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The *PROCEEDINGS* OF THE INDIANA ACADEMY OF SCIENCE is a journal dedicated to promoting scientific research and the diffusion of scientific information, to encouraging communication and cooperation among scientists, and to improving education in the sciences.

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Cover: Mudpuppies (*Necturus maculosus*) are large, wholly aquatic salamanders found throughout many of Indiana's rivers and streams. Smooth skin and large, red gills distinguish the Mudpuppy from Indiana's other large, stream-dwelling salamander, the Hellbender (*Cryptobranchus alleganiensis*). Unlike Hellbenders, however, Mudpuppies commonly inhabit both streams and lakes. They are found at substantial depths in Lake Michigan and in water less than a meter deep in the rocky streams of southern Indiana. This extreme variation in habitat use makes Mudpuppies difficult to sample for in a standardized way. As a result, much of our records for Mudpuppies come from incidental captures by fisherman. Though Mudpuppies are known from most Indiana watersheds, we know very little about their population dynamics. However, recent observations of Mudpuppy nests in southern Indiana and numerous new county records scattered throughout the state are encouraging signs. Upper left: Image of an adult mudpuppy from southeastern Indiana in a shallow, rocky stream. Upper right: Close up image of an adult Mudpuppy, showing feathery gills. Lower left: Recently deposited Mudpuppy eggs on the underside of a rock in a southeastern Indiana stream. Female mudpuppies guard their nests and offer some protection to eggs and young larvae from predatory fish and invertebrates. Lower right: The Salamander Mussel (*Simpsonaias ambigua*) is only found in streams with Mudpuppies because their larvae are obligate parasites on the gills of Mudpuppies.

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Figures 1–4.—Right chelicerae of species of *Centruroides* from Timbuktu. 1. Dorsal view; 2. Prolateral view of moveable finger; 3. *Centruroides* holotype male; 4. *Centruroides* female. Scale = 1.0 mm.

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RECENT RECORDS FOR MUDPUPPIES (*NECTURUS MACULOSUS*) IN INDIANA WITH NOTES ON PRESUMED DECLINES THROUGHOUT THE MIDWEST

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ABSTRACT. Mudpuppies (*Necturus maculosus*) have the broadest distribution of any fully aquatic salamander in North America but population trends are poorly understood. There are no demographic data for Mudpuppies in Indiana despite indications of population declines. Considering the dramatic decline of Hellbenders (*Cryptobranchus alleganiensis*) in Indiana, it is important to understand Mudpuppy population trends to ensure that similar declines are not occurring. Thirteen new county records for Mudpuppies are presented and the first published evidence of breeding in the state in almost a century. Salamander Mussel (*Simpsonaias ambigua*) records and the geographic inconsistencies between the known distributions of these two intimately-linked species are also discussed. Mudpuppies should be the subject of more extensive monitoring and conservation efforts to better understand their conservation needs.

Keywords: Mudpuppy, salamander mussel, Indiana

INTRODUCTION

Hellbender (*Cryptobranchus alleganiensis*) populations in the Midwestern United States experienced drastic declines during the mid-1900's (Wheeler et. al 2003). Some biologists (Davis et al. 1998; Minton 1998) have linked this decline with a presumed decline of Mudpuppies (*Necturus maculosus*), and Minton (2001) explicitly mentioned that Mudpuppies and Hellbenders in Indiana have declined for similar reasons. Yet Mudpuppy population dynamics remain understudied and poorly understood (Matson 2005). The most intensive study of an Indiana Mudpuppy population, now almost a century old (Evermann & Clark 1918), comes from Lake Maxinkuckee in Marshall County. This report also detailed the only published account of a Mudpuppy nest in Indiana. Most other Indiana records for Mudpuppies come from scattered museum specimens and anecdotal reports obtained from anglers who occasionally capture the salamanders by accident (Piatt 1931; Allyn & Shockley 1939; Minton 2001).

Additional insight into Mudpuppy distribution in Indiana comes from a freshwater mussel species that cannot reproduce in their absence. Mudpuppies are the only known host for Salamander Mussel (*Simpsonaias ambigua*) larvae (Howard 1915); thus you will not find *Simpsonaias* unless *Necturus* is present. The Mudpuppy and Salamander Mussel are "Species of Special Concern" in Indiana (IDNR 2013), while the Salamander Mussel is a "Lower Risk, Conservation-dependent Species" globally (IUCN 2013). There is a need for more extensive monitoring of this widespread salamander in order to better understand current population trends and management needs. A compilation of recent survey efforts and historic records is presented to better assess the distribution and current status of Mudpuppies in Indiana.

METHODS

Quantitative sampling.—Searches for Mudpuppies were conducted in streams at Big Oaks National Wildlife Refuge (BONWR) in Jennings County and Ripley County, Indiana. BONWR is a 20,234 ha refuge consisting of varied habitat ranging from grasslands and shrublands to mature forest. Numerous head-

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water streams within the upper Muscatatuck River watershed dissect the refuge flowing east to west. These streams are shallow and clear with abundant limestone cliffs, ledges, and flat rocks. Visual encounter searches were conducted for Mudpuppies during June 2012 and May–June 2013 in Otter Creek, Graham Creek, and Little Graham Creek. During these surveys, groups of 3–13 surveyors overturned large, flat, submerged rocks in search of Mudpuppies and nests. Our search method follows Matson (1998) and the timing of our surveys (May and June) corresponds to previous observations of nests (Petranka 1998; Matson 2005). Minnow traps baited with Ol' Roy canned dog food were used from 7 December 2012–12 January 2013 to trap for adult Mudpuppies in Otter Creek. Minnow traps were placed in a paired setup with a Frabill vinyl-coated minnow trap and a Promar medium minnow trap placed at each trapping location. Leaf packs, undercut banks, log jams, and large rocks were targeted when placing traps within streams.

Additional records.—We compiled records for Mudpuppies collected incidentally while sampling fish statewide during other projects of the Wildlife Diversity Program, Indiana Department of Natural Resources, and investigated reports received of Mudpuppies caught by anglers and other biologists. These were verified by photo or specimen when possible. HerpNET and museums were also searched for collection records.

RESULTS

BONWR.—109.5 person hours were spent searching for Mudpuppies during June 2012 and May–June 2013. Eleven adult Mudpuppies and four nests were observed during visual encounter surveys in Otter Creek and Little Graham Creek at BONWR. Individuals were identified based on unique markings and scars noted in photographs. We sampled Otter Creek (88.3 person hours, 1078 cover objects, 4 km covered) more extensively than Graham Creek (21.2 person hours, 370 cover objects, 2 km covered) and Little Graham Creek (5 person hours, 80 cover objects, 100 m covered). During 2012, we found five adult Mudpuppies in Otter Creek, one of which was guarding a clutch of recently hatched larvae. Larvae were dispersed enough to make counting clutch size impossible. These Mudpuppies were observed in three different stretches of Otter Creek (two sites in Jennings County and one in Ripley County) and all were at similar depths (<0.5 m), under large, flat rocks (>80 cm at widest point), and in calm clear portions of the creek.

During 2013, four adult Mudpuppies were found in two stretches of Otter Creek (Jennings County) and a single adult from Little Graham Creek (Jennings County). Two Mudpuppies from Otter Creek and one from Little Graham Creek were females guarding nests. Both Otter Creek nests were located under large, embedded, flat rocks over medium-sized cobble substrate with a single entranceway, and were within 10 m of sites where Mudpuppies were found during 2012. The nests contained 52 and 132 eggs respectively. The eggs were deposited recently as was evident by the lack of development. A live Salamander Mussel was found under both nest rock sites. The nest in Little Graham Creek was also under a large, flat rock over cobble substrate with a single entranceway and contained 63 eggs that were well developed.

Between 7 December 2012 and 12 January 2013, minnow traps were used in the same stretches of Otter Creek in which visual searches were conducted during the previous summer. Traps were placed out for a total of 158 trap nights and the number of traps out at a given time was variable. Day time water temperature (measured daily while traps were out) varied from 0.1° C–10.1° C. During this sampling, only a single Mudpuppy was captured. It was captured beneath an uprooted tree below Northwest Exit Road Bridge (Jennings County) using a Frabill vinyl-coated minnow trap submerged 0.5 m underwater in a leaf pack following a mild, rainy night.

Additional state-wide records.—Twelve additional county records were compiled by the Indiana Department of Natural Resources and are herein reported (Table 1). Most records come from specimens captured during electrofishing surveys or photo documented reports from fishermen. Jagger (2008) reported the only published county record since Minton (2001).

DISCUSSION

Mudpuppy populations have declined in Illinois (Davis et al. 1998; Mierzwa 1998), Indiana (Minton 1998, 2001), and Ohio (Davis et al. 1998), yet detailed population data are unavailable. Population trends are also unknown in Minnesota and Wisconsin where

Table 1.—Additional, unpublished Mudpuppy records collected in Indiana since Minton's 2001 publication. All reported specimens are housed in the collection of the Nongame Aquatic Biologist at the Atterbury Fish and Wildlife Area with the exception of the Steuben County record (housed at the Field Museum of Natural History). Vouched records (either photographs or preserved specimens) are noted when appropriate.

County	Waterbody	Date	Voucher	Catalog number
Cass	Deer Creek	07/25/02	Photo	NECTURUS02001
Elkhart	St. Joseph River	06/22/12	Photo	NECTURUS12002
Greene	Richland Creek	09/30/02	Specimen	BEF02253
Greene	Plummer Creek	05/27/03		BEF03015
Greene	Beech Creek	04/25/12	Photo	BEF12024
LaPorte	Little Kankakee River	10/02/13		BEF13163
LaPorte	Lake Michigan	12/15/10		NECTURUS10001
Lawrence	E. Fork White River	12/17/12	Photo	NECTURUS12001
Martin	E. Fork White River	02/11/03		BEF03004
Newton	Kankakee River	10/25/08	Photo	NECTURUS08001
Noble	Crooked Lake	05/04/09		NECTURUS09001
Steuben	Hamilton Lake	Unknown		FMNH 2838
Washington	Delaney Creek	10/07/97	Photo	NECTURUS97001

Mudpuppies are frequently collected in large quantities for biological supply companies (Casper 1998; Moriarty 1998). Mudpuppies have likely declined due to poor water quality and siltation in streams (Casper 1998; Davis et al. 1998; Minton 2001), but overharvesting (Casper 1998; Moriarty 1998) and lampricide application (Matson 1998) potentially threaten local populations. Based on the observations and opinions of numerous biologists, Mudpuppy populations have declined, but the extent of this decline is unknown.

Even with the addition of 13 new county records (Fig. 1), the distribution of Mudpuppies in Indiana has not been described fully. Recent statewide freshwater mussel surveys (Wildlife Diversity Program, Indiana Department of Natural Resources), basin surveys by other researchers (Watters 1988, 1996, 1998; Harmon 1989, 1990, 1992a, b, 1996; Cummings et al. 1991; Lewis 1991; Ecological Specialists, Inc. 1993, 1998; Anderson 1994; Commonwealth Biomonitoring 2004), and a review of museum collections indicate that there are at least 10 counties without Mudpuppy records, where the Salamander Mussel has been found (Fig. 1). Most of these mussel surveys were haphazard (Strayer & Smith 2003), thus the actual distribution of the Salamander Mussel, much like Mudpuppies, could be under-represented. The numerous streams in which Mudpuppies occur where the Salamander Mussel remains undocumented lends further credence to this idea.

Declining Hellbender populations might imply conservation threats to Mudpuppy populations, but the Salamander Mussel is clearly a better indicator of such problems. Unfortunately our understanding of Salamander Mussel distribution in Indiana is only marginally better than that of Mudpuppies. It seems that the Salamander Mussel has declined or is extirpated from numerous streams (personal observation), as evidenced by finding only weathered dead or subfossil shells in many streams (Fig. 1).

Though substantial search effort resulted in relatively few Mudpuppy captures, reproducing populations of Mudpuppies were documented in two streams, Otter and Little Graham Creeks, at BONWR. Harmon (1989) found Salamander Mussels downstream from BONWR in Big Creek and Graham Creek, indicating that Mudpuppies may remain present in these streams.

Visual encounter searches at BONWR, were time consuming and capture rates were low. This method can also disrupt nest rocks and perhaps reduce clutch survivorship. Using nest boxes, similar to those designed for Hellbenders in Missouri (Briggler & Ackerson 2012), would minimize these disturbances and allow researchers to monitor breeding success. Minnow traps are effective tools for sampling Mudpuppies (Chellman & Parish 2010) suggesting that our low capture rates were likely the result of adverse weather. This method should be more effective during milder weather.

The survey of BONWR, along with numerous incidental reports, allow us to fill in some of

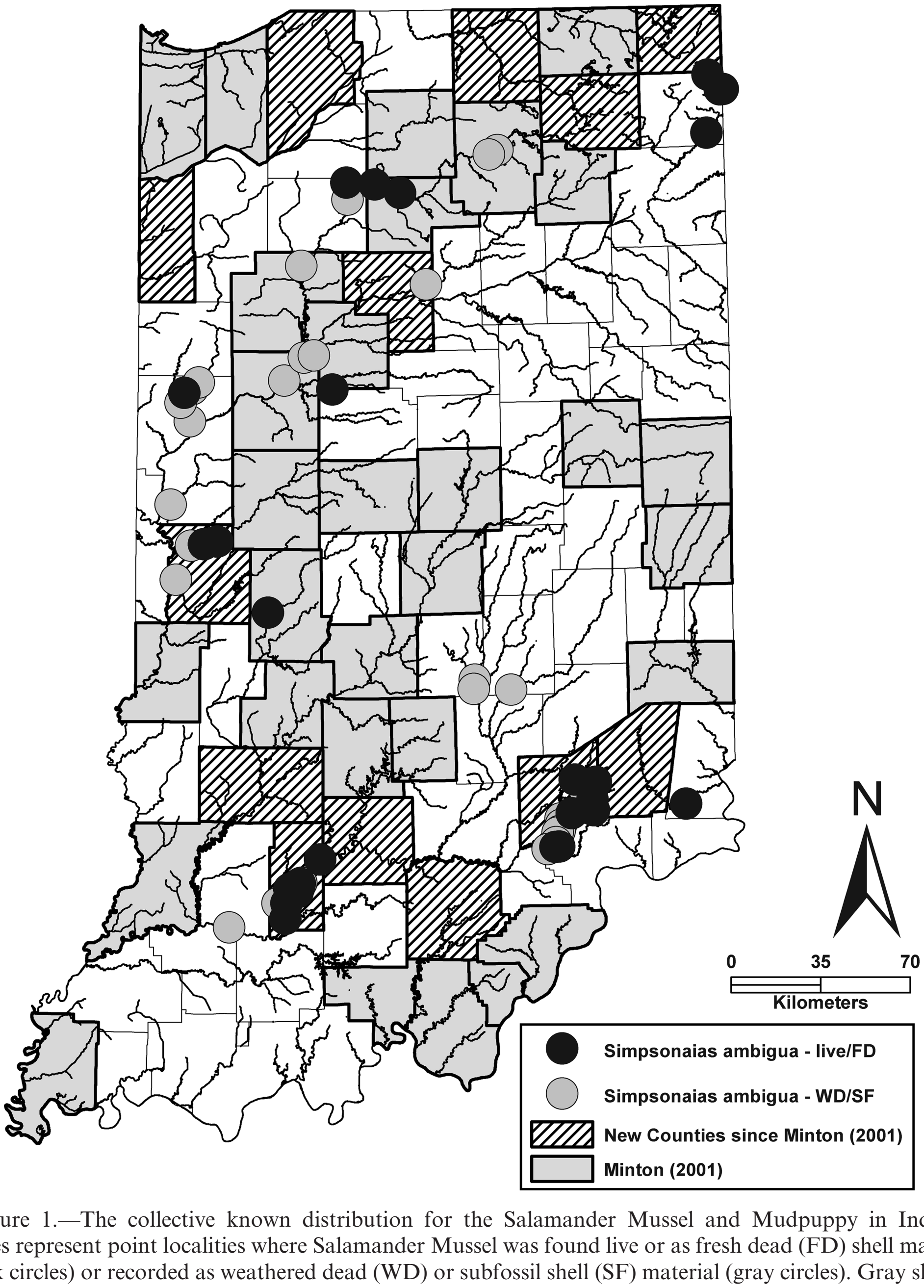

Figure 1.—The collective known distribution for the Salamander Mussel and Mudpuppy in Indiana. Circles represent point localities where Salamander Mussel was found live or as fresh dead (FD) shell material (black circles) or recorded as weathered dead (WD) or subfossil shell (SF) material (gray circles). Gray shaded counties represent Mudpuppy records reported by Minton (2001) and cross-hatched counties represent new records since 2001. A recently published Parke County record (Jagger 2008) was also included in the latter category.

the gaps in our knowledge of Mudpuppy distribution in Indiana. Furthermore, we identify the Upper Muscatatuck River watershed as a potentially important conservation area for both the Mudpuppy and Salamander Mussel.

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LITERATURE CITED

Allyn, W.P. & C.H. Shockley. 1939. A preliminary survey of the surviving species of Caudata of Vigo County and vicinity. Proceedings of the Indiana Academy of Science 48:238–243.

Anderson, R.M. 1994. Mussel survey of Jefferson Proving Grounds section of Otter Creek. DNR Memorandum, Indianapolis, Indiana.

Briggler, J.T. & J.R. Ackerson. 2012. Construction and use of artificial shelters to supplement habitat for Hellbenders (*Cryptobranchus alleganiensis*). Herpetological Review 43:412–416.

Casper, G.S. 1998. Review of the status of Wisconsin amphibians. Pp. 199–205. *In* Status and Conservation of Midwestern Amphibians (M.J. Lannoo, Ed.). University of Iowa Press, Iowa City, Iowa.

Chellman, I.C. & D.L. Parrish. 2010. Developing methods for sampling Mudpuppies in Vermont tributaries of Lake Champlain. Final Report, State Wildlife Grants Program. Vermont Fish and Wildlife, Waterbury, Vermont.

Commonwealth Biomonitoring. 2004. Bioassessment in the Tippecanoe River watershed. 2003 and 2004. Final Report for The Nature Conservancy-Indiana Chapter, Indianapolis, Indiana.

Cummings, K.S., C.A. Mayer & L.M. Page. 1991. Survey of the freshwater mussels (Mollusca: Unionidae) of the Wabash River drainage. Phase III: White River and selected tributaries. Final Report to the Nongame and Endangered Wildlife Program, Division of Fish & Wildlife, IDNR, Indianapolis, Indiana.

Davis, J.G., P.J. Krusling & J.W. Ferner. 1998. Status of amphibian populations in Hamilton County, Ohio. Pp. 155–165. *In* Status and Conservation of Midwestern Amphibians (M.J. Lannoo, Ed.). University of Iowa Press, Iowa City, Iowa.

Ecological Specialists, Inc. 1993. Mussel habitat suitability and impact analysis of the Tippecanoe River. Final Report to the Nongame and Endangered Wildlife Program, Division of Fish & Wildlife, IDNR, Indianapolis, Indiana.

Ecological Specialists, Inc. 1998. Unionid survey upstream and downstream of 16 point sources in the Tippecanoe River. Final Report to the U.S. Fish and Wildlife Service, Bloomington Field Office, Bloomington, Indiana.

Evermann, B.W. & H.W. Clark. 1918. The turtles and batrachians of the Lake Maxinkuckee region. Proceedings of the Indiana Academy of Science 26:472–518.

Harmon, J.L. 1989. Freshwater bivalve mollusks (Bivalvia: Unionidae) of Graham Creek, a small southeastern Indiana stream. Malacology Data Net 2:113–121.

Harmon, J.L. 1990. Survey of the freshwater mussels (Bivalvia: Unionidae) of Sugar Creek, East Fork White River drainage, in central Indiana. Final Report to the Nongame and Endangered Wildlife Program, Division of Fish & Wildlife, IDNR, Indianapolis, Indiana.

Harmon, J.L. 1992a. Survey of the freshwater mussels (Bivalvia: Unionidae) of the Vernon Fork of the Muscatatuck River with emphasis on the impact area of proposed structure 42, a water supply reservoir. Final Report to the Nongame and Endangered Wildlife Program, Division of Fish & Wildlife, IDNR, Indianapolis, Indiana.

Harmon, J.L. 1992b. Survey of the freshwater mussels (Bivalvia: Unionidae) of Big Creek and quantitative evaluation of state listed mussel species in Big and Graham Creeks. Final Report to the Nongame and Endangered Wildlife Program, Division of Fish & Wildlife, IDNR, Indianapolis, Indiana.

Harmon, J.L. 1996. Survey of the freshwater mussels (Bivalvia: Unionidae) of the Muscatatuck National Wildlife Refuge. Final Report to the U.S. Fish and Wildlife Service, Muscatatuck National Wildlife Refuge, Seymour, Indiana.

Howard, A.D. 1915. Some exceptional cases of breeding among the Unionidae. The Nautilus 29:4–11.

IDNR (Indiana Department of Natural Resources). 2013. Indiana's State Endangered species. At http://www.in.gov/dnr/fishwild/files/fw-Endangered_Species_List.pdf. Accessed on 14 November 2013.

IUCN (International Union for Conservation of Nature). 2013. The IUCN Red List of Threatened Species. Version 2013.1. At http://www.iucnredlist.org. Accessed on 14 November 2013.

Jagger, F.B. 2008. *Necturus maculosus*. Mudpuppy. Geographic distribution. Herpetological Review 39:477.

Lewis, R.B. 1991. Freshwater mussel (Mollusca: Unionidae) survey of Sugar Creek in Parke, Montgomery, Boone, and Clinton Counties of Indiana. Final Report to the Nongame and Endangered Wildlife Program, Division of Fish & Wildlife, IDNR, Indianapolis, Indiana.

Matson, T.O. 1998. Evidence for home ranges in Mudpuppies and implications for impacts due to episodic applications of Lampricide TFM. Pp. 278–287. *In* Status and Conservation of Midwestern Amphibians (M.J. Lannoo, Ed.). University of Iowa Press, Iowa City, Iowa.

Matson, T.O. 2005. *Necturus maculosus* (Rafinesque, 1818) Mudpuppy. Pp. 870–871. *In* Amphibian Declines: The Conservation Status of United States Species (M.J. Lannoo, Ed.). University of California Press, Berkeley and Los Angeles, California.

Mierzwa, K.S. 1998. Status of northeastern Illinois amphibians. Pp. 115–124. *In* Status and Conservation of Midwestern Amphibians (M.J. Lannoo, Ed.). University of Iowa Press, Iowa City, Iowa.

Minton, S.A. 1998. Observations on Indiana amphibian populations: A forty-five-year overview. Pp. 217–220. *In* Status and Conservation of Midwestern Amphibians (M.J. Lannoo, Ed.). University of Iowa Press, Iowa City, Iowa.

Minton, S.A. 2001. Amphibians and Reptiles of Indiana. Indiana Academy of Science, Indianapolis, Indiana. 404 pp.

Moriarty, J.J. 1998. Status of amphibian in Minnesota. Pp. 166–168. *In* Status and Conservation of Midwestern Amphibians (M.J. Lannoo, Ed.). University of Iowa Press, Iowa City, Iowa.

Petranka, J.W. 1998. Salamanders of the United States and Canada. Smithsonian Institute Press, Washington D.C. 587 pp.

Piatt, J. 1931. Herpetological report of Morgan County, Indiana. Proceedings of the Indiana Academy of Science 40:361–368.

Strayer, D.L. & D.G. Smith. 2003. A Guide to Sampling Freshwater Mussel Populations. American Fisheries Society, Bethesda, Maryland. 103 pp.

Watters, G.T. 1988. A survey of the freshwater mussels of the St. Joseph River system, with emphasis on the federally endangered white cat's paw pearly mussel. Final Report to the Nongame and Endangered Wildlife Program, Division of Fish & Wildlife, IDNR, Indianapolis, Indiana.

Watters, G.T. 1996. 1996 survey the mussels of the Fish Creek drainage. Final Report to the Nature Conservancy-Indiana Chapter, Indianapolis, Indiana.

Watters, G.T. 1998. Freshwater mussel survey of the St. Joseph River drainage, exclusive of Fish Creek. Final Report to The Nature Conservancy-Indiana Chapter, Indianapolis, Indiana.

Wheeler, B.A., E. Prosen, A. Mathis & R.F. Wilkinson. 2003. Population declines of a long-lived salamander: A 20+-year study of Hellbenders, *Cryptobranchus alleganiensis*. Biological Conservation 109:151–156.

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TREE-RING ANALYSIS IN INDIANA WITH SPECIAL ATTENTION TO DENDROARCHAEOLOGY

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ABSTRACT. Although tree-ring studies have been performed in Indiana since the 1930s, their study in the state has been sporadic, and many gaps in our current understanding of tree growth and forest dynamics exist. Most notably, very little information regarding tree growth prior to the nineteenth century has been made available in the state. The tree rings in timbers of historically erected buildings can be used to build centuries-long chronologies for Indiana and the Mid-Ohio River Valley. Through tree-ring analysis, especially of nineteenth century buildings found throughout the region, study of past growth patterns and dynamics of the old-growth forest that once covered the state is possible.

Keywords: Tree rings, dendrochronology, dendroarchaeology

INTRODUCTION

Tree rings, the annual increments of wood that are deposited around the circumference of woody plants each year, offer a unique opportunity to study growth patterns in trees and forests over extended time periods. Dendrochronology is the science of assigning accurate calendar dates to individual tree rings and using these accurately dated rings to interpret past influences on tree growth. Dendrochronological techniques have been used to examine the influence of various factors on growth rates. For instance, dendrochronological methods have been used to study a wide range of topics including forest fire history, insect outbreaks, ecological phenomena (e.g., disturbance events such as wind storms), and climatic influences on tree growth (Fritts & Swetnam 1989; Schweingruber 1989).

The width of individual tree rings varies from year to year. Tree-ring width in a given year is governed by a suite of biotic and abiotic factors with larger rings being formed during years of favorable growing season conditions and smaller rings in less favorable years. Factors affecting individual tree-ring widths include, but are not limited to, age-related growth trends, climate, and disturbances which originate from within or outside a forested stand. The size of a tree ring is, therefore, an aggregate response to the conditions experienced by a tree in a given year (Cook 1987). The variation in ring width permits dendrochronologists to study long-term growth rates and tease out the influence of different variables on tree growth (e.g., how droughts of varying degrees of severity affect growth in given years).

Additionally, variation in tree-ring widths allows for samples to be crossdated. Crossdating is the process of matching the pattern of small and large tree rings among numerous trees throughout a particular forested stand or geographic region (e.g., Stokes & Smiley 1968; Fritts 1976). Consistent crossdating among tree rings is essential for ensuring that the exact calendar date is assigned to individual tree rings. In Indiana, drought events result in decreases in radial growth rates with the size of individual rings decreasing as stress during a particular year increases. Reliably dated chronologies (i.e., compilation of accurately dated and measured tree rings from a number of trees) can, therefore, be created in a given region. For example, common crossdating signals in southeastern Indiana oak (*Quercus* L.) trees includes very small tree rings (in relation to neighboring tree rings) in 1930, 1936, 1944, and 1984 and smaller than normal rings in 1954, 1970, 1988, 1994, and 2000 (Fig. 1). If the pattern of rings in a putatively dated tree-ring sample crossdates or matches this pattern, the sample is considered to be dated accurately and that the exact year of formation of each tree ring is known. Since droughts affect fairly large areas simultaneously, relatively homogeneous signals throughout an area are created, and crossdating is possible.

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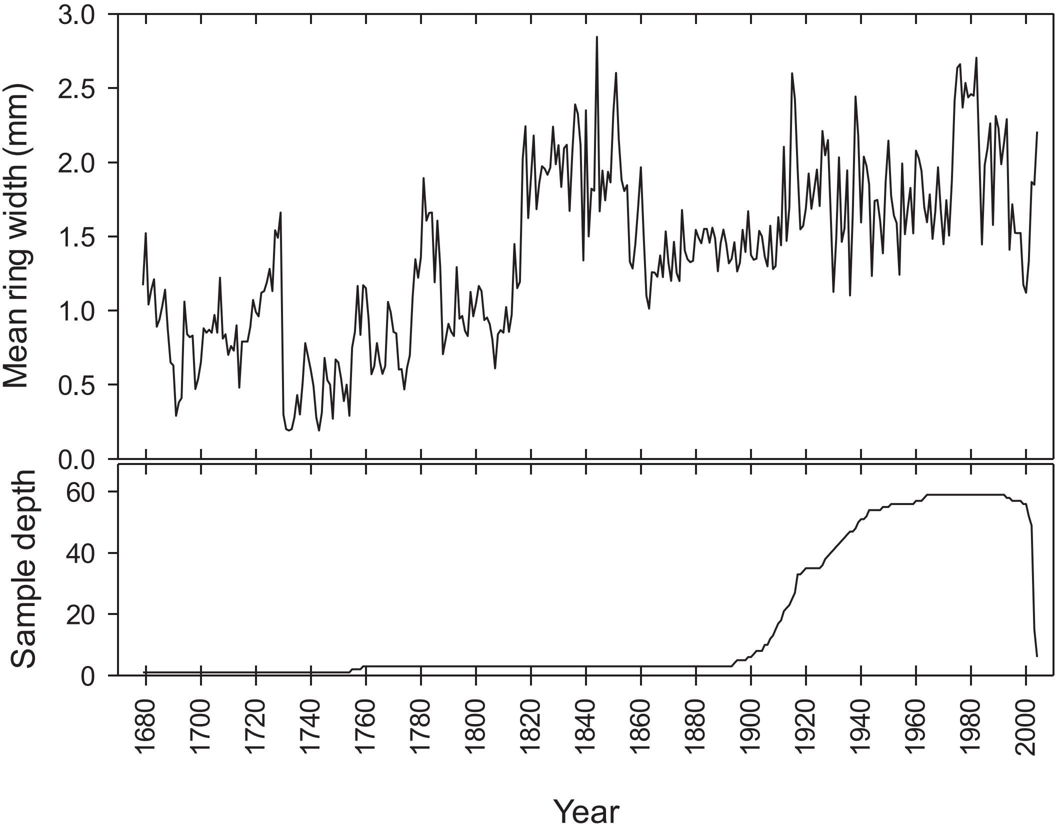

Figure 1.—Mean tree-ring chronology for white oaks (*Quercus* subgenus *Lepidobalanus*) growing in Happy Valley, a forested ravine of Hanover College (Jefferson County, Indiana). Sample depth refers to the number of tree rings dated and measured for a particular year.

Dendrochronology in Indiana.—Tree-ring studies, designed to answer very different questions, have been performed sporadically in Indiana since the 1930s. For example, Edwin Lincoln Moseley identified the pattern of wide and narrow tree rings in oaks throughout several Midwestern states, including Indiana, in the 1930s. Moseley was interested in historic climate cycles/patterns and hoped to create a method of predicting future droughts, Ohio River floods, and lake levels (Moseley 1939; Stuckey 1998). Diller (1935) and Friesner & Friesner (1941) studied the impact of climatic variables including temperature and precipitation on tree growth in the state in the northern half of Indiana and Marshall County, respectively. In 1934, Florence Hawley Senter who was serving as the Director of the Laboratory of Dendrochronology at the University of Chicago (Nash 1999) studied trees from Clinton, Crawford, Knox, Marion, Noble, Owen, and Parke Counties (Hawley 1941). She was working to determine if dendrochronological analysis would be possible in the Midwest United States. If so, chronologies would be created in the hopes of dating Native American ruins in the region. As an example of the different groups interested in tree-ring sciences, the work performed by Hawley and the lab in Chicago was funded by the Indiana Historical Society (Hawley 1941).

More recent studies have focused on stress leading to oak mortality (Pederson 1998); interactive effects of acidic deposition, drought, and insect attack on various oak species (LeBlanc 1998); impact of early season water balance on white oak (*Quercus alba* L.) growth (LeBlanc & Terrell 2001); impact of soil texture on tree growth (Charton & Harman 1973); and the effects of insect outbreaks (Speer et al. 2010) on tree growth. Cook produced an oak chronology for Pulaski Woods in north central

Indiana (International Tree-Ring Data Bank 2013) while creating a continental-wide tree-ring database.

Most tree-ring studies in the state have focused on oaks (*Quercus* L. spp.) especially those in the "white oak" subgenus [*Quercus* subgenus *Lepidobalanus* including white oak (*Q. alba*), chestnut oak (*Q. prinus* L.), or post oak (*Q. stellata* Wangenh.)]. Species in the white oak group are often chosen for dendrochronological analysis in eastern North America due to their reliable crossdating and consistent response to climatic variables such as temperature, precipitation, and drought indices (e.g., Wedel & Hawley 1941; Sheppard et al. 1988; Rubino & McCarthy 2000). In Indiana, studies on other species have been performed to a lesser degree (Diller 1935; Friesner & Friesner 1941; LeBlanc 1998; Pederson 1998; Sparks & Bishop 2009; Speer et al. 2010). These species include American beech (*Fagus grandifolia* Ehrh.), bitternut hickory (*Carya cordiformis* (Wangenh.) K. Koch), sassafras (*Sassafras albidum* (Nutt.) Nees.), sugar maple (*Acer saccharum* Marshall), tulip poplar (*Liriodendron tulipifera* L.), white ash (*Fraxinus americana* L.), and various oaks in the red/black group (*Quercus* subgenus *Erythrobalanus*) including black (*Q. velutina* Lam.), northern red (*Q. rubra* L.), and pin oak (*Q. palustris* Muenchh.). Taxonomy and nomenclature follow Gleason & Cronquist (1991). The above is not intended to be a thorough historical review of all dendrochronological studies performed in the state but rather a representation of the various types of work that have been done.

Despite the long use of dendrochronology to explore tree growth in the state, many gaps in our knowledge persist. For example, tulip poplar, an important species in many forest types of Indiana, has had only very limited study; from 1930 to 1939 Friesner & Friesner (1941) studied tulip poplar growth in Marshall County in relation to climate. Limited analysis of other widely distributed species such as ash, both white and green (*Fraxinus pennsylvanica* Marshall), has been reported; for example, Speer et al. (2010) created a 74 year-long white ash chronology using 18 trees. Similar voids in dendrochronological study exist for other common trees such as sweet gum (*Liquidambar styraciflua* L.), sugar maple, red maple (*Acer rubrum* L.), beech, elm (*Ulmus* L. spp.), black walnut (*Juglans nigra* L.), and various hickory species (*Carya* Nutt. spp.). A better understanding of forest growth and dynamics will surely require new or additional studies of these and other common species found throughout the state.

Dendroarchaeology in Indiana.—Using standard dendrochronological techniques, the dating of tree rings in living trees is relatively straightforward. Since the year of sampling is known, calendar dates can be given to individual tree rings starting with assignment of the current year to the outermost ring (if the growing season for that year has begun) and assigning the corresponding previous year to each ring until the pith or center of the sample is reached. If the sample crossdates with other samples, confident dates can be given to the individual tree rings in the sample, and it can be included in a regional chronology.

However, date assignment is not always this straightforward. For example, if a sample is obtained from a timber of a structure with an unknown construction date (which is almost always the case), assignment of the dates in which the rings were formed is more complicated. Dating of such a sample could be performed using dendroarchaeological techniques. Dendroarchaeology is a sub-field of dendrochronology that deals specifically with the sampling of historically erected buildings and other wooden objects to tap the tree-ring information found within their timbers. In dendroarchaeological studies, the formation date of an individual tree ring is unknown. The date, however, can be determined by crossdating the pattern of small and large rings in a sample of unknown age with accurately dated chronologies that have been prepared from the same geographic region (Fig. 2).

Crossdating is a highly reliable method for dating wood of unknown age, and dendroarchaeological techniques have proven to be powerful and effective research tools, especially in dry climates such as the American Southwest (Nash 1999). Crossdating and dendroarchaeological techniques also have been used throughout the world in such places as the Mediterranean [work of P.I. Kuniholm's Cornell Lab; Manning & Bruce (2009)], Canada (e.g., Robichaud & Laroque 2008), and Japan (Hoshino et al. 2008), to name a few. Despite the mesic climate of the eastern United States, dendroarchaeological analysis has been successfully performed

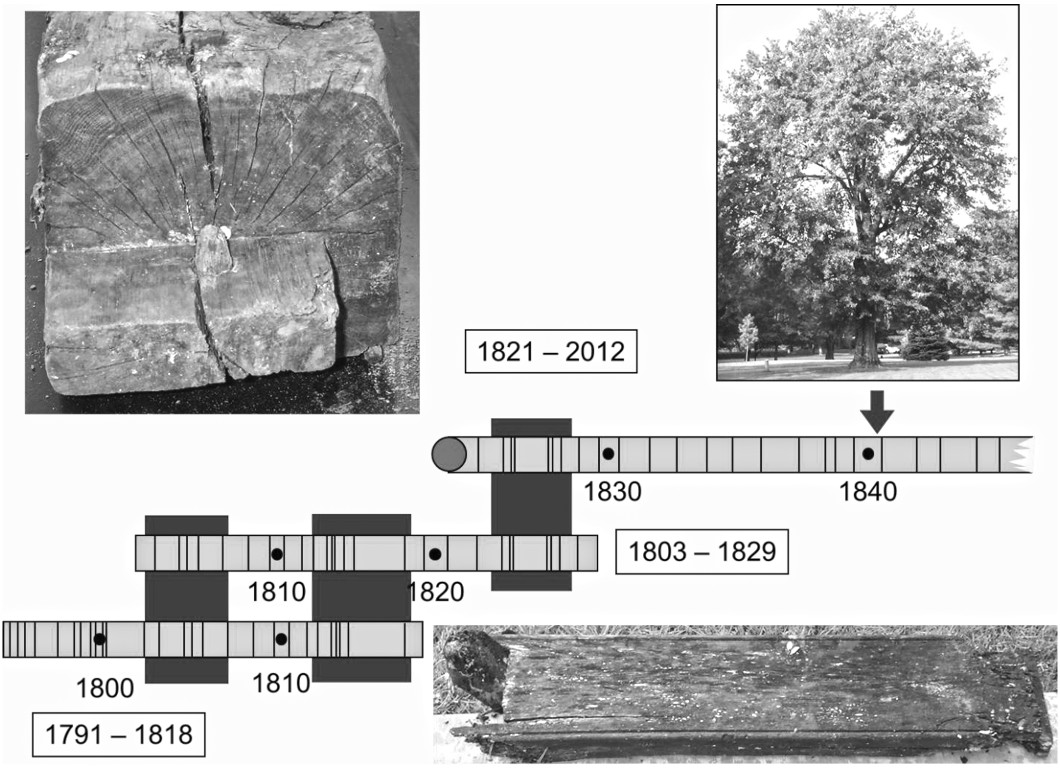

Figure 2.—Dating samples of unknown age (beam and floor joist in this example) are performed by crossdating the tree-ring patterns in the samples with chronologies of dated tree rings. The shaded areas represent unique growth patterns that make pattern matching and crossdating possible. Note: the sample lengths used in this illustration are much shorter than those that are needed to perform an actual analysis.

throughout the region including Michigan (Harley et al. 2011), Georgia (Wight & Grissino-Mayer 2004), Florida (Grissino-Mayer et al. 2010), Virginia (Bortolot et al. 2001), Arkansas (Stahle 1979), and the Southeast [H.D. Grissino-Mayer's University of Tennessee-Knoxville Lab; Grissino-Mayer (2009)].

With European colonization and subsequent settlement of Indiana, few forests were spared the axe. Consequently, the archetypical "virgin" or "old-growth" forested ecosystem is a true rarity in Indiana. The loss of the original forests witnessed by early explorers and settlers was already lamented by Hoosier scientists as early as 1895. A.W. Butler, in his presidential address to the Indiana Academy of Science (Butler 1896) remarked on the loss of "tall trees" and "heavy timber" especially in southern portions of the state. This lack of old trees was also noted by Hawley (1941) in her early work in the Midwest and Indiana in the 1930s (Senter 1938a, b). As a result, most of the chronologies created for the state are relatively short in length, with few extending into and prior to the nineteenth century. Undoubtedly, this problem has been further exasperated in recent decades as many of the remaining old trees have either died or have been cut down.

Studying past forest growth patterns and dynamics, however, is not impossible. Indiana's rich natural (and cultural) past are preserved in the historically erected buildings found throughout the state. Early construction utilized timber standing on site—trees were felled and incorporated into buildings as beams, rafters, floor boards, joists, and braces (Senter 1938a; Hutslar 1992; Roberts 1996). A plethora of nineteenth century Hoosier barns, churches, agricultural outbuildings, homes, and mills can be found in nearly all parts of the state. In these structures we find remnants of the vast, uninterrupted forest seen by the earliest of European settlers and explorers. The tree-ring patterns in these timbers offer a truly unique opportunity to examine the old-growth forests of the state.

Using dendroarchaeological techniques and crossdating structural timbers, one is able to obtain much information from the resulting chronology of accurately dated tree rings. For example, one can determine and/or verify the construction date or modification of buildings (e.g., Stahle 1979; Bortolot et al. 2001; Wight & Grissino-Mayer 2004; Grissino-Mayer et al. 2010; Harley et al. 2011). Additionally, after successfully crossdating a specimen, one is able to investigate plant-environment interactions such as the influence of climate, disturbance events, and human impacts on tree and forest growth by studying long- and short-term growth patterns of previous centuries. Accurately dated tree rings can also prove invaluable as proxy data for studying and recreating climatic regimes of the state and region.

Creation of Hoosier Chronologies.—Despite the loss of old trees from throughout the region, creation of centuries-long, quality tree-ring chronologies is feasible. Over the past decade, the author has developed chronologies from living and recently killed trees. Chronologies from chinquapin oak, (*Quercus muehlenbergii* Engelm.), American beech, tulip poplar, ash, elm, and hickory have been created in southern Indiana. Also during this time the author has sampled scores of nineteenth and early twentieth century buildings throughout the state and the Mid-Ohio River Valley. After a decade of collecting, analyzing, and crossdating hundreds of timbers and tree samples from throughout the region, the creation of replicated and reliably dated multiple centuries-long chronologies for several major forest species has been achieved. These chronologies have been formed by combining tree-ring series from living trees, downed logs, and timbers of historic buildings. In the face of global climate change and various perturbations such as acid deposition and introduction of invasive plants and pests (e.g., emerald ash borer) to local forests, creating such chronologies may be of great importance when investigating past forest growth conditions and responses.

Dendroarchaeological analysis in Indiana is possible and has a great potential to expand tree-ring chronologies in a greatly understudied region of the United States (Senter 1938a, b). Locating potential structures for analysis has been achieved by delivering lectures to civic groups, historical societies, and Historic Landmarks Foundation of Indiana's BarnAgain conferences. Property managers and owners attending these lectures have been pivotal in providing permission or contacts to sample barns, houses, smokehouses, and other types of buildings in the state. Managers are very interested in verifying construction dates of the properties they steward; there is no shortage of potential buildings to date and analyze (Senter 1938a). Dendroarchaeological analysis offers an objective way to date historically erected structures when other lines of evidence (e.g., deed and tax records or oral history) are nonexistent or unreliable.

The goal of this article is to provide a general introduction to both the tree-ring studies that have been performed in Indiana and to the dendroarchaeological analyses that the author has performed in the region over the past decade. Additionally, the author hopes to show the great potential of dendroarchaeology in the Midwest by continuing, 80 years later, the seminal work Florence M. Hawley Senter began in the 1930s when she dated timber from an old Indiana cabin (Senter 1938b). The *Proceedings of the Indiana Academy of Science* is an ideal venue to present the Hoosier history (natural and cultural) that the author has studied and will study using tree-ring analyses. The findings of a dendroarchaeological analysis of a log structure from New Harmony, Indiana are included in this volume of the *Proceedings*. These results represent the first modern contribution of dendroarchaeological analysis to the *Proceedings*. At one time, dendroarchaeology was considered "the greatest single contribution ever made to American archaeology" (Haury 1935). In temperate regions such as Indiana, tree-ring dating of prehistoric archaeological wood objects will likely not be possible because of poor wood preservation. On the other hand, chronologies reaching as early as the 1400s have been created through the study of historic buildings. Dendroarchaeology truly has the potential to enable the study of the once unknown past.

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LITERATURE CITED

Bortolot, Z.J., C.A. Copenheaver, R.L. Longe & J.A.N. Van Aardt. 2001. Development of a white oak chronology using live trees and a post-civil war cabin in south-central Virginia. Tree-Ring Research 57:197–203.

Butler, A.W. 1896. A century of changes in the aspects of Nature. Proceedings of the Indiana Academy of Science 5:3–42.

Charton, F.L. & J.R. Harman. 1973. Dendrochronology in northwestern Indiana. Annals of the Association of the American Geographers 63: 302–311.

Cook, E.R. 1987. The decomposition of tree-ring series for environmental studies. Tree-Ring Bulletin 47:37–59.

Diller, O.D. 1935. The relation of temperature and precipitation to the growth of beech in northern Indiana. Ecology 16:72–81.

Friesner, R.C. & G.M. Friesner. 1941. Abstract. Relation of annular ring formation to rainfall as illustrated in six species of trees in Marshall County, Indiana. Proceedings of the Indiana Academy of Science 50:57–58.

Fritts, H.C. 1976. Tree Rings and Climate. Academic Press, New York, New York. 567 pp.

Fritts, H.C. & T.W. Swetnam. 1989. Dendroecology: a tool for evaluation variations in past and present forest environments. Advances in Ecological Research 19:111–188.

Gleason, H.A. & A. Cronquist. 1991. Manual of Vascular Plants of Northeastern United States and Adjacent Canada, 2nd ed. New York Botanical Garden, Bronx, New York. 910 pp.

Grissino-Mayer, H.D. 2009. An introduction to dendroarchaeology in the southeastern United States. Tree-Ring Research 65:5–10.

Grissino-Mayer, H.D., L.N. Kobziar, G.L. Harley, K.P. Russell, L.B. LaForest & J.K. Oppermann. 2010. The historical dendroarchaeology of the Ximénex-Fatio House, St. Augustine, Florida, U.S.A. Tree-Ring Research 66:61–73.

Harley, G.L., H.D. Grissino-Mayer, L.B. LaForest & P. McCauley. 2011. Dendrochronological dating of the Lund-Spathelf House, Ann Arbor, Michigan, USA. Tree-Ring Research 67:117–121.

Haury, E.W. 1935. Tree rings—the archaeologist's time piece. American Antiquity 1:98–108.

Hawley, F. 1941. Tree-Ring Analysis and Dating in the Mississippi Drainage. Occasional Papers, No. 2. The University of Chicago Press, Chicago, Illinois. 110 pp.

Hoshino, Y., T. Okochi & T. Mitsutani. Dendrochronological dating of vernacular folk crafts in northern central Japan. Tree-Ring Research 64:109–114.

Hutslar, D.A. 1992. Log Construction in the Ohio Country, 1750–1850. Ohio University Press, Athens, Ohio. 265 pp.

International Tree-Ring Data Bank. 2013. Contributors of the International Tree-Ring Data Bank. IGBP PAGES/World Data Center for Paleoclimatology, NOAA/NGDC Paleoclimatology Program, Boulder, Colorado. At: http: www.ngdc.noaa.gov/paleo/ftp-treering.html.

LeBlanc, D.C. 1998. Interactive effects of acidic deposition, drought, and insect attack on oak populations in the midwestern United States. Canadian Journal of Forest Research 28:1184–1197.

