03*	Hon	OA	*p=2.73E-05*
			Lake	*p=2.73E-03*
			Nor	*p=9.75E-04*
		JBU	OA	*p=2.15E-03*
			Lake	*p=1.37E-03*
			Nor	nod.
		WW	OA	nod.
			Lake	nod.
			Nor	nod.
Reach Mean TDS (ppm)	*p=1.17E-22*	Hon	OA	*p=5.59E-03*
			Lake	nod.
			Nor	*p=2.21E-03*
		JBU	OA	***p=4.29E-05***
			Lake	***p=2.37E-07***
			Nor	***p=6.52E-04***
		WW	OA	***p=4.28E-07***
			Lake	***p=3.80E-07***
			Nor	***p=7.63E-07***
Reach Mean EC (μS)	*p=1.13E-21*	Hon	OA	*p=1.48E-03*
			Lake	*nod.*
			Nor	*p=2.66E-03*
		JBU	OA	***p=6.17E-04***
			Lake	***p=9.78E-06***
			Nor	***p=3.40E-03***
		WW	OA	***p=6.16E-07***
			Lake	***p=6.21E-07***
			Nor	***p=1.31E-06***
Reach Mean Temp (°C)	nod.			

Parameter	ANOVA	SCR	FCR	Difference
Reach Diversity	*p=7.49E-08*	Hon	OA	*p=1.34E-07*
			Lake	nod.
			Nor	*p=4.46E-03*
		JBU	OA	*p=1.12E-04*
			Lake	nod.
			Nor	*p=8.57E-03*
		WW	OA	*p=6.32E-05*
			Lake	nod.
			Nor	*p=3.18E-04*
Reach Mean pH	*p=2.72E-07*	Hon	OA	*p=4.79E-05*
			Lake	*p=4.52E-05*
			Nor	*p=5.42E-05*
		JBU	OA	***p=1.03E-02***
			Lake	nod.
			Nor	***p=3.63E-03***
		WW	OA	nod.
			Lake	nod.
			Nor	nod.
Reach Mean NO_3^- (ppm)	*p=1.04E-05*	Hon	OA	nod.
			Lake	nod.
			Nor	*p=6.41E-03*
		JBU	OA	*p=2.73E-05*
			Lake	*p=1.22E-03*
			Nor	nod.
		WW	OA	nod.
			Lake	nod.
			Nor	***p=2.84E-03***
Reach Mean PO_4^{3-} (ppm)	*p=8.64E-12*	Hon	OA	nod.
			Lake	nod.
			Nor	nod.
		JBU	OA	nod.
			Lake	nod.
			Nor	nod.
		WW	OA	***p=3.64E-04***
			Lake	***p=2.97E-04***
			Nor	***p=2.61E-04***
Reach Mean O_2 (ppm)	n.d.			

Table 3. List of orders and families of aquatic insects and crustacean taxa collected, identified, and counted in Sager and Flint creeks. Numbers at the end of each taxon indicates the pollution-tolerance value according to Sarver (2005). Mean number of individuals of each recorded family per reach (M/R) value indicates either no significant difference in the abundance between the streams (n.d.) or the stream in which the taxon dominated (Sager Creek=SC, Flint Creek-FC) and the p-value of the difference in abundance; n=48 for each mean calculation.

Insecta							
Coleoptera	**M/R**	**Diptera**	**M/R**	**Ephemeroptera**	**M/R**	**Lepidoptera**	**M/R**
Elmidae(4)	nd	Ceratopogonidae(6)	nd	Baetidae(4)	SC *p=7.6E-03*	Pyralidae(5)	nd
Psephenidae(4)	nd	Chironomidae(6)	SC *p=1.0E-02*	Caenidae(7)	nd		
		Empididae(6)	nd	Ephemiridae(4)	FC *p=1.5E-03*		
		Simuliidae(6)	nd	Heptageniidae(4)	nd		
		Tipulidae(3)	FC *p=9.7E-03*	Isonychiidae(2)	FC *p=2.1E-06*		
				Leptohyphidae(4)	FC *p=3.6E-04*		
				Leptophlebiidae(2)	nd		
Megaloptera		**Odonata**		**Plecoptera**		**Trichoptera**	
Corydalidae(4)	FC *p=1.02E-07*	Calopterygidae(5)	nd	Capniidae(1)	FC *p=3.2E-03*	Helicopsychidae(3)	FC *p=5.2E-04*
Sialidae(7.5)	nd	Coenagrionidae(9)	SC *p=3.6E-06*	Perlidae(3)	FC *p=2.4E-09*	Hydropsychidae(4)	nd
		Gomphidae(7)	FC *p=7.0E-04*			Hydroptilidae(4)	nd
		Libellulidae(9)	nd			Leptoceridae(4)	nd
Crustacea						Limnephilidae(3)	nd
	M/R					Philopotamidae(3)	nd
Amphipoda(6.9)	FC *p=4.5E-04*					Polycentropidae(6)	nd
Isopoda(8)	FC *p=5.6E-05*						

The Hon, JBU, and WW Reach Indices were all significantly higher when compared to the OA Reach Index (Table 2). The WW Reach Index was also significantly higher than the Nor Reach Index (Table 2). However, the Lake Reach Index was significantly higher than both the Hon and JBU Reach Indices (Table 2). There were no significant differences between the other comparisons.

Diversity Index.--- The t-test analysis of the Overall Diversity of Sager Creek and Flint Creek indicated that Flint Creek had significantly higher diversity than Sager Creek (Table 1). As expected, the ANOVA of the Reach Diversity from all 6 reaches also indicated a statistical difference (Table 2). The t-test analysis of the 6 reaches also revealed statistical difference between most reaches (Fig. 3). The Reach Diversity for OA was significantly higher than that of the Hon, JBU and WW reaches (Table 2). The Nor Reach Diversity was also significantly higher than that of the Hon, JBU and WW reaches (Table 2). Only the Lake reach showed no significant difference with any of the Sager Creek reaches.

Overall Diversity.--- The t-test analysis of the M/R values for each of the insect families and 2 crustacean taxa revealed no significant differences in 18 of the 32 groups. However, 14 groups did show significant differences in abundance per stream. Of these, 3 groups were significantly more abundant in Sager Creek, while 11 were more abundant in Flint Creek. Of the 3 Sager Creek groups, 2 ranked in the top-half

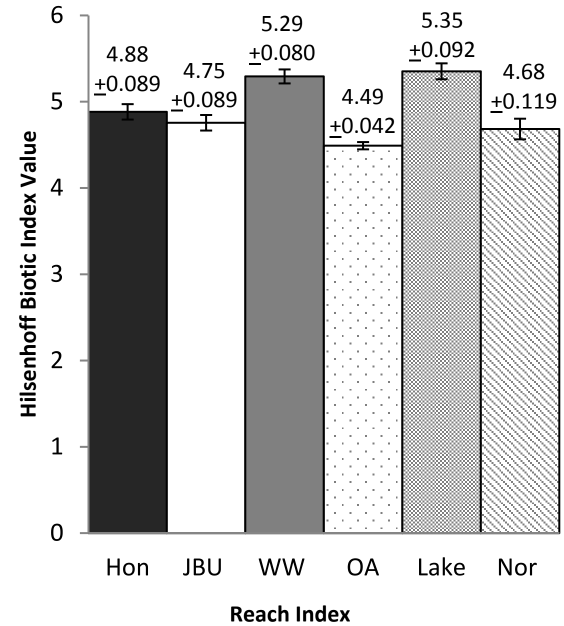

Fig. 2. The stream reach-specific mean FBI (Reach Index) for both Sager (Hon, JBU, WW) and Flint (OA, Lake, Nor) reaches. Standard error bars, and mean ± standard error are indicated.

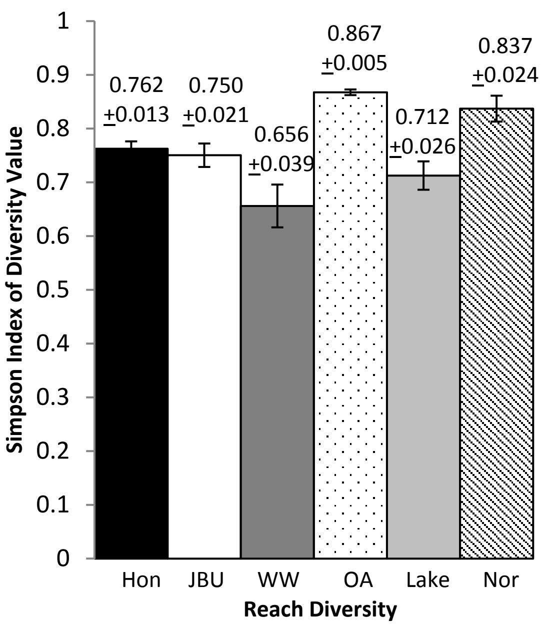

Fig. 3. The stream reach-specific mean SID (Reach Diversity) for both Sager (Hon, JBU, WW) and Flint (OA, Lake, Nor) reaches. Standard error bars, and mean ± standard error are indicated.

(6-10) of the pollution tolerance values. Of the 11 Flint Creek groups, only 3 were ranked in the top-half of pollution tolerance while 8 were ranked in the lower half (0-4) (Table 3).

Physiochemical Parameters.---The t-test analysis of the Stream Mean for water-flow and all tested physiochemical properties are found in Table 1. The most significant differences were seen in water-flow, TDS, EC and dissolved PO_4^{3-}. Significant differences were also found in pH and dissolved O_2. Only temperature and dissolved NO_3^- showed no significant differences at the overall stream level.

ANOVA tests of Reach Mean for water-flow and all tested physiochemical properties are found in Table 2. As in the Stream Mean, the ANOVA for the Reach Mean for temperature showed no significant differences across all 6 stream reaches. Interestingly, the Reach Mean for dissolved NO_3^- did show significant differences at the stream-reach level counter to what was seen at the overall-stream level, and the Reach Mean for dissolved O_2 did not show any significant differences at the stream-reach level, counter to the overall-stream level. The largest differences were seen in water-flow, TDS, EC and dissolved PO_4^{3-}.

Table 2 also contains the t-test analyses of the Reach Mean, for all physiochemical parameters, across the 6 stream reaches. These tests reveal that the Flint Creek flow is relatively stable throughout the study area. However, Sager Creek begins with relatively low flow and increases throughout the study area (Fig. 4). Flow in the Hon reach was significantly lower when compared to the OA, Lake, and Nor reaches. The same was true of the JBU reach when compared to the OA and Lake reaches, however it was not significantly different from the Nor reach. Only the WW reach had sufficient flow to show no significant difference with any of the Flint Creek reaches (Table 2).

T-tests of the Reach Mean for TDS revealed that Flint Creek has a relatively stable level of TDS, while Sager Creek has an ever-increasing level throughout the study area (Fig. 5). It also revealed that there were significant differences in almost every comparison. Only the Hon reach when compared to the Lake reach showed no significant differences (Table. 2).

Electrical conductivity (EC) is directly correlated to TDS, thus it is not surprising that the t-test analyses of the Reach Mean for EC are essentially the same as for TDS. EC is relatively stable throughout the Flint Creek study area while showing an ever increasing level throughout Sager Creek (Fig. 6). Again, with the exception of the Hon to Lake comparison, all other cross stream-reach comparisons showed significant differences (Table 2).

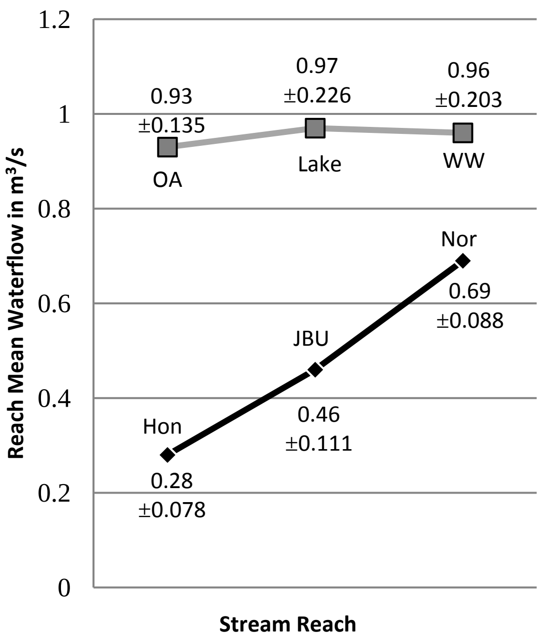

Fig. 4. The stream reach-specific mean (Reach Mean) for water-flow in both Sager (Hon, JBU, WW) and Flint (OA, Lake, Nor) reaches. Mean ± standard error are indicated.

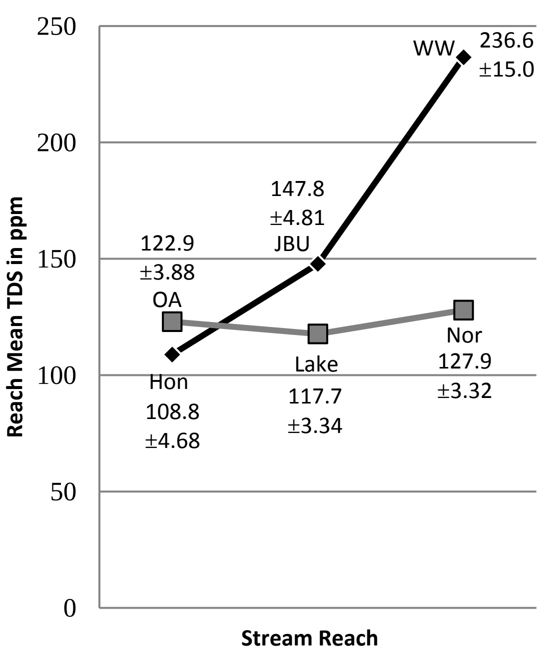

Fig. 5. The stream reach-specific mean (Reach Mean) for total dissolved solids (TDS) in both Sager (Hon, JBU, WW) and Flint (OA, Lake, Nor) reaches. Mean ± standard error are indicated. ppm=parts per million.

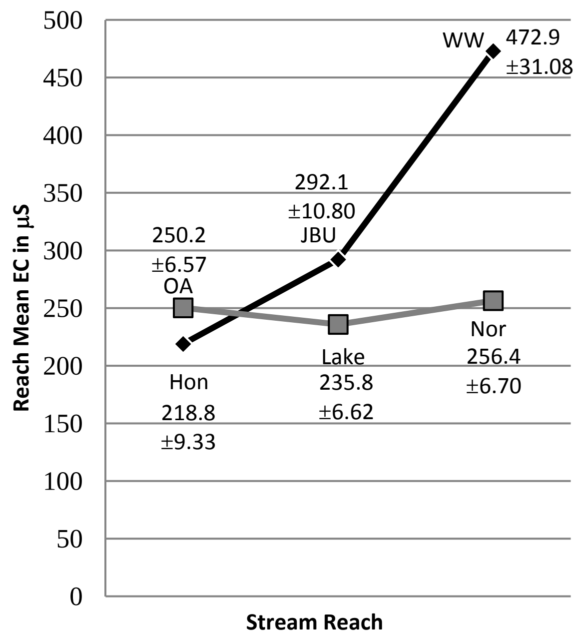

Fig. 6. The stream reach-specific mean (Reach Mean) for electrical conductivity (EC) in both Sager (Hon, JBU, WW) and Flint (OA, Lake, Nor) reaches. Mean ± standard error are indicated. µS/cm= microsiemens per centimeter.

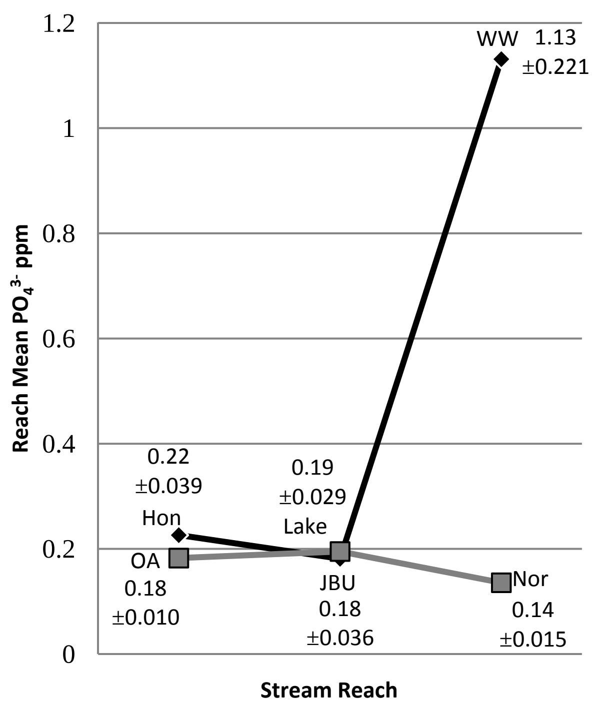

Fig. 7. The stream reach-specific mean (Reach Mean) for dissolved PO_4 in both Sager (Hon, JBU, WW) and Flint (OA, Lake, Nor) reaches. Mean ± standard error are indicated. ppm=parts per million.

T-test analyses of the Reach Mean for dissolved PO_4^{3-} revealed that levels remain relatively stable and constant throughout all of the Flint Creek study area and most of the Sager Creek study area. However an extremely dramatic change in dissolved PO_4^{3-} is seen in the WW reach of Sager Creek (Fig. 7). This reach's level of dissolved PO_4^{3-} was significantly higher than all of the dissolved PO_4^{3-} levels of all 3 Flint Creek reaches (Table 2). Although not included on this table, further analysis of the WW reach indicated that its dissolved PO_4^{3-} level was significantly higher than both the Hon reach ($p=6.67E\text{-}04$) and the JBU reach ($p=5.35E\text{-}04$).

Of the physiochemical parameters tested, Reach Mean values for pH and dissolved NO_3^- seemed to have the most stream specific variation. The pH Reach Mean of the Hon reach was only slightly basic, but increased for the JBU reach and held stable for the WW reach. However the pH Reach Mean of the OA reach was more basic than the Hon reach, then increased substantially at the Lake reach before dropping down to its original level at the Nor reach (Fig. 8). T-test significant differences between the pH Reach Mean values can be seen in Table 2.

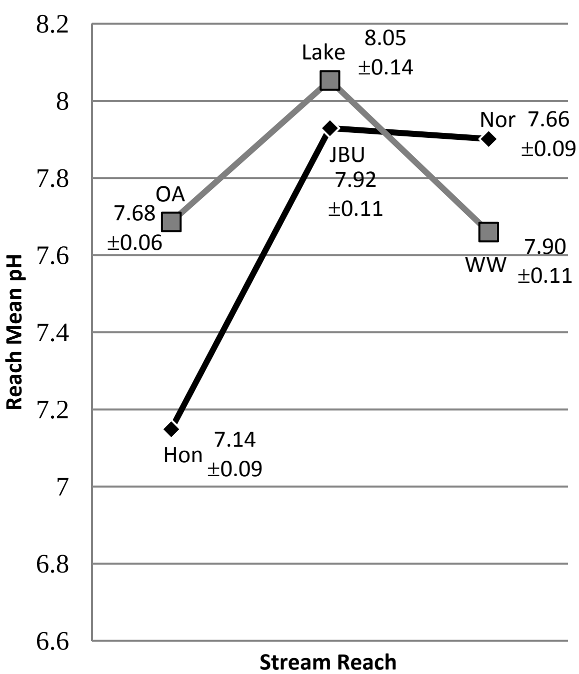

Fig. 8. The stream reach-specific mean (Reach Mean) for pH in both Sager (Hon, JBU, WW) and Flint (OA, Lake, Nor) reaches. Mean + standard error are indicated.

Finally, the t-tests of the Reach Mean for dissolved NO_3^- revealed that FC had a steadily decreasing value throughout the study area while SC had much more erratic values. Both the Hon and WW reaches had comparable dissolved NO_3^- levels while the JBU dissolved NO_3^- level was substantially lower (Fig. 9). Significant difference in the levels of dissolved NO_3^- can be seen in Table 2.

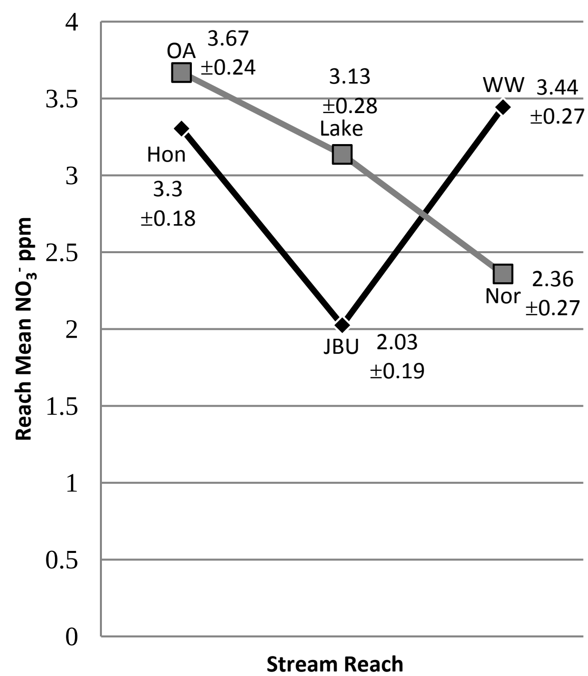

Fig. 9. The stream reach-specific mean (Reach Mean) for dissolved NO_3 in both Sager (Hon, JBU, WW) and Flint (OA, Lake, Nor) reaches. Mean + standard error are indicated. ppm=parts per million.

Discussion

According to Paul and Meyer, (2001), streams that are impaired by urban development (i.e. urban stream syndrome) should see effects in three critical areas; physical, chemical and biological. Although some of these effects are seen in both Sager and Flint creeks, Sager Creek shows a higher degree of urban stream syndrome than Flint Creek.

Physical.---Siloam Springs, AR was incorporated as a township in 1881. At that time, much of the industry of the city revolved around tourism as many of the springs that fed into Sager Creek were advertised to have "healing properties". Thus, much of the downtown area was built directly on or around Sager Creek (Warden 2010). As the town has matured into a small city the amount of impervious surface (buildings, roads, parking lots, etc.,) that covers the watershed has increased substantially.

In addition, no less than 3 dams were constructed across the stream channel to make the water more accessible to citizens, with one dam currently still in place. In 1892 a severe flood took the lives of 3 citizens and destroyed much of the Siloam Springs downtown area (Warden 2010). As a result the main flow of the stream was channeled, first by the construction of stone and mortar walls and later by concrete retaining walls. At least 20 bridges have also been built across Sager Creek to allow for the passage of cars, trains, golf carts and pedestrians.

