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Cover: From 2012 through 2017 Ball State University student teams have transformed the former Car Doctors brownfield in Muncie, IN, to a community green space. Many participating students gained academic credit for an 'immersive learning' experience. Top left: South end of property at the start of project; Top right: Student teams planting tall grass prairie; Bottom left: Clay material being spread over barren areas in preparation for capping with topsoil; Bottom right: South side of the property as of early 2017. For additional details concerning this fascinating project, see the article entitled "Remediation of an Indiana Brownfield: a Student Immersive Learning Experience" by John Pichtel, Faye Lichtsinn, Keegan Waggener and Amanda Howe in this issue. (Photographs by John Pichtel)

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DEGENERATIVE DISC DISEASE IN THE LOWER ILLINOIS RIVER VALLEY: MIDDLE WOODLAND, LATE WOODLAND, & MISSISSIPPIAN COMPARISONS

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ABSTRACT. Assessing degenerative disc disease during changes in prehistoric subsistence-settlement patterns can help to elucidate changes in levels of physical activity patterns during this transition. The present study compares rates and severity of degenerative disc disease in adults during the Middle Woodland, Late Woodland, and Mississippian periods in the Lower Illinois River Valley. The disease was assessed by sex and two age-at-death categories (young ≤ 45 years, old = 45+ years) in 317 individuals. Results indicate that degenerative disc scores for both young and old females decreased between the Middle Woodland (A.D. 150–400) and Late Woodland periods (A.D. 400–1050), then increased in the Mississippian period (A.D. 1050–1500). These trends were statistically significant in the older female category. Male individuals, both young and old, demonstrated significant decreases in degenerative disc scores between the Late Woodland and Mississippian periods. Within a single period, both young and older males were found to have higher degenerative disc scores than females during the Late Woodland period, while older females were found to have higher degenerative disc scores than older males during the Mississippian period. It is interesting to note the opposing directionality in male and female trends between the Late Woodland and Mississippian periods at these sites. The trends of increasing female degenerative disc scores at the same time as decreasing male scores with the intensification of maize agriculture may indicate cultural or technological changes.

Keywords: Pete Klunk mounds, Schild mounds, maize, agriculture, vertebral osteophytosis

INTRODUCTION

Degenerative joint disease is an overarching term that refers to the breakdown of joints from continuous “wear and tear” due to physical activity or repetitive motion(s), as well as aging (Jurmain 1990; Wentz 2010; Myszka et al. 2014). This process may occur as osteoarthritis at the synovial (apophyseal) facet joints and degenerative disc disease (DDD), or vertebral osteophytosis, at the (symphyseal) intervertebral joint. Important as a differential diagnostic, other non-infectious reactive processes also involve the vertebral column (e.g., diffuse idiopathic skeletal hypertrophy [DISH] and ankylosing spondylitis), and mechanical injury may have contributing factors that likely influence incidence and severity. These factors can include age, body mass, sex, genetics, trauma, and physical activity to name just a few (e.g., Brown et al. 2008; Rojas-Sepulveda et al. 2008; Bernard et al. 2010; Chang 2010; Duncan et al. 2012; Listi & Manhein 2012; Goode et al. 2013; Suri et al. 2013, Myszka et al. 2014).

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DDD (vertebral osteophytosis) occurs at the intervertebral joint, consisting of two adjacent vertebral bodies and the intervertebral disc. It is most commonly attributed to continual pressure on the vertebral column inducing breakdown of the intervertebral disc (Kim et al. 2012; Van der Merwe et al. 2006). The vertebrae then attempt to stabilize the joint through reactive bone changes including progressive osteophytes, lipping, and porosity of the superior and (mostly commonly) inferior surfaces of the vertebral body (Fardon & Milette 2001; Rojas-Sepulveda et al. 2008). Schmorl’s nodes have also sometimes been attributed to DDD, but while these marks may relate to herniation of the nucleus pulposus, why and to what degree DDD contributes to these phenomena is unknown (Plomp et al. 2012). For this reason, Schmorl’s nodes were not included in the present analysis.

Recent studies also have found that the vertebrae farthest from the direct line of gravity between the top of the head and the floor (e.g., C5, T9, T10, L4) are most likely to show signs of DDD. This is believed to be due to higher levels of pressure on those vertebrae (Knusel et al. 1997; Van der Merwe et al. 2006; Kim et al. 2012) and

relates to the argument that Maat et al. (1995) make that DDD is more strongly affected by weight-bearing, while vertebral osteoarthritis may be more affected by specific body movements. The association between DDD and enthesal changes, particularly in the lower limb, provides further evidence that DDD is associated with weight-bearing physical activity (Myszka et al. 2014).

Assessing DDD during prehistoric subsistence-settlement transitions provides insights into the changes in physical activity that accompanied changes in food procurement strategies (Stout 1978; Ruff et al. 1984; Bridges 1991, 1992; Bridges et al. 2000; Robling & Stout 2003). Such a subsistence-settlement transition occurred in the Lower Illinois River Valley from the Middle Woodland period (A.D. 150–400), through the Late Woodland period (A.D. 400–1050), to the Mississippian period (A.D. 1050–1500) (Pickering 1984; Rose 2008; Husmann 2011; Spencer 2014). Degenerative disc disease in the Lower Illinois River Valley has not been well studied. The present preliminary assessment of DDD is one piece in a larger study that assesses changes in health status associated with the transition from early horticulture to agriculture in the Lower Illinois River Valley (Husmann 2011). The human remains investigated are from two sites that straddle three adaptational periods: the Pete Klunk mound group and the Schild mound group (Fig. 1). These remains are currently housed in the Osteology and Paleopathology Laboratory at Indiana University, Bloomington.

ARCHAEOLOGICAL CONTEXT

The Middle Woodland period in this area dates to A.D. 150–400. The subsistence strategy in the Lower Illinois Valley during the Middle Woodland complex was both foraging (nuts, wild plants, animals) and horticulture of the local cultigens known as the Eastern Agricultural Complex (EAC), which included goosefoot (*Chenopodium berlandieri* Moq.), knotweed (*Polygonum erectum* L.), and marsh elder (*Iva annua* L.) (Gregg 1974; Yerkes 2005). With this combination of foraging and horticulture, there is still some debate about the level of sedentism during the Middle Woodland period. Yerkes (2006) suggests that high mobility was still necessary to obtain sufficient food and dietary fiber in Ohio, while Mangold & Schurr (2006) advocate a similar situation for northern Indiana Middle Woodland groups.

The Late Woodland period in the Lower Illinois River Valley dates from A.D. 400–1050 (Reynolds et al. 2015). During the Late Woodland, people continued to cultivate the EAC. However, we also begin to see some of the first forays into maize consumption in this area (Asch et al. 1979; Studenmund 2000; Rose 2008). Rose (2008) completed isotopic analyses on skeletal remains from the Late Woodland components of the Knight, Joe Gay, and Yokem sites. She found little evidence of maize consumption in the early Late Woodland remains, but by the latter part of the Late Woodland, many more remains (over half of the remains sampled from Yokem) were found to have ^{13}C levels indicative of maize consumption. However, her analysis of skeletal remains from the Schild site produced more negative ^{13}C levels throughout the Late Woodland period, an indication that these individuals were not yet uniformly participating in significant maize consumption. Simon (2014) has posited that these Late Woodland forays into maize represent early “experiments” with maize that ultimately failed, necessitating a later re-introduction of maize to the area. Her research, however, focuses only on the botanical evidence for maize and does not consider the skeletal isotopic data. The isotopic data support a more continual progression throughout the region, though additional research is necessary to elucidate how the botanical and isotopic data fit together.

According to other studies of skeletal remains, Late Woodland females (including those from Pete Klunk, Koster, Schild, Yokem, Ledders, Hacker, and Kuhlmann) had an increase in upper limb strength compared to females of the Middle Woodland period (including remains from Pete Klunk, Gibson, and L’Orient) (Bridges et al. 2000). This additional robustness may be the result of increased food processing demands and is most likely due to increased consumption of the EAC starchy seed plants. Buikstra et al. (1987, 1994) also have noted both the minimal maize consumption and the beginning of sexual distinctions in diet during the Late Woodland period. Their studies were completed on numerous sites from the Lower Illinois River Valley, including Ledders, Helton, Schild, and Gibson, and found that males were consuming more protein while females were consuming more carbohydrates. Settlement patterns in the Illinois River Valley varied during the Late Woodland: some sites likely had year round occupation while others appear to be seasonal (VanDerwarker et al. 2015).

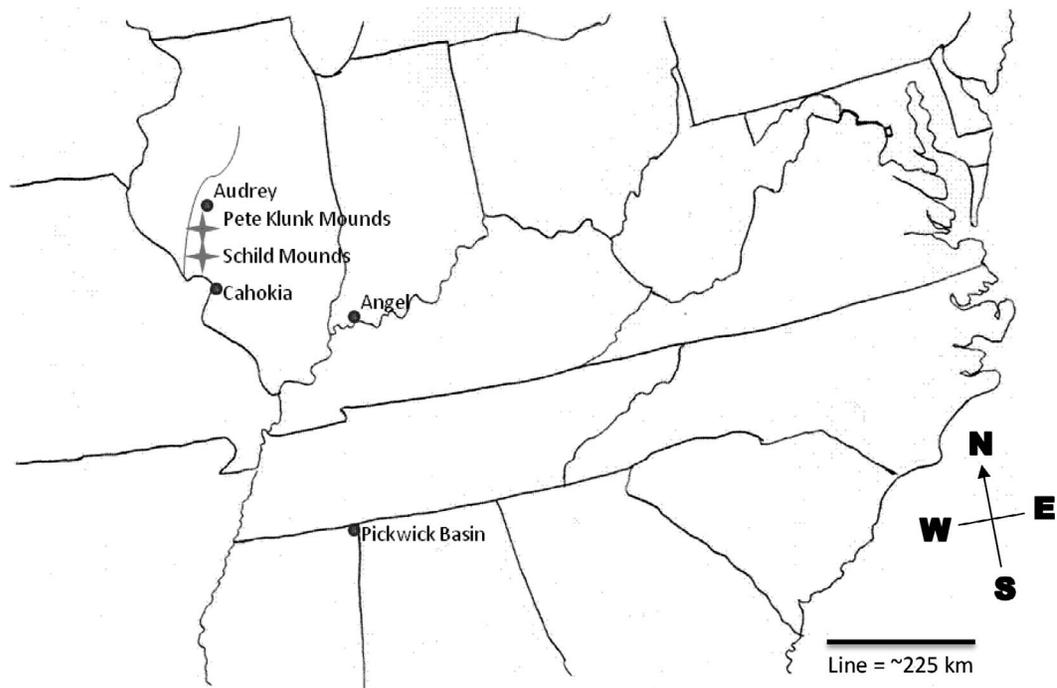


Figure 1.—Map of the Eastern United States with locations of the Pete Klunk & Schild mound groups with other relevant sites (1" = ~ 200 km)

Maize agriculture ultimately took off in the Lower Illinois River Valley by the Mississippian period (A.D. 1050–1500) resulting in much more sedentary, aggregated village settlements than either of the previous periods (Reynolds et al. 2015). With this increase in maize consumption, Bridges et al. (2000) found a decrease in female upper limb strength indicators compared to the Late Woodland. Possible food processing-related interpretations of these data include lower processing requirements for maize or potentially increased efficiency in processing, possibly related to new soaking or boiling technologies (Bridges et al. 2000). With these new soaking or boiling technologies came the advent of gruel. Gruel allowed weaning to occur at an earlier age and, thus, may have facilitated (along with immigration into these sites from surrounding areas) the population growth (Buikstra et al. 1986). Thus, the period from the Middle Woodland through the Mississippian demonstrated an overall change from foraging and horticulture of local cultigens to a dependence on maize agriculture in order to sustain the population. This span also demonstrated new innovations in food processing as the subsistence patterns changed.

The Pete Klunk and Schild mound groups.—The Pete Klunk mound group (11C4) in Calhoun County, Illinois was excavated during the 1960 and 1961 seasons by Gregory Perino and consists of 14 mounds running north–south on a bluff overlooking the Illinois River (Perino 1968). The mounds were associated with the Kampsville village site and are believed to be part of the mortuary population for that site. Excavations of these mounds uncovered 300 Middle Woodland burials and about 80 Late Woodland burials. Early uncalibrated radiocarbon dates placed the Middle Woodland component in Mound 1 at A.D. 175 ± 75 (Perino 1968). The Late Woodland component was dated to A.D. 613 ± 110 in Crematory B of Mound 8, and to A.D. 780 ± 120 in Crematory A of Mound 10 (Perino 1973a).

Perino excavated the Schild mounds in 1962 and 1963. This site included nine Late Woodland mounds and two non-mounded knolls (A & B) containing interments of Mississippian origin (Perino 1973b). Within the mounds were over 200 Late Woodland burials and approximately 250 Late Woodland – Emergent Mississippian

Table 1.—Classifications and descriptions of degenerative disc disease (DDD) scoring.

Original classification	Specific classification	Numeric score	Description
A	A	1	No sign of additional bone growth, porosity, or pitting
	A-	2	
B	B+	3	Minimal additional bone growth, no restriction of movement
	B	4	
	B-	5	
C	C+	6	Bone growth present, but no fusion and/or only minor restriction of movement
	C	7	
	C-	8	
D	D+	9	Prolific additional bone growth resulting in limited movement or fusions of vertebrae
	D	10	

burials. Recent calibrated radiocarbon dates average around A.D. 830 for the Late Woodland component at Schild (see Spencer 2014 for more detailed dates).