LeBlanc, D. & M. Terrell. 2001. Dendroclimatic analyses using Thornthwaite-Mather-type evapotranspiration models: a bridge between dendroecology and forest simulation models. Tree-Ring Research 57:55–66.

Manning, S.W. & M.J. Bruce. 2009. Tree-rings, Kings, and Old World Archaeology and Environment: Papers Presented in Honor of Peter Ian Kuniholm. Oxbow Books, Oxford, UK. 332 pp.

Moseley, E.L. 1939. Long time forecasts of Ohio River floods. Ohio Journal of Science 39:220–231.

Nash, S.E. 1999. Time, Trees, and Prehistory: Tree-ring Dating and Development of North American Archaeology, 1914 to 1950. University of Utah Press, Salt Lake City, Utah. 294 pp.

Pederson, B.S. 1998. The role of stress in the mortality of Midwestern oaks as indicated by growth prior to death. Ecology 79:79–93.

Robichaud, A. & C.P. Laroque. 2008. Dendroarchaeology in southwestern Nova Scotia and the construction of a regional red spruce chronology. Tree-Ring Research 64:17–25.

Roberts, W.E. 1996. Log Buildings of Southern Indiana. Trickster Press, Bloomington, Indiana. 228 pp.

Rubino, D.L. & B.C. McCarthy. 2000. Dendroclimatological analysis of white oak (*Quercus alba* L., Fagaceae) from an old-growth forest of southeastern Ohio, USA. Journal of the Torrey Botanical Society 127:240–250.

Schweingruber, F.H. 1989. Tree Rings: Basics and Applications of Dendrochronology. Kluwer Academic Publishers, Dordrecht, Holland. 276 pp.

Senter, F.H. 1938a. Can we fix prehistoric dates in the Middle West by tree rings? Indiana History Bulletin 15:118–128.

Senter, F.H. 1938b. Dendrochronology in two Mississippi drainage tree-ring areas. Tree-Ring Bulletin 5:3–6.

Sheppard, P.R., E.R. Cook & G.C. Jacoby. 1988. Dendrochronology in national parks: a Mammoth Cave case study. Park Science 9:5–6.

Sparks, J.K.F. & G.I. Bishop. 2009. Evaluation of S*assafras albidum* for dendrochronology. Tree-Ring Research 65:157–161.

Speer, J.H., K. Clay, G. Bishop & M. Creech. 2010. The effect of periodical cicadas on growth of five tree species in Midwestern deciduous forests. American Midland Naturalist 164:173–186.

Stahle, D.W. 1979. Tree-ring dating of historic buildings in Arkansas. Tree-ring Bulletin 39: 1–28.

Stokes, M.A. & T.L. Smiley. 1968. An Introduction to Tree-Ring Dating. University of Chicago Press, Chicago, Illinois. 73 pp.

Stuckey, R.L. 1998. Forecasting long-range weather conditions. Pp. 127–136. *In* Edwin Lincoln Moseley (1865–1948): Naturalist, Scientist, Educator. (R.E. Niederhofer & R.L. Stuckey, Eds.). RLS Creations, Columbus, Ohio.

Wedel, M.M. & F. Hawley. 1941. Reflection of precipitation and temperature in tree growth. Pp. 45–49. *In* Tree-Ring Analysis and Dating in the Mississippi Drainage (F. Hawley, Ed.). Occasional Papers, No. 2. The University of Chicago Press, Chicago, Illinois.

Wight, G.D. & H.D. Grissino-Mayer. 2004. Dendrochronological dating of an Antebellum Period house, Forsyth County, Georgia, U.S.A. Tree-Ring Research 60:91–99.

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USING TREE-RING GROWTH PATTERNS TO DATE THE CONSTRUCTION OF A NINETEENTH CENTURY DOGTROT HOUSE IN POSEY COUNTY, INDIANA

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ABSTRACT. Dendroarchaeology is a sub-field of dendrochronology (tree-ring science) that deals with the sampling of historically constructed buildings to tap the tree-ring information found within their timbers. Dendroarchaeological studies provide an accurate and reliable means of determining the construction date of a building through a process called crossdating (matching the pattern of small and large tree rings in samples with unknown dates to samples with rings of known age). Crossdating is a highly reliable method for dating wood of unknown age, and dendroarchaeological techniques have proven to be powerful and effective research tools. The goal of this investigation was to provide a possible construction date for the Grayson dogtrot house located in a museum setting in New Harmony, Indiana. Dogtrots are a type of folk housing popular throughout the southern United States, but rare in Indiana. Tree-ring analysis of the tulip poplar timbers in the house suggests that it was built after the initiation of the 1852 growing season, and the results of this study will be used in the interpretation of this unique architectural resource.

Keywords: Dendrochronology, dendroarchaeology, tree-rings, tulip poplar, vernacular architecture

INTRODUCTION

Dendrochronology is the study of tree rings that have been dated to their precise year of formation. Analysis of the growth patterns observed in tree-ring chronologies (series of accurately measured and dated tree rings and their widths) can be extremely informative when exploring a wide variety of phenomena. Dendrochronological techniques have been used, for example, to study the timing of insect outbreaks, forest stand dynamics, forest fires, and the influence of climate on tree growth. Dendrochronological investigations are not limited to the study of tree rings found solely in living trees. Dendroarchaeology, a subfield of dendrochronology, focuses on the study and analysis of tree rings found in the timbers of historically constructed buildings and artifacts.

The tree-ring record preserved in the timbers of historically constructed buildings provides a unique opportunity to study historic tree growth (Stahle 1979; Therrell 2000). Through the accurate dating of the tree rings found in such timbers, dendroarchaeologists are able to create extended tree-ring chronologies that can be used for a variety of studies and purposes. For example, tree rings obtained from historically constructed buildings have been used for reconstructing and studying climate (e.g., precipitation, drought severity, and temperature) for periods lacking instrumental records, studying local human impacts (e.g., settlement), determining the construction date of historic buildings, and managing historic properties (e.g., Stahle 1979; Therrell 2000; Bortolot et al. 2001; Towner et al. 2001; Thun 2005).

European colonization and the subsequent clearing of forests in eastern North America have greatly hampered the creation of long-term tree-ring chronologies using living trees (Stahle 1979). However, by using the tree-ring patterns found in the timbers of historically constructed buildings, which often contain old-growth timber, it is possible to reach farther back in time than with methods using only living trees (Senter 1938; Stahle 1979). Therefore, the use of historic buildings is essential for the creation of informative, long-term regional tree-ring chronologies. Such chronologies can be useful for studying past tree and forest growth patterns. Please see Rubino (2014, in this issue) for a detailed description of dendro-

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chronological and dendroarchaeological analysis in Indiana.

Grayson House.—The Grayson dogtrot house is located near the intersection of North and West Streets in the historic town of New Harmony, Indiana. The village was founded on the Wabash River by German Harmonists in 1814 and later became a short-lived utopian experiment when industrialist Robert Owen purchased the town in 1825. The house is displayed with a collection of other log structures that were moved to the museum location. Unfortunately, very little is known about the house. An examination of building notes and newspaper articles located in the New Harmony offices of the Indiana State Museum and Historic Sites suggest that the house has no historic connection to the town's founding. It was moved from a location on what is now Indiana Highway 68 in the 1960s to be used as a pottery studio by the University of Evansville. It was purchased and restored in 1977 by Historic New Harmony, a partnership between the University of Southern Indiana and the Indiana State Museum and Historic Sites (Indiana State Museums and Historic Sites 2013). Interpretive signage at the cabin identifies it as the Macluria Double Log Cabin with a 1775 construction date. However, State Historic Site files attribute the cabin to the Grayson family. Unfortunately, neither of these family names could be located in historic census data or plat maps for Harmony and Robb Townships (the location of Indiana Highway 68 between New Harmony and Poseyville). We refer to it here as the Grayson House to be consistent with the site's interpretive materials. A construction date of 1775 was attributed to the house. Based on the region's settlement patterns, this date seemed much too early, thereby making the house a worthy candidate for dendroarchaeological study.

The Grayson House is a single story structure comprised of an open, central hallway flanked by two rectangular rooms. Chimneys are located at each end, and a common roof covers the entire structure (Figs. 1–3). The wide hallway runs the depth of the house, and is sometimes termed a "breezeway, passage, dogrun, or possumtrot; but it is most generally called a dogtrot" (Montell & Morse 1995). Typically, a family slept in one room and cooked in the other. The trot provided a covered outdoor living space for warm southern climates (Glassie 1968; Jordan 1985; Kniffen 1986).

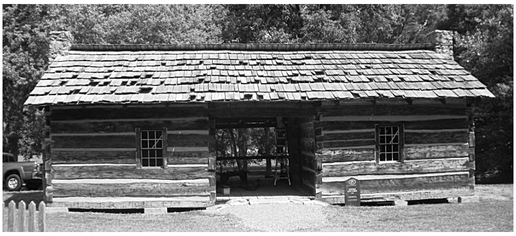

Figure 1.—Grayson dogtrot south façade. Dogtrot houses have two rectangular rooms and a central hall, all under a continuous roof. The open hall is known as a trot.

The Grayson dogtrot is made from planked logs that measure 46–61 cm (18–24 in) in height, and approximately 23 cm (8 in) in thickness. The house is seven logs in height, and modern chinking fills the spaces between the timbers. It is corner timbered with half-dovetail notching (Fig. 4). A modern porch has been added to the north façade (Fig. 3).

The dogtrot house form has European antecedents, although scholars argue whether it is descended from English, German, or Finnish and Scandinavian traditions (Glassie 1968; Jordan 1985; McAlester 1988; Roberts 1996). Early colonists brought the design to the Delaware Valley where it was carried by the wave of settlement into the American interior via the Middle Atlantic migration stream. This eighteenth and nineteenth century movement of settlers extended southwest from Pennsylvania through eastern Tennessee, then across northern Mississippi and Alabama. Settlers then brought it north into western Kentucky and the southern regions of Indiana, Illinois, and Ohio (Kniffen 1986; Roberts 1996). The open trot allowed breezes to cool the structure in southern climates and is the reason the highly functional house form was used for an extended time. While the house form is common in Appalachia and the upper South, it is rare in the Hoosier state; southern Indiana marks the northern reaches of its diffusion.

The goal of this investigation was to determine the likely construction date of the Grayson House through dendroarchaeological analysis. Construction dates for buildings can be suggested by accurately crossdating the outermost ring of an individual timbers. The outermost ring will be the year in which an

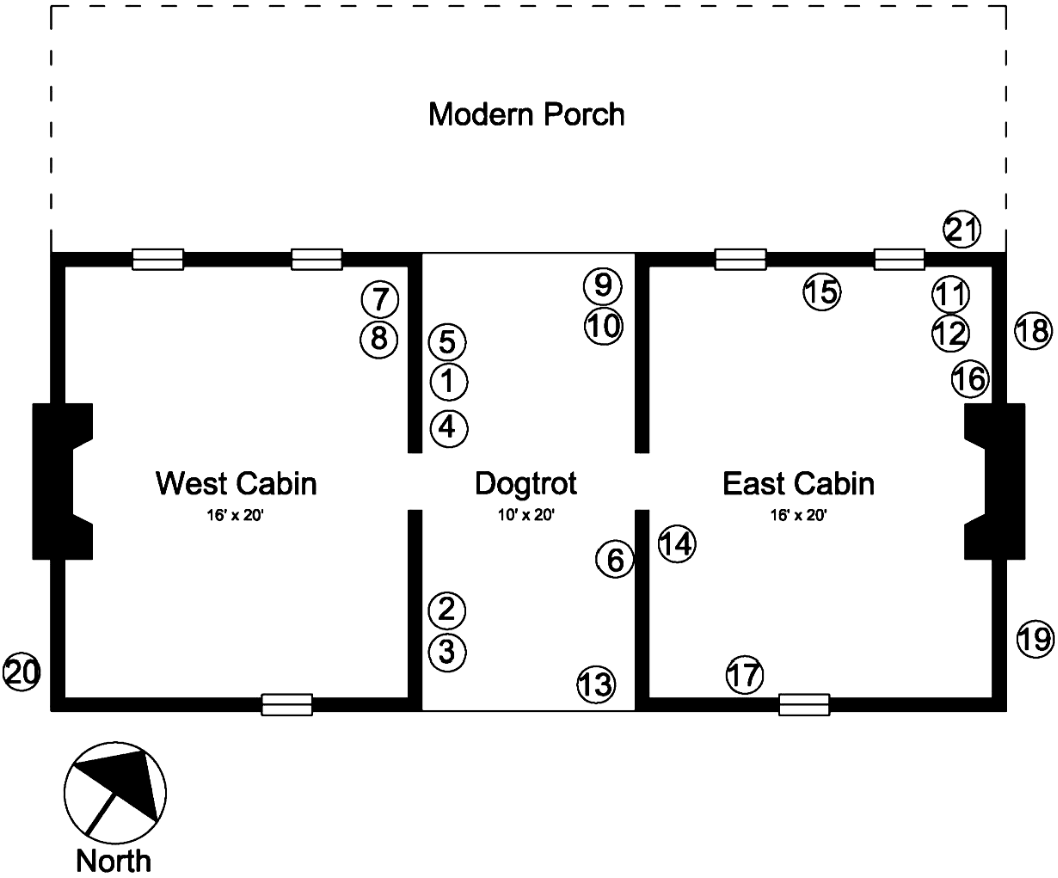

Figure 2.—Plan of Grayson dogtrot showing sampling locations. Note: measurements are given in feet, the unit most likely used by the builders of the structure. See Table 1 for details of each provenience.

individual tree died. For hypothesized construction dates to be accurate, the outermost ring of a timber must represent the last year of growth for the tree; the outermost ring must either be adjacent to bark or be associated with the wane of the piece of lumber. Wane can be identified by noting a uniform, rounded outer surface of a timber that is free of any tool marks (e.g., those created by hatchet, ax, adze, chisel, or saw). If wane is present but bark is not, the outermost ring of the timber represents the last ring formed by a tree, and the bark most likely fell off or was removed. When a

Figure 3.—Grayson house east façade showing end chimney and porch.

Figure 4.—Close up of southeast corner of the Grayson house showing half-dovetail notching.

number of timbers from a structure have similar (or comparable) death dates, one can infer a likely construction date.

METHODS AND MATERIALS

Samples were obtained from the building by coring timbers using a battery-powered drill (1.3 cm chuck) and a dry wood boring bit (Forest Research Tools, Knoxville, Tennessee). Sampling focused on wane-bearing and bark-bearing timbers since determining construction date of the house was the major goal of this investigation. Since repair and renovation are common in log buildings, sampling was performed throughout the structure so that an accurate date of initial construction could be determined (Fig. 2). Each timber in the house was carefully inspected to make certain that either bark or wane was present. Prior to coring the timber, a permanent marker was used to color the outermost surface of the wood or bark to ensure that it was kept intact during the coring process. The bit was drilled into the timber until it passed the approximate center or pith of the timber or until a void was reached.

For several timbers, multiple cores were extracted to increase sample size, to provide a better opportunity for dating if one core was undatable (e.g., extensive insect damage or wounds), and to ensure that the outermost ring did in fact represent wane. Replicate sampling of an individual timber is especially beneficial when working with tulip poplar (*Liriodendron tulipifera* L.) because it is prone to containing missing rings (missing rings result from a tree not forming a complete ring around its entire circumference or any ring at all in a given year due to injury or stressful growing conditions).

Cores were stored in labeled PVC pipe to protect them during transport. Each sample or core was assigned a unique identification containing three portions: a three-letter structure identification (MAC), a two digit provenience (individual timber) identification, and a letter indicating the individual series sampled from a provenience. For example sample MAC03B identifies a replicate series (B) obtained from the third provenience (03) sampled from the structure.

Cores were glued into individually labeled mounting boards so that the vessels were aligned vertically for later surface preparation, ring measurement, and dating. Each core was sanded with progressively finer grits of sandpaper (Stokes & Smiley 1968) to expose the tree-ring structure. Each core was sanded with a belt sander with ANSI 80-, 120-, 180-, and 220-grit sanding belts. A palm sander was then used with ANSI 220-, 320-, and 400-grit sandpaper (Orvis & Grissino-Mayer 2002). Each core was then hand sanded/polished with 30 and 15 micron sanding film.

Starting with the innermost (oldest) tree ring, years—not dates—were assigned to each ring using a boom dissection microscope at 40×. The innermost ring was assigned year 1, the next year 2, and so on until the outermost ring was numbered. The resulting tree-ring series were then considered to be "floating" since individual rings were assigned arbitrary years and not calendar dates (Grissino-Mayer 2001). For each floating series, a skeleton plot was manually created. Skeleton plots are prepared to graphically highlight the pattern of small and large rings in the samples (Stokes & Smiley 1968). The skeleton plots of each series were compared to each other to identify common growth patterns and potential marker years (e.g., abnormally small rings).

The ring widths of each sample were measured to the nearest 0.01 mm with a boom dissecting scope (45× magnification), VELMEX unislide measuring device (VELMEX Inc., Bloomfield, NY), ACU-RITE linear encoder (ACU-RITE Inc., Jamestown, NY), and Quick-Check digital readout device (Metronics Inc., Bedford, NH) connected to a computer. The program MEDIR (Version 1.13; Krusic et al. 1997) was used during the measurement process to create computerized ring-width series consisting of years and measurements for each sample.

The outermost ring in each series with wane was not measured since it is not possible to know if the ring was fully formed (i.e., the tree could have been harvested during the growing season). The innermost ring of most samples could not be measured since sawing, hewing, cracking, or decay does not follow a ring boundary, and the ring would be incomplete. The innermost ring of a series can be measured only if pith is present since the innermost ring would be fully present and adjacent to the pith. Measurement of an entire series is not always possible if the sample has an irregular growth pattern due to scar tissue or growth anomalies associated with branching. When such patterns were encountered, measuring was performed

Table 1.—Series information for each of the dated tulip poplar timbers sampled from the Grayson House, New Harmony, Indiana. “First” and “last” refer to the first and last years present in each of the series. If more than one sample was taken from an individual timber, the provenience description is given only once and not for each of the series. Mean and SD refer to ring widths of each series (mm). In the “Outer Ring” column, “w” = wane; “b” = bark; blank = outer ring is not the last ring formed on the log, and the death date of the timber is undeterminable. See Fig. 2 for sample locations.

Series	First	Last	Outer Ring	Years	Mean	SD	Provenience
MAC01A	1803	1851	w	47	0.42	0.22	West pen, east wall
MAC01B	1774	1851	b	76	0.64	0.56	
MAC01C	1738	1792		53	0.63	0.30	
MAC01D	1713	1738		24	1.52	0.57	
MAC01E	1738	1757		18	0.79	0.32	
MAC02A	1761	1851	w	87	0.57	0.51	West pen, east wall
MAC02B	1681	1761		79	1.28	0.64	
MAC03A	1772	1834		60	1.05	0.46	West pen, east wall
MAC03B	1703	1851	w	145	0.81	0.52	
MAC03C	1709	1772		62	0.67	0.32	
MAC03D	1834	1851	w	16	0.54	0.11	
MAC04I	1780	1852	w	71	0.63	0.46	West pen, east wall
MAC05A	1730	1852	w	121	0.92	0.60	West pen, east wall
MAC06A	1760	1851	w	90	1.35	0.82	East pen, west wall
MAC11A	1797	1827		29	1.86	0.78	Ceiling beam
MAC12A	1719	1812		91	1.21	0.59	Ceiling beam
MAC12I	1799	1852	w	52	0.69	0.25	
MAC14A	1770	1852	b	71	1.00	0.66	East pen, west wall
MAC14B	1808	1852	b	43	0.66	0.18	
MAC15A	1767	1808		40	2.17	1.17	East pen, north wall
MAC16A	1758	1852	b	93	1.49	1.38	East pen, east wall
MAC17B	1759	1852	b	92	1.22	1.16	East pen, south wall
MAC18A	1745	1824		76	2.07	1.22	East pen, east wall
MAC18B	1827	1852	b	24	0.90	0.45	
MAC19A	1783	1852	w	68	1.42	0.63	East pen, east wall
MAC20A	1802	1852	w	49	0.88	0.39	West pen, west wall
MAC21A	1708	1806		97	1.17	0.50	East pen, north wall

only in the region where normal growth was observed. Inclusion of incomplete rings and abnormally-formed rings in the ring-width series was avoided since the true ring width is not determinable and subsequent inclusion of such measurements would likely bias growth pattern analyses.

Calendar date assignment to individual rings in the floating series was achieved by cross-dating the samples against local chronologies with known dates (see Rubino 2014, in this issue for an example). Chronologies are comprised of dated and measured tree rings created by studying numerous trees in an area. These local chronologies consist of living trees and crossdated timbers from other regional structures. Crossdating was performed by using skeleton plots and by using ring-width measurements via the computer program COFECHA (Holmes 1997). COFECHA utilizes a correlation procedure to enhance time-series characteristics (the pattern of small and large rings) in the samples. COFECHA assists in date assignment of floating tree-ring series by comparing the measured floating series to measured series with known, verified dates. Following a run of COFECHA, a list of possible calendar dates for dating each of the floating series is provided (Holmes 1997; Grissino-Mayer 2000). These tentative dates were then compared to the growth patterns observed in the skeleton plots of each sample to assist in final calendar date assignment.

COFECHA was also used to verify date assignments (i.e., quality control). COFECHA breaks each series into consecutive 50-year segments overlapping by 25 years (Grissino-Mayer 2000; Holmes 1997). The correlation of each of the segments is then checked against all other series. If a correlation coefficient for

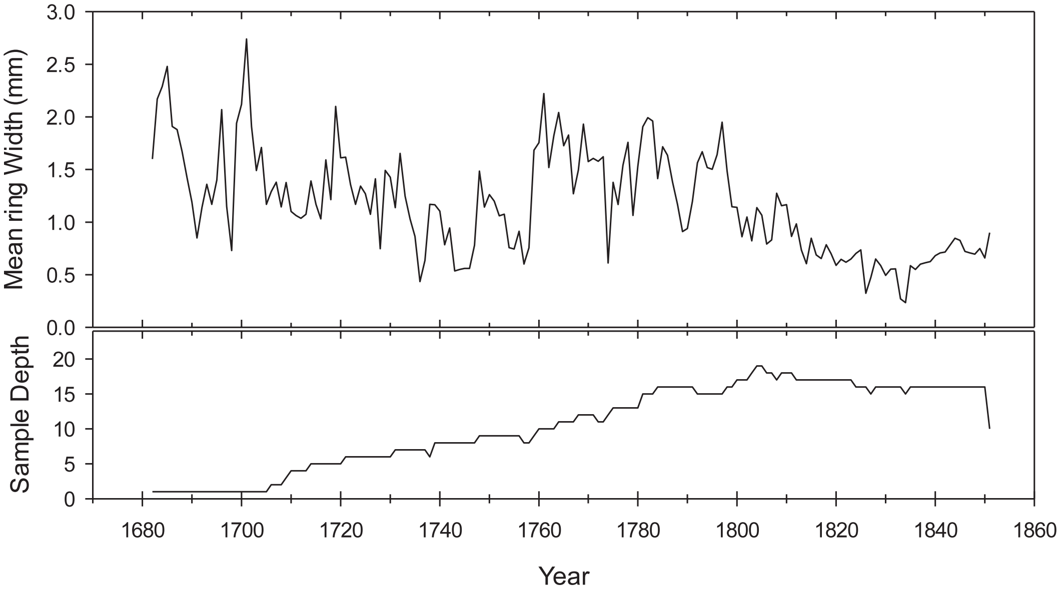

Figure 5.—Tree-ring chronology prepared from tulip poplar timbers of the Grayson House, New Harmony, Indiana. Sample depth is the number of samples that had a ring present at an individual year.

a 50 year segment has an *r*-value > 0.33 (associated probability of 0.01) the crossdating is verified, and date assignment is likely successful.

Each timber type was identified to the lowest possible taxonomic rank (species or subgenus) using macroscopic and microscopic wood anatomy characteristics (Panshin & de Zeeuw 1980). Subsamples for wood identification were obtained by removing paper-thin sections of wood with a double-edged razor blade.

RESULTS

Samples were obtained from American (white) elm (*Ulmus americana* L.), "White" oak (*Quercus* subgenus *Lepidobalanus*), hickory (*Carya* sp.), and tulip poplar timbers. Taxonomy and nomenclature follows Gleason & Cronquist (1991). Twenty-one proveniences were sampled from throughout the structure (Fig. 2). Dates were successfully assigned to 27 samples from16 different proveniences. Confident date assignment was not possible for 5 of the proveniences due to extensive insect damage and decay that did not allow extraction of long enough cores to permit reliable crossdating or extensive growth suppressions that consisted of tree rings exhibiting little to no change in ring width from year to year (accurate crossdating necessitates ring-width variation).

Tulip poplar was the dominant timber type sampled from the structure (Table 1). Two white elm (MAC09 and MAC10), two "white" oaks (MAC08 and MAC13), and one hickory (MAC07) were also sampled; none of these crossdated. All of the tulip poplar timbers crossdated and yielded a 172 year-long chronology (1682–1851) consisting of 1774 accurately dated and measured tree rings (Table 1; Fig. 5). Mean ring width was 1.06 mm (SD = 0.63).

To determine the strength and quality of dating among the samples, each series was broken into 50 year-long segments overlapping by 25 years (e.g., 1700–1749, 1725–1774, 1750–1799, etc.). Each segment was then correlated against all other series in the chronology. Additionally, each of the complete series was correlated against all other series in the chronology. Correlation analysis was performed with the ring width measurements for each year in each of the series. Strong and significant correlations were found among the 50 year-long segments and among all series in the chronology (Table 2). These significant correlations suggest that accurate crossdating was achieved. Skeleton plots (not shown) also suggest successful crossdating among the

Table 2.—Segment (50 year-long segments overlapping by 25 years) and series correlation coefficients for crossdated tulip poplar timbers from the Grayson House. A correlation coefficient > 0.33 indicates a significant correlation ($P < 0.01$) for 50 year segments. Time span is the period for which tree rings were measured for each series.

Series	Time Span	1700–1749	1725–1774	1750–1799	1775–1824	1800–1849	1825–1874	Series *r*
MAC01A	1804–1850					0.66		0.66
MAC01B	1775–1850				0.67	0.73	0.69	0.69
MAC01C	1739–1791		0.49	0.50				0.51
MAC01D	1714–1737	0.82						0.82
MAC01E	1739–1756		0.43					0.43
MAC02A	1764–1850			0.67	0.59	0.66	0.63	0.64
MAC02B	1682–1760	0.63	0.64					0.65
MAC03A	1774–1833			0.68	0.71	0.69		0.68
MAC03B	1706–1850	0.68	0.73	0.65	0.66	0.67	0.67	0.66
MAC03C	1710–1771	0.65	0.61					0.55
MAC03D	1835–1850						0.21	0.21
MAC04I	1781–1851				0.35	0.34	0.33	0.36
MAC05A	1731–1851		0.60	0.69	0.60	0.62	0.64	0.62
MAC06A	1761–1850			0.72	0.56	0.42	0.42	0.59
MAC11A	1798–1826				0.49			0.49
MAC12A	1721–1811	0.47	0.59	0.54	0.61			0.54
MAC12I	1800–1851					0.49	0.49	0.49
MAC14A	1781–1851				0.62	0.75	0.75	0.66
MAC14B	1809–1851					0.74		0.74
MAC15A	1768–1807			0.39				0.40
MAC16A	1759–1851			0.72	0.51	0.52	0.49	0.61
MAC17B	1760–1851			0.67	0.58	0.44	0.42	0.55
MAC18A	1748–1823		0.78	0.76	0.70			0.72
MAC18B	1828–1851						0.81	0.81
MAC19A	1784–1851				0.49	0.33	0.30	0.37
MAC20A	1803–1851					0.60		0.60
MAC21A	1709–1805	0.78	0.86	0.73	0.72			0.74

individual timbers from the house. Crossdating was greatly aided by noting decreased growth rate (in relation to neighboring rings) in 1728, 1736, 1752, 1774, 1833 (missing ring in two of the timbers), and 1834 (Fig. 5).

Calendar date assignment to individual tree rings was performed and assessed by correlating a master chronology (a mean chronology comprised of all crossdated tree rings from the house's timbers) with local regional chronologies. The ring-width master chronology of the Grayson House correlated significantly with all other regional chronologies (Table 3). The consistent, significant correlations suggest that accurate calendar date assignment was achieved.

DISCUSSION

An 1851 or 1852 death date was found in all timbers for which a death date was determinable (i.e., bark or wane present). The timbers

Table 3.—Correlation results of 50 year-long segments (overlapping by 25 years) of the Grayson House mean master chronology with regional tulip poplar chronologies from Indiana. Correlations for 50 year-long segments are significant ($P < 0.01$) if $r > 0.33$.

Chronology	Span	1682–1731	1707–1756	1732–1781	1757–1806	1782–1831	1802–1851
Corydon	1575–1901	0.40		0.56	0.55	0.41	0.33
Jefferson County	1457–1889			0.62	0.66	0.48	0.33
Switzerland County	1613–1856	0.42	0.48	0.69	0.70	0.52	0.46
Washington County	1637–1882	0.40	0.33	0.60	0.62	0.54	0.45
New Harmony (1)	1686–1858		0.64	0.76	0.63	0.55	0.75
New Harmony (2)	1704–1885		0.49	0.81	0.74	0.65	0.66

showed uniform hewing marks and thicknesses. Also, the timbers had little or no extraneous tooling (e.g., empty mortises) suggesting that the timbers had been recycled from other buildings. Since timbers from throughout the house had similar death dates, we conclude that the house was most likely originally constructed in its current layout/format as a dogtrot house after the initiation of the 1852 growing season. We are not certain (nor is Historic New Harmony) how the 1775 construction date was assigned, but it should be corrected in light of tree-ring evidence.

Crossdating (using skeleton plots and correlation analysis) among tree-ring series was achieved in this investigation using only tulip poplar. Tulip poplar is not commonly used for dendrochronological and dendroarchaeological investigations and has been considered a species of minor importance to dendrochronology because it has been reported to only crossdate within and between trees on a site-by-site basis (Grissino-Mayer 1993). However, we found a very consistent signal (repeated pattern of small and large tree rings among samples) in the samples at both the within (all Grayson house samples) and among (regional tulip poplar chronologies) site levels using correlation analysis (Tables 2 and 3). We suggest further investigation of the potential of tulip poplar for tree-ring studies. In this investigation the samples exhibited consistent annual ring variation and sensitivity to extraneous growth factors.

For this analysis, we utilized dendrochronology within a framework of interpretive archaeology to explain historic sites to public and academic audiences (Wilkie 2009). Along with Historic New Harmony, we are working with local museums and individuals to interpret historic timber buildings in public history settings (Baas & Rubino 2012). The authors aim to continue investigating New Harmony structures, specifically Harmonist houses and community buildings.

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LITERATURE CITED

Baas, C. & D.L. Rubino. 2012. Using tree-ring growth patterns to date the construction of a beater hay press barn in Allensville, Switzerland County, Indiana. Indiana Archaeology 7:35–57.

Bortolot, Z.J., C.A. Copenheaver, R.L. Longe & J.A.N. Van Aardt. 2001. Development of a white oak chronology using live trees and a post-civil war cabin in south-central Virginia. Tree-Ring Research 57:197–203.

Glassie, H. 1968. Pattern in the Material Folk Culture of the Eastern United States. University of Pennsylvania Press, Philadelphia, Pennsylvania. 344 pp.

Gleason, H.A. & A. Cronquist. 1991. Manual of Vascular Plants of Northeastern United States and Adjacent Canada, 2nd ed. New York Botanical Garden, Bronx, New York. 910 pp.

Grissino-Mayer, H.D. 1993. An updated list of species used in tree-ring research. Tree-Ring Bulletin 53:17–43.

Grissino-Mayer, H.D. 2001. Evaluating crossdating accuracy: a manual and tutorial for the computer program COFECHA. Tree-Ring Research 57: 205–221.

Holmes, R.L. 1997. The Dendrochronology Program Library. Pp. 40–71. *In* The International Tree-Ring Data Bank Program Library Version 2.1 User's Manual (H.D. Grissino-Mayer, R.L. Holmes & H.C. Fritts, Eds.). Laboratory of Tree-Ring Research, The University of Arizona, Tucson, Arizona.

Indiana Museum and State Historic Sites. 2013. Interpretive notes and newspaper clippings from New Harmony office.

Jordan, T.G. 1985. American Log Buildings: An Old World Heritage. University of North Carolina Press, Chapel Hill, North Carolina. 240 pp.

Kniffen, F.B. 1986. Folk housing: key to diffusion. Pp. 3–26. *In* Common Places: Readings in American Vernacular Architecture (D. Upton & J.M. Vlach, Eds.). University of Georgia Press, Athens, Georgia.

Krusic, P.J., R.L. Holmes & J.C. King. 1997. MEDIR Version 1.13 measurement program. Pp. 18–20. *In* The International Tree-Ring Data Bank Program Library Version 2.1 User's Manual (H.D. Grissino-Mayer, R.L. Holmes & H.C. Fritts, Eds.). Laboratory of Tree-Ring Research, The University of Arizona, Tucson, Arizona.

McAlester, V. & L. McAlester. 1988. A Field Guide to American Houses. Alfred A. Knopf, New York, New York. 525 pp.

Montell, L.W. & M. L Morse. 1995. Kentucky Folk Architecture. University Press of Kentucky, Lexington. 120 pp.

Orvis, K.H. & H.D. Grissino-Mayer. 2002. Standardizing the reporting of abrasive papers used to surface tree-ring samples. Tree-Ring Research 58:47–50.

Panshin, A.J. & C. de Zeeuw. 1980. Textbook of Wood Technology: Structure, Identification, Properties, and Uses of the Commercial Woods of the United States and Canada, 4th ed. McGraw-Hill Inc., New York, New York. 722 pp.

Roberts, W.E. 1996. Log Buildings of Southern Indiana. Trickster Press, Bloomington, Indiana. 228 pp.

Rubino, D.L. 2014. Tree-Ring Analysis in Indiana with Special Attention to Dendroarchaeology. Proceedings of the Indiana Academy of Science 123(1):7–13.

Senter, F.H. 1938. Dendrochronology: can we fix prehistoric dates in the Middle West by tree rings? Indiana History Bulletin 15:118–128.

Stahle, D.W. 1979. Tree-ring dating of historic buildings in Arkansas. Tree-Ring Bulletin 39:1–28.

Stokes, M.A. & T.L. Smiley. 1968. An Introduction to Tree-Ring Dating. University of Chicago Press, Chicago, Illinois. 73 pp.

Therrell, M.D. 2000. The historic and paleoclimatic significance of log buildings in southcentral Texas. Historical Archaeology 34:25–37.

Thun, T. 2005. Norwegian conifer chronologies constructed to date historical timber. Dendrochronologia 23:63–74.

Towner, R., D. Grow, J. Psaltis & A. Falzone. 2001. The importance of sample context in dendroarchaeological interpretation: an example from northwestern New Mexico, USA. Tree-Ring Research 57:75–88.

Wilkie, L.A. 2009. Interpretive historical archaeologies. Pp. 333–345. *In* International Handbook of Historical Archaeology (T. Majewski & D. Gaimster, Eds.). Springer, Tucson, Arizona.

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IMPACTS OF *GALERUCELLA CALMARIENSIS* AND *G. PUSILLA* ON *LYTHRUM SALICARIA* IN INDIANA

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ABSTRACT. *Lythrum salicaria,* an invasive wetland hydrophyte native to Eurasia, has spread across Indiana since 1900. Two *Galerucella* spp. have been utilized as biological control agents for *L. salicaria* in Indiana since 1994. This study examines the impact of *Galerucella* spp. over an 8-10 year period at four Indiana wetlands. *Galerucella* abundance varied substantially over time but had low Spearman's ρ at three sites ($\rho = 0.21$ to 0.40) due to rapid decreases following reduction in *Lythrum*. In contrast, hydrophyte species richness and percent cover were both correlated with time and had higher ρ (0.37 to 0.69) at three sites. The number of *Lythrum* inflorescences and stem density were reduced at all four sites, with inflorescences showing the strongest correlation ($\rho = -0.46$ to -0.78). Although variation was observed between sites, the introduction of *Galerucella* spp. resulted in significant declines in *L. salicaria* at each of the wetlands. Their impacts at these wetlands strongly suggest that *Galerucella* spp. can play a major role in controlling this invasive plant species.

Keywords: *Galerucella*, *Lythrum salicaria*, purple loosestrife, biological control, invasive species, Indiana wetlands

INTRODUCTION

Lythrum salicaria, a wetland hydrophyte native to Eurasia, is considered an invasive species in North America. Although the first observation of *L. salicaria* in North America occurred in New England in 1814 (Mal et al. 1992) and the earliest record in Indiana is from 1900, its spread was considered minor until after 1940 (Stuckey 1980). Today, *L. salicaria* is found throughout Indiana, though it is most common in the northern counties. Additionally, *L. salicaria* occurs in 47 of the contiguous States (it is absent from Florida) (Blossey et al. 2001).

Lythrum salicaria can form extensive stands which many have characterized as monotypic (Malecki et al. 1993). The slightly square stems can grow in clumps of 30–50 from a single taproot, while a terminal spike of tightly clustered flowers may exceed 1 meter in length (Mal et al. 1992). As a result, a single plant can produce over 2.5 million seeds in a single growing season (Malecki et al. 1993). This prolific seed production, coupled with their persistence in a seed bank, permits high recruitment of seedlings even after removal of adult plants (Welling & Becker 1990). Likewise, adult plants are highly competitive (Gaudet & Keddy 1995; Mal et al 1997; Weihe & Neely 1997; Farnsworth & Ellis 2001), especially in non-flooded conditions (Weiher et al. 1996). This results in reductions in species richness (Schooler & McEvoy 2006) as well as density of associated species such as grasses and sedges (Gabor et al. 1996).

Due to its large showy floral display, *L. salicaria* may reduce seed set of native species such as *L. alatum* through competition for pollinators (Brown et al. 2002). Other studies (Brown & Mitchell 2001; Da Silva & Sargent 2011) demonstrate that pollination of *L. alatum* or *Decodon verticillatus* with mixtures that include *L. salicaria* pollen reduce seed set by as much as 1/3.

Replacement of *Typha* (and other graminoids) by *L. salicaria* may have a notable

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impact on wetland function. Its more phosphorus rich leaves drop earlier and decompose more quickly (Bärlocher & Biddiscombe 1996; Emery & Perry 1996; Grout et al. 1997) and likely lead to changes in soil chemistry when compared to communities dominated by *Typha angustifolia* or *Phragmites austalis* (Templer et al. 1996).

Whitt et al. (1999) found that avian species diversity decreased in wetlands where *L. salicaria* was a dominant species. Muskrat (*Agelaius phoeniceus*) use and long-billed marsh wren (*Cistothorus palustris*) nesting were shown to be lower in *L. salicaria* stands than adjacent *Typha* spp. stands (Rawinski 1982; Rawinski & Malecki 1984). Lor (2000) examined the use of *L. salicaria* by many avian species for feeding and nesting. Virginia rail (*Rallus limicola*), sora (*Porazana coaolina*), least bittern (*Ixobrychus exilis*), American bittern (*Botaurus lentiginosus*), and pied-billed grebe (*Podilymbus podicpes*) were found to avoid *L. salicaria* stands. Meanwhile, nearby areas with *Typha* spp. provided habitat for many of these species.

While many studies have identified negative effects of *L. salicaria*, there remains some debate as to whether it is causing damage to native wetland communities. Hager & Vinebrooke (2004) studied six Minnesota wetlands and found that species richness was significantly greater in wetlands that had been invaded by *L. salicaria* compared to *Typha angustifolia* wetlands. A review of the relationship of *L. salicaria* with native flora and fauna by Anderson (1995) determined that, from the literature existing at the time of his review, the affect of *L. salicaria* on wetland ecosystems and native species was unclear. Additionally, Farnsworth & Ellis (2001) found no significant effect between density of *L. salicaria* stems and species richness.

Biological control.—Classical biological control involves the introduction of natural plant enemies to control the introduced plant species (Hight & Drea 1991). In its native European range, *L. salicaria* does not form large monotypic or dominant stands due to control of reproduction and growth provided by native insect herbivores (Blossey et al. 1994) as well as genetic differences in growth patterns (Chun et al. 2010).

Attempts to control *L. salicaria* began in the 1950's, but initial efforts employing flooding, cutting, and burning were largely unsuccessful (Skinner et al. 1994; Blossey et al. 2001). Hand pulling plants was the most successful of early control methods, but required pulling of entire rootstocks, which is highly labor intensive. Chemical control has primarily utilized glyphosate, 2,4-D, or triclopyr; however, because of large and long-persisting seed bank (Welling & Becker 1990), spraying must be repeated (Skinner et al. 1994; Blossey et al. 2001), perhaps every two to three years (Gabor et al. 1996). Additionally, the non-selective nature of chemical control reduced populations of sedges, grasses, cattails, and other native wetland plants (Skinner et al. 1994; Gabor et al. 1996).

Due to the lack of effective control methods and the continued spread of *L. salicaria*, a biological control program was established in North America (Hight & Drea 1991; Malecki et al. 1993; Blossey et al. 2001). Ultimately four insect species were approved for release, *Hylobius transversovittatus* Goeze (a root-mining weevil), *Nanophyes marmoratus* Goeze (a flower feeding weevil), *Galerucella calmariensis* L., and *G. pusilla* Duft (two leaf-beetles) (Blossey et al. 2001). Insect releases began in the United States in 1992 as did the monitoring of their impact on *L. salicaria* and its associated wetland community (Blossey et al. 2001). Results of these releases have been published from across the United States, from two to ten years post-release (Piper 1996; Dech & Nosko 2002; Landis 2003; Grevstad 2006).

The earliest results of biological control of *L. salicaria* occurred in Washington State (Piper 1996) following the release of *Galerucella* spp. in 1992 and 1993. Surveys conducted in 1993 and 1994 found that *Galerucella* spp. had become established at all eight release sites. In another study Dech & Nosko (2002) found that, in Ontario, establishment of *Galerucella* spp. was slow and the populations remained small throughout the three year study period. The *G. pusilla* population crashed in the second year after release, and only minor feeding damage was observed by *Galerucella* spp. over this short time period. In contrast, Landis (2003) found 100% establishment of 24 releases made in Michigan between 1994 and 1999. Within 4–5 years, large populations were observed at all three 1999 release sites. In potted *L. salicaria*, Stamm-Katovich et al. (1999) found that biomass was reduced after a single growing season when *Galerucella* spp. were present. In a natural setting, several years

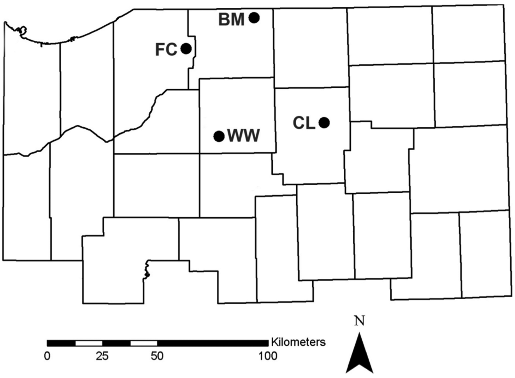

Figure 1.—*Galerucella* spp. transect analysis sites in northern Indiana, USA. BM=Bonneyville Mills, CL=Chapman Lake FC=Fish Creek, WW=Wilson Wetland.

often are required for the impacts of *Galerucella* spp. to be observable (Piper 1996; Dech & Nosko 2002; Landis 2003). Once established, though, Landis (2003) measured suppression of flowering, up to a 98% reduction in stem numbers, and an increase in site species richness. These results, however, did not occur at all sites or in a uniform time. A Minnesota wetland showed a 95% reduction in flowering, and a 50% reduction in stem height four years after release (Blossey & Skinner 2000). In this case, the number of stems per square meter remained constant throughout the sampling period. Grevstad (2006), in New York State, examined the ten-year impacts of *Galerucella* spp. and found that 24 of 36 release sites had *Galerucella* spp. present after ten years. Although only a single site had no persisting *L. salicaria*, as an apparent result of *Galerucella* spp., stem height was reduced by 26% between 1994 and 2004 and reduced flowering was observed at sites with moderate to high plant damage.

In 1994 the Indiana Department of Natural Resources (IDNR), Division of Nature Preserves began releasing *Galerucella* spp. to various wetlands. Additional releases continue as deemed necessary. In order to evaluate the impact of biological control agents in Indiana, the IDNR began monitoring at one release sites in 1996, two additional sites in 1997, and a fourth site in 1999 following the protocol of Blossey (1994). In this study we report the impacts of *Galerucella* spp. on *L. salicaria* and the potential changes in wetland species richness at these Indiana sites.

Table 1.—*Galerucella* abundance categories.

Abundance category	Number of egg masses, larvae, or adults
1	0
2	1–9
3	10–49
4	50–99
5	100–499
6	500–1000
7	<1000

METHODS

Sites and transects.—The releases described in this study occurred in 1996 at three sites and 1999 at the fourth site. The 1996 releases were at Fish Creek, Wilson Wetland, and Bonneyville Mills and the 1999 release site was at Chapman Lake (Fig. 1).

The Fish Creek site (on private property in LaPorte County, Indiana) was a fen grading into a sedge meadow along the west side of the creek. The wetland extended several kilometers above and below the sampling site and, at the start of the study, purple loosestrife was abundant throughout the drainage.

Wilson Wetland (at Culver Academy in Marshall County) was a long, narrow, constructed wetland created by adding sinuosity to a ditched stream channel. The transition from upland to wetland was fairly abrupt and the soils did not have the accumulated organic matter typical of more natural wetlands. Purple loosestrife was also present upstream from the sampling site, and in nearby roadside ditches.

Bonneyville Mill (located along the Little Elkhart River in Bonneyville Mill County Park, Elkhart County) was a ca. 0.2 hectare wetland in a backwater just below the dam for the mill pond. The substrate was a floating mat of plant roots over muck deposits, the least stable substrate of the sites.

Table 2.—Midpoints used for percent cover and percent damage variables.