The main flow of the stream begins from an underground aquifer called Box Spring. The water from Box Spring emerges onto the Siloam Springs city golf course where it serves as a “water hazard” for approximately 325 m. A previous water quality assessment of Box Springs (GBM[c] & Associates 2005) is available, however, a direct comparison between this historic study and the current study is difficult at best. In the current study, no samples were collected directly from Box Spring. The closest sampling site was the Honeycutt site which is downstream from the golf course. Also, in the historic study, the physiochemical tests utilized to test water quality were different from the tests used in the current study. However, for those tests that were similar some variations between the historic Box Springs water quality and the current Honeycutt site water quality are apparent. Honeycutt site water is slightly more acidic (pH 7.14) than historic Box Spring water (pH 6.3). Honeycutt site water contains less NO_3^- (3.3 ppm) than historic Box Spring water (5.1 ppm), but Honeycutt site water contains more PO_4^{3-} (0.22 ppm) than historic Box Spring water (0.052 ppm).

Approximately 11.4×10^7 liters/day of waste water effluent is released into Sager Creek from the Siloam Springs wastewater treatment plant (Wakefield 2013). All of these physical alterations to the natural hydrology and geomorphology of Sager Creek are found along the ~5.2 km study area.

The ~7.04 km Flint Creek study area has some of the same urban disturbances that are seen in Sager Creek only to a greater degree. For example, Flint Creek flows through a golf course for approximately 2.0 km. It has 3 intact dams; the largest of these was constructed in 1946 and forms the Siloam Springs City Lake. The spillway of the dam alters the flow of Flint Creek from its original stream bed, forcing an alternate route for approximately 350 m before rejoining the original stream bed. Water from the lake was originally utilized as a drinking water reservoir, but is currently employed as coolant for the nearby Southwestern Electric Power Company (SWEPCO) power plant. Between 6-10 million gallons of water is cycled through the power plant every day (Siloam Spring 2009).

However, Flint Creek shows either a reduced amount or complete lack of certain physical effects when compared to Sager Creek. For example, there are only 7 bridges for car or railroad traffic in the Flint Creek study area. This is a much smaller number of bridges per stream area, compared to Sager Creek; (*Flint Creek ~1 bridge/km of stream versus Sager Creek ~4 bridges/km of stream*). The city of Gentry is located within the Flint Creek watershed, but no part of the city is built around the main flow of the stream. Thus the amount of impervious surface surrounding Flint Creek is much smaller when compared to Sager Creek. In addition, most notably, the wastewater effluent from Gentry does not empty into Flint Creek.

There were only 2 physical effects that the present study investigated: temperature and water flow. It is plausible to expect that the Flint Creek Lake reach, which is just downstream of the Siloam Springs City Lake, would show elevated temperature due to the lake water’s usage as coolant for the SWEPCO power plant. However, water removed from the lake for coolant is not returned to the lake but is instead released into a separate watershed. Thus it is not surprising that all of the statistical analyses for temperature showed no statistical differences between the streams or across any of the individual stream reaches.

Flint Creek has a relatively stable flow rate throughout the study area while Sager Creek shows an ever increasing rate of flow (Fig. 4). The percentage increase due to natural sources, (such as the influx of groundwater or the confluence of small springs), versus urban disturbances, (such as runoff from impervious surfaces or the influx of waste water effluent), is not known. However, a quick correlation study failed to show strong relationships between water flow and any other studied parameter. Other studies, however, have demonstrated that the types of physical changes described above result in detrimental effects on the hydrology and geomorphology of streams (Neller 1988, Booth and Jackson 1997, Hart and Finnelli 1999, Meyer and Wallace 2001, Brueggen-Boman and Bouldin 2012). Indirect effects of these physical alterations would also be reflected in changes within the biological systems (Klein 1979). For example, the level of impervious surface covering a watershed has become a very accurate predictor of urban impacts on streams (McMahon and Cuffney 2000).

Chemical.---Several previous studies have indicated that urbanization of streams tends to increase almost all chemical constituents within the stream including levels of dissolved metals, hydrocarbons, ammonium, dissolved solids, electrical conductivity and oxygen demand (Porcella and Sorenson 1980, Lenat and Crawford 1994, Latimer and Quinn 1998, USGS 1999). Many of these constituents were not investigated in this study. However, the effects of urbanization were strongly indicated in Sager Creek by the changing levels of total dissolved solids (TDS), electrical conductivity (EC) and dissolved PO_4^{3-}.

EC is a literal measurement of a solutions ability to conduct an electrical current. The number of dissolved ions in the solution (i.e. TDS), will obviously have a direct impact on the EC. The correlation between the two measurements is generally accepted as TDS (ppm) * 2 = EC (μS/cm) (McPherson 1995).

In laboratory studies, elevated levels of TDS have been shown to be detrimental to aquatic life. The mean TDS of rivers around the world is 120 ppm and detrimental effects on invertebrates have been detected at TDS levels of 280 ppm. However, the detrimental effects are both ion and species specific (Weber-Scannell et al. 2007). In lotic environments, pollution intolerant families of Ephemeroptera, Plecoptera and Trichoptera, seemed to be the most effected by elevated TDS. In general, a reduction in overall stream diversity was inversely correlated with an increase in populations of pollution tolerant macroinvertebrates (Timpano et al. 2010).

The results of this study indicate stable levels of TDS and EC within Flint Creek, while Sager Creek shows consistently elevating levels of both parameters (Figs. 5 and 6). The ions responsible for these elevated levels in Sager Creek are unknown, however, in other urban areas, elevated levels of these parameters are consistent with waste water treatment effluent and non-point source runoff from impervious services (Paul and Meyer 2001).

Waste water treatment effluent can also be a major source of dissolved PO_4^{3-} (LaValle 1975). The effluent from the Siloam Springs waste water treatment plant seems to have a significant influence on the levels of dissolved PO_4^{3-} compared to all other reaches within both Sager Creek and Flint Creek (Fig. 7). A previous publication, (Haggard et al. 2004) had already demonstrated this elevated PO_4^{3-} level downstream from the Siloam Springs waste water treatment plant. However, at that time, a limit for the amount of PO_4^{3-} that could be released from the plant had not been established. In December of 2009, the EPA's National Pollutant Discharge Elimination System (NPDES) permit program established a 30 day average limit of 1.0 ppm PO_4^{3-} release. In 2005, the annual average PO_4^{3-} released at the Siloam Springs waste water treatment plant was 3.5 ppm. Since 2013 the average annual release has been 0.4 ppm; an 88% reduction in PO_4^{3-} release (Myers 2014). However, in 2013, monthly averages ranged as low as 0.135 ppm up to 0.761 ppm with 4 months above 0.66 ppm. The NPDES permit also allows the treatment plant's weekly average to be as high as 1.5 ppm and still be in compliance (Myers 2014). Since the PO_4^{3-} samples in this study were grab samples collected and processed in a single day, and these samples represent the sum of all the PO_4^{3-} found in each creek rather than the PO_4^{3-} lever in effluent only, it is understandable how the PO_4^{3-} levels below the plant could be as high as indicated.

Though PO_4^{3-} is an essential nutrient for all forms of life, elevated levels can overstimulate algal growth, which can result in substantial trophic changes in the stream (USEPA 2010). Thus, an elevated level of PO_4^{3-}, particularly from waste water treatment effluent, is a definitive indicator of urban disturbance (USGS 1999, Winter and Duthie 2000).

Effects of urban stream syndrome were not clearly reflected in the measured values for pH (Fig. 8). Although there were significant differences between measured values of pH in the reaches of Sager Creek versus Flint Creek, all of the values fell within the suitable range of pH values (6.5-9.0) as established by the EPA (USEPA 1986). This was not surprising as most changes in the pH of urban streams occur during rain events.

Many urban areas have combined sewers that collect domestic sewage, industrial wastewater and rainwater runoff. This wastewater is then transported to a wastewater treatment facility. However during heavy rain events, the volume of waste water may be greater than the capacity of the sewer system or treatment plant. These combined sewers are designed to overflow during these events and discharge wastewater directly into the nearby streams (USEPA 2012a). During these events, swings in pH values, (4-8.7) can be seen depending on the amount of storm water versus domestic sewage is found in the combined sewer overflow (Kominkova 2012). Since no samples in this study were taken during rain events, significant changes in pH were not expected.

Streams effected by urban stream syndrome typically show elevated levels of dissolved NO_3^- (USGS 1999). However, this study did not find consistently high levels of NO_3^- in any of the studied reaches (Fig. 9). There were significant differences between some of the reaches, (Table 3), however, none of the dissolved NO_3^- levels are particularly alarming as none of them approach the maximum contamination levels of 10 ppm in drinking water (USEPA 2012b). Thus, the levels of dissolved NO_3^- in Sager Creek and Flint Creek do not clearly indicate urban disturbance in either stream.

Low levels of dissolved NO_3^- may be one of the reasons why levels of dissolved O_2 were high in both streams. Although a significant difference between the Sager Creek dissolved O_2 Stream Mean and Flint Creek dissolved O_2 Stream Mean was indicated (Table 2), both stream's Stream Mean values were slightly higher than maximum levels of dissolved O_2 for the mean stream temperatures (USEPA 2012c). When analyzed at the stream reach level, no significant differences were indicated for either stream (Table 3). High levels of NO_3^- laden pollution typically causes depleted O_2 levels within aquatic systems (Daniel et al. 2002, Kominkova 2012). Since there was no indication of consistently high levels of dissolved NO_3^- in either stream, it's probable that dissolved O_2 levels were equilibrated with atmospheric O_2 levels throughout both streams. Therefore, the levels of dissolved O_2 in both Sager and Flint creeks also do not indicate urban disturbance.

Biological.---The effects of urbanization on biological organisms has been demonstrated in microbes, invertebrates, fish, algae and plants. However more work seems to have been done on invertebrates than any other group (Paul and Meyer 2001). The general effect of urbanization is an overall decrease in invertebrate diversity. This is especially true in the sensitive orders of Ephemeroptera, Plecoptera and Trichoptera. However, pollution tolerant invertebrates such as the Chironomidae, oligochaete worms and some stream gastropods actually increase in abundance due to urbanization (Pratt et al. 1981, Hachmoller et al 1991, Thorne et al. 2000).

The results of this study confirmed these same results. Although the Overall Index of both SC and FC were not significantly different (Table 2.), the Reach Index of Sager Creek's reaches showed a strong tendency to be higher (i.e. more organic pollution) than the Flint Creek reaches. The one exception to this trend was the Flint Creek Lake Reach Index (Fig. 2). This reach is just downstream of the Siloam Springs City Lake and therefore shows the greatest level of physical disturbance. The approximately 350 m of altered flow is often across bedrock material rather than the gravel and cobble stream bed that dominates all other reaches. This reach's altered geomorphology is assumed to be the reason for the unusually high Reach Index when compared to the other Flint Creek reaches.

The Overall Diversity of Flint Creek compared to Sager Creek, however, is an indication of urban disturbance as there is an overall decrease in macroinvertebrate diversity in Sager Creek (Table 2). Additionally, the M/R values in Table 1 indicate the higher level of diversity within the Flint Creek reaches compared to the Sager Creek reaches. These results are consistent with a stream showing urban stream syndrome, particularly the increase in pollution tolerant arthropods in the Sager Creek reaches compared to the Flint Creek reaches and the lack of pollution intolerant arthropods, especially the Plecopterans, in Sager Creek.

Conclusion

The results of this study corroborated the findings of the Steuer's (2010) formula, specifically that Sager Creek shows a much higher degree of urban stream syndrome than Flint Creek. In recognition of the declining health of Sager Creek, the city of Siloam Springs has taken measures to improve the water quality of the Sager Creek watershed. This includes multimillion dollar improvements to the Siloam Springs wastewater treatment plant, the purchasing of land and the creation of wetlands along the headwaters and tributaries of Sager Creek, riparian zone restoration along the main channel of the stream, and the removal of one low-water bridge (Della Rosa 2010a,b). However, substantial improvement in the overall health of Sager Creek may require even more drastic measures and considerable time. According to Steuer's (2010) formula, a disturbance metric of 15 is the threshold where invertebrate taxa richness begins to dramatically decline and watersheds with rankings over 30 were found on the segment of the regression curve with the lowest slope. Thus, significant investments in mitigating activity such as the restoration of forested land within the Sager Creek watershed as well as more extensive wetlands may be necessary to see much improvement in stream health (Moore and Palmer 2005).

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Measuring Pain Withdrawal Threshold using a Novel Device in "Pseudo-continuous" Mode

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Running title: Pain Withdrawal Measurements in "Pseudo-continuous" Mode

Abstract

The study of pain and analgesia is an important area of biomedical research that has led to a significant number of advances in the treatment of acute and chronic pain. This study introduces a novel approach to mechanical testing of pain withdrawal of a rat hind paw to a stimulus. This systematic method involves a modified electronic esthesiometer controlled by an IDEA drive that allows for consistency in experiments. The device gives the experimenter computer control of the step size and velocity of approach of the probe stimulus. We discuss here some of the limitations in the current techniques used and illustrate how this device will result in reduced errors during an experiment. The standard method primarily involves manually raising the probe towards the animal. The data presented herein shows how the computer controlled *pseudo-continuous* mode of operation is effective in determining the pain threshold with a lesser deviation from the mean.

Introduction

In 1864, three surgeons, S. W. Mitchell, G. R. Morehouse, and W. W. Keen, produced one of the first publications addressing neurological disorders in their book, "Gunshot wounds and other injuries of the nerves." This book is one of the first publications to address the idea that neurological disorders can be characterized by pain in the affected area (Xinning et al. 2014). Today, clinical and basic science research shows that chronic neuropathic pain is caused by lesions in the peripheral or central nervous systems present in many varied forms (Dworkin et al 2003, Kim and Chung 1992). The behavioral study of pain has led to a significant number of advances in the treatment of acute and chronic pain. These types of experiments include measuring the withdrawal threshold of limb to a thermal, mechanical, electrical, or chemical stimulus. Mechanical testing of pain response can reveal either mechanical allodynia or hyperalgesia. A limb withdrawal in response to a light touch, a pressure, or a brushing evidences allodynia, which is pain to a normally non painful stimuli (Bove 2006). Hyperalgesia is increased sensitivity pain as a result of peripheral nerve damage.

The current method used for quantifying mechanical pain is based on an early esthesiometer, developed by the German physiologist M. von Frey who utilized horse hairs of varying lengths and diameters that would buckle under a specific force. The pain threshold was determined as the bending force of the weakest filament applied that resulted in limb withdrawal in the tested animal or human. Recently the horse hair has been replaced with nylon (Semmes and Weinstein monofilaments) with increasing diameters that bend when a specific value is reached (Weinstein 1993). The advent of electronic force transducers has produced new forms of esthesiometry; these are either electronically controlled by a motor or manually moved by the experimentalist. To use an electronic esthesiometer, a motor controls a probe, to which a force transducer is connected. The probe applies pressure in a linear motion to an area of skin until the threshold is reached, at which point the subject moves the limb and the probe is removed (Moller et al. 1998). Although both the electronic and manual method of esthesiometry rely on the transference of a force, there is a difference in the outcome of the methods. For example, transfer of force in the manual system may not be constant each time (Chong and Cros 2004), while the electronic system may have a rather continuous motion, and thus monitor a true reaction to the stimulus. The purpose here is to present a novel method of measuring and testing hyperalgesia to a mechanical stimulus. This proposition is aimed to increase experimental sensitivity and reproducibility.

Materials and Methods

The device consists of a captive actuator with a 38.1 mm (1.5 in.) stroke. An anti-rotation cap allows the shaft to actuate without an external guide mechanism and is designed to lift up to 2 kg of mass. The mass of the transducer atop the motor is 100 g. This mass plus the reaction force on the animal's plantar surface are within the limits of the motor. The minimum step size is 0.006 mm. The motor is computer controlled using a programmable IDEA drive (HaydonKerk Motion Solutions). The drive is electronic with a fully programmable control unit that uses a Graphic User Interface, giving the experimenter access to control the rate and size of the steps.

The cylindrical force transducer delivers the stimulus on the same plane and axis as the linear actuator (Figure 1). The flow chart in Figure 1 shows the basic electronic schematic. The data acquisition card varied between the WINDAQ system and the CLAMPEX data acquisition systems. The design is flexible to work with any signal data acquisition systems available.

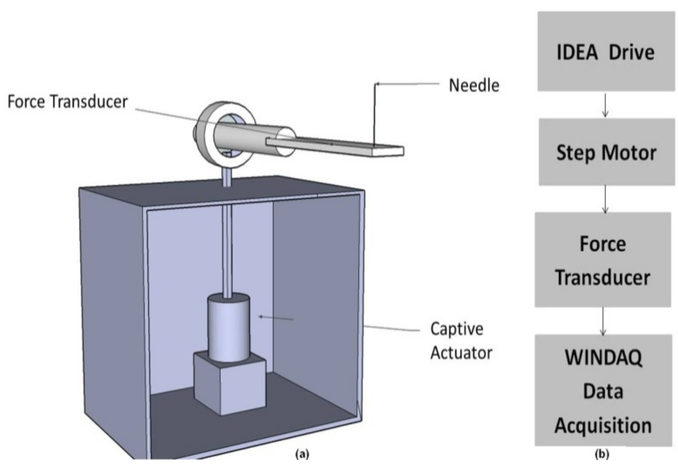

Figure 1. (a) Diagram of the automated device used the measure the PWT. (b) Flow chart of the components in the design.

The experiment was performed on Sprague-Dawley rats, under IACUC protocol #3393 (Evaluation of efficacy of novel analgesic compounds in rat model of neuropathy, University of Arkansas for Medical Sciences). The rats were kept in the test cages for at least 30 minutes prior to experiment. This allowed them to acclimatize to the test environment. They were resting during the experiments.

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The experiments were conducted on three male rats and each test was repeated five times for each rat. The device was placed under each rat and the probe approached the rat at a rate of 1mm/sec. The experiment stopped after the hind paw was withdrawn indicating the pain threshold. The data were compared using the SigmaStat statistical package.

Results and Discussion

The motor functions in a *pseudo-continuous* mode. This means that during testing, the very small increments in the step size will be an almost "continuous" motion. The exact force representing the pain threshold can be determined since the experiment is identical for each test. The results presented here show how the technique allows for the maximum threshold force to be determined as well as the reduced deviation from the mean values.

Figure 2 shows typical results from the experiments. The background noise shown is due to small vibrations that may exist in the laboratory, which is generally averaged out. . The actual force can be determined using Newton's second law, $F = mg$, where m is the mass (kg) and g = 9.8m/s^2.

Figure 2a and 2b show an increase in force, followed by a sharp drop. The peak is recorded as the pain threshold response for that experiment. From the similarities in the results, it can be seen that the programmable esthesiometer does not alter the experiment but rather the way the data is recorded: the force is increased in set increments. The manual esthesiometer has been considered to move in incremental steps as well, however, the experimenter's approach to the animal is subjective and may not always be constant.

Figure 3 shows the overall data obtained from 5 experiments on 3 different rats using the automated programmable esthesiometer and the manual esthesiometer. In each case the experiment was performed 5 times. The error bars represent the standard deviation of the values obtained from the experiment and will be used here to compare the two different methods to determining the pain withdrawal

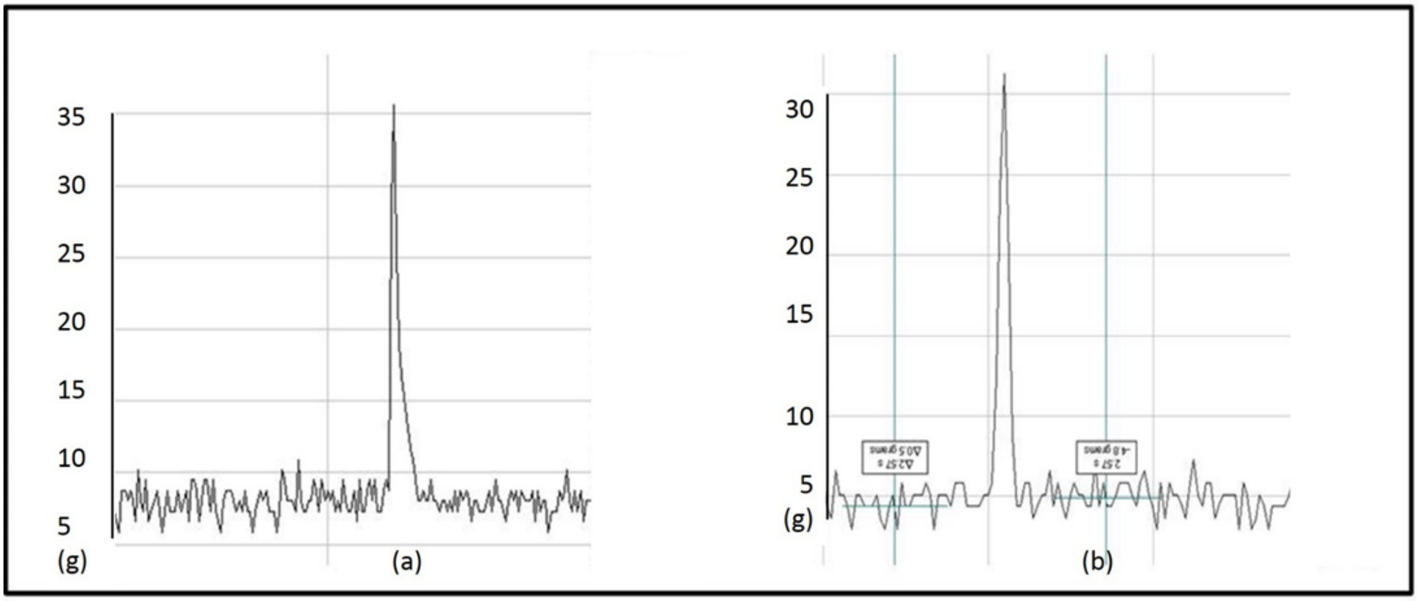

Figure 2. (a) Typical data using programmable esthesiometer. (b) Typical data using manual esthesiometer. The horizontal axis (not shown) is time. The vertical axis is grams. The vertical axis shows the raw data obtained from the experiments. The force is then calculated using F=mg, where m is the mass (kg) and $g = 9.8m/s^2$

threshold. The data obtained from the manual experiment shows between 50-60% standard deviation, while the data obtained from the automated device shows 10-25% deviation from the mean. This is at least two-fold decrease in error using the programmable esthesiometer.