The two knolls at Schild contained nearly 300 Mississippian individuals (Perino 1971) with recent calibrated radiocarbon dates that average around A.D. 1030 (Spencer 2014). Unfortunately, the habitation site with which Schild was affiliated remains something of an enigma (Perino 1971; Goldstein 1980). Delaney-Rivera (2004, 2007) completed a ceramic analysis that proposed that the Audrey village site and the Schild mounds were part of the same sociopolitical climate, yet she posits that these two sites have their origins in two different groups. The Audrey site is thought to be the result of Mississippian colonization from the area around Cahokia, known as the American Bottom, while the Schild mounds were associated with descendants of the local Late Woodland population who had acculturated to a Mississippian lifestyle due to interactions and movements of people. The Schild site is located roughly 15 km south of the Audrey site.

METHODS

The superior and inferior surfaces of all present vertebral bodies were examined for signs of DDD. All vertebrae present were aligned in anatomical position prior to observation. Of the 317 individuals assessed, only 26 individuals did not include representation from all three vertebral regions assessed (cervical, thoracic, and lumbar). Of these 26 individuals, 11 did not include any lumbar vertebrae (four from the Middle Woodland, six from the Late Woodland, and one Mississippian), while 15 did not include any cervical vertebrae (four from the Middle Woodland, six from Late Woodland, and five Mississippians). In addition,

the average number of vertebrae present from each region was as follows: cervical – 5.8 vertebrae, thoracic – 10.7 vertebrae, and lumbar – 4.4 vertebrae. Thus, these individuals were all quite well preserved.

Evidence of DDD, including osteophytic lipping, porosity, and pitting of the vertebral surfaces was categorized as A, B, C, or D. See Table 1 for descriptions of these categories. For photographs comparable to the four stage system initially utilized here, see Jurmain & Kilgore (1995).

A system of pluses and minuses (A, A-, B+, B-, etc.) was later implemented for additional detail resulting in scores assigned between one and ten (A=1, D=10) and all vertebrae were re-analyzed according to the new system. The vertebrae with the worst degeneration were scored first and then the number of vertebrae that demonstrated that level of degeneration was evaluated to determine if the score for the vertebral column as a whole should be at a less severe level. For example, if the worst degeneration present on an individual set of vertebrae was a 7 (C), but it only occurred on two or three vertebrae, the vertebral column from this individual would be more likely to receive a 5 (B-) or a 6 (C+) while an individual where half or more of the available vertebrae demonstrated that 7 (C) level of degeneration would receive a 7 for the entire vertebral column. This provides a broad assessment to evaluate differences through time among male and female adults. All individuals were scored by the author within a six month time-frame and several individuals were rechecked to ensure consistency across time. For additional detail, see Husmann (2011).

All available remains also were evaluated morphologically for signs of systemic infection

Table 2.—Age data for each period. MW = Middle Woodland, LW = Late Woodland, Miss = Mississippian.

Period	N	Mean	Standard deviation	Median
MW	110	36.6	9.7	35.7
LW	77	37.0	9.8	37.0
Miss	130	41.7	10.9	42.0

(e.g., periostosis, osteomyelitis, lytic lesions) or other pathological conditions that may present confounding variables (e.g., DISH, ankylosing spondylitis, trauma). Any remains with confounding or predisposing conditions were excluded from the analysis.

All statistical analyses were completed using SPSS 23 (IBM Corp. 2013). Non-parametric tests (Kruskal-Wallis tests, Mann-Whitney U tests, and associated Dunn-Bonferroni post-hoc tests) were utilized because normality tests were not supported. However, equal variances were calculated using the Brown-Forsythe method ($p = 0.405$).

The skeletal sample.—Sample selection was limited to adults, as determined by the eruption of the third molar, and the presence of the eighth or ninth thoracic vertebrae (these were necessary for additional analyses not presented here). The adult age criteria were used because DDD is generally not present until around age twenty (Jurmain 1990; Maat et al. 1995; Van der Merwe et al. 2006).

The total sample for this study consisted of the following adults: 117 individuals found in Middle Woodland contexts, 81 individuals found in Late Woodland contexts, and 132 individuals found in Mississippian contexts. The Middle Woodland sample, by necessity, came completely from the Pete Klunk mounds, while the Mississippian sample was equally limited to Schild. The Late Woodland sample was drawn from both sites depending on the presence of vertebrae and the

ability to determine other necessary parameters (age, sex, etc.). The present study evaluates two sites in close proximity (Pete Klunk & Schild) in order to reduce the confounding influence of genetics and attempts to control for age and sex by completing separate analyses based on these factors. Age was determined using multifactorial methods developed by Lovejoy et al. (1985) with ecto-cranial suture closure (Buikstra & Ubelaker 1994), pubic symphyses (Brooks & Suchey 1990), auricular surfaces (Buckberry & Chamberlain 2002), and first ribs (DiGangi et al. 2009) as the components. See Table 2 for descriptive statistics for age in each period examined. Sex was determined by assessing the pelvis and/or skull, as preservation allowed, and following the standards put forth by Buikstra & Ubelaker (1994). Additional explanation and examples of age calculations may be found in Husmann (2011).

Analyses of these data were undertaken by period (Middle Woodland, Late Woodland, or Mississippian) and segregated by sex and age (Table 3). Individuals aged above and below 45 years were analyzed separately to help identify the effects related to aging in each cultural period. The age of 45 years was chosen due to the potential impact that hormone-related changes accompanying the onset of menopause may have to the individual's ability to maintain bone density. In modern populations, this perimenopausal period generally occurs in the mid-forties (Moore et al. 2014; McKinley et al. 2015).

RESULTS

The results will be presented in two subdivisions: young to middle adults (aged below 45 years) and older adults (aged above 45 years). These must be presented separately due to the confounding factor that age (especially postmenopausal ages) can have on DDD scores and the fact that previous studies (Bridges 1991; Pickering 1994) have focused solely on the young

Table 3.—Sample sizes divided by age, sex, and period association.

Sample demographics	Middle woodland	Late woodland	Mississippian
< 45			
Females:	52	34	40
Males:	33	25	38
45+			
Females:	13	9	30
Males:	12	9	22
Total	110	77	130

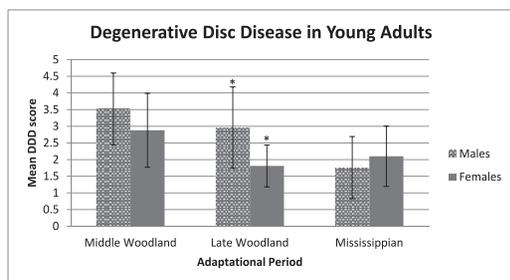


Figure 2.—Graph of the mean degenerative disc disease scores for male and female remains aged less than forty-five during the Middle Woodland, Late Woodland, and Mississippian periods. (* indicates periods in which male and female scores were significantly different.)

to middle adults. Within each section, I will present the results comparing males across periods and females across periods (Middle Woodland, Late Woodland, and Mississippian), followed by the results of pairwise comparisons between males and females within each period.

Young to middle adults.—The average DDD scores for young male and female remains are listed in Table 4 and trends are shown in Fig. 2. For males, lower DDD scores were noted in individuals from the Late Woodland contexts than those from the Middle Woodland contexts. This trend continued into the Mississippian period, which showed the lowest mean scores of all three groups. For young males, Kruskal-Wallis tests demonstrated that the differences in DDD levels among the three periods were significant ($p < 0.000$). Pairwise comparisons found a significant difference between males from Middle Woodland and Mississippian complexes ($p < 0.000$) and also between males from the Late Woodland and Mississippian complexes ($p = 0.009$).

For females, a decrease in DDD scores was found between the Middle Woodland and the Late Woodland periods, with a slight increase during the Mississippian (see Fig. 2). Kruskal-

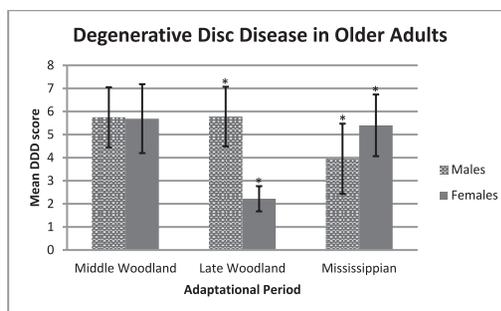


Figure 3.—Graph of the mean degenerative disc disease scores for male and female remains aged more than forty-five years during the Middle Woodland, Late Woodland, and Mississippian periods. (* indicates periods in which male and female scores were significantly different.)

Wallis tests demonstrate no significant differences between young females in the different periods ($p = 0.061$).

Mann-Whitney U tests between males and females within each period revealed no significant differences during the Middle Woodland and Mississippian periods. However, during the Late Woodland period males were found to have significantly higher DDD scores than females ($p = 0.025$).

Older adults.—The average DDD scores for older male and female remains are listed in Table 4 and trends are shown in Fig. 3. For males, a drop in DDD scores is seen between the Late Woodland and Mississippian periods. However a Kruskal-Wallis test returned no significant differences in DDD scores among any of the periods in question for males in this age bracket.

For females, a sizeable drop in mean DDD scores was seen during the Late Woodland, but the scores rebounded during the Mississippian period (see Fig. 3). Thus, significant differences were found between the Middle and Late Woodland ($p = 0.009$) and between the Late Woodland and Mississippian ($p = 0.004$).

Table 4.—Mean degenerative disc disease (DDD) scores for male and female remains from the Middle Woodland (MW), Late Woodland (LW), and Mississippian (Miss) periods. SD = standard deviation.

Mean DDD scores	Males < 45	Females < 45	Males > 45	Females > 45
MW	3.52 (SD: 2.15)	2.88 (SD: 2.22)	5.75 (SD: 2.60)	5.69 (SD: 2.98)
LW	2.96 (SD: 2.44)	1.81 (SD: 1.26)	5.78 (SD: 2.59)	2.22 (SD: 1.09)
Miss	1.76 (SD: 1.87)	2.10 (SD: 1.81)	3.95 (SD: 3.05)	5.40 (SD: 2.67)

Mann-Whitney U tests also were run between males and females within each period. Significant differences were seen in both the Late Woodland and the Mississippian periods. During the Late Woodland, males were found to have higher DDD scores ($p < 0.000$) while during the Mississippian, females were found to have higher DDD scores ($p = 0.037$).

DISCUSSION

Young to middle adults.—This analysis found no significant differences between DDD scores of young male and young female remains aged less than 45 years when the remains were found in Middle Woodland or Mississippian contexts. This is consistent with Bridges (1992) review of arthritis in the prehistoric Americas, who reported no significant differences in appendicular or vertebral osteoarthritis (OA) between males and females of hunter-gatherer societies in the prehistoric Americas (most similar to Middle Woodland, though admittedly Middle Woodland individuals were practicing some horticulture). However, these results are not consistent with studies of agricultural populations outside of the American Midwest, which found significant differences in DDD scores between males (higher) and females (lower) (Van der Merwe et al. 2006; Wentz 2010; Kim et al. 2012). This difference may relate to unidentified biological differences between the populations studied, the terrain being worked, or cultural divisions of labor in those societies that were unlike those at Klunk and Schild. In addition, it must also be remembered that DDD may be more strongly affected by weight-bearing, while vertebral OA may be more affected by specific body movements (Maat et al. 1995), so these differences may also relate to different specific body movements between the sexes while weight-bearing activities remained comparable.

Pickering (1984) also found no significant differences in OA scores between the sexes based upon his analysis of young to middle adult appendicular and vertebral remains from Middle Woodland, Late Woodland, and Mississippian components of the Lower Illinois River Valley, including the Pete Klunk and Schild mound groups. However, in the present analysis, Late Woodland young male remains from these sites were found to have significantly higher degenerative disc scores than young female remains. Pickering (1984) may not have found these results

because he split the Late Woodland period into early and late components, which he grouped with the Middle Woodland and Mississippian periods, respectively. Unfortunately, in doing so, he lost some of the resolution for the Late Woodland period itself. However, weight-bearing versus non-weight-bearing activities also may be affecting these results.

Cross-period analyses for young males demonstrated a general trend of decreasing degenerative disc scores from the Middle Woodland to the Mississippian period with the Late Woodland to Mississippian transition showing a significant decrease. This is consistent with the general trend toward decreasing degenerative scores with the advent of intensive agriculture, as noted by Bridges (1992), who reviewed OA literature, and Jurmain (1990), who examined vertebral OA and DDD. On the other hand, Bridges (1992) also found the Lower Illinois River Valley (including Klunk and Schild) to be an exception to this rule and suggested regional variation with the new subsistence economy as one explanation. Nonetheless, she does note that the Lower Illinois River Valley trend she reports (increasing degenerative scores) was generally less prevalent in males, which she attributes to less involvement of males in agricultural related chores.

For young to middle aged females, the cross-period analysis demonstrated no significant results. This does not fit with the pattern of increasing OA with agriculture in the Lower Illinois River Valley, as reported by Bridges (1992). Pickering (1984) overall also found more severe degenerative disease scores for females with the intensification of maize agriculture, though differences in the categorizations of age and period make direct comparisons difficult. One interpretation of these results would be that the weight-bearing activities for young to middle aged females were fairly consistent, or at least comparable, throughout this transition while the need for other movements (e.g., twisting) were increasing.