Coded value	Category midpoint	Category range
A	0%	0%
B	3%	1–5%
C	15%	5–25%
D	37.5%	25–50%
E	62.5%	50–75%
F	87.5%	75–100%

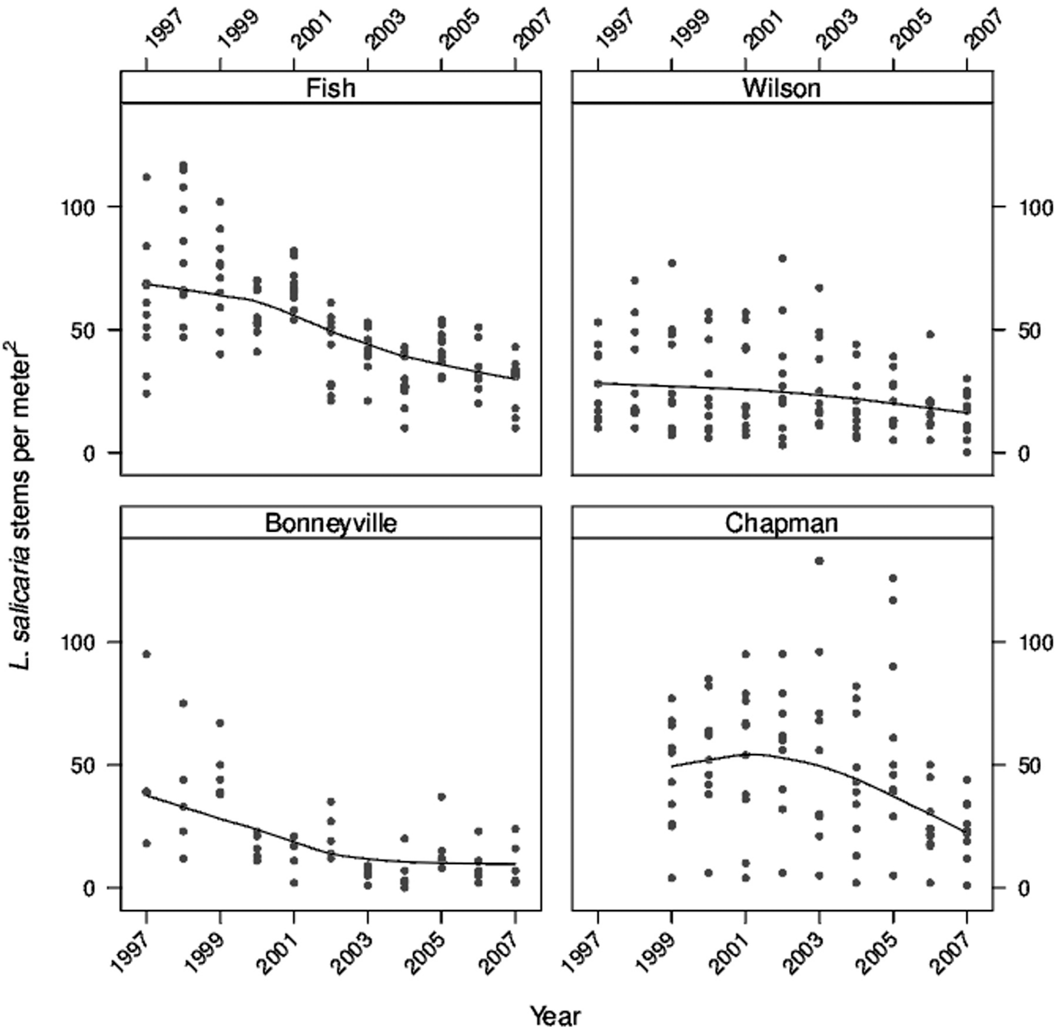

Figure 2.—Correlation between the number of *Lythrum salicaria* stems per quadrat over time.

The Chapman Lake site (on private property, Kosciusko County) was a marly flat adjacent to the shore of Big Chapman Lake. The sampling site was a ca. 1.2 hectare wetland dominated by sedges (*Carex* spp.) and abundant cattail (*Typha* spp.). It was the driest of the wetland sites in this study. Other wetlands were scattered around the shore of the lake and many of them had stands of purple loosestrife.

At all sites except Bonneyville Mills, two transects were oriented parallel to one another and one meter square quadrats were placed every five meters for a total of ten quadrats. Spacing between transects varied based on site size, shape, and distribution of *L. salicaria* within the wetland. At Bonneyville Mills the site was exceptionally small, which restricted the number and positioning of the quadrats. Five quadrats were arranged along two transects. Transects were perpendicular to one another with one quadrat at the intersection and the remaining quadrats place three meters from the intersection in either direction along the transects.

Sampling.—Monitoring was conducted following the protocol outlined by Blossey (1994). This involved sampling each site in the spring and fall, from 1997 through 2007. Spring sampling was conducted between May 18th and June 19th and included insect and vegetative sampling. Each quadrat was surveyed to determine: the estimated abundance of *Galer-*

Table 3.—Spearman rank-sum correlations between year and number of stems, percent cover, and number of inflorescences by *Lythrum salicaria*; species richness; and *Galerucella* abundance. Spearman's followed by an asterisk (*) are significant.

Site	Variable	Spearman's ρ	*p*-value
Bonneyville Mills	Number of stems	−0.64*	<0.0001
	% cover	−0.57*	<0.0001
	Number of inflorescences	−0.46*	0.0006
	Species richness	−0.16	0.25
	Galerucella abundance	0.10	0.48
Chapman Lake	Number of stems	−0.32*	0.002
	% cover	−0.19	0.08
	Number of inflorescences	−0.52*	<0.0001
	Species richness	0.43*	0.0001
	Galerucella abundance	0.21	<0.05
Fish Creek	Number of stems	−0.68*	<0.0001
	% cover	−0.64*	<0.0001
	Number of inflorescences	−0.74*	<0.0001
	Species richness	0.69*	<0.0001
	Galerucella abundance	0.40*	<0.0001
Wilson Wetland	Number of stems	−0.21*	0.03
	% cover	−0.37*	<0.0001
	Number of inflorescences	−0.78	<0.0001
	Species richness	0.39*	<0.0001
	Galerucella abundance	0.21*	0.03

ucella spp. in each life stage (eggs, larvae, adults), percent damage to *L. salicaria*, percent cover of *L. salicaria*, number of *L. salicaria* stems, percent cover of *Typha* spp., number of *Typha* spp. stems. Additionally, in each quadrat the height of the five tallest *L. salicaria* stems and the five tallest *Typha* spp. stems were recorded. Each quadrat also was surveyed to list all present plant species. *Galerucella* abundances for each life stage were recorded as a categorical variable as shown in Table 1. Percent damage and percent cover were assigned to categories as shown in Table 2.

Fall sampling occurred from September 1st to 22nd. At this time *L. salicaria* has finished blooming and *Galerucella* spp. activity had ended. Since insect activity had ceased, no measure of beetle abundance or damage was made. The same measures of *L. salicaria* and *Typha* spp. were made as during the spring sampling. The five tallest *L. salicaria* plants in each quadrat were surveyed for the following: number of inflorescence, length of the terminal inflorescence, number of flower buds in 5 cm of inflorescence. The total number of inflorescence in each quadrat also was recorded. Finally, a list of plant species present in each quadrat was compiled.

Data analysis.—Data obtained from the five tallest plants in each quadrat were averaged to obtain a single value for each quadrat per year. *Galerucella* spp. abundances presented some challenge. The numbers of each life stage were estimates and were assigned to a category (Table 1). Additionally, the categories had unequal ranges, some of which were quite large; because of this using midpoints as estimates seemed misleading. Therefore, to analyze *Galerucella* spp. abundance the three categorical values were averaged for each quadrat in each year. These calculated abundances are of little use in describing the actual number of beetles (except for an abundance of 1, which means *Galerucella* spp. were not present); rather, they provide an indication of the relative size of the population. Percent cover and percent damage values were defined as the midpoint of the category range (Table 2).

Release timing (Chapman Lake) and replication (Bonneyville Mills) varied between sites, preventing analysis across sites. Therefore, each site was analyzed separately. Statistical analysis and graph plotting was done using R 2.14.2 (R Development Core Team 2011) and the *lattice* package (Sarkar 2008). Fall data were used to assess *L. salicaria* response over time. Data were not normally distributed, and therefore required the use of non-parametric methods. Spearman's rank-sum correlations were calcu-

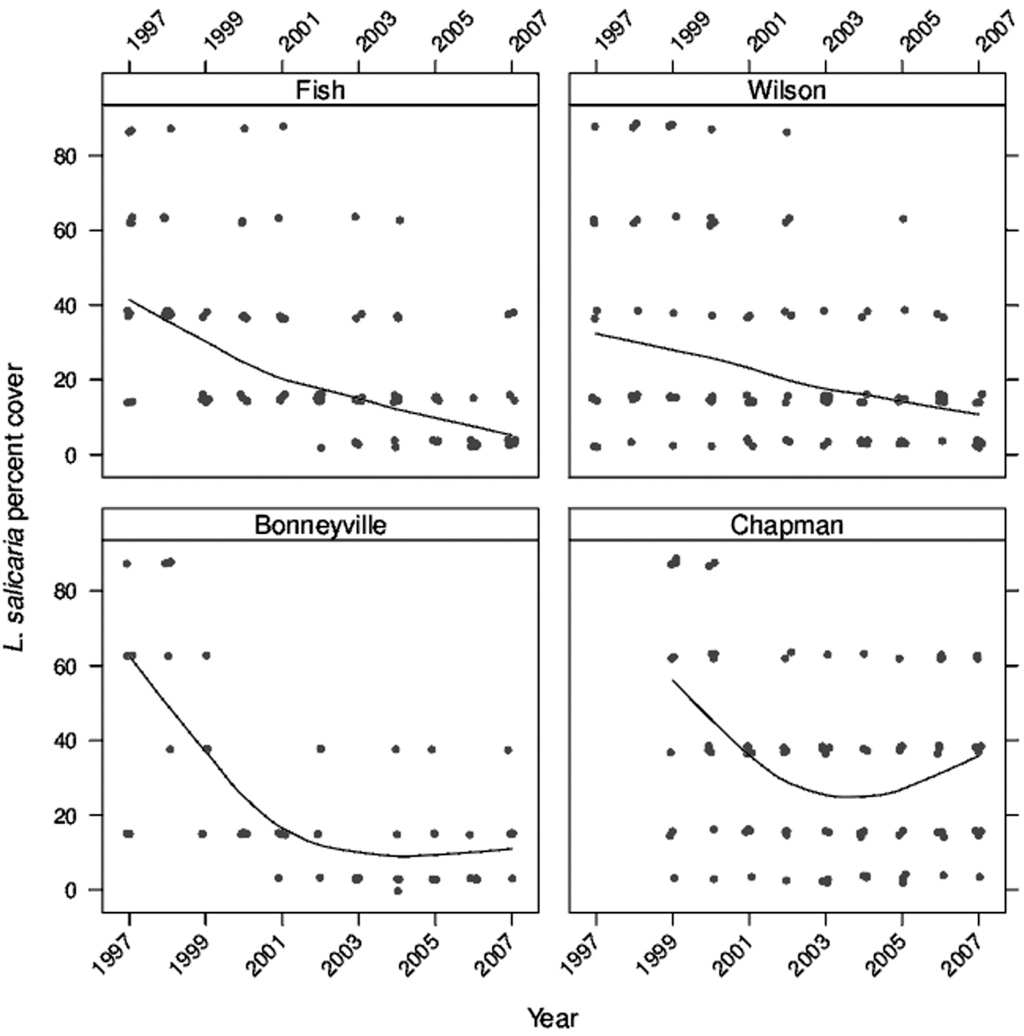

Figure 3.—Change in *Lythrum salicaria* percent cover class over time. Jitter has been added to keep points from overlapping, since both variables are essentially categorical.

lated to examine the relationship between year of observation and the following variables: species richness, number of stems, percent cover of *L. salicaria*, and number of inflorescence. Additionally, to aid in graphical interpretation, best fit lines were drawn through scatterplots of the same data using the *smooth* option of the *xyplot* command which can be found in the *lattice* package. This utilizes loess smoothing which fits the line to the scatterplot using local polynomial regression, providing a graphical view of general trends in the data.

In order to provide comparisons to Landis (2003) and Farnsworth & Ellis (2001), the relationship between *L. salicaria* stem density and species richness was examined. These data were normally distributed at all sites, so a simple linear regression was used. The number of *L. salicaria* stems per quadrat (stem density) was the independent variable and species richness was the dependent variable.

RESULTS

Spearman's ρ showed significant negative correlation at all sites between year and number of stems per square meter ($p < 0.001$) (Fig. 2). Fish Creek ($\rho = -0.68$) and Bonneyville Mills ($\rho = -0.64$) had the highest correlations. At Chapman Lake ρ was -0.32 and at Wilson Wetland ρ was -0.21 (Table 3).

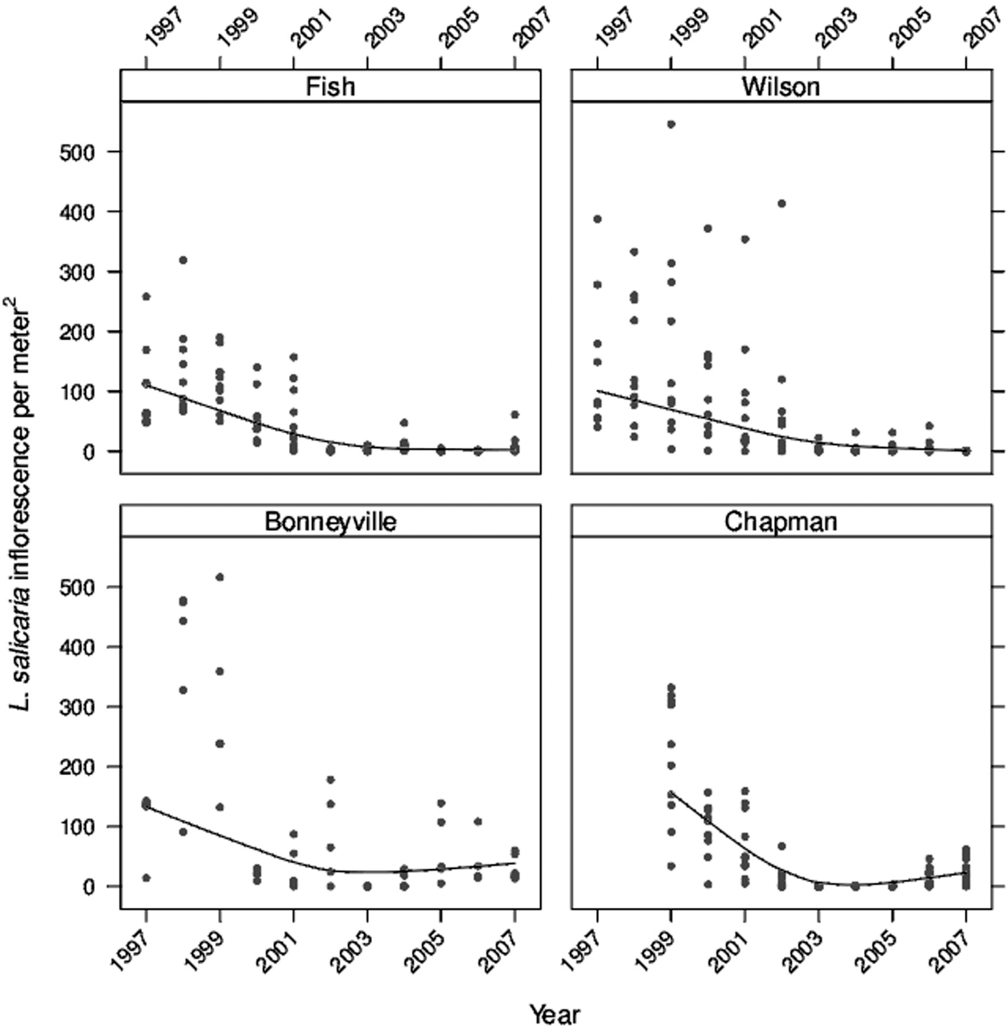

Figure 4.—Correlation between total number of *Lythrum salicaria* inflorescences over time.

Percent cover of *L. salicaria* was significantly correlated to year ($p < 0.001$) at all sites except Chapman Lake ($p = 0.08$) (Fig. 3). Fish Creek again showed the highest correlation ($\rho = -0.64$). Spearman's ρ was −0.57 at Bonneyville Mills and −0.37 at Wilson Wetland (Table 3).

Correlations between year and number of inflorescence per quadrat was significant at all sites ($p < 0.001$) (Fig. 4). The highest correlation was at Wilson Wetland, where ρ was −0.78. Spearman's ρ was −0.74 at Fish Creek, −0.52 at Chapman Lake, and −0.46 at Bonneyville Mills (Table 3). In comparison to other metrics, the number of inflorescence showed a consistently high ρ.

Spearman's ρ showed significant positive correlation between species richness and year at Chapman Lake ($p < 0.001$), Fish Creek ($p < 0.001$), and Wilson Wetland ($p < 0.001$), but not at Bonneyville Mills ($p = 0.25$) (Fig. 5). Fish Creek had a ρ of 0.69, the highest of the sites. Correlations at Chapman Lake and Wilson Wetland were noticeably lower with ρ's of 0.43 and 0.39 respectively (Table 3).

The abundance of *Galerucella* spp. was significantly correlated with time at Fish Creek ($p < 0.0001$), Wilson Wetland ($p < 0.03$), and Chapman Lake ($p < 0.05$) and was insignificant at Bonneyville Mills ($p = 0.18$). At the same time, the Spearman's correlations were consis-

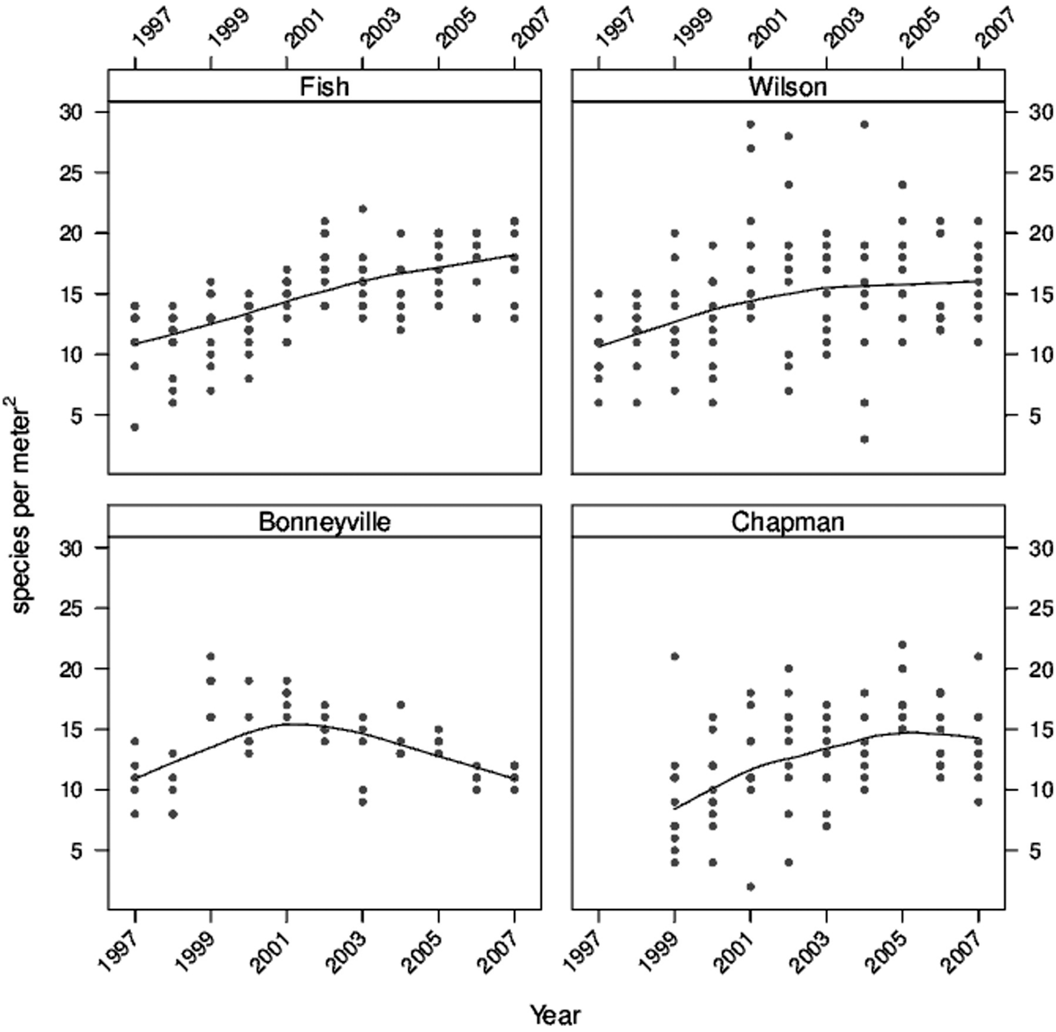

Figure 5.—Change in total autumn species richness over time.

tently low being 0.40 at Fish Creek, 0.21 at Wilson Wetland, and 0.21 at Chapman Lake.

Species richness showed a significant relationship to *L. salicaria* stem density at Chapman Lake ($p = 0.01$; $F(88) = 6.38$, $r = 0.26$), Fish Creek ($p < 0.0001$; $F(108) = 43.74$, $r = 0.54$), and Wilson Wetland ($p = 0.02$; $F(102) = 5.39$, $r = 0.22$), but was not significant at Bonneyville Mills ($p = 0.79$; $F(53) = 0.07$, $r = 0.04$) (Fig. 7). When analyzed across all four sites collectively the relationship was highly significant ($p < 0.0001$; $F(363) = 26.04$, $r = 0.26$).

DISCUSSION

The response of *L. salicaria* to *Galerucella* spp. varied between sites, with some sites showing a very strong correlation between plant characteristics and time, while others showed only weak correlations.

While all sites showed a negative correlation between number of stems and year, this relationship was very strong at Fish Creek and Bonneyville Mills, but weaker at Chapman Lake, and nearly insignificant at Wilson Wetland. One potential explanation for this is that heavy feeding by *Galerucella* spp. may reduce plant growth early in the season. Following this reduction in height and flowering, plants have been shown to respond by producing new stems (Blossey & Skinner 2000). Regardless, it is clear that in the years following release of *Galerucella* spp., the number of stems decreased at all sites.

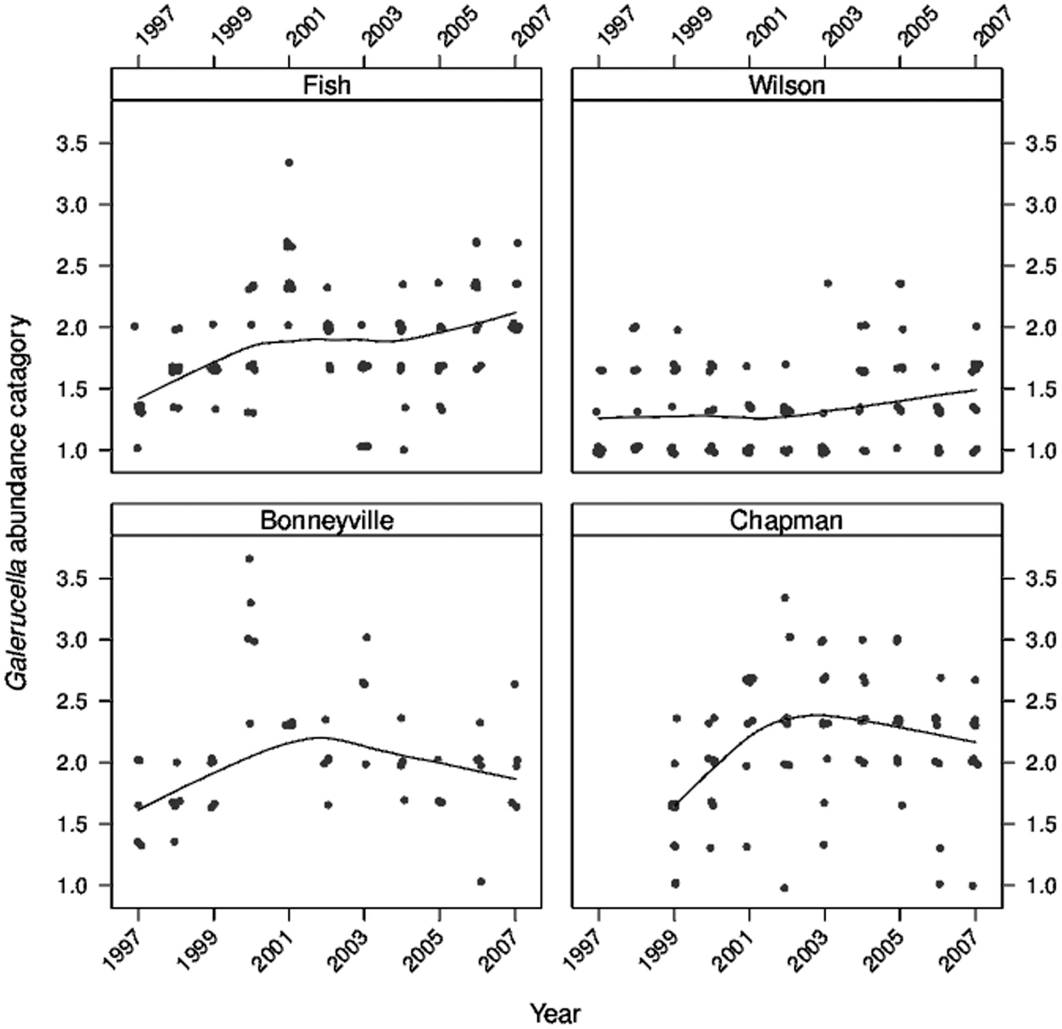

Figure 6.—Change in *Galerucella* abundance over time. Abundance is calculated as the mean categorical value for the three life stages, as described in Methods, and jitter has been added to keep points from overlapping.

Blossey & Skinner (2000) observed similar reduction in stem densities at one site.

Percent cover by *L. salicaria* showed substantial negative correlation with time at three sites (Bonneyville Mills, Wilson Wetland, Fish Creek), but not at Chapman Lake. This could be caused by several factors. Possibly, this illustrates a recovery of *L. salicaria* at the Chapman Lake site. It was the driest of the four sites and potentially *L. salicaria* has greater competitive ability under these non-flooded conditions (Weiher et al. 1996). At the same time, though, other measures of *L. salicaria* health did not show similar trends. Alternatively, these minor changes may be due to the categorical estimates of cover class or differences in personnel making these observations in subsequent years. Meanwhile, the other three sites demonstrate a significant decrease in cover of *L. salicaria* over time.

All four sites exhibited a correlation between the number of *L. salicaria* inflorescences and year. Consistent reductions in flowering were observed three to five years after biocontrol release, similar to Landis (2003). Flowering was essentially terminated at some point for all sites and in the final year (2007) remained extremely low. The prevention or reduction of flowering could result in major long term impacts on *L. salicaria* populations. Seed accumulation

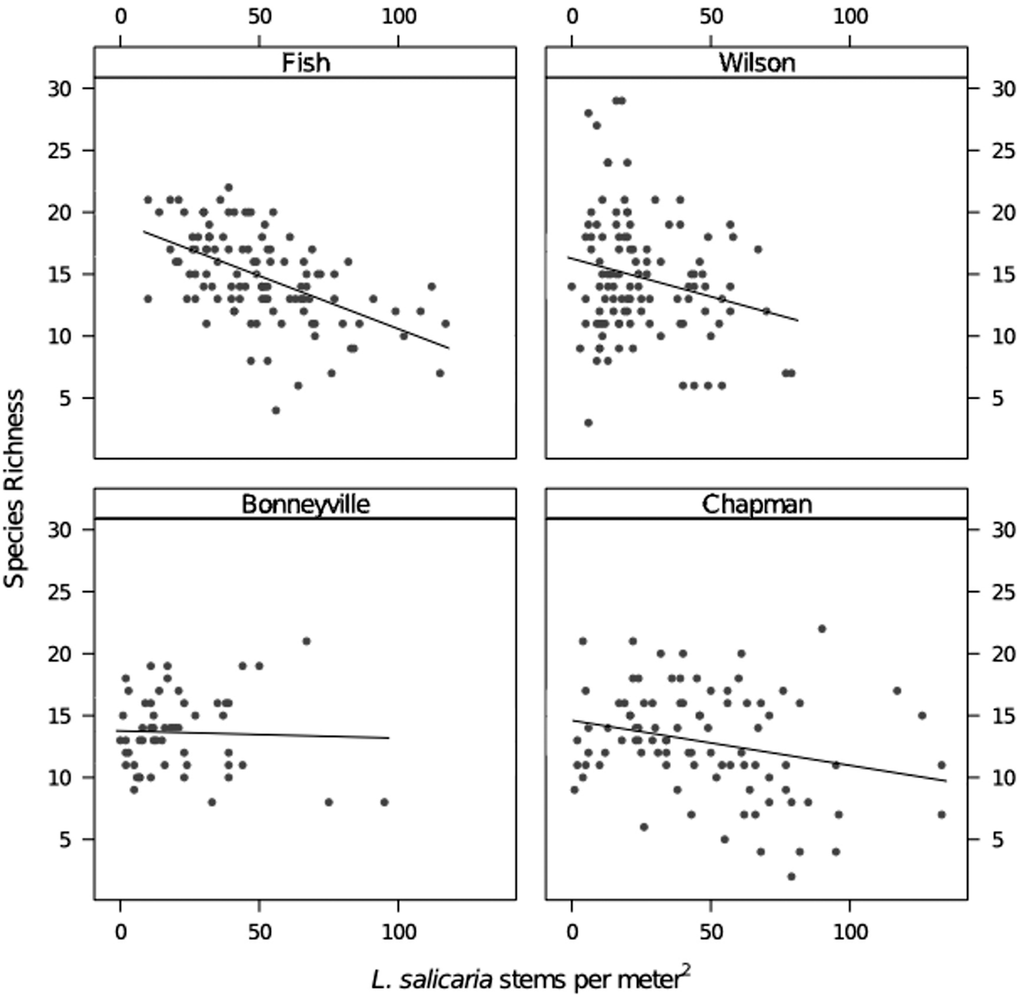

Figure 7.—Linear regression of the relationship between density of *L. salicaria* and plant species richness by site.

in these wetlands was likely high, but if the flowering remains very low, the existing seed bank could become depleted, leading to further reductions in *L. salicaria* in the future.

Species richness was not significantly correlated with year at Bonneyville Mills, but was significant at the other three sites. At Bonneyville Mills, species richness increased steadily for the first 3–5 years, but decreased unexpectedly over the next five years. During the same time period, all measures of *L. salicaria* health continued to decrease.

The correlation identified between *L. salicaria* stem density and species richness at three of the four sites supports the findings of Landis (2003). However, Farnsworth & Ellis (2001) found no noteworthy relationship one year after release of *Galerucella* spp. This suggests that a longer period of time is needed to observe these impacts. Also, the relationship of stem density and species richness was not significant at Bonneyville Mills. This may have been due to natural species richness within that wetland, particularly since it was such a small area. The smaller number of quadrats within the site may have caused any relationship which did occur to remain unnoticeable.

The correlation of *Galerucella* spp. abundance with time was low, even at sites where it was significant, with the highest correlation being 39.9% at Fish Creek. Examination of the scatter plots and best-fit lines (Fig. 6) better demonstrates the temporal patterns. A single year with high *Galerucella* spp. abundances occurred at each of the sites except Wilson Wetland. These one-year spikes occurred in 2001 at Fish Creek, 2000 at Bonneyville Mills, and 2002 at Chapman Lake. These spikes in *Galerucella* spp. populations were followed by declined over the next several years. Past studies have shown that large populations of *Galerucella* spp. can decimate *L. salicaria* in an area (Landis 2003). Once this happens, *Galerucella* spp. are forced to disperse to new locations in order to find additional food plants. This is likely occurring at these sites, i.e., several years after release the number of *Galerucella* spp. is sufficiently high to provide substantial control of *L. salicaria* but this control forces the population to disperse to a new area.

CONCLUSIONS

Considering the changes at each of the four sites several conclusions can be drawn. First, not all sites show the same response to *Galerucella* spp. over the 8–10 year time frame. These variations are likely due to subtle differences between sites, which may be biotic, abiotic, or anthropomorphic differences. Despite these differences, the overall trends remain. Species richness generally increased each year after *Galerucella* spp. were released. Similarly, *L. salicaria* stem density, percent cover, and number of inflorescence decreased over time. The general trends suggest that the impact of *Galerucella* spp. is substantial, though the level of control and the impacts of the beetles may be variable between sites.

The one impact which was significant at all sites was a reduction in flowering. This also is the most visible change, as the absence of large purple spike inflorescence is a dramatic change from uncontrolled populations. Early studies of biological control insects suggested that a combination of several insects may best control *L. salicaria* (Malecki et al. 1993). However, one of the approved biological control agents is *Nanophyes marmoratus*, a flower weevil. Considering the impact *Galerucella* spp. had on flower production, significant populations of both insects are likely not sustainable within a wetland.

Finally, our study of biocontrol of *L. salicaria* by *Galerucella* spp. adds a longer time frame than most previous investigations and demonstrates a significant inhibition of vegetative growth and reproductive effort at each of the wetlands in this study. However, *L. salicaria* was more inhibited at some sites than others. In addition to reducing the vigor of *L. salicaria*, species richness generally increased. These negative effects on *L. salicaria*, while important for restoring wetland ecosystem function, did not eliminate this invasive species from these sites.

LITERATURE CITED

Anderson, M.G. 1995. Interactions between *Lythrum salicaria* and native organisms: a critical review. Environmental Management 19:225–231.

Bärlocher, F. & N.R. Biddiscombe. 1996. Geratology and decomposition of *Typha latifolia* and *Lythrum salicaria* in a freshwater marsh. Archiv fuïr Hydrobiologie 136:309–325.

Blossey, B. 1994. Purple loosestrife monitoring protocol. Department of Natural Resources, Cornell University. Unpublished.

Blossey, B., D. Schroeder, S.D. Hight & R.A. Malecki. 1994. Host specificity and environmental impact of two leaf beetles (*Galerucella calmariensis* and *G. pusilla*) for biological control of purple loosestrife (*Lythrum salicaria*). Weed Science 42:134–140.

Blossey, B. & L. Skinner. 2000. Design and importance of post-release monitoring. Pp. 693–706. *In* Proceedings of the X International Symposium on Biological Control of Weeds. (N.R. Spencer, Ed.). Montana State University, Bozeman, Montana.

Blossey, B., L.C. Skinner & J. Taylor. 2001. Impact and management of purple loosestrife (*Lythrum salicaria*). Biodiversity and Conservation 10:1787–1807.

Brown, B. & R. Mitchell. 2001. Competition for pollination: effects of pollen of an invasive plant on seed set of a native congener. Oecologia 129:43–49.

Brown, B.J., R.J. Mitchell & S.A. Graham. 2002. Competition for pollination between an invasive species (purple loosestrife) and a native congener. Ecology 83:2328–2336.

Chun, Y.J., C.G. Kim & K.A. Moloney. 2010. Comparison of life history traits between invasive and native populations of purple loosestrife (*Lythrum salicaria*) using nonlinear mixed effects model. Aquatic Botany 93:221–226.

Da Silva, E.M. & R.D. Sargent. 2011. The effect of invasive *Lythrum salicaria* pollen deposition on seed set in the native species *Decodon verticillatus*. Botany 89:141–146.

Dech, J.P. & P. Nosko. 2002. Population establishment, dispersal, and impact of *Galerucella pusilla* and *G. calmariensis*, introduced to control purple loosestrife in Central Ontario. Biological Control 23:228–236.

Emery, S.L. & J.A. Perry. 1996. Decomposition rates and phosphorus concentrations of purple loosestrife *Lythrum salicaria* and cattail *Typha* spp. Hydrobiologia 323:129–138.

Farnsworth, E.J. & D.R. Ellis. 2001. Is purple loosestrife (*Lythrum salicaria*) an invasive threat to freshwater wetlands? Conflicting evidence from several ecological metrics. Wetlands 21:199–209.

Gabor, T.S., T. Haagsma & H.R. Murkin. 1996. Wetland plant responses to varying degrees of purple loosestrife removal in southeastern Ontario, Canada. Wetlands 16:995–98.

Gaudet, C.L. & P.A. Keddy. 1995. Competitive performance and species distribution in shoreline plant communities: a comparative approach. Ecology 76:280–291.

Grevstad, F.S. 2006. Ten-year impacts of biological control agents *Galerucella pusilla* and *G. calmariensis* (Coleoptera: Chrysomelidae) on purple loosestrife (*Lythrum salicaria*) in Central New York State. Biological Control 39:1–8.

Grout, J.A., C.D. Levings & J.S. Richardson. 1997. Decomposition rates of purple loosestrife (*Lythrum salicaria*) and Lyngbyei's Sedge (*Carex lyngbyei*) in Fraser River Estuary. Estuaries and Coasts 20:96–102.

Hager, H.A. & R.D. Vinebrooke. 2004. Positive relationships between invasive purple loosestrife (*Lythrum salicaria*) and plant species diversity and abundance in Minnesota wetlands. Canadian Journal of Botany 82:763–773.

Hight, S.D. & J.J. Drea. 1991. Prospects for a classical biological control project against purple loosestrife (*Lythrum salicaria* L.). Natural Areas Journal 11:151–157.

Landis, D.A. 2003. Establishment and impact of *Galerucella calmariensis* L. (Coleoptera: Chrysomelidae) on *Lythrum salicaria* L. and associated plant communities in Michigan. Biological Control 28:78–91.

Lor, S. 2000. Population status and breeding biology of marsh birds in Western New York. [thesis], Ithaca (NY), Cornell University.

Mal, T.K., J. Lovet-Doust & L. Lovet-Doust. 1997. Time-dependent competitive displacement of *Typha angustifolia* by *Lythrum salicaria*. Oikos 79:26–33.

Mal, T.K., J. Lovet-Doust, L. Lovet-Doust & G. Mulligan. 1992. The biology of Canadian weeds. 100. *Lythrum salicaria*. Canadian Journal of Plant Science 72:1305–1330.

Malecki, R.A., B. Blossey, S.D. Hight, D. Schroeder, L.T. Kok & J.R. Coulson. 1993. Biological control of purple loosestrife. BioScience 43:680–686.

Piper, G. 1996. Biological control of the wetlands weed purple loosestrife (*Lythrum salicaria*) in the Pacific northwestern United States. Hydrobiologia 340:291–294.

R Development Core Team. 2011. R: A Language and Environment for Statistical Computing. R Foundation for Statistical Computing, Vienna, Austria.

Rawinski, T.J. 1982. The ecology and management of purple loosestrife (*Lythrum salicaria* L.) in central New York. [thesis], Cornell University.

Rawinski, T.J. & R.A. Malecki. 1984. Ecological relationships among purple loosestrife, cattail and wildlife at the Montezuma National Wildlife Refuge. New York Fish and Game Journal 31:81–87.

Sarkar, D. 2008. Lattice: Multivariate Data Visualization with R. Springer, New York.

Schooler, S.S. & P.B. McEvoy. 2006. Relationship between insect density and plant damage for the golden loosestrife beetle, *Galerucella pusilla*, on purple loosestrife (*Lythrum salicaria*). Biological Control 36:100–105.

Skinner, L.C., W.J. Rendal & E.L. Fuge. 1994. Minnesota's purple loosestrife program: history, findings, and management recommendations. Special publication 145, Minnesota Department of Natural Resources.

Stamm-Katovich, E.J., R.L. Becker & D.W. Ragsdale. 1999. Effect of *Galerucella* spp. on survival of purple loosestrife (*Lythrum salicaria)* roots and crowns. Weed Science 47:360–365.

Stuckey, R.L. 1980. Distributional history of *Lythrum salicaria* (purple loosestrife) in North America. Bartonia 47:3–20.

Templer, P., S. Findaly & C. Wigand. 1996. Sediment chemistry associated with native and non-emergent macrophytes of Hudson River marsh ecosystem. Pp. 1–32. *In* Final Reports of the Tibor T. Polgar Fellowship Program, 1995. (J. Waldman, W. Nieder & E. Blair, Eds.). Hudson River Foundation, NY.

Weihe, P.E. & R.K. Neely. 1997. The effects of shading on competition between purple loosestrife and broad-leafed cattail. Aquatic Botany 59:127–138.

Weiher, E., I.C. Wisheu, P.A. Keddy & D.R. Moore. 1996. Establishment, persistence, and management implications of experimental wetland plant communities. Wetlands 16:208–218.

Welling, C.H. & R.L. Becker. 1990. Seed bank dynamics of *Lythrum salicaria* L.: implications for control of this species in North America. Aquatic Botany 38:303–309.

Whitt, M.B., H.H. Prince & R.R. Cox, Jr. 1999. Avian use of purple loosestrife dominated habitat relative to other vegetation types in a Lake Huron wetland complex. The Wilson Bulletin 11:105–114.

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FOOD HABITS OF BOBCATS IN INDIANA

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ABSTRACT. The food habits of bobcats (*Lynx rufus*) have been reported throughout North America but there are few published accounts from the midwestern United States where conversion of native habitats to agricultural use prompted historic declines in regional populations. We determined food habits of bobcats in Indiana by examining the stomach contents of 159 carcasses obtained primarily from collisions with vehicles and trap-related mortalities in 38 counties between 1990 and 2010. Thirty-eight stomachs were either empty or had only vegetation or woody debris. Mammalian prey was found in 94.2% of the remaining stomachs, whereas avian remains were present in 14 stomachs (11.6%). Leading prey items consumed year-round were eastern cottontail rabbits (*Sylvilagus floridanus*; 35.5% occurrence), small mammals (e.g., *Microtus* spp., *Peromyscus* spp.; 26.4% occurrence), and tree squirrels (e.g., *Sciurus niger*, *S. carolinensis*; 15.7% occurrence). White-tailed deer (*Odocoileus virginianus*) and muskrat (*Ondatra zibethicus*) were consumed exclusively in fall-winter, but seasonality of other prey species was not observed. Frequencies of most foods varied between sexes and among age classes but differences were not statistically significant. Bobcats in Indiana exploited a wide array of mammalian prey consistent with other studies throughout North America, and principal food items (e.g., rabbits, small mammals, sciurids) were equally as important in other midwestern states where diet has been assessed.

Keywords: Bobcat, diet, food habits, Indiana, lagomorphs, *Lynx rufus*

INTRODUCTION

Bobcats (*Lynx rufus*) are the most common and widely distributed felid endemic to North America (Anderson & Lovallo 2003). The species' historical range is vast, encompassing the continental United States and extending from central Mexico north into British Columbia (Young 1958; Hall 1981). Bobcats inhabit diverse environments (e.g., coniferous and broadleaf forests, grasslands, savannahs, deserts) as evidenced by their extensive geographic distribution. As opportunistic predators, the abundance and stability of available prey populations influence their use of habitats (Litvaitis et al. 1986; Koehler & Hornocker 1989). Their food habits have been described from different regions of North America (Rollings 1945; Pollack 1951; Progulske 1955; Fritts & Sealander 1978; Bailey 1979; Jones & Smith 1979; Maehr & Brady 1986; Knick 1990). Bobcats, as all Felidae, are obligate carnivores and prefer prey items weighing between 700 g and 5.5 kg (Rosenzweig 1966). As a whole, mammals comprise the bulk of their diet, and multiple sources (see reviews in Rolley 1987 and Anderson & Lovallo 2003) report lagomorphs (e.g., *Lepus* spp., *Sylvilagus* spp.) to be a common food item rangewide. Principal prey species, however, vary regionally: medium-sized rodents (e.g., *Neotoma* spp., *Sigmodon* spp.) dominate diets in southern states (Kight 1962; Beasom & Moore 1977; Miller & Speake 1978; Jones & Smith 1979) and white-tailed deer (*Odocoileus virginianus*) become more important at northern latitudes (Marston 1942; McCord 1974). Small mammals (e.g., *Microtus* spp., *Peromyscus* spp.) are regularly consumed and were the leading prey of bobcats in southern Illinois (Woolf & Nielsen 2002).

Bobcat populations in the midwestern United States experienced widespread regional declines due to unregulated take and conversion of native habitats to anthropogenic land uses, largely extensive row-crop agriculture, following European settlement (Erickson et al. 1981; Hamilton & Fox 1987; Rolley 1987). Published accounts of the species' food habits from this altered landscape are limited to Illinois (Woolf & Nielsen 2002) and Iowa (Brockmeyer & Clark 2007). Whitaker &

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Mumford (2009) provide the only known data from Indiana bobcats in which they describe the stomach contents of three individuals. Our purpose is to provide a comprehensive assessment of the food habits of bobcats in Indiana. We document principal prey items and seasonal variation in food consumption and compare our findings with studies conducted throughout the bobcat's range.

METHODS

Bobcat carcasses were collected from September 1990 through March 2010. Stomachs were removed by cutting the distal end of the esophagus and anterior portion of the duodenum and contents were emptied onto trays for examination. Most items were identified macroscopically but a dissecting microscope was often used to differentiate among cricetids. Each item was identified to the lowest level we were able to confirm, and diet was assessed using percent occurrence and percent volume. Due to small sample sizes, taxonomically and guild-related species were combined into prey groups (e.g., *Sciurus* spp., small mammals) for analysis. The number of individuals of each item present was noted when possible. The volume of each item in stomachs was estimated visually if $>$ 1 item was present. Each sample was assigned to one of two intervals to determine seasonal variation: spring-summer (1 April to 30 September) or fall-winter (1 October to 31 March). Bobcats were aged as juveniles ($<$ 1 year), yearlings (1–2 years), or adults ($>$ 2 years) based on tooth eruption patterns (Jackson et al. 1988), the presence of an open apical root foramen, or cementum annuli analysis of a lower canine (Matson's Laboratory, LLC; Milltown, Montana). We used chi-square (X^2) analyses to compare frequencies among sexes, age classes, and seasons.

RESULTS

Sample effort.—A total of 159 bobcat carcasses (40 adult males, 32 adult females, 22 yearling males, 19 yearling females, 20 juvenile males, 20 juvenile females, and five males and one female of undetermined age) were collected from 38 (41.3%) of 92 Indiana counties. Most originated from the southern half of the state, including 83 (52.2%) from seven counties (Gibson, Greene, Lawrence, Martin, Pike, Posey, Warrick) in southwestern Indiana (Fig. 1). Only eight samples, all of which were collected during fall-winter, came from six counties in the northern half of the state. Sources of carcasses included bobcats that had been struck by vehicles (n = 123), incidentally killed in traps legally set for other furbearers (n = 26), illegally shot (n = 6), struck by a train (n = 1), and died from undetermined causes (n = 3). Samples were unevenly distributed throughout the year with most (81.1%; n = 129) obtained during fall-winter months (Fig. 2). Although our study spanned nearly 20 years, most carcasses (81.1%; n = 129) were collected during the 42-month period from October 2006 through March 2010.

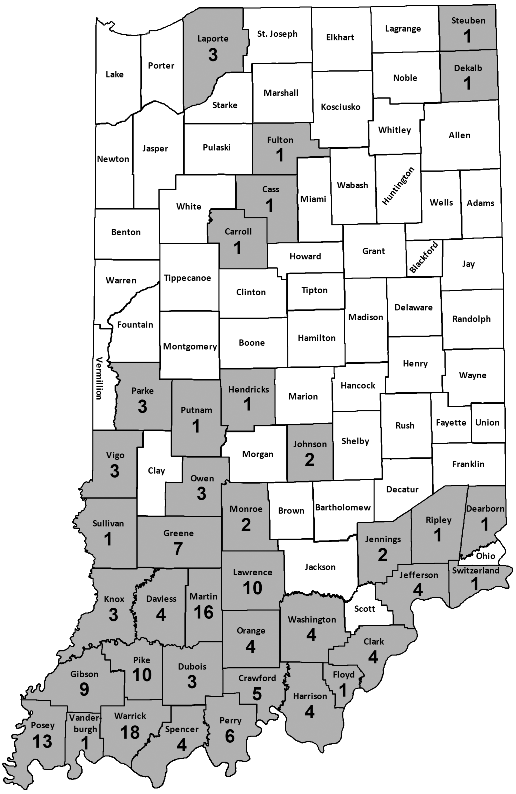

Figure 1.—Number of bobcat carcasses (n = 159) by county examined for stomach contents in Indiana, September 1990 through March 2010.