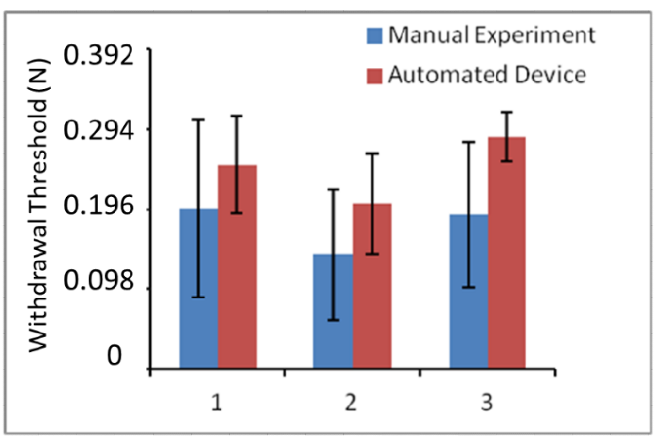

Figure 3. Comparison of withdrawal thresholds obtained from the manual experiment and the automated device ($p<0.1$).

While the actual thresholds obtained from the two experiments are not significantly different, and should not be, the automated device produced a tighter group of data when compared to the manual method. In all 3 subjects, the same trend is observed. The automated device shows a reduction in the percent variation from the mean compared to the manual experiment. The rats in the experiment were all normal control rats. There are usual variations between the rats themselves because of size. However, the purpose here is not to compare the pain withdrawal threshold but to compare the difference obtained in the threshold from the two described methods.

Conclusion

While the technique of using a force transducer is not new, the method we have described is new and permits an innovative way of collecting data. Each iteration of the experiment is guaranteed to be performed in exactly the same way each time. The rate of approach to the animal is kept constant regardless of the experimenter. The goal of this paper was not to compare the actual values obtained. Rather, the data presented illustrate that using a programmable esthesiometer operating in small increment steps (*pseudo-continuous)* results in a greater consistency in each experiment and yields data with lesser variation. Thus this new method can could ultimately lead to better models of behavioral response and a better understanding the factors contributing to these various ailments.

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The Introduced Dirt-Colored Seed Bug, *Megalonotus sabulicola* (Hemiptera: Rhyparochromidae): New for Arkansas

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Over the past decade, we have published several new records of Hemiptera from Arkansas in this journal (Chordas et al. 2005, 2011, Chordas and Kovarik 2008a, 2008b, Chordas and Kremers 2009). We now document the introduced dirt-colored seed bug, *Megalonotus sabulicola* (Thomson, 1870) for the first time from Arkansas.

On 20 April 2013, CTM and HWR collected various hemipterans at a watershed 8.0 km N of Huntsville off St. Hwy. 23W at Withrow Springs State Park (36.156195°N, 93.733369°W), Madison County (Fig. 1). Specimens were collected with a standard insect sweep net or an aquatic dip net and placed in individual vials of 70% ethanol. Bugs were forwarded to the senior author for identification, photography, and deposition of voucher specimens into the C.A. Triplehorn Insect Collection (The Ohio State University, Columbus, Ohio). Ashlock and Slater (1988), Maw et al. (2000), Scudder and Foottit (2006), and Wheeler (1992) were used as distribution references. Wheeler (1989, 1992) were used as taxonomic and identification references.

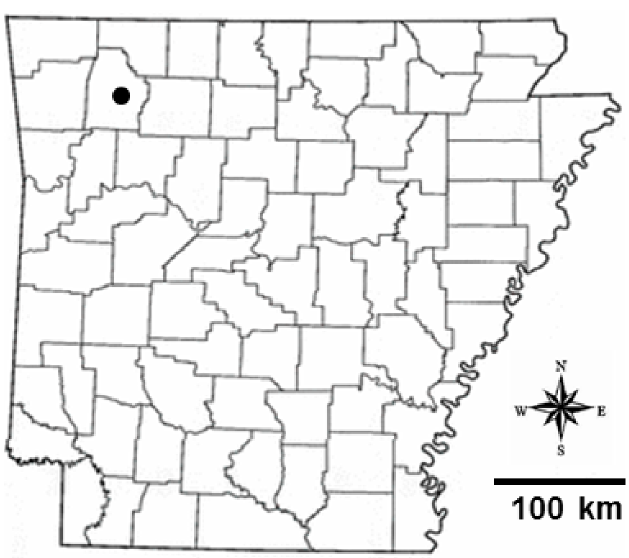

Figure 1. Location of study site (dot) in Madison County, Arkansas.

A single noteworthy specimen of *M. sabulicola* (Fig. 2) was collected in emergent vegetation. Our specimen matches the identifying characters and figure in Wheeler (1989). The bug possesses a mottled brown-testaceous hemelytra with a piceous head, pronotum and scutellum (Fig. 2A). It also has a prominent spine on the lateral, ventral aspect of the swollen profemur (Fig. 2B) and an antennal segment II almost all yellow with black at the distal edge (Fig. 2C). The tibia are yellowish with both the meso- and metafemurs are yellow proximally and piceous distally (Figs. 2A-B).

Figure 2. A. Dorsal habitus of *Megalonotus sabulicola*. Bar = 1 mm. B. Ventrolateral profemur spines (arrows). C. Antennae.

Megalonotus sabulicola was introduced from Europe in the early 1900's and has been reported to feed on seeds of knapweed, *Centaurea* spp. (Wheeler 1989). Documented mainly from both Atlantic and Pacific coastal regions of North America (see distribution herein), *M. sabulicola* was unexpected for Arkansas.

Tabulation of 21 records for *M. sabulicola* (Fig. 4) are as follows: USA (18 records): **Arkansas** [***New Record***], California, Connecticut, Delaware, Idaho, Massachusetts, Maryland, Michigan, New Jersey, New

York, North Dakota, Oregon, Pennsylvania, Rhode Island, Utah, Virginia, West Virginia, Washington. Canada (3 records): British Columbia, Ontario, Quebec.

Our Arkansas record represents a significant range extension of >1,000 km from the closest reported location (western Virginia) for this species. Previous records were primarily concentrated along the eastern and western portions of the U.S. and Canada with a few additional records in the Great Lakes region and a report from North Dakota (Fig. 3).

In summary, more data and collections are needed to determine the persistence of this bug in Arkansas. How and when it arrived in the state would be pure speculation at this point. For detailed biological information on *M. sabulicola*, see Wheeler (1989).

Figure 3. Distribution of *Megalonotus sabulicola* north of Mexico. Light shading (previous records); dark shading (new state record).

Acknowledgments

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New Records of Ectoparasites and Other Epifauna from *Scalopus aquaticus* and *Blarina carolinensis* in Arkansas

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Running Title: Ectoparasites of Soricomorpha in Arkansas

Compared to its surrounding states, little is known about the ectoparasites infesting the wild mammals of Arkansas (Schiefer and Lancaster 1970, Whitaker and Wilson 1974, Whitaker et al. 2007, McAllister et al. 2013). Recently, McAllister et al. (2013) suggested that additional ectoparasite surveys on mammals should be undertaken, particularly on insectivores. Here, we report information on some ectoparasites and other epifaunistic arthropods collected from the eastern mole, *Scalopus aquaticus*, and southern short-tailed shrew, *Blarina carolinensis*, from Arkansas. Ectoparasites have previously been reported from eastern moles by Whitaker and Schmelz (1974), Whitaker and Wilson (1974), Whitaker et al. (2007) and others and from southern short-tailed shrews by Whitaker et al. (1994), Nims et al. (2004, 2008), Whitaker et al. (2007) and Sylvester et al. (2012).

Twenty-four southern short-tailed shrews were collected using live traps between August 2012 and April 2013 from Union Co. and 3 eastern moles were collected using kill traps between May 2013 and June 2014 from Union (*n*=2) and Benton (*n*=1) cos. After being euthanized following American Society of Mammalogists guidelines (Sikes et al. 2011), individuals were examined for ectoparasites following standard methods (Gardner 1996). Ectoparasites and other arthropods were processed and identified using appropriate guides (Peck 1982, Whitaker 1982, Benton 1983, Lukoschus et al. 1988). Voucher specimens of hosts are deposited in the mammal collection at Henderson State University (HSU) in Arkadelphia, Arkansas. Representative ectoparasites are deposited in the General Ectoparasite Collection in the Department of Biology at Georgia Southern University, Statesboro (accession nos. L3564; L3569; L3584; L3587-L3589; L3680). We provide a taxonomic summary of the ectoparasites and other epifauna collected below.

Coleoptera: Leptinidae

Leptinus spp. beetles are epifaunistic arthropods of small mammals in the Holarctic region (Peck 1982). They are not true ectoparasites because they feed on dead host skin, sebaceous secretions and material in host mammal nests. In North America, *L. americanus* occurs west of the Mississippi River and east of the Rocky Mountains, whereas *L. orientamericanus* Peck occurs east of the Mississippi River and *L. occidentamericanus* Peck is found west of the Rocky Mountains (Peck 1982). *Leptinus americanus* has been reported previously from Washington County, Arkansas, from a tree stump (*n*=2) and from a mammal nest (*n*=28) (Peck 1982). We collected *L. americanus* from one of 24 southern short-tailed shrews and 2 of 3eastern moles. This species has previously been reported from *Scalopus aquaticus* (Whitaker and Schmeltz 1974), but *B. carolinensis* represents a newly reported host record. Whitaker et al. (1994) reported *L. americanus* from *B. carolinensis* from South Carolina; however, since 1982, beetles east of the Mississippi River have been treated as *L. orientamericanus* as detailed by Peck (1982).

Siphonaptera: Ctenophthalmidae

Ctenopthalmus pseudagyrtes is a common flea on shrews, moles, and other small mammals throughout much of North America although most records are from eastern States and Provinces (Hopkins and Rothschild 1956, Durden et al. 2012). We collected *C. pseudagyrtes* from both individuals of *S. aquaticus* (one male, one female flea) from Union Co. and from one of 24 individuals of *B. carolinensis* (ome male flea) from Union Co. This species has been previously collected from *S. aquaticus* from Arkansas (Schiefer and Lancaster 1970); *B. carolinensis* is a new host record. It has also been recorded previously from nearby Missouri and Texas (Kollars et al. 2007, McAllister and Wilson 2012).

One of 24 southern short-tailed shrews was infested with two *Doratopsylla blarinae* (one male, one female). Whitaker et al. (1994) reported this species from *B. carolinensis* in South Carolina but this collection represents a new state record for Arkansas. This flea appears to be widely distributed as an ectoparasite of *Blarina* spp., mostly in eastern North America (Hopkins and Rothschild 1956, Whitaker et al. 1994, Ritzi et al. 2005, Durden et al. 2012).

Acari: Laelapidae

Echinonyssus blarinae was collected from one of 24 southern short-tailed shrews. This mite has previously been reported from seven species of insectivores (including both *B. carolinensis* and *S. aquaticus*) and from at least 16 States and Provinces combined (Whitaker and Wilson 1974, Ritzi et al. 2005, Whitaker et al. 1994, 2007, Nims et al. 2008, Sylvester et al. 2012). However, this is the first record from Arkansas.

Haemogamasus harperi is a relatively large mite that is ectoparasitic mainly on shrews and moles (Whitaker and Wilson 1974, Whitaker et al. 2007). It has previously been reported from both *S. aquaticus* and *B. carolinensis* (Whitaker and Schmelz 1974, McAllister and Wilson 2012, Sylvester et al. 2012). One of two moles was infested with two *H. harperi* (both females) in this survey. This collection represents a new state record for this mite from Arkansas.

Acari: Listrophoridae

Olistrophorus blarina was collected from one of 24 southern short-tailed shrews. This tiny fur mite has previously been collected from *B. brevicauda*, *B. carolinensis* and *B. hylophaga* and has been reported from eight U.S. States (Whitaker and Wilson 1974, Whitaker et al. 1994, 2007, Ritzi et al. 2005, Nims et al. 2008). However, this represents the first record of this mite from Arkansas.

Acari: Glycyphagidae

One hypopial deutonymph of *Glycyphagus hypudaei* was collected from one of 24 *B. carolinensis.* This tiny fur mite has been collected from more than 70 species of mammals (including *B. carolinensis*), mainly rodents and insectivores, in North America with previous records from more than 25 States and Provinces combined (Whitaker and Wilson 1974, Whitaker et al. 1994, 2007, Nims et al. 2004, Ritzi et al. 2005). Nevertheless, this represents the first record of *G. hypudaei* from Arkansas.

Acari: Myobiidae

One of 24 southern short-tailed shrews was infested with 2 fur mites, *Protomyobia blarinae*. This small mite has previously been recorded from *B. brevicauda*, *B. carolinensis* and *B. hylophaga* from the States/Provinces of Georgia, Indiana, Kansas, Manitoba, New Brunswick, New York, Ontario and South Carolina (Whitaker et al. 1994, 2007, Ritzi et al. 2005, Nims et al. 2008). However, this is the first record of *P. americana* from Arkansas.

In conclusion, we record 2 species of fleas and 5 species of mites from 2 species of soricomorphs in Arkansas with all 7 of these arthropod species representing new state records. Whitaker and Wilson (1974) summarized 8 species of ectoparasitic/ epifaunistic mites from mammals in Arkansas and Whitaker et al. (2007) added a ninth species to the state list. Based on the examination of just 26 soricomorph specimens, we have increased the Arkansas list by 56%. Clearly, the ectoparasite fauna, particularly the mite fauna, of Arkansas mammals is inadequately documented. Therefore, we recommend additional ectoparasite surveys of Arkansas mammals.

Acknowledgments

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Natural History Notes and Records of Vertebrates from Arkansas

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Running Title: Natural History Notes and Records of Vertebrates from Arkansas

Although vertebrates are a commonly studied group of animals, the distribution and natural history of many species within Arkansas is still not well understood or documented. However, recently several new distribution and natural history notes have been published in a continuing series regarding Arkansas's vertebrates (e.g. Tumlison and Robison 2010, Connior et al. 2011, Connior et al. 2012, Connior et al. 2013). Thus, we continue to augment current literature with new records of distribution and provide notes on the natural history of selected vertebrates from Arkansas. All specimens (physical or photographic) are deposited in the vertebrate collections at Arkansas State University (ASUMZ), Henderson State University (HSU), or Auburn University (AUM; currently unaccessioned with DAN field numbers).

CLASS ACTINOPTERYGII

Noturus exilis Nelson – Slender Madtom. The slender madtom is widely distributed throughout the White and Arkansas River systems and inhabits small to medium- sized, permanent, spring-fed streams (Robison and Buchanan 1988). Robison and Buchanan (1988) reported collecting "ripe females" during late April and May. However, nothing is known on the size of the embryo clusters during nesting in Arkansas. On 16 June 2013, CTM and MBC collected two embryo clusters from Flint Creek, near Springtown, Benton Co. that had 86 embryos and 39 embryos with a wet weight on 1.78 g and 0.84 g, respectively. MBC also collected an individual embryo cluster that had 154 embryos with a wet weight of 3.12 g from Water Creek, near Mull, Searcy Co. on 14 June 2013. Vives (1987) reported a single egg cluster from the same Flint Creek in Oklahoma that contained 44 embryos. Other studies reported an average clutch size of 72 embryos with a maximum clutch size of 124 in 5 clusters from the North Fork White River in Missouri (Burr and Mayden 1984) and 51 embryos with a maximum clutch size of 74 from southern Illinois (Mayden and Burr 1981). It is interesting to note that we report the largest embryo count from four states.

Noturus gyrinus (Mitchill) – Tadpole Madtom. Five specimens of *N. gyrinus* were taken by DAN, HWR, and CTM on 24 October 2013 from Rolling Fork River at Johnson Bridge Rd. (Co. Rd. 12) W of Union, Sevier Co. (34.0647°N, 94.3801°W), representing only the second time this species has been reported from the Rolling Fork River system (Robison and Buchanan 1988).

Fundulus blairae Wiley and Hall - Western Starhead Topminnow. Two specimens of *F. blairae* were taken by DAN, HWR, and CTM on 24 October 2013 from Rolling Fork River at Johnson Bridge Rd. (Co. Rd. 12) W of Union, Sevier Co. (34.0647°N, 94.3801°W), representing the first documentation of this topminnow from the Rolling Fork River system (Robison and Buchanan 1988).

Fundulus chrysotus (Gunther) – Golden Topminnow. Robison and Buchanan (1988) noted that the golden topminnow was "strictly a lowland species inhabiting oxbow lakes, sluggish areas of creeks, and swampy backwater overflows of rivers", and their distribution map documented collections from the Mississippi Alluvial Plain and Gulf Coastal Plain in Arkansas. Updated records (McAllister et al. 2006) largely represented localities well within these same physiographic regions with the exception of specimens from the Arkansas River in Crawford Co., and a new record from Hot Spring Co. in the Ouachita River drainage. The Hot Spring Co. record extended the range along the Ouachita drainage only slightly from previous records in Clark Co., but to the edge of the Gulf Coastal Plain. This fish had been reported from the Saline River drainage only as far north as Bradley Co. (Robison and Buchanan, 1988) until a new record from Grant Co. extended the known range upstream (McAllister et al. 2010). We report a new county record from Hurricane Creek (34.61282°N; 92.52614°W) in Saline Co. near Bryant (HSU 3491) collected by M. Benson on 6 April 2013. This location is near the interface of the Gulf Coastal Plain and Ouachita Mountain provinces. Additionally, four specimens of *F. chrysotus* were collected on 24 October 2013 from a remnant oxbow off the Little River, at the first bridge S of the Little River on St. Hwy. 41, Little River Co. (33.9087°N, 94.3968°W) and represent the first record from the Little River system in Arkansas (Robison and Buchanan 1988).

Lepomis symmetricus Forbes – Bantam Sunfish. The Bantam Sunfish is a rare lowland species of the Coastal Plain in Arkansas (Robison and Buchanan, 1988). A single specimen of this species was collected on 24 October 2013 from a remnant oxbow off the Little River, at the first bridge S of the Little River on St. Hwy. 41, Little River Co. (33.9087°N, 94.3968°W). The water was tannin stained with no current over a mud substrate with abundant vegetation (*Myriophyllum*, *Utricularia*, *Nuphar*). These specimens represent the first record of this sunfish documented from the Little River system in Arkansas (Robison and Buchanan 1988).

Aphredoderus sayanus Gilliams – Pirate Perch. Five specimens of *A. sayanus* were collected by DAN, HWR, and CTM on 24 October 2013 from Rolling Fork River at Johnson Bridge Rd. (Co. Rd. 12) W of Union, Sevier Co. (34.0647°N, 94.3801°W) and represent the first record from the Rolling Fork River system (Robison and Buchanan 1988).

Esox niger LeSueur – Chain Pickerel. One juvenile specimen of *E. niger* was collected by DAN, HWR, and CTM on 24 October 2013 from Rolling Fork River at Johnson Bridge Rd. (Co. Rd. 12) W of Union, Sevier Co. (34.0647°N, 94.3801°W) and represents the first documented record of the Chain Pickerel from the Rolling Fork River system (Robison and Buchanan 1988).

Etheostoma spectabile pulchellum Distler– Plains Orangethroat Darter. The form of the Orangethroat Darter inhabiting the Little River system is known as *E. spectabile pulchellum* (Distler 1968, Robison and Buchanan 1988). Eleven specimens of *E. s. pulchellum* were collected by DAN, HWR, and CTM on 24 October 2013 from Rolling Fork River at Johnson Bridge Rd. (Co. Rd. 12) W of Union, Sevier Co. (34.0647°N, 94.3801°W) and although rather common in the Little River system, this collection is the first documented report of this species from the Rolling Fork River.

Percina phoxocephala Nelson – Slenderhead Darter. Robison and Buchanan (1988) showed only six records of *P. phoxocephala* for the entire state of Arkansas. Two specimens of *P. phoxocephala* were collected on 24 October 2013 from mainstem Little River at Little River Country Club at Billingsley's Corner, Little River Co. (33.9265°N, 94.4143°W). The collection of this darter species is particularly noteworthy as it is only the second report of *P. phoxocephala* from the Arkansas portion of the Little River system (Robison and Buchanan 1988) and the seventh in the entire state.

CLASS AMPHIBIA

Eurycea tynerensis Moore and Hughes – Oklahoma Salamander. *Eurycea tynerensis* is comprised of both the metamorphic and paedomorphic types (Bonett and Chippendale 2004). Life history mode is correlated with streambed habitat and microstructure (Tumlison and Cline 2003, Bonett and Chippendale 2006). Most populations exhibit a single life history mode with metamorphosing populations existing in poorly sorted clastic material and paedomorphic populations in streams containing large chert gravel (Bonett and Chippindale 2006). Herein, we report on a population occurring near Mull, Marion Co. that contained both life history modes. Ireland (1976) reported that larvae from NW Arkansas transform to adults at 38-42 mm snout-vent length (SVL). Of 17 individuals that were collected on 31 Dec 2013, 15 were paedomorphic (39.7 ± 6.1, range 23-47 mm SVL) with the remaining 2 being metamorphic. The 2 metamorphic individuals (41, 47 mm SVL) that were collected were found under large rocks at the periphery of the streambed. Interestingly, one of the paedomorphic individuals exhibited an unknown fungal infection (Fig. 1). Another population located at 3 km S Mull, near Water Creek, Searcy Co. contained mainly paedomorphic individuals; however, one (adult male SVL 43 mm) of eight individuals collected on 7 March 2014 exhibited the metamorphic body type.