Older adults.—While many other studies have not examined older adults due to the relationship between age and degenerative diseases (Bridges 1991, Pickering 1984), this study specifically includes older adults in the analysis since these are the individuals that have been engaged in repetitive tasks (whatever they may be) the longest and thus are more likely to have their effects on the bones. By keeping the analyses separate from the younger

adults, the differences due purely to aging should be somewhat consistent across periods. This analysis found no significant differences between degenerative disc scores of male and female remains aged more than 45 years recovered from Middle Woodland contexts, which is consistent with the analysis of the young to middle adults. However, male remains from the Late Woodland period had significantly higher degenerative disc scores than female remains from this period. This is consistent with the analysis completed on young to middle adult remains for these periods and may suggest similar workloads throughout the life span. Of course, it must be acknowledged that the sample sizes are limited for this analysis (i.e., eight Late Woodland males and nine Late Woodland females aged over 45). In addition, female remains were found to have higher degenerative disc scores than male remains from the Mississippian period, which was not seen in the younger adults or reported with other populations (Van der Merwe et al. 2006; Wentz 2010; Kim et al. 2012). One factor in this aberration may be the potential for decreasing vertebral bone density caused by increased acidity in the diet (from increased maize consumption), though this would then be expected in other populations with high acidity diets as well.

Cross-period analyses for male remains aged over 45 years found no significant differences, though a marginally lower average degenerative disc score was noted for male remains from the Mississippian period than from Middle Woodland or Late Woodland contexts. Again, this lack of statistical significance may relate to the limited sample size in these categories. By contrast, in the cross-period analyses of females aged over 45 years, remains associated with the Late Woodland period had significantly lower degenerative disc scores than either of the other two periods assessed. This is consistent with the general directionality of the results from younger females, though the older females showed significant differences while the young female analyses did not reach significance (possibly because they had not been participating in the activities as long as the older females).

Comparisons.—Within this series of trends, a few of particular interest come to light. First, in both younger and older males there is a drop in degenerative disc scores between the Late Woodland and the Mississippian. This is

indicative of some variation in the weight-bearing activities between these sites and the wide variety of those reviewed by Bridges (1992). Meanwhile, in both younger and older females, there is a drop in degenerative disc scores between the Middle Woodland and the Late Woodland. These may be related to changes in harvesting methods between these periods since this work is most commonly attributed to women (Bridges 1992; Pickering 1984). During the Late Woodland, innovations in harvesting of the EAC plants may have made these activities easier (Bridges 1989a, 1989b; Buikstra et al. 1986). With the reliance on maize in the Mississippian period, some of these innovations may have been discarded as less efficient or effective, resulting in an increase of female degenerative disc scores back toward their previous levels. Pickering (1984) also noted that increased carrying loads (upper spine) and increased bending with short handled hoes (lower spine) may be required for agricultural intensification. These may be reflected in the DDD scores since they are weight-bearing activities, and thus are more likely to appear as DDD than OA (Maat et al. 1995).

It is interesting to note that the directionality of these DDD changes (decreasing from Middle to Late Woodland, and increasing from Late Woodland to Mississippian) is in direct contrast to upper limb strength indicators (higher in Late Woodland compared to Middle Woodland or Mississippian) (Bridges et al. 2000). This combination of upper limb and DDD trends suggests that women in the Late Woodland were spending more time at upper limb repetitive activities and less time in weight-bearing activities than the preceding or following periods. Thus, it is possible that the females could have spent more time using processing techniques that utilized the upper limb and took stress off of the vertebral column during the Late Woodland (e.g., nut pounding), but spent more time during the other periods on activities that were easier on the upper limb, but more stressful for the weight-bearing vertebral column (e.g., harvesting or carrying loads).

Another interesting trend is that the directionality of the changes in degenerative disc scores between the Late Woodland and the Mississippian periods is positive (increasing) for females, but negative (decreasing) for males, regardless of age category. This is consistent with a change in the sexual division of labor between the Late Wood-

land and Mississippian periods at these sites more than simply just the decreased utilization of innovations from previous agricultural endeavors. If the men were continuing to do fewer weight-bearing activities, why weren't the women? One possible explanation may be a cultural reason that female scores increased while male degenerative disc scores continued to decline, especially since this trend began shortly after the sex-related dietary distinctions noted by Buikstra et al. (1987, 1994). Age alone cannot be used to explain these opposing trends since there is a larger sample of older remains during the Mississippian period than either the Middle or Late Woodland periods for both males and females (Table 3), yet this opposing directionality in degenerative disc trends. If age were the major contributing factor, one would expect to see increased DDD scores in both sexes given that the average age was higher for both males and females. An alternative interpretation of these data could be that the innovations the males had found during the Late Woodland continued to work with the intensification of maize agriculture while the innovations that the females were using were no longer viable. However, this has not been supported by any other analyses.

Future directions.—Future analyses of the vertebral regions (lumbar, thoracic, cervical) separately and an assessment of the asymmetry of the vertebrae would be beneficial. Analyzing vertebral regions separately would elucidate their differential responses to load bearing, stability, and biomechanical processes (Bridges 1994; Jurmain & Kilgore 1995). Evaluating the asymmetries of the vertebrae and their associated osteophytes may help to parse out which changes result from differences in activities and which are simply part of the aging process (Bridges 1994). In addition, future analyses of physical activity markers on the appendicular skeleton would be beneficial in determining what types of activities might help account for the differences seen here.

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CHECKLIST OF INDIANA FUNGI I: MACROFUNGI

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ABSTRACT. A checklist of macrofungi was compiled for Indiana as part of a larger effort to document fungi within the state. Our study compiled records of Indiana fungi from digitized specimen data available online through the Mycology Collections data Portal (<http://mycoportal.org>). These data were supplemented with records from the scientific literature. While several small checklists of Indiana fungi exist, the majority of these being published previously in the Proceeding of the Indiana Academy of Science, our study represents the first to comprehensively compile all the available data on Indiana fungi. Overall, more than 19,000 records of Indiana fungi were examined, with 1410 species of macrofungi being documented in this publication. These species represent 24 fungal orders from two major phyla, with 757 species in this checklist being reported in the literature for the first time here. Our study also recovered records documenting other groups of Indiana fungi, such as microfungi, which will be covered in subsequent publications.

Keywords: Ascomycota, Basidiomycota, checklist, Eumycota, fungi, taxonomy

INTRODUCTION

A wide range of natural communities exist in Indiana, with six general ecoregions recognized by Woods and colleagues (1998): the Eastern Corn Belt Plains, Huron/Erie Lake Plains, Interior Plateau, Interior River Valleys and Hills, Central Corn Belt Plains, and Southern Michigan/Northern Indiana Drift Plains. While over half the land area of the state is now devoted to agriculture (Vilsack & Clark 2012), it was once largely forested, and the previously dominant oak-hickory and beech-maple plant communities persist primarily as isolated stands (Friesner 1936; Clark 1994; Whitaker et al. 2012). Although the majority of agricultural land in Indiana is devoted to row crops (Vilsack & Clark 2012), some areas in the state support a high level of vascular plant diversity. For example, the Indiana Dunes National Lakeshore, encompassing some 6,000 ha, has nearly 1,200 native plant species (Wilhelm 1990) in a mosaic of habitats that include xeric sand dunes, sedge meadows, oak savanna, pannes, bogs, fens, kettle lakes, and mesic prairies. Considering the potential for tight linkages between vascular plants and their fungal counterparts (Van der Heijden et al. 1998), a high level of fungal diversity might also be expected across the state.

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Members of the kingdom Eumycota (Fungi) are filamentous, or sometimes unicellular, heterotrophic organisms with absorptive nutrition. The diverse forms include mushrooms, rusts, smuts, and yeasts. Many fungal species (e.g., mycorrhizal forms) are essential symbiotic partners of vascular plants, providing them with nutrients such as phosphorus (Bolan 1991), while others are key in cycling carbon through the decay of organic matter (Lindahl et al. 2002). Recent research also suggests fungal pathogens are an increasing threat to the health of animals, plants, and even ecosystems (Fisher et al. 2012). Given this diversity of forms and of ecological roles, it is reasonable to expect fungal diversity to be vast, with estimates ranging from 1.5 to 5.1 million species worldwide (Blackwell 2011). In comparison to this extensive diversity, the number of described species (ca. 100,000) is proportionally low, and understanding of fungal diversity even in temperate regions, such as Indiana, is deficient. At the same time, recent projects aimed at digitizing our natural history collections (e.g., Baird 2010; Conway 2010; Beaman & Cellinese 2012; Blagoderov et al. 2012) have facilitated access to data on organismal diversity. Taking inspiration from these digitization efforts, this study relied largely on digital resources to document our current knowledge of fungal diversity in Indiana.

Over 19,000 digitized records of fungal specimens were examined online in public databases. To supplement these data, we carried out online searches of records of fungi from the state in the

scientific literature. While records of Indiana fungi have been published in the broader literature (e.g., Schmit et al. 1999; Harmon & Latin 2005; Wilson et al. 2014), a wealth of digitized publications (e.g., Van Hook 1911, 1912, 1913, 1916, 1921a, 1921b, 1922, 1924, 1925, 1926, 1928, 1929a, 1929b, 1930, 1935; Jackson 1918, 1921a, 1921b; Anderson & Anderson 1920; Fink & Fuson 1920; Van Hook & Busted 1935) were discovered from the Proceedings of the Indiana Academy of Science, with documentation of fungal diversity in the state beginning with the first published volume (Underwood 1892) and continuing into the twenty-first century (Ruch et al. 2001). This study summarizes the sizable fungal diversity known to exist across Indiana. Due to length, the Indiana fungal checklist will be published in three parts: macrofungi (e.g., bracket fungi, mushrooms, and puffballs), microfungi (e.g., molds and rusts), and lichenized fungi (i.e., lichens) of Indiana.

METHODS

The preliminary fungal species list was compiled through a search of Indiana specimens housed in 72 public collections, universities, and natural history museums across the United States and Canada. The specimen records from these institutions are available online through the Mycology Collections data Portal (MyCoPortal, <http://mycoportal.org>). MyCoPortal is the official repository of data for the Macro- and Microfungi Collections Consortia (MaCC and MiCC, respectively) Thematic Collections Networks, digitization efforts funded by the NSF's Advancing Digitization of Biological Collections (ADBC) program. Data from MyCoPortal are served on the World Wide Web using Symbiota software (Gries et al. 2014). Its database currently contains over three million fungal specimen records from around the world, with more records being added daily and digitization efforts funded until 2018. Records obtained from MyCoPortal specimen data have been supplemented with records of Indiana fungi from the scientific literature. A number of historical mycological publications from the Proceedings of the Indiana Academy of Science (Arthur 1897; Lee 1929; Busted 1936; Herre 1944; Cottingham 1947, 1948, 1949, 1950, 1951, 1952, 1954, 1956; Wolf 1972; Evans-Ruhl et al. 1982; Schoknecht 1982; Hyczyk 1998) were accessed online through the Academy's digital archive (<https://journals.iupui.edu/index.php/ias/index>). Several additional pub-

lications from other journals (Reddick 1907; Cooke 1975; Avis et al. 2006, 2008, 2017; Gurung et al. 2011; Avis 2012; Leacock et al. 2016) were also used in compiling the complete list of Indiana fungi.

In order to verify current taxonomic status and check synonymy for each of the scientific names included in the checklist, the online fungal nomenclature databases MycoBank (<http://www.mycobank.org>) and Index Fungorum (<http://www.indexfungorum.org>) were consulted. The system of classification used here follows that outlined in MycoBank. The taxa included within this checklist of macrofungi generally have fruiting bodies that reach well over a centimeter in height or diameter (though see the discussion below). Each species entry in the checklist is annotated to indicate if the scientific name was sourced from MyCoPortal specimen records and/or a reference from the literature (see the 'Key' below). If the species had not been previously reported in the literature as occurring in Indiana, then the annotation 'NR' indicates a new published record for the state. All statistics (viz., numbers of new records and species) are considered only at the level of species; however, citations for new records are indicated (but not counted) for infraspecific taxa, such as varieties. Infraspecific taxa (e.g., *Helvella macropus* var. *brevis*) were included whenever encountered in databases or the literature and also were listed as the binomial (*Helvella macropus*) when it was not known if other species-level records represented a distinct infraspecific taxon (e.g., *Helvella macropus* var. *macropus*).

RESULTS AND DISCUSSION

Over 19,000 records were recovered in MyCoPortal and the scientific literature documenting a wide range of fungi from the state of Indiana (Figs. 1–3). After updating the taxonomic names and checking for synonymy, as well as excluding microfungi, lichens, and slime molds (protozoans traditionally included in mycological collections), 1410 species of macrofungi were recovered. Of these 757 (54%) are reported from Indiana for the first time, adding greatly to our knowledge of fungal diversity in the state. Overall, the species included in this macrofungi checklist represent two major phyla (Ascomycota, Basidiomycota) in the Eumycota. Within these phyla, 24 fungal orders are represented, most abundantly in the Basidiomycota (19 orders containing 1326 species). The complete list of all non-lichenized fungi



Figure 1–3.—Indiana macrofungi. A wide range of fungal diversity can be found within the state of Indiana, including macroscopic fruiting bodies recognizable as (1) mushrooms (*Marasmius* sp.), (2) turkey-tail fungi (*Trametes* sp.), and (3) stink horns (*Mutinus* sp.).

is being made available online through the MyCoPortal (<http://mycoportal.org>, see the Macrofungi of North America: Regions research checklist page), and will be updated regularly as more data become available.