Thirty stomachs (18.9%), which included eight from spring-summer and 22 from fall-winter, were empty and excluded from further analysis. We omitted eight additional stomachs that contained only vegetation or woody debris that was likely ingested inciden-

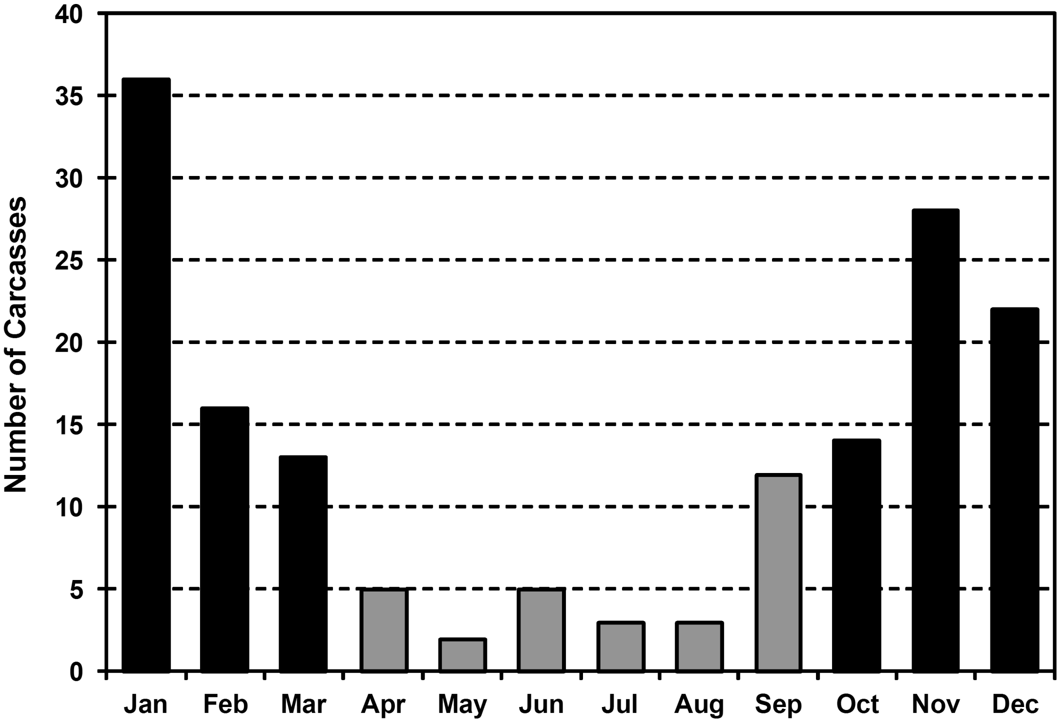

Figure 2.—Number of bobcat carcasses (n = 159) by month examined for stomach contents in Indiana, September 1990 through March 2010. Black bars denote fall-winter samples; gray bars denote spring-summer samples.

tally or consumed in trap-related mortalities. Thus, our final analysis is based on 121 carcasses composed of 64 males and 57 females, including 58 adults, 31 yearlings, 28 juveniles, and four not aged. Of these, 20 were obtained during spring-summer and 101 were obtained during fall-winter.

Prey items.—Mammalian prey comprised the bulk of the bobcat's diet in Indiana and was found in 94.2% (n = 114) of the stomachs (Table 1). Avian remains were present in 14 stomachs (11.6%). We found no evidence of other taxa (e.g., reptiles, amphibians, fish, insects). Although frequency of most species or prey groups appeared to vary between sexes (Fig. 3), the differences were not significant ($X^2 = 2.73$, df = 5, $P = 0.74$). There were no statistically significant age-related differences ($X^2 = 10.82$, df = 8, $P = 0.21$) among prey groups that were consumed by all three age classes (Fig. 4).

The eastern cottontail rabbit (*Sylvilagus floridanus*) was the most common food item in our study (Table 1). Overall, rabbit remains were found in 43 stomachs (35.5%) that comprised an average of 32.0% of the total volume and were the only prey animal in 27 stomachs (22.3%). Small mammals, as a group, were the second most prevalent food item; they were present in 32 stomachs (26.4%) that comprised an average of 18.3% of the total volume and were the only prey group in 20 (16.5%) stomachs. *Microtus* spp. and *Peromyscus* spp. were the most commonly taken small mammals, which included the prairie vole (*M. ochrogaster*), woodland vole (*M. pinetorum*), and white-footed mouse (*P. leucopus*). The meadow jumping mouse (*Zapus hudsonius*), southern bog lemming (*Synaptomys cooperi*), and Soricidae (shrews) were also identified. Sciurids (i.e., *Sciurus niger*, *S. carolinensis*) were found in 19 stomachs (15.7%) with a mean of 13.4% of the total volume. They were the only prey group found in 11 stomachs (9.1%). White-tailed deer was present in 15 stomachs (12.4%), of which 13 (86.7%) contained exclusively deer remains; percent volume averaged 11.5%. Muskrat

Table 1.—Contents of bobcat stomachs collected in Indiana, 1990–2010. Spring-Summer defined as April through September; Fall-Winter defined as October through March. Small mammals include *Peromyscus* spp., *P. leucopus*, *Microtus* spp., *M. ochrogaster*, *M. pinetorum*, *Zapus hudsonius*, *Synaptomys cooperi*, Soricidae (shrews), and unidentified small mammals. Sciurids include *Sciurus niger*, *S. carolinensis*, and unidentified sciurids. Birds include *Anas platyrhynchos*, *Fulica americana*, *Otus asio*, unidentified passerines, and unidentified birds.

	Spring-Summer (n = 20)		Fall-Winter (n = 101)		Combined (n = 121)	
Species or prey group	Percent occurrence	Mean percent volume	Percent occurrence	Mean percent volume	Percent occurrence	Mean percent volume
Mammals	95.0	93.5	94.1	92.4	94.2	92.6
Sylvilagus floridanus	45.0	41.9	33.7	30.0	35.5	32.0
Small mammals	25.0	13.7	26.7	19.2	26.4	18.3
Sciurids	35.0	28.0	11.9	10.5	15.7	13.4
Odocoileus virginianus	0.0	0.0	14.9	13.7	12.4	11.5
Ondatra zibethicus	0.0	0.0	5.9	5.4	5.0	4.5
unidentified mammal	10.0	10.0	14.9	13.5	14.0	12.9
Birds	10.0	6.5	11.9	7.6	11.6	7.4

(*Ondatra zibethicus*) was found in six stomachs (5.0%), all of which were adults, and was the only prey species in five (83.3%) of those samples. Mammalian remains that could not be identified to species or prey group because of their advanced stage of digestion were found in 17 stomachs (14.0%). Birds, as a group, were an occasional food of bobcats in Indiana. Remains, typically feathers, were present in 14 stomachs (11.6%) that comprised an average of 7.4% of the total volume. Passeriformes were the most common taxa among birds, although waterfowl (e.g., *Anas platyrhynchos*, *Fulica americana*) and a Strigiformes (i.e., *Otus asio*) were also noted.

Excluding stomachs with either deer or unidentifiable mammalian remains (n = 30), 65 of 91 stomachs (71.4%) contained a single species or prey group (e.g., small mammals, sciurids). Twenty-five additional stomachs (27.5%) had two prey groups whereas only one contained parts from three different groups (rabbit, small mammals, passerine). Number of individuals in each stomach averaged 1.7 ± 1.4 but most (64.8%) contained a single individual regardless of species or prey group. All stomachs that had squirrel remains (n = 19) contained only one individual. Similarly, 41 of 43 stomachs (95.3%) with rabbit parts had evidence of a single rabbit. In contrast, 22 of 32 (68.8%) stomachs that contained small mammals had > 1 individual ($\bar{x}$ = 2.6 ± 1.9; range = 1–8).

Seasonal variation.—Bobcats in Indiana consumed all species or prey groups in both seasons, except white-tailed deer and muskrat which were found only in stomachs from fall-winter months (14.9% and 5.9% of the seasonal sample, respectively; Table 1). There were no statistically significant differences (X^2 = 3.77, df = 3, P = 0.29) among prey groups that were consumed year-round, although sciurids and rabbits were present in a higher percentage of the stomachs in spring-summer than fall-winter (Table 1).

DISCUSSION

Our results are similar to many of the food habit studies that have been conducted throughout the geographic range of the bobcat. Mammals dominated the diet in Indiana and our three leading prey items (i.e., rabbits, small mammals, sciurids) were equally important in other midwestern states (Woolf & Nielsen 2002, Brockmeyer & Clark 2007). Cottontail rabbits were the principal prey in Indiana and lagomorphs (i.e., rabbits, hares, jackrabbits) have been the top food item for bobcat populations that inhabit vastly different North American environments (Korschgen 1957; Nussbaum & Maser 1975; Epperson 1978; Fritts & Sealander 1978; Bailey 1979; Story et al. 1982; Parker & Smith 1983; Litvaitis et al. 1986; Brockmeyer & Clark 2007). Rabbits comprised a smaller portion of the total diet in Indiana (35.5% occurrence) compared to studies in other midwestern and central plains states including Nebraska (68%, Epperson 1978), Iowa (60%, Brockmeyer & Clark 2007), and Missouri

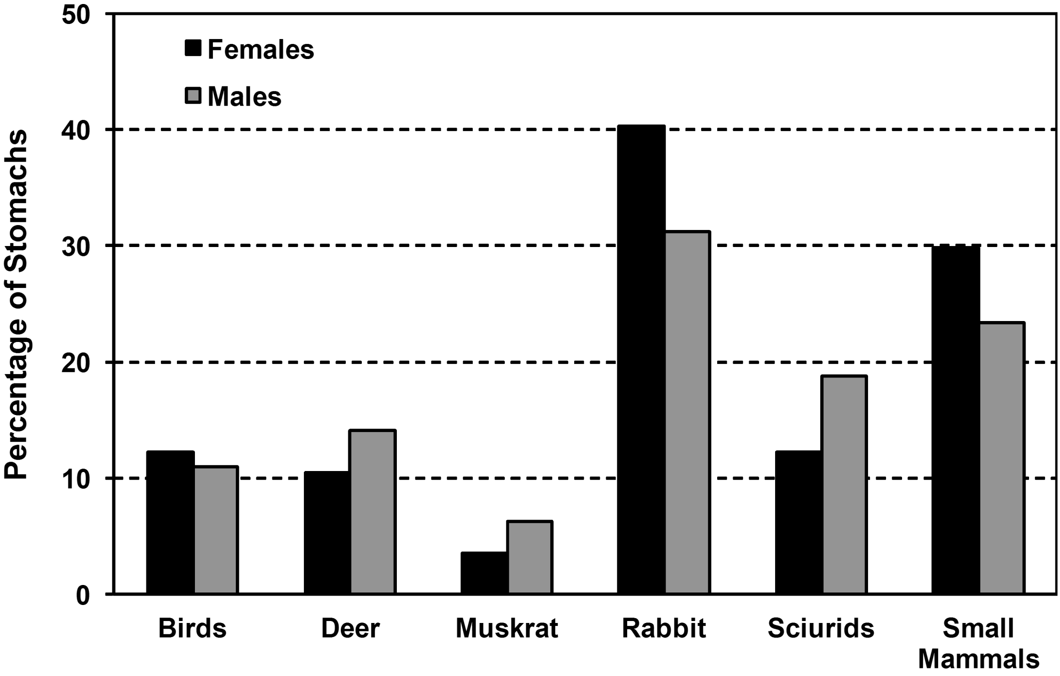

Figure 3.—Percentage of stomachs of female (n = 57) and male (n = 64) bobcats containing multiple species or prey groups in Indiana, September 1990 through March 2010.

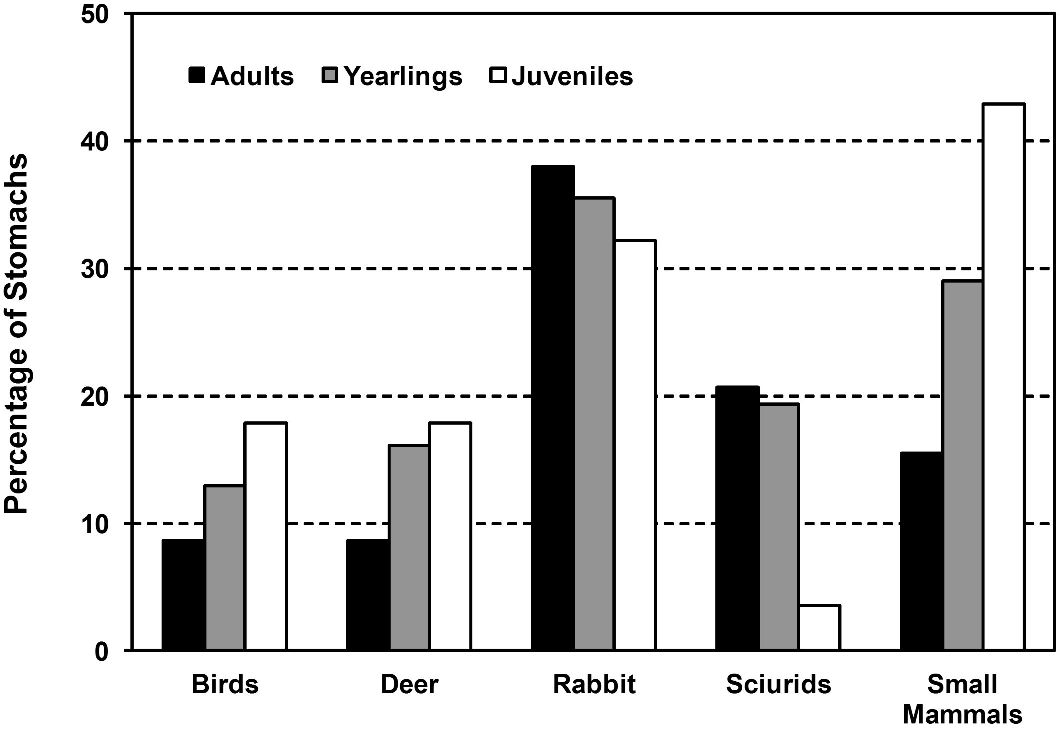

Figure 4.—Percentage of stomachs of adult (n = 58), yearling (n = 31), and juvenile (n = 27) bobcats containing multiple species or prey groups in Indiana, September 1990 through March 2010.

(52.2%, Korschgen 1957). Conversely, occurrence of rabbits in the diet of Indiana bobcats was greater than that reported in neighboring Illinois (22.7%, Woolf & Nielsen 2002).

Collectively, small mammals are a consistent element of bobcat diets and they comprised a relatively important prey group in Indiana (26.4% occurrence). Occurrence of small mammals in our study was slightly greater than reported for populations in Arkansas, (21.3%, Fritts & Sealander 1978) and Iowa (~20%, Brockmeyer & Clark 2007) whereas higher frequencies were noted in Idaho (*Microtus* spp. seasonal occurrence 40.0% and 65.2%, Koehler & Hornocker 1989), Illinois (32.8%, Woolf & Nielsen 2002), and Tennessee (*M. pinetorum* 27.3%, Story et al. 1982). *Microtus* spp. was frequently consumed although small mammals, as a group, provide little sustenance individually compared to larger items. Story et al. (1982) suggested voles are either easily captured or bobcats have developed efficient hunting strategies to warrant their pursuit. Our data support this contention and parallel data reported by Woolf & Nielsen (2002) in that most stomachs with small mammals (68.8% in Indiana, 51.9% in Illinois) commonly contained multiple individuals.

Frequency of tree squirrels in the diet of bobcats varies geographically and is uncommon or absent from arid southwestern sections of their range (Beasom & Moore 1977; Jones & Smith 1979; Delibes & Hiraldo 1987). Consumption of squirrels in Indiana (15.7% occurrence) was within the range (15–25%) reported for populations that occupy more forested areas (Progulske 1955; Korschgen 1957; Fritts & Sealander 1978; Litvaitis et al. 1984) including fragmented woodlands characteristic of midwestern landscapes (Woolf & Nielsen 2002; Brockmeyer & Clark 2007). Birds occur in most diet studies but typically constitute a minor component because they are usually active during the day which limits contact with primarily nocturnal predators (Tewes et al. 2002). Avian remains were found in 11.6% of bobcat stomachs in Indiana, similar to that reported in Illinois (10.1%, Woolf & Nielsen 2002) but considerably greater than Iowa (~2%, Brockmeyer & Clark 2007).

Previous studies (Fritts & Sealander 1978; Litvaitis et al. 1984) hypothesized age-related differences in bobcat diets may be due to inexperience, inadequate hunting skills, or variation in body mass that influences optimal prey size. Moreover, the diet of juveniles may be influenced by their mother, particularly for philopatric individuals that still occupy their natal range. Although differences were not significant, we found small mammals, an abundant and easily captured prey item, in the stomachs of more juveniles than adults (42.9% versus 15.5%). Conversely, adult bobcats preyed on tree squirrels, which probably demand advanced hunting skills, more frequently than juveniles (20.7% versus 3.6%). Similar age-related patterns for these two prey groups were reported from Iowa (Brockmeyer & Clark 2007).

Remains of white-tailed deer were found in a higher percentage of juveniles (17.9%) and yearlings (16.1%) than adults (8.6%), which differs from Litvaitis et al. (1984) and Brockmeyer & Clark (2007) in which juveniles consumed deer less often than older individuals. Bobcats are known to kill ungulates (Marston 1942; McCord 1974; Bailey 1979; Koehler & Hornocker 1989; Labisky & Boulay 1998), but the majority of deer eaten by bobcats is believed to represent carrion left from hunt-related losses, offal from field-dressed deer, road-kills, or winter starvation. Smaller and less experienced juveniles may be more apt than adult bobcats to take advantage of carrion, particularly in winter when other prey may be unavailable or less abundant. Although the few spring-summer samples limited our ability to detect seasonal differences in diet, only deer and muskrat were restricted to the fall-winter season. Further, 10 of the 15 stomachs (66.7%) with deer remains were collected from mid-November through December, a period that coincides with deer firearms season in Indiana.

The type and variety of prey consumed by bobcats are influenced by prey availability (Anderson & Lovallo 2003). It was not practical, however, to evaluate prey availability in our study because data were collected throughout the state during a 20-year span. As a result, spatial and temporal variation in prey populations was unknown as was their potential to influence bobcat habitat use. Food, however, was not believed to limit increases in bobcat populations in Illinois (Woolf & Nielsen 2002) or Iowa (Brockmeyer & Clark 2007), and results from our study support this premise. Overall, bobcats in Indiana exploited a wide array of prey consistent with earlier studies, particularly those conducted in midwestern states, and their diet did not vary based on sex, age class, or season. Primary prey species occur throughout Indiana and are common or abundant in suitable habitats (Simon et al. 2002, Whitaker & Mumford 2009). Most of our samples originated from southwest Indiana (Fig. 1) where the landscape is characterized by riparian woods and forested tracts interspersed among early-stage vegetation (e.g., grassy fields, shrubby areas, reclaimed strip mines) and cultivated fields. The mixture of open and forested habitats in this region likely supports a diverse and plentiful prey base needed to sustain viable bobcat populations in Indiana.

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LITERATURE CITED

Anderson, E.M. & M.J. Lovallo. 2003. Bobcat and lynx. Pp. 758–786. *In* Wild Mammals of North America: Biology, Management, and Conservation, 2nd ed (G.A. Feldhamer, B.C. Thompson & J.A. Chapman, eds.). Johns Hopkins University Press, Baltimore, Maryland.

Bailey, T.N. 1979. Den ecology, population parameters and diet of eastern Idaho bobcats. Pp. 62–69. *In* Bobcat Research Conference Proceedings: Current Research on Biology and Management of *Lynx rufus* (P.C. Escherich & L. Blum, eds.). Scientific and Technical Series 6. National Wildlife Federation, Washington, D.C.

Beasom, S.L. & R.A. Moore. 1977. Bobcat food habit response to change in prey abundance. The Southwest Naturalist 21:451–457.

Brockmeyer, K.J. & W.R. Clark. 2007. Fall and winter food habits of bobcats (*Lynx rufus*) in Iowa. Journal of the Iowa Academy of Science 114:40–43.

Delibes, M. & F. Hiraldo. 1987. Food habits of the bobcat in two habitats of the southern Chihuahuan Desert. The Southwestern Naturalist 32:457–461.

Epperson, C.J. 1978. The biology of the bobcat (*Lynx rufus*) in Nebraska. M.S. Thesis, University of Nebraska, Lincoln, Nebraska. 258.

Erickson, D.W., D.A. Hamilton & F.B. Samson. 1981. The status of the bobcat in Missouri. Transactions of the Missouri Academy of Science 15:49–59.

Fritts, S.H. & J.A. Sealander. 1978. Diets of bobcats in Arkansas with special reference to age and sex differences. Journal of Wildlife Management 42:533–539.

Hall, E.R. 1981. The Mammals of North America, 2nd ed. John Wiley and Sons, New York, New York. 1181.

Hamilton, D.A. & L.B. Fox. 1987. Wild furbearer management in the midwestern United States. Pp. 1100–1115. *In* Wild Furbearer Management and Conservation in North America (M. Novak, J.A. Baker, M.E. Obbard & B. Malloch, eds.). Ontario Ministry of Natural Resources, Toronto.

Jackson, D.L., E.A. Gluesing & H.A. Jacobson. 1988. Dental eruption in bobcats. Journal of Wildlife Management 52:515–517.

Jones, J.J. & N.S. Smith. 1979. Bobcat density and prey selection in central Arizona. Journal of Wildlife Management 43:666–672.

Kight, J. 1962. An ecological study of the bobcat *Lynx rufus* (Schreber) in west-central South Carolina. M.S. Thesis, University of Georgia, Athens, Georgia. 52.

Knick, S.T. 1990. Ecology of bobcats relative to exploitation and a prey decline in southeastern Idaho. Wildlife Monograph No. 108. 42.

Koehler, G.M. & M.G. Hornocker. 1989. Influences of seasons on bobcats in Idaho. Journal of Wildlife Management 53:197–202.

Korschgen, L.J. 1957. Food habits of coyotes, foxes, house cats and bobcats in Missouri. Missouri Conservation Commission Fish Game Division. P-R Series 15.

Labisky, R.F. & M.C. Boulay. 1998. Behaviors of bobcats preying on white-tailed deer in the Everglades. American Midland Naturalist 139:275–281.

Litvaitis, J.A., J.A. Sherburne & J.A. Bissonette. 1986. Bobcat habitat use and home range size in relation to prey density. Journal of Wildlife Management 50:110–117.

Litvaitis, J.A., C.L. Stevens & W.W. Mautz. 1984. Age, sex, and weight of bobcats in relation to winter diet. Journal of Wildlife Management 48:632–635.

Maehr, D.S. & J.R. Brady. 1986. Food habits of bobcats in Florida. Journal of Mammalogy 67:133–138.

Marston, M.A. 1942. Winter relations of bobcats to white-tailed deer in Maine. Journal of Wildlife Management 6:328–337.

McCord, C.M. 1974. Selection of winter habitat by bobcats (*Lynx rufus*) on the Quabbin Reservation, Massachusetts. Journal of Mammalogy 55:428–437.

Miller, S.D. & D.W. Speake. 1978. Prey utilization by bobcats on quail plantations in southern Alabama. Proceedings from the Annual Conference of the Southeast Fish and Wildlife Agencies 32:100–111.

Nussbaum, R.A. & C. Maser. 1975. Food habits of the bobcat (*Lynx rufus*) in the Coast and Cascade Ranges of western Oregon in relation to present management policies. Northwest Science 49:261–266.

Parker, G.R. & G.E. Smith. 1983. Sex- and age-specific reproductive and physical parameters of the bobcat (*Lynx rufus*) on Cape Breton Island, Nova Scotia. Canadian Journal of Zoology 61:1771–1782.

Pollack, E.M. 1951. Food habits of the bobcat in the New England states. Journal of Wildlife Management 15:209–213.

Progulske, D.R. 1955. Game animals utilized as food by bobcat in the southern Appalachians. Journal of Wildlife Management 19:249–253.

Rolley, R.E. 1987. Bobcat. Pp. 670–681. *In* Wild Furbearer Management and Conservation in North America (M. Novak, J.A. Baker, M.E. Obbard & B. Malloch, eds.). Ontario Ministry of Natural Resources, Toronto.

Rollings, C.T. 1945. Habits, foods and parasites of the bobcat in Minnesota. Journal of Wildlife Management 9:131–145.

Rosenzweig, M.L. 1966. Community structure in sympatric carnivora. Journal of Mammalogy 47:602–612.

Simon, T.P., J.O. Whitaker, Jr., J.S. Castrale & S.A. Minton. 2002. Revised checklist of the vertebrates of Indiana. Proceedings of the Indiana Academy of Science 111:182–214.

Story, J.D., W.J. Galbraith & J.T. Kitchings. 1982. Food habits of bobcats in eastern Tennessee. Journal of the Tennessee Academy of Science 57:29–32.

Tewes, M.E., J.M. Mock & J.H. Young. 2002. Bobcat predation on quail, birds, and mesomammals. Pp. 65–70. *In* Quail V: Proceedings of the Fifth National Quail Symposium (J. DeMaso, W.P. Kuvlesky, Jr., F. Hernández & M.E. Burger, eds.). Texas Parks and Wildlife Department, Austin, Texas.

Whitaker, J.O. & R.E. Mumford. 2009. Mammals of Indiana. Indiana University Press, Bloomington, Indiana. 661.

Woolf, A. & C. Nielsen. 2002. The bobcat in Illinois. Special Report Cooperative Wildlife Research Laboratory, Southern Illinois University, Carbondale, Illinois.

Young, S.P. 1958. The bobcat of North America. Wildlife Management Institute, Washington, D.C. 193.

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SEX RATIOS OF THE BIG BROWN BAT, *EPTESICUS FUSCUS*, AT AN URBAN-RURAL INTERFACE

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ABSTRACT. Wildlife responds to urbanization in a variety of ways. Some species, including the big brown bat (*Eptesicus fuscus*), take advantage of anthropogenic landscapes and can thrive in association with humans. The species is often found in association with humans, and is known to exploit urban environments. Females of many bat species, including the big brown bat, are sexually segregated during summer when females roost communally and males individually. The purpose of this study was to examine if there is gender bias in the distribution of this otherwise ubiquitous species across an urban/rural interface associated with conservation lands owned by the Indianapolis International Airport. Using a long-term data set, we compared sex ratios of big brown bats captured from a rural area south of Interstate 70 to the more urbanized northern region north of Interstate 70. Both areas were dominated by female big brown bats, but a greater proportion of males were captured in the rural area.

Keywords: Urbanization, big brown bat, *Eptesicus fuscus*, sex ratio, habitat

INTRODUCTION

Urbanization is known to affect wildlife in different ways (McKinney 2002; Duchamp & Swihart 2008). Many species can be negatively impacted by human development (McKinney 2006), while some species are known to take advantage of man-made structures (i.e., dwellings, outbuildings) and act as exploiters and/or adapters (Whitaker & Gummer 1992; Ordenana et al. 2010). While several studies have focused on community diversity and how urbanization impacts large portions of local fauna (Kurta & Teramino 1992; Sparks et al. 1998; Gehrt & Chelsvig 2004; Whitaker et al. 2004; Ulrey et al. 2005; Marchetti et al. 2006; Ordenana et al. 2010), there is limited research explaining how urbanization and urban sprawl affect the sexual distribution of different wildlife.

Bat diversity can serve as a reliable indicator of habitat quality and level of disturbance (Medellín et al. 2000). Some species thrive in an anthropogenically-disturbed environment (Gehrt & Chelsvig 2004; Oprea et al. 2009), while others are rarely found in association with humans. The big brown bat (*Eptesicus fuscus*) is relatively adaptable to human presence and development (Williams & Brittingham 1997; Duchamp et al. 2004; Neubaum et al. 2007), and is often found using human-made structures such as homes, barns, and outbuildings (Whitaker & Gummer 1992; Duchamp et al. 2004; Whitaker et al. 2004).

The Indianapolis International Airport (IND) began funding annual bat assessments as early as 1991 as part of mitigation for the federally endangered Indiana myotis (*Myotis sodalis*). Associated with additional airport development in 2001, a Habitat Conservation Plan (HCP) was designed (by American Consulting, Inc. in concert with the Indianapolis Airport Authority, IAA), approved by U.S. Fish & Wildlife Service, and implemented shortly thereafter. Due to the consistency of net site protocol since the HCP began, IAA has much data on the distribution, abundance, and richness of the bat species at this urban-rural study site (Whitaker et al. 2004; Ulrey et al. 2005; Damm et al. 2011; Whitaker et al. 2011), as well as bat foraging (Duchamp et al. 2004; Sparks et al. 2005a, 2005b; Walters et al. 2007) and roosting habits (Ritzi et al. 2005; Whitaker et al. 2006).

None of the prior studies examined whether urbanization influences sex ratios among bats

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captured at the site. Big brown bats are a reasonable choice for such a study because unlike some other local species, there are no larger-scale differences amongst the sexes in migratory behavior (Whitaker et al. 2007), and many individuals are captured each year (Whitaker et al. 2004).

METHODS

Study area.—The Indianapolis International Airport (IND; 39°42′57″N, 86°16′07″W) is situated on the southwestern edge of Indianapolis, a major US metropolis. The study area was located to the southwest of IND on lands purchased by the Indianapolis Airport Authority and was bordered by US Highway 40 and Indiana Highway 67 to the north and south, respectively (Fig. 1). Indiana Highway 267 bordered the study site to the west. Interstate Highway 70 (I-70) was chosen as a halfway point as it bisected the study site into northern and southern sections, with the area north of I-70 being more developed owing to an increasing warehouse district and the sample sites are immediately adjacent to the airport. The percentage of urban ground cover within 2 km of the net site to the north ranged from 27.6–43.1 percent (Table 1). The area south of I-70 is a matrix of agricultural and residential parcels with many small, scattered woodlots ranging approximately 30–40 ha in area. Urban ground cover in the south ranged from 4.4 to 19.4 percent. All 10 of the net sites used in this study were located along the East Fork of White Lick Creek (WLC), a medium-sized perennial stream which runs north to south through the study area. The terminal sites measure approximately 10.7 km apart. This stream bisects the study area from the east side of Mooresville in the south to the west side of Indianapolis to the north. The banks of WLC are mostly wooded, with the dominant woody species being box elder (*Acer negundo*), eastern cottonwood (*Populus deltoides*), hackberry (*Celtis occidentalis*), sycamore (*Platanus occidentalis*), green ash (*Fraxinus pennsylvanica*), and black walnut (*Juglans nigra*). Most open areas are either cultivated or developed. The woodlots that are not adjacent to the WLC are dominated by black walnut (*Juglans nigra*), bitternut hickory (*Carya cordiformis*), shagbark hickory (*Carya ovata*), shellbark hickory (*Carya laciniosa*), northern red oak (*Quercus rubra*), white oak (*Quercus alba*), sugar maple (*Acer saccharum*), honey locust (*Gleditsia triacanthos*), and American elm (*Ulmus americana*). As part of the airport's mitigation activities, properties have been purchased and small (30–40 acre) woodlots planted along the WLC.

Mist netting.—The bat community was sampled annually from 15 May–15 August of 2002 – present day. Data from 2002–2010 is used in this study. Mist-netting was conducted for two reasons: 1) to monitor and annually assess the overall bat community at the airport, and 2) to radio-tag Indiana myotis for roosting and foraging data. Standardized data taken from every bat included species, sex, reproductive status, length of right forearm, and body mass in grams. Each individual also received an individually numbered aluminum wing band (Porzana Ltd., United Kingdom) placed on the right or left forearm for male and female, respectively.

Netting sessions were conducted at 10 sites along White Lick Creek, four to the north and six to the south of I-70. Each site was sampled three times in a season. At each site, two mist nets were placed in such a way as to seal the flyway along the creek. All nets were set in place by dusk (approximately 2100 hr) and consisted of two and/or three 9 m × 2.6 m mist nets. Nets remained in place until at least 0115 hr, unless adverse weather required them to be taken down earlier.

Data analysis.—Sex ratios of the big brown bat were categorized based on sex and region in Microsoft Excel 2007. Ratios were then compared to look for differences in the number of each sex which could possibly be due to greater urbanization. Sex ratios for the big brown bat were compared using a G-Test for independence in program R v.2.13.0 (R Development Core Team). Recaptures were excluded from analyses. The G-Tests were run using code written by Peter Hurd (http://www.psych.ualberta.ca/~phurd/cruft/g.test.r) and were corrected using a William's correction. Similar G-Tests tests were also run to examine the difference within each sex in the two regions.

RESULTS

Female big brown bats were the dominant sex in nearly all years for both north and south (Fig. 2); in 2009 in the southern area both male and female totals equaled 24 individuals each (Table 2). The ratio of female to male big brown bats from 2002–2010 was 3.25:1 in the

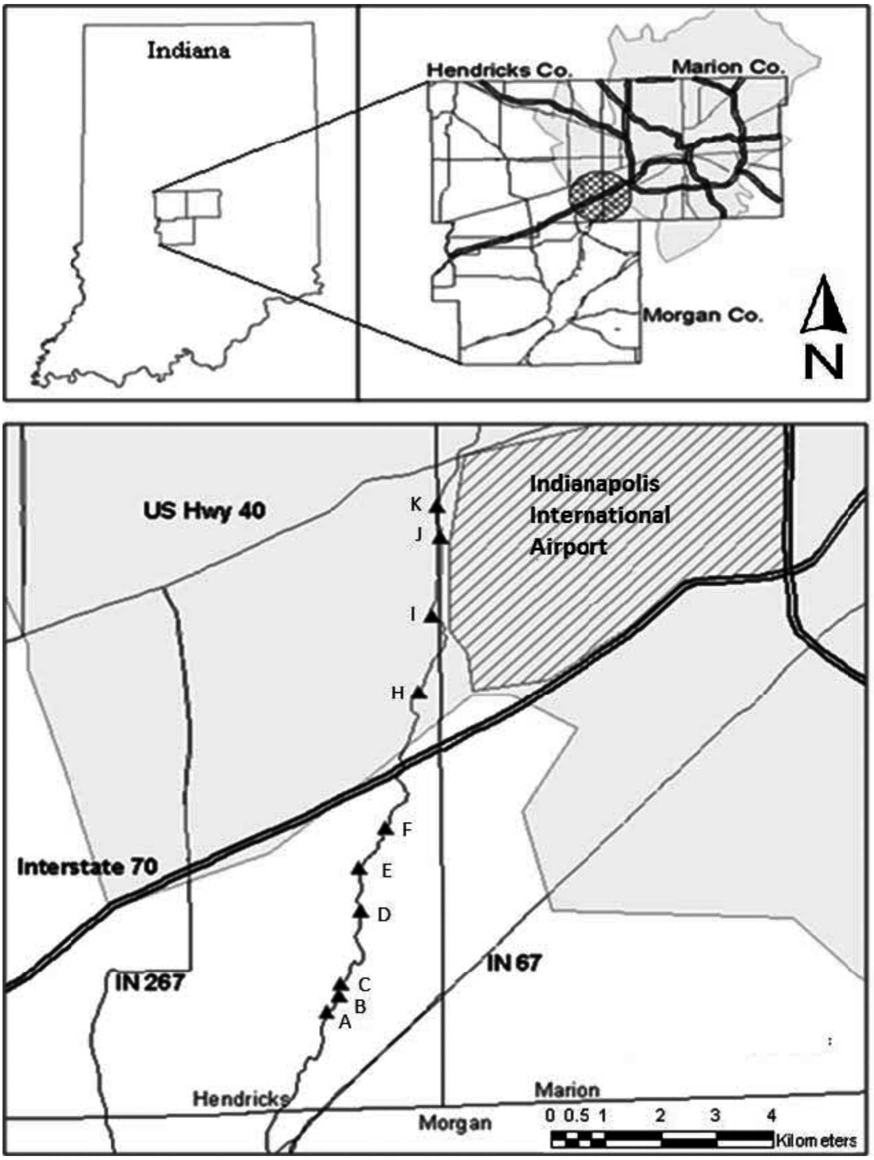

Figure 1.—Location of the study area within the state of Indiana (top left) and greater Indianapolis Metroplex (top right). Bottom shows an overview of the study area, with major roads and the East Fork of White Lick Creek. Net sites are denoted by black triangles. Thatched area represents the Indianapolis International Airport.

northern areas and 2.18:1 in the south. A total of 332 (76.5%) females and 102 (23.5%) males were captured in the north, and 325 (68.6%) females and 149 (31.4%) males were tallied in the south.

There was a significant female bias in all captures throughout the study area (G = 7.15, *d.f.* = 1, p = 0.0075). Comparatively, proportions of females within the urban north and rural south were similar (G = 0.075, *d.f.* = 1, p = 0.78), but males were proportionally more abundant in the south (G = 8.84, *d.f.* = 1, p = 0.0030).

DISCUSSION

To date, few studies have examined possible differences in sex ratio of vertebrates between an urban and rural area. These results show

Table 1.—Percent of urban and forested ground cover within 2-kilometers of each net site at the study area near the Indianapolis International Airport, Indianapolis, Indiana.

Net site	Percent urban	Percent forested
A	9.8	29.9
B	7.7	29.6
C	9.3	29.9
D	10.0	21.7
E	4.4	28.1
F	19.4	17.7
H	34.7	15.4
I	30.5	8.8
J	27.6	17.4
K	43.1	18.3

that at this location, urban landscape is a significant variable affecting the number of males present, while female ratios of this species did not change. This differs from the results of Kurta & Teramino (1992), who observed no difference in sex ratio with this same species in the Detroit, Michigan area. In another study, Kurta & Matson (1980) found that in Michigan, there was a significantly greater number of *E. fuscus* males than females. They attributed this to longer lifespan for males. Many members of this species seldom move very far from their natal colony (Mills et al. 1975), which implies that the ratios shown are reasonably representative of our study site.

Female big brown bat captures were approximately the same in both the urbanized north and more rural south areas of the project site. Female big brown bats often form maternity colonies in great numbers (Kurta & Baker 1990; Whitaker & Mumford 2009). The presence of maternity colonies in both the rural and urban portions of this study area (Duchamp et al. 2004; Whitaker et al. 2004) likely explains the overall

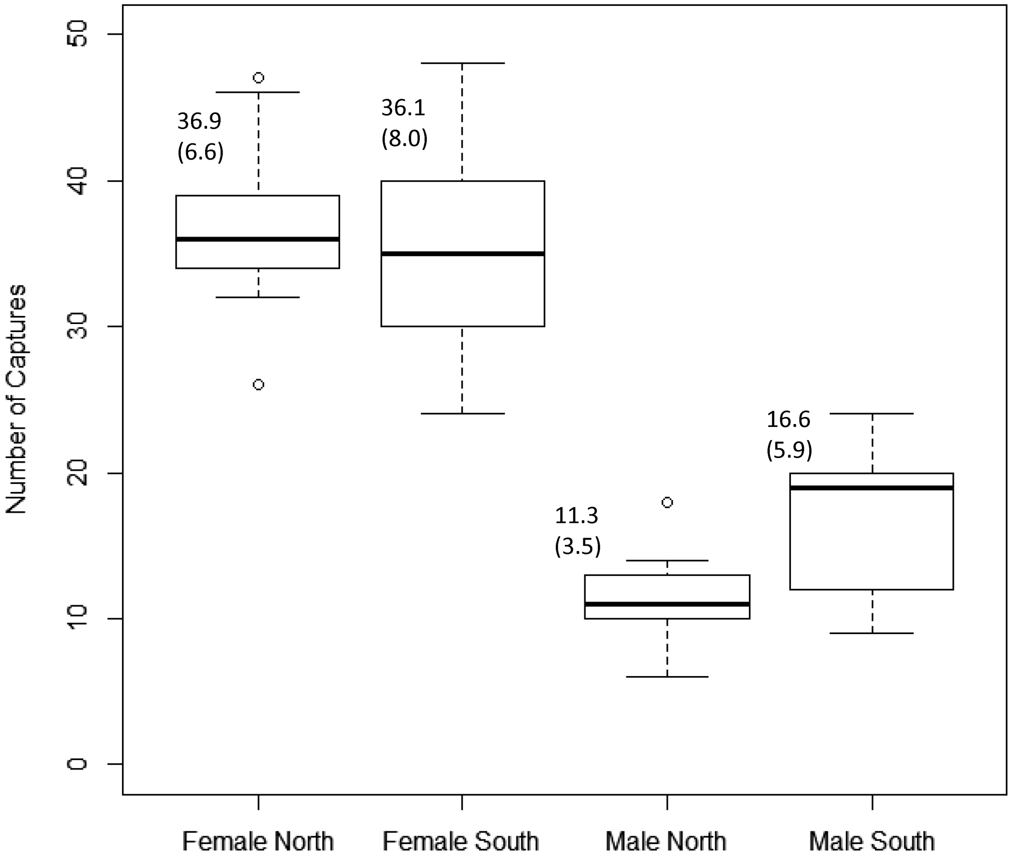

Figure 2.—Boxplot representing the mean number of male and female *Eptesicus fuscus* captures in the north and south regions of the Indianapolis International Airport Conservation Properties. Hollow circles represent outliers. Numbers represent the mean captures per year, with standard deviation in parentheses.

Table 2.—Number of male and females big brown bats captured from 2002 through 2010 in the north and the south portions of the study area near the Indianapolis International Airport, Indianapolis, Indiana.

		Year									
		2002	2003	2004	2005	2006	2007	2008	2009	2010	Total
Female	North	26	35	39	46	32	36	47	37	34	332
	South	48	46	33	35	40	30	29	24	40	325
	Total	74	81	72	81	72	66	76	61	74	657
Male	North	6	18	12	14	11	8	10	10	13	102
	South	24	12	12	19	20	10	19	24	9	149
	Total	30	30	24	33	31	18	29	34	22	251
Total		104	111	96	114	103	84	105	95	96	908

bias toward females. Males are typically solitary in the summer, and thus may be spread more evenly throughout the area. Furthermore, lactation requires both a high caloric diet and ready access to water, which may bias females to foraging along WLC where both resources are abundant. Males, conversely, may be just as abundant but are able to exploit smaller foraging and drinking patches. Such a behavioral difference might help alleviate competition with both female big brown bats and the other eight species of bats that occur along WLC.

Of particular interest is the potential for these data to provide insight into changes in big brown bat distributions following the January 2011 detection of White-Nose Syndrome (WNS) in Indiana. This fungal disease has caused marked declines in many cave-hibernating bats across the eastern United States and adjacent Canada (Turner et al. 2011; Francl et al. 2012). Some big brown bats are known to be killed by WNS (Blehert et al. 2008), but summer capture rates indicate the species is able to persist after the arrival of this disease (Francl et al. 2012). To date gender-bias in mortality has not been explored, but a changing sex ratio at the Indianapolis Airport may be an early sign that disruptive, differential mortality exists. Conversely, the rapid disappearance of other species from the community may allow male big brown bats greater access to foraging and roosting areas associated with WLC.

Finally, many species have the ability to control the gender of their offspring by either producing more juveniles of one gender (i.e., a difference in the primary sex ratio) or by behavioral activities that ensure differential survival of one gender. Big brown bats produce twin pups and have a balanced primary sex ratio (Burnett & Kunz 1982). Females have been observed nursing both male and female juveniles (D. W. Sparks unpublished), and thus have the opportunity to provide differential levels of care. Such ability may prove important if the adult in question has a compromised ability to forage or care for young following infection with WNS.

ACKNOWLEDGMENTS

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LITERATURE CITED

Blehert, D.S., A.C. Hicks, M. Behr, C.U. Meteyer, B.M. Berlowski-Zier, E.L. Buckles, J.T.H. Coleman, S.R. Darling, A. Gargas, R. Niver, J.C. Okoniewski, R.J. Rudd & W.B. Stone. 2008. Bat white-nose syndrome: An emerging fungal pathogen? Science 323:227.

Burnett, C.D. & T.H. Kunz. 1982. Growth rates and age estimation in *Eptesicus fuscus* and comparison with *Myotis lucifugus*. Journal of Mammalogy 63:33–41.

Damm, J.P., K.R. Pearman, N.S. Gikas, D.W. Sparks & J.O. Whitaker, Jr. 2011. Mammals of the Indianapolis International Airport conservation properties, Hendricks County, Indiana, with county records. Proceedings of the Indiana Academy of Science 120:96–103.

Duchamp, J.E., D.W. Sparks & J.O. Whitaker, Jr. 2004. Foraging-habitat selection by bats at an urban-rural interface: comparison between a successful and less successful species. Canadian Journal of Zoology 82:1157–1164.

Duchamp, J.E. & R.K. Swihart. 2008. Shifts in bat community structure related to evolved traits and features of human-altered landscapes. Landscape Ecology 23:849–860.

Francl, K.E., W.M. Ford, D.W. Sparks & V. Brack, Jr. 2012. Capture and reproductive trends of summer bat communities in West Virginia: assessing the impact of white nose syndrome. Journal of Fish and Wildlife Management 3:33–42.

Gehrt, S.D. & J.E. Chelsvig. 2004. Bat activity in an urban landscape: species-specific patterns along an urban gradient. Ecological Applications 14:625–635.

Kurta, A. & R.H. Baker. 1990. Eptesicus fuscus. Mammalian Species 356:1–10.

Kurta, A. & J.O. Matson. 1980. Disproportionate sex ratio in the big brown bat (*Eptesicus fuscus*). American Midland Naturalist 104:367–369.

Kurta, A. & J.A. Teramino. 1992. Bat community structure in an urban park. Ecography 15:257–261.

Marchetti, M.P., J.L. Lockwood & T. Light. 2006. Effects of urbanization on California's fish diversity: differentiation, homogenization and the influence of spatial scale. Biological Conservation 127:310–318.

McKinney, M.L. 2002. Urbanization, biodiversity, and conservation. Bioscience 52:883–890.

McKinney, M.L. 2006. Urbanization as a major cause of biotic homogenization. Biological Conservation 127:247–260.

Medellín, R.A., M. Equihua & M.A. Amin. 2000. Bat diversity and abundance as indicators of disturbance in neotropical rainforests. Conservation Biology 14:1666–1675.

Mills, R.S., G.W. Barrett & M.P. Farrell. 1975. Population dynamics of the big brown bat (*Eptesicus fuscus*) in southwestern Ohio. Journal of Mammalogy 56:591–604.

Neubaum, D.J., K.R. Wilson & T.J. O'Shea. 2007. Urban maternity-roost selection by big brown bats in Colorado. Journal of Wildlife Management 71:728–736.

Oprea, M., P. Mendes, T.B. Vieira & A.D. Ditchfield. 2009. Do wooded streets provide connectivity for bats in an urban landscape?. Biodiversity and Conservation 18:2361–2371.

Ordenana, M.A., K.R. Crooks, E.E. Boydston, R.N. Fisher, L.M. Lyren, S. Siudyla, C.D. Haas, S. Harris, S.A. Hathaway, G.M. Turschak, A.K. Miles & D.H. Van Vuren. 2010. Effects of urbanization on carnivore species distribution and richness. Journal of Mammalogy 91:1322–1331.

Ritzi, C.M., B.L. Everson & J.O. Whitaker, Jr. 2005. Use of bat boxes by a maternity colony of Indiana myotis (*Myotis sodalis*). Northeasten Naturalist 12:217–220.

Sparks, D.W., J.A. Laborda & J.O. Whitaker, Jr. 1998. Bats of the Indianapolis International Airport as compared to a more rural community of bats at Prairie Creek. Proceedings of the Indiana Academy of Science 107:171–179.

Sparks, D.W., C.M. Ritzi, J.E. Duchamp & J.O. Whitaker, Jr. 2005a. Foraging habitat of the Indiana bat (*Myotis sodalis*) at an urban-rural interface. Journal of Mammalogy 86:713–718.