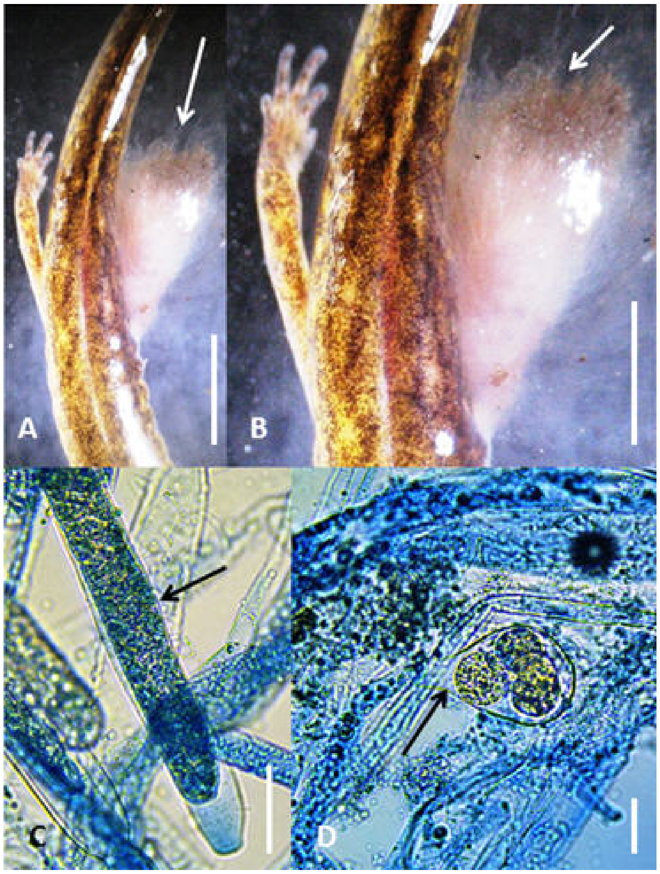

Figure 1. *Eurycea tynerensis* with unknown fungal infection. (A) Specimen showing infection (arrow) on left hind leg and foot; scale bar = 5mm. (B) Closer view of same specimen showing complete involvement of limb (arrow); scale bar = 2.5 mm. (C) Photomicrograph showing fungal filament (arrow); scale bar = 50 µm. (D) Photomicrograph showing oogonium (arrow); scale bar = 25 µm.

Ambystoma talpoideum (Holbrook) – Mole Salamander. On 9 Jan 2014, an adult female was collected crossing Jackson Street, 1 km N of Jct. between Jackson St. and US 167, 8 km S El Dorado, Union Co. This individual possessed an extra digit (6 total) on its hind limb (Fig. 2). Although polydactyly has been reported for other amybstomatids, including *A. tigrinum* and *A. macrodactylum* (Johnson et al. 2003), this is the first report to our knowledge of polydactyly in *A. talpoideum*.

Lithobates sylvaticus Le Conte– Wood Frog. On 7 March 2014, a large population of wood frogs was discovered in a cattle pond near Mull, Marion Co. This area is also a known locale for the Ozark highlands leech, *Macrobdella diplotertia.* An adult male wood frog (SVL 59 mm) was trapped in a funnel trap that also contained an adult *M. diplotertia.* Upon collection, two wounds were discovered (one on the abdomen and one on the dorsum) from where a leech had attached and consumed a bloodmeal. This is the first report of a *M. diplotertia* feeding on a wood frog; however, other ranid frogs including *L. catesbeianus*, *L. clamitans*, and *L. palustris* were reported as hosts at this same location (Connior and Trauth 2010).

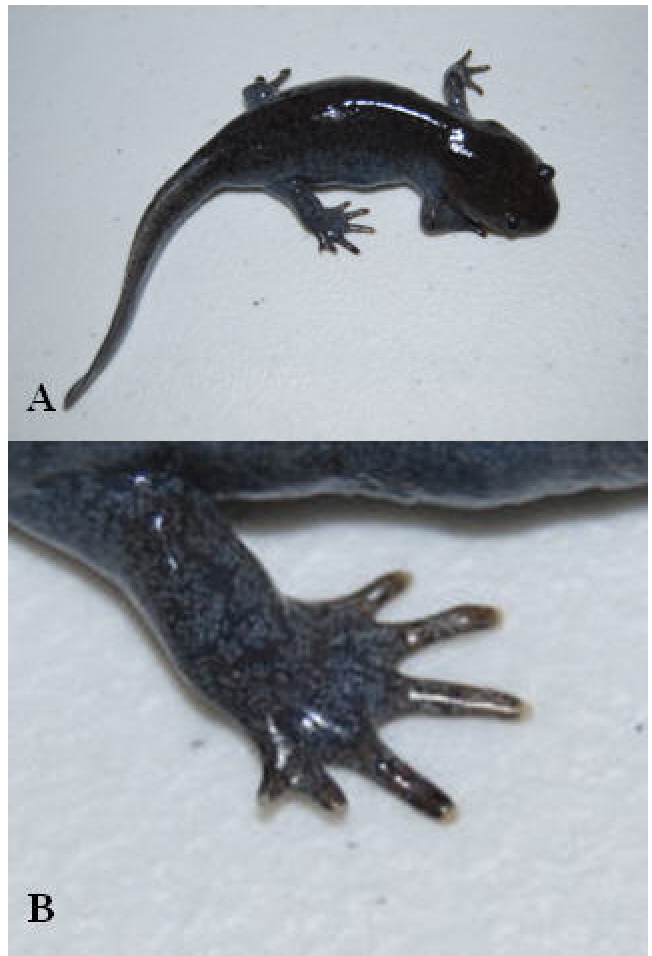

Figure 2. *Ambystoma talpoideum* exhibiting polycatctyly. A) Entire specimen B) close-up showing the extra digit on the hindlimb.

CLASS REPTILIA

Anolis carolinensis (Voigt) - Northern Green Anole. On 24 Feb 2013, MBC discovered an adult male (SVL 65 mm) from 2 km N of Junction City, Union Co., that was missing the lower half of the hind limb at the knee area (Fig. 3). It is unclear if this amputation was the result of a congenital defect or an injury, such as a failed predation attempt.

Figure 3. *Anolis carolinensis* from Union Co. Arkansas showing missing lower left hind limb.

CLASS MAMMALIA

Sorex longirostris Bachman - Southeastern Shrew. The southeastern shrew is collected only rarely in Arkansas, and has been documented from only a few counties in the interior Highlands region (Garland and Heidt 1989). On 7 May 2013, a southeastern shrew (HSU 703) was taken in a pitfall trap set to catch beetles in the Iron Springs Picnic Area, St. Hwy 7, N of Jessieville, Garland Co. Standard measurements are: total length 80 mm, tail length 33 mm, hind foot length 10 mm, ear length 5 mm. This specimen represents a new county record for Arkansas. The southern short-tailed shrew, *Blarina carolinensis*, was taken in the same pitfall.

Blarina brevicauda (Say)– Northern short-tailed shrew. The northern short-tailed shrew is known only from the Ozark Plateau and Boston Mountains in northern Arkansas (Pfau et al. 2011). On 6 March 2014, a single adult *B. brevicauda* was found dead in a small decorative pond 3 km S Mull, near Water Creek, Searcy Co. This is a new county record filling a distributional hiatus from adjacent Newton and Van Buren Cos. (Pfau et al. 2011).

Blarina carolinensis (Say) – Southern Short-tailed shrew. On 11 Feb 2013, a single female, containing 3 embryos, was collected from the vicinity of El Dorado, Union Co. This embryo count falls within the reported range for litter sizes of 2-6 (McCay 2001) and the breeding season of February to October reported by Sealander and Heidt (1990).

Cryptotis pava (Say) – Least Shrew. On 8 November 2013, a single adult female *Cryptotis parva* was collected from 3.2 km ESE Bruno, Marion Co. that contained 4 embryos. This is similar to the reported mean of 4.9 (Whitaker 1974) and falls within the reported breeding season for Arkansas of February to November (Sealander and Heidt 1990). This is a new county record filling a distributional hiatus between nearby Newton and Stone cos. (Pfau et al. 2011).

Three adult specimens were collected in a pitfall trap set (7 April - 23 May 2013) in a Baird's pocket gopher burrow system near Gillham in Sevier Co. This least shrew was a non-target species captured in a pitfall trap targeting pocket gopher insect inquilines, which was set in a burrow and completely sealed off from the surface. Other vertebrates (*i.e.*, reptiles [Connior et al. 2008, Connior and Chordas 2012] and small mammals [e.g. *Peromyscus maniculatus* Connior et al. 2011]) have been captured in pocket gopher burrows and mounds in Arkansas. Although Vaughan (1961) reported catching eastern moles, *Scalopus aquaticus* (Soricomorpha), this is the first documented record of a shrew (Soricomorpha) being captured inside a pocket gopher burrow.

Tamias striatus (Linnaeus)- Eastern Chipmunk. Searcy Co.: ~5 km W Harriet; State Hwy 27. DOR. 10 Nov 2013. 35.974857°N; 92.571149°W. Sealander and Heidt (1990) reported that chipmunks had been seen in Searcy Co, but this is the first museum record. Recently, Sasse (2003) reported chipmunks from adjacent Marion Co. to the north.

Castor canadensis Kuhl- Beaver. Citing Bradt (1939), Sealander and Heidt (1990) noted that beaver in Arkansas have offspring from April through June. That reference actually recorded observations from the northern United States, but more recent research demonstrated an earlier breeding season in the south (Baker and Hill 2003). The earliest estimated date of birth in Mississippi was mid-February (Wigley et al. 1983). We obtained 4 fetuses from a beaver trapped 23 February 2014 from Bayou Meto in northern Pulaski Co., which were well furred and near parturition (Fig. 4). Birth would have occurred in February or early March, thus this reproductive observation is the earliest reported for Arkansas.

Acknowledgments

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Commission issued Scientific Collecting Permits to CTM, MBC, HWR and RT.

Figure 4. Four fetuses from a beaver trapped 23 February 2014 from Bayou Meto in northern Pulaski County, AR.

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First Record of Ribbon Worms (Nemertea: Tetrastemmatidae: *Prostoma*) from Arkansas

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Running Title: Nemertea from Arkansas

Ribbon worms (Phylum Nemertea) are well known coelomate marine organisms; however, few biologists are aware of the presence of freshwater forms in North America (Gibson and Moore 1976, Sundberg and Gibson 2008). Members of this phylum are unique in having an eversible muscular proboscis that lies free inside of a rhynchocoel above the alimentary canal and is used for grasping prey (Hickman et al. 2011). Freshwater nemerteans are hermaphroditic and often protandric (Kolasa 1991). Ribbon worms have been previously reported from adjacent Louisiana (Harman 1962), Oklahoma (Harrell 1969) and Texas (Ziser 2011); however, to date, this phylum has not been documented from Arkansas despite numerous intensive aquatic macroinvertebrate surveys in the state by Cather and Harp (1975), Harp and Harp (1980), Farris and Harp (1982), Guntharp and Harp (1982), Huggins and Harp (1983), Cochran and Harp (1990), Chordas et al. (1996), and Harp and Robison (2006).

On 10 July 2011, 15 specimens of an unknown species of ribbon worm were collected from the Ouachita River at Little Hope Road, 0.4 km S of St. Hwy. 88 in Pine Ridge, Montgomery County (34.581364°N, 93.883678°W) (Fig. 1). Ribbon worms were collected in the main river from a shallow riffle where submerged plants of hornleaf riverweed, *Podostemum ceratophyllum* Michx., occurred abundantly. At the collecting site the water was 25 to 38 cm deep, the water temperature was 23.5°C, and the air temperature was 34°C. At this locale, the Ouachita River is characterized physicochemically by water temperatures ranging from near 0°C in winter months to 25°C in summer, dissolved oxygen of 9.5-11.4 mg/l, pH 6.9-7.1, alkalinity (total) 25.2- 36.8 mg/l, chloride 11.2-26.0 mg/l, total dissolved solids 14-53, turbidity (NTU) 2.0-4.2, hardness, Ca++, Mg++ of 28.2-36.9 mg/l, sulfates 3.7-56 mg/l, total phosphorus 0.029-2.033, and total suspended solids 3.0-5.1 (J Wise, *pers. comm*). While these data are not intended to be indicative of physicochemical limits of this nemertean worm, they are suggestive of the general type of water quality found at this upland locality.

Figure 1. The Ouachita River study site where *Podostemum ceratophyllum* (submerged and not visible) occurred abundantly.

In our search for Arkansas ribbon worms we purposefully sought out *Podostemum* vegetation as this had been shown to be a reliable microhabitat for collecting nemerteans. The senior author (PGD) had previously collected nemerteans from the sediment attached to *Podostemum* in western North Carolina (Chattooga River) and northwestern Alabama (Cypress Creek, Lauderdale County and Gin Creek, Marion County). *Podostemum* has long been known as an important habitat for macroinvertebrates (Hutchens et al. 2004, Nelson and Scott 1962) but we know of no previous published reports linking ribbon worms to *Podostemum*. At the Ouachita River site, *Podostemum* was removed by hand from its attachment to rock in the flowing stream. Care was taken to acquire the sediment bound by *Podostemum* at the rock surface. Samples were placed in plastic bags, stored in a cooler, and processed in a motel room within several hours of collecting. Processing followed the procedure known as the oxygen depletion method (Schockaert 1996) described in some detail by Young (2001). Materials collected from the Ouachita River were placed in the bottom halves of six clear glass jars ranging in size from 0.96 to 7.6 l (1 qt to 2 gal). Stone weights (rocks of golf ball size and larger) were added to hold the vegetation in the lower half of the jars and the jars were then filled with habitat water. The stone weights prevent vegetation from rising and obscuring one's view. Within 5 hrs, 4 stagnant jars yielded a total of 10 nemerteans made visible with the aid of a strong light aimed through the backs and sides of the jars. The worms clung to the inner glass walls as they glided towards the water's surface. By the next morning, a total of 15 pinkish ribbon worms measuring 4 to 8 mm in length were collected by pipet and preserved in 70% v/v ethanol.

Figure 2. Ribbon worm collected from *Podostemum*. Scale bar = 1.5 mm.

Our collection of ribbon worms represents the first documentation of the Phylum Nemertea in Arkansas. Unfortunately, we were unable to determine the exact identity of ribbon worm (Fig. 2). Eight specimens were sent to C. Laumer for identification using DNA sequencing. Mr. Laumer reported (*pers. comm.*) that preliminary DNA analysis of the haplotypes from the Arkansas *Prostoma* specimens were identical to a particular haplotype seen elsewhere in the USA and Australia. He suggested that we use the name currently being listed in GenBank as *Prostoma* cf. *eilhardi* for the form we report herein.

Acknowledgments

We thank B. Crump, USDA Forest Service biologist, Ouachita National Forest, for her assistance in our quest to find *Podostemum* localities and ribbon worms in the Ouachita Mountains. Her professionalism, wide knowledge of the area, and enthusiasm for the project aided our effort immensely. In addition, we also thank G. Leeds, retired USDA Forest Service biologist, Ozark National Forest (ONF), L. Leeds, retired USDA Forest Service engineer (ONF), and J. Kremers, Clarksville, for assisting us in the Ozark Mountains. These knowledgeable individuals kindly showed us localities in the Ozarks, chauffeured us to the various sites, and ably assisted us in our collecting efforts. In addition, L. and S. Leeds graciously provided us food, shelter, and use of their home as our laboratory while in their company. Appreciation is also expressed to J. Wise (ADEQ) for supplying water quality data for the Ouachita River. Lastly, we wish to acknowledge two ribbon worm experts, C. Laumer (Harvard University), who conducted the DNA analyses and provided a name to use for this manuscript, and N. Van Steenkiste (Hasselt University, Belgium), who showed PGD his first freshwater nemertean and how to use the oxygen depletion method.

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New Host and Location Record for the Bat Bug *Cimex adjunctus* Barber 1939, with a Summary of Previous Records

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Running Title: New Host and Location Record for the Bat Bug Cimex adjunctus Barber 1939

Abstract

In June 2009, 14 Rafinesque's big-eared bats (*Corynorhinus rafinesquii*) were collected from an abandoned house near Shepherd, San Jacinto County, Texas. Three individuals harbored bat bugs which were subsequently identified as *Cimex adjunctus* Barber 1939. This is the first record of this species from *C. rafinesquii.* In August 2013, 10 *C. rafinesquii* were collected from a maternity colony in Drew County in southeastern Arkansas. Four of the bats harbored bat bugs, which were identified as *C. adjunctus*. This is the first record of this bat bug from Arkansas. A summary of previous state and host records of the insect is provided, as is a summary of parasite records from *C. rafinesquii.*

Results and Discussion

Rafinesque's big eared bat (*Corynorhinus rafinesquii*) roosts in abandoned buildings, caves, hollow trees, and under bridges across the southeastern United States (Jones 1977, Trousdale and Beckett 2004). This bat commonly forms colonies ranging in number from a few individuals to 100 or more.

On 28 June 2009, 14 *C. rafinesquii* were hand-collected from an abandoned house near Shepherd, San Jacinto County, Texas. A maternity colony of *C. rafinesquii* numbering >50 individuals occupied this roost each year from 2005-2009 and in 2012 (surveys were not conducted in other years). Three *Cimex adjunctus* Barber 1939 were found on the torso and uropatagium of 3 lactating adult female bats. No external parasites were noted on 8 additional adult females or 3 juvenile males also captured at the same location and time. Bat bugs were preserved in ethanol and deposited in the Gibson Entomarium in the Biology Department at Stephen F. Austin State University. This is the first record of *C. adjunctus* from Rafinesque's big-eared bat.

A maternity colony of over 100 individuals of *C. rafinesquii* roosts in the Taylor House, an abandoned antebellum building located at the edge of an agricultural field adjacent to Bayou Bartholomew in Drew County, Arkansas. On August 2, 2013, 10 individuals were captured with a hand net. All of the captured bats were females; 3 were lactating. Four of the bats harbored bat bugs, which were collected and preserved in 95% ethanol. All bats were released unharmed at the point of capture. The preserved bat bugs were sent to the Department of Entomology and Plant Pathology at Auburn University, Alabama, where they were identified as *Cimex adjunctus*. Specimens were deposited in the insect collection of the Museum of Natural History at Auburn. This represents the first record of *C. adjunctus* from Arkansas.

Cimex adjunctus is found over most of the eastern United States and southeastern Canada and has been recorded as far west as Colorado (Usinger 1966). It has previously been reported from *Eptesicus fuscus* in Illinois, Indiana (Webster and Whitaker 2005), Missouri (Bowles et al. 2013), Kansas (Sparks et al. 2003), Michigan (Dood and Kurta 1982), Alabama (Durden et al. 1992), Kentucky, Colorado, Georgia, and Florida (Usinger 1966), from *Lasionycteris noctivagans* in Nebraska (Usinger 1966) and South Dakota (Swier 2003), and from *Nycticeus humeralis* in Kansas (Sparks et al. 2003), Missouri (Bowles et al. 2013), Indiana, Kentucky, North Carolina, South Carolina, West Virginia, Alabama, Florida, and Texas (Usinger 1966). This bat bug is known from *Myotis californicus* in Colorado (Usinger 1966), from *Myotis lucifigus* in Missouri (Palmer and Gunier 1975), Michigan (Dood and Kurta 1988), Pennsylvania (Dick et al. 2003), Tennessee (Reeves et al. 2007), West Virginia (Wilson 1943), Colorado, Indiana, Virginia, Vermont (Usinger 1966), and Nova Scotia (Poissant et al. 2010), from *Myotis septentrionalis* in Indiana (Ritzi and Whitaker 2003), New Hampshire (Sasse and Pekins 2000), and Nova Scotia (Poissant and Broders 2008), from *Myotis sodalis* in Indiana (Usinger 1966) and Michigan (Dood and Kurta 1982), from *Myotis thysanodes* in South Dakota (Turner and Knox Jones 1968), and from roosts of *Myotis austroriparius* in South Carolina (Reeves 2001). *Cimex adjunctus* was collected from *Tadarida brasiliensis* and its roosts in Georgia (Spears et al. 1999). It has also been recorded from unknown hosts in Ohio, Iowa, Maine, Rhode Island, Delaware, New Jersey, Maryland, New Hampshire, New York (Barber 1939, Usinger 1966), Manitoba, Ontario, Quebec, and Newfoundland (Maw et al. 2000), and from Pennsylvania, where the type specimens were collected (Barber 1939).

Although *C. rafinesquii* is widespread, there are relatively few studies of its behavior and ecology, and few parasites have been recorded. The bat bug *Cimex pilosellus* has been reported from *C. rafinesquii* in Arkansas (Steward et al. 1986). Mites recorded from Rafinesque's big-eared bats include *Chiroptoglyphus*, *Macronyssus,* and *Teinocoptes* (Whitaker et al. 2007). Reported endoparasites include tapeworms (*Vampirolepis* sp.) and two species of nematodes (*Physaloptera* sp. and *Capillaria palmata*—McAllister et al. 2005).

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Fecundity of Arkansas Tarantulas *Aphonopelma hentzi* (Girard)

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Running Title: *A. hentzi* Reproduction in Arkansas

Aphonopelma hentzi (Girard) is a species of theraphosid spider found throughout arid regions of the eastern expanse of the southwestern United States. Its range has been stated as occurring from the Mississippi river west into New Mexico, north to Colorado and Missouri and south into southern Texas and possibly Mexico (Hamilton et al. 2011). There has been some uncertainty surrounding the taxonomy of *Aphonopelma* species in Arkansas. Morphological data, county records, and public survey data was compiled by Warriner (2008) who concluded that *A. hentzi* was the sole species of theraphosid present in Arkansas. Using mitochondrial DNA markers, Hamilton et al. (2011) suggested that no other species exists north of the Colorado River Basin in Texas. Populations of this spider occurring in Arkansas are primarily found in the upland portions of the state (Warriner 2008) where their occurrence has been strongly correlated to xeric habitats with well drained soils such as cedar glades and disturbed areas (Baerg 1958). *Aphonopelma hentzi* was the first species of tarantula described in the US (Girard 1854) and is the largest spider within the state. There is a distinct lack of published information concerning the ecology of this species in Arkansas and none could be found specifically concerning populations endemic to the Ouachita Mountains physiographic region. The bulk of information to be found comes from studies conducted over several decades by the famous William J. Baerg at a field site in the Ozark Mountains near the University of Arkansas, Fayetteville (Baerg 1958).

The main purpose of this research was to increase the body of knowledge surrounding *A. hentzi* populations occurring within Arkansas with special interest placed on reproductive data.

Egg sacs were collected between June and August from 3 sites over a period of 5 years (2009-2013). Sites were located in both the Ozarks and Ouachita Mountains physiographic regions. Site 1 consisted of Ozark glade habitat along the western border of Lake Leatherwood in Carroll County (Table 1). Site 2 was an open canopied disturbed roadside ditch amongst recently burned pine in the Ouachita National Forest approximately 3 km west of HWY 71 on Buffalo Road, Scott County (Table 1). Site 3 was a power line right-of-way in the Ouachita National Forest approximately 2 km west of HWY 71 on Poteau Mountain Road, Scott County (Table 1). Once located, females were coaxed away from their burrow entrances and egg sacs collected by hand. The sacs were placed into cotton pillow cases and then into a cooler for transport. The females were then photographed (Figure 1) and their overall carapace length measured in mm with a flexible ruler from Carolina Biological Supply Company (Table 1). Collected egg sacs were opened with scissors (Figure 2) under a dissecting scope and numbers of first instar spiderlings/eggs were recorded. After counting, the first instar *A. hentzi* were either placed in 95% v/v ethanol or were observed for several weeks and released.