Numerous basidiomycetous corticioid fungi (e.g., *Botryobasidium*, *Corticium*, *Ramaricium*, *Septobasidium*, *Subulicystidium*, *Tulasnella*), having little topology but growing to cover a large area, are included among the macrofungi following mycological tradition. Again following tradition, ascomycetous fungi recorded from Indiana (e.g., *Hypoxyton*, *Kretzschmaria*, *Rosellinia*) with perithecial ascomata (viz., pyrenomycetes) will be included in the checklist of microfungi, even though their growth habit is superficially similar to that of corticioid fungi. The same is true for a number of other minute ascomycetes (e.g., *Cordyceps*, *Dasyscyphus*, *Isaria*, *Mitrula*) that skirt the boundaries between macro- and micro-fungi, as well as those that cover fruiting bodies of

macrofungi as parasites (e.g., *Hypomyces*). However, included among the macrofungi are a few of the smaller ascomycetous genera (e.g., *Bulgaria*, *Camarops*, *Daldinia*, *Galiella*, *Leotia*, *Microglossum*, *Microstoma*, *Peziza*, *Xylaria*, *Xylocoremium*) known to generally produce somewhat conspicuous ascomata.

There has been considerable mycological activity in the state, with the earliest specimens collected in 1804 and the first documentation in the literature dating to the late 1800's (Rose 1886). After a more intense period of activity, roughly 1885–1925, workers have consistently collected specimens in the field throughout the decades (Fig. 4). The fungal species in the Indiana checklist demonstrate the wide range of fungal diversity that exist in the state, and a number of species in the macrofungi checklist have importance to humans, such as deadly poisonous mushrooms (e.g., *Amanita phalloides*, see Pringle & Vellinga 2006) and choice edibles (e.g., *Cantharellus* and

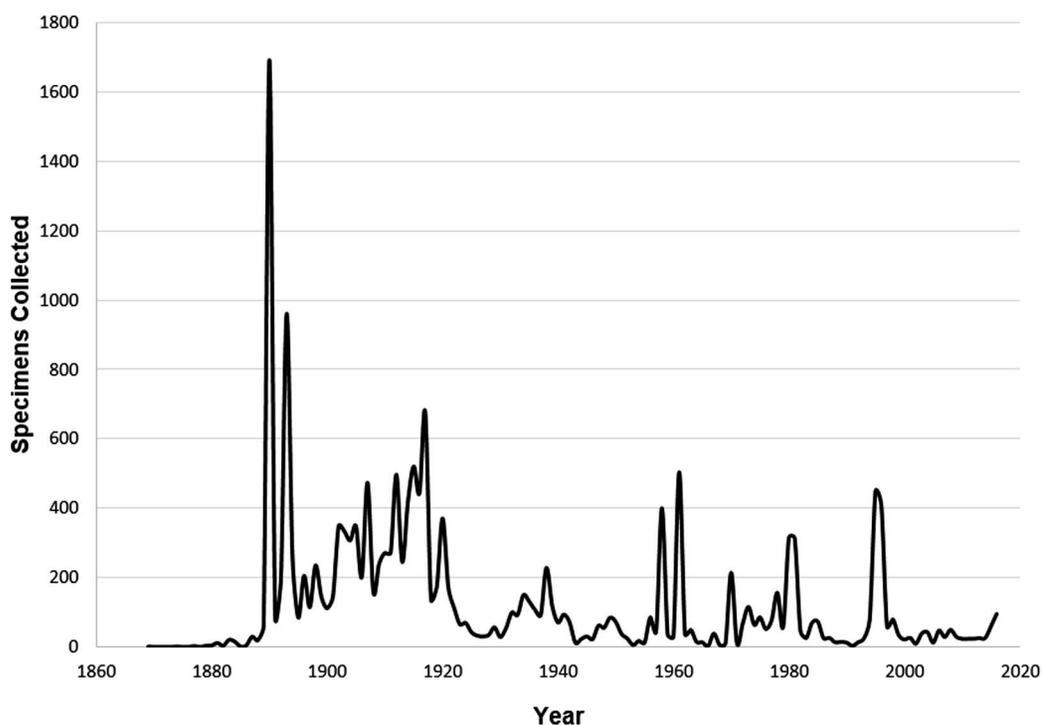


Figure 4.—Number of fungal specimens collected per year from Indiana and deposited within various collections in the United States and Canada (an early specimen collected in 1804 is not included here in order to simplify the plot).

Morchella, see Molina et al. 1993). Estimates of the global diversity for fungi posit that there may be six fungal species for every vascular plant species (Hawksworth 1991; Blackwell 2011). If we apply this ratio to Indiana, then a comparison of the total number of fungal species (including macro-, microfungi, and lichens) recovered in our overall study (ca. 3000) against the number of vascular plant species known for state (ca. 2800; pers. comm. Kay Yatskiyevych) suggests that many more fungal species are in need of documentation. Many species of fungi absent from our Indiana checklists likely represent taxa already known to science. Indeed, the author's record for *Amanita muscaria* var. *guessowii*, never before reported from the state, was discovered without much effort on the campus of Purdue University Northwest (PNW) while preparing this manuscript (a voucher specimen was sent to Purdue's Arthur Fungarium). Noting the magnitude of this gap (perhaps thousands of 'missing' species of Indiana fungi), it is equally likely that a number of fungal species new to science await discovery in the state. These facts highlight the

great need for continued mycological research within the state, as suggested previously by Ruch and colleagues (2001).

We acknowledge that this checklist may contain inaccuracy given the scope of taxa included (and the restricted focus of the lead author) as well as the need to revise some of the species recorded here in light of new data (e.g., see O'Donnell et al. 2011 for the morels). Overall, however, we hope this publication represents solid progress toward a more complete understanding of fungal diversity in the state of Indiana.

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The authors thank Drs. Peter Avis and Patrick Leacock, as well as Stephen Russell, for making data from their mycological projects easily accessible for use in this publication, and we very much appreciate Mr. Russell's taxonomic review of the species included here. The lead author would also like to personally thank Drs. Barbara Thiers (MaCC) and Andrew N. Miller (MiCC) for spearheading the preparation of ADBC proposals (now

funded) and for serving as PIs overseeing the digitization efforts of fungal specimens, which have made literally millions of fungal specimen records available online.

CHECKLIST OF INDIANA MACROFUNGI

Annotation key.—Annotations [in brackets] follow each taxon, indicating records from Indiana that are published in the literature for the first time here (NR) and/or citing the source for each record, either from the literature (†) or from specimens (‡) deposited in collections (single or multiple records representing each taxon reported here can be searched individually at <http://mycoportal.org>).

ASCOMYCOTA

Bolinales

Camarops petersii (Berk. & M.A. Curtis) Nannf. [†, ‡]

Geoglossales

Geoglossum difforme Fr. [‡, NR]
Geoglossum nigratum Cooke [‡, NR]
Trichoglossum farlowii (Cooke) E.J. Durand [‡, NR]
Trichoglossum hirsutum (Pers.) Boud. [†, ‡]
Trichoglossum variabile (E.J. Durand) Nannf. [‡, NR]
Trichoglossum velutipes (Peck) E.J. Durand [‡, NR]
Trichoglossum walteri (Berk.) E.J. Durand [‡, NR]

Helotiales

Bulgaria inquinans (Pers.) Fr. [†]
Leotia lubrica (Scop.) Pers. [†, ‡]
Microglossum olivaceum (Pers.) Gillet [‡, NR]
Microglossum rufum (Schwein.) Underw. [†, ‡]

Pezizales

Aleuria aurantia (Pers.) Fuckel [†, ‡]
Disciotis venosa (Pers.) Boud. [†]
Galiella rufa (Schwein.) Nannf. & Korf [†, ‡]
Gyromitra brunnea Underw. [†, ‡]
Gyromitra caroliniana (Bosc.) Fr. [‡, NR]
Gyromitra esculenta (Pers.) Fr. [†, ‡]
Gyromitra gigas (Krombh.) Cooke [†]
Gyromitra infula (Schaeff.) Quél. [‡, NR]
Helvella acetabulum (L.) Quél. [†, ‡]
Helvella albella Quél. [‡, NR]

Helvella atra Oeder [‡, NR]
Helvella elastica Bull. [†, ‡]
Helvella lactea Boud. [‡, NR]
Helvella lacunosa Afzel. [†, ‡]
Helvella macropus (Pers.) P. Karst. [‡, NR]
Helvella macropus var. *brevis* Peck [‡, NR]
Helvella queletii Bres. [‡, NR]
Helvella subglabra N.S. Weber [‡, NR]
Helvella sulcata Afzel. [‡, NR]
Helvella villosa Schaeff. [‡, NR]
Jafnea semitosta (Berk. & M.A. Curtis) Korf [‡, NR]
Microstoma floccosum (Schwein.) Raitv. [†, ‡]
Mitrophora semilibera (DC.) Lév. [†, ‡]
Morchella angusticeps Peck [†, ‡]
Morchella conica Pers. [†, ‡]
Morchella crassipes (Vent.) Pers. [†]
Morchella deliciosa Fr. [‡, NR]
Morchella esculenta (L.) Pers. [†, ‡]
Morchella hybrida (Sowerby) Pers. [‡, NR]
Otidea onotica (Pers.) Fuckel [‡, NR]
Pachyella clypeata (Schwein.) Le Gal [‡, NR]
Peziza aurantiaca Bull. [‡, NR]
Peziza badia Pers. [†, ‡]
Peziza badiocconfusa Korf [‡, NR]
Peziza bronca Peck [‡, NR]
Peziza cerea Sowerby [‡, NR]
Peziza citrina (Hedw.) Pers. [†, ‡]
Peziza dehni Rabenh. [‡, NR]
Peziza domiciliana Cooke [†, ‡]
Peziza odorata Peck [‡, NR]
Peziza repanda Pers. [†, ‡]
Peziza succosa Berk. [†, ‡]
Peziza sulcata Pers. [†]
Peziza sylvestris (Boud.) Sacc. & Traverso [‡, NR]
Peziza vesiculosa Bull. [†, ‡]
Plectania occidentalis (Schwein.) Seaver [†, ‡]
Sarcoscypha austriaca (Beck ex Sacc.) Boud. [‡, NR]
Sarcoscypha coccinea (Scop.) Sacc. [†, ‡]
Sarcoscypha occidentalis (Schwein.) Sacc. [†, ‡]
Tuber dryophilum Tul. & C. Tul. [‡, NR]
Tuber rufum Pollini [†]
Urnula craterium (Schwein.) Fr. [†, ‡]
Verpa bohemica var. *bispora* (Sorokin) Syd. [†]
Verpa conica (O.F. Müll.) Sw. [†]

Xylariales

Daldinia childiae J.D. Rogers & Y.M. Ju [‡, NR]
Daldinia concentrica (Bolton) Ces. & De Not. [‡, ‡]
Daldinia simulans Child [‡, NR]
Daldinia vernicosa Ces. & De Not. [†, ‡]
Xylaria castorea Berk. [†, ‡]

Xylaria comosa (Mont.) Mont. [†]
Xylaria corniformis (Fr.) Fr. [‡, NR]
Xylaria cornu-damae (Schwein.) Berk. [†, ‡]
Xylaria cubensis (Mont.) Fr. [‡, NR]
Xylaria digitata (L.) Grev. [‡, NR]
Xylaria hypoxylon (L.) Grev. [†, ‡]
Xylaria longiana Rehm [‡, NR]
Xylaria longipes Nitschke [‡, NR]
Xylaria mali Fromme [‡, NR]
Xylaria polymorpha (Pers.) Grev. [†, ‡]
Xylaria subterranea (Schwein.) Sacc. [†]
Xylaria tentaculata Ravenel ex Berk. [‡, NR]
Xylocoremium flabelliforme (Schwein.) J.D. Rogers [‡, NR]

BASIDIOMYCOTA

Agaricales

Agaricus abruptibulbus Peck [†, ‡]
Agaricus arvensis Schaeff. [†, ‡]
Agaricus bitorquis (Quél.) Sacc. [†, ‡]
Agaricus campestris L. [†, ‡]
Agaricus comtulus Fr. [‡, NR]
Agaricus diminutivus Peck [‡, NR]
Agaricus dorsalis Peck [†]
Agaricus haerens Peck [†, ‡]
Agaricus magniceps Peck [‡, NR]
Agaricus moelleri Wasser [‡, NR]
Agaricus placomyces Peck [†, ‡]
Agaricus scitulus Massee [†]
Agaricus silvaticus Schaeffer [†, ‡]
Agaricus sylvicola (Vittad.) Peck [†, ‡]
Agaricus xanthoderma Genev. [‡, NR]
Agrocybe acericola (Peck) Singer [‡, NR]
Agrocybe arvalis (Fr.) Heim & Romagn. [‡, NR]
Agrocybe dura (Bolton) Singer [†, ‡]
Agrocybe erebia (Fr.) Singer [‡, NR]
Agrocybe firma (Peck) Singer [‡, NR]
Agrocybe gibberosa (Fr.) Fayod [‡, NR]
Agrocybe howeana (Peck) Singer [‡, NR]
Agrocybe pediades (Fr.) Fayod [†, ‡]
Agrocybe praecox (Pers.) Fayod [†, ‡]
Amanita abrupta Peck [†, ‡]
Amanita aestivalis Singer ex Singer [‡, NR]
Amanita bisporigera G.F. Atk. [†, ‡]
Amanita brunnescens var. *brunnescens* G.F. Atk. [†, ‡]
Amanita brunnescens var. *pallida* L. Krieg. [‡, NR]
Amanita caesarea (Scop.) Pers. [†]
Amanita ceciliae (Berk. & Broome) Bas [‡, NR]
Amanita chlorinosma (Peck) Lloyd [†, ‡]
Amanita citrina (Schaeff.) Pers. [‡, NR]
Amanita citrina f. *lavendula* (Coker) Veselý [‡, NR]