Sparks, D.W., C.M. Ritzi & B.L. Everson. 2005b. Nocturnal behavior and roosting ecology of a juvenile *Lasiurus cinereus* near Indianapolis, Indiana. Proceedings of the Indiana Academy of Science 114:70–72.

Turner, G.O., D.M. Reeder & J.T.H. Coleman. 2011. A five-year assessment of mortality and geographic spread of white-nose syndrome in North American bats and a look to the future. Bat Research News 52:13–27.

Ulrey, W.A., D.W. Sparks & C.M. Ritzi. 2005. Bat communities in highly impacted areas: comparing Camp Atterbury to the Indianapolis Airport. Proceedings of the Indiana Academy of Science 114:73–76.

Walters, B.L., C.M. Ritzi, D.W. Sparks & J.O. Whitaker, Jr. 2007. Foraging behavior of eastern red bats (*Lasiurus borealis*) at an urban-rural interface. American Midland Naturalist 157:365–373.

Whitaker, J.O. Jr. & S.L. Gummer. 1992. Hibernation of the big brown bat, *Eptesicus fuscus*, in buildings. Journal of Mammalogy 73:312–316.

Whitaker, J.O. Jr., B.L. Walters, J.P. Damm & N.S. Gikas. 2011. 2010 Monitoring program for the Indiana myotis (*Myotis sodalis*) near the site of the future Six Points Interchange in Hendricks and Marion Counties, Indiana as required under the Six Points Interchange Habitat Conservation Plan. Submitted to BAA Indianapolis for the Indianapolis Airport Authority, Indianapolis International Airport.

Whitaker, J.O. Jr., V. Brack, Jr., D.W. Sparks, J.B. Cope & S. Johnson. 2007. Bats of Indiana. Publication number 1. Indiana State University, Center for North American Bat Research and Conservation. 59 pp.

Whitaker, J.O. Jr. & R.E. Mumford. 2009. Mammals of Indiana Indiana University Press, Bloomington, Bloomington, Indiana. 661 pp.

Whitaker, J.O. Jr., D.W. Sparks & V. Brack, Jr. 2004. Bats of the Indianapolis International airport area, 1991–2001. Proceedings of the Indiana Academy of Science 113:151–161.

Whitaker, J.O. Jr., D.W. Sparks & V. Brack, Jr. 2006. Use of artificial roost structures by bats at the Indianapolis International Airport. Environmental Management 38:28–36.

Williams, L.M. & M.C. Brittingham. 1997. Selection of maternity roosts by big brown bats. Journal of Wildlife Management 61:359–368.

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2014. Proceedings of the Indiana Academy of Science 123(1):49–56

OCCURRENCE AND SUSPECTED FUNCTION OF PREMATERNITY COLONIES OF EASTERN PIPISTRELLES, *PERIMYOTIS SUBFLAVUS*, IN INDIANA

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ABSTRACT. During summer, some pregnant female Eastern Pipistrelles form colonies in buildings but most typically roost in clusters of live or dead leaves in trees. We provide evidence that some that ultimately roost in leaf clusters form temporary colonies in or on buildings in early spring, prior to moving to the leaf clusters where they give birth. We call these prematernity colonies, and define them as those formed for a short time following hibernation and before the bats move to their maternity roosts. Prematernity colonies form from late April to early May and individuals relocate to leaf clusters from late May to early June. The bats showed strong fidelity to prematernity roosts, returning annually. Time of occupancy during any one year averaged 26 days. Nine bats were radio-tracked during the transition from building to tree roosts and were found to use several different trees. Bats showed interannual fidelity to building roosts. Buildings may help colonies re-form after individuals migrate from their hibernacula. Also, they could provide a warmer or more stable microclimate for pregnant females.

Keywords: Bats, bat roosts, Eastern Pipistrelles, prematernity colonies

INTRODUCTION

Bats regularly roost in anthropogenic structures, with commensal roosting behavior in buildings becoming nearly obligate in some species. For example, *Myotis lucifugus* (LeConte) (Little Brown Myotis) and *Eptesicus fuscus* (Beauvois) (Big Brown Bat) often form maternity colonies in human structures (Whitaker & Gummer 1992, 1993). Presumably these species formed maternity colonies in trees prior to the arrival of Europeans. Benefits of buildings may include protection from the elements (including a warmer and more stable microclimate), as well as relative permanence as most buildings likely remain available for relatively long periods as compared to tree roosts. Of 401 bat roosts located in buildings in Indiana between 1987 and 1992 (Whitaker & Gummer 1993), 330 were of Big Brown Bats, 58 of Little Brown Myotis, 12 of Eastern Pipistrelles (*Perimyotis subflavus*) (F. Cuvier), and one of the Evening Bat (*Nycticeius humeralis* Rafinesque). This indicates that relatively few Eastern Pipistrelles use buildings as roost sites in Indiana. (Note—We have retained the common name Eastern Pipistrelle for this species because this name has long been in use and because there is already a tricolored bat, *Glyphonycteris sylvestris* (Phyllostomidae), of Central and South America (see Whitaker et al. 2011).

Many species of hibernating bats leave hibernacula in spring and move to their summer roost areas where females form maternity colonies and give birth. Little is known about their behavior during the period following hibernation and prior to forming maternity colonies. However, the females of many temperate bat species return to the same maternity roost structure each year, whether it be a tree (Indiana Myotis, *Myotis sodalis* Miller & Allen; Northern Myotis, *M. septentrionalis* Trouessart), a building (*Myotis lucifugus*, *Eptesicus fuscus*), or a cave or mine (Gray Bat, *Myotis*

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grisescens A. H. Howell) (Whitaker & Mumford 2009).

Eastern Pipistrelles roost primarily in clusters of dead leaves in trees in Indiana (Veilleux et al. 2003) and Arkansas (Perry & Thill 2007), although in Nova Scotia, Poissant et al. (2010) found them roosting exclusively in *Usnea trichodea* Ach., a lichen which typically occurs in conifers. Annual fidelity to summer (maternity) roost areas was reported for foliage roosting Eastern Pipistrelles by Veilleux & Veilleux (2004a). However, they exhibit fidelity to broad roost areas, rather than to a single roost (leaf cluster) site, since specific foliage roosts are ephemeral and may not be available across years (Veilleux & Veilleux 2004b).

Whitaker (1998) studied the life history patterns of six colonies of Eastern Pipistrelles in building roosts in Indiana. Five of the six served as maternity roosts in which bats had their young and remained for the majority of the maternity season. One roost (Jackson Roost in 1990) was abandoned before the maternity season and therefore, no young were produced at the site. Eight bats in the Jackson Roost first arrived on 21 May, and the colony increased to a maximum of 13 bats. Most bats left that colony by 23 June. We now suspect that this was a prematernity colony as described in this paper. We have since identified three additional prematernity roosts which were used only during spring, before bats moved to the more typical foliage roosts. The goal of this paper is to describe the behavior patterns we observed in these three roosts. The use of a prematernity roost has not been previously described. Specifically our goals were to determine the size of the colonies, when they form, when they relocate, whether the bats move to foliage roosts when they leave, and if they show fidelity to the prematernity roost site.

METHODS

We studied three Eastern Pipistrelle prematernity colonies in Indiana: the Parshall-Layton colony, the Lowry colony, and the Clifty Falls colony. The first two colonies contained only pregnant females, as determined by palpation. The Parshall-Layton colony was identified in 1995 in Seelyville, Vigo County. Individuals roosted along the rafters of an open porch/breezeway about 12 m high. The bats clustered in the open and usually roosted in the same specific location. The site was used by bats during each successive year until 2003 when nearly all the nearby trees (roost trees?) were removed and the bats apparently relocated elsewhere. The Lowry colony was observed in 2002, 2003, and 2008. The bats roosted in a small shed near a house in Clinton, Vermillion County. Bats had been present at the roost in early spring for at least three years prior to 2002 and had returned to the roost during each year through 2008. Most bats were usually in one main cluster. The Clifty Falls colony was discovered in 1999 at Clifty Falls State Park, Jefferson County. Individuals roosted under the eaves of the main building of the park nature center and have returned to the site during each year of the study.

The owners/occupants of each of the buildings watched for the bats each year and contacted us when they arrived. The authors conducted exit counts each evening until the bats relocated and residents continued to observe the roosts and informed us if the bats returned. The small size of the three colonies and the tendency to roost in well lit, accessible areas, allowed data collection regarding arrival and departure dates as well as direct counts of individuals during observation periods. Some bats from two of the colonies were banded to determine if they returned to the same roost during successive years.

To investigate where bats roosted after leaving the Lowry roost sites, 0.35 or 0.45 g radio transmitters (Model LB-2N or LB-2, Holohil Inc., Ontario, Canada or Model LTM, Titley Electronics, New Ballina, Australia) were used. A small amount of fur was trimmed from between the scapula using scissors, and the transmitter was affixed using a non-toxic, surgical adhesive (Skin-Bond, Smith+Nephew, Largo, Florida). A single individual from the Parshall-Layton colony was radio-tagged twice in 1998 and once in 2001, and nine individuals from the Lowry colony were radio-tagged in 2008. Transmitters averaged 6% of body mass. Using a VHF radio-receiver (Model TRX2000S, Wildlife Materials, Carbondale, Illinois) and a 3-element Yagi antenna, bats were tracked to their roosts on each day, until the transmitter battery failed or the transmitter fell from the bat.

RESULTS

The bats in the Parshall-Layton and Lowry colonies were all pregnant females, as indicated

Table 1.—Results of bat banding at the Parshall/Layton roost, Vigo County, Indiana: 1998–2003. N = new bat for year; R = return from previous year; BNR = bands not read; UBB= unbanded bats; NBNR = new banded bats, but no returns.

Band number	1998	1999	2000	2001	2002	2003
Y202	N	R	R	R	R	R
W1006		N		R		R
W1085		N		R		R
W1086		N		R		R
W1088		N		R		
BNR			5		4	1
UBB	6					
NBNR		2	2	4	4	4
N =	7	7	8	9	9	9

by palpating the abdomen. Bats in the Clifty Falls colony were not handled but were assumed to be pregnant females since their behavior was similar to that of the Parshall-Layton and Lowry colonies.

We monitored the Parshall-Layton colony (Table 1) for six years (1998 through 2003), although limited data were collected in 2002. Dates of first arrival ranged from 26 April to 18 May, with colony size ranging from 7 to 9 individuals (Fig. 1). Dispersal from the prematernity roost, presumably to tree roosts, ranged from 23 May to 12 June (no data for 2002 or 2003). Some individuals were absent for portions of each year; residence time (number of days between first arrival and latest occupancy) ranged from 20 to 32 days ($\bar{x}$ = 26 days). A single female from the Parshall-Layton roost (Y202) was banded on the forearm in 1998 with a plastic split-ring band (Table 1). An additional six bats were banded in 1999. Three of these individuals were recaptured at the prematernity roost in 2001 and 2003. Bat Y202 returned to the building roost for all six years of observation. This bat was radio-tracked in 1998 to a white oak tree (*Quercus alba* L.) located 600 m from the Parshall-Layton roost where it remained from

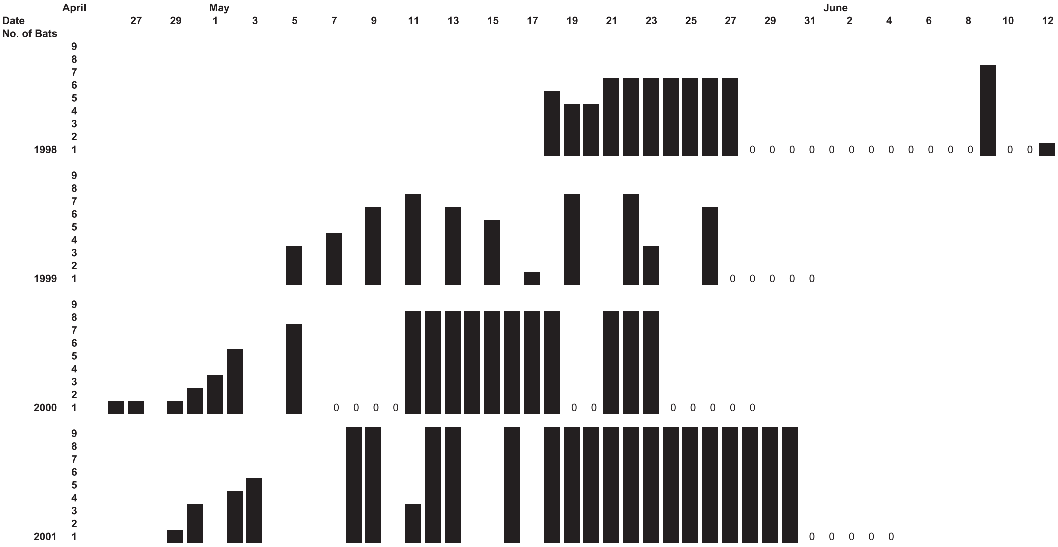

Figure 1.—Numbers of Eastern Pipistrelles on various dates at Parshall-Layton colony, Vigo County, Indiana: 1998–2001. A zero indicates no bats were present; a blank indicates no observation was made. This figure does not include the years 2002 and 2003 since complete population assessments were not made. Residents of the properties notified us when the bats arrived each year, and also watched to be sure the bats did not return after our last observations.

Table 2.—Results of bat banding at the Lowry roost, Clinton, Vermillion County, Indiana: 2002, 2003, and 2008. N = new bat for year, R = return from previous year.

Band number	2002	2003	2008
4001	N	R	
4002	N	R	R
4003	N		
4004	N	R	R
4005	N		
4006	N		
4007	N	R	R
4008	N	R	
4009	N		
4010	N		R
4012	N	R	
4013	N		
4014	N	R	
4201		N	R
4202–4206*		N (5)	
6151			N
6152			N
6155			N
6192			N
6200			N
N =	13	13	10

* These 5 bats were banded in 2003, but were not seen in 2008.

10 through 13 June. Emergence counts were conducted during this time and two to four bats were observed emerging from this tree. On each of those days, one of the emerging bats was the radio-tagged Eastern Pipistrelle Y202. This bat was radio-tagged again on 30 May 2001 and subsequently located on 31 May and 1 June, roosting in two different trees, each approximately 200 m from the white oak used in 1998. On 2 June, 2001, bat Y202 roosted in the white oak used in 1998.

At the Lowry roost, data were collected during three years (2002, 2003, and 2008; Table 2). Dates of first arrival ranged from 27 April to 27 May. Colony size ranged from 10 to 13 individuals. Dates of latest occupancy ranged from 25 May to 8 June. As in the Parshall-Layton colony, some individuals were absent for portions of 2003. Residence time ranged from five to 29 days ($\bar{x}$ = 16 days). A total of 13 individuals, appearing to comprise the entire colony, was banded in 2002 with seven returning in 2003 and five in 2008 (Table 2). On 27 May 2008, 11 females were present at the Lowry roost, including five banded in 2002 and 2003. Nine were radio-tagged (Table 3, Figs. 2, 3). One bat (6155) disappeared, and the remaining 8 were tracked for up to 6 days ($\bar{x}$ = 3.8, 1-6). Bat 6200 roosted for 2 days in a cluster of leaves in the top of a fallen red oak (*Quercus rubra* L.), then roosted in four different trees. Bat 6192 roosted two days in the shed, then one day in a tree, and bat 4201 two days in the shed, then four days in different trees. Bat 4010 roosted one day in the shed, and one day in a cluster of leaves in a small oak tree. Bat 4004 roosted in three separate days in three different oak trees. Another bat (4002) remained in the shed one night then roosted in a tree, whereas bat 4007 roosted two days in a cluster of leaves in an oak tree. Thus, the bats moved into clusters of leaves in trees not far from the prematernity shed.

Of these bats individuals were in the shed seven times, and all entered leaf clusters in trees on 20 occasions ($\bar{x}$ = 2.5, 1–6) within 200 m of the shed. The bats roosted in a total of 13 trees including red and white oak (*Quercus*), hickory (*Carya*), elm (*Ulmus*), maple (*Acer*), and one tulip poplar (*Liriodendron tulipifera* L.).

The bats have formed prematernity colonies at Clifty Falls since at least 1999 (Table 4), but data were collected only during three years (2001, 2002, and 2003). Other than one bat that appeared 28 April, dates of first arrival ranged between 1 and 20 May. Dates of latest occupancy were between 24 May and 8 June. Residence time ranged from 19 to 24 days ($\bar{x}$ = 22.5 days). These bats were not handled, not banded, and no radio-telemetry was conducted.

Dates of first arrival at the building roosts generally occurred between late April and late May and mean group size during this period was 11 bats (range 9–13). Residence time in prematernity roosts ranged from five to 32 days. Prematernity colonies generally relocated to tree roosts from late May to early June, with the latest observed relocation date of 25 June. We suspect that females were nearing parturition when they moved from the buildings to roost in foliage. The timing of these observations was similar to those for these bats in buildings as reported by Whitaker (1998).

DISCUSSION

The data indicate that during summer, some female Eastern Pipistrelles living in woods have

Table 3.—Eastern Pipistrelles (*Perimyotis subflavus*) radio-tagged 27 May 2008 from the Lowry colony in Clinton, Vermillion County, Indiana. X = not found that day; bat left, or radio lost. NV. = bat not actually viewed. Distance and direction from shed. Unid. = unidentified. Numbers in parentheses on the table correspond to tree numbers. It was not always possible to determine in exactly which tree a bat was roosting.

Bat	28 May 08	29 May 08	30 May 08	31 May 08	1 June 08	2 June 08
6200	NV. 150 m NE cluster of leaves in top of fallen white oak tree (10)	Same as on 28 May	NV. 90 m N bitternut hickory or red or white oak tree (7)	NV. 35 m NE in red or white oak tree (2)	NV. 195 m N in bitternut hickory tree (15)	NV. 100 m N in red or white oak tree (8)
6192	Shed	Shed	NV. 155 m N in bitternut hickory or white oak tree with A04201 (14)	Radio on ground near 30 May roost site		
4201	Shed	Shed	NV. 155 m N in bitternut hickory or white oak tree with A06192 (14)	NV. 145 m N in tulip poplar (13)	In tree in yard 200 m W of prematernity roost (16)	NV. 145 m N in bitternut hickory or white oak tree (12)
4010	Shed	NV. 185 m NE in small American elm trees (11)	Shed	Shed	X	X
4004	100 m NE in cluster of dead white oak leaves 2.5 m off ground in sm dead tree (9)	NV. 50 m N in bitternut hickory or white or red oak tree (4)	NV. 70 m N in bitternut hickory or sugar maple (6)	Radio on ground 50 m E		
4002	X	NV. 60 m N in bitternut hickory (5)	X	X	X	X
4007	95 m SE in cluster of dead red oak leaves in American elm 3 m off ground (1)	Same as 28 May (1)	X	X	X	X
6151	40 m NE in cluster of dead white oak leaves in sm tree 2.5 m off ground (roosting with 1 other bat) (3)	Same as 28 May (3)	X	X	X	\X
6155	X	X	X	X	X	X
Total # Bats in shed	6	4	2	2	0	0

Figure 2.—Eastern Pipistrelle A6151 roosting with another untagged Eastern Pipistrelle in a cluster of dead oak leaves about 2.5 m off the ground.

relatively long staging periods following emergence from hibernation and prior to entering leaf clusters. During this time they may form small temporary colonies (prematernity colonies) in or on buildings prior to moving to the clusters of dead leaves (foliage roost sites) in which they give birth. We believe that the main function of this behavior is to serve as a permanent gathering place for the bat colony because there is no main summer roost to return to since the clusters are temporary and do not remain in place from year to year. The leaf clusters utilized were located in the vicinity of the prematernity roost; all were within 200 m of the Lowery roost.

In addition, since warmer roost temperatures speed gestation (Racey 1973), it has been suggested that the function of the prematernity colonies might be to provide a more stable microclimate than the leaf clusters. However, the Parshall-Layton and Clifty Falls colonies are both exposed to the elements. The Lowry colony is located in a shed, thus the microclimate may be more stable there. The prematernity colonies are or will be maternity colonies, but differ in that they break up prior to using leaf clusters (Veilleux & Veilleux 2004b; Veilleux

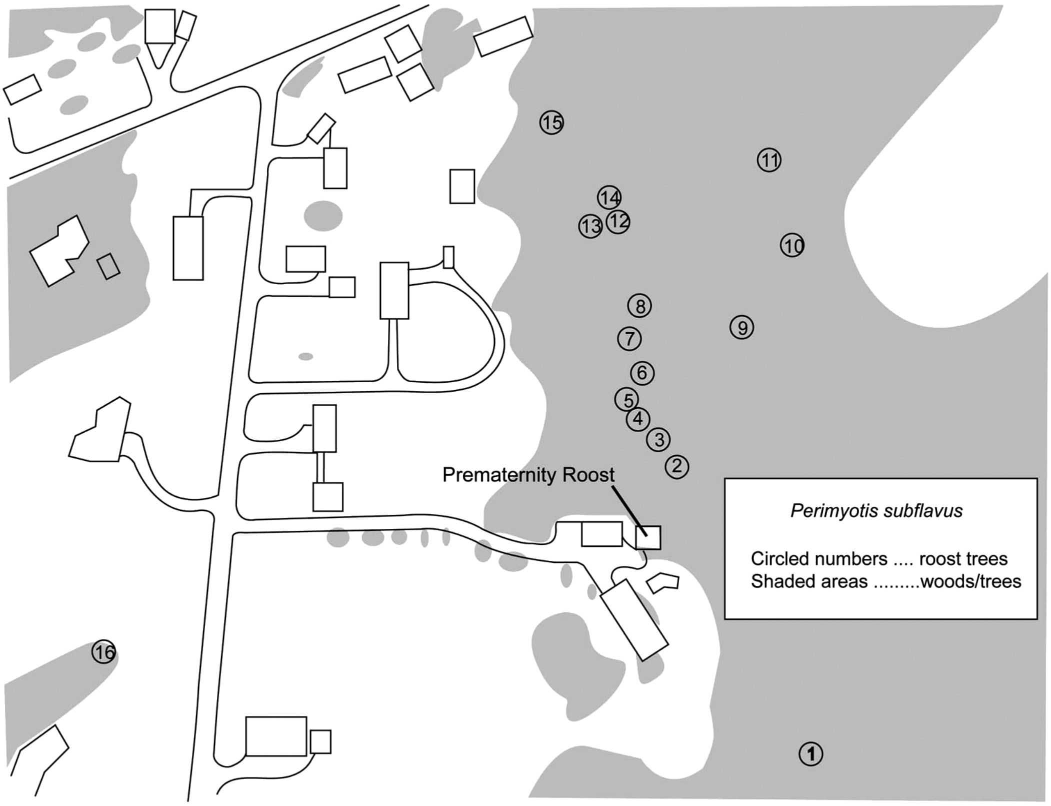

Figure 3.—Prematernity roost and roost trees of *Perimyotis subflavus* southwest of Clinton, Vermillion County, Indiana.

Table 4.—Observations of Eastern Pipistrelles at Clifty Falls State Park, Jefferson County, Indiana: 2001–2003. There was a single bat present on 28 April 2002. N = no observations.

		2001	2002	2003
May	1			2
	2			4
	3			5
	4			3
	5		5	N
	6		N	5
	7		8	4
	8		N	5
	9		N	6
	10		11	N
	11		11	5
	12		N	N
	13	0	N	N
	15	0	N	7
	16	0	N	10
	17	0	N	9
	18	0	N	5
	19	0	N	N
	20	5	11	N
	21	N	0	7
	22	N	0	12
	23	10		13
	24	0		0
	25	0		0
	26			
	27			
	28			
	29			
	30			
	31			
June	1			
	2			
	3	13		
	4			
	5	13		
	6			
	7			
	8	2		

et al. 2003). We know of no other bat species that behaves this way.

Whitaker (1998) reported parturition of Eastern Pipistrelles in maternity colonies in buildings occurring between 30 May and 11 July, with most young born between 12 June and 1 July. In Indiana, the earliest parturition in foliage roosting Eastern Pipistrelles was reported in late June (29 and 26 June, 1999 and 2000, respectively; Veilleux & Veilleux 2004b). The dates in buildings (Whitaker 1998) may be related to higher and more stable temperatures than are probably found in foliage roosts; Eastern Pipistrelles in foliage may receive higher exposure to the cooling effects of wind and rain (Veilleux et al. 2003).

Bats in prematernity roost sites showed annual fidelity to these sites. One bat banded in 1998 at the Parshall-Layton colony returned to the roost for five successive years and three bats banded in 1999 were later observed there in 2001 and 2003. At the Lowry colony, seven of 13 bats banded in 2002 returned in 2003, and four of the original bats were present at the roost five years later in 2008, along with one that was banded in 2003.

We suspect that originally these bats formed prematernity colonies using large trees or other environmental markers and then moved from there into nearby areas with ample leaf clusters where they had their young. With the advent of buildings, prematernity colonies became associated with these structures (inside or outside) and spread from there to nearby areas with leaf clusters. The Parshall-Layton prematernity colony disappeared in 2003, after the nearby trees were removed.

It is not known why these bats split up into the small groups in leaf clusters, but it is probably because clusters of leaves are so small as compared to buildings. It appears that the bats have evolved to fit leaf clusters, as suggested by their color which resembles that of dead leaves. The splitting of a colony into small numbers in leaf clusters could be a means of avoiding predation or a seeking of sites with favorable temperature and humidity conditions. Our samples are small, but there is no evidence that the bats in the prematernity colonies mix with pipistrelles forming maternity colonies in buildings.

Barclay & Kurta (2007) describe three broad types of roost switching by bats: episodic (between various life episodes such as hibernation to maternity colonies or to staging sites, or after the young become volant), emergency (to avoid a predator such as a raccoon or snake), and recurrent (such as back and forth from tree to tree). The prematernity colonies described here appear to be an example of an episodic roost switching behavior, with the prematernity colonies forming after hibernation and prior to the dispersal to foliage roost sites.

Big Brown and Little Brown bats may have entered buildings accidentally or while searching for places to roost and found them suitable as maternity roost sites, and they are now the bats that commonly roost in buildings. However, a few do form maternity colonies in buildings or under bridges. Eastern Pipistrelles may have taken a different approach to the adoption of roosts in buildings by gathering in or on buildings as prematernity colonies and then in some instances remaining in buildings as maternity roosts. It would be a simple behavioral change from forming prematernity colonies in buildings, to using buildings for production of young.

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LITERATURE CITED

Barclay, R.R. & A. Kurta. 2007. Ecology and behavior of bats roosting in tree cavities and under bark. Pp. 17–57. *In* Bats in Forests: Conservation and Management (M.J. Lacki, J.P. Hayes & A. Kurta, eds.). The Johns Hopkins University Press, Baltimore, Maryland.

Perry, R.W. & R.E. Thill. 2007. Tree roosting by male and female Eastern Pipistrelles in a forested landscape. Journal of Mammalogy 88:974–981.

Poissant, J.A., H.G. Broders & G.M. Quinn. 2010. Use of lichen as a roosting substrate by *Perimyotis subflavus*, the Tricolored Bat, in Nova Scotia. Écoscience 17:372–378.

Racey, P.A. 1973. Environmental factors affecting the length of gestation in heterothermic bats. Journal of Reproduction and Fertility, Supplement 19:175–189.

Veilleux, J.P. & S.L. Veilleux. 2004a. Intra-annual and interannual fidelity to summer roost areas by female Eastern Pipistrelles, *Pipistrellus subflavus*. American Midland Naturalist 152:196–200.

Veilleux, J.P. & S.L. Veilleux. 2004b. Colonies and reproductive patterns of tree-roosting female Eastern Pipistrelle bats in Indiana. Proceedings of the Indiana Academy of Science 113:60–65.

Veilleux, J.P., J.O. Whitaker, Jr. & S.L. Veilleux. 2003. Tree-roosting ecology of reproductive female Eastern Pipistrelles, *Pipistrellus subflavus*, in Indiana. Journal of Mammalogy 84:1068–1075.

Whitaker, J.O. Jr. 1998. Life history and roost switching in six summer colonies of Eastern Pipistrelles in buildings. Journal of Mammalogy 79:651–659.

Whitaker, J.O. Jr. & S.L. Gummer. 1992. Hibernation of the Big Brown Bat, *Eptesicus fuscus*, in buildings. Journal of Mammalogy 73:312–316.

Whitaker, J.O. Jr. & S.L. Gummer. 1993. The status of the Evening Bat, *Nycticeius humeralis*, in Indiana. Proceedings of the Indiana Academy of Science 102:283–291.

Whitaker, J.O. Jr. & R.E. Mumford. 2009. Mammals of Indiana. Indiana University Press, Bloomington, Indiana. 661.

Whitaker, J.O. Jr., A. Kurta & T.C. Carter. 2011. Retention of the common name Eastern Pipistrelle for *Perimyotis subflavus*. Bat Research News 52:53–54.

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2014. Proceedings of the Indiana Academy of Science 123(1):57–64

A DEVICE FOR MEASURING THE FLEXURAL STIFFNESS OF INSECT WINGS, OR HOW TO MAKE A WING-BAR GIZMO

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ABSTRACT. Flexural stiffness is an important property of many biological structures, including insect wings, but measuring it can prove challenging when the structures to be measured are small and light or have a low elastic modulus. We have designed, constructed, and tested a rugged and inexpensive device for measuring flexural stiffness. The apparatus was validated by testing with fine gage copper wire and comparing our results with those obtained from standard test equipment used for tensile testing. It is shown that results can be obtained with the wings of small butterflies. Preliminary findings on *Strymon melinus* (Hübner), the Gray Hairstreak, showed that the stiffness measurements of the butterfly wings were repeatable and therefore the testing mechanism was not damaging the wings. Little variation was found between the dorsal and ventral direction in the experimental measurements. The stiffness tester provides a simple, low cost, means to measure the flexural stiffness of small and light biological structures. This device is well within reach, and provides a means, of quality research in a small college or university setting.

Keywords: Flexural stiffness, insect wings, experimental apparatus

INTRODUCTION

Interest in the mechanical properties of insect wings has primarily been motivated by the desire to understand the mechanics of insect flight. For instance, an important application has been replicating essential features of these structures in biomimetic micro-air-vehicles (Karpelson et al. 2008). While a good portion of the work has been related to understanding the micro-scale and unsteady fluid mechanics, there is also significant interest in measuring and understanding the structural properties of the insect wings. Understanding the mechanics of insect flight may elucidate subjects as diverse as the energetics of foraging to the constraints on non-flight related wing functions (Dudley 2000).

The structural property of a wing that relates to the strength of the wing and also the deformation during flight is the flexural stiffness. The flexural stiffness of a structure is the product of Young's modulus, E, which is a material property describing the relationship between stress and strain, and the second area moment of inertia, I, which is a geometric quantity. Flexural stiffness represents the resistance to deformation under a load at a specific length along the structure. A higher flexural stiffness indicates less deflection occurs for equally applied forces. The flexural stiffness of a cantilevered structure, EI (Fig. 1) relates the displacement of the end of a structure, δ, the force applied to the beam, F, and the length from the mounted end at which the force is applied, L:

$$EI = \frac{FL^3}{3\delta} \tag{1}$$

Early examinations and cataloging of the structure of insect wings were documented by Comstock (1918) and Martynov (1925). Subsequently, Rees (1975) explored the corrugated structure and its contribution to the wing's flexural stiffness, noting that the leading edges are more strongly corrugated. Kesel et al. (1998) examined the folded structure of dragonfly and common house-fly wings, finding the overall wing experiences stress-stiffening as the folds are straightened. This result accords with the microstructure and intermolecular interactions among chitin chains in chitin microfibrils that make up the wing. Vincent & Wegst (2004) present a review showing that the mechanical properties of cuticle vary widely, spanning several orders of magnitude, depending on molecular and protein arrangements and water

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Figure 1.—Geometry of a generic beam deflection, illustrating the measured values used to calculate flexural stiffness of a structure. Length (L) from the fixed end to the location at which force (F) is applied and the deflection (δ) of the structure at the force location. These three measured values are used in equation 1 to determine flexural stiffness.

content. Dirks & Taylor (2012) examined the structure of the wing veins in a locust and observed that cross-veins act to prevent the growth of cracks that form from defects in the wing material. Vein spacing and wing material properties could strike a balance between strength and weight in the wing.

In a search for a relationship between flexural stiffness and other morphological parameters, Steppan (2000) constructed a wing bending apparatus to measure the flexural stiffness profiles of dried wings from ten different species of butterflies. The testing apparatus was a loading bar which pushed down on a wing that was mounted horizontally by mounting one or two mm of the basal attachment regions of the wings between two glass microscope slides. This loading bar applied a line load on the wing, perpendicular to the wing span, to mimic the aerodynamic loading that occurs on a wing in flight. Deflection of the wing was determined by measuring the displacement of the bar with a linear variable differential transformer attached to the loading bar. Force was measured by a transducer that appears to have been connected directly to the loading bar.

Combes & Daniel (2003a) measured the flexural stiffness of sixteen different insect species in both the span-wise and chord-wise directions by pushing with a pin on a single point, located 70% of the way along the wing length. The pin was mounted on a flexible beam that was used to measure displacement and instrumented with a force transducer that measured the force pushing on the wing. In the second part of the study (Combes & Daniel, 2003b), they used a laser to illuminate the wing before and after deformation, and optical analysis was used to measure the displacement profile across the wing. By assuming various stiffness profiles and matching the displacement results, the local stiffness values of the wing were determined.

Measurement of small forces (on the order of millinewtons) and displacements (on the order of tens of micrometers) are needed to determine the flexural stiffness of insect wings. However, the expense of equipment typically used can be a road block to collecting these data and all of the studies cited here required force transducers and other expensive measurement equipment: an apparatus similar to the ones reported in these studies could cost as much as $15,000.

This paper describes a rugged, simple to use, and inexpensive apparatus for measuring the flexural stiffness of butterfly wings. The apparatus was designed, tested and implemented with less than $1000 for materials purchased. The apparatus was constructed to use an analytical balance, which is equipment typically found in a biology or chemistry laboratory. Machining time was provided free of charge by the departmental machinist, and would have added a few hundred dollars to the overall cost. This design and procedure provide an easily accessible and affordable opportunity for more biomechanical measurements of flexural stiffness of insect wings, and may find additional applications with other biological materials with small stiffness such as feathers or small bones.

METHODS

Design of wing bar testing apparatus.—The test apparatus was constructed to measure the flexural stiffness of a butterfly wing in bending mode (Fig. 2). The load applied to measure the flexural stiffness was applied in a line force, along a line perpendicular to the major branches of the medial and cubital veins. The three species that this device was designed to test were the *Strymon melinus* (Hübner) or Gray Hairstreak, *Cupido comyntas* or Eastern Tailed Blue, and *Celastrina ladon* or Spring Azure. The wings of all these species are similarly sized, however larger species could easily be tested within the apparatus device. Maximum size is limited only by the traverse length of the micromanipulator stage.

The apparatus consisted of an analytical balance to measure the applied force and a pair of orthogonally-mounted micromanipulators (Model number NT37-936, Edmund Scientific,

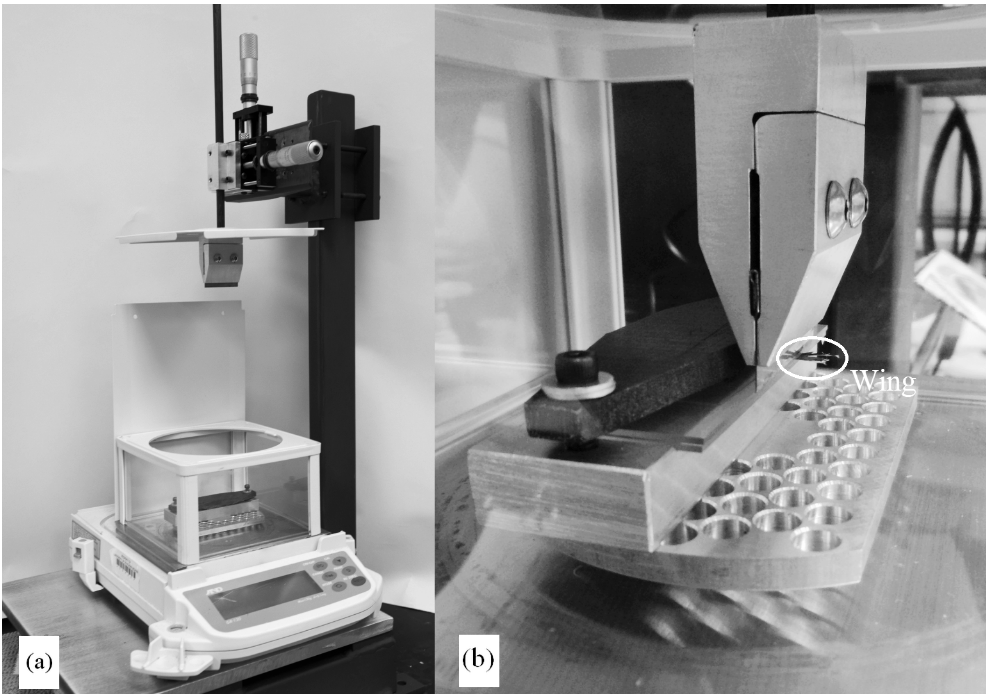

Figure 2.—(a) Photo of the wing bar testing apparatus with the micromanipulator stages visible. The enclosure of the balance was modified to reduce the distance between the wing and the micromanipulators and to minimize any potential effect of vibrations. (b) The wing-bar and a wing mounted in the glass slides. The mounting device that held the slides was inserted in place of the balance pan, and the holes drilled in the aluminum support matched the weights of the mounting device and balance pan.

Tonawanda, NY) to provide displacement in the horizontal (along the wing) and vertical (pressing on the wing) directions. The micromanipulators had 0.01 mm resolution marks, and were mounted to translational stages that were attached to an arm that was suspended above an aluminum plate and steel base for the balance. This arm and base were fabricated in the machine shop from rectangular steel bar stock and an aluminum plate of 13.5 mm thickness. Two aspects of the construction were critical for repeatable and accurate data: the overall sturdiness of the arm and the right angle that the arm formed with the base. Attached to the translating stages was an 8.0 mm diameter carbon fiber rod that extended downward to the balance and held a dulled, single-edged razor blade that provided the line force to the wing. The razor blade was slightly dulled so that there was no chance of slicing the wing, but it was still sharp enough to provide a narrow application of the load distributed across the anterior-posterior axis of the wing. The tall sliding wind screens of the analytical balance were removed and replaced with shorter ones to decrease the distance between the wing and the micromanipulators (Fig. 2a), reducing any potential vibrations in the rod.

Tested wings were glued between two glass microscope slides with cyanoacrylate glue as per Steppan (2000). The pan of the balance was replaced with a mounting device that held the slides by a piece of Bakelite (5 mm thickness) screwed to an aluminum block (11.4 mm). The screws passed through the aluminum block and into a perforated aluminum plate (5.4 mm) that served as a modified balance pan (Fig. 2b). On its bottom side, the modified pan had a milled conical protrusion that matched the dimensions of the pin from the original balance pan. The holder was aligned on the modified balance pan so that the edge of the holder passed over the center of the pin of the balance pan. Two aspects of the construction of the slide holder were critical, first that the mounting device was a sufficient replacement for the balance pan, in both total weight (not too much heavier) and in the mounting connection. The second critical aspect of the holder was that the slide and the wing be held sturdily and orthogonal to the razor blade supplying the force. Other than these critical aspects, the specific details of the wing mounting device are not critical.

Testing Procedure.—Butterfly wings and copper wire specimens used for validation were first mounted between a sandwich of two glass slides, the edges of which were kept flush to each other. Cover slips were used as spacers between the glass slides as needed and no more than 1 mm of the wing base was used to glue the wing between the two slides in the sandwich. After the glue set, the mounted wings were checked to insure flatness and perpendicular alignment of the leading edge to the edge of the glass slides. Straightened pieces of soft copper wire (16 AWG and 24 AWG, ca. 20 mm length) were mounted in a similar fashion. The mounted specimens were then screwed into the holder on the modified balance pan.

Screws were tightened on either end of the holder to fix the slide and specimen sandwich in place as well as to fix the holder to the modified balance pan. After measuring the length of the specimen, the micromanipulators were used to position the razor blade wing bar over different percentage distances along the span. At each distance where measurements were collected, the wing bar was carefully moved into a position in which it was just touching the specimen and the balance displayed a zero reading. Then, five to seven displacements of the wing were made by using the micromanipulators to advance the wing bar downward in 10 μm increments. At each of these successive deflections, the scale was read and the resulting *EI* was estimated from the slope of the linear regression of force on deflection following Equation 1.

It should be noted that Equation 1 was derived assuming a *linear elastic* response of a structure (Callister 1994). If the structure modeled using this equation does not behave linearly or elastically, the equation cannot be used to derive the flexural stiffness (the product *EI*). Any non-linearity would mean that the deformations had surpassed the elastic range or that the wing had been damaged in the testing procedure, in which case the wing data would have to be discarded.

Validation Experiments.—To confirm that the device could accurately measure the properties of samples, five samples of copper wire were tested, using a slightly modified procedure with small pieces of the wire as spacers between the slides instead of glass coverslips. Measuring force and displacement on a copper wire allowed for the calculation of Young's modulus, *E*, since the area moment of inertia, *I*, can be calculated for cylindrical wire from its diameter and the slope of the force deflection curves obtained can be substituted into Equation 1 to solve for *E*. Samples of the same wire were subjected to tensile testing in an Instron (UTM, model 5592-F1, Grove City, PA) 10 kN tensile tester and the slopes of the early elastic region of the stress-strain curve were used to estimate Young's modulus. According to isotropic theory, the modulus should be the same in tension or bending (Beer et al. 1992); however, in real metals the elastic modulus varies depending on the crystallographic orientation (Callister 1994).

RESULTS

Copper Wire.—For copper wire, the relationship between applied force and deflection was strongly linear across distances between 7 and 22 mm and deflections between 10 and 50 μm (Fig. 3). Since the slopes of the force-deflection curves increased slightly at distances closer to the mounting fixture, the calculated values of *E* decreased accordingly. Tensile testing of samples taken from a different part of the same copper wires generated values between 52 and 83 GPa with 3 of 4 tests yielding values between 78 and 83 GPa (Fig. 4). The value for the isotropic elastic modulus of pure copper was found to be 110 GPa (Beer et al. 1992), which is also shown in Fig. 4. The range of the anisotropic elastic moduli was found to be 67 to 191 GPa (Callister 1994). The elastic moduli calculated from the measured force-displacement ranged from 72 to 106 GPa.

Butterfly Wings.—Representative force-deflection data from the hind wing of *Strymon melinus* (Hübner), the Gray Hairstreak, show strong linearity at distances between 3.20 and 9.59 mm from the mounting fixture and over deflections from 10 to 250 μm (Fig. 5). This linearity in force-displacement was found in every wing that was tested, indicating that the wings were within the linear elastic region of deformation. Therefore, Equation 1 could be

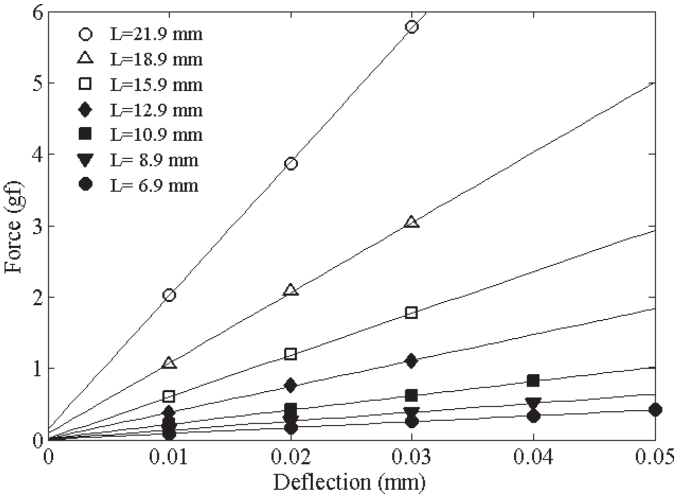

Figure 3.—Force vs. deflection for a series of tests on one copper wire at varying lengths, *L*. The linearity of the force-deflection curves confirms the validity of Equation 1. Linear beam theory is appropriate for this application. Forces reported in gram-force (gf), because the analytical balance measured in gram-force.

used validly to determine the flexural stiffness, *EI*. In addition, a series of dry wings were tested multiple times bending in the dorsal and ventral directions, to verify that no hysteresis effects could be observed from multiple trials of bending the same wing. If the wing were damaged by the device while it was being tested, subsequent tests on the same wing would yield different results. This was not the case (Fig. 6), and the device seemed capable of bending the wings without causing damage.

Flexural stiffness was found to vary across the wingspan, in general increasing distally. Some variation in flexural stiffness was ob-

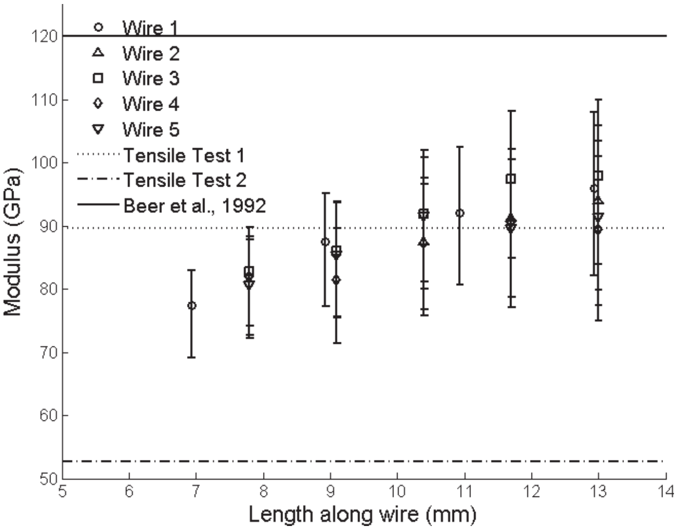

Figure 4.—Elastic modulus, *E*, for four different copper wires, measured at different lengths along the wire. The straight lines represent the elastic modulus for a different section of the same wires, measured in a tensile test, or given in a textbook. The error bars on the data points represent the total uncertainty for each point as determined by the cumulative effect of all measurements.

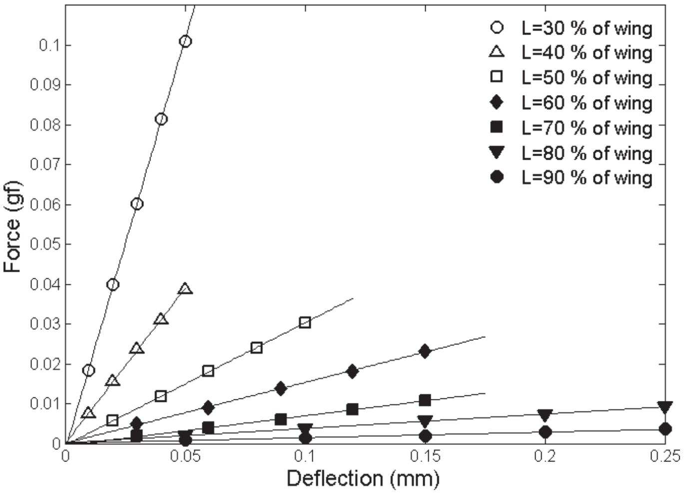

Figure 5.—Representative force-deflection data for a wing of a Grey Hairstreak butterfly at increasing lengths from the wing mount. The linearity of the wing deflections is evident in this plot, and every wing tested had a linear force-deflection relationship. Forces reported in gram-force (gf) because the analytical balance measured in gram-force.

served between tests on the dorsal and the ventral side of the wings, but no final conclusions can be made without more testing.