Table 1: Date and location of collection, carapace length of adult female (CLAF), offspring number, and body length of first instars (LFI).

Site	ID	Date Collected	Coordinates	CLAF (mm)	Offspring #	LFI (mm)
1	A	8 Aug 2009	36°26’18.15” N 93°45’32.34” W	19.0	268	4.5
1	B	5 July 2010	36°26’16.41” N 93°45’37.78” W	19.0	743	4.0
1	C	31 July 2010	36°26’47.53” N 93°45’8.12” W	21.0	278	4.5
2	A	16 June 2012	34°58'10.48" N 94° 8’58.67" W	22.0	813*	n/a
3	A	17 June 2012	34°58’37.94” N 94°6’46.48” W	21.0	694	3.5
3	B	27 July 2013	34°58’37.94” N 94°6’46.48”W	19.0	467	4.5
3	C	28 July 2013	34°58’26.69” N 94°6’42.17”W	24.0	780	5.0

*unhatched eggs


Figure 1. Adult female at Site 2 with egg sac 16 June, 2012.


Figure 2. First instars counted and returned to opened egg sac.

Three egg sacs were collected from Sites 1 and 3 and one from Site 2. Carapace length of the adult females (CLAF) from which egg sacs were collected ranged from 19.0 mm to 24.0 mm with a mean of 20.7mm (Table 1). First instar number per egg sac ranged from 268-780 with a mean of 538.3. The body length of first instar spiderlings (LFI) ranged from 3.5-5.0 mm with a mean of 4.3 mm. One egg sac contained 813 eggs that were still yet to hatch.

The data collected over 5 seasons showed that on average females observed were slightly larger than those from previous studies. The largest female of this study was encountered from Site 3 and had a carapace length more than 4mm longer than any specimen described from the state by Warriner (2008) and also outsized a "well-fed" captive female described by Baerg (1958) at 23.3 mm. Female size however did not appear to correlate to offspring number. The number of offspring per egg sac was highly variable and had a range of 268-813 which fell within the range noted by Baerg (1958). *A. hentzi* egg sacs collected from the Ouachita Mountains physiographic region of Arkansas had a higher mean number of offspring (688.5) than specimens from the Ozarks (429.7) (Table 1).

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Haemogregarina sp. (Apicomplexa: Haemogregarinidae), *Telorchis attenuata* (Digenea: Telorchiidae) and *Neoechinorhynchus emydis* (Acanthocephala: Neoechinorhynchidae) from Map Turtles (*Graptemys* spp.), in Northcentral Arkansas

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Running Title: Haemogregarine, Trematode and Acanthocephalan Records

Little is known about the hematozoa and helminths of turtles of Arkansas. McAllister and King (1980) and McAllister et al. (1995) reported haemogregarines from the red-eared slider, *Trachemys scripta elegans* and alligator snapping turtle, *Macrochelys temminckii*, respectively. Fragmentary information is available on acanthocephalans (Ward and Hopkins 1931, Cable and Fisher 1957, Fisher 1960) and a nematode (McAllister et al. 1995). However, the only complete endoparasite survey to date on turtles of the state is that of Rosen and Marquardt (1976) on *T. s. elegans*. It is obvious that more turtles need to be surveyed for hemoparasites and helminths. Here we report new geographic and host records for a haemogregarine, a digene trematode and an acanthocephalan in map turtles, *Graptemys* spp. from the state.

On 25 May 2013, a juvenile Ouachita map turtle, *Graptemys ouachitensis* (carapace length [cl] = 57 mm, ASUMZ 33041) was collected by dipnet from the Lakeview Boat Dock, Baxter County (36.370576°N, 92.554544°W). On 25 July 2013, an adult male common map turtle, *Graptemys geographica* (cl = 125 mm, ASUMZ 33042) was collected by hand from Crooked Creek, Marion County (36.245225°N, 92.715755°W). Both turtles were killed with an intraperitoneal injection of concentrated Chloretone and their plastrons were removed with a bone saw to expose visceral contents. Thin smears were made of blood samples taken from the heart, fixed in absolute methanol, stained with Wright's stain, rinsed in neutral buffer and examined by light microscopy for hematozoa. The entire gastrointestinal tract from the cloaca to esophagus and urinary bladder was removed, washed in 0.6% w/v saline, split longitudinally, and examined for helminths under a stereomicroscope. Trematodes were stained with acetocarmine and mounted in Canada balsam. Acanthocephalans were placed on slides with a drop of glycerol and studied as temporary mounts. Voucher specimens of hosts are deposited in the Arkansas State University Museum of Zoology (ASUMZ) Herpetological Collection, State University. Voucher specimens of parasites were deposited in the United States National Parasite Collection, Beltsville, Maryland. Scientific and common names of turtles follow the TIGR Reptile Database (Uetz and Hošek 2013).

A digene trematode was found in the *G. ouachitensis* while a haemogregarine and an acanthocephalan were recovered from the *G. geographica*. Data is presented below in annotated format.

Apicomplexa: Adeleorina: Haemogregarinidae

Haemogregarina sp. Danilewsky, 1885 – About 2% of the red blood cells of *G. geographica* contained an intraerythrocytic hematozoan thought to belong to the genus *Haemogregarina* (USNPC 107976). Banana-shaped immature gamonts were most often observed (Fig. 1). They were very similar to the "type IV" morphological type reported from Lonoke County *T. s. elegans* by McAllister and King (1980). McAllister et al. (1995, Fig. 3) also reported large immature gamonts from *M. temminckii* from Ouachita County similar of those from *G. geographica*. In addition, Acholonu (1974) reported *Haemogregarina pseudemydis* in Mississippi map turtle, *Graptemys pseudogeographic kohnii* (syn. *Graptemys kohnii*) from Louisiana. Haemogregarines are most commonly reported from aquatic turtles with leeches serving as the only known invertebrate hosts and vectors (Telford 2009). We document a new host record for a haemogregarine in *G. geographica*.

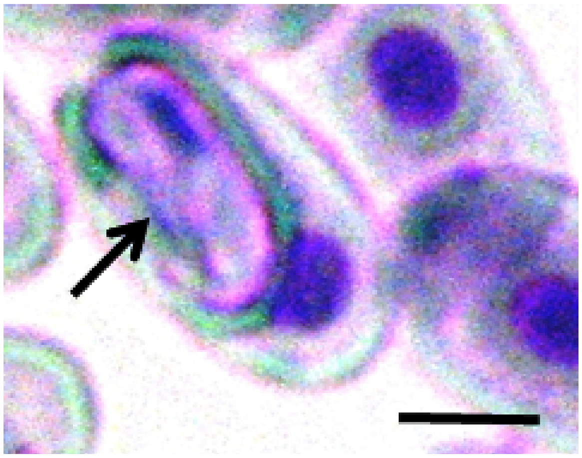

Figure 1. Gamont (arrow) of hematozoan from *Graptemys geographica*. Scale bar = 10 µm.

Trematoda: Digenea: Plagiorchiida: Telorchiidae

Telorchis attenuata Goldberger, 1911 – Numerous (> 100) digene specimens fitting the description of *T. attenuata* (Fig. 2, USNPC 107963) and confirmed using the key to North American species of *Telorchis* provided by MacDonald and Brooks (1989) were found in the intestine of *G. ouachitensis*. This trematode was previously reported in common snapping turtles, *Chelydra serpentina* from Ohio (Rausch 1947) and painted turtles, *Chrysemys picta* from Indiana (Goldberger 1911), Iowa, Maryland (MacDonald and Brooks 1989), Michigan (Esch and Gibbons 1967), Nebraska (Brooks and Mayes 1975), Ohio (Rausch 1947, Platt 1977), Wisconsin (Guilford1959) and British Columbia, Canada (MacDonald and Brooks 1989), and *T. scripta* from Mexico (Moravec and Vargas-Vásquez. 1998) and Spain (Cardells et al. 2013). Previously in the state, Rosen and Manis (1976) reported *Telorchis stunkardi* Chandler, 1923 from the three-toed amphiuma, *Amphiuma tridactylium* and Rosen and Marquardt (1978) reported *Telorchis corti* Stunkard, 1915 and *Telorchis singularis* (Bennett, 1935) Wharton, 1940 from *T. scripta* from Lake Conway (see MacDonald and Brooks 1989). Brooks and Mayes (1976) previously reported *Telorchis chelopi* MacCallum, 1919 (syn. *Telorchis gutturosi* Brooks and Mayes, 1976) from false map turtle, *Graptemys pseudogeographica pseudogeographica* from Nebraska. We document a new host and new geographic record for *T. attenuata*.

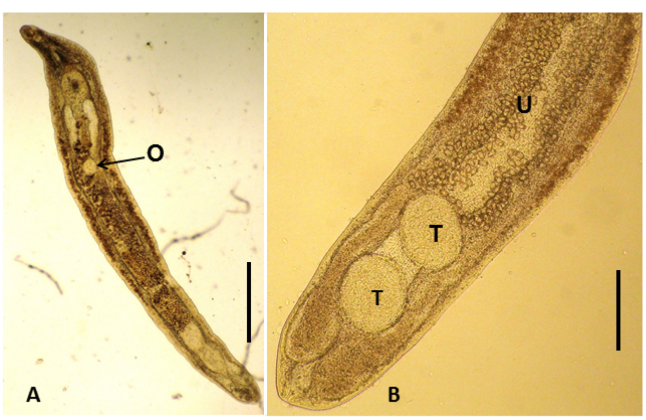

Figure 2. *Telorchis attenuata* (unstained) from *Graptemys geographica*. (A) Entire worm showing ovary (O); scale bar = 100 µm. (B) Lower half of worm showing tandem testes (T) and uterus (U) with ova; scale bar = 25 µm.

Acanthocephala: Eoacanthocephala: Neoechinorhychida: Neoechinorhynchidae

Neoechinorhynchus emydis (Leidy, 1851) Van Cleave, 1916 – Of the acanthocephalans we examined from the intestinal tract of *G. geographica* that included immatures and both sexes, every gravid female (USNPC 107211) represented *N. emydis* (Fig. 3), confirmed by the anatomy of the eggs and posterior ends (Barger and Nickol 2004). There were more than 200 individual worms in this host (Fig. 3A). Previous hosts of *N. emydis* include *G. geographica*, G. pseudogeographica, Texas map turtle, Graptemys

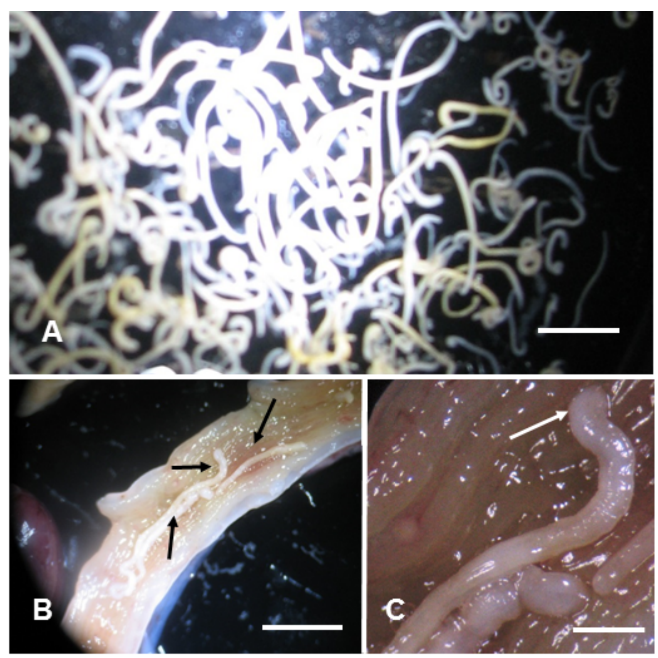

Figure 3. *Neoechinorhynchus emydis*. A. Gross view of acanthocephalans removed from intestinal tract showing intensity of infection. Scale bar = 1 mm. B. In situ view showing three worms in intestine (arrows). Scale bar = 10 mm. C. Closer view of worm with proboscis embedded in intestinal mucosa (arrow). Scale bar = 2 mm.

versa, *C. serpentina*, river cooter, *Pseudemys concinna*, *T. scripta*, spotted turtle, *Clemmys guttata*, wood turtle, *Glyptemys insulpta*, and Blanding's turtle, *Emydoidea blandingii* (Hopp 1954, Ernst and Ernst 1977, Barger 2004). This acanthocephalan has been reported most often from the eastern half of the upper Mississippi River drainage, including Illinois, Indiana, Mississippi, Ohio, Oklahoma and Texas (Williams 1953, Everhart 1958, Barger 2004), and now Arkansas. In addition, Rosen and Marquardt (1978) reported four species of *Neoechinorhynchus* (but not *N. emydis*) from *T. s. elegans* from the state. Thus, we document a new distributional record for *N. emydis* in the Arkansas.

Turtles are hosts of numerous described and undescribed hematozoans and helminths (Ernst and Ernst 1977, 1979, Telford 2009). Because Arkansas supports 19 species and subspecies of turtles within four families (Trauth et al. 2004), we suggest additional surveys on larger samples of turtles from the state as several species remain to be examined for hematozoans and endoparasites. The inclusion of DNA sequence analysis would be particularly helpful to identify some parasite species which have limited morphological traits (i.e., haemogregarines). As such, we predict additional new host and distributional records, including the possibility of discovery of new species.

We thank P.R. Pilitt (USNPC) and Dr. S.E. Trauth (ASUMZ) for expert curatorial assistance. The Arkansas Game & Fish Commission provided Scientific Collecting Permits to CTM and MBC.

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New Host Records for *Mesocestoides* sp. Tetrathyridia (Cestoidea: Cyclophyllidea) in Anurans (Bufonidae, Ranidae) from Arkansas, with a Summary of North American Amphibian Hosts

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Running Title: New Host Records for *Mesocestoides*

Adult *Mesocestoides* (Cestoidea) are cosmopolitan parasites of placental mammals and birds and rarely humans (Padgett et al. 2012). The tetrathyridial stage is often found in the body cavity and encapsulated in tissues of vertebrate second intermediate hosts such as amphibians, reptiles, and rodents. Although there have been numerous attempts to transmit tetrathyridia to definitive hosts, none have been successful. Indeed, this is one of the few tapeworms for which the life cycle has yet to be conclusively demonstrated. In addition, there are no morphological characters that allow assignment of tetrathyridia to a given species. Over the last three decades in Arkansas, *Mesocestoides* sp. tetrathyridia have been previously reported by McAllister and associates from numerous amphibians and reptiles (see McAllister et al. 2014). The purpose of this report is to document three new host records for *Mesocestoides* sp. in common anurans of the state. We also provide a summation of the North American amphibians known to harbor *Mesocestoides*.

Between September 2013 and February 2014, 99 frogs and toads including 29 dwarf American toads, *Anaxyrus americanus charlesmithi*, 6 Fowler's toads, *Anaxyrus fowleri* and 64 southern leopard frogs, *Lithobates sphenocephalus utricularius* were collected by hand from various sites in Union County. Specimens were euthanized with a concentrated Chloretone solution and a mid–ventral incision was made and the viscera and body cavity examined for *Mesocestoides* sp. When suspected encapsulated tapeworms were observed, they were excised with a bit of tissue and preserved in 10% v/v neutral buffered formalin. Free cestodes were collected and preserved in 70% v/v ethanol, stained with Semichon's acetocarmine and mounted in Kleermount®. For light microscopy of plastic–embedded tissues, we followed the methods of Bozzola and Russell (1999). Following fixation, tissues were dehydrated in a graded series of increasing ethanol solutions (70–100% v/v), placed in a 50/50 % v/v acetone/plastic mixture for overnight infiltration, and then embedded in Mollenhauer's Epon-Araldite #2 (Dawes 1988). For thick sectioning (approximately 1 µm in thickness) and staining, we used glass knives on an LKB Ultrotome (Type 4801A) with Ladd® multiple stain (LMS), respectively. For photomicroscopy, we used a Nikon Eclipse 600 epifluorescent light microscope with a Nikon DXM 1200C digital camera (Nikon Instruments Inc, Melville, NY). Voucher specimens of hosts were deposited in the Arkansas State University Herpetological Museum (ASUMZ), State University, Arkansas, U.S.A. or the Henderson State University (HSU) Collection, Arkadelphia, Arkansas. Voucher specimens of *Mesocestoides* sp. were deposited in the United States National Parasite Collection (USNPC 107667-107668), Beltsville, Maryland.

Four of 29 (14%) *A. a. charlesmithi* (54-70 mm SVL males and females) collected from Calion Lake (*n* = 3) and Grady Bell Road (*n* = 1) were found to harbor free tetrathyridia of *Mesocestoides* in their body cavity (Fig. 1A). One of 6 (17%) *A. fowleri* (62 mm SVL female) collected from 9.5 km S of El Dorado and 1.5 km N of jct. of Jackson Street and US 167 had tetrathyridia in its liver (Fig. 2). In addition, 6 of 64 (9%) *L. s. utricularius* (54 mm SVL female, 5 males 52-67 mm SVL) from Calion Lake (*n* = 1) Grady Bell Road (*n* = 1), 19th Street (*n* = 1), and 19th Street/Champagnolle Road (*n* = 3) had tetrathyridia in their mesenteries (Fig. 1B). All tetrathyridia from all hosts possessed characteristic individual features of a single invaginated scolex, a generally deep invagination canal (Fig. 1A), a prominent single excretory pore at the end opposite the scolex, and a solid hindbody. No tetrathyridium possessed a divided scolex, somatic bud, or any tegumental or excretory anomalies such as those reported rarely from tetrathyridia in some aberrant acephalic tetrathyridia from other host species (see recent review by Conn et al. 2011).

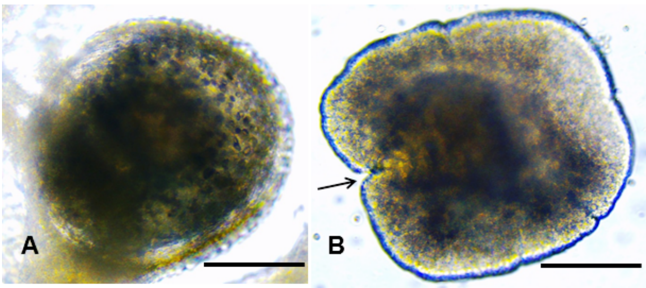

Figure 1. Tetrathyridia (unstained whole mounts) from anurans. A. Free tetrathyridium from *Anaxyrus americanus charlesmithi*. Bar = 250 µm. B. Free tetrathyridium from *Lithobates sphenocephalus utricularius* showing invagination canal (arrow). Bar = 250 µm.

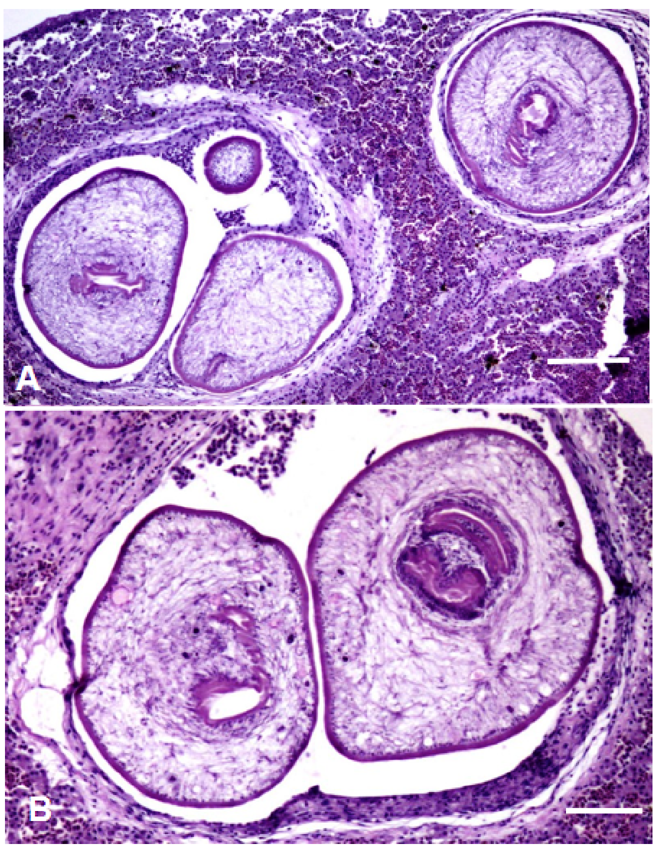

Figure 2. *Mesocestoides* sp. tetrathyridia from liver of *Anayxrus fowleri*. A. Four tetrathyridia encapsulated in tissue; scale bar = 50 µm. B. Closer view of two tetrathyridia in encapsulation; scale bar = 25 µm.

A summary of North American anuran hosts of *Mesocestoides* sp. was provided nearly 25 years ago by McAllister and Conn (1990) and we update that list herein and add caudate hosts to it (Table 1). Interestingly this tapeworm remains to be reported from a number of U.S. states, and as far as we can determine, it has only been reported to date from amphibians from 11 (22%), states including Arkansas, California, Iowa, Kansas, Michigan, Nebraska, New York, Oklahoma, South Dakota, Texas, and Wisconsin (Table 1). In all, *Mesocestoides* sp. has been reported from six species of salamanders of the world and 18 species/subspecies of frogs and toads from North America. There are numerous states east of the Mississippi River that support a great diversity of amphibians (i.e., Kentucky, North Carolina, Tennessee) and we predict that examination of several of those species could result in new geographic distribution records in those states and perhaps new host records for *Mesocestoides* sp.

In conclusion, we document 3 new host records for *Mesocestoides* sp. in anurans from Arkansas. This parasite has now been reported from nine of 59 (15%) amphibians of the state (Trauth et al. 2004).

We thank P.R. Pilitt (USNPC) and Dr. R. Tumlison (HSU) for expert curatorial assistance. The Arkansas Game & Fish Commission provided Scientific Collecting Permits to CTM and MBC.

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Table 1. Summary of reports of *Mesocestoides* sp. in North American amphibians.