Amanita cokeri (E.-J. Gilbert & Kühner) E.-J. Gilbert [‡, NR]
Amanita cothurnata G.F. Atk. [†]
Amanita daucipes (Berk. & Mont.) Lloyd [‡, NR]
Amanita excelsa (Fr.) Bertill. [†, ‡]
Amanita farinosa Schwein. [†, ‡]
Amanita flavoconia G.F. Atk. [†, ‡]
Amanita flavorubescens G.F. Atk. [†, ‡]
Amanita floccocephala G.F. Atk. [†]
Amanita fulva Pers. [‡, NR]
Amanita gemmata (Fr.) Bertill. [‡, NR]
Amanita junquillea Quél. [†]
Amanita magnivelaris Peck (Fr.) P. Kumm. [‡, NR]
Amanita multisquamosa Peck [‡, NR]
Amanita muscaria (L.) Lam. [†, ‡]
Amanita muscaria var. *guessowii* Veselý [‡, NR]
Amanita pantherina (DC.) Krombh. [‡, NR]
Amanita phalloides (Fr.) Link [†, ‡]
Amanita rubescens Pers. [†, ‡]
Amanita rubescens f. *alba* (Coker) Sartory & L. Maire [‡, NR]
Amanita solitaria (Bull.) Mérat [†, ‡]
Amanita spreta (Peck) Sacc. [†, ‡]
Amanita strobiliformis (Paulet ex Vittad.) Bertill. [†, ‡]
Amanita subphalloides (Murrill) Murrill [‡, NR]
Amanita thiersii Bas [‡, NR]
Amanita vaginata (Bull.) Lam [†, ‡]
Amanita verna (Bull.) Lam. [†]
Amanita virosa Bertill. [†, ‡]
Arachnion album Schwein. [‡, NR]
Armillaria caligata (Viv.) Gillet [‡, NR]
Armillaria calvescens Bérubé & Dessur. [‡, NR]
Armillaria gallica Marxm. & Romagn. [‡, NR]
Armillaria mellea (Vahl) P. Kumm. [†, ‡]
Armillaria nardosmia Ellis [†]
Armillaria tabescens (Scop.) Emel [†, ‡]
Arrhenia epichysium (Pers.) Redhead, Lutzoni, Moncalvo & Vilgalys [‡, NR]
Arrhenia peltigerina (Peck) Redhead [†]
Asterophora lycoperdoides (Bull.) Ditmar [‡, NR]
Baeospora myriadophylla (Peck) Singer [†]
Bolbitius coprophilus (Peck) Hongo [‡, NR]
Bolbitius tener Berk. [†]
Bolbitius titubans (Bull.) Fr. [†, ‡]
Bovista aestivalis (Bonord.) Demoulin [‡, NR]
Bovista dermoxantha (Vittad.) De Toni [‡, NR]
Bovista longispora Kreisel [‡, NR]
Bovista minor Morgan [‡, NR]
Bovista pila Berk. & M.A. Curtis [‡, NR]
Bovista plumbea Pers. [‡, NR]
Bovista pusilla (Batsch) Pers. [‡, NR]
Bovistella ohiensis (Ellis & Morgan) Morgan [†, ‡]

- Bovistella radicata* (Durieu & Mont.) Pat. [‡, NR]
Calvatia craniiformis (Schwein.) Fr. ex De Toni [‡, †]
Calvatia cyathiformis (Bosc) Morgan [†, †]
Calvatia excipuliformis (Scop.) Perdeck [‡, NR]
Calvatia gigantea (Batsch) Lloyd [†, †]
Calvatia lepidophora (Ellis & Everh.) Coker & Couch [‡, NR]
Calvatia lilacina (Mont. & Berk.) Henn. [‡, NR]
Calvatia rugosa (Berk. & M.A. Curtis) D.A. Reid [†, †]
Calvatia utriformis (Bull.) Jaap [†]
Calyptella campanula (Nees) W.B. Cooke [‡, NR]
Cheimonophyllum candidissimum (Berk. & M.A. Curtis) Singer [‡, NR]
Chlorophyllum agaricoides (Czern.) Vellinga [‡, NR]
Chlorophyllum molybdites (G. Mey.) Massee [†, †]
Chondrostereum purpureum (Pers.) Pouzar [‡, NR]
Clavaria appalachiensis Coker [‡, NR]
Clavaria fragilis Holmsk. [†, †]
Clavaria inaequalis O.F. Müll. [†]
Clavaria rosea Dalman [‡, NR]
Clavaria tetragona Schwein. [†]
Clavulinopsis corniculata (Schaeff.) Corner [‡, NR]
Clavulinopsis fusiformis (Sowerby) Corner [†, †]
Clavulinopsis laeticolor (Berk. & M.A. Curtis) R.H. Petersen [‡, NR]
Clitocybe americana H.E. Bigelow [‡, NR]
Clitocybe aeruginosa H.E. Bigelow [‡, NR]
Clitocybe albidula Peck [†]
Clitocybe candida Bres. [†, †]
Clitocybe cartilaginea (Bull. ex Pers.) Bres [†]
Clitocybe catinus (Fr.) Quél. [†]
Clitocybe clavipes (Pers.) P. Kumm. [‡, NR]
Clitocybe connata (Schumach.) Gillet [‡, NR]
Clitocybe dealbata (Sowerby) P. Kumm. [†, †]
Clitocybe diatreta (Fr.) P. Kumm. [‡, NR]
Clitocybe dilatata P. Karst. [‡, NR]
Clitocybe ditopa (Fr.) Gillet [†]
Clitocybe eccentrica X.H. Wang & P.G. Liu [†]
Clitocybe elephantina Murrill [‡, NR]
Clitocybe fragrans (With.) P. Kumm. [‡, NR]
Clitocybe gibba (Pers.) P. Kumm. [†, †]
Clitocybe intermedia Kauffman [‡, NR]
Clitocybe laccata var. *pallidifolia* Peck [†, †]
Clitocybe martiorum J. Favre [‡, NR]
Clitocybe multiceps Peck [†]
Clitocybe nebularis (Batsch) P. Kumm [†, †]
Clitocybe ochropurpurea (Berk.) Sacc. [†]
Clitocybe odora (Bull.) P. Kumm. [†, †]
Clitocybe patuloides (Peck) Peck [†]
Clitocybe phaeophthalma (Pers.) Kuyper [†, †]
Clitocybe phyllophila (Pers.) P. Kumm. [†]
Clitocybe piceina Peck [†]
Clitocybe regularis Peck [‡, NR]
Clitocybe sinopica (Fr.) P. Kumm. [†]
Clitocybe subconnexa Murrill [‡, NR]
Clitocybe trullisata Ellis [‡, NR]
Clitocybula abundans (Peck) Singer [†, †]
Clitocybula lacerata (Scop.) Singer ex Métrod [‡, NR]
Clitopilus caespitosus Peck [†]
Clitopilus hrbanovii (Velen.) Singer [‡, NR]
Clitopilus noveboracensis (Peck) Sacc. [†]
Clitopilus prunulus (Scop.) P. Kumm. [†, †]
Collybia amabilipes (Peck) Peck [†]
Collybia cookei (Bres.) J.D. Arnold [‡, NR]
Conocybe crispa (Longyear) Singer [†]
Conocybe cryptocystis (G.F. Atk.) Singer [‡, NR]
Conocybe tenera (Schaeff.) Fayod [†, †]
Coprinellus deliquescens (Bull.) P. Karst. [†]
Coprinellus disseminatus (Pers.) J.E. Lange [†, †]
Coprinellus domesticus (Bolton) Vilgalys [†]
Coprinellus ephemerus (Bull.) Redhead [†]
Coprinellus micaceus (Bull.) Vilgalys, Hopple & Jacq. Johnson [†, †]
Coprinopsis atramentaria (Bull.) Redhead, Vilgalys & Moncalvo [†, †]
Coprinopsis lagopus (Fr.) Redhead, Vilgalys & Moncalvo [†, †]
Coprinopsis nivea (Pers.) Redhead, Vilgalys & Moncalvo [‡, NR]
Coprinopsis picacea (Bull.) Redhead [†]
Coprinopsis radiata (Bolton) Redhead, Vilgalys & Moncalvo [†, †]
Coprinopsis variegata (Peck) Redhead, Vilgalys & Moncalvo [‡, NR]
Coprinus comatus (O.F. Müll.) Pers. [†, †]
Coprinus comatus var. *ovatus* (Scop.) Quél. [†]
Coprinus ebulbosus Peck [†]
Coprinus quadrifidus Peck [†]
Coprinus rotundisporus Peck [†]
Cortinarius alboviolaceus (Pers.) Fr. [†, †]
Cortinarius armillatus (Fr.) Fr. [†, †]
Cortinarius autumnalis Peck [†, †]
Cortinarius caperatus (Pers.) Fr. [†]
Cortinarius cinnabarinus Fr. [†, †]
Cortinarius cinnamomeus (L.) Gray [†]
Cortinarius claricolor (Fr.) Fr. [‡, NR]
Cortinarius collariatus E. Horak & M.M. Moser [†]
Cortinarius collinitus (Sowerby) Gray [†]
Cortinarius corrugatus Peck [‡, NR]
Cortinarius cylindripes Kauffman [†]
Cortinarius distans Peck [†, †]

- Cortinarius duracinus* Fr. [†, ‡]
Cortinarius gentilis (Fr.) Fr. [‡, NR]
Cortinarius hinnuleus Fr. [†]
Cortinarius iodeoides Kauffman [‡, NR]
Cortinarius iodes Berk. & M.A. Curtis [‡, NR]
Cortinarius michiganensis Kauffman [†]
Cortinarius obliquus Peck [†]
Cortinarius ochraceus Peck [†]
Cortinarius olearioides Rob. [‡, NR]
Cortinarius semisanguineus (Fr.) Gillet [‡, NR]
Cortinarius sericeps Peck [‡, NR]
Cortinarius torvus (Fr.) Fr. [‡, NR]
Cortinarius violaceus (L.) Gray [‡, NR]
Crepidotus applanatus (Pers.) P. Kumm. [†, ‡]
Crepidotus applanatus var. *globiger* (Berk.) Pilát [‡, NR]
Crepidotus calolepis (Fr.) P. Karst. [†]
Crepidotus cocophyllus (Berk.) Sacc. [‡, NR]
Crepidotus ellipsoideus Hesler & A.H. Sm. [‡, NR]
Crepidotus fulvotomentosus (Peck) Peck [†, ‡]
Crepidotus fusisporus Hesler & A.H. Sm. [†]
Crepidotus herbarum (Peck) Peck [‡, NR]
Crepidotus hygrophanus Murrill [‡, NR]
Crepidotus hundellii Pilát [†]
Crepidotus mollis (Schaeff.) Staude [†, ‡]
Crepidotus nephrodes (Berk. & M.A. Curtis) Sacc. [‡, NR]
Crepidotus versutus (Peck) Peck [†, ‡]
Crepidotus vulgaris Hesler & A.H. Sm. [‡, NR]
Crinipellis setipes (Peck) Singer [‡, NR]
Crinipellis zonata (Peck) Sacc. [†, ‡]
Crucibulum crucibuliforme (Scop.) V.S. White [†, ‡]
Cyathus olla (Batsch) Pers. [†, ‡]
Cyathus poeppigii Tul. & C. Tul. [‡, NR]
Cyathus stercoreus (Schwein.) De Toni [†, ‡]
Cyathus striatus (Huds.) Willd. [†, ‡]
Cylindrobasidium evolvens (Fr.) Jülich [‡, NR]
Cylindrobasidium torrendii (Bres.) Hjortstam [‡, NR]
Cyphella fasciculata Berk. & M.A. Curtis [‡, NR]
Cyphella mellea Burt [‡, NR]
Deconica subviscida Peck [‡, NR]
Disciseda candida (Schwein.) Lloyd [‡, NR]
Entoloma abortivum (Berk. & M.A. Curtis) Donk [†, ‡]
Entoloma alboumbonatum Hesler [‡, NR]
Entoloma asprellum (Fr.) Fayod [†]
Entoloma byssisedum (Pers.) Donk [‡, NR]
Entoloma carneogriseum (Berk. & Broome) Noordel. [†]
Entoloma clypeatum (L.) P. Kumm. [†]
Entoloma gracilipes (Peck) Malloch [‡, NR]
Entoloma grande Peck [†]
Entoloma jubatum (Fr.) P. Karst. [†]
Entoloma lividum (Bull.) Quél. [†]
Entoloma murrayi (Berk. & M.A. Curtis) Sacc. [‡, NR]
Entoloma rhodopolium (Fr.) P. Kumm. [†, ‡]
Entoloma salmoneum (Peck) Sacc. [‡, NR]
Entoloma sericellum (Fr.) P. Kumm. [‡, NR]
Entoloma serrulatum (Fr.) Hesler [‡, NR]
Entoloma sinuatum (Bull. ex Pers.) P. Kumm. [†]
Entoloma strictipes (Peck) Hesler [‡, NR]
Entoloma strictius (Peck) Sacc. [†, ‡]
Entoloma subcostatum G.F. Atk. [†]
Entoloma undatum (Gillet) M.M. Moser [†]
Entoloma vernum S. Lundell [‡, NR]
Fistulina hepatica (Schaeff.) With. [†]
Flammula betulina Peck [†]
Flammula praecox Peck [‡, NR]
Flammulaster erinaceellus (Peck) Watling [‡, NR]
Flammulaster gracilis (Quél.) Watling [‡, NR]
Flammulina velutipes (Curtis) Singer [†, ‡]
Galera ovalis (Fr.) Gillet [†]
Galerina autumnalis (Peck) A.H. Sm. & Singer [‡, NR]
Galerina marginata (Batsch) Kühner [‡, NR]
Galerina unicolor (Vahl) Singer [†, ‡]
Gerronema strombodes (Berk. & Mont.) Singer [‡, NR]
Gymnopilus aeruginosus (Peck) Singer [†]
Gymnopilus bellulus (Peck) Murrill [‡, NR]
Gymnopilus junonius (Fr.) P.D. Orton [†, ‡]
Gymnopilus liquiritiae (Pers.) P. Karst. [‡, NR]
Gymnopilus sapineus (Fr.) Maire [‡, NR]
Gymnopilus acervatus (Fr.) Murrill [†, ‡]
Gymnopus alkalivirens (Singer) Halling [‡, NR]
Gymnopus androsaceus (L.) J.L. Mata & R.H. Petersen [‡, NR]
Gymnopus aquosus (Bull.) Antonín & Noordel. [‡, NR]
Gymnopus atratoides (Peck) Murrill [†]
Gymnopus bififormis (Peck) Halling [‡, NR]
Gymnopus confluens (Pers.) Antonín, Halling & Noordel. [†, ‡]
Gymnopus dichrous (Berk. & M.A. Curtis) Halling [‡, NR]
Gymnopus dryophilus (Bull.) Murrill [†, ‡]
Gymnopus erythropus (Pers.) Antonín [†, ‡]
Gymnopus foetidus (Sowerby) J.L. Mata & R.H. Petersen [†, ‡]
Gymnopus fusipes (Bull.) Gray [†]
Gymnopus hariolorum (Bull.) Antonín [†]
Gymnopus hygrophoroides (Peck) Murrill [†, ‡]
Gymnopus iocephalus (Berk. & M.A. Curtis) Halling [‡, NR]
Gymnopus luxurians (Peck) Murrill [‡, NR]