DISCUSSION

A testing apparatus (Fig. 2) has been developed and validated that can be used to measure small forces and displacements. The apparatus was constructed from inexpensive or available equipment, and its design could easily be replicated by researchers at any size institution. The force measurement was performed on an analytical balance, which was already available, and the wing-holding mount was made to be removable so that the scale's use was not hindered. By using the balance a force gage was not required; a force gage of sensitivity similar to the balance, such as those used by other researchers (Combes & Daniel 2003b; Smith et al. 2000; Steppan 2000), can cost approximately $2000. This is not the first instance of this cost-reducing procedure: Mountcastle & Daniel

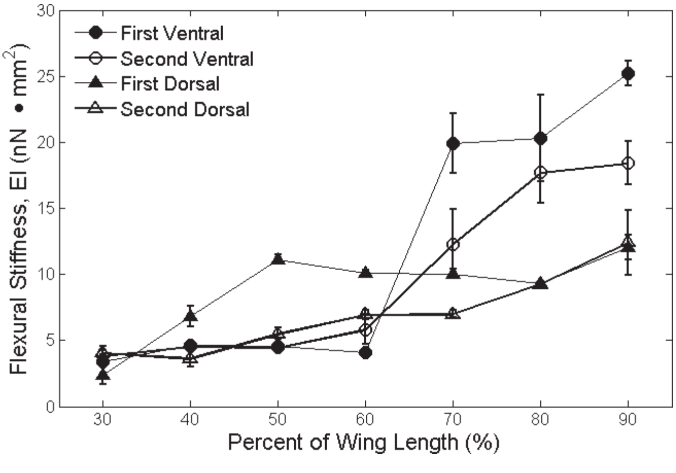

Figure 6.—Flexural stiffness measurements at different percentages of the total wing length along a Grey Hairstreak butterfly wing. This figure shows that there is little change in flexural stiffness with respect to the order in which each side of the wing, dorsal versus ventral, is tested first. The same stiffness profiles are observed, indicating that no damage is done to the wings by the testing procedure.

(2010) used an analytical balance to measure the force applied to a bending wings by mounting a pin to the pan of the balance and lowering the wing onto the pin. Two stages with manipulators were obtained for approximately $700, which allowed for positioning the deflection bar and applying the deflection; previous researchers have also used linear translators to apply deflection to the wings, and quotes for similar equipment exceeded $10,000. The cost of the raw materials for the present apparatus was $55; in our case the manufacturing was provided gratis by the departmental machinist, but labor time could add $500 to $1000 to the total cost. A trained undergraduate student operator can test as many as eight wings a day on this apparatus.

Validation of this apparatus has been performed with copper wire. Force-deflection data were found to be linear, confirming that elastic modulus can be calculated using Equation 1. Furthermore, the calculated values of the elastic modulus in bending, E, compared well to the values of the elastic modulus found in tensile tests and textbook values (Fig. 4). For a homogeneous material, the elastic modulus should be the same under tension and bending, but this is not always the case.

Steppan (2000) tested ten different species of butterflies in his wing bar apparatus. The stiffness patterns of the dry wings from all the different species (which he reported in tabular form) differed from each other, but when they were normalized and averaged across all the species, the profiles (presented in a plot) showed a peak in stiffness around the 50% location. Bending the wings in the dorsal or ventral direction made little difference in the stiffness profiles for most of the tested species. Stiffness patterns from fresh and dry wings from *Vanessa cardui* (L.), the Painted Lady, were tested and the dried wings were found to have higher stiffness than the fresh wings, although the shapes of the stiffness pattern along the length of the wing remained similar to each other. The shape of the preliminary stiffness profiles that were found with the present apparatus showed agreement with some of the species that Steppan tested, and little difference was found between pushing from the dorsal or ventral direction in the preliminary data (Fig. 6). However, the results presented here from bent wings are not sufficient to draw any conclusions.

The flexural stiffness measurements made by Combes & Daniel (2003a, 2003b) were done by applying a force at a single point, 70% along the wing. Flexural stiffness was highly correlated with the size of the wing, but not with the vein patterns. Observing the spatial deflection optically, they determined the flexural stiffness profiles for two species. They noted, however, that their force-deflection curves were non-linear. By applying a line load to the wing, we only observed linear force-displacement curves (Fig. 5). In a separate study of wing flexing, Combes & Daniel (2003c) demonstrated the inertial forces in the flapping wings are much more significant for deformation than the aerodynamic loading. This indicates that aero-elastic models need not be coupled to correctly model the deformations of a flapping wing, and implies that the structural properties of the wing are the most important element in understanding the motion and aerodynamics of an insect wing. To that end, an inexpensive testing device that can measure structural properties may be a help to other researchers in both mechanical and aerodynamic aspects of insect flight.

The design of the apparatus and testing method permits flexural stiffness testing in different directions and of different parts of the wing by varying the mounting direction of samples being affixed between the glass slides. Flexural stiffness tests can be performed on any small material that can be mounted under the wing bar, and the scale and manipulator resolutions are such that many biological samples could be reasonably tested.

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LITERATURE CITED

Beer, F.P., E.R. Johnston & J.T. Dewolf. 1992. Mechanics of Materials. McGraw-Hill, New York. 740 pp.

Callister, W.D. 1994. Materials Science and Engineering: An Introduction. Wiley, New York. 560 pp.

Combes, S.A. & T.L. Daniel. 2003a. Flexural stiffness in insect wings. I. Scaling and the influence of wing venation. Journal of Expermental Biology 206:2979–2987.

Combes, S.A. & T.L. Daniel. 2003b. Flexural stiffness in insect wings. II. Spatial distribution and dynamic wing bending. Journal of Expermental Biology 206:2989–2997.

Combes, S.A. & T.L. Daniel. 2003c. Into thin air: contributions of aerodynamic and inertial-elastic forces to wing bending in the hawkmoth *Manduca sexta*. Journal of Expermental Biology 206:2999–3006.

Comstock, J.H. 1918. The Wings of Insects. The Comstock Publishing Company, Ithaca, New York. 470 pp.

Dirks, J.H. & D. Taylor. 2012. Veins improve fracture toughness of insect wings. PLoS One 7:e43411.

Dudley, R. 2000. The Biomechanics of Insect Flight: Form, Function, Evolution. Princeton University Press, Princeton, New Jersey. 476 pp.

Karpelson, M., G.Y. Wei & R.J. Wood. 2008. A Review of Actuation and Power Electronics Options for Flapping-Wing Robotic Insects. Pp. 779–786. *In* IEEE International Conference on Robotics and Automation, Pasadena, California.

Kesel, A.B., U. Philippi & W. Nachtigall. 1998. Biomechanical aspects of the insect wing: an analysis using the finite element method. Computers in Biology and Medicine 28:423–437.

Martynov, A.V. 1925. Uber zwei Grundtypen der Flugel bei den Insecten und ihre Evolution. Zeitschrift fur Morphologie und Oecologie der Tiere 4:465–450.

Mountcastle, A.M. & T.L. Daniel. 2010. Vortexlet models of flapping flexible wings show tuning for force production and control. Bioinspiration and Biomimetics 5:045005.

Rees, C.J. 1975. Form and function in corrugated insect wings. Nature 256:200–203.

Smith, C.W., R. Herbert, R.J. Wootton & K.E. Evans. 2000. The hind wing of the desert locust (*Schistocerca gregaria* Forskal). II. Mechanical properties and functioning of the membrane. Journal of Expermental Biology 203:2933–2943.

Steppan, S.J. 2000. Flexural stiffness patterns of butterfly wings (Papilionoidea). Journal of Research on the Lepidoptera 35:61–77.

Vincent, J.F. & U.G. Wegst. 2004. Design and mechanical properties of insect cuticle. Arthropod Structure and Development 33:187–199.

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FIRST REPORT OF THE SNOW CRANE FLY *CHIONEA SCITA* WALKER, 1848 (DIPTERA: TIPULOIDEA: LIMONIIDAE) FROM INDIANA

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ABSTRACT. The snow crane fly *Chionea scita* Walker, 1848 (Diptera: Tipuloidea: Limoniidae) is reported from Indiana for the first time based on one male adult from New Bellsville, Brown County. This represents a slight westward expansion of the geographic range of this species, with the nearest records being from southeastern Michigan and east-central Kentucky. This is the first report of the species from inside a human structure. Only one other *Chionea* species has been reported from Indiana.

Keywords: Midwest, distribution, record data, *Chionea stoneana*

Crane flies (Diptera: Tipuloidea) are typically slender-bodied flies with fragile, long and slender legs and elongate, narrow wings as adults. They are among the most abundant and familiar true flies, commonly found in the suburban landscape and often mistakenly identified by non-entomologists as oversized mosquitoes; however, even those tipuloid flies with elongate mouthparts cannot bite. This is the largest single grouping of true flies, with over 15,000 species worldwide. As might be expected from such a large group, an amazing variety of forms and habits are found within its ranks (Alexander & Byers 1981; Borror et al. 1989; Oosterbroek 2014; Pers. Observ.).

The genus *Chionea* Dalman, 1816 (Limoniidae) is a peculiar group of crane flies known as the snow crane flies. *Chionea* adults are wingless and are usually encountered walking on snow (Byers 1983; Schrock 1992). They are small, brown, hairy insects, with a distinctly spiderlike appearance (Alexander & Byers 1981). Without need for bulky wing muscles, female adults may carry many eggs. Very little is known, however, about exact oviposition sites for most species. Larvae are known to occur in the spring and summer, but they are seldom collected; some species have been found associated with rodent burrows or nests, perhaps feeding on the feces found therein. As with many other coprophages, snow crane flies may serve as intermediate hosts for tapeworms. Snow crane flies pupate through the late summer and autumn, with adults usually emerging in winter. Adults may be long-lived, surviving for several months before reaching the end of their life cycle. In some cases, adults are known to carry ring-like capsules of immature nematodes between their head and thorax, serving as a means of dispersal for the roundworms. These and additional aspects of *Chionea* biology and ecology in North America are detailed, discussed and illustrated by Byers (1983, 1995) and Schrock (1992).

Fifty-six *Chionea* species are known globally, and eighteen are known from North America (Byers 1995; Oosterbroek 2014). Previously, only one species has been reported from Indiana: *C. stoneana* Alexander, 1940. The specimens on which this historical report is based were collected 23–26 December 1974 from Posey Township, Clay County, in southwestern Indiana (Byers 1983).

The single male adult I collected on 23 December 2013 from New Bellsville, Brown County (39°08′16″N, 86°07′03″W), represents the first record of *C. scita* Walker, 1848 from Indiana and a slight western extension to the species' range of geographic distribution. This also represents only the second report of the genus *Chionea* from Indiana, the previous one being based on specimens collected about 40 years ago (see above).

Chionea scita has been reported from Vermont westward to Michigan and southward to northern Georgia and South Carolina, but it

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was thought to occur only in the Appalachian Mountains south of Pennsylvania (Byers 1983, 1995; Petersen et al. 2005). The closest records to Indiana are from southeastern Michigan (Highland Recreation Area, Oakland Co., 8–12 November 1952) and east-central Kentucky (Wind Cave, Pulaski Co., 24 October 1970); these comprise the previously westernmost records of the species (Byers 1983, 1995).

The specimen newly reported herein was collected during mid-afternoon (around 3:30 pm EST) on a cold (−2°C) and cloudy day, from the recently opened doorway of an outbuilding with a concrete floor. The individual had been active inside the rather dark building, which has only a few small windows. At the time of collection, there was no snow cover, but there had been snow three days prior until it was melted by nearly two days of steady rain.

Remarkably, *C. scita* is perhaps the only North American *Chionea* species collected primarily from surfaces other than snow (Byers 1983). Notable numbers of specimens have been collected from caves in West Virginia and Kentucky and from cavities in soil along the shore of a marsh in Michigan. The possibility that the microhabitat of this species is associated with rodents has not been ruled out (Byers 1983). This new report is the first documented occurrence inside a human structure, although a previous report from Pennsylvania was based on a specimen collected from a window screen near lights at night (Byers 1983).

The Indiana specimen was identified using Byers (1983, 1995), and it is deposited in the Purdue University Entomological Research Collection, West Lafayette, Indiana, USA. I note here that the dististyles of this specimen are slightly more evenly attenuate than those figured by Byers (1983: Figs. 98 & 99) for *C. scita*.

LITERATURE CITED

Alexander, C.P. 1940. Undescribed species of crane-flies from the eastern United States and Canada (Dipt.: Tipulidae). Part VI [concl.]. Entomological News 51:99–103.

Alexander, C.P. & G.W. Byers. 1981. Tipulidae, Pp. 153–190. *In* Manual of Nearctic Diptera, Volume 1 (J.F. McAlpine, B.V. Petersen, G.E. Shewell, H.J. Teskey, J.R. Vockeroth & D.M. Wood, Eds.). Agriculture Canada, Monograph No. 27. 674 pp.

Borror, D.J., C.A. Triplehorn & N.F. Johnson. 1989. An Introduction to the Study of Insects, 6th edition. Saunders College Publishing, Ft. Worth, Texas. 875 pp.

Byers, G.W. 1983. The crane fly genus *Chionea* in North America. The University of Kansas Science Bulletin 52(6):59–195.

Byers, G.W. 1995. New species of Nearctic snow crane flies of the genus *Chionea* (Diptera: Tipulidae). Proceedings of the Entomological Society of Washington 97:508–514.

Dalman, J.W. 1816. *Chionea araneoides*. Ett nytt inlandskt insekt af tvaningarnas Ordning. Kungliga Svenska Vetenskapsakademiens Handlingar (3) 19:102–105.

Merritt, R.W., G.W. Courtney & J.B. Keiper. 2003. Diptera (flies, mosquitoes, midges, gnats). Pp. 324–340. *In* Encyclopedia of Insects (V.H. Resh, V.H. & R.T. Cardé, Eds.). Academic Press, Amsterdam, Netherlands. 1266 pp.

Oosterbroek, P. 2014. Catalogue of the Craneflies of the World (Diptera, Tipuloidea: Pediciidae, Limoniidae, Cylindrotomidae, Tipulidae). At: http://nlbif.eti.uva.nl/ccw/index.php (Accessed 16 January 2014).

Petersen, M.J., C.R. Parker & E. Bernard. 2005. The crane flies (Diptera: Tipuloidea) of Great Smoky Mountains National Park. Zootaxa 1013:1–18.

Schrock, J.R. 1992. Snow Flies. The Kansas School Naturalist 38(2):1–16.

Walker, F. 1848. List of the specimens of dipterous insects in the collection of the British museum. Part 1. The Trustees, British Museum (Natural History), London. 229 pp.

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EFFECT OF WATER ON BLOW FLY (DIPTERA: CALLIPHORIDAE) COLONIZATION OF PIGS IN NORTHWEST INDIANA

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ABSTRACT. Forensic entomology is the use of insects in the criminal justice system. Blow flies (Diptera: Calliphoridae) are early colonizers of carrion and any information on factors that influence their oviposition (egg laying) is of vital importance to forensic entomologists. This study examined the effect that being placed in a water environment had on blow fly oviposition. Six pigs were used in this study: three were in water and three were on land (control). Pigs were checked daily to document the arrival time of adult flies, fly eggs, fly larvae, the start of larval migration, and the end of larval migration. Data were analyzed using t-tests to determine if significant differences existed in the timing of blow fly life events between control pigs and pigs in water. Significant differences were seen in the timing of adult flies, fly eggs, fly larva, start of larval migration, and the end of larval migration. Pigs in the water environment initially sank but floated on the water's surface after four days. Colonization by blow flies occurred five days after field placement on the pigs in water. There was an average of a five day difference in postmortem interval (PMI) estimations between control pigs and pigs in water. The results from this study will be valuable to forensic entomologists because it provides important information about blow fly oviposition, growth and development on pigs in a water environment.

Keywords: Blow fly, water, oviposition, forensic entomology

INTRODUCTION

Forensic entomology is the use of insects in the criminal justice system (Greenberg 1991; Haskell & Williams 2008; Byrd & Castner 2010). There are three main areas of forensic entomology: urban, stored product pests, and medico-legal (Catts & Goff 1992; Hall 1995; Byrd & Castner 2010). Medico-legal forensic entomology focuses on the use of insects in determining the amount of time that has passed since insect colonization. Colonization by blow flies (Diptera: Calliphoridae) usually occurs within the first few hours after death and is used to estimate the postmortem interval (PMI) (Haskell & Williams 2008). The PMI is the period of time between death and corpse discovery. Establishing the PMI is important to investigators because it helps limit the number of possible suspects, or validate testimonies.

The PMI is calculated using a system of accumulated degree hours (ADH), or accumulated degree days (ADD). Based on life history characteristics and larval development times of different fly species, ADH (or ADD) is a measure of thermal energy required for insect larvae to reach a specific life stage. The ADH can be applied to determine an approximate time since death (Kamal 1958; Anderson 2000; Byrd & Allen 2001).

A number of different factors can influence blow fly oviposition (egg laying) such as weather, chemicals, and fly access to the corpse. An example of a barrier to oviposition that impacts fly access to a corpse would be an aquatic environment (submersion in water). A body placed in or around water could have an effect on insect colonization and subsequently the PMI. Most aquatic forensic entomology studies focus on aquatic insect species (Haskell et al. 1989; Vance et al. 1995; Keiper et al. 1997) and not blow flies colonizing pigs in water. Tomberlin & Adler (1998) studied the effect of water submersion on rats in the summer and winter months in South Carolina. They found that no flies colonized rats in water during the winter, and three species (*Cochliomyia macellaria* (Fabricius), *Lucilia sericata* (Meigen), and *Sarcophaga bullata* (Parker)) colonized in summer months. The rats in water had a four day delay in oviposition during the summer. Similarly, while conducting research on blow fly colonization on pigs in a manmade pond in Malaysia, Chin et al. (2008) found a four day delay in oviposition on the pigs in water.

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This study examined the effects of a water environment on blow fly oviposition and subsequent life stages. Pigs were used as the carrion model because they have been found to be the best substitute for humans in forensic entomology research studies (Haskell & Williams 2008). Researchers have hypothesized that the pigs in water would have a delay in oviposition that is related to the submersion of the pig.

METHODS

Research was performed in Valparaiso, Indiana, from 23 September — 14 October, 2013. The research area was a clearing in a wooded area on Valparaiso University's campus (approximate GPS coordinates 41°27′39.3″N, 87°03′02.3″W). Research was conducted inside a metal dog kennel measuring 6.1 m × 6.1 m × 1.8 m to prevent predation. Orange snow fencing covered the top of the kennel to allow sun and fly access but deter predators.

Six frozen pigs (*Sus scrofa*) were obtained from Birky Farms in Kouts, Indiana, and thawed for 15 hours in a room without fly access prior to field placement. Bugajski et al. (2011) determined that freezing prior to field placement does not significantly impact blow fly activity. A random number generator was used to place pigs inside of 1 m plots within the research area. Two treatments were examined, pigs that were placed on land (control) and pigs that were placed in water. Each treatment had three replicates. The water environment was created by filling 62 L plastic containers one-half full (31 L) with Valparaiso city water (Fig. 1). Pigs were placed into the container of water at the start of the experiment and stayed in the water for its entirety. Because pigs were not held under the water by weights, they went through periods of submersion and floatation on the surface.

The pigs were checked once daily at 1500 hr to document the arrival time of adult flies, fly eggs, fly larvae, the start of larval migration, and the end of larval migration. Daily samples of adult flies and fly larvae were taken from each pig and preserved in 70% ethanol. Specimens were identified using taxonomic keys (Stojanovich et al. 1962; Whitworth 2008).

Data were analyzed with SPSS® Statistics 18 software using t-tests to determine if significant differences existed in the timing of blow fly life events between control pigs and pigs in water (SPSS 2009). The species compositions of adult and larval flies were graphed.

Figure 1.—Pig in a 62 L plastic container with water. Containers were filled with 31 L of water prior to pig placement.

RESULTS

Significant differences existed in the first appearance of adult flies (t = −3, df = 4, P = 0.04), eggs (t = −10, df = 4, p = 0.001), larva (t = −11, df = 4, $P < 0.001$), larval migration (t = −7.18, df = 4, P = 0.002), and the end of larval migration (t = −391.0, df = 4, $P <$ 0.001).

The main difference in species diversity between the control and pigs in water was the absence of *Calliphora* and *Ophyra* spp. adult flies on water pigs (Fig. 2). Adult flies on the control pigs were comprised of 47% *Lucilia coeruleiviridis* (Macquart), 20% *Phormia regina* (Meigen), 26% *Ophyra* spp., and 7% *Calliphora* spp. (Fig. 2). Water pigs had less diversity in adult flies and were comprised of 72% *L. coeruleiviridis* and 18% *P. regina* (Fig. 2). Larval diversity was reduced from adult diversity in both treatments. Larval composition on pigs in water was made up of 59% *L. coeruleiviridis* and 41% *P. regina* (Fig. 3). The only larva represented on control pigs was *L. coeruleiviridis* (Fig. 3).

The average temperature during the experiment was 15° C; the highest recorded temperature was 27° C. ADH calculations correctly estimated control pigs to have been in the field since 23 September 2013. However, there was greater variation in the PMI estimations for the water pigs. The estimates for the water pigs were 27—29 September 2013. This represents a

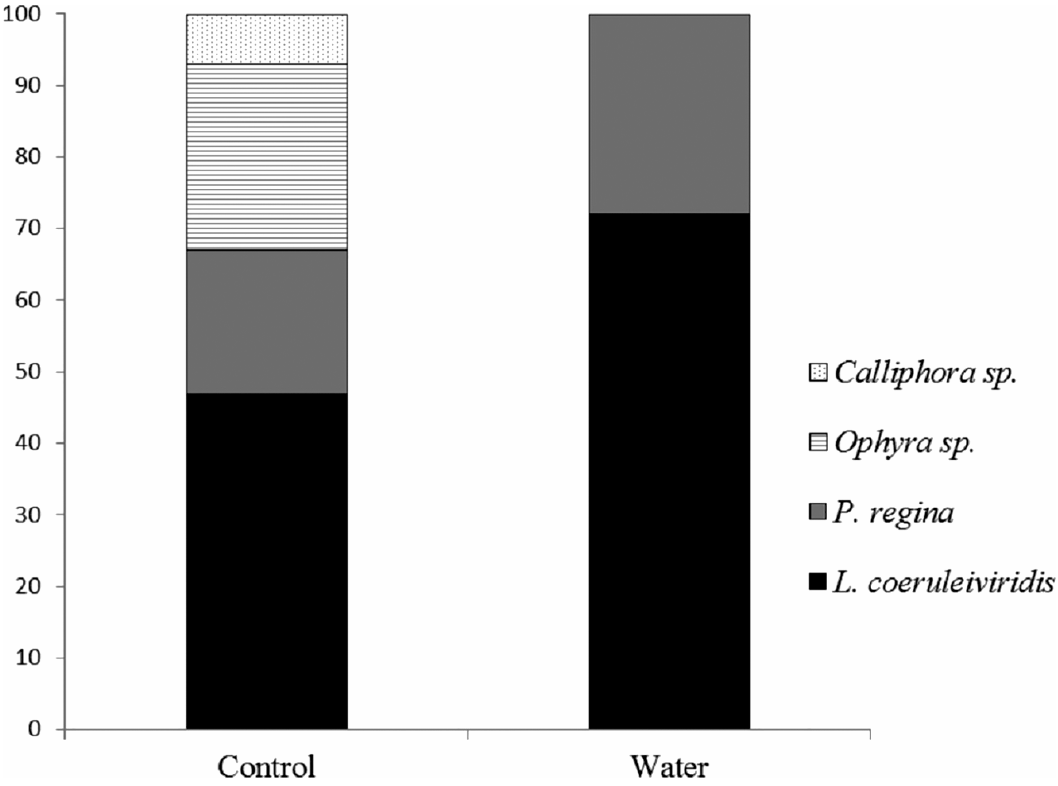

Figure 2.—Species composition of adult blow flies on water and control pigs.

four to six day delay difference from their actual placement.

DISCUSSION

Since decomposition progresses differently than on land, water presents unique challenges for forensic entomologists examining insect evidence. Haefner et al. (2004) characterized the stages of decomposition for submerged pigs as follows: submerged fresh, early floating, early floating decay, advanced floating decay, and sunken remains. They found that it took two to 13 days for the carcass to rise to the surface depending on the water habitat and time of year.

Pigs in this experiment followed the progression outlined by Haefner et al. (2004) and floated after 4 days (Fig. 4). They remained on the water surface for the remainder of the experiment. Colonization by blow flies occurred five days after placement in the field. This is similar to the four day delay in oviposition observed by Tomberlin & Adler (1998) and Chin et al. (2008). Large maggot masses were seen on the floating pigs 12 days after placement (Fig. 5). The initial sinking of the pigs delayed oviposition, but the blow fly life cycle continued the same as control pigs once the water pigs reached the floating stage. Every blow fly life stage was significantly delayed on water pigs when compared to the control. The control pigs were completely skeletonized at the conclusion of the experiment, while the pigs in water had large amounts of soft tissue remaining.

The blow fly life stage most impacted by water was larval migration. On land larvae will migrate into soil cracks or crevices for protection during the pupation stage. In a water environment, larval migration into water results in death either through drowning and decomposition, or through predation by aquatic organisms. The loss of larvae in turn eliminates the long pupal stage that is critical to successful PMI calculations. Kamal (1958) found that the pupal stage accounts for approximately half of the blow fly life cycle, making it the longest of all the life stages. Pupation follows larval migration and water

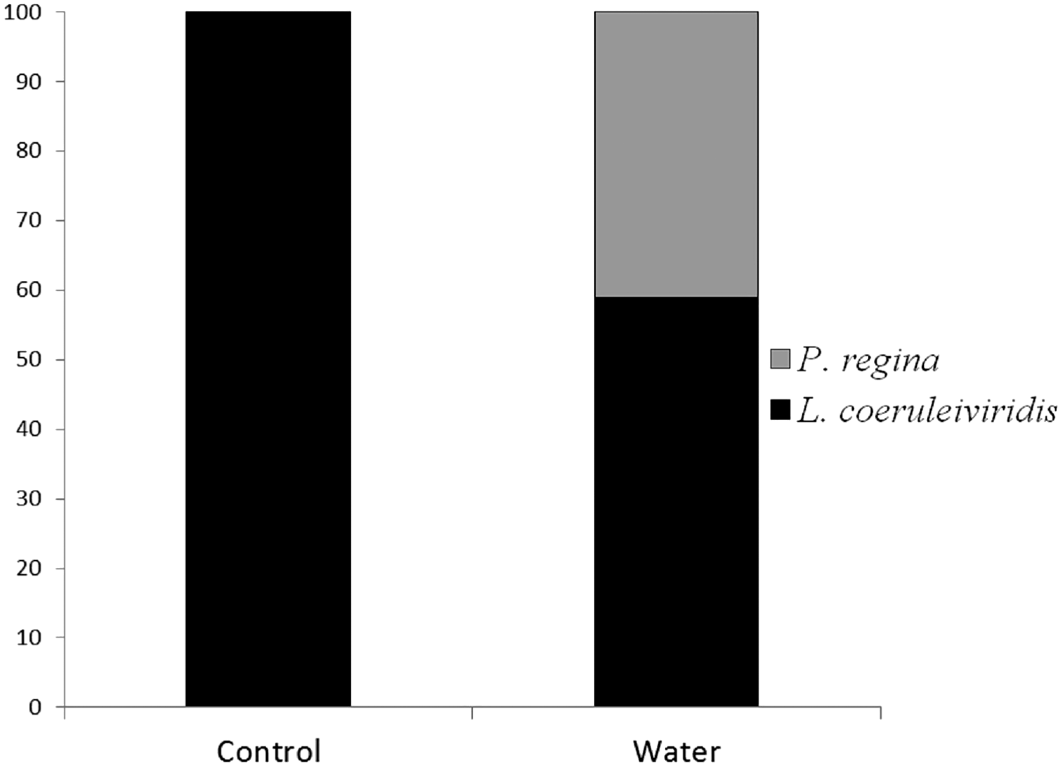

Figure 3.—Species composition of larval blow flies on water and control pigs.

eliminates the possibility of this stage. If a body is found in water and it has been colonized by blow flies that are absent from the body at the time of discovery, forensic entomologists should consider that maggots were present and died during migration.

The difference found in PMI estimations between control and pigs in water is the essential finding in this experiment. The PMI estimations for the pigs in water were an average of five days later than estimations for the control pigs. Furthermore, a greater variability in PMI estimations was seen in pigs in water when compared to the control. The control pig replicates all had the same PMI estimation date, while the pigs in water had

Figure 4.—Pig floating on the water's surface (4 days after placement).

Figure 5.—Large maggot mass on floating pig (12 days after placement).

estimation dates spanning a three day period. Since PMI estimations are based on maggot growth, the timing of oviposition is critical and the significant difference in oviposition timing between water and control pigs is the reason there is a difference.

This research provides important information about blow fly oviposition, growth and development on pigs in a water environment. Due to the variability of decomposition in water, more research needs to be conducted on pigs in a water environment to accurately estimate the PMI. Variables might include water temperature and season of the year, size of the pig, water quality, and depth of the water. Greater understanding of blow fly behavior when water is involved could complement aquatic forensic entomology studies especially when aquatic insects are limited.

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LITERATURE CITED

Anderson, G.S. 2000. Minimum and maximum development rates of some forensically important Calliphoridae (Diptera). Journal of Forensic Science 45:824–832.

Bugajski, K.N., C.C. Seddon & R.E. Williams. 2011. A comparison of blow fly (Diptera: Calliphoridae) and beetle (Coleoptera) activity on refrigerated only versus frozen-thawed pig carcasses in Indiana. Journal of Medical Entomology 48:1231–1235.

Byrd, J. & J. Allen. 2001. The development of the black blow fly, *Phormia regina* (Meigen). Forensic Science International 120:79–88.

Byrd, J. & J. Castner. 2010. Forensic Entomology: The Utility of Arthropods in Legal Investigations, 2nd ed. CRC Press, Inc., Boca Raton, Florida. 681 pp.

Catts, E. & M. Goff. 1992. Forensic entomology in criminal investigations. Annual Review of Entomology 37:253–272.

Chin, H.C., M.A. Marwi, J. Jeffery & B. Omar. 2008. Insect succession on a decomposing piglet carcass placed in a man-made freshwater pond in Malaysia. Tropical Biomedicine 25:23–29.

Greenberg, B. 1991. Flies as forensic indicators. Journal of Medical Entomology 28:565–77.

Haefner, J.N., J.R. Wallace & R.W. Merritt. 2004. Pig decomposition in lotic aquatic systems: the potential use of algal growth in establishing a postmortem submersion interval (PMSI). Journal of Forensic Sciences 49:330–336.

Hall, M. 1995. Trapping the flies that cause myiasis: their responses to host-stimuli. Annals of Tropical Medicine and Parasitology 89:333–357.

Haskell, N.H., D.G. McShaffrey, D.A. Hawley, R.E. Williams & J.E. Pless. 1989. Use of aquatic insects in determining submersion interval. Journal of Forensic Sciences 34:622–632.

Haskell, N. & R. Williams. 2008. Entomology and Death: A Procedural Guide, 2nd ed. Forensic Entomology Partners, Clemson, South Carolina. 182 pp.

Kamal, A. 1958. Comparative study of thirteen species of sarcosaprophagous Calliphoridae and Sarcophagidae (Diptera) I. Bionomics. Annals of the Entomological Society of America 51:261–270.

Keiper, J.N., E.G. Chapman & B.A. Foote. 1997. Midge larvae (Diptera: Chironomidae) as indicators of postmortem submersion interval of carcasses in a woodland stream: a preliminary report. Journal of Forensic Sciences 42:1074–1079.

SPSS for Windows, Rel. 18.0.0. 2009. SPSS Inc., Chicago, Illinois.

Stojanovich, C.J., H.D. Pratt & E.E. Bennington. 1962. Fly larvae: Key to some species of public health importance. US Department of Health, Education and Welfare. Public Health Service, Communicable Disease Center, Training Branch, Atlanta, Georgia. At: http://www.cdc.gov/nceh/ehs/Docs/Pictorial_Keys/Flies.pdf.

Tomberlin, J.K. & P.H. Adler. 1998. Seasonal colonization and decomposition of rat carrion in water and on land in an open field in South Carolina. Journal of Medical Entomology 35: 704–709.

Vance, G.M., J.K. VanDyk & W.A. Rowley. 1995. A device for sampling aquatic insects associated with carrion in water. Journal of Forensic Sciences 40:479–482.

Whitworth, T.L. 2006. Keys to the genera and species of blow flies (Diptera: Calliphoridae) of America north of Mexico. Proceedings of the Entomological Society of Washington 108: 689–725.

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THE VASCULAR FLORA AND VEGETATIONAL COMMUNITIES OF COFFMAN WOODS NATURE PRESERVE, WAYNE COUNTY, INDIANA

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ABSTRACT. Coffman Woods Nature Preserve (CWNP), owned by the Whitewater Valley Land Trust, Inc., was acquired in two steps. The first 18.2 ha (45 acres) was funded by from the Rocky Express Gas Pipeline (REX) grant program. The additional 27.9 ha (69 acres) was funded principally by the Indiana Bicentennial Nature Trust and partly by the Indiana Heritage Trust. The preserve is located along the east fork of the Whitewater River in south-central Wayne County, Indiana, in Abington Township. CWNP harbors significant regional plant diversity with 378 taxa representing 250 genera and 87 families. The twelve families containing ~59% of the documented species were the Asteraceae (49 spp.), Poaceae (39), Cyperaceae (19), Brassicaceae (18), Apiaceae (15), Fabaceae (14), Liliaceae (14), Lamiaceae (12), Polygonaceae (12), Ranunculaceae (11), Scrophulariaceae (10), and the Rosaceae (8). Of the 378 species documented, 282 [~ 75%], are native and 96 [~ 25%] are exotics, and 20 represented Wayne County Records. Although none of the plants documented at the site have state or federal status, two species are on the Indiana Watch List, i.e., *Prenanthes crepidinea* and *Veratrum woodii*. A detailed physiognomic analysis revealed that the native species consisted of 55 woody species, 183 herbaceous vines or forbs, 39 graminoids, and five ferns or fern allies. Of the 96 exotics, 11 were woody, 66 were herbaceous vines or forbs, and 19 were grasses. The major habitats occurring at CWNP are roadside and a roadside field, an old-field, steep-sloping mesic woods (the majority of the site), a small floodplain woods, a flat upland woods, a circumneutral hardwood forest seep, a small man-made pond, and the Whitewater River corridor including the riverbank, a sandy shoreline, and several sand-gravel shorelines and islands. Plants characteristics of each habitat are listed. Floristic Quality Index (FQI) for native species was 58.4 and a mean Coefficient of Conservatism (mean C) was 3.5. For all species FQI = 50.5 and the mean C = 2.6. These numbers indicate that CWNP is a site with high natural quality that is being compromised by exotics. The most invasive exotics were *Alliaria petiolata* and *Lonicera maackii* in the sloping woodland, *Poa trivialis* and *Ranunculus ficaria* var. *bulbifera* in the floodplain woods, and *Artemisia annua*, *Brassica nigra*, *Humulus japonicus* and *Phalaris arundinacea* along the Whitewater River corridor.

Keywords: Floristic Quality Index (FQI), flora – Indiana, circumneutral hardwood forest seep, floodplain woods, deciduous forest, old-field flora

INTRODUCTION

In 2009 the Whitewater Valley Land Trust, Inc. (WVLT) received a Rocky Express Gas Pipeline (REX) Migratory Bird Mitigation grant to help purchase nine properties in the Whitewater River Watershed. (These lands are being conserved, in part, by funding and technical assistance made available as mitigation for impacts caused by the construction and maintenance of Rockies Express Pipeline, LLC in partnership with the U.S. Fish and Wildlife Service.) Coffman Woods Nature Preserve (CWNP), which lies on the east side of the east fork of the Whitewater River just north of Abington, Indiana in southern Wayne County, was one of the properties purchased. Funding from the REX grant program reflects the value that U.S. Fish and Wildlife Service placed on this site as a nature preserve to serve as a permanent habitat for the native flora and fauna of this region.

Because it offered two major habitat-types not previously studied, e.g., a fairly steep-sloping mesic woodland and a river corridor, this study of CWNP was undertaken as part of our continuing effort to determine the flora and floral communities of east-central Indiana.

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There have been no formal published studies regarding the flora of CWNP. However, one of the requirements of the REX grant was to create a list of the vascular plants at each site. In 2011 in consultation with WVLT, Don Ruch compiled this list for Coffman Woods. The list was based on three forays into the site, i.e., late spring, mid-summer, and late summer/early fall. Ruch reported 243 species of plants, including 182 natives and 61 exotics.

As stated in earlier works (Ruch et al. 2008a, 2008b, 2009, 2012), an inventory of resources is the necessary first step in developing a long-term resource management plan. An inventory is the simplest means to document species diversity and is a fundamental step in monitoring changes that may occur in species composition. Additionally, measures of diversity, i.e., species richness, are frequently seen as indicators of the well-being of ecological systems (Magurran 1988). (Species richness is simply a count of species and does not take into account the abundances of the species or their relative abundance distributions.) Thus, the goals in this study were (1) to inventory the vascular flora; (2) to determine the floristic quality of the site; (3) to describe the various habitats and summarize floral dominance for each; and (4) to identify areas of special concern (e.g., areas with rare or threatened plants, if any, and communities sensitive to disturbance). Based on the finding of this study, long term resource management of the site has already been discussed with Mike Hoff, President of the WVLT.

SITE DESCRIPTION

Located in south-central Wayne County, Indiana (Fig. 1), Coffman Woods Nature Preserve is a 46 ha (~ 114 acres) property located just north and across the Whitewater River from Abington, Indiana, i.e., located in the southeast quarter of Abington Township or at ~ 39°44′03N and 84°57′25″W [NAD 83] at the gate to the logging road at the southern end of the property along Potter Shop Road; this location is marked with an X in Fig. 2. The property is part of the riparian corridor along the Whitewater River. The preserve is bordered on the south by Potter Shop Road, the Whitewater River along the southern third of the west border and by privately owned land along the northern two-thirds of the west border and the north border. The east border is adjacent to the remaining land of the Coffman family. The site was acquired in two acquisitions. The first purchase (2012), using money from the REX grant, included the western 45 acres along the river and the majority of the slope woodland. The second purchase (2013), using money from the Bicentennial Nature Trust, Indiana Heritage Trust, and WVLT, included 69 acres of the flatter upland east of the original purchase.

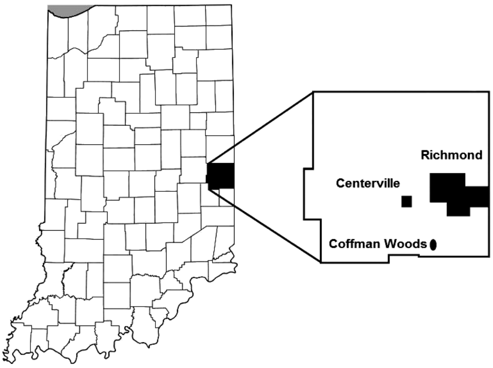

Figure 1.—Maps indicating the location of Coffman Woods Nature Preserve within Wayne County (right) and the location of Wayne County within the state of Indiana (left). The site lies approximately 10.5 km south of downtown Richmond.

Although CWNP contains a number of smaller habitat types, the majority of the site is a fairly steep, mesic, sloping woodland (Fig. 2). Within the sloping woodland several seasonal creeks have cut small rocky ravines. Also, a creek with water throughout the year flows generally to the southwest, entering the property in the northeast corner and flowing off the property in the west-central section of the woodland. Other habitat types present include the roadside and roadside field along Potter Shop Road, an old agricultural field along the southern third of the eastern border and a man-made pond, less than one-acre in size, on the east-central border. The northern 20-25% of the woodlands is a flat, upland woods with little slope. This upland woodland is bordered on the north by row-crop fields. Lying on the steep slope in the northwestern corner of the property is a circumneutral hardwood forest seep. [A circumneutral hardwood forest seep is a groundwater-fed wetland, having neutral or weakly acidic soil water, on organic soil (typically circumneutral muck) within a forest. These communities are characterized by slowly flowing water during at least

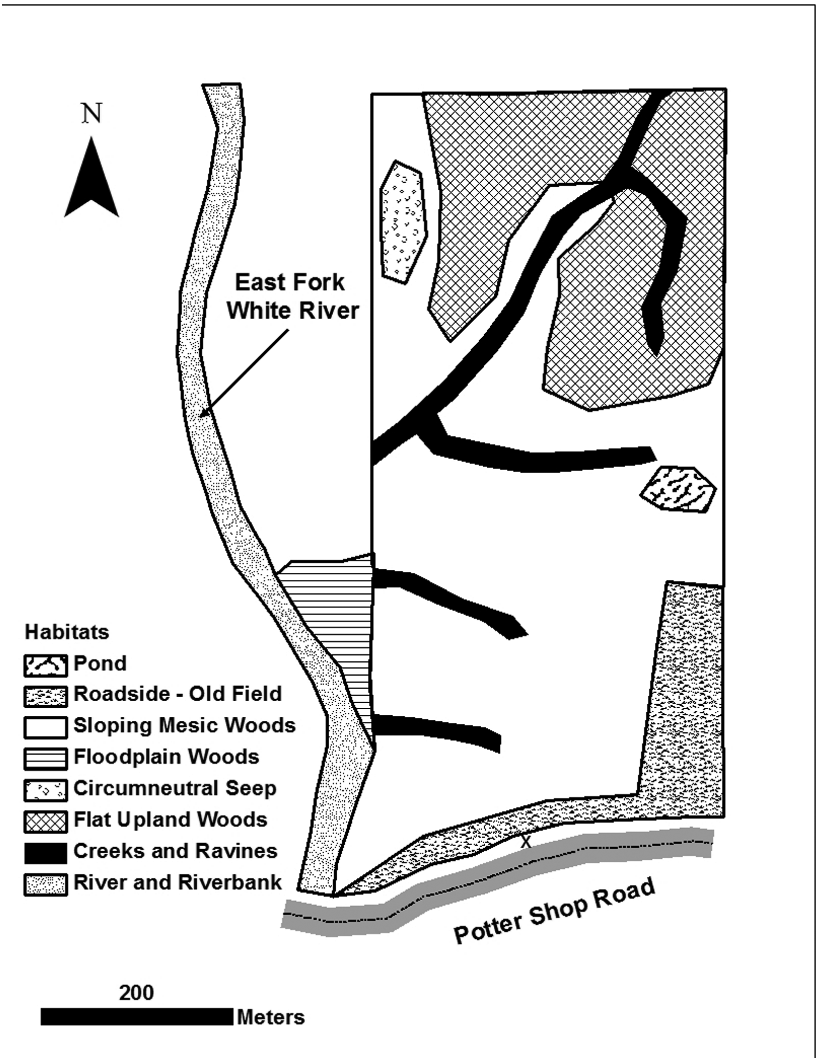

Figure 2.—Diagram illustrating the distribution of the major habitats at Coffman Woods Nature Preserve, Wayne County, Indiana. To the left of the dotted line is the original purchase of the property in 2012, and to the right of the line is the area of the second purchase (2013).

part of the year and mixture of seepage indicator and other wetland plants (Division of Forests and Lands 2014; WSAG 2005).] This seep is ~ 10 m wide and 30 m long. A small floodplain woodland is located in the southern third of the property along the river. This woodland is contiguous with a much larger, privately owned floodplain woods to the north. The final major habitat is the Whitewater River and river corridor, which is quite diverse from both the floristic and topo-edaphographic perspective. Much of the shoreline is sandy or sand and gravel and is underwater seasonally or periodically due to rainfall. These shorelines rise slowly upward through sandy and silty soil into the floodplain woods. Located near the sand and gravel shoreline, is one large sand and gravel island. The southern half of the river corridor is a riverbank which is elevated from 1 – 2.5 m above the river. At the base of the slope at several locations is a shoreline of silt, sand, and rocks that supports plant growth.

According to Homoya et al. (1985) and Wiseman & Berta (2013), CWNP lies in the transition zone between the Tipton Till Plain [Central Till Plain] and the Switzerland Hills

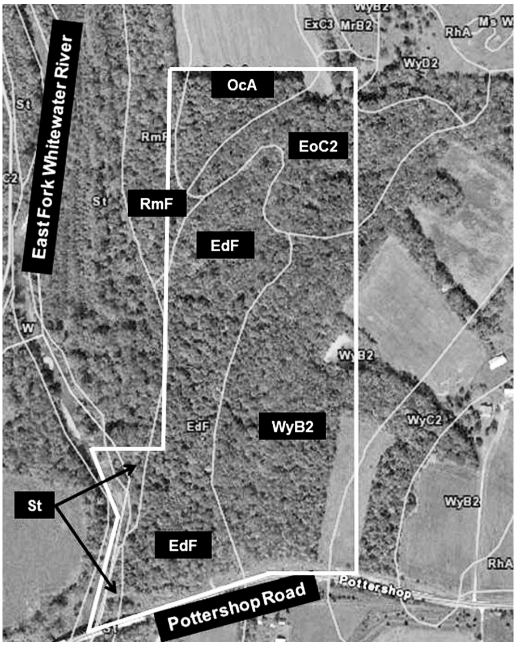

Figure 3.—Soil types and location in Coffman Woods Nature Preserve, Wayne County, Indiana. EdF = Eden flaggy silty clay loam, EoC2 = Eldean loam, OcA = Ockley silt loam, RmF = Rodman gravelly loam, St = Stonelick loam. (From WSS, 2013).

region. The preserve is within the Whitewater Watershed (USGS Cataloging Unit 05080003, EPA 2013). The Whitewater River flows south into Brookville Lake. From there it flows south and east into Ohio where it merges with the Miami River near Elizabethtown. The Miami River is tributary to the Ohio River.

The soils of CWNP range from loam to silty clay loam (Fig. 3) (Blank 1987; WSS 2013). The soil along the river corridor and the floodplain woodland is Stonelick loam, which is occasionally flooded, but well drained. The soil of the sloping woodland is Eden flaggy silty clay loam having a 25-40% slope and is eroded and well drained. The soil in the upper sloping woods and old-field at the top of the hill are Wynn silt loam characterized as having a 2-6% slope, are well drained, and eroded. The soils in the flatter woods in the northern section of the site are Eldean loam, 2-6% slope, well drained, and eroded, and Ockley silt loam, 1-2% slope and well drained. The soil around the circumneutral forest seep is Rodman gravely loam, being gravelly loam to gravelly sandy loam, having a 25-50% slope, and being an excessively drained, hydrologic soil (Blank 1987; WSS 2013).

BRIEF HISTORY OF LAND USE

Based on aerial photographs from 1950 to 1976, except for a small area in the southern-third of the eastern side of the upper slope, nearly the entire slope was wooded (IHAPI 2008). Much of the 69 acres of the flatter upland in the second purchase was non-woodland. Mr. Phil Coffman purchased the property in 1973 from the Sherwood Family. It should be noted that the original Coffman property extended along the river south of the current location of Potter Shop Road. In the early 1990s, the state built the current bridge over the Whitewater River. Because the new bridge is located north of the former bridge, 5.54 acres of the original Coffman land was lost. The current border of the CWNP was established with the extension of Potter Shop Road to the new bridge. This extension also created the roadside and roadside field habitats.