Host	State	Prevalence*	Reference
Caudata			
Ambystomatidae			
Ambystoma maculatum	Arkansas	1/25 (4%)	McAllister et al. (2014)
Plethodontidae			
Batrachoseps pacificus	California	1/174 (1%)	Goldberg et al. (2000a)
Desmognathus brimleyorum	Arkansas	8/41 (20%)	McAllister et al. (1995c)
D. conanti	Arkansas	1/26 (4%)	McAllister et al. (2013)
Plethodon albagula	Arkansas	1/17 (6%)	McAllister et al. (2014)
P. sequoyah	Oklahoma	2/25 (8%)	McAllister and Bursey (2004)
Anura			
Bufonidae			
Anaxyrus americanus americanus	Iowa, South Dakota	11/415 (3%)	James and Ulmer (1967)
	Iowa	4/101 (4%)	Ulmer and James (1976)
	Michigan**	10/71 (15%)	Muzzall and Andrus (2014)
	Wisconsin	6/47 (13%)	Bolek and Coggins (2000)
		5/30 (17%)	Bolek and Coggins (2003)
		3/39 (8%)	Yoder and Coggins (2007)
A. a. charlesmithi†	Arkansas	4/29 (14%)	This report
A .cognatus	Iowa, South Dakota	1/17 (6%)	James and Ulmer (1967)
A. fowleri†	Arkansas	1/6 (17%)	This report
	Michigan**	6/31 (19%)	Muzzall and Andrus (2014)
A. houstonensis	Texas	2/17 (12%)	Thomas et al. (1984)
Incilius nebulifer	Texas	3/23 (13%)	McAllister et al. (1989)
Hylidae			
Hyla chrysoscelis	Wisconsin	8/65 (12%)	Bolek and Coggins (1998)
Pseudacris crucifer	Wisconsin	2/79 (3%)	Yoder and Coggins (2007)
P. streckeri	Texas	3/42 (7%)	McAllister (1987)

Table 1. *continued*. Summary of reports of *Mesocestoides* sp. in North American amphibians.

Host	State	Prevalence	Reference
Ranidae			
Lithobates berlandieri	Texas	1/2 (50%)	McAllister and Conn (1990)
L. blairi	Iowa	4/16 (25%)	Goldberg et al. (2000b)
	Kansas	1/18 (6%)	Goldberg et al. (2000b)
	Nebraska	5/55 (9%)	Goldberg et al. (2000b)
L. clamitans	Michigan	49/239 (21%)	Muzzall et al. (2001)
	Wisconsin	6/412 (2%)	Williams and Taft (1980)
L. palustris	Arkansas	1/26 (4%)	McAllister et al. (1995a)
L. pipiens	Iowa, South Dakota	27/1568	James and Ulmer (1967)
	Iowa	10/491 (2%)	Ulmer and James (1976)
	Michigan	4/43 (9%)‡	Gillilland and Muzzall (1999)
	New York	1/34 (3%)	McAllister and Conn (1990)
	Wisconsin	1/22 (5%)	Williams and Taft (1980)
		13/31 (42%)	Bolek and Coggins (2003)
L. sphenocephalus utricularius†	Arkansas	6/64 (9%)	This report
L. sylvaticus	Arkansas	3/42 (7%)	McAllister et al. (1995b)
	Wisconsin	9/78 (12%)	Yoder and Coggins (2007)
Scaphiopodidae			
Scaphiopus bombifrons	Oklahoma	2/3 (67%)	McAllister et al. (2005)
S. hurterii	Oklahoma	3/14 (21%)	McAllister et al. (2005)

*Prevalence = number infected/number examined (%).
†New host record.
‡froglets < five mo. old only.
**Note added in proof: After this paper went to press, Muzzall and Andrus (2014) reported *Mesocestoides* sp. in *Anaxyrus a. americanus* and *Anaxyrus fowleri* from western Michigan.

New Host and Distribution Records of the Leech *Placobdella multilineata* Moore, 1953 (Hirudinida: Glossiphoniidae)

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Running Title: New Host and Distribution Records of *Placobdella multilineata*

Placobdella multilineata was described by Moore (1953) based on free-living specimens collected from New Orleans, Louisiana and Norman, Oklahoma (Meyer 1968). *Placobdella multilineata* is a blood-feeding leech with relatively low host specificity, being reported from alligators, amphiuma, and turtles (Sawyer and Shelley 1976, Forrester and Sawyer 1974, Saumure and Doody 1998). The geographic range of *P. multilineata* includes the southeastern United States and extends northward through the Mississippi Valley as far north as Illinois and Iowa (Klemm 1982, 1985). Although, it is a relatively common species, it was only recently reported from Arkansas (Moser et al. 2006, McAllister and Moser 2012).

Between 2007-2014, leeches were collected as follows: a single individual of *P. multilineata* was collected from a broad-banded watersnake (*Nerodia fasciata confluens*) from Big Cane Conservation Area, Butler County, Missouri (36°29’56”N 90°29’40”W) on 6 June 2007; a single free-living individual of *P. multilineata* was collected from Jonesboro, Craighead County, Arkansas (35°45’20.94”N 90°42’43.93”W) on 12 February 2012; ten free-living individuals of *P. multilineata* were collected from Lukfata Creek, McCurtain County, Oklahoma (33°58’05.51”N, 94°45’57.06”W) on 8 October 2011; single individuals of *P. multilineata* were collected from a red-eared slider (*Trachemys scripta elegans*) and a northern diamond-backed water snake (*Nerodia rhombifer*) from a cattle tank in Broken Bow at Lukfata, McCurtain County, Oklahoma (34°00’22.03”N, 94°45’53.81”W) on 11 June and 13 June 2012, respectively; a single individual of *P. multilineata* was collected from a red-eared slider (*T. scripta elegans*) from 7 km east of Harrell on Highway 278, Calhoun County, Arkansas (33°32’09.4”N 92°19’49.5”W) on 11 January 2012; a single free-living individual of *P. multilineata* was collected from Spring Mill off US Highway 69, Independence County, Arkansas (35°49’42”N 91°43’24”W) on 25 July 2013; a single individual of *P. multilineata* was collected from an eastern musk turtle (*Sternotherus odoratus*) from intersection of county road 407 and county road 409, Jonesboro, Craighead County, Arkansas (35°46’08”N 90°42’51”W) on 8 March 2014. Specimens were prepared as described by Moser et al. (2006).

Molecular analyses were conducted on newly collected material according to Richardson et al. (2010). Purified PCR products were sequenced using the HCO2198 primer and the LCO1490 primer for the Cytochrome c oxidase subunit I products by the W. M. Keck Foundation Biotechnology Resource Laboratory at Yale University. The DNA sequences were aligned using Clustal W version 2 (Larkin et al. 2007) and checked manually using SeaView 4 (Gouy et al. 2010) and then analyzed using PAUP* 4.0b10 (Swofford 2002), deposited in GenBank (http://www.ncbi.nlm.nih.gov/genbank/), and compared to other leech DNA sequences contained within Genbank. Uncorrected p distance was calculated using PAUP*.

Leeches were identified with the assistance of taxonomic keys (Klemm 1982, 1985) and examination of the type series of *P. multilineata* (USNM 36383-36484, USNM 36413, USNM 36428, USNM 36435). Voucher specimens of leeches were deposited in the Invertebrate Zoology Collections of the Department of Invertebrate Zoology, National Museum of Natural History (USNM), Smithsonian Institution, Washington, D. C. (USNM 1253384-1253390) and the Peabody Museum of Natural History at Yale University (YPM IZ 58313-58315, 58392 and 67729).

Molecular comparison of 637 nucleotides of CO-I revealed an intraspecific difference of 1.1% (7 nucleotides) between two specimens of *P. multilineata* collected from Lukfata Creek, Oklahoma (GenBank KM396760 & KM396761). An intraspecific difference of 1.3% (8 nucleotides) was found between *P. multilineata* collected from Lukfata Creek, Oklahoma and a specimen of *P. multilineata* (GenBank AY962464) collected from Maurepas Swamp, Louisiana. Comparison of CO-I sequence data of three specimens of *P. multilineata* (GenBank KM396760, KM396761 and AY962464) revealed differences of 13.6 % to 14.0% (86 to 89 nucleotides) from five specimens of *Placobdella parasitica* collected from its type locality (Minnesota; GenBank KF058895 – KF058899), differences of 16.7% to 17.8% (106 to 113 nucleotides) from five specimens of *Placobdella papillifera* from Connecticut (GenBank KC505241–KC505245), differences of 16.4% to 18.0% (104 to 115 nucleotides) from three specimens of *Placobdella ali* from Connecticut and New York (GenBank HM347040–HM347042), and differences of 15.5% to 16.6% (99 to 105 nucleotides) from five specimens of *Placobdella rugosa* from North Dakota (GenBank JX412986–JX412990).

Placobdella multilineata is a relatively large and sharply dorsoventrally flattened species. It is characterized by its five precise longitudinal rows of papillae, narrow, uninterrupted (sometimes interrupted) dorsal-medial line, and stripes on the ventral surface. Examination of the type series *P. multilineata* (USNM 36383-36484, USNM 36413, USNM 36428, USNM 36435) and specimens collected in this study revealed a pattern of two rows of three pre-anal papillae, followed by two pairs of prominent paramedial papillae (Fig. 1). This distinct pre-anal papillae pattern also occurs in *P. ali* and *P. rugosa* (Hughes and Siddall 2007, Moser et al. 2012).

In summary, *P. multilineata* is reported from Missouri for the first time. Recorded hosts for *P. multilineata* are presented in Table 1. New host records in this study include broad-banded watersnake (*N. fasciata confluens*), northern diamond-backed water snake (*N. rhombifer*), red-eared slider (*T. scripta elegans*), and eastern musk turtle (*S. odoratus*). *Placobdella multilineata* has now been reported from 17 species and subspecies of alligators, amphiumas, crocodiles, snakes and turtles.

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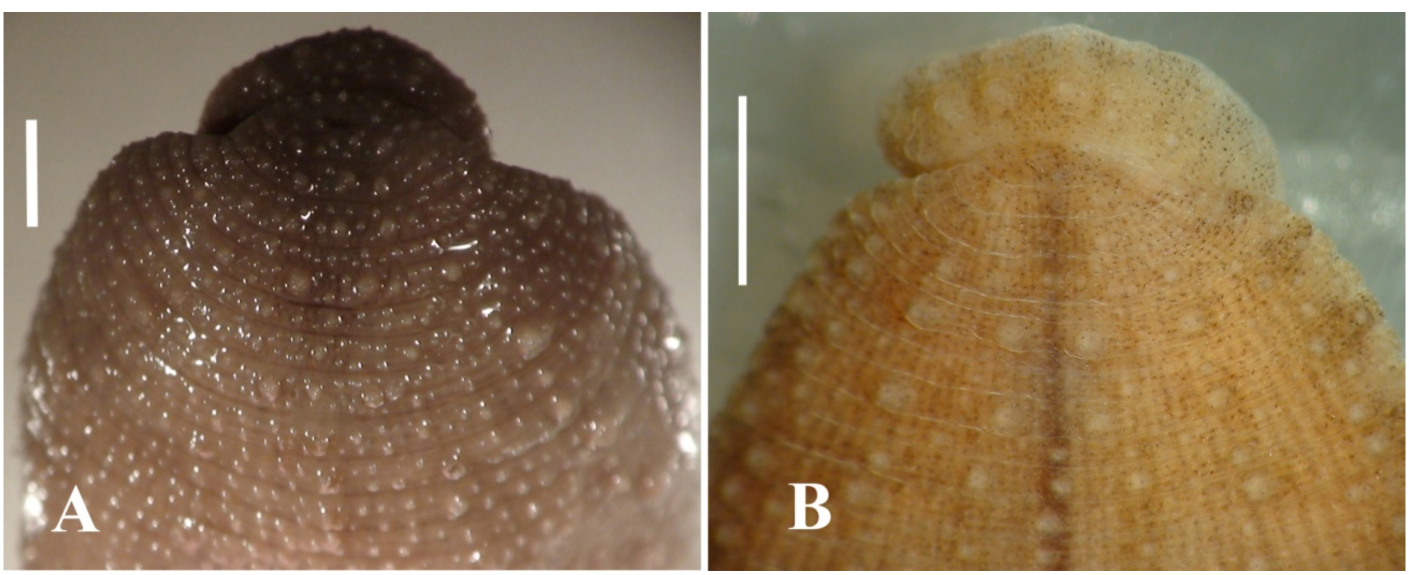

Figure 1. Posterior end of *Placobdella multilineata* showing papillar pattern of pre-anal region.
A) USNM 36384. Paratype, scale bar equals 2 mm. B) USNM 1253387, specimen collected from Lukfata Creek, McCurtain County, Oklahoma in present study. Scale bar equals 2 mm.

Table 1. Reported Hosts of *Placobdella multilineata* Moore 1953

Host	Common name	Reference
Alligator mississippiensis	American alligator	Forrester and Sawyer 1974, Glassman et al. 1979, Khan et al. 1980, Cherry and Ager 1982, Brantley and Platt 1991
*Crocodylus porosus**	estuarine crocodile	Yang and Davies 1985a,b
Amphiuma tridactylum	three-toed amphiuma	Saumure and Doody 1998
Chelydra serpentina	snapping turtle	Stone 1976, Sawyer and Shelley 1976, Readel et al. 2008
Clemmys muhlenbergii	bog turtle	Saumure and Carter 1998, Saumure and Beane 2001
Chrysemys picta	painted turtle	Readel et al. 2008
Kinosternon leucostomum	white-lipped mud turtle	Rossow et al. 2013
Kinosternon scorpioides	scorpion mud turtle	Rossow et al 2013
Macrochelys temminckii	alligator snapping turtle	Forrester and Sawyer 1974
Rhinoclemmys funereal	black river turtle	Rossow et al. 2013
Sternotherus carinatus	razor-backed musk turtle	McAllister and Moser 2012
Sternotherus odoratus	eastern musk turtle	this study
Trachemys scripta	pond slider	Readel et al. 2008
Trachemys scripta elegans	red-eared slider	this study
Trachemys scripta scripta	yellow-bellied slider	Sawyer and Shelley 1976
Nerodia rhombifer	n. diamond-backed watersnake	this study
Nerodia fasciata confluens	broad-banded watersnake	this study

*Accidental infestation at the Beijing Zoo, People's Republic of China

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New Host and Geographic Distribution Records for the Fish Leech *Myzobdella reducta* (Meyer, 1940) (Hirudinida: Piscicolidae)

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Running Title: New Host and Geographic Records for *Myzobdella reducta*

Myzobdella reducta is an opportunistic sanguivorous fish leech originally described by Meyer (1940) from the slenderhead darter, *Percina phoxocephala*, in Illinois as *Piscicolaria reducta*. Based primarily on molecular data, Williams and Burreson (2006) synonomyzed the genus *Pisicolaria* with *Myzobdella*. *Myzobdella reducta* has been reported from a wide variety of fishes from Florida, Georgia, Illinois, Kansas, Kentucky, Maine, Michigan, Minnesota, Nebraska, New Jersey, New York, Ohio, Oklahoma, Pennsylvania, Tennessee, West Virginia, and Wisconsin in the United States and Ontario in Canada (Richardson et al. 2012). In addition, Klemm (1985) indicated, within a distribution map, that *P. reducta* had been reported from Connecticut, Delaware, Louisiana, and Massachusetts although corroborating literature was not referenced.

Herein, we document new host and geographic distribution records of *M. reducta* based on field collections and examination of holdings in museums. Newly collected material was processed as described by McAllister et al. (2012). Voucher specimens were deposited in the Invertebrate Zoology Collections of the Yale Peabody Museum of Natural History (YPM), Yale University, New Haven, Connecticut, U.S.A., or the Harold W. Manter Laboratory (HWML), University of Nebraska State Museum, University of Nebraska-Lincoln, Lincoln, Nebraska, U.S.A. Nomenclature for leeches discussed in this paper follows Klemm et al. (2014). Nomenclature for fishes discussed in this paper follows Page et al. (2013).

On 10 September 2012 and 9 September 2013, fish were examined from the Montague Power Canal Reservoir, an impoundment of the Connecticut River in Franklin County, Massachusetts (42° 35’ 29N, 72° 34’ 41W). One – 6 (mean 2.4) individuals of *M. reducta* occurred on 21 of 31 (67.7%) tessellated darters (*Etheostoma olmstedi*) representing a new host record. The relative distribution of site of attachment for 39 individuals of *M. reducta* on 21 tessellated darters is given in Figure 1. In addition, single individuals of *M. reducta* were collected from 3 of 73 (4.1%) yellow perch (*Perca flavescens*), 3 of 318 (0.9%) bluegill sunfish (*Lepomis macrochirus*), 1 of 5 chain pickerel (*Esox niger*), and 1 of 19 largemouth bass (*Micropterus salmoides*). *Myzobdella reducta* was also collected from 2 of 18 rock bass (*Ambloplites rupestris*), 1 from 1 and 2 from another. Examination of 2 American eels (*Anguilla rostrata*), 26 smallmouth bass (*Micropterus dolomieu*), 3 carp (*Cyprinus carpio*), one channel catfish (*Ictalurus punctatus*), 17 shiners (*Notropis* sp.), 4 pumpkinseed (*Lepomis gibbosus*), and

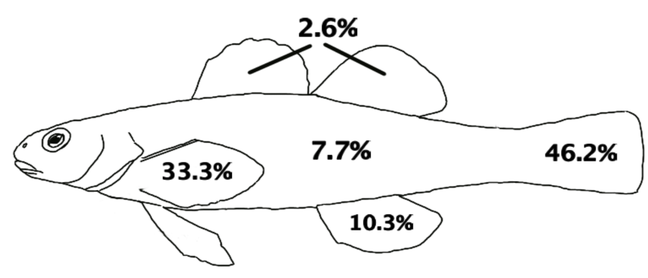

Figure 1. Relative distribution of attachment sites of 39 specimens of *Myzobdella reducta* on the tessellated darter.

one goldfish (*Carassius auratus*) failed to reveal the presence of *M. reducta*. These findings represent the first definitive report of *M. reducta* from Massachusetts and new host records for *A. rupestris*, *E. olmstedi*, and *M. salmoides*.

Examination of material in the Museum of Comparative Zoology (MCZ) at Harvard University in Cambridge, Massachusetts revealed a specimen of *M. reducta* (MCZ56486) collected by D. G. Smith from an individual of *E. olmstedi* from the Missisquoi River in Swanton, Franklin County, Vermont on 23 August 1983 representing a new geographic distribution record for Vermont.

On 3 August 2013, 4 individuals of *M. reducta* were collected from 2 *L. macrochirus* collected from Community Lake in Wallingford, New Haven County, Connecticut, 1 from 1 and 3 from the other. This represents the first definitive report of *M. reducta* from Connecticut.

On 25 October 2013, fish were examined from Little River at Cow Creek Crossing, McCurtain County, Oklahoma for the presence of leeches. Single individuals of *M. reducta* were collected from a crystal darter, *Crystellaria asprella*, and a highland stoneroller, *Campostoma spadiceum*. Two individuals of *M. reducta* were taken from a logperch, *Percina caprodes*. The occurrence of *M. reducta* on *C. asprella* and *C. spadiceum* represent new host records. *Myzobdella reducta* was previously reported from a channel catfish, *Ictalurus punctatus*, from Lake Texoma, Marshall County, Oklahoma by Nagel (1976).

On 5 June 2010 individuals of *M. reducta* were taken from 5 blacktail shiners, *Cyprinella venusta*, collected from Big Sandy Creek near Beaver Slide Trail in the Big Thicket National Preserve, Polk County, Texas, representing a new host record and geographic distribution record. Three shiners were infested with single individuals of *M. reducta*. One shiner was infested with 2 individuals and one was infested with 3 individuals.

On 14 April 2013 a single individual of *M. reducta* was taken from a spotted bass, *Micropterus punctulatus*, collected at Caddo River lower dam, Clark County, Arkansas. On 27 April, 2013, 2 redear sunfish, *Lepomis microlophus* were found to be infested with *M. reducta*, 1 from 1 and 2 from the other. On 2 May, 2013, a warmouth, *Lepomis gulosus* was found to be infested with 6 individuals of *M. reducta*. On 25 October 2013 a single individual of *M. reducta* was taken from an orangebelly darter, *Etheostoma radiosum*, from Rolling Fork River at Johnson Bridge Road, Sevier County, Arkansas.

Examination of museum specimens deposited in the vertebrate collection of the Biology Department at Henderson State University, Arkadelphia, Arkansas revealed a single specimen of *M. reducta* from a shadow bass, *Ambloplites ariommus*, collected from a tributary of the Ouachita River in Hot Spring County, Arkansas on 2 February 1999. The occurrence of *M. reducta* on *A. ariommus, E. radiosum, L. macrolophus,* and *M. punctulatus* represent new host records. *Myzobdella reducta* was previously reported from a pirate perch, *Aphredoderus sayanus*, from Spring Creek in Independence County, Arkansas (McAllister et al. 2012). In addition, Klemm (1982) previously reported *M. reducta* from Arkansas but no specific data were provided (Klemm 1982, Moser et al. 2006).

In summary, *M. reducta* is reported definitively for the first time from Massachusetts and Connecticut and is reported for the first time from Vermont and Texas. *Myzobdella reducta* is reported for the first time from *Amploplites ariommus* (shadow bass), *Ambloplites rupestris* (rock bass), *Campostoma spadiceum* (highland stoneroller), *Crystellaria asprella* (crystal darter), *Etheostoma olmstedi* (tessellated darter), *Etheostoma radiosum* (orangebelly darter), *Lepomis macrolophus* (redear sunfish), *Micropterus punctulatus* (spotted bass) and *Micropterus salmoides* (largemouth bass). New distribution and host information is summarized in Table 1.

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Table 1. Summary of new reports of *Myzobdella reducta* from 6 states of the USA. (*) indicates new locality record or host record.