- Gymnopus peronatus* (Bolton) Gray [†, ‡]
Gymnopus polyphyllus (Peck) Halling [‡, NR]
Gymnopus spongiosus (Berk. & M.A. Curtis) Halling [‡, NR]
Gymnopus subnudus (Ellis ex Peck) Halling [‡, NR]
Gymnopus subsulphureus (Peck) Murrill [‡, NR]
Hebeloma albidulum Peck [†]
Hebeloma album Peck [†]
Hebeloma crustuliniforme (Bull.) Quél. [‡, NR]
Hebeloma mesophaeum (Pers.) Quél. [†]
Hebeloma sinapizans (Paulet) Gillet [‡, NR]
Hemingsomyces candidus (Pers.) Kuntze [‡, NR]
Hohenbuehelia angustata (Berk.) Singer [‡, NR]
Hohenbuehelia approximans (Peck) Singer [‡, NR]
Hohenbuehelia horakii Courtec. [‡, NR]
Hohenbuehelia petaloides (Bull.) Schulzer [†, ‡]
Hydropus floccipes (Fr.) Singer [‡, NR]
Hygrocybe acutoconica (Clem.) Singer [‡, NR]
Hygrocybe acutoconica var. *cuspidata* (Peck) Arnolds [‡, NR]
Hygrocybe borealis (Peck) Bon [†]
Hygrocybe calyptriformis (Berk. & Broome) Fayod [‡, NR]
Hygrocybe cantharellus (Schwein.) Murrill [‡, NR]
Hygrocybe ceracea (Wulfen) P. Kumm. [†, ‡]
Hygrocybe chlorophana (Fr.) Wünsche [‡, NR]
Hygrocybe coccinea (Schaeff.) P. Kumm. [†, ‡]
Hygrocybe colemanniana (A. Bloxam) P.D. Orton & Watling [‡, NR]
Hygrocybe conica (Schaeff.) P. Kumm. [†, ‡]
Hygrocybe irrigata (Pers.) Bon [‡, NR]
Hygrocybe laeta (Pers.) P. Kumm. [‡, NR]
Hygrocybe lepida Arnolds [‡, NR]
Hygrocybe marginata var. *concolor* (A.H. Smith) A.E. Bessette, A.R. Bessette, W.C. Roody & W.E. Sturgeon [‡, NR]
Hygrocybe marginata var. *marginata* (Peck) Murrill [‡, NR]
Hygrocybe marginata var. *olivacea* (A.H. Smith & Hesler) A.E. Bessette, A.R. Bessette, W.C. Roody & W.E. Sturgeon [‡, NR]
Hygrocybe miniata (Fr.) P. Kumm. [†, ‡]
Hygrocybe nitida (Berk. & M.A. Curtis) Murrill [‡, NR]
Hygrocybe obrussea (Fr.) Wünsche [‡, NR]
Hygrocybe pratensis (Schaeff.) Murrill [†, ‡]
Hygrocybe psittacina (Schaeff.) P. Kumm. [†, ‡]
Hygrocybe punicea (Fr.) P. Kumm. [†, ‡]
Hygrocybe virginea (Wulfen) P.D. Orton & Watling [‡, NR]
Hygrophorus eburneus (Bull.) Fr. [†]
Hygrophorus flavodiscus Frost [†]
Hygrophorus laurae Morg. [†]
Hygrophorus occidentalis A.H. Sm. & Hesler [‡, NR]
Hygrophorus pudorinus (Fr.) Fr. [†, ‡]
Hygrophorus roseibrunneus Murrill [†]
Hygrophorus russula (Schaeff.) Kauffman [†]
Hygrophorus sordidus Peck [†, ‡]
Hygrophorus tennesseensis A.H. Sm. & Hesler [‡, NR]
Hypholoma candolleianum (Fr.) Maire [‡, NR]
Hypholoma capnoides (Fr.) P. Kumm. [‡, NR]
Hypholoma cutifractum Peck [†]
Hypholoma echiniceps G.F. Atk. [‡, NR]
Hypholoma hydrophilum (Bull.) Quél. [‡, NR]
Hypholoma incertum (Peck) A.H. Sm. [‡, NR]
Hypholoma lateritium (Schaeff.) P. Kumm. [†, ‡]
Hypsizygus ulmarius (Bull.) Redhead [†, ‡]
Inocybe albodisca Peck [†, ‡]
Inocybe calospora Quél. [†, ‡]
Inocybe cookei Bres. [‡, NR]
Inocybe decipientoides Peck [‡, NR]
Inocybe fibrillosa Peck [†]
Inocybe geophylla (Sowerby) P. Kumm. [†, ‡]
Inocybe griseovelata Kühner [‡, NR]
Inocybe lacera (Fr.) P. Kumm. [‡, NR]
Inocybe lilacina (Peck) Kauffman [†]
Inocybe maculata Boud. [‡, NR]
Inocybe pallidipes Ellis & Everh. [‡, NR]
Inocybe praetervisa Quél. [‡, NR]
Inocybe pyriodora (Pers.) P. Kumm. [‡, NR]
Inocybe radiata Peck [‡, NR]
Inocybe rimosa (Bull.) P. Kumm. [†, ‡]
Inocybe sororia Kauffman [‡, NR]
Inocybe unicolor Peck [‡, NR]
Kuehneromyces mutabilis (Schaeff.) Singer & A.H. Sm. [†]
Laccaria amethystina Cooke [‡, NR]
Laccaria bicolor (Maire) P.D. Orton [‡, NR]
Laccaria laccata (Scop.) Cooke [†, ‡]
Laccaria ochropurpurea (Berk.) Peck [†, ‡]
Laccaria proxima (Boud.) Pat. [‡, NR]
Laccaria tortilis (Bolton) Cooke [‡, NR]
Laccaria trullisata (Ellis) Peck [‡, NR]
Lachnella villosa (Pers.) Gillet [‡, NR]
Lacrymaria lacrymabunda (Bull.) Pat. [†, ‡]
Lepiota alluviina (Peck) Morgan [†]
Lepiota aspera (Pers.) Quél. [†, ‡]
Lepiota augustana (Britzelm.) Sacc. [†]
Lepiota castanea Quél. [‡, NR]
Lepiota conspurcata (Willd.) Morgan [†]
Lepiota clypeolaria (Bull.) P. Kumm. [‡, NR]
Lepiota cristata (Bolton) P. Kumm. [†, ‡]
Lepiota farinosa Peck [†]
Lepiota fischeri Kauffman [‡, NR]

- Lepiota granosa* Morgan [†]
Lepiota miamensis (Morgan) Sacc. [†]
Lepiota naucinoides (Peck) Peck [†]
Lepiota subincarnata J.E. Lange [‡, NR]
Lepiota virescens (Speg.) Morgan [†]
Lepista flaccida (Sowerby) Pat. [†, ‡]
Lepista irina (Fr.) H.E. Bigelow [‡, NR]
Lepista nuda (Bull.) Cooke [‡, NR]
Lepista personata (Fr.) Cooke [†, ‡]
Leptonia rosea var. *marginata* Largent [‡, NR]
Leucoagaricus americanus (Peck) Vellinga [†, ‡]
Leucoagaricus caeruleascens (Peck) J.F. Liang, Zhu L. Yang & J. Xu [†]
Leucoagaricus leucothites (Vittad.) Wasser [†, ‡]
Leucoagaricus rubrotinctus (Peck) Singer [†, ‡]
Leucocoprinus cepistipes (Sowerby) Pat. [†, ‡]
Leucocoprinus flavescens (Morgan) H.V. Sm. [‡, NR]
Leucopaxillus albissimus (Peck) Singer [†, ‡]
Leucopaxillus cerealis (Lasch) Singer [‡, NR]
Leucopaxillus giganteus (Sowerby) Singer [†]
Leucopaxillus laterarius (Peck) Singer & A.H. Sm. [‡, NR]
Leucopaxillus paradoxus (Costantin & L.M. Dufour) Boursier [‡, NR]
Lindmeria trachyspora (Bourdot & Galzin) Pilát [‡, NR]
Lycoperdon americanum Demoulin [‡, NR]
Lycoperdon echinatum Pers. [‡, NR]
Lycoperdon ericaeum Bonord. [‡, NR]
Lycoperdon fuscum Huds. [‡, NR]
Lycoperdon marginatum Vittad. [‡, NR]
Lycoperdon molle Pers. [‡, NR]
Lycoperdon nigrescens Wahlenb. [‡, NR]
Lycoperdon peckii J.B. Morgan [‡, NR]
Lycoperdon pedicellatum Gardezi [‡, NR]
Lycoperdon perlatum Pers. [†, ‡]
Lycoperdon pulcherrimum Berk. & M.A. Curtis [‡, NR]
Lycoperdon pyriforme Schaeff. [†, ‡]
Lycoperdon radicum Reichard [‡, NR]
Lycoperdon rimulatum Peck [‡, NR]
Lycoperdon stellare (Peck) Lloyd [‡, NR]
Lycoperdon umbrinum Pers. [†, ‡]
Lycoperdon wrightii Berk. & M.A. Curtis [‡, NR]
Lyophyllum decastes (Fr.) Singer [‡, NR]
Lyophyllum plexipes (Fr.) Kühner & Romagn. [†]
Macrocyttidia cucumis (Pers.) Joss. [‡, NR]
Macrolepiota procera (Scop.) Singer [†, ‡]
Marasmiellus candidus (Fr.) Singer [†]
Marasmiellus papillatus (Peck) Redhead & Halling [‡, NR]
Marasmiellus vaillantii (Pers.) Singer [‡, NR]
Marasmius armeniacus Gilliam [‡, NR]
Marasmius capillaris Morgan [†, ‡]
Marasmius cohaerens (Pers.) Cooke & Quél. [†, ‡]
Marasmius copelandii var. *olidus* (Gilliam) Desjardin [‡, NR]
Marasmius delectans Morgan [†, ‡]
Marasmius falcatipes Desjardin [‡, NR]
Marasmius felix Morgan [‡, NR]
Marasmius fulvoferrugineus Gilliam [‡, NR]
Marasmius glabellus Peck [†]
Marasmius graminum (Lib.) Berk. [†, ‡]
Marasmius haematocephalus (Mont.) Fr. [†]
Marasmius oreades (Bolton) Fr. [†, ‡]
Marasmius pulcherripes Peck [‡, NR]
Marasmius rotula (Scop.) Fr. [†, ‡]
Marasmius setosus (Sowerby) Noordel. [‡, NR]
Marasmius siccus Schwein. [†, ‡]
Marasmius spissus Gilliam [‡, NR]
Marasmius strictipes (Peck) Singer [‡, NR]
Marasmius sullivantii Mont. [‡, NR]
Marasmius velutipes Berk. & M.A. Curtis [†]
Marasmius vialis Peck [‡, NR]
Megacollybia platyphylla (Pers.) Kotl. & Pouzar [†, ‡]
Megacollybia rodmanii R.H. Petersen, K.W. Hughes & Lickey [‡, NR]
Megacollybia rodmanii f. *murina* R.H. Petersen & K.W. Hughes [‡, NR]
Melanoleuca alboflavida (Peck) Murrill [‡, NR]
Melanoleuca melaleuca (Pers.) Murrill [†, ‡]
Merismodes anomala (Pers.) Singer [‡, NR]
Morganella subincarnata (Peck) Kreisel & Dring [‡, NR]
Mycena acicula (Schaeff.) P. Kumm. [‡, NR]
Mycena albiceps (Peck) Gilliam [‡, NR]
Mycena alcalina (Fr.) P. Kumm. [†]
Mycena clavularis (Batsch) Sacc. [‡, NR]
Mycena corticola (Pers.) Gray [‡, NR]
Mycena epipterygia (Scop.) Gray [†, ‡]
Mycena epipterygia var. *griseoviridis* (A.H. Sm.) Maas Geest. [‡, NR]
Mycena filopes (Bull.) P. Kumm. [‡, NR]
Mycena galericulata (Scop.) Gray [†, ‡]
Mycena haematopus (Pers.) P. Kumm. [†, ‡]
Mycena hemisphaerica Peck [‡, NR]
Mycena inclinata (Fr.) Quél. [‡, NR]
Mycena leaiana (Berk.) Sacc. [†, ‡]
Mycena leptophylla (Peck) Sacc. [†]
Mycena luteopallens Peck [‡, NR]
Mycena murina (Murrill) Murrill [‡, NR]
Mycena niveipes (Murrill) Murrill [†]
Mycena parabolica (Fr.) Quél. [†]
Mycena pectinata (Murrill) Murrill [‡, NR]
Mycena polygramma (Bull.) Gray [†]
Mycena pulcherrima (Peck) Sacc. [‡, NR]