On December 9, 1958, Mr. Paul Quigg, property owner at the time, placed 62.5 acres in the Indiana Classified Forest and Wildlands Program (IDNR 2013). Mr. Quigg added another 13.8 acres on the south end of the property on February 19, 1963. The later 13.8 acres was reduced to 8.256 acres when the County reworked Potter Shop Road and took some of the land for the right-of-way. In 1975 the Coffmans logged the site by removing all trees with a diameter greater than 30.5 cm (12 in). A letter to Coffman from Greg Yapp dated June 15, 1979 refers to a harvest when Coffman bought the farm that “removed most of the large merchantable trees.” According to the Coffman’s, the woodland purchased by the Whitewater Valley Land Trust has remained undisturbed since 1975. As a final note, to the knowledge of everyone mentioned above, although the site had been selectively logged prior to the 1970s, it had never been clear cut.

METHODS

During the 2012 growing season, a foray was made every seven to ten days into the study area. Forays were made into every major habitat type, and an effort was made to cover all areas within these habitats. Voucher specimens for each species were collected and deposited in the Ball State Herbarium (BSUH).

Table 1.—Physiognomic analysis of the vascular flora documented at Coffman Woods Nature Preserve, Wayne County, Indiana. A = annual, B = biennial, H = herbaceous, P = perennial, W = woody.

	Native species summary		Exotic species summary	
	Number	% of total	Number	% of total
# of species	282	74.6%	96	25.4%
Tree	36	9.5%	4	1.1%
Shrub	11	2.9%	5	1.3%
W-Vine	8	2.1%	2	0.5%
H-Vine	3	0.8%	1	0.3%
P-Forbs	129	34.1%	26	6.9%
B-Forbs	10	2.6%	14	3.7%
A-Forbs	41	10.8%	25	6.6%
P-Grass	14	3.7%	8	2.1%
A-Grass	6	1.6%	11	2.9%
P-Sedge	16	4.2%	0	0.0%
A-Sedge	3	0.8%	0	0.0%
Fern	5	1.3%	0	0.0%

Notes on vegetation consisted of a species list with visual estimates of distribution patterns and relative abundance (see catalog of vascular plants, Appendix 1). Additionally, seasonal changes in the dominant vegetation (based on time of flowering) were noted for the various habitats. Nomenclature follows the USDA Plants Database (USDA 2013). A Floristic Quality Index (FQI) for CWNP was determined using the program developed by the Conservation Design Forum in conjunction with Rothrock (2004). This program also calculates the mean Coefficient of Conservatism (mean C), and the mean Wetland Indicator Status (mean W). Additionally, it presents a detailed physiognomic analysis of the flora, both native and exotic. For a detailed description of how the FQI is determined and an explanation of C-values, see Swink & Wilhelm (1994), Rothrock (2004), and Rothrock & Homoya (2005). Briefly, C-values, which range from zero to ten, are an index of the fidelity of an individual species to undisturbed plant communities characteristic of the region prior to European settlement. The higher the C-value the more conserved the species is to an undisturbed habitat. All exotics are given a C value of 0. The FQI is determined by multiplying the mean C for all species present by the square root of the total number of species. [For native FQI and mean C, only the native species are used.] A FQI greater than 35 suggests that a site has remnant natural quality and contains some noteworthy remnants of natural heritage of the region (Rothrock & Homoya 2005; Swink & Wilhelm 1994).

RESULTS

The catalog of the vascular flora documented at Coffman Woods Nature Preserve is listed in Appendix 1. The vascular flora consisted of 378 taxa representing 250 genera and 87 families. Thirty-three families [~ 38%] are represented by only one species and 16 families [~ 18%] are represented by only two species. As seen in Appendix 1, the twelve plant families having the highest number of species are the Asteraceae (49 species), Poaceae (39 species), Cyperaceae (19 species, including 12 *Carex* spp.), Brassicaceae (18 species), Apiaceae (15 species), Fabaceae (14 species), Liliaceae (14 species), Lamiaceae (12 species), Polygonaceae (12 species), Ranunculaceae (11 species), Scrophulariaceae (10 species), and the Rosaceae (8 species). These twelve families account for 221 of the 378 species or ~ 59% of the species documented. It should be noted that the USDA Plants Database follows the Cronquist system of plant taxonomy.

The physiognomic analysis of the flora in CWNP revealed some interesting information on plant composition (Table 1). Of the 378 plants documented, 282 [~ 74.6%] are native and 96 [~ 25.4%] are exotics. Of the 282 native species, 55 [~ 19.6%] are woody, 183 [64.8%] are herbaceous vines or forbs, 39 [~ 13.9%] are graminoids, and five [~ 1.7%] are ferns and their allies. Of the 96 exotics, 11 [~ 11.5%] are woody, 66 [~ 68.7%] are vines or forbs, and 19 [~ 19.8%] are grasses.

The Floristic Quality Index and mean Coefficients of Conservatism (mean C) for the native species is 58.4 and 3.5, respectively, and for all species, including the exotics is 50.5 and 2.6, respectively. An examination of Appendix 1 reveals that one species, *Ranunculus hispidus* var. *caricetorum*, has a Coefficient of Conservatism (C) = 10 and one species, *Carex careyana*, has a C = 9. Nine species have a C = 8, i.e., *Aristolochia serpentaria*, *Collinsonia canadensis*, *Fagus grandifolia*, *Hepatica nobilis* var. *acuta*, *Sanicula trifoliata*, *Sedum ternatum*, *Symphyotrichum prenanthoides*, *Symplocarpus foetidus* and *Trillium grandiflorum*. Twenty species have a C = 7. In comparison, 239 species [~ 63.4%] have C-values ≤ 3, i.e., 125 species with C = 0 (including 29 native species), 30 species with C = 1, 32 species with C = 2, and 52 species with C = 3.

Based upon the Indiana Natural Heritage Data Center's records for Wayne County [this is the same plant list in the computer database of Keller et al. (1984)], the USDA Plant Database (2013), Overlease & Overlease (2007), the species listed at Hayes Arboretum (Ruch et al. 2007), and at Lick Creek Summit Nature Preserve (Ruch et al. 2008a), 20 species documented in CWNP are reported for the first time and represent Wayne County records. Among these 20 species are the exotics *Aegopodium podagraria* and *Digitaria ciliaris*, and the natives *Cyperus squarrosus*, *Prenanthes crepidinea*, *Rudbeckia triloba*, *Veratrum woodii* and *Wolffia punctata*. None of the species documented at the site have state RTE status (IDNR Nature Preserves 2013), but two species are on the state watch list, i.e., *Prenanthes crepidinea* and *Veratrum woodii*. Although *Pinus strobus* is listed as state rare, all the white pines in the preserve were planted and they do not appear to be naturalizing.

DESCRIPTIONS OF THE MAJOR HABITATS

Coffman Wood Nature Preserve encompasses several habitats, each with rather distinctive plant communities (Fig. 2). The communities are separated by topographic features, water regime, and history of human use. The major habitats at CWNP are listed earlier in the Site Description section. Below is a more detailed description of the plants in each habitat.

Roadside, roadside field and old-field.—These sites are typically very dry, although the roadside field had a ditch that held water through mid-summer. Although woody species are present, such as *Fraxinus pennsylvanica* and *Juglans nigra*, the majority of the vegetation is composed of forbs and graminoids. The dominant grasses include *Andropogon virginicus*, *Bromus inermis*, *Dactylis glomerata*, *Panicum virgatum*, *Poa pratensis*, *Schedonorus arundinaceus*, *Sporobolus vaginiflorus* and *Tridens flavus*. Sedges occurring in the wetter sites include *Carex granularis*, *C. vulpinoidea*, *Scirpus atrovirens* and *S. pendulus*. Dominant forbs or forbs occurring only in this habitat type are *Asclepias* spp., especially *A. tuberosa*, *Barbarea vulgaris*, *Medicago lupulina*, *Melilotus officinalis*, *Securigera varia*, *Solidago altissima*, *Solidago nemoralis*, *Symphyotrichum pilosum* and *Typha* spp. Less frequently occurring forbs are those typical of old-fields, such as *Ambrosia artemisiifolia*, *Cichorium intybus*, *Cirsium arvense*, *Conium maculatum*, *Daucus carota*, *Eupatorium serotinum*, *Lepidium* spp., *Oenothera biennis*, *Pastinaca sativa*, *Symphyotrichum novae-angliae*, *Trifolium* spp., *Vernonia gigantea* and *Veronica arvensis*.

Sloping mesic woodland.—Most of the site consists of a steep, mesic woodland. The slope runs east to west down the hill to the Whitewater River, dropping approximately 42.5 m (140 ft). from a height of 286 m (940 ft) to 244 m (800 ft) above sea level over a distance of approximately 244 m (800 ft). A south to north transect is dissected by three seasonal creeks with small rocky ravines. The vegetation along these creeks and ravines is generally the same as the surrounding woodlands. The eastern one-third of this habitat, especially along the southern one-half of the border, contains a much younger, successional woodland than does the rest of the sloping woodland. Based on aerial photographs, this area consisted of agricultural fields in the past (IHAPI 2008). Although younger, this site does contain the same tree species as the rest of the woodland slope but is dominated by *Lonicera maackii* and many planted *Pinus strobus*. Also, *Asplenium platyneuron* occurs only in this younger woodland and is abundant.

For the rest of the sloping, mesic woodland, the canopy is composed of a variety of deciduous tree species; however, the two dominant species are *Acer saccharum* and *Celtis occidentalis*, both abundant and widespread.

Other common canopy species include *Acer nigrum*, *Carya ovata*, *Fraxinus* spp., especially *F. quadrangulata*, *Gleditsia triacanthos*, *Juglans nigra*, *Quercus muehlenbergii* and *Ulmus americana*. The most common vines are *Menispermum canadense*, *Toxicodendron radicans* and *Vitis* spp. The spring floral display is outstanding and includes *Asarum canadense*, *Cardamine bulbosa*, *C. concatenata*, *Carex albursina*, *C. careyana*, *Claytonia virginica*, *Cryptotaenia canadensis*, *Cystopteris protrusa*, *Geum vernum*, *Elymus macgregorii*, *Festuca subverticillata*, *Hydrophyllum appendiculatum*, *H. macrophyllum*, *Jeffersonia diphylla*, *Maianthemum racemosum*, *Osmorhiza longistylis*, *Packera obovata*, *Phlox divaricata*, *Poa sylvestris*, *Polygonatum biflorum*, *Sanguinaria canadensis*, *Sanicula* spp., *Stylophorum diphyllum*, which covered many acres, *Tradescantia virginiana* and *Viola* spp. Later flowering forbs include *Ageratina altissima*, *Campanulastrum americanum*, *Circaea lutetiana*, *Geum canadense*, *Phryma leptostachya*, *Solidago flexicaulis* and *Tradescantia subaspera*.

Just inside the fence on the southern property line is a shrub dominated successional woodland. This shrub-strip, up to 15 m wide, ends when the slope becomes very steep. This layer is dominated by *Lonicera maackii*, *Celtis occidentalis*, *Gleditsia triacanthos*, *Juglans nigra* and *Maclura pomifera*. Additionally, a number of other tree species typical of the woodland slope occur here.

Floodplain woodland.—The southern end [~ 245 m] of the property on the west side borders the Whitewater River. A small but well established wooded floodplain occurs in this section. The dominant trees in this floodplain are *Acer negundo*, *A. saccharinum*, *Aesculus glabra*, *Platanus occidentalis* and *Populus deltoides*. Although a diversity of herbaceous plants occurs throughout the floodplain, the most common species are *Cryptotaenia canadensis*, *Elymus virginicus*, *Glyceria striata*, *Laportea canadensis*, *Poa trivialis*, *Pilea pumila*, *Tovara virginiana*, *Ranunculus ficaria* var. *bulbifera*, *Rudbeckia laciniata*, *Silphium perfoliatum*, *Symphyotrichum lanceolatum*, *S. lateriflorum*, *S. prenanthoides*, *Tradescantia subaspera* and *Verbesina alternifolia*. This site is severely infested with the exotic *R. ficaria* var. *bulbifera*. Along the riverbank, *Ligustrum obtusifolium* occurred in two locations, the only sites on the property where it is found. The southern-end of the riverbank is elevated from 1 – 2.5 m above the river. Although this elevated bank contains many of the woody species typical of the slope woods, it also contained four species found almost exclusively here, i.e., *Asimina triloba*, *Carpinus caroliniana virginiana*, *Quercus macrocarpa* and *Symphoricarpos orbiculatus*.

Flat upland woodland.—The northern-most ~ 185 m (600 ft) is a relatively flat, dry woodland. It lies adjacent to a cow pasture on the northern border of the property. It is a mixed deciduous woodland but the most common tree species is *Acer saccharum*. This is the only site where *Fagus grandifolia* was found. The herbaceous layer is very similar to the sloping mesic woodland. *Agastache nepetoides* [rare] and *Corydalis flavula* [abundant] occur only in this habitat.

Circumneutral hardwood forest seep.—This site lies on a steep slope in the northwestern corner of the property. Woody species occurring in the seep include *Fraxinus nigra*, *Hydrangea arborescens*, *Ostrya virginiana*, *Sambucus nigra* ssp. *canadensis* and *Viburnum lentago*. The most common herbaceous species are *Cardamine bulbosa*, *C. pensylvanica*, *Cinna arundinacea*, *Equisetum hyemale*, *Galium triflorum*, *Impatiens capensis*, *Pilea pumila* and *Symplocarpus foetidus*. Plants growing on the edge of the seep and nowhere else on the property include *Collinsonia canadensis*, *Hybanthus concolor*, *Sanicula trifoliata*, *Uvularia grandiflora* and *Veratrum woodii*. Two species often associated with seeps but not found in CWNP are *Caltha palustris* and *Packera aurea*.

Whitewater River corridor (including the riverbank, a sandy shoreline, and several sand-gravel shorelines and islands).—These habitats contained the greatest diversity of plant species. Some of the plants occurring in these sites include *Abutilon theophrasti*, *Alisma subcordatum*, *Amaranthus albus*, A. *tuberculatus*, *Artemisia annua*, three *Bidens* spp., *Brassica nigra*, *Cicuta maculata*, *Commelina communis*, four *Cyperus* spp. including *C. erythrorhizos* and *C. squarrosus*, two *Echinochloa* spp., *Eclipta prostrata*, *Eleusine indica*, four *Eragrostis* spp., *Eupatorium perfoliatum*, *Humulus japonicus*, *Leucospora multifida*, *Lindernia dubia*, *Lobelia siphilitica*, *Ludwigia palustris*, *Mimulus* spp., *Mollugo verticillata*, *Muhlenbergia frondosa*, *Nasturtium officinale*, *Penthorum sedoides*, *Persicaria* spp., *Phalaris arundinacea*, *Plantago rugelii*, *Polanisia dodecandra*, many *Populus deltoides* seedlings, *Ranunculus ficaria* var. *bulbifera*, *R. hispidus* var. *caricetorum*, *Rorippa*

Table 2.—Comparison of the native Floristic Quality Index and mean Coefficient of Conservatism for sites in Wayne County, Indiana, and for sites across east-central Indiana. Sites are arranged by ascending FQI values for native species. Mean C = native mean Coefficient of Conservations, FQI = Floristic Quality Index, * = unpublished data (Ruch), NP = Nature Preserve. (Prast et al. 2014; Rothrock 1997; Ruch et al. 2004, 2007, 2008a, 2012; Stonehouse et al. 2003; Tungesvick et al. 2011).

Site	# Native spp./# total spp.	Native FQI	Mean C	Hectares	County
Coffman Woods NP	282/378	58.4	3.5	33	Wayne
Lick Creek Summit NP	304/387	68.9	4.0	16	Wayne
Duning Woods NP*	308/380	71.9	4.1	49	Wayne
Hayes Arboretum	375/525	72.0	3.7	203	Wayne
Mississinewa Woods NP	233/311	46.2	3.0	15	Randolph
Munsee Woods NP	300/399	55.0	3.2	18	Delaware
Fogwell Forest NP	210/240	59.3	4.1	11	Allen
McVey Memorial Woods*	295/378	60.0	3.5	115	Randolph
Botany Glen	295/357	68.5	4.0	18	Grant
Ginn Woods	364/441	74.1	3.9	61	Delaware
Mounds State Park	478/584	96.2	4.4	117	Madison

spp., *Rumex* spp. including *Rumex altissimus*, *Salix nigra*, *Setaria* spp., *Solanum ptycanthum*, *Verbascum* spp., *Verbena* spp., and *Veronica anagallis-aquatica*. Many more species occur along the river corridor (Appendix 1). Other than shoreline plants, no submerge aquatic vegetation is present.

Pond.—This is a small, less than one acre, man-made pond located in the east-central section of the study area (Fig. 2). It was created when a dam was constructed on the downhill side [west side]. The borders of the pond are either young successional woodlands dominated by *Lonicera maackii* or old-fields having the forbs listed in the Roadside … old-field section earlier. However, several plant species occurring only at this site or are most abundant at this site include *Bidens tripartita*, *Celastrus orbiculatus*, *Ipomoea pandurata*, *Leersia oryzoides*, *Mentha arvensis*, *Prunella vulgaris*, *Typha x glauca* and *Wolffia punctata*. On the northern-bank of the drainage ditch from the pond and just inside the border of the woodland, one bush of *Euonymus alatus* was found.

DISCUSSION

The vascular flora at Coffman Woods Nature Preserve (CWNP) included the same core of plants, and subsequently plant families, reported for other sites in east central Indiana (Prast et al. 2014; Rothrock et al. 1993; Rothrock 1997; Ruch et al. 1998, 2002, 2004, 2007, 2008a, b, 2009, 2012; Stonehouse et al. 2003; Tungesvick 2011). The 13 plant families, accounting for nearly 60% of the plants reported at CWNP and all the sites referred to above, are the Apiaceae, Asteraceae, Brassicaceae, Caprifoliaceae, Cyperaceae, Fabaceae, Lamiaceae, Liliaceae, Poaceae, Polygonaceae, Ranunculaceae, Rosaceae, and Scrophulariaceae (see Appendix 1). In the current study, these families accounted for 227 of the 378 species documented or ~60%.

The Floristic Quality Index (FQI) for the native vascular flora of CWNP was 58.4, with a mean Coefficient of Conservatism (mean C) of 3.5. These values suggest that this site is of remnant natural quality and contains some noteworthy remnants of natural heritage of the region (Rothrock & Homoya 2005; Swink & Wilhelm 1994). A comparison of the floristic quality indices of other WVLT properties (i.e., Lick Creek Summit and Durning Woods Natures Preserves) located along the Whitewater River in Wayne County to CWNP is seen in Table 2. Although the floristic quality indices are similar, the values for CWNP are somewhat lower due to fewer native species being documented and the lower mean C (Rothrock 2004; Rothrock & Homoya 2005; Ruch et al. 2010; Swink & Wilhelm 1994). The lower values at CWNP reflect the increased human impact when compared to the other sites (Ruch et al. 2008a; Mike Hoff, President of WVLT, Pers. Comm.). Table 2 also compares CWNP to other woodland sites which have been studied in east-central Indiana. Sites having similar or more human impact over the years, such as McVey Memorial Woods, Mississinewa Woods Nature Preserve, and Munsee Woods Nature Preserve, have similar native FQI and mean C values. Although having similar FQI values as CWNP, Botany Glen and Fogwell Forest Nature Preserve are both smaller sites and having higher mean C values.

The FQI and mean C for all species, both native and exotics, provides a further comprehensive understanding of the flora at CWNP. For all species the FQI = 50.5, or 7.9 units lower than the FQI for native species alone. Likewise, for all species, including exotics, the mean C is 2.6 or 0.9 units lower than the mean C for native species alone. Rothrock & Homoya (2005) have suggested that natural quality of an area is compromised when exotic diversity lowers mean C ≥ 0.7 units. Based on these numbers and on visual observations, it would appear that the exotic flora is having a negative impact on the native flora.

Ninety-six exotic species were found at CWNP or > 25% of the total species documented. While the majority of the exotic species were infrequent and occurred in the roadside field, the old-field near the top of the hill, or along the river corridor, some species were particularly invasive. Within the sloping mesic woodland, *Lonicera maackii* and *Alliaria petiolata* are a problem. *Lonicera* is growing in large colonies along the southern-third of the eastern border near the top of the hill and along the entire southern border. However, it is not growing so thickly as to prevent a herbaceous layer. Following our recommendation, the staff of WVLT began removing this exotic species. *Alliaria*, however, is growing throughout the woodland and would be virtually impossible to remove without major damage to the site. Fortunately, it is growing interspersed with other vegetation and not forming dense colonies at the expense of other species. The floodplain woodland is heavily infested with *Poa trivialis* and *Ranunculus ficaria* var. *bulbifera*. In the larger floodplain woodland just north of CWNP, *Ranunculus ficaria* is growing in a large colony covering nearly three-fourth of an acre. Lastly, the river corridor, especially the sand-gravel shoreline and island, has heavy infestations of *Artemisia annua*, *Brassica nigra*, *Humulus japonicus* and *Phalaris arundinacea*. The *Humulus* and *Phalaris* are growing so thickly in spots that they are definitely suppressing the growth of native species.

In summary, although Coffman Woods Nature Preserve is of modest size, its floristic quality, its location within the Whitewater River riparian corridor, its diversity of habitats, and its remnant woodland structure and species richness make it a significant resource for the conservation of regional biodiversity. The Rockies Express Pipeline, LLC in partnership with the U.S. Fish and Wildlife Service, the Bicentennial Nature Trust, the Indiana Heritage Trust, and Whitewater Valley Land Trust, Inc. are to be commended for recognizing the value of this site as a permanent habitat for the native flora and fauna of this region.

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APPENDIX 1

CATALOG OF VASCULAR FLORA AT COFFMAN WOODS

(Arranged alphabetically by family in each phylum)

Listed are voucher specimens for all species observed at Coffman Woods, Wayne County, Indiana. Nomenclature follows the USDA Plants Database (USDA 2013). Each species report contains the following information: (1) current scientific name based on the USDA Plants Database; (2) current taxonomic synonyms, if appropriate; (3) common names(s) based on Gleason & Cronquist (1991), Swink & Wilhelm (1994), Yatskievych (2000), and the USDA Plant Database (2013); (4) typical habitats(s) within the study site; (5) a visual estimate of its relative abundance; (6) it coefficient of conservation (C-value) for Indiana (Rothrock 2004); and (7) the Ball State University Herbarium (BSUH) number(s). Exotic species are listed in all capital letter; # = Wayne County records.

PHYLUM EQUISETOPHYTA

Equisetaceae (Horsetail Family)

Equisetum hyemale L. var. *affine* (Engelm.) A.A. Eaton; Common scouring rush or horsetail; Seep; Rare but locally abundant; C = 2; BSUH 18394.

PHYLUM POLYPODIOPHYTA

Aspleniaceae (Spleenwort Family)

Asplenium platyneuron (L.) Britton, Sterns & Poggenb. var. *platyneuron*; Ebony-spleenwort; Younger, drier upland slope woods; Common; C = 3; BSUH 18480.

Dryopteridaceae (Wood Fern Family)

Cystopteris protrusa (Weath.) Blasdell; SYN: *Cystopteris fragilis* (L.) Bernh. var. *protrusa* Weath.; Lowland bladderfern; Woodlands; Common; C = 4; BSUH 18178.

Ophioglossaceae (Adder's-tongue Family)

Botrychium dissectum Spreng. var. *obliquum* (Muhl. ex Willd.) Clute; Cut-leaf or lace-frond grape-fern, bronze fern; Slope woods; Rare; C = 3; BSUH 18196, 18533.

Botrychium virginianum (L.) Swartz; Rattlesnake fern; Woodlands; Common; C = 4; BSUH 18256.

PHYLUM PINOPHYTA

Cupressaceae (Redwood or Cypress Family)

Juniperus virginiana L. var. *virginiana*; Eastern red cedar; Woodland edge; Rare; C = 2; BSUH 18098, 18149.

Pinaceae (Pine Family)

Pinus strobus L.; Eastern white pine; Upper half of slope woods, planted; Infrequent but locally common; C = 5; BSUH 18221, 18549.

PHYLUM MAGNOLIOPSIDA

Acanthaceae (Acanthus Family)

Ruellia strepens L.; Smooth ruellia, limestone wild petunia; Woodlands; Infrequent; C = 4; BSUH 18479.

Aceraceae (Maple Family)

Acer negundo L.; Boxelder; Woodlands; Abundant; C = 1; BSUH 18126, 18528.

Acer nigrum Michx. f.; SYN: *Acer saccharum* Marshall var. *nigrum* (Michx. f.) Britton; Black maple; Woodlands; Common; C = 6; BSUH 18245, 18445.

Acer saccharinum L.; Silver or soft maple; Floodplain w oods; Abundant; C = 1; BSUH 18521.

Acer saccharum Marshall; Sugar maple; Woodlands; Abundant; C = 4; BSUH 18502.

Alismataceae (Water-Plantain Family)

Alisma subcordatum Raf.; SYN: *Alisma plantago-aquatica* L. ssp. *subcordatum* (Raf.) Hultén; American or small-flowered water-plantain; Riverbank & shore; Rare; C = 2; BSUH 18406.

Sagittaria latifolia Willd.; Common or broad-leaf arrowhead; Riverbank; Rare; C = 3; BSUH 18547.

Amaranthaceae (Amaranth Family)

Amaranthus albus L.; Tumbleweed; Sand & gravel bar and sandy shore; Abundant; C = 0; BSUH 18367.

AMARANTHUS RETROFLEXUS L.; Rough green amaranth, redroot amaranth; Sand & gravel bar, old-field; Infrequent; C = 0; BSUH 18349.

Amaranthus tuberculatus (Moq.) J.D. Sauer; SYN: *Acnida altissima* (Riddell) Moq. ex Standl.; Common water-hemp, rough-fruited amaranth; Sand & gravel bar, sandy shore; Common; C = 1; BSUH 18407.

Anacardiaceae (Cashew Family)

Toxicodendron radicans (L.) Kuntze ssp. *negundo* (Greene) Gillis; Common or Eastern poison ivy; Woodland; Abundant, widespread; C = 1; BSUH 18087.

Toxicodendron radicans (L.) Kuntze ssp. *radicans*; Common or Eastern poison ivy; Woodland edge; Infrequent; C = 1; BSUH 18123.

Annonaceae (Custard-Apple Family)

Asimina triloba (L.) Dunal; Pawpaw; Floodplain woods; Infrequent but locally common; C = 6; BSUH 18170, 18263.

Apiaceae (Carrot Family)

AEGOPODIUM PODAGRARIA L.; Goutweed; Floodplain woods; Rare; C = 0; BSUH 18274.

Angelica atropurpurea L.; Purple-stemmed angelica; Floodplain woods; Infrequent; C = 6; BSUH 18493.

Chaerophyllum procumbens (L.) Crantz var. *procumbens*; Wild or spreading chervil; Woodlands; Abundant; C = 2; BSUH 18150, 18543.

Cicuta maculata L. var. *maculata*; Spotted water hemlock; Riverbank and floodplain woods; Infrequent; C = 6; BSUH 18466.

CONIUM MACULATUM L.; Poison hemlock; Riverbank & shore, old-fields; Infrequent; C = 0; BSUH 18507.

Cryptotaenia canadensis (L.) DC.; Canadian honewort; Woodlands; Abundant; C = 3; BSUH 18501.

DAUCUS CAROTA L.; Queen Anne's-lace, wild carrot; Old-fields; Infrequent; C = 0; BSUH 18456.

Erigenia bulbosa (Michx.) Nutt.; Harbinger-of-spring; Woodlands; Abundant; C = 5; BSUH 18202.

Heracleum maximum W. Bartram; SYN: *Heracleum lanatum* Michx., *Heracleum sphondylium* L. ssp. *montanum* (Schleich. ex Gaudin) Briq.; Common cow parsnip; Floodplain woods; Infrequent but locally common; C = 6; BSUH 18088.

Osmorhiza longistylis (Torr.) DC.; Aniseroot, long-style sweetroot; Woodlands; Abundant; C = 3; BSUH 18539.

PASTINACA SATIVA L.; Wild parsnip; Old-fields; Infrequent; C = 0; BSUH 18229.

Sanicula canadensis L. var. *canadensis*; Canada sanicle, Canada black-snakeroot; Woodlands; Infrequent; C = 2; BSUH 18443.

Sanicula odorata (Raf.) Pryer & Philippe; SYN: *Sanicula gregaria* E.P. Bicknell; Clustered sanicle, clustered black-snakeroot; Woodlands; Abundant; C = 2; BSUH 18121.

Sanicula trifoliata E.P. Bicknell; Large-fruited black-snakeroot, beaked sanicle; Border of woodland seep; Rare; C = 8; BSUH 18396.

Thaspium trifoliatum (L.) A. Gray var. *aureum* (L.) Britton; Meadow parsnip; Floodplain woods; Infrequent; C = 5; BSUH 18109.

Apocynaceae (Dogbane Family)

Apocynum cannabinum L.; American Indian hemp, dogbane; Old-fields; Infrequent; C = 2; BSUH 18236, 18355.

Araceae (Arum Family)

Arisaema triphyllum (L.) Schott ssp. *triphyllum*; Jack-in-the-pulpit; Woodland creek bank; Rare; C = 4; BSUH 18494.

Symplocarpus foetidus (L.) Salisb. ex W.P.C. Barton; Skunk cabbage; Seep; Rare but locally abundant; C = 8; BSUH 18399.

Aristolochiaceae (Birthwort Family)

Aristolochia serpentaria L.; Virginia snakeroot; Woodland; Infrequent; C = 8; BSUH 18478.

Asarum canadense L.; SYN: *Asarum reflexum* E.P. Bicknell; Canadian wild ginger; Woodlands; Abundant; C = 5; BSUH 18164.

Asclepiadaceae (Milkweed Family)

Asclepias syriaca L.; Common milkweed; Old-field; Infrequent; C = 1; BSUH 18242.

Asclepias tuberosa L. var. *tuberosa*; Butterfly-weed; Old-field; Common; C = 4; BSUH 18460.

Asclepias verticillata L.; Whorled milkweed; Old-field, very dry site; Rare; C = 4; BSUH 18308.

Asteraceae (Aster Family)

Ageratina altissima (L.) King & H. Rob. var. *altissima*; SYN: *Eupatorium rugosum* Houtt.; White snakeroot; Woodlands; Abundant; C = 2; BSUH 18357.

Ambrosia artemisiifolia L. var. *elatior* Descourtils; Common or annual ragweed; Old-field; Infrequent; C = 0; BSUH 18324.

Ambrosia trifida L. var. *trifida*; Great or giant ragweed; Floodplain woods; Infrequent; C = 0; BSUH 18312.

ARCTIUM MINUS (Hill) Bernh.; Common or lesser burdock; Old-field; Rare; C = 0; BSUH 18439.

ARTEMISIA ANNUA L.; Sweet or annual wormwood, sweet sagewort; Riverbank, sandy/gravel bars, floodplain woods; Common; C = 0; BSUH 18291, 18292.

Bidens cernua L.; Nodding bur-marigold, nodding beggar's-tick; Riverbank and floodplain woods; Common; C = 2; BSUH .

Bidens frondosa L.; Common or devil's beggar's-tick; Riverbank and floodplain woods; Infrequent; C = 1; BSUH 18297.

Bidens tripartita L.; SYN: *Bidens comosa* (A. Gray) Wiegand; Three-parted or three-lobed beggar's-tick; Riverbank, floodplain woods, pond; Common; C = 2; BSUH 18303.

CICHORIUM INTYBUS L.; Chicory; Old-field; Infrequent; C = 0; BSUH 18481.

CIRSIUM ARVENSE (L.) Scop.; Canada thistle; Old-fields; Infrequent; C = 0; BSUH 18340.

Cirsium discolor (Muhl. ex Willd.) Spreng.; Field or pasteur thistle; Old-field; Rare; C = 3; BSUH 18304, 18360.

Conyza canadensis (L.) Cronquist var. *canadensis*; SYN: *Erigeron canadensis* L.; Canadian horseweed, common muleweed; Old-fields; Infrequent; C = 0; BSUH 18322.

Eclipta prostrata (L.) L.; SYN: *Eclipta alba* (L.) Hassk.; Yerba de tajo, false daisy; Riverbank and sand/gravel bars; Infrequent; C = 3; BSUH 18363.

Erigeron annuus (L.) Pers.; Eastern or annual daisy fleabane, whitetop; Old-fields; Infrequent; C = 0; BSUH 18484.

Erigeron philadelphicus L. var. *philadelphicus*; Common or Philadelphia fleabane; Woodlands; Infrequent; C = 3; BSUH 18127.

Eupatorium perfoliatum L. var. *perfoliatum*; Common boneset; Floodplain woods, sandy shore; Infrequent; C = 4; BSUH 18316.

Eupatorium serotinum Michx.; Late boneset, late-flowering thoroughwort; Old-fields; Infrequent; C = 0; BSUH 18307, 18358.

Eutrochium purpureum (L.) E.E. Lamont var. *purpureum*; SYN: *Eupatoriadelphus purpureus* (L.) King & H. Rob., *Eupatorium purpureum* L. var. *purpureum*; Sweet-scented or purple-node Joe-Pyeweed; Woodland seep; Rare; C = 5; BSUH 18387.

GAILLARDIA PULCHELLA Foug. var. *PULCHELLA*; Blanket-flower, Indian blanket; Old-fields; Infrequent; C = 0; BSUH 18457.

GALINSOGA QUADRIRADIATA Ruiz & Pav.; Common quickweed, Peruvian daisy, shaggy soldier; Floodplain woods, river bank; Infrequent; C = 0; BSUH 18333.

Helenium autumnale L. var. *autumnale*; Common sneezeweed; River bank; Rare; C = 3; BSUH 18279.

Helianthus tuberosus L.; Jerusalem-artichoke; Riverbank; Rare; C = 2; BSUH 18310.

Heliopsis helianthoides (L.) Sweet var. *helianthoides*; False sunflower, smooth oxeye; Woodland; Infrequent; C = 4; BSUH 18211.

Lactuca floridana (L.) Gaertn. var. *floridana*; Woodland or blue lettuce; Woodlands; Infrequent but widespread; C = 5; BSUH 18319.

LACTUCA SERRIOLA L.; Prickly lettuce; Roadside field; Common; C = 0; BSUH 18220.

Packera glabella (Poir.) C. Jeffrey; SYN: *Senecio glabellus* Poir.; Yellowtop, butterweed; Floodplain woods; Infrequent; C = 0; BSUH 18156, 18534.

Packera obovata (Muhl. ex Willd.) W.A. Weber & Á. Lőve; SYN: *Senecio obovatus* Muhl. ex Willd.; Round-leaved ragwort; Woodlands; Abundant; C = 7; BSUH 18151.

Polymnia canadensis L.; Small-flowered or white-flowered leafcup; Woodlands; Abundant; C = 3; BSUH 18233.

Prenanthes altissima L. var. *altissima*; Tall rattlesnake-root, white lettuce; Woodlands; Common; C = 5; BSUH 18487.

Prenanthes crepidinea Michx.; Great white lettuce, nodding rattlesnake-root, Midwestern white lettuce; Floodplain woods; Rare; C = 7; BSUH 18188.

Rudbeckia laciniata L. var. *laciniata*; Cut-leaved coneflower; Floodplain woods; Abundant; C = 3; BSUH 18375.

Rudbeckia triloba L. var. *triloba*; Three-lobed coneflower, brown-eyed Susan; Sandy river bank; Rare; C = 3; BSUH 18378.

Silphium perfoliatum L. var. *perfoliatum*; Cup-plant; Floodplain woods; Infrequent; C = 4; BSUH 18296, 18416, 18492.

Solidago altissima L.; SYN: *Solidago canadensis* L. var. *scabra* Torr. & A. Gray; Tall goldenrod, Canada goldenrod; Old-fields; Common; C = 0; BSUH 18283, 18306.

Solidago caesia L. var. *caesia*; Blue-stemmed goldenrod; Woodlands; Infrequent; C = 7; BSUH 18197.

Solidago flexicaulis L.; SYN: *Solidago latifolia* L.; Zigzag goldenrod; Woodlands, especially around the woodland seep; Common; C = 6; BSUH 18300, 18400.

Solidago gigantea Aiton; Late or giant goldenrod; Floodplain woods; Infrequent but locally common; C = 4; BSUH 18320.

Solidago nemoralis Aiton var. *nemoralis*; Old-field or gray goldenrod; Old-fields, especially in dry sites; Infrequent; C = 3; BSUH 18287.

SONCHUS ASPER (L.) Hill; Spiny sow-thistle; Old-fields; Infrequent; C = 0; BSUH 18227.

Symphyotrichum cordifolium (L.) G.L. Nesom; SYN: *Aster cordifolius* L. var. *cordifolius*; Common blue heart-leaved aster, common blue wood aster; Woodlands; Common; C = 5; BSUH 18286.

Symphyotrichum lanceolatum (Willd.) G.L. Nesom ssp. *lanceolatum* var. *lanceolatum*; SYN: *Aster lanceolatus* Willd., *Aster simplex* Willd.; White panicled aster; Woodlands, especially in the floodplain; Common; C = 3; BSUH 18194.

Symphyotrichum lateriflorum (L.) Á. Lőve & D. Lőve var. *lateriflorum*; SYN: *Aster lateriflorus* (L.) Britton; Calico, goblet or side-flowering aster; Woodlands; Abundant; C = 3; BSUH 18284.

Symphyotrichum novae-angliae (L.) G.L. Nesom; SYN: *Aster novae-angliae* L.; New England aster; Old-fields; Infrequent; C = 3; BSUH 18198, 18219, 18359.

Symphyotrichum pilosum (Willd.) G.L. Nesom var. *pilosum*; SYN: *Aster pilosus* Willd. var. *pilosus*; Heath, awl or hairy white old-field aster, goodbye-meadow; Old-fields; Common; C= 0; BSUH 18289.

Symphyotrichum prenanthoides (Muhl. ex Willd.) G.L. Nesom; SYN: *Aster prenanthoides* Muhl. ex Willd.; Zigzag or crooked-stem aster; Floodplain woods, especially near the river; Infrequent; C = 8; BSUH 18281.

TARAXACUM OFFICINALE F.H. Wigg. ssp. *OFFICINALE*; Common dandelion; Old-field, woodlands; Infrequent; C = 0; BSUH 18148.

Verbesina alternifolia (L.) Britton ex Kearney; SYN: *Actinomeris alternifolia* (L.) DC.; Wingstem; Floodplain woods; Abundant; C = 3; BSUH 18351.

Vernonia gigantea (Walter) Trel. ssp. *gigantea*; Tall or giant ironweed; Old-fields; Infrequent; C = 2; BSUH 18354.

Xanthium strumarium L. var. *canadense* (Mill.) Torr. & A. Gray; SYN: *Xanthium chinense* Mill.; Cocklebur; Floodplain woods, river bank; Infrequent but locally common; C = 0; BSUH 18428, 18429.

Balsaminaceae (Touch-Me-Not Family)

Impatiens capensis Meerb.; SYN: *Impatiens biflora* Walter; Orange jewelweed, spotted tough-me-not; Woodland seep; Abundant; C = 2; BSUH 18451.

Impatiens pallida Nutt.; Yellow jewelweed, pale touch-me-not; Floodplain woods; Infrequent; C = 4; BSUH 18228.

Berberidaceae (Barberry Family)

Jeffersonia diphylla (L.) Pers.; Twinleaf; Woodlands; Abundant; C = 7; BSUH 18171.

Podophyllum peltatum L.; May-Apple; Woodlands; Abundant; C = 3; BSUH 18183.

Betulaceae (Birch Family)

Carpinus caroliniana Walter ssp. *virginiana* (Marshall) Furlow; Musclewood, blue beech, American hornbeam; Woodlands, especially in the floodplain; Common; C = 5; BSUH 18086, 18386.

Ostrya virginiana (Miller) K. Koch; Ironwood, hop-hornbeam; Woodlands; Common; C = 5; BSUH 18485.

Bignoniaceae (Trumpet-Creeper Family)

Catalpa speciosa (Warder) Warder ex Engelm.; Northern catalpa; Floodplain woods; Rare; C = 0; BSUH 18372.

Boranginaceae (Borage Family)

Hackelia virginiana (L.) I.M. Johnst.; Stickseed, beggars-lice; Woodlands; Common; C = 0; BSUH 18409.

MYOSOTIS SCORPIOIDES L.; True forget-me-not; Riverbank; Rare; C = 0; BSUH 18083.

Brassicaceae (Mustard Family)

ALLIARIA PETIOLATA (M. Bieb.) Cavara & Grande; SYN: *Alliaria officinalis* Andrz. ex M. Bieb.; Garlic mustard; Woodlands; Abundant; C = 0; BSUH 18153.

Arabis laevigata (Muhlenb. ex Willd.) Pori. var. *laevigata*; Smooth rockcress; Woodlands; Common; C = 5; BSUH 18532.

BARBAREA VULGARIS R. Br.; Yellow rocket; Old-fields; Common; C = 0; BSUH 18140.

BRASSICA NIGRA (L.) W.D.J. Koch; Black mustard; Sandy shore; Common; C = 0; BSUH 18452.

CAPSELLA BURSA-PASTORIS (L.) Medik.; Shepherd's purse; Old-fields; Infrequent; C = 0; BSUH 18145.

Cardamine bulbosa (Schreb. ex Muhl.) Britton, Sterns & Poggenb.; SYN: *Cardamine rhomboidea* (Pers.) DC.; White or bulbous springcress; Woodland seep and floodplain woods; Common but abundant in the seep; C = 4; BSUH 18173.

Cardamine concatenata (Michx.) O. Schwarz.; SYN: *Dentaria laciniata* Muhl. ex Willd.; Cut-leaved toothwort; Woodlands; Abundant; C = 4; BSUH 18187.

Cardamine douglassii (Torr.) Britton; Purple spingcress, limestone bittercress; Woodlands; Common; C = 5; BSUH 18203.

Cardamine pensylvanica Muhl. ex Willd.; Pennsylvania bittercress; Creek bank, floodplain woods near seep; Infrequent; C = 2; BSUH 18159.

DRABA VERNA L.; SYN: *Erophila verna* (L.) Besser; Early whitlow-grass, spring draba; Roadside field; Infrequent; C = 0; BSUH 18205.

HESPERIS MATRONALIS L.; Dame's rocket; Woodlands, especially the floodplain; Common; C = 0; BSUH 18117.

Iodanthus pinnatifidus (Michx.) Steud.; Purple-rocket; Woodlands; Infrequent; C = 6; BSUH 18084.

LEPIDIUM CAMPESTRE (L.) W.T. Aiton; Field peppergrass, field pepperweed, cow cress; Old-fields; Infrequent; C = 0; BSUH 18144.

Lepidium virginicum L. var. *virginicum*; Common peppergrass, poor-man's-pepper; Old-fields; Infrequent; C = 0; BSUH 18540.

NASTURTIUM OFFICINALE R. Br.; SYN: *Rorippa nasturtium-aquaticum* (L.) Hayek; Water-cress; Old-fields in wet areas, riverbank; Infrequent; C = 0; BSUH 18241, 18258.

Rorippa palustris (L.) Besser ssp. *fernaldiana* (Butters & Abbe) Jonsell; SYN: *Rorippa islandica* (Oeder) Borbas var. *fernaldiana* Butters & Abbe; Common yellow cress; Riverbank; Infrequent; C = 2; BSUH 18335.

RORIPPA SYLVESTRIS (L.) Besser; Creeping yellow cress; Riverbank; Infrequent but locally common; C = 0; BSUH 18261, 18420.

THLASPI ARVENSE L.; Field pennycress; Floodplain woods; Rare; C = 0; BSUH 18079, 18094.

Campanulaceae (Bellflower Family)

Campanulastrum americanum (L.) Small; SYN: *Campanula americana* L.; Tall or American bellflower; Woodlands; Abundant; C = 4; BSUH 18329.

Lobelia inflata L.; Indian-tobacco; Woodland; Rare; C = 3; BSUH 18546.

Lobelia siphilitica L. var. *siphilitica*; Great blue lobelia; Floodplain woods, riverbank; Infrequent; C = 3; BSUH 18417.

Cannabaceae (Indian Hemp Family)

HUMULUS JAPONICUS Siebold & Zucc.; Japanese hops; Sandy areas along the riverbank, floodplain woods; Abundant; C = 0; BSUH 18081, 18282, 18491.

Capparaceae (Caper Family)

POLANISIA DODECANDRA (L.) DC. ssp. *DODECANDRA*; SYN: *Polanisia graviolens* Raf.; Redwhisker clammyweed; Sand/gravel bars; Infrequent; C = 0; BSUH 18404, 18424.

Caprifoliaceae (Honeysuckle Family)

LONICERA JAPONICA Thunb.; Japanese honeysuckle; Woodland edge along southern end; Rare; C = 0; BSUH 18184.

LONICERA MAACKII (Rupr.) Maxim.; Amur honeysuckle, Manchurian bush honeysuckle;

Woodlands; Common and locally abundant; C = 0; BSUH 18544.

Sambucus nigra L. ssp. *canadensis* (L.) R. Bolli; SYN: *Sambucus canadensis* L.; American black elderberry; Woodlands near creeks, woodland seep; Infrequent; C = 2; BSUH 18246.

Symphoricarpos orbiculatus Moench.; Coralberry; Woodland, especially SW quarter; Common; C = 1; BSUH 18264, 18361, 18410.

Viburnum lentago L.; Nannyberry, sheepberry; Woodland seep; Rare; C = 5; BSUH 18362.

Viburnum prunifolium L.; Black haw; Woodlands; Common; C = 4; BSUH 18090.

Caryophyllaceae (Pink Family)

SAPONARIA OFFICINALIS L.; Soapwort, bouncing-bet; Sandy area of floodplain woods; Infrequent; C = 0; BSUH 18341.

Silene stellata (L.) W.T. Aiton; Starry catchfly, widow's-frill; Woodland; Infrequent but locally common; C = 5; BSUH 18401.

Silene virginica L. var. *virginica*; Firepink; Woodlands, southern half; Infrequent; C = 7; BSUH 18503.

STELLARIA MEDIA (L.) Vill. ssp. *MEDIA*; Common chickweed; Woodlands; Abundant; C = 0; BSUH 18152.

Stellaria pubera Michx.; Great or star chickweed; Woodlands; Common; C = 7; BSUH 18168.

Celastraceae (Staff-Tree Family)

CELASTRUS ORBICULATUS Thunb.; Oriental bittersweet; Woodland edge around pond; Rare but locally abundant; C = 0; BSUH 18234.

EUONYMUS ALATUS (Thunb.) Siebold; Winged euonymus, winged burning-bush; Woodland near pond; Rare; C = 0; BSUH 18383.

EUONYMUS FORTUNEI (Turcz.) Hand-Mazz. var. *RADICANS* (Siebold ex Miq.) Rehder; Wintercreeper; Slope woodland near floodplain; Rare; C = 0; BSUH 18444.

Euonymus obovatus Nutt.; Running strawberry-bush; Slope woodland, especially SW quarter; Common; C = 7; BSUH 18254.