State	Host	Catalog number(s)
Arkansas	*Ambloplites ariommus* (shadow bass)* *Etheostoma radiosum* (orangebelly darter)* *Lepomis gulosus* (warmouth) *Lepomis microlophus* (redear sunfish)* *Micropterus punctulatus* (spotted bass)*	YPM67720 YPM67721 YPM67727 YPM67726 YPM67725
Connecticut*	*Lepomis macrochirus* (bluegill sunfish)	YPM67718-67719
Massachusetts*	*Ambloplites rupestris* (rock bass)* *Esox niger* (chain pickerel) *Etheostoma olmstedi* (tessellated darter)* *Lepomis macrochirus* (bluegill sunfish) *Micropterus salmoides* (largemouth bass)* *Perca flavins* (yellow perch)	YPM67762-67764 YPM67758 YPM67711-67716 YPM58310 & 58311 YPM67759 YPM67760 & 67761
Oklahoma	*Campostoma spadiceum* (highland stoneroller)* *Crystellaria asprella* (crystal darter)* *Percina caprodes* (logperch)	YPM67722 YPM67723 YPM67724
Texas*	*Cyprinella venusta* (blacktail shiner)	HWML64634
Vermont*	*Etheostoma olmstedi* (tessellated darter)*	MCZ56486

New Records and Notes on the Ecology of the Northern Long-Eared Bat (*Myotis septentrionalis*) in Arkansas

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Running title: New Records and Notes on the Ecology of the Northern Long-Eared Bat

The northern long-eared bat (*Myotis septentrionalis*) has been a common insectivorous bat in much of eastern North America, including Arkansas, which is located near the southwestern edge of its range. While this species is expected to occur throughout the Ozarks and Ouachita Mountains, it has only been previously documented in 19 of 75 Arkansas counties (Harvey and McDaniel 1983, Saugey et al. 1989, Sealander and Heidt 1990, Saugey et al. 1993, Wilhide et al. 1998a, Tumlison et al. 2002, Sasse and Saugey 2008).

In the northeastern United States, there have been significant losses in many bat populations due to white-nose syndrome. Analyses have thus indicated declines in northern long-eared bat summer capture rates and hibernating winter cave populations (Francl et al. 2012, Ingersoll et al. 2013). In 2013, the U.S. Fish and Wildlife Service proposed listing the northern-long eared bat as an endangered species and it is now considered as such within Arkansas (U.S. Fish and Wildlife Service 2013).

We examined 1,464 known *M. septentrionalis* collection events from 1938-2014 that were collected by the Arkansas Game and Fish Commission and report on new records of this species in 16 additional counties (Figure 1).

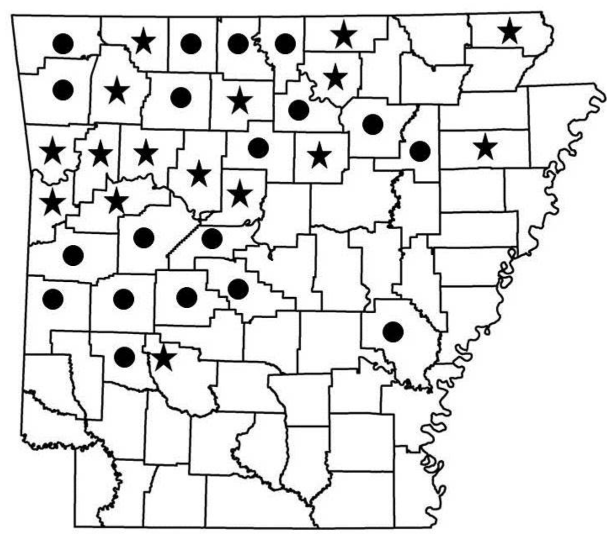

Figure 1. Distribution of the northern long-eared bat in Arkansas. “Stars” indicate new county records. “Solid circles” indicate historical records from Saugey et al (1993), Sasse and Saugey (2008), Sealander and Heidt (1990), Tumlison et al (2002).

Carroll Co.

A single male was captured by MJH inside Bennett Cave on October 27, 1979.

Clark Co.

On January 28, 1994 1 male and 1 female were found by DAS in an abandoned mine south of Amity in the northwestern part of the county.

Clay Co.

On June 19, 2006 1 male and 2 females were captured by TSR in a mist net in Sec. 12 T19N R3E.

Cleburne Co.

On July 29, 2013 a male was found alive on a homeowner’s deck in the town of Tumbling Shoals and submitted to the Arkansas Department of Health for rabies testing and was identified by DAS.

Conway Co.
On March 17, 2004 2 males were captured by DAS in a mist net set over a road in Sec. 5 T5N R18W. One male and one female were caught by DAS in this same location on July 5, 2004.

Crawford Co.
On December 31, 1997 a single bat was observed inside a crevice cave in Sec. 17 T12N R31W (Bill Puckette, personal communication).

Franklin Co.
On January 3, 1997 a single bat was seen in each of two difference crevice caves in Sec. 8 T11N R27W (Bill Puckette, personal communication).

Fulton Co.
On March 26, 2012 a female from the town of Sturkie was submitted to the Arkansas Department of Health for rabies testing and identified by DAS.

Izard Co.
On August 16, 2002 4 bats were observed in Bergren Cave (G.O. Graening, personal communication).

Johnson Co.
On July 6, 2004 4 males were captured by JLJ in a mist net set over a stream in Sec. 19 T12N R21W. Three more males were captured by JLJ at the same site on July 7, 2004.

Logan Co.
A female was captured by MLC in a mist net on August 21, 2001 on the Cold Springs Ranger District, Ozark-St. Francis National Forests.

On May 20, 2004 8 males and 9 females were captured by DAS in a mist net in Sec. 24 T6N R25W.

Madison Co.
On March 8, 1976 4 males were collected by J. Priday 22.4 km north of Fredericktown and specimens were deposited in the museum of Arkansas State University (Specimens ASU 2130, 2188-2190).

Poinsett Co.
On May 8, 2013 an adult male was captured by TSR in a mist net set over a trail in Sec. 36 T12N R1E on the Earl Buss Bayou DeView Wildlife Management Area.

Pope Co.
On June 12, 2001 2 females were captured by MLC in a mist net on the Bayou Ranger District, Ozark-St. Francis National Forests.

On June 29, 2004 one female was captured by JLJ in a mist net set over a road in Sec. 2 T9N R18W.

Searcy Co.
On July 20, 2008 4 males were captured by DBS in a mist net set over a road in Sec. 19 T15N R18W. One female was captured by DBS in a different site in that section on June 29, 2009.

Sebastian Co.
On August 3, 2005 a female was captured in a mist net set over a trail in Sec. 4 T3N R31W (Lisa Gatens, personal communication).

Examination of records maintained by the Arkansas Game and Fish Commission and the authors of this paper found other observations on the life history of this species worthy of note.

The northern long-eared bat is generally found only in the Ouachita and Ozark mountains of western and northern Arkansas. They have been found only occasionally in the Delta region in the eastern part of the state and do not seem to be common in bottomland hardwoods despite their use of this habitat in other parts of their range (Carter and Feldhamer 2005, Fokidis et al. 2005, Medlin et al. 2006). On September 4, 1986 a male bat from Stuttgart in Arkansas County was submitted to the Arkansas Department of Health for rabies testing and was identified by DAS. This species was commonly captured by TSR in mist nets set in bottomland hardwood habitat on the Dave Donaldson Black River Wildlife Management Area in Clay County during the summer of 2006 and one was captured on the Earl Buss Bayou DeView Wildlife Management Area in Poinsett County in 2013.

Caves were utilized by northern long-eared bats throughout the year. However, more than 10 hibernating bats were seen in only 11 caves and more than 100 bats in only 2 caves. Fitton Cave in Newton County is the only cave that has supported relatively large numbers of hibernating northern long-eared bats;

Arkansas Game and Fish Commission records indicate that surveys found only 1-5 hibernating bats prior to 1997, but the population has grown since to as high as 391 in 2014.

Small numbers of males used caves as roosts throughout the summer but females were found roosting inside a cave during this period only once when 2 were captured by DAS inside Spillway Mine in Garland County on May 27, 2008. At Reed Cave in Marion County, 7 males were clustered together with at least 30 Ozark big-eared bats (*Corynorhinus townsendii ingens*) on June 20, 1995 (Wilhide et al. 1998b).

Northern long-eared bats appear to leave hibernation in mid-to late March but spring records are rare. A female was submitted for rabies testing from West Fork in Washington County on March 11, 2003 and a female from Fulton County was submitted on March 26, 2012 and both were identified by DAS. Bats have been captured in mist nets set over roads and ponds as early as March 17, 2004 at a site in Conway County, and at 3 sites in Boone County from March 25-30, 2008.

Female bats were found by DAS roosting in Spillway Mine in Garland County in late April and May. Two females, one of which was pregnant, were observed there with 4 males on May 27, 2008. On April 20, 2010, 12 females that may have been in the early stages of pregnancy were found in the mine and 8 days later 10 pregnant females, and 1 female, for which reproductive status was not recorded, were captured there. On April 20, 2011, 16 pregnant females roosted in the mine.

During mist netting conducted by the authors in summer months from 1996-2013, pregnant females were captured from May 3-June 24, lactating females from May 19-July 20, and the first capture of volant juveniles occurred from June 6-July 20.

The northern long-eared bat does not commonly roost in buildings or other manmade structures (Krochmal and Sparks 2007, Henderson and Broders 2008, Timpone et al. 2010); however there were several occurrences of this in Arkansas. At least 2 male and 10 female bats were captured inside a private home in Newport on July 23, 1999 (Grippo and Massa 2000). On August 16, 2013 a male and female bat were captured by PNJ while roosting on the side of a log cabin in Newton County.

Acknowledgments

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Size and Age Records for an Arkansas Specimen of the American Bullfrog, *Lithobates catesbeianus* (Anura: Ranidae)

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Running Title: Size and Age Records for an Arkansas Specimen of *Lithobates catesbeianus* (Anura: Ranidae)

The American Bullfrog (*Lithobates catesbeianus*), North America's largest anuran, is widely distributed throughout the lower 48 United States, and populations have been introduced into western Canada and Hawaii (Dodd 2013) as well as many other regions of the world (Kraus 2009), such as China (Wang and Li 2009). This species occurs in nearly every county in Arkansas (Trauth et al. 2004), and is a prized game animal. The largest body size ever recorded for the American Bullfrog is 204.2 mm snout-vent length (SVL), measured from an adult female collected from Cleveland County, Oklahoma in 1995; its mass was 908.6 g (Lutterschmidt et al. 1996). In Arkansas, the body size normally ranges from 90-152 mm SVL (McKamie and Heidt 1974, Trauth et al. 2004) but, in rare instances, can reach nearly 190 mm. In the following, we report on the largest known specimen of this species ever reported from Arkansas and provide an estimate of its age using skeletochronology.

Skeletochronology, a histological technique for determining annual growth increments by counting lines of arrested growth (LAGs) in bones in temperate amphibians and reptiles, has been shown to be a reliable method for age determination (Castanet and Smirina 1990). A number of recent skeletochronological studies have used ranid frogs (e.g., Bastien and Leclair 1992, Tsiora and Kyriakopoulou-Sklavounou 2002, Lai et al. 2005, Kyriakopoulou-Sklavounou et al. 2008, Liao 2011, Sarasola-Puente et al. 2011) to determine growth, size, age of maturity, and longevity in frog populations throughout the world.

On April 27, 2013, one of us (TAW) collected a gravid female American Bullfrog (Fig. 1) from Gum Slough Ditch at its intersection with St. Hwy 230 (35º, 54', 33.53"N; 90º, 50', 50.78"W) approximately 3.6 km west of Bono (Craighead County), Arkansas. The specimen was brought to the Department of Biological Sciences at Arkansas State University, photographed, and massed (784.5 g). Its body length measured 195 mm from snout tip to groin. The frog was deposited into the Arkansas State University herpetological collection (ASUMZ 32687).

In order to determine an age estimate of this specimen, the diaphyseal portion of the left tibiofibula and a phalangeal segment of the 2nd digit of the right pes were removed and placed into decalcifying agent (1% v/v hydrochloric acid) for 3 days and then

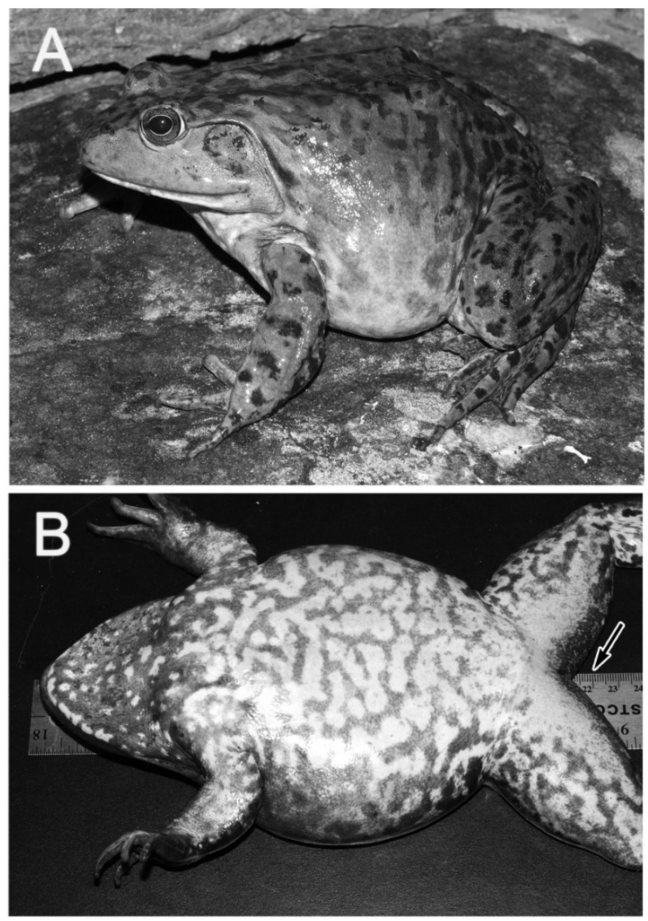

Figure 1. Adult female American Bullfrog, *Lithobates catesbeianus* (ASUMZ 32687), from Craighead County, Arkansas. A. Left lateral view of specimen. B. Ventral surface of specimen lying supine on a metric ruler; arrow points to 220 mm.

transferred into 50% v/v ethanol for temporary storage. Later, these bones were dehydrated in a series of graded ethanol solutions in preparation for paraffin infiltration and embedding. Bones were then sectioned with a rotary microtome at a thickness of 10 µm and affixed to microscope slides using Haupt's adhesive. Slides were stained with hematoxylin (6 min) and eosin (45 sec) using a standard histological protocol (Presnell and Schreibman 1997). A Nikon Eclipse 600 epi-fluorescent light microscope with a Nikon DXM 1200C digital camera was utilized to obtain photomicrographs.

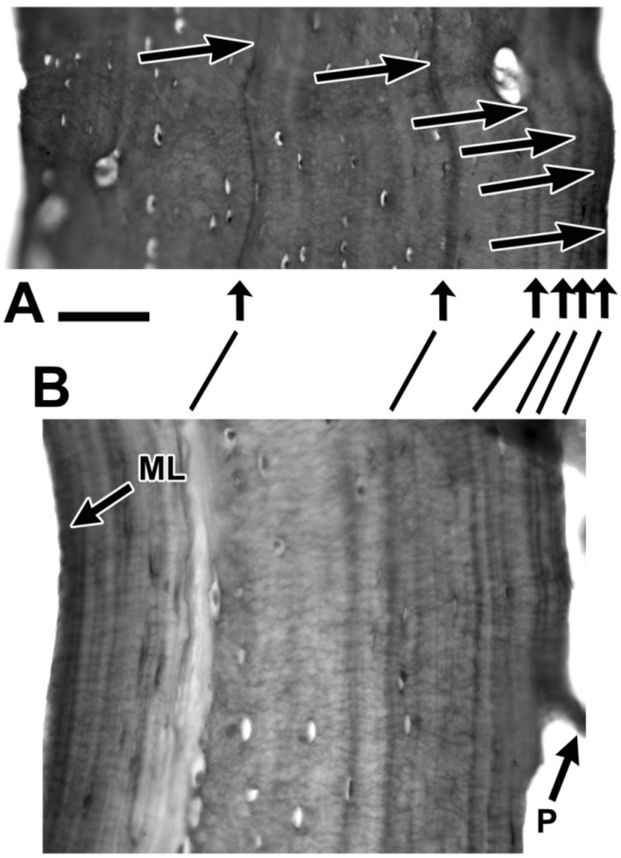

Figure 2. Transverse sections of different regions of the diaphysis of the tibiofibula of the adult female American Bullfrog, *Lithobates catesbeianus* (ASUMZ 32687). A. Arrows point to lines of arrested growth (LAGs); scale bar = 50 µm. B. Similar sectional plane as shown in A. Lines link LAGs between A and B. ML = metamorphosis line; P = periosteum.

The results of skeletochronology of the tibiofibula of ASUMZ 32687 revealed an age estimate of six yr based upon number of LAGS (Fig. 2). In addition, no endosteal resorption was evident. Several false LAGs were observed, however, and these were especially evident during the first year of life and also during year 2 through 4 (see Fig. 2B). The maximum age of an American Bullfrog is 7 years, 3 months and 24 days, recorded from a specimen held in captivity at the Philadelphia Zoo (Snider and Bowler 1992).

Acknowledgments

Deposition of the American Bullfrog into the Arkansas State University herpetological collection was authorized by the Arkansas Game and Fish Commission under Scientific Collection Permit No. 020520134 to SET.

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Natural History Notes and New County Records for Ozarkian Millipeds (Arthropoda: Diplododa) from Arkansas, Kansas and Missouri

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Running Title: Natural History Notes on Millipeds

Over the past decade, there have been numerous new geographic records documented for milliped distributions in Arkansas (see McAllister et al. 2013 and refs.) and, to a lesser degree, for Kansas and Missouri (Gunthorp 1913, 1921, Chamberlin 1928, Shelley and Snyder 2012), but little is known about their natural history in these states (Youngsteadt 2008, 2009). Here we summarize observations on several Ozark millipeds within six orders and nine families that the senior author made over the last eight years, and add several new geographic distribution records.

Unless noted, specimens were collected from under logs or rocks in woodland habitat. Specimens were maintained in 11 or 16 cm diameter clear-plastic deli dishes provided with clay, wood, rock, and/or soil as a substrate to approximate the natural microhabitat. Millipeds were kept in a general purpose room that had windows, but was also artificially lighted when too dark for other purposes. The temperature varied with time of day and season from about 13 to 29°C (55 to 85°F). The most common food items provided were baker's yeast and compost in the blackened stage that was derived largely from oak/sweet gum leaves and scrap fruits and vegetables. Tetramin® tropical fish food was sometimes provided and, occasionally, carrot or potato peels, raw ground beef, or freshly killed insects. Photographs/photomicrographs were taken with a Canon Power Shot SX-100, 110 or 160 IS digital camera, either directly or through an ocular of a stereomicroscope. Some were taken through the clear plastic of the deli dish. Each annotated account below begins with the taxon studied along with the longest time one of the individuals lived, the collection sites and dates the millipeds were collected, Voucher specimens (photovouchers) of millipeds representing new county records are on deposit in the Sam Noble Oklahoma Museum of Natural History, Norman, Oklahoma.

Platydesmida: Andrognathidae

Brachycybe lecontii Wood – lived at least 1.3 yrs. Lake Leatherwood Park, Eureka Springs, Carroll County, Arkansas (27 Nov., 29 Dec. 2011); ca. 16 km SSE of Ozark, Christian County, Missouri (22 Apr., 11 May 2013. These pink to red millipeds were about 2 cm long and found in colonies under logs. They apparently feed on the microorganisms that live on the rotting wood. To molt, these millipeds curled up in a protected place and shed their skins after about 10 days. They did not eat their exoskeletons. Mating and egg-laying were not observed, but about 24 eggs appeared that were being tended by an adult that had its anterior half wrapped around them (Fig. 1A). This adult was not sexed, but male brooding of eggs has been documented in several *Brachycybe* species (Shear 1999, Kudo et al. 2010). The eggs hatched in 21 days, during which time the adult remained in the same place with them. The eggs were 0.6 mm in diameter, but swelled a bit before hatching. The hatchlings young (Figs. 1B-C) had five pairs of legs (Fig. 1C) and were 2.0 to 2.5 mm long; an older hatchling is shown in Fig. 1D. None survived more than several days.

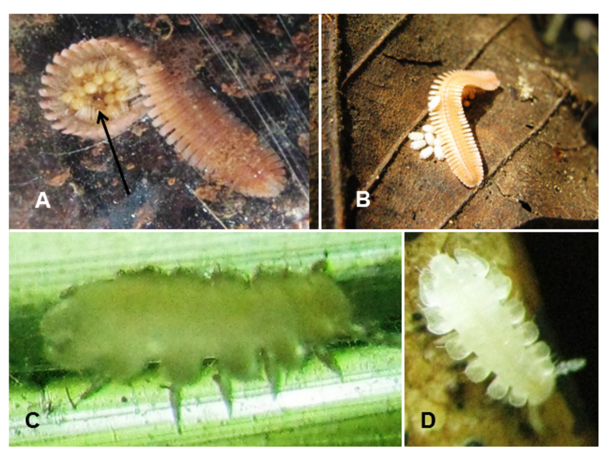

Figure 1. *Brachycybe lecontii*. A. Adult brooding eggs. B. Newly-hatched young. C. Six days old. D. New hatchling.

We also documented a new county record for *B. lecontii* collected on 22 Apr. 2013, ca. 16 km SSE of Ozark, Christian County, Missouri (see Shelley et al. 2005b).

Chordeumatida: Cleidogonidae

Various species. – lived 1-2 mos. Greene and Taney cos., Missouri and Carroll Co., Arkansas (Jan. - Apr., Oct.-Dec., 2007-2013). *Tiganogona* is a common local genus, but some of the specimens may have belonged to other genera (i.e., *Cleidogona*). The longest mating (Fig. 2A) lasted at least 18 hrs during which the female would sometimes walk around with the male twisting his body and using his posterior and middle legs to keep up. Although no eggs were observed, hatchlings (Fig. 2B) that appeared in March were 1.0-1.2 mm long, had five pairs of legs, eye spots, and curved setae. None lived long enough to molt.

Figure 2. A. Cleidogonid mating pair. B. Hatchling; scale bar = 500 µm.

Trichopetalidae

Trigenotyla parca Causey. – Nov.; lived 5 months. Lake Leatherwood Park, Eureka Springs, Carroll County, Arkansas (27 Nov. 2011). This single male was 0.8 cm long and it had sticky droplets at the bases of its long segmental setae. If the sticky droplets were touched with a needle, they would stretch into strands as the needle was withdrawn as they did in the closely related *Causeyella* (Youngsteadt 2008). Shear (2008) discussed milliped spinnerets and a possible homology between the segmental setae (with their sticky droplets) and the spinnerets.