- Mycena pura* (Pers.) P. Kumm. [†, ‡]
Mycena rutilantiformis (Murrill) Murrill [‡, NR]
Mycena subcaerulea (Peck) Sacc. [‡, NR]
Mycetinis scorodoni (Fr.) A.W. Wilson & Desjardin [‡, NR]
Nolanea howellii Peck [‡, NR]
Omphalia alboflava Moorgan [†]
Omphalina pyxidata (Bull.) Quél. [‡, NR]
Omphalotus olearius (DC.) Singer [†, ‡]
Ossicaulis lignatilis (Pers.) Redhead & Ginns [†]
Ozonium omnivorum Shear [‡, NR]
Panaeolina foenicicii (Pers.) Maire [†]
Panaeolus campanulatus (Bull.) Quél. [†]
Panaeolus papilionaceus (Bull.) Quél. [†, ‡]
Panaeolus semiovatus (Sowerby) S. Lundell & Nannf. [†]
Panaeolus solidipes (Peck) Sacc. [†]
Panellus pusillus (Pers. ex Lév.) Burds. & O.K. Mill. [†]
Panellus serotinus (Pers.) Kühner [‡, NR]
Panellus stipticus (Bull.) P. Karst. [†, ‡]
Parasola plicatilis (Curtis) Redhead, Vilgalys & Hopple [†, ‡]
Phaeosolenia inconspicua (Sacc.) Donk [‡, NR]
Pholiota adiposa (Batsch) P. Kumm. [†, ‡]
Pholiota aggericola (Peck) Sacc. [†]
Pholiota angustipes (Peck) Sacc. [†]
Pholiota aurivella (Batsch) P. Kumm. [‡, NR]
Pholiota cerasina Peck [†]
Pholiota curvipes (P. Kumm.) Quél. [†]
Pholiota flammans (Batsch) P. Kumm. [†]
Pholiota luteofolius (Peck) Sacc. [†]
Pholiota polychroa (Berk.) A.H. Sm. & H.J. Brodie [‡, NR]
Pholiota populnea (Pers.) Kuyper & Tjall.-Beuk. [†]
Pholiota squarrosoides (Peck) Sacc. [†]
Pholiota subsquarrosa (Fr.) Sacc. [†, ‡]
Pholiota veris A.H. Sm. & Hesler [‡, NR]
Pholiotina rugosa (Peck) Singer [†]
Phyllostopsis nidulans (Pers.) Singer [†, ‡]
Pleurotus atropellitus Peck [†]
Pleurotus cystidiosus O.K. Mill. [‡, NR]
Pleurotus dryinus (Pers.) P. Kumm. [†]
Pleurotus ostreatus (Jacq.) P. Kumm. [†, ‡]
Pleurotus populinus O. Hilber & O.K. Mill. [‡, NR]
Pleurotus pulmonarius (Fr.) Quél. [†, ‡]
Pleurotus sapidus Quél. [†, ‡]
Pleurotus serotinoideus (Peck) Sacc. [†]
Pleurotus serotinus (Pers.) P. Kumm. [†]
Pluteus americanus (P. Banerjee & Sundberg) Justo, E.F. Malysheva & Minnis [‡, NR]
Pluteus cervinus (Schaeff.) P. Kumm. [†, ‡]
Pluteus cervinus var. *albus* Peck [†]
Pluteus chrysophaeus (Schaeff.) Quél. [‡, NR]
Pluteus chrysophlebius (Berk. & M.A. Curtis) Sacc. [‡, NR]
Pluteus cincus Cke. [†]
Pluteus fuliginosus Murrill [‡, NR]
Pluteus fulvobadius Murrill [‡, NR]
Pluteus leoninus (Schaeff.) P. Kumm. [†]
Pluteus longistriatus (Peck) Peck [†, ‡]
Pluteus nanus (Pers.) P. Kumm. [†, ‡]
Pluteus pellitus (Pers.) P. Kumm. [‡, NR]
Pluteus petasatus (Fr.) Gillet [‡, NR]
Pluteus podospileus Sacc. & Cub. [‡, NR]
Pluteus romellii (Britzelm.) Sacc. [‡, NR]
Pluteus salicinus (Pers.) P. Kumm. [†]
Pluteus umbrosus (Pers.) P. Kumm. [†]
Porodisculus pendulus (Schwein. ex Fr.) Murrill [‡, NR]
Porotheleum fimbriatum (Pers.) Fr. [‡, NR]
Psathyrella ammophila (Durieu & Lév.) P.D. Orton [‡, NR]
Psathyrella candolleana (Fr.) Maire [‡, NR]
Psathyrella cernua (Vahl) Hirsch [†]
Psathyrella conopilus (Fr.) A. Pearson & Dennis [†]
Psathyrella echiniceps (G.F. Atk.) A.H. Sm. [‡, NR]
Psathyrella epimyces (Peck) A.H. Sm. [‡, NR]
Psathyrella gracilis (Fr.) Quél. [‡, NR]
Psathyrella hydrophila (Bull.) Maire [‡, NR]
Psathyrella hymenocephala (Peck) A.H. Sm. [‡, NR]
Psathyrella incerta (Peck) A.H. Sm. [‡, NR]
Psathyrella longipes (Peck) A.H. Sm. [†]
Psathyrella piluliformis (Bull.) P.D. Orton [†]
Psathyrella sarcocephala (Fr.) Singer [†, ‡]
Pseudoclitocybe cyathiformis (Bull.) Singer [†]
Pseudofistulina radicata (Schwein.) Burds. [‡, NR]
Pterula plumosa (Schwein.) Fr. [†, ‡]
Radulomyces confluens (Fr.) M.P. Christ. [‡, NR]
Resinomycena rhododendri (Peck) Redhead & Singer [‡, NR]
Resupinatus alboniger (Pat.) Singer [‡, NR]
Resupinatus applicatus (Batsch) Gray [†, ‡]
Rhizomarasmius pyrrhocephalus (Berk.) R.H. Petersen [‡, NR]
Rhodocybe popinalis (Fr.) Singer [†]
Rhodocollybia butyracea (Bull.) Lennox [‡, NR]
Rhodocollybia maculata (Alb. & Schwein.) Singer [†, ‡]
Rhodotus palmatus (Bull.) Maire [†, ‡]
Schizophyllum commune Fr. [†, ‡]
Simocybe centunculus (Fr.) P. Karst. [†, ‡]
Stropharia coronilla (Bull.) Quél. [‡, NR]

- Stropharia hardii* G.F. Atk. [‡, NR]
Stropharia rugosoannulata Farl. ex Murrill [‡, NR]
Stropharia semiglobata (Batsch) Quél. [†, ‡]
Stropharia viridula (Schaeff.) Morgan [†]
Tetrapyrgos nigripes (Fr.) E. Horak [‡, NR]
Tricholoma album (Schaeff.) P. Kumm. [†]
Tricholoma caligatum (Viv.) Ricken [‡, NR]
Tricholoma columbetta (Fr.) P. Kumm. [‡, NR]
Tricholoma equestre (L.) P. Kumm. [†]
Tricholoma fulvum (Fr.) Bigeard & H. Guill. [†]
Tricholoma fumescens (Peck) Sacc. [†, ‡]
Tricholoma leucophyllum Ovrebo & Tylutki [‡, NR]
Tricholoma resplendens (Fr.) P. Karst. [‡, NR]
Tricholoma saponaceum (Fr.) P. Kumm. [‡, NR]
Tricholoma sciodes (Pers.) C. Martín [‡, NR]
Tricholoma sejunctum (Sowerby) Quél. [†, ‡]
Tricholoma subsplendens (Murrill) Sacc. & Trotter [‡, NR]
Tricholoma subsaponaceum Peck [‡, NR]
Tricholoma sulphureum (Bull.) P. Kumm. [†]
Tricholoma terreum (Schaeff.) P. Kumm. [‡, NR]
Tricholoma virgatum (Fr.) P. Kumm. [‡, NR]
Tricholoma zelleri (D.E. Stuntz & A.H. Sm.) Ovrebo & Tylutki [‡, NR]
Tricholomopsis formosa (Murrill) Singer [‡, NR]
Tricholomopsis rutilans (Schaeff.) Singer [‡, NR]
Tubaria furfuracea (Pers.) Gillet [‡, NR]
Tubaria pellucida (Bull. & Vent.) Sacc. [‡, NR]
Tulostoma berkeleyi Lloyd [‡, NR]
Tulostoma brumale Pers. [‡, NR]
Tulostoma fimbriatum Fr. [‡, NR]
Tulostoma fimbriatum var. *campestre* (Morgan) G. Moreno [‡, NR]
Tulostoma longii Lloyd [‡, NR]
Tulostoma melanocyclus Bres. [‡, NR]
Tulostoma pygmaeum Lloyd [‡, NR]
Tulostoma simulans Lloyd [‡, NR]
Tulostoma striatum G. Cunn. [‡, NR]
Typhula subfasciculata Ellis & Everh [‡, NR]
Vascellum cruciatum (Rostk.) P. Ponce de León [†, ‡]
Vascellum curtisii (Berk.) Kreisel [‡, NR]
Vascellum pratense (Pers.) Kreisel [‡, NR]
Volvariella bombycina (Schaeff.) Singer [†, ‡]
Volvariella gloiocephala (DC.) Boekhout & Enderle [†, ‡]
Volvariella hypopithys (Fr.) Shaffer [‡, NR]
Volvariella pusilla (Pers.) Singer [†, ‡]
Xeromphalina campanella (Batsch) Kühner & Maire [†, ‡]
Xeromphalina kauffmanii A.H. Sm. [‡, NR]
Xeromphalina tenuipes (Schwein.) A.H. Sm. [‡, NR]
Xerula furfuracea (Peck) Redhead, Ginns & Shoemaker [‡, NR]
Xerula megalospora (Clem.) Redhead, Ginns & Shoemaker [‡, NR]
Xerula limonispora R.H. Petersen [‡, NR]
Xerula longipes (Quél.) Maire [†]
Xerula radicata (Relhan) Dörfelt [†, ‡]
Xerula sinopudens R.H. Petersen & Nagas. [‡, NR]
- Atheliales**
Athelia arachnoidea (Berk.) Jülich [‡, NR]
Byssocorticium atrovirens (Fr.) Bondartsev & Singer ex Singer [‡, NR]
- Auriculariales**
Auricularia americana Parmasto & I. Parmasto ex Audet [‡, NR]
Auricularia auricula-judae (Bull.) J. Schröt. [†, ‡]
Seismosarca alba Lloyd [‡, NR]
Tremellodendron schweinitzii G.F. Atk. [†]
- Boletales**
Astraeus hygrometricus (Pers.) Morgan [†, ‡]
Aureoboletus auriporus (Peck) Pouzar [†, ‡]
Austroboletus gracilis (Peck) Wolfe [†]
Boletellus chrysenteroides (Snell) Snell [‡, NR]
Boletellus russellii (Frost) E.-J. Gilbert [‡, NR]
Boletinus cavipes (Opat.) Kalchbr. [‡, NR]
Boletus auripes Peck [†]
Boletus badius (Fr.) Fr. [†]
Boletus bicolor var. *bicolor* Raddi [†, ‡]
Boletus bicolor var. *subreticulatus* A.H. Sm. & Thiers [‡, NR]
Boletus calopus Pers. [†]
Boletus campestris A.H. Sm. & Thiers [†]
Boletus conicus Rav. [†]
Boletus curtisii Berk. [‡, NR]
Boletus edulis Bull. [†, ‡]
Boletus frostii J.L. Russell [†, ‡]
Boletus fumosiceps (Murrill) Murrill [†]
Boletus glabellus Peck [‡, NR]
Boletus illudens Peck [‡, NR]
Boletus indecisus Peck [†]
Boletus inedulis (Murrill) Murrill [†]
Boletus innixus Frost [†]
Boletus luridus Schaeff. [†]
Boletus luridiformis Rostk. [†]
Boletus magnisporus Frost [†]
Boletus mariae A.H. Sm. & Thiers [‡, NR]
Boletus miniatoolivaceus Frost [‡, NR]