Chenopodiaceae (Goosefoot Family)

CHENOPODIUM ALBUM L. var. *ALBUM*; Lamb's-quarters, pigweed; Old-fields, sandy/gravel bar along river; Infrequent; C = 0; BSUH 18438.

Commelinaceae (Spiderwort Family)

COMMELINA COMMUNIS L.; Common or Asiatic day-flower; Sandy shore of the river; Rare; C = 0; BSUH 18210.

Tradescantia subaspera Ker Gawl. var. *subaspera*; Zigzag spiderwort, broad-leaved spiderwort; Woodlands, especially the floodplain; Common; C = 4; BSUH 18446.

Tradescantia virginiana L.; Virginia spiderwort; Woodlands, especially the slope woods; Infrequent but locally abundant; C = 7; BSUH 18120.

Convolvulaceae (Morning-Glory Family)

Calystegia sepium (L.) R. Br.; Common hedge bindweed; Riverbank; Infrequent; C = 1; BSUH 18408.

IPOMOEA HEDERACEA Jacq.; Ivy-leaved morning-glory; Sand/gravel bar; Rare; C = 0; BSUH 18421.

Ipomoea pandurata (L.) G. Mey.; Wild potato, man-of-the-earth; Woodland edge around pond; Rare but locally abundant; C = 3; BSUH 18217.

Cornaceae (Dogwood Family)

Cornus drummondii C.A. Mey.; Rough leaved dogwood; Woodland edge; Infrequent; C = 2; BSUH 18249.

Crassulaceae (Stonecrop Family)

Penthorum sedoides L.; Ditch-stonecrop; Riverbank, sandy shore; Infrequent; C = 2; BSUH 18373.

Sedum ternatum Michx.; Wild stonecrop; Woodlands; Common and widespread; C = 8; BSUH 18530.

Cucurbitaceae (Gourd Family)

Sicyos angulatus L.; One-seed bur cucumber; Floodplain woods; Rare; C = 3; BSUH 18350.

Cuscutaceae (Dodder Family)

Cuscuta gronovii Willd. ex Schult. var. *gronovii*; Common dodder, scaldweed; Woodland seep, floodplain forest; Infrequent but locally abundant; C = 2; BSUH 18353.

Cyperaceae (Sedge Family)

Carex albursina E. Sheld.; Blunt-scaled wood sedge; Woodlands; Common; C = 7; BSUH 18116.

Carex blanda Dewey; Common wood sedge; Woodlands; Abundant; C = 1; BSUH 18137.

Carex careyana Torr. ex Dewey; Carey's wood sedge; Woodlands; Common; C = 9; BSUH 18189, 18476.

Carex conjuncta Booth; Green-headed fox sedge, soft fox sedge; Woodlands; Infrequent; C = 6; BSUH 18136.

Carex granularis Muhl. ex Willd; Pale sedge; Old-fields; Common; C = 2; BSUH 18093.

Carex grisea Wahlenb.; Wood gray sedge, inflated narrow-leaf sedge; Woodlands; Abundant; C = 3; BSUH 18103, 18135, 18138.

Carex hystericina Willd.; Porcupine sedge; Roadside ditch in wet area; Rare; C = 5; BSUH 18512.

Carex jamesii Schwein.; Grass sedge; Woodlands; Common; C = 4; BSUH 18529.

Carex rosea Schkuhr ex Willd.; Curly-styled wood sedge, rosy sedge; Woodlands; Infrequent; C = 5; BSUH 18139.

Carex sparganioides Willd; Loose-headed bracted sedge; Woodlands; Infrequent; C = 4; BSUH 18104, 18105.

Carex tribuloides Wahlenb.; Awl-fruited oval sedge, blunt broom sedge; Riverbank; Rare; C = 5; BSUH 18391.

Carex vulpinoidea Michx. var. *vulpinoidea*; Brown fox sedge; Old-fields; Rare; C = 2; BSUH 18244.

Cyperus erythrorhizos Muhl.; Redroot flatsedge; Sand/gravel bar and sandy shoreline; Common; C = 1; BSUH 18368, 18427.

Cyperus odoratus L. SYN: *Cyperus ferruginescens* Boeckeler; Rusty flatsedge, fragrant flatsedge; Sand/gravel bar and sandy shoreline; Common; C = 1; BSUH 18327, 18434.

Cyperus squarrosus L.; Bearded flatsedge; Sand/gravel bar and sandy shoreline; Rare; C = 2; BSUH 18426.

Cyperus strigosus L.; False nutsedge, long-scaled or straw-colored nutsedge; Sand/gravel bar and sandy shoreline; Common; C = 0; BSUH 18328, 18423.

Schoenoplectus tabernaemontani (C.C. Gmel.) Pella; SYN: *Scirpus validus* Vahl.; Great or soft-stem bulrush; Sandy area of floodplain woods; Rare; C = 4; BSUH 18325, 18343.

Scirpus atrovirens Willd.; Black or dark green bulrush; Old-fields; Rare but locally common; C = 4; BSUH 18464.

Scirpus pendulus Muhl.; Red or rufous bulrush, nodding bulrush; Old-fields; Rare; C = 2; BSUH 18240.

Dipsacaceae (Teasel Family)

DIPSACUS FULLONUM L.; SYN: *Dipsacus sylvestris* Huds., *Dipsacus fullonum* L. ssp. *sylvestris* (Huds.) Clapman; Common teasel, Fuller's teasel; Old-field; Infrequent; C = 0; BSUH 18243.

Euphorbiaceae (Spurge Family)

Acalypha rhomboidea Raf.; SYN: *Acalypha virginica* L. var. *rhomboidea* (Raf.) Cooperr.; Common three-seeded mercury; Woodlands and river corridor; Abundant; C = 0; BSUH 18318.

Chamaesyce maculata (L.) Small; SYN: *Euphorbia supina* Raf., *Euphorbia maculata* L.; Creeping spurge, spotted sandmat; Roadside and sandy-gravel shoreline; Infrequent but locally common; C = 0; BSUH 18206.

Chamaesyce nutans (Lag.) Small; SYN: *Euphorbia nutans* Lag.; Nodding spurge, eyebane; Roadside; Infrequent; C = 0; BSUH 18380.

Euphorbia dentata Michx. var. *dentata*; SYN: *Poinsetta dentata* (Michx.) Klotzsch & Garcke; Toothed spurge; Old-field; Rare; C = 0; BSUH 18288.

Fabaceae (Pea or Bean Family)

Amphicarpaea bracteata (L.) Fernald; Hog-peanut; Floodplain woods; Abundant; C = 5; BSUH 18440.

Cercis canadensis L. var. *canadensis*; Eastern redbud; Woodland edge; Infrequent; C = 3; BSUH 18154, 18505.

Gleditsia triacanthos L.; Honey locust; Woodlands; Common; C = 1; BSUH 18147.

Gymnocladus dioicus (L.) K. Koch; Kentucky coffee-tree; Woodlands; Common; C = 4; BSUH 18270, 18548.

MEDICAGO LUPULINA L.; Black medic; Old-fields; Common; C = 0; BSUH 18095.

MEDICAGO SATIVA L. ssp. *SATIVA*; Alfalfa; Old-fields; Rare; C = 0; BSUH 18458.

MELILOTUS ALBUS Medik.; SYN: *Melilotus officinale* (L.) Lam.; White sweet clover; Old-fields; Common; C = 0; BSUH 18483.

MELILOTUS OFFICINALIS (L.) Lam.; Yellow sweet clover; Old-fields; Common; C = 0; BSUH 18097.

Robinia pseudoacacia L.; Black locust; Woodlands; Infrequent; C = 1; BSUH 18265.

SECURIGERA VARIA (L.) Lassen; SYN: *Coronilla varia* L.; Crown-vetch; Old-fields; Abundant; C = 0; BSUH 18510.

TRIFOLIUM HYBRIDUM L.; Alsike or hybrid clover; Old-field; Rare; C = 0; BSUH 18336.

TRIFOLIUM PRATENSE L.; Red clover; Old-field; Rare; C = 0; BSUH 18130.

TRIFOLIUM REPENS L.; White clover; Old-field; Infrequent; C = 0; BSUH 18541.

VICIA CRACCA L.; Cow vetch; Old-field; Infrequent; C = 0; BSUH 18268.

Fagaceae (Beech Family)

Fagus grandifolia Ehrh.; American beech; Woodlands; Rare, only four trees observed; C = 8; BSUH 18113.

Quercus macrocarpa Michx.; Bur oak; Woodland especially along the river; Infrequent; C = 5; BSUH 18251, 18252, 18275.

Quercus muehlenbergii Engelm; Yellow oak, chinquapin or chinkapin oak; Woodlands; Abundant; C = 4; BSUH 18384, 18511, 18536.

Quercus rubra L.; Northern red oak; Woodlands; Infrequent; C = 4; BSUH 18385, 18482, 18506.

Fumariaceae (Fumitory Family)

Corydalis flavula (Raf.) DC.; Short-spurred corydalis, yellow corydalis, yellow fumewort; Woodlands, especially the northern half; Abundant; C = 3; BSUH 18192.

Dicentra canadensis (Goldie) Walp.; Squirrel-corn; Woodlands; Abundant; C = 7; BSUH 18167.

Dicentra cucullaria (L.) Bernh.; Dutchman's-breeches; Woodlands; Abundant; C = 6; BSUH 18161.

Geraniaceae (Geranium Family)

Geranium maculatum L.; Wild geranium, spotted geranium; Woodlands; Abundant; C = 4; BSUH 18162.

Grossulariaceae (Gooseberry Family)

Ribes cynosbati L.; Dogberry, Eastern prickly gooseberry; Woodlands; Infrequent; C = 4; BSUH 18185.

Hippocastanaceae (Horse-Chestnut Family)

Aesculus glabra Willd. var. *glabra*; Ohio buckeye; Woodlands; Abundant; C = 5; BSUH 18169, 18475.

Hydrangeaceae (Hydrangea Family)

Hydrangea arborescens L.; American or wild hydrangea; Woodland seep; Rare; C = 7; BSUH 18395.

Hydrophyllaceae (Waterleaf Family)

Hydrophyllum appendiculatum Michx.; Appendaged waterleaf; Woodlands; Abundant; C = 6; BSUH 18114, 18538.

Hydrophyllum macrophyllum Nutt.; Large-leaved or hairy waterleaf; Woodlands; Common; C = 7; BSUH 18077, 18495.

Phacelia purshii Buckley; Miami-mist; Woodlands; Abundant; C = 3; BSUH 18125.

Juglandaceae (Walnut Family)

Carya cordiformis (Wangenh.) K. Koch; Bitternut hickory; Woodlands; Common; C = 5; BSUH 18260.

Carya ovata (Miller) K. Koch; Shagbark hickory; Woodlands; Common; C = 4; BSUH 18465.

Juglans nigra L.; Black walnut; Woodlands; Abundant; C = 2; BSUH 18473.

Juncaceae (Rush Family)

Juncus dudleyi Wiegand; Dudley's rush; Old-field; Infrequent; C = 2; BSUH 18100, 18277.

Lamiaceae (Mint Family)

Agastache nepetoides (L.) Kuntze; Yellow giant-hyssop; Woodland, northern end; Rare; C = 4; BSUH 18299.

Collinsonia canadensis L.; Horsebalm, richweed; Woodland around the seep; Infrequent; C = 8; BSUH 18352.

GLECHOMA HEDERACEA L.; Gill-over-the-ground, ground ivy, creeping Charlie; Floodplain woods; Infrequent but locally common; C = 0; BSUH 18141.

LAMIUM PURPUREUM L. var. *PURPUREUM*; Purple deadnettle; Woodlands; Infrequent but widespread; C = 0; BSUH 18143.

Lycopus americanus Muhl. ex W. Barton; American bugleweed, American water horehound; Floodplain woods; Abundant; C = 3; BSUH 18314.

Lycopus uniflorus Michx. var. *uniflorus*; Northern bugleweed, Northern water-horehound; Floodplain woods; Abundant; C = 5; BSUH 18321.

Mentha arvensis L.; SYN: *Mentha canadensis* L.; Field or wild mint; Riverbank, edge of pond; Infrequent; C = 4; BSUH 18313.

Monarda clinopodia L.; SYN: *Monarda fistulosa* L. var. *clinopodia* (L.) Cooperr.; Basil-balm, white bergamot; Woodlands; Infrequent, occurring in patches; C = 7; BSUH 18467.

PRUNELLA VULGARIS L. ssp. *VULGARIS*; Selfheal, heal-all; Border of pond; Rare; C = 0; BSUH 18356.

Scutellaria lateriflora L. var. *lateriflora*; Mad-dog skullcap; Sandy shore of floodplain woods; Infrequent; C = 4; BSUH 18370.

Stachys tenuifolia Willd.; SYN: *Stachys hispida* Pursh.; Smooth hedge-nettle; Floodplain woods; Infrequent; C = 4; BSUH 18455.

Teucrium canadense L. var. *canadense*; SYN: *Teucrium canadense* L. var. *virginicum* (L.) Eaton; Canada or American germander; Floodplain woods; Rare but locally common; C = 3; BSUH 18330.

Lauraceae (Laurel Family)

Lindera benzoin (L.) Blume; Spicebush; Woodlands; Infrequent; C = 5; BSUH 18078.

Lemnaceae (Duckweed Family)

Lemna minor L.; Small or common duckweed; Whitewater River; Infrequent; C = 3; BSUH 18415.

Wolffia punctata Griseb.; Watermeal; Pond; Abundant; C = 5; BSUH 18369.

Liliaceae (Lily Family)

Allium burdickii (Hanes) A.G. Jones; SYN: *Allium tricoccum* Aiton var. *burdickii* Hanes; Narrowleaf wild leek; Woodlands; Abundant; C = 6; BSUH 18176, 18237.

Allium canadense L. var. *canadense*; Wild garlic; Floodplain woods; Common; C = 1; BSUH 18132.

ASPARAGUS OFFICINALIS L.; Asparagus; Old-fields; Rare; C = 0; BSUH 18509.

Camassia scilloides (Raf.) Cory; Early-blooming wild hyacinth; Woodlands; Abundant; C = 5; BSUH 18107, 18531.

Erythronium americanum Ker Gawl. ssp. *americanum*; Yellow trout-lily, yellow dog-tooth violet; Woodlands; Common, locally abundant; C = 5; BSUH 18191, 18200.

HEMEROCALLIS FULVA (L.) L.; Orange day-lily; Riverbank; Rare; C = 0; BSUH 18468.

Maianthemum racemosum (L.) Link ssp. *racemosum*; SYN: *Smilacina racemosa* (L.) Desf.; Feathery Solomon's plume, false Solomon's-seal, feathery false lily-of-the-valley; Woodlands; Infrequent but locally common; C = 4; BSUH 18089, 18115.

ORNITHOGALUM UMBELLATUM L.; Common star-of-Bethlehem; Floodplain woods; Rare; C = 0; BSUH 18111.

Polygonatum biflorum (Walter) Elliott var. *biflorum*; Smooth Solomon's-seal; Woodlands; Abundant; C = 4; BSUH 18542.

Trillium grandiflorum (Michx.) Salisb.; Large-flowered trillium, big white trillium; Woodlands; Abundant, occurring in large patches; C = 8; BSUH 18535.

Trillium sessile L.; Sessile trillium, toad-shade; Woodlands; Abundant; C = 4; BSUH 18182.

Trillium sessile L. f. *luteum*; Yellow-form of the sessile trillium; Woodlands; Infrequent but widespread; C = 4; BSUH 18193.

Uvularia grandiflora Sm.; Large-flowered bellwort, big merrybells; Border of woodland seep; Rare; C = 7; BSUH 18190, 18382.

Veratrum woodii J.W. Robbins ex Alph. Wood; SYN: *Melanthium woodii* (J.W. Robbins ex Alph. Wood) Bodkin; False hellebore, wood's bunchflower; Border of woodland seep; Rare; C = 7; BSUH 18397.

Malvaceae (Mallow Family)

ABUTILON THEOPHRASTI Medik.; Velvetleaf; Sand/gravel bar; Rare; C = 0; BSUH 18207, 18280.

Menispermaceae (Moonseed Family)

Menispermum canadense L.; Moonseed; Woodlands; Common; C = 3; BSUH 18142.

Molluginaceae (Carpetweed Family)

MOLLUGO VERTICILLATA L.; Carpetweed; Sandy shore, sand/gravel bar; Infrequent; C = 0; BSUH 18374.

Moraceae (Mulberry Family)

MACLURA POMIFERA (Raf.) C.K. Schneider; Osage-orange, hedge apple; Floodplain woods, riverbank; Infrequent; C = 0; BSUH 18488.

MORUS ALBA L.; SYN: *Morus tatarica* L.; White mulberry; Woodlands, especially the edge, floodplain woods; Infrequent; C = 0; BSUH 18332, 18474.

Morus rubra L. var. *rubra*; Red mulberry; Woodlands; Infrequent; C = 4; BSUH 18393.

Oleaceae (Olive Family)

Fraxinus americana L.; White or American ash; Woodlands; Common; C = 4; BSUH 18266, 18504.

Fraxinus nigra Marshal; Black ash; Woodland seep; Rare; C = 7; BSUH 18411.

Fraxinus pennsylvanica Marshall; SYN: *Fraxinus pennsylvanica* Marshall var. *subintegerrima* (Vahl.) Fernald; Green ash; Woodlands; Infrequent; C = 1; BSUH 18271.

Fraxinus quadrangulata Michx.; Blue ash; Woodlands; Common C = 7; BSUH 18128.

LIGUSTRUM OBTUSIFOLIUM Siebold & Zucc.; Border privet; Floodplain woods on the riverbank; Rare; C = 0; BSUH 18131, 18490.

Onagraceae (Evening Primrose Family)

Circaea lutetiana L. ssp. *canadensis* (L.) Asch. & Magnus; SYN: *Circaea latifolia* Hill; Common or broadleaf enchanter's-nightshade; Woodlands; Abundant; C = 2; BSUH 18215, 18239.

Epilobium coloratum Biehler; Purple-leaf or cinnamon willowherb; Sandy shore of river; Rare; C = 3; BSUH 18422.

Ludwigia palustris (L.) Elliott; Marsh purslane, marsh seedbox; Shoreline of river; Rare but locally abundant; C = 3; BSUH 18405.

Oenothera biennis L.; SYN: *Oenothera pycnocarpa* Atk. & Bartlett; Common evening-primrose; Old-fields; Infrequent; C = 0; BSUH 18323.

Oxalidaceae (Wood Sorrel Family)

Oxalis dillenii Jacq.; SYN: *Oxalis stricta* auct. non L.; Slender yellow wood sorrel; Old-field, woodlands; Infrequent; C = 0; BSUH 18091.

Papaveraceae (Poppy Family)

CHELIDONIUM MAJUS L.; Lesser celandine; Sandy shore of river; Rare; C = 0; BSUH 18524.

Sanguinaria canadensis L.; Bloodroot; Woodlands; Abundant; C = 5; BSUH 18180, 18201.

Stylophorum diphyllum (Michx.) Nutt.; Wood poppy, celandine poppy; Woodlands; Abundant; C = 7; BSUH 18160, 18526.

Phytolaccaceae (Pokeweed Family)

Phytolacca americana L. var. *americana*; Pokeweed; Floodplain woods; Rare; C = 0; BSUH 18449.

Plantaginaceae (Plantain Family)

PLANTAGO LANCEOLATA L.; English plantain, buckhorn; Old-fields; Infrequent; C = 0; BSUH 18092.

Plantago rugelii Decne. var. *rugelii*; American or blackseed plantain; Sandy shoreline of river [very large plants]; Infrequent; C = 0; BSUH 18225.

Platanaceae (Plane-Tree Family)

Platanus occidentalis L.; American sycamore, buttonwood; Floodplain woods; Infrequent; C = 3; BSUH 18523.

Poaceae (Grass Family)

AGROSTIS STOLONIFERA L.; SYN: *Agrostis stolonifera* L. var. *palustris* (Huds.) Farw; Creeping bent-grass; Sandy shoreline of river; Rare; C = 0; BSUH 18461.

Andropogon virginicus L. var. *virginicus*; Broom-sedge, Virginia bluestem; Old-fields; Common; C = 1; BSUH 18290, 18305.

BROMUS COMMUTATUS Schrad.; SYN: *Bromus racemosus* L.; Hairy chess; Old-fields; Rare; C = 0; BSUH 18106, 18518.

BROMUS INERMIS Leyss. var. *INERMIS*; Smooth or Hungarian brome; Old-fields; Abundant; C = 0; BSUH 18513.

BROMUS JAPONICUS Thunb.; SYN: *Bromus arvensis* L.; Japanese chess, field brome; Old-fields; Infrequent; C = 0; BSUH 18463.

Cinna arundinacea L.; Common woodreed; Woodland around the seep; Common here, absent elsewhere; C = 4; BSUH 18390.

DACTYLIS GLOMERATA L.; Orchard grass; Old-fields; Common; C = 0; BSUH 18101.

DIGITARIA CILIARIS (Retz.) Koeler; SYN: *Digitaria sanguinalis* (L.) Scop var. *ciliaris* (Retz.) Parl.; Southern crabgrass; Sandy shoreline of river; Infrequent; C = 0; BSUH 18365.

ECHINOCHLOA CRUS-GALLI (L.) P. Beauv.; Barnyard-grass; Sandy shoreline of river, sand/gravel bar; Common; C = 0; BSUH 18348, 18366.

Echinochloa muricata (P. Beauv.) Fernald var. *muricata*; Rough barnyard-grass; Sandy shore of river, sand/gravel bar; Common; C = 1; BSUH 18222, 18435.

ELEUSINE INDICA (L.) Gaertn.; Crowfoot-grass, yard-grass, Indian goosegrass; Sand/gravel bar; Common; C = 0; BSUH 18208.

Elymus hystrix L. var. *hystrix*; SYN: *Hystrix patula* Moench; Eastern bottlebrush grass; Woodlands; Infrequent; C = 5; BSUH 18436.

Elymus macgregorii R. Brooks; SYN: *Elymus virginicus* L.; Early wild rye; Woodlands; Abundant; C = 3; BSUH 18272, 18273, 18515.

Elymus villosus Muhl.; Downy wild rye; Woodlands; Common; C = 4; BSUH 18345.

Elymus virginicus L.; Virginia wild rye; Floodplain woods; Common; C = 3; BSUH 18344.

ERAGROSTIS CILIANENSIS (All.) Vign. ex Janchen; Stink-grass; Sand/gravel bar; Infrequent; C = 0; BSUH 18412.

Eragrostis hypnoides (Lam.) Britton, Sterns & Poggenb.; Teal or creeping love-grass; Sandy shoreline of river [where the river receded]; Common; C = 3; BSUH 18389.

ERAGROSTIS MINOR Host; SYN: *Eragrostis poaeoides* P. Beauv. ex Roem. & Schult.; Low or little love-grass; Sand/gravel bar; Infrequent; C = 0; BSUH 18414.

Eragrostis pectinacea (Michx.) Nees ex Steud. var. *pectinacea*; Carolina or tufted love-grass; Sand/gravel bar; Infrequent; C = 0; BSUH 18413.

Festuca subverticillata (Pers.) E. Alexeev.; Nodding fescue; Woodlands; Abundant; C = 4; BSUH 18517.

Glyceria striata (Lam.) Hitchc.; Fowl manna grass; Floodplain woods; Abundant; C = 4; BSUH 18516.

Leersia oryzoides (L.) Sw.; Rice cut-grass; Border of pond; Abundant locally; C = 2; BSUH 18302.

Leersia virginica Willd.; White grass; Woodlands; Common; C = 4; BSUH 18377.

Muhlenbergia frondosa (Poir.) Fernald; Common satin grass, wirestem muhly; Riverbank and shoreline; Infrequent but locally abundant; C = 3; BSUH 18433.

Panicum dichotomiflorum Michx. var. *dichotomiflorum*; Knee-grass, fall panic grass; Sand/gravel bar; Infrequent; C =0; BSUH 18295.

Panicum philadelphicum Bernh.; Philadelphia panic grass; Sandy shoreline of river; Rare; C = 4; BSUH 18388.

Panicum virgatum L. var. *virgatum*; Switchgrass; Old-field; Common; C = 4; BSUH 18364, 18381.

PHALARIS ARUNDINACEA L.; Reed canary-grass; Floodplain woods, riverbank and shore; Infrequent but locally abundant; C = 0; BSUH 18108.

POA ANNUA L.; Speargrass, annual bluegrass; Old-fields; Infrequent; C = 0; BSUH 18146.

POA COMPRESSA L.; Canada bluegrass; Woodlands; Common; C = 0; BSUH 18462.

POA PRATENSIS L. ssp. *PRATENSIS*; Kentucky bluegrass; Old-fields; Abundant; C = 0; BSUH 18099, 18134.

Poa sylvestris A. Gray; Woodland bluegrass; Woodlands; Abundant; C = 5; BSUH 18133.

POA TRIVIALIS L.; Rough bluegrass; Floodplain; Abundant; C = 0; BSUH 18102.

SCHEDONORUS ARUNDINACEUS (Schreb.) Dumort., nom. cons.; SYN: *Schedonorus phoenix* (Scop.) Holob, *Festuca elatior* L. var. *arundinacea* (Schreb.) Wimm., *Festuca arundinacea* Schreb.; Tall fescue; Old-fields; Common; C = 0; BSUH 18514.

SETARIA FABERI R. Herrm.; Nodding or giant foxtail; Sand/gravel bar, sandy shoreline of river; Rare; C = 0; BSUH 18342.

SETARIA PUMILA (Poir.) Roem. & Schult. ssp. *pumila*; SYN: *Setaria glauca* (L.) P. Beauv.; Yellow foxtail; Old-fields, sand/gravel bar; Infrequent; C = 0; BSUH 18379.

SETARIA VIRIDIS (L.) P. Beauv. var. *viridis*; Green foxtail, green bristle-grass; Sand/gravel bar; Rare; C = 0; BSUH 18326, 18402.

Sporobolus vaginiflorus (Torr. ex A. Gray) Alph. Wood var. *vaginiflorus*; Poverty-grass, poverty drop-seed; Old-fields; Infrequent; C = 1; BSUH 18309.

Tridens flavus (L.) A. Hitchc. var. *flavus*; SYN: *Triodia flava* (L.) Smyth; Purpletop tridens; Old-fields; Common; C = 1; BSUH 18432.

Polemoniaceae (Phlox Family)

Phlox divaricata L. ssp. *divaricata*; Wild blue or woodland phlox; Woodlands; Abundant; C = 5; BSUH 18177.

Phlox paniculata L.; Garden, summer or fall phlox; Floodplain woods; Infrequent; C = 3; BSUH 18338.

Polemonium reptans L. var. *reptans*; Spreading Jacob's-ladder, Greek valerian; Woodlands; Abundant; C = 5; BSUH 18181.

Polygonaceae (Smartweed Family)

FALLOPIA JAPONICA (Houtt.) Ronse Decr.; SYN: *Polygonum cuspidatum* Siebold & Zucc.;

Japanese knotweed; Sandy shoreline of river; Rare, one plant; C = 0; BSUH 18486.

Fallopia scandens (L.) Holub; SYN: *Polygonum scandens* L. var. *scandens*; Climbing false buckwheat; Floodplain woods; Infrequent; C = 0; BSUH 18278.

PERSICARIA CESPITOSA (Blume) Nakai, var. *LONGISETA* (Bruijn) C. F. Reed; SYN: *Polygonum cespitosum* Blume var. *longisetum* (Bruijn) A.N. Steward, *Polygonum longisetum* Bruijn, *Persicaria longiseta* (Bruijn) Kitagawa; Creeping smartweed, Oriental lady's-thumb; Woodlands, sandy shoreline of river; Common; C = 0; BSUH 18247, 18301, 18431.

Persicaria lapathifolia (L.) Gray; SYN: *Polygonum lapathifolium* L.; Heart's-ease, dock-leaved smartweed, curly-top knotweed; Sandy shoreline of river; Abundant; C = 0; BSUH 18231, 18315.

PERSICARIA MACULOSA Gray; SYN: *Polygonum persicaria* L., *Polygonum dubium* Stein, *Polygonum maculata* (Raf.) Gray, *Persicaria vulgaris* Webb & Moq.; Spotted lady's-thumb; Riverbank and sandy shoreline of river; Common; C = 0; BSUH 18080, 18230, 18430.

Persicaria pensylvanica (L.) Small; SYN: *Polygonum pensylvanicum* L.; Pinkweed, Pennsylvania smartweed; Sandy shoreline of river; Infrequent; C = 0; BSUH 18419.

Persicaria punctata (Elliott) Small var. *leptostachya* (Meisn.) Small; SYN: *Polygonum punctatum* Elliott var. *confertiflorum* (Meisn.) Fassett; Water or dotted smartweed; Floodplain woods, sandy shoreline of river; Abundant; C = 3; BSUH 18223.

POLYGONUM AVICULARE L.; SYN: *Polygonum monspeliense* Pers.; Common, field or prostrate knotweed; Sandy shoreline of river; Common; C = 0; BSUH 18232.

Rumex altissimus Alph. Wood; Pale dock; Sandy shoreline of river; Common; C = 2; BSUH 18347, 18489.

RUMEX CRISPUS L.; Curly dock, sour dock; Sandy shoreline of river; Infrequent; C = 0; BSUH 18257, 18262.

RUMEX OBTUSIFOLIUS L; Bitter dock, blunt-leaved dock; Floodplain woods, sandy shoreline of river; Infrequent; C = 0; BSUH 18448.

Tovara virginiana (L.) Raf.; SYN: *Polygonum virginianum* L., *Persicaria virginiana* (L.) Gaertn.; Woodland knotweed, jumpseed; Woodlands; Abundant; C = 3; BSUH 18371.

Portulacaceae (Purslane Family)

Claytonia virginica L. var. *virginica*; Spring beauty; Woodlands; Common; C = 2; BSUH 18163.

PORTULACA OLERACEA L.; Purslane, little hogweed; Sand/gravel bar; Rare; C = 0; BSUH 18437.

Primulaceae (Primrose Family)

LYSIMACHIA NUMMULARIA L.; Moneywort, creeping Jenny; Floodplain woods, seasonal creek beds; Common; C = 0; BSUH 18497.

Ranunculaceae (Buttercup Family)

Clematis virginiana L.; Virgin's-bower, devil's darning needles; Riverbank; Rare; C = 3; BSUH 18469.

Delphinium tricorne Michx.; Dwarf, spring, or rock larkspur; Woodlands; Abundant; C = 5; BSUH 18174.

Enemion biternatum Raf.; SYN: *Isopyrum biternatum* (Raf.) Torr. & A. Gray; Eastern false rue-anemone; Woodlands; Infrequent but locally abundant; C = 5; BSUH 18155.

Hepatica nobilis Schreb. var. *acuta* (Pursh) Steyerm.; SYN: *Hepatica acutiloba* DC.; Sharp-leaved hepatica, liverleaf; Slope woods near floodplain, border of woodland seep; Common; C = 8; BSUH 18253.

Ranunculus abortivus L.; Small-flowered crowfoot, little-leaf buttercup, kidney-leaved buttercup or crowfoot; Floodplain woods; Common; C = 0; BSUH 18158.

RANUNCULUS FICARIA L. var. *BULBIFERA* Marsden-Jones; Lesser celandine, fig buttercup; Woodlands, especially floodplain woods; Abundant [extremely]; C = 0; BSUH 18204, 18525.

Ranunculus hispidus Michx. var. *caricetorum* (Greene) T. Duncan; Swamp buttercup; Floodplain woods, sandy shoreline of river; Infrequent but locally abundant; C = 10; BSUH 18082, 18454, 18520.

Ranunculus hispidus Michx. var. *hispidus*; Hispid buttercup; Woodlands; Infrequent; 7; BSUH 18119, 18165, 18250, 18537.

Ranunculus micranthus Nutt. ex Torr. & A. Gray; Rock buttercup, rock crowfoot; Upland woods [drier sites]; Common; C = 4; BSUH 18166.

Ranunculus recurvatus Poir. var. *recurvatus*; Hooked buttercup, hooked crowfoot; Woodlands along creeks; Infrequent; C = 5; BSUH 18157.

Ranunculus sceleratus L. var. *sceleratus*; Cursed buttercup, cursed crowfoot; Sandy shoreline of river; Rare; C = 3; BSUH 18453.

Rosaceae (Rose Family)

Agrimonia pubescens Wallr.; Downy or soft agrimony; Woodlands; Common; C = 5; BSUH 18376.

Crataegus punctata Jacq.; Dotted hawthorn; Woodlands; Infrequent; C = 2; BSUH 18285.

Geum canadense Jacq. var. *canadense*; White avens; Woodlands; Common; C = 1; BSUH 18477.

Geum vernum (Raf.) Torr. & A. Gray; Spring avens; Woodlands; Abundant; C = 1; BSUH 18179.

Prunus serotina Ehrh. var. *serotina*; Wild black cherry; Woodlands; Common; C = 1; BSUH 18472, 18499.

ROSA MULTIFLORA Thunb.; Multiflora rose; Woodlands; Common; C = 0; BSUH 18096.

Rosa setigera Michx.; Climbing prairie-rose, Illinois rose; Woodland edge; Infrequent; C = 4; BSUH 18459.

Rubus occidentalis L.; Black raspberry; Woodland edge on east side; Infrequent; C = 1; BSUH 18235.

Rubiaceae (Madder Family)

Galium aparine L.; Cleaves, annual bedstraw, stickwilly; Woodlands; Abundant; C = 1; BSUH 18124.

Galium concinnum Torr. & A. Gray; Shining bedstraw; Woodlands; Infrequent; C = 5; BSUH 18442.

Galium triflorum Michx.; Sweet-scented or fragrant bedstraw; Floodplain woods, border of seep; Infrequent; C = 5; BSUH 18392.

Rutaceae (Rue Family)

Zanthoxylum americanum Miller; Common prickly ash; Woodlands; Infrequent but locally common, widespread; C = 3; BSUH 18498.

Salicaceae (Willow Family)

Populus deltoides Marshall var. *deltoides*; Eastern cottonwood; Woodlands, especially floodplain woods; Common; C = 1; BSUH 18522.

SALIX FRAGILIS L.; Crack willow; Sandy shore of river; Rare; C = 0; BSUH 18441.

Salix nigra Marshall; Black willow; Floodplain woods, sandy shore of river; Infrequent; C = 3; BSUH 18276.

Saxifragaceae (Saxifrage Family)

Heuchera americana L. var. *americana*; Common alumroot; Woodlands; Infrequent; C = 7; BSUH 18500.

Scrophulariaceae (Figwort Family)

Leucospora multifida (Michx.) Nutt.; SYN: *Conobea multifida* (Michx.) Benth.; Conobea, narrowleaf paleseed; Sand/gravel bar; Abundant; C = 3; BSUH 18425.

Lindernia dubia (L.) Pennell var. *anagallidea* (Michx.) Cooperr.; SYN: *Lindernia anagallidea* (Michx.) Pennell; Yellowseed false pimpernel; Sand/gravel bar; Common; C = 3; BSUH 18209.

Mimulus alatus Aiton; Winged or sharpwing monkey-flower; Sand/gravel shore of river; Common; C = 4; BSUH 18214.

Mimulus ringens L. var. *ringens*; Allegheny monkey-flower; Sand/gravel shore of river; Common; C = 4; BSUH 18226.

Penstemon calycosus Small; SYN: *Penstemon laevigatus* Aiton; Eastern or smooth beard-tongue; Woodlands; Infrequent; C = 4; BSUH 18269.

Scrophularia marilandica L.; Eastern or late figwort, carpenter's square; Floodplain woods; Common; C = 5; BSUH 18331.

Veronica anagallis-aquatica L.; SYN: *Veronica catenata* Pennell; Water speedwell; Floodplain woods; Common; C = 5; BSUH 18259.

VERONICA ARVENSIS L.; Corn speedwell; Old-fields; Infrequent; C = 0; BSUH 18129.

VERBASCUM BLATTARIA L.; Moth mullein; Sandy shoreline of river; Rare; C = 0; BSUH 18450.

VERBASCUM THAPSUS L.; Common or wooly mullein; Sandy shoreline of river; Rare; C = 0; BSUH 18447.

Simaroubaceae (Quassia family)

AILANTHUS ALTISSIMA (Mill.) Swingle; Tree-of-heaven; Riverbank; Rare; C = 0; BSUH 18470.

Smilacaceae (Catbrier Family)

Smilax ecirrhata (Engelm. ex Kunth) S. Watson; Upright carrion-flower; Woodlands; Infrequent; C = 5; BSUH 18216.

Smilax tamnoides L.; SYN: *Smilax hispida* Muhl. ex Torr.; Bristly greenbrier; Woodlands; Common; C = 3; BSUH 18255.

Solanaceae (Nightshade Family)

DATURA STRAMONIUM L.; Jimsonweed; Woodland edge/row crop field; Rare but locally common; C = 0; BSUH 18195.

Solanum ptycanthum Dunal; SYN: *Solanum nigrum* auct. non L., *Solanum americanum* auct. non Mill.; Eastern or West Indian black nightshade; Floodplain woods, sandy shore of river; Infrequent; C = 0; BSUH 18224, 18403.

Tiliaceae (Linden Family)

Tilia americana L.; Basswood, American linden; Woodlands; Common; C = 5; BSUH 18496.

Typhaceae (Cattail Family)

TYPHA ANGUSTIFOLIA L.; Narrow-leaved cattail; Old-fields; C = 0; BSUH 18267.

TYPHA × *GLAUCA* Godr.; Hybrid cattail; Roadside field and pond; C = 0; BSUH 18339.

Ulmaceae (Elm Family)

Celtis occidentalis L.; Northern or common hackberry; Woodlands; Common; C = 3; BSUH 18118.

Ulmus americana L. White or American elm; Woodlands; Common; C = 3; BSUH 18471, 18527.

Ulmus rubra Muhl.; Slippery or red elm; Woodlands; Infrequent; C = 3; BSUH 18545.

Urticaceae (Nettle Family)

Boehmeria cylindrica (L.) Sw.; Small-spike false nettle; Floodplain woods; Common; C = 3; BSUH 18317.

Laportea canadensis (L.) Weddell; Canadian wood nettle; Woodlands, especially floodplain woods; Abundant; C = 2; BSUH 18311.

Pilea pumila (L.) A. Gray var. *pumila*; Canadian clearweed; Woodlands; Abundant; C = 2; BSUH 18199, 18293.

Urtica dioica L. ssp. *gracilis* (Aiton) Seland.; SYN: *Urtica dioica* L. var. *procera* (Muhl. ex Willd.) Weddell, *Urtica procera* Muhl. ex Willd.; Tall or stinging nettle; Sandy shore of floodplain woods; Infrequent; C = 1; BSUH 18334.

Valerianaceae (Valerian Family)

Valeriana pauciflora Michx.; Large-flowered valerian; Floodplain woods; Infrequent; C = 7; BSUH 18110, 18519.

Valerianella umbilicata (Sull.) Alph. Wood; Navel-fruited corn salad; Floodplain woods; Infrequent; C = 5; BSUH 18112.

Verbenaceae (Vervain Family)

Phryma leptostachya L.; American lopseed; Woodlands; Abundant; C = 4; BSUH 18218, 18238.

Verbena hastata L. var. *hastata*; Blue vervain, swamp verbena; Sandy shoreline of river; Locally common; C = 3; BSUH 18212.

Verbena urticifolia L. var. *urticifolia*; White vervain; Sandy shoreline of river; Locally common; C = 3; BSUH 18337.

Violaceae (Violet Family)

Hybanthus concolor (T.F. Forst.) Spreng.; Eastern green violet; Woods around woodland seep; Rare but locally common; C = 6; BSUH 18294, 18398.

Viola pubescens Aiton var. *pubescens*; Downy yellow violet; Woodlands; Common; C = 5; BSUH 18175.

Viola sororia Willd.; SYN: *Viola papilionacea* Pursh p.p.; Common blue violet; Woodlands; Abundant; C = 1; BSUH 18172.

Viola striata Aiton; Striped white violet, striped cream violet; Woodlands; Abundant; C = 4; BSUH 18186.

Vitaceae (Grape Family)

Parthenocissus quinquefolia (L.) Planch.; Virginia creeper, woodbine; Woodlands; Abundant; C = 2; BSUH 18122.

Vitis riparia Michx.; Frost or riverbank grape; Woodlands; Infrequent; C = 1; BSUH 18085.

Vitis vulpina L.; Frost grape; Woodlands; Common; C = 3; BSUH 18508.

LITERATURE CITED

Blank, J.R. 1987. Soil Survey of Wayne County, Indiana. USDA. Soil Conservation Service, Washington, D.C. 149.

Division of Forests and Lands. 2014. Circumneutral Hardwood Forest Seeps. At: http://www.nhdfl.org/about-forests-and-lands/bureaus/natural-heritage-bureau/photo-index/circumneutral-hardwood-forest-seep.aspx (Accessed 20 December 2014).

EPA (U.S. Environmental Protection Agency). 2013. Surf Your Watershed. At: http://cfpub.epa.gov/surf/locate/index.cfm.

Gleason, H.A. & A. Cronquist. 1991. Manual of Vascular Plants of Northeastern United States and Adjacent Canada. New York Botanical Garden, Bronx, New York. 910 pp.

Homoya, M.A., D.B. Abrell, J.R. Aldrich & T.W. Post. 1985. The Natural Regions of Indiana. Proceedings of the Indiana Academy of Science 94:245-268.

IDNR. 2013. Indiana Classified Forest and Wildlands Program. At: http://www.in.gov/dnr/forestry/4801.htm.

IDNR Nature Preserves. 2013. Endangered, Threatened, Rare and Extirpated Plants of Indiana. At: http://www.in.gov/dnr/naturepreserve/files/np-etrplants042513.pdf.

IHAPI (Indiana Historical Aerial Photo Index). 2008, At: http://129.79.145.7/arcims/IHAPI/index.html.

Keller, C., T. Crovello & K. Guild. 1984. Floristic database program (See C. Keller 1986. The computerization of regional floristic data. Proceedings of the Indiana Academy of Science 95: 412).

Magurran, A.E. 1988. Ecological Diversity and Its Measurement. Princeton University Press, Princeton, New Jersey. 179 pp.

Overlease, W. & E. Overlease. 2007. 100 Years of Change in the Distribution of Common Indiana Weeds. Purdue University Press, West Lafayette, Indiana. 270 pp.

Prast, Z.B., D.G. Ruch, D. LeBlanc, M. Russell, K.S. Badger & P.E. Rothrock. 2014. The vascular flora and vegetational communities of Munsee Woods Nature Preserve, Delaware County, Indiana. Proceedings of the Indiana Academy of Science 123, In Press.

Rothrock, P.E. 1997. The vascular flora of Fogwell Forest Nature Preserve, Allen County, Indiana. Proceedings of the Indiana Academy of Science 106:267-290.

Rothrock, P.E. 2004. Floristic quality assessment in Indiana: The concept, use and development of coefficients of conservatism. Final Report for ARN A305 4 53, EPA Wetland Program Development Grant CD975586 01. 96 p. At: http://www.in.gov/idem/water/planbr/401/publications.html.

Rothrock, P.E., H. Starcs, R. Dunbar & R.L. Hedge. 1993. The vascular flora of Mounds State Park, Madison County, Indiana. Proceedings of the Indiana Academy of Science 102:161-199.

Rothrock, P.E. & M.A. Homoya. 2005. An evaluation of Indiana's Floristic Quality Assessment. Proceedings of the Indiana Academy of Science 114:9-18.

Ruch, D.G., A. Schoultz & K.S. Badger. 1998. The flora and vegetation of Ginn Woods, Ball State University, Delaware County, Indiana. Proceedings of the Indiana Academy of Science 107:17-60.

Ruch, D.G., B.G. Torke, C.R. Reidy, K.S. Badger & P.E. Rothrock. 2002. The flora and vegetational communities of Wilbur Wright Fish and Wildlife Area, Henry County, Indiana. Proceedings of the Indiana Academy of Science 111:147-176.

Ruch, D.G., C.R. Reidy, B.G. Torke, K.S. Badger & P.E. Rothrock. 2004. Additions to the flora of Ginn Woods, Delaware County, Indiana. Proceedings of the Indiana Academy of Science 113:1-6.

Ruch, D.G., B.G. Torke, K.S. Badger, C.R. Reidy, P.E. Rothrock, R. Waltz, E.G. Urly, J.L. Chance & L. Click. 2007. The vascular flora and vegetational communities of Hayes Arboretum in Wayne County, Indiana. Proceedings of the Indiana Academy of Science 116:11-41.

Ruch, D.G., B.G. Torke, K.S. Badger, B.R. Hess, B.N. Christian & P.E. Rothrock. 2008a. The vascular flora and vegetational communities of Lick Creek Summit Nature Preserve in Wayne County, Indiana. Proceedings of the Indiana Academy of Science, (1), 117:29-54.

Ruch, D.G., B.G. Torke, B.R. Hess, K.S. Badger & P.E. Rothrock. 2008b. The vascular flora and vegetational communities of the wetland complex on the IMI Property in Henry County near Luray, Indiana. Proceedings of the Indiana Academy of Science, (2), 117:142-158.

Ruch, D.G., B.G. Torke, B.R. Hess, K.S. Badger & P.E. Rothrock. 2009. The vascular flora in three prairie cemeteries in Henry County, Indiana. Proceedings of the Indiana Academy of Science, (1), 119:35-51.

Ruch, D.G., B.G. Torke, K.S. Badger & P.E. Rothrock. 2010. The vascular flora and plant communities of the Bennett wetland complex in Henry County, Indiana. Proceedings of the Indiana Academy of Science, (1), 118:39-54.

Ruch, D.G., K.S. Badger, B.C. Daugherty, B.G. Torke & P.E. Rothrock. 2012. The vascular flora and vegetational communities of Mississinewa Woods in Randolph County, Indiana. Proceedings of the Indiana Academy of Science 121:23-44.

Stonehouse, A.L., K.S. Badger, D.G. Ruch & P.E. Rothrock. 2003. A floristic inventory and description of the structure and composition of the plant communities of Botany Glen, Grant County, Indiana. Proceedings of the Indiana Academy of Science 112:135-159.

Swink, F. & G. Wilhelm. 1994. Plants of the Chicago Region. 4th edition. Indiana Academy of Science, Indianapolis, Indiana. 921.

Tungesvick, K., D.G. Ruch, B.G. Torke, K.S. Badger & P.E. Rothrock. 2011. Additions to the flora of Mounds State Park and Preserve, Madison County, Indiana. Proceedings of the Indiana Academy of Science 120:1-11.

USDA. 2013. Natural Resources Conservation Services Plants National Database. At: http://plants.usda.gov/index.html.

WSAG (Wetland Science Advisory Group). 2005. Recommendations of the wetland science advisory group or rare and ecologically important wetland type definitions. At: www.in.gov/idem/files/rareecowetland.doc (Accessed on 8 January 2015).

WSS. 2013. Web Soil Survey, Natural Resource Conservation Service. At: http://websoilsurvey.sc.egov.usda.gov/app/HomePage.htm (Accessed on 20 July 2013).

Wiseman, D.J. & S.M. Berta. 2013. Indicator Species Analysis: An Alternative Approach to Ecosystems Geography. At: http://wiseman.brandonu.ca/article2.htm.

Yatskievych, K. 2000. Field Guide to Indiana Wildflowers. Indiana University Press, Bloomington, Indiana. 357 pp.

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