Callipodida: Abacionidae

Abacion spp. **–** lived one yr. E of Springfield near Turners, Greene County, Missouri (17, 20 Apr., 12 Jun. 2007); N side of McDaniel Lake, N of Springfield, Greene County, Missouri (15 Apr. 2009); N of Wakefield, Clay County, Kansas (28 May 2011); Lake Leatherwood Park, Eureka Springs, Carroll County, Arkansas (28 Oct. 2011). Two of five *Abacion texense* collected in Kansas produced silken molting cocoons (Fig. 3), but their construction was not observed and both died in the cocoons before molting. Enghoff and Akkari (2011) described a dense callipodidan cocoon in more detail and noted that it was the first such report since 1874. That gap between publications tends to characterize the present state of milliped biology. Enghoff and Akkari had hoped others would look for similar cocoons and add more detail.

The *A. texense* from northcentral Kansas represent a new county record for Clay County (see McAllister and Shelley 2010). They were collected from under rocks in prairie habitat.

Figure 3. *Abacion texense* showing silky cocoon.

Polydesmida: Xystodesmidae

Apheloria virginiensis reducta Chamberlin. – lived four mos. Ca. 16 km SSE of Ozark Christian County, Missouri (23 May 2007, 18 May 2011); Beachler Ridge, ca. 19 km SSE of Ozark, Christian County, Missouri (2 Apr., 26 Oct. 2010, 3 Mar., 6 May 2011). These four cm long millipeds were black with bright yellow markings. Data suggested a one yr life cycle: adults were collected in the spring, mating occurred in May and June, young appeared in July (and lived three mos), adults died in late spring or summer, and sub-adults were collected in October.

When males attempted to mate, the females attempted to avoid them. If the male did attain the belly to belly mating position, he curled his head and anterior segments around the front of the female's head and apparently tried to push it back, while the female tried to keep her head tucked. Such female resistance resulted in many unsuccessful mating attempts. In general, matings were relatively short, the longest lasting about 30 min.

Although eggs were never seen, young did appear in a dish that also contained other kinds of millipeds.

Since the young did not resemble the hatchlings of the other kinds, it was assumed they were *Apheloria*.

The first instars moved slowly and spent most of their time in the soil. They were white, 1.2-1.5 mm long, had three pairs of legs and seven segments counting the epiproct. They had longer setae than the first instars of the other polydesmidans; the lateral mid-body setae were about two-thirds the width of the body. There were three of these setae per side per segment including dorso-laterals, laterals, and ventro-laterals. The laterals stuck straight out with only a slight curve. Second instars were 2.1-2.2 mm long, had 10 segments and six pairs of legs. Third instars were about 3.2 mm long with 11 pairs of legs. A possible fourth instar was 3.6 mm long with mid-body setae about one-fourth the width of the body. Stages progressed as follows: a first instar was molting on 16 Aug.; a second instar was seen on 20 Aug.; a third instar was seen on 14 Sept.; the possible fourth instar was seen on 19 Oct.

All specimens from Christian County, Missouri document a new county record; previous reports from the state include Barry, Cole, Franklin, Howell, Oregon, Pulaski, Shannon St. Louis, Stone and Taney counties (see Shelley and McAllister 2007).

Euryuridae

Auturus evides (Bollman). – lived five mos. Hatchlings (Fig. 4A) appeared in June and lived two yrs. E of Springfield near Turners, Greene County, Missouri (30 Mar., 18 May 2007, sometime before 9 May 2008, inadvertently introduced with wood); N side of Fellows Lake, Greene County, Missouri (1 Jan. 2012). These 3.5 cm long millipeds mated frequently from March to May in a manner typical of polydesmidans: belly to belly with the front of the male curled over the head of the female and his legs firmly enclosing her, particularly toward the front. If the animal was upside down, it was not unusual for the posterior part to be twisted so the legs were on the ground. Mating lasted for over an hour.

Eggs were laid in hollows beneath the soil in clusters of 12 to 30. They were tan and about 0.45 mm in diameter. They swelled somewhat before hatching and became more grayish and translucent. Hatching was not synchronous, but proceeded for two or more weeks from a given clutch. The hatchlings (Fig. 4A) were slow and lethargic compared to those of *Pseudopolydesmus pinetorum* (herein), and did not form a flock. A few survived to adulthood.

Molting took place in an igloo-like chamber constructed of fecal pellets shaped by the everted rectum; the chambers sometimes had a chimney-like structure on the side (Fig. 4B). Chambers varied with the size of the builder, but one with an outside diameter of 14 mm had walls 2 mm thick. Construction took about a day and the milliped spent about 10 days in it before the skin was shed. It might spend another three days in the chamber before exiting, usually without eating the skin.

We document a new county record (Greene County) in Missouri for *A. evides*. Shelley (1982) previously reported this milliped from 25 other counties of the state.

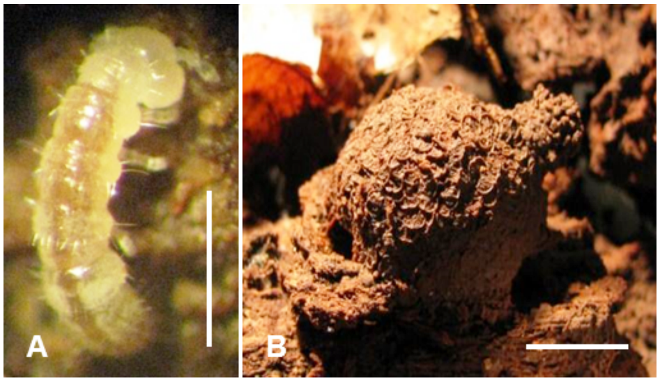

Figure 4. *Auturus evides*. A. Hatchling; scale bar = 1 mm. B. Molting chamber; scale bar = 5 mm.

Polydesmidae

Scytonotus granulatus (Say). – lived three mos. Young appeared in April and May. E of Springfield near Turners, Greene County, Missouri (30 Mar. 2007, 4 Feb., 13 Nov. 2008); N side of McDaniel Lake, Greene County, Missouri (20 Feb. 2009); Beachler Ridge, ca. 19 km SSE of Ozark, Christian County, Missouri (2 Apr. 2010). These bumpy-backed millipeds were about 1.2 cm long. Mating occurred around spring, and in one observed case the male mounted the female from behind, crawled forward, and then turned belly to belly with his anterior end curled around her head. His legs completely surrounded her, including her legs. The longest mating lasted at least 3.5 hrs.

On 22 Apr., a female had built an open-topped igloo-like egg chamber, apparently of fecal material (construction was not observed) in which she laid 12 eggs. The eggs were white to tan, spherical, and had a diameter of 0.4 mm. By 3 May they were less spherical, enlarged to about 0.5 mm in diameter, grayer and more translucent, and embryonic structure was apparent in some. By 5 May, one egg was somewhat crescent shaped and about 0.8 mm long. They all hatched later that day (except one that was apparently infertile) and the hatchlings dispersed.

The first instars were white, 1.1 to 1.4 mm long, had seven segments counting the epiproct, and three pairs of legs. Like the adults, they had bumps on their backs but were covered with short, hooked setae. Successive instars were increasingly coordinated, but remained white with hooked setae through the fifth instar, at which the last one died at about three months old.

We document a new county record (Greene County) for *S. granulatus* in Missouri. Shelley et al. (2005a) previously reported this milliped from Boone, Calloway, Chariton, Christian, Cole, Dent and Phelps counties.

Polydesmidae

Pseudopolydesmus pinetorum (Bollman). – Springfield, Greene County, Missouri (20 Aug. 2007, 13 Mar. 2011); Beachler Ridge, ca. 19 km SSE of Ozark, Christian County, Missouri (13 Oct. 2008); N of Wakefield, Clay County, Kansas (28 May 2011). This account summarizes some observations made by Youngsteadt (2009) and adds new information. These millipeds were about 2 cm long and had a one yr life cycle. The females built igloo-like egg chambers constructed of fecal material and laid several clutches of eggs in the spring (Figs. 5A and 5B) before they and the males died in the summer. The fecal material used for chamber construction was shaped by the everted rectum and the eggs were laid in the chamber as construction proceeded. The eggs hatched synchronously after about 8 to 18 days, depending on temperature, after which the hatchlings spent another two days in the chamber before one of them made a hole in the chamber wall through which they all departed, one after another (Fig. 5D). These first instars stayed together as a flock (Fig. 5C). Molting took about 10 days and occurred in chambers comparable to egg chambers. Adulthood was reached after seven molts. Some reached this stage and mated in the fall, but others overwintered in their sixth or seventh stadia to become adult and mated in the spring. Regardless, egg laying was centered near springtime. Mating in this species lasted up to two days.

If prodded with a paintbrush bristle, the first instars produced a clear secretion from a pair of stalked lateral pores on the fifth segment; the secretion was assumed to be repellant. More recent testing demonstrated that if an object was inoculated with the secretion and held in front of a first instar, the first instar stopped moving, laid its antennae back, and either retreated or changed direction. Second and third instars and adults also responded, but first and second instars of *A. evides* (herein) did not obviously do so.

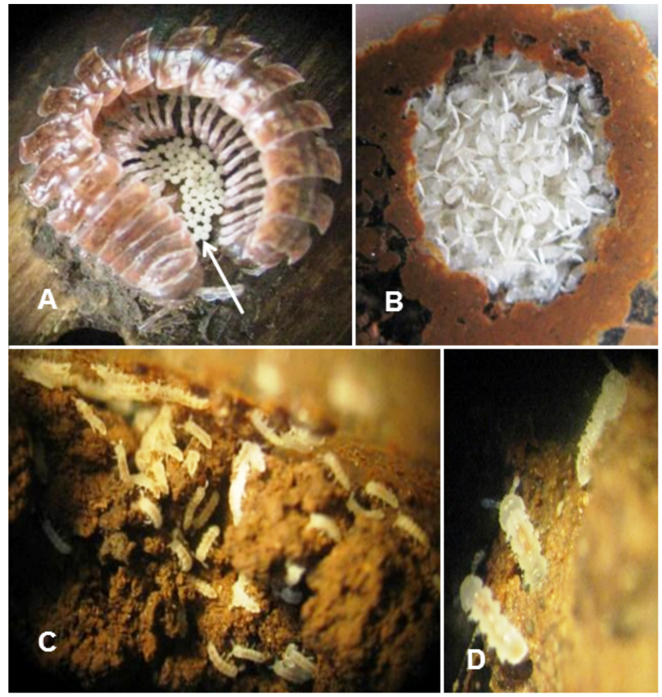

Figure 5. *Pseudopolydesmus pinetorum*. A. Female laying eggs (arrow). B. Egg chamber with hatchlings. C. Flock of first instars. D. Hatchlings exiting egg chamber.

With continued prodding, a first instar also produced what looked like a clear droplet of fluid from its rectum, and this was reported as a droplet by Youngsteadt (2009). However, further observation suggested that the “droplet” was more likely an everted rectum. In addition to being everted when individuals were prodded, they were also commonly everted as individuals moved about in a flock; every once in a while an individual would lift its abdomen a bit and rather quickly evert and retract its rectum. Since these millipeds are blind, the idea emerges that the everted rectum might emit a signaling substance that helps organize the flock. However, no individual movement or movement of the flock as a whole seemed to be influenced by the eversions.

As a test for possible cryptic species, males and females from different locations were placed together to see if they would mate; all did. The most distant locations were separated by about 422 km (264 mi), and included a male from Springfield, Missouri (Greene County) and two females from Wakefield, Kansas (Clay County). Offspring were also produced, but since one of the females laid eggs prior to the mating, neither was likely virgin. Overall, the longest-surviving colony of this species died out in its third generation.

As previous records of *P. pinetorum* reported by Withrow (1988) are from an unpublished dissertation, we take this opportunity to document new county records for *P. pinetorum* from Christian and Greene counties, Missouri, and Clay County, Kansas.

Julida: Parajulidae

Species 1 – lived two months and was released. N side of McDaniel Lake, Greene County, Missouri (20, 25 Feb. 2009). These 4 cm long millipeds (Fig. 6) resembled snakes in the way they twisted and curled, particularly during mating, which lasted just over an hour for the longest record.

Figure 6. Parajulids mating.

Species 2 – lived two mos. Greenway trail by prairie plots ca. 0.8 km S of Pershing School, Springfield, Greene County, Missouri (8, 12 Nov. 2011). These 4.5 cm long millipeds were generally similar to species 1 above. Four matings or attempted matings were observed in November but none lasted more than a few minutes. In one case when a male met a female, they quickly coiled into a mating knot, but then quickly separated. The knotting was almost snap quick when they met. Later, two were observed to touch, but one quickly retreated.

Spirobolida: Spirobolidae

Narceus americanus (Palisot de Beauvois). – the original adults lived six mos, but one juvenile was released when nearing three years old. E of Springfield near Turners, Greene County, Missouri (17 Apr., 10 Aug. 2007). One young that appeared in Apr. lived about 11 mos; one that appeared in Aug. was released when nearing 3 years old. These 5 or 6 cm long millipeds ate rotting wood and compost and their fecal pellets, the color and texture of wood, suggested that wood was the major component. Juveniles reduced dead oak leaves to veins but left most other kinds uneaten.

Mating was observed in Sept.; the longest lasted at least five hours. Coupling was typical for millipeds with the head and anterior part of the male curved over the head of the female, but as mating progressed, the male moved its anterior part slowly back and forth over the female's head in approximately two-second cycles.

A single egg appeared in May. It was grayish-white, slightly ovoid, and the long dimension was about 1.5-1.6 mm. It was deposited on the bottom of the dish under a mud capsule. The smallest juveniles observed were 1 to 1.5 cm long.

These millipeds (Fig. 7) coiled in a protected spot to molt, which took about 10 days for juveniles and 20 for adults. In one case in which a 1.5-2 cm long individual was observed, its skin finally split between the head and collum and the milliped crawled out through the slit. About 5 segments were added during the molt: it emerged with 40-42 segments counting the collum and epiproct from an exoskeleton that had 35-36.

Figure 7. *Narceus americanus*.

Regarding growth, a 1.5 cm long juvenile grew to 3.5 – 4 cm in about 11 months; another that was 1+ cm long grew to about 5.5 cm in almost three years.

We document a new county record for *N. americanus* in Greene County. Missouri. Shelley et al. (2006) previously reported this milliped from 22

Missouri counties.

In summary, we have provided some new natural history information for several millipeds collected from three states that help augment previously published accounts. In addition, we report nine new geographic distribution records for some of these millipeds. We still need more information on milliped ecology and natural history and undoubtedly, with additional study, that should become available to diplopodologists in the near future.

Acknowledgments

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The editorial staff also extends our heartfelt appreciation for the expertise, assistance and valuable time provided by our colleagues who acted as reviewers for the Journal. Our expert reviewers are recruited from within Arkansas, North America, Europe, South America, Australia and Asia. Only through the diligent efforts of all those involved that gave freely of their time, can we continue to produce a high quality scientific publication.

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Proposed timetable for manuscript processing

2 days before AAS annual meeting: authors e-mail manuscript to Managing Editor (istill@atu.edu).

AAS annual meeting: authors submit electronic (on CD) and hard copy to editorial staff at the meeting.

End of May: Initial editorial review. Manuscripts sent to reviewers.

End of July: All reviews received. Editorial decisions on reviewed manuscripts. Manuscripts returned to authors for response to reviewers' critiques. Please email the Managing Editor if you fail to receive your review by the 31st July.

End of August: Authors return revised manuscripts to Managing Editor, 28 days after editorial decision/reviewers critiques were e-mailed. Corresponding author submits publication charges to the Editor-in-Chief (mhemmati@atu.edu): Mailing address: Mostafa Hemmati, P.O. Box

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The prompt return of revised manuscripts and payment of publication costs is critical for processing of the *JOURNAL* by the *JOURNAL* staff. If the corresponding author will be unable to attend to the manuscript within the framework of this schedule, then it is the responsibility of the corresponding author to make arrangements with a coauthor to handle the manuscript. NB. The corresponding author will be responsible for submitting the total publication cost of the paper by August 31st. Failure to pay the publication charges by the deadline will prevent processing of the manuscript, and the manuscript will be added to the manuscripts received from the following year's meeting.

Preparation of the Manuscript

A. General considerations

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A.E. Firstauthor[1*], B.F. Second[1], C.G. Third[2], and D.H. Lastauthor[1] (12 point font, normal, centered)

[1]Department of Biology, Henderson State University, Arkadelphia, AR 71999
[2]Arkansas Game and Fish Commission, 915 E. Sevier Street, Benton, AR 72015 (10 point font, italic, centered)

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Figure 1. Layout of the title section for a submitted manuscript.

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3. Materials and Methods

Sufficient details should be included for readers to repeat the experiment. Where possible reference any standard methods, or methods that have been used in previously published papers. Where kits have been used, methods are not required: include the manufacturer's name and location in brackets e.g. "RNA was prepared using the RNeasy Plus Micro Kit (Qiagen, USA)."

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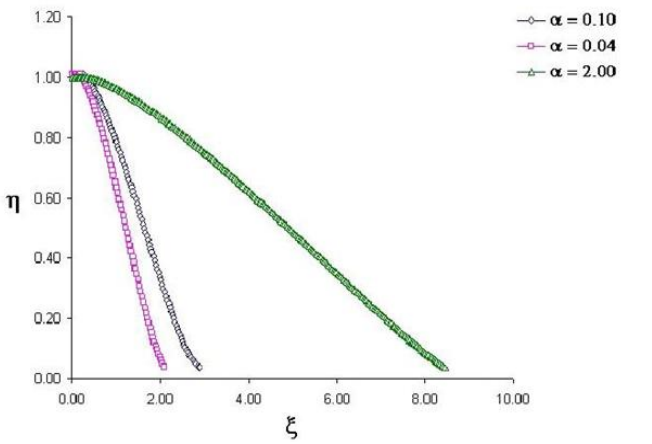

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Author(s) of the part. Year. Title of the part. *In* author(s) or editor(s) of the book. Title of the book. Publisher (Place of publication). Pages of the part.

Weins JA. 1996. Wildlife in patchy environments: Metapopulations, mosaics, and management. *In*: McCullough DR, editor. Metapopulations and wildlife conservation. Island Press (Washington, DC). 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. Pergamon (New York). p 122-30.

Dissertations and Theses – General format is given first.

Author. Date of degree. Title [type of publication – dissertation or thesis]. Place of institution: name of institution granting the degree. Total number of pages. Availability statement.

The availability statement includes information about where the document can be found or borrowed if the source is not the institution's own library.

Stevens WB. 2004. An ecotoxilogical analysis of stream water in Arkansas [dissertation]. State University (AR): Arkansas State University. 159 p.

Millettt PC. 2003. Computer modeling of the tornado-structure interaction: Investigation of structural loading on a cubic building [MS thesis]. Fayetteville (AR): University of Arkansas. 176 p. Available from: University of Arkansas Microfilms, Little Rock, AR; AAD74-23.

Stevens WB. 2004. An ecotoxilogical analysis of stream water in Arkansas [dissertation]. State University (AR): Arkansas State University. 159 p.

<u>Published Conference Proceedings</u> – General format is given first.

Author(s)/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.

<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.

<u>Electronic Journal Articles and Electronic Books</u> should be cited as standard journal articles and books except add an availability statement and date of accession following the page(s):

653 p. Available at: www.usfw.gov/ozarkstreams. Accessed 2004 Nov 29.

<u>Online resources</u>

Citation depends on the requirement of the particular website. Otherwise use the “electronic journal article” format.

US Geological Survey (USGS). 1979. Drainage areas of streams in Arkansas in the Ouachita River Basin. Open file report. Little Rock (AR): USGS. 87 p. <www.usgs.gov/ouachita> Accessed on 2 Dec 2005.

<u>Cited in text as:</u> (USGS 1979)

<u>Multiple Citations are Cited in text as:</u>

(Harris and Gordon 1988, Steiner et al. 1992, Johnson 2006).

8. Submission of Obituaries and *In Memoria*

The Executive Committee and the Journal of the Arkansas Academy of Science welcome the opportunity to pay appropriate professional honor to our departed Academy colleagues who have a significant history of service and support for the Academy and Journal. The editorial staff will consider obituaries for former executive committee members to be included in the Journal. Additional obituaries not meeting these criteria will be forwarded to be posted on the Academy website. We would request that paid up members of the Academy that wish to write an obituary provide a one to two page professional description of the scientist’s life that should include details of his/her contribution to the Academy and publication record. The format should follow the two column format and 11pt Times New Roman font. A color or black-and-white photograph to fit in one column should also be provided.

REVIEW PROCEDURE

Evaluation of a paper submitted to the *JOURNAL* begins with critical reading by the Managing Editor. The manuscript is then submitted to referees for critical review for scientific content, originality and clarity of presentation. To expedite review, authors should provide the names and current e-mail address of at least three reviewers within the appropriate field, with whom they have not had a collaboration in the past two years. Potential reviewers that the authors wish to avoid due to other conflicts of interest can also be provided. Attention to the preceding paragraphs will also facilitate the review process. Reviews will be returned to the author together with a judgement regarding the acceptability of the manuscript for publication in the *JOURNAL*. The authors will be requested to revise the manuscript where necessary. Time limits for submission of the manuscript and publication charges will be finalized in the accompanying letter from the Managing Editor (see "Proposed timetable for manuscript processing"). The authors will then be asked to return the revised manuscript, together with a cover letter detailing their responses to the reviewers' comments and changes made as a result. The corresponding author will be responsible for submitting the total publication cost of the paper to the Editor-in-Chief, when the revised manuscript is sent to the Managing Editor. Failure to pay the publication charges in a timely manner will prevent processing of the manuscript. If the time limits are not met, the paper will be considered withdrawn by the author. Please note that this revised manuscript will be the manuscript that will enter into the bound journal. Thus, authors should carefully read for errors and omissions so ensure accurate publication. A page charge will be billed to the author of errata. All final decisions concerning acceptance or rejection of a manuscript are made by the Managing Editor (Ivan H. Still) and/or the Editor-in-Chief (Mostafa Hemmati).

Please note that all manuscript processing, review and correspondence will be carried out electronically using e-mail. Thus, authors are requested to add the e-mail addresses of the editors (istill@atu.edu and mhemmati@atu.edu), and the *JOURNAL* email address (jarksci@yahoo.com) to their accepted senders' list to ensure that they receive all correspondence.

Reprint orders should be placed with the printer, not the Managing Editor. Information will be supplied nearer publication of the *JOURNAL* issue. The authors will be provided with an electronic copy of their manuscript after the next annual meeting.

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