- Boletus miniatopallescens* A.H. Sm. & Thiers [‡, NR]
Boletus modestus Peck [†, ‡]
Boletus nigrellus Peck [†]
Boletus ornatipes Peck [†, ‡]
Boletus pallidus Frost [†, ‡]
Boletus peckii Frost [‡, NR]
Boletus pseudosensibilis A.H. Sm. & Thiers [‡, NR]
Boletus reticulatus Schaeff. [†]
Boletus retipes Berk & M.A. Curtis [†]
Boletus rubropunctus Peck [‡, NR]
Boletus sensibilis Peck [‡, NR]
Boletus speciosus Frost [†]
Boletus subfraternus Coker & Beers [†, ‡]
Boletus subglabripes Peck [†]
Boletus subsanguineus Peck [†, ‡]
Boletus subvelutipes Peck [†, ‡]
Boletus tenax A.H. Sm. & Thiers [‡, NR]
Boletus variipes Peck [‡, NR]
Boletus vermiculosus Peck [†]
Bothia castanella (Peck) Halling [‡, NR]
Chalciporus piperatoides (A.H. Sm. & Thiers) T.J. Baroni & Both [‡, NR]
Chalciporus piperatus (Bull.) Bataille [‡, NR]
Chroogomphus ochraceus (Kauffman) O.K. Mill. [‡, NR]
Coniophora arida (Fr.) P. Karst. [‡, NR]
Coniophora olivacea (Fr.) P. Karst. [‡, NR]
Coniophora puteana (Schumach.) P. Karst. [†]
Cyanoboletus pulverulentus (Opat.) Gelardi [†]
Gomphidius flavipes Peck [‡, NR]
Gyroporus castaneus (Bull.) Quéf. [†, ‡]
Gyroporus cyanescens (Bull.) Quéf. [†, ‡]
Gyroporus cyanescens var. *violaceotinctus* Watling [‡, NR]
Gyrodon merulioides (Schwein.) Singer [†, ‡]
Hygrophoropsis aurantiaca (Wulfen) Maire [†, ‡]
Leccinum albellum (Peck) Singer [†, ‡]
Leccinum crocipodium (Letell.) Watling [‡, NR]
Leccinum holopus var. *holopus* (Rostk.) Watling [‡, NR]
Leccinum holopus var. *americanum* A.H. Sm. & Thiers [‡, NR]
Leccinum insigne A.H. Sm., Thiers & Watling [‡, NR]
Leccinum oxydabile (Singer) Singer [‡, NR]
Leccinum pseudoscabrum (Kallenb.) Šutara [‡, NR]
Leccinum rugosiceps (Peck) Singer [‡, NR]
Leccinum scabrum (Bull.) Gray [†, ‡]
Leccinum snellii A.H. Sm., Thiers & Watling [‡, NR]
Leccinum subglabripes (Peck) Singer [‡, NR]
Leccinum subleucophaeum E.A. Dick & Snell [‡, NR]
Leucogyrophana pinastri (Fr.) Ginns & Weresub [‡, NR]
Melanogaster broomeanus Berk. [‡, NR]
Paxillus involutus (Batsch) Fr. [†, ‡]
Phylloporus leucomyelinus Singer [‡, NR]
Phylloporus rhodoxanthus (Schwein.) Bres. [†, ‡]
Pisolithus arrhizus (Scop.) Rauschert [‡, NR]
Plicaturopsis crispa (Pers.) D.A. Reid [†, ‡]
Pseudoboletus parasiticus (Bull.) Šutara [‡, NR]
Pseudomerulius aureus (Fr.) Jülich [‡, NR]
Retiboletus griseus (Frost) Manfr. Binder & Bresinsky [‡, NR]
Rhizopogon roseolus (Corda) Th. Fr. [‡, NR]
Rubinoboletus ballouii (Peck) Heinem. & Ramme-
 loo [‡, NR]
Scleroderma areolatum Ehrenb. [†, ‡]
Scleroderma aurantium (L.) Pers. [‡, NR]
Scleroderma bovista Fr. [‡, NR]
Scleroderma cepa Pers. [‡, NR]
Scleroderma citrinum Pers. [†, ‡]
Scleroderma flavidum Ellis & Everh. [‡, NR]
Scleroderma polyrhizum (J.F. Gmel.) Pers. [†, ‡]
Scleroderma septentrionale Jeppson [‡, NR]
Scleroderma verrucosum (Bull.) Pers. [†]
Serpula lacrymans (Wulfen) P. Karst. [†, ‡]
Strobilomyces confusus Singer [‡, NR]
Strobilomyces strobilaceus (Scop.) Berk. [†, ‡]
Suillus acidus (Peck) Singer [‡, NR]
Suillus americanus (Peck) Snell [‡, NR]
Suillus collinitus (Fr.) Kuntze [†]
Suillus granulatus (L.) Roussel [‡, NR]
Suillus grevillei (Klotzsch) Singer [‡, NR]
Suillus hirtellus (Peck) Kuntze [‡, NR]
Suillus luteus (L.) Roussel [†, ‡]
Suillus pictus A.H. Sm. & Thiers [†, ‡]
Suillus purpureus (Fr. & Hök) Kuntze [†]
Suillus salmonicolor (Frost) Halling [‡, NR]
Suillus subaureus (Peck) Snell [‡, NR]
Tapinella atrotomentosa (Batsch) Šutara [‡, NR]
Tapinella panuoides (Fr.) E.-J. Gilbert [†, ‡]
Tylopilus alboater (Schwein.) Murrill [‡, NR]
Tylopilus eximius (Peck) Singer [†]
Tylopilus felleus (Bull.) P. Karst. [†, ‡]
Tylopilus indecisus (Peck) Murrill [‡, NR]
Tylopilus rubrobrunneus Mazzer & A.H. Sm. [‡, NR]
Tylopilus sordidus (Frost) A.H. Sm. & Thiers [‡, NR]
Veligaster nitidus (Berk.) Guzmán & Tapia [†]
Xanthoconium affine (Peck) Singer [†, ‡]
Xanthoconium affine var. *maculosus* (Peck) Singer [‡, NR]

Xanthoconium purpureum Snell & E.A. Dick [‡, NR]
Xanthoconium separans (Peck) Halling & Both [‡, ‡]
Xerocomellus chrysenderon (Bull.) Šutara [‡, ‡]
Xerocomus rubellus (Krombh.) Quél. [‡, ‡]
Xerocomus subtomentosus (L.) Quél. [‡, ‡]
Xerocomus truncatus Singer, Snell & E.A. Dick [‡, NR]

Cantharellales

Botryobasidium medium J. Erikss. [‡, NR]
Botryobasidium subcoronatum (Höhn. & Litsch.) Donk [‡, NR]
Botryobasidium vagum (Berk. & M.A. Curtis) D.P. Rogers [‡, NR]
Botryohypochnus isabellinus (Fr.) J. Erikss. [‡, NR]
Cantharellus chicagoensis Leacock, J. Riddell, Rui Zhang & G.M. Muell. [‡, ‡]
Cantharellus cibarius Fr. [‡, ‡]
Cantharellus cinnabarinus (Schwein.) Schwein. [‡, ‡]
Cantharellus flavus M.J. Foltz & T.J. Volk [‡, NR]
Cantharellus ignicolor R.H. Petersen [‡, NR]
Cantharellus infundibuliformis (Scop.) Fr. [‡, ‡]
Cantharellus lateritius (Berk.) Singer [‡, ‡]
Cantharellus lutescens (Pers.) Fr. [‡]
Cantharellus minor Peck [‡, ‡]
Cantharellus tubaeformis Fr. [‡]
Cantharellus wrightii Berk. & M.A. Curtis [‡]
Ceratobasidium stevensii (Burt) P.H.B. Talbot [‡, NR]
Clavulina amethystina (Bull.) Donk [‡]
Clavulina cinerea (Bull.) J. Schröt. [‡, ‡]
Clavulina cristata (Holmsk.) J. Schröt. [‡]
Craterellus convolvulatus (A.H. Sm.) Eyssart. & Buyck [‡, NR]
Craterellus cornucopioides (L.) Pers. [‡, ‡]
Craterellus dubius Peck [‡, NR]
Craterellus fallax A.H. Sm. [‡, NR]
Haplotrichum croceum (Mont.) Partr. & Morgan-Jones [‡, NR]
Haplotrichum curtisii (Berk.) Hol.-Jech. [‡, ‡]
Haplotrichum simile (Berk.) Hol.-Jech. [‡, NR]
Hydnellum aurantiacum (Batsch) P. Karst. [‡, NR]
Hydnellum caeruleum (Hornem.) P. Karst. [‡, NR]
Hydnellum conrescens (Pers.) Banker [‡, NR]
Hydnellum peckii Banker [‡, NR]
Hydnellum scrobiculatum (Fr.) P. Karst. [‡, NR]
Hydnellum spongiosipes (Peck) Pouzar [‡, NR]
Hydnum albiceps Berk. & Ravenel ex Cooke [‡, NR]

Hydnum albidum Peck [‡, ‡]
Hydnum combinans Peck [‡]
Hydnum flabelliforme Berk. [‡, NR]
Hydnum membranaceum Bull. [‡, NR]
Hydnum mucidum Pers. ex J.F. Gmel. [‡]
Hydnum repandum L. [‡, ‡]
Hydnum umbilicatum Peck [‡, NR]
Hydnum velutinum Fr. [‡, NR]
Multiclavula mucida (Pers.) R.H. Petersen [‡, ‡]
Pseudocraterellus undulatus (Pers.) Rauschert [‡, NR]
Rhizoctonia solani J.G. Kühn [‡, ‡]
Tulasnella bifrons Bourdot & Galzin [‡, NR]
Tulasnella pinicola Bres. [‡, NR]
Tulasnella pruinosa Bourdot & Galzin [‡, NR]
Tulasnella violacea (Johan-Olsen) Juel [‡, NR]
Tulasnella violea (Quél.) Bourdot & Galzin [‡, NR]

Corticiales

Corticium roseocarneum (Schwein.) Hjortstam [‡, ‡]
Corticium roseum Pers. [‡, NR]
Cytidia salicina (Fr.) Burt [‡, NR]
Dendrothele acerina (Pers.) P.A. Lemke [‡, NR]
Dendrothele candida (Schwein.) P.A. Lemke [‡, ‡]
Dendrothele macrodens (Coker) P.A. Lemke [‡, NR]
Dendrothele nivosa (Berk. & M.A. Curtis ex Höhn. & Litsch.) P.A. Lemke [‡, ‡]
Mycolindtneria trachyspora (Bourdot & Galzin) Rauschert [‡, NR]
Punctularia strigosozonata (Schwein.) P.H.B. Talbot [‡, NR]

Dacrymycetales

Calocera cornea (Batsch) Fr. [‡, ‡]
Dacrymyces australis Lloyd [‡, NR]
Dacrymyces chrysospermus Berk. & M.A. Curtis [‡, NR]
Dacrymyces ellisii Coker [‡, NR]
Dacrymyces stillatus Nees [‡, NR]
Dacryopinax spathularia (Schwein.) G.W. Martin [‡, ‡]

Exobasidiales

Exobasidium symploci Ellis & G. Martin [‡, NR]
Exobasidium vaccinii (Fuckel) Woronin [‡, ‡]

Gaestrales

Gastrum arenarium Lloyd [‡, NR]
Gastrum coronatum Pers. [‡, NR]
Gastrum elegans Vittad. [‡, NR]
Gastrum fimbriatum Fr. [‡, NR]

Geastrum lageniforme Vittad. [‡, NR]
Geastrum minimum Schwein. [†, ‡]
Geastrum morgani Lloyd [‡, NR]
Geastrum rufescens Pers. [‡, NR]
Geastrum saccatum Fr. [†, ‡]
Geastrum triplex Jungh. [†, ‡]
Geastrum velutinum Morgan [‡, NR]
Myriostoma coliforme (Dicks.) Corda [‡, NR]
Sphaerobolus stellatus Tode [‡, NR]

Gloeophyllales

Gloeophyllum sepiarium (Wulfen) P. Karst. [†, ‡]
Gloeophyllum trabeum (Pers.) Murrill [†, ‡]
Neolentinus suffrutescens (Brot.) T.W. May & A.E. Wood [†, ‡]

Gomphales

Clavariadelphus pistillaris (L.) Donk [†, ‡]
Corallium formosum (Pers.) G. Hahn [†]
Gomphus floccosus (Schwein.) Singer [‡, NR]
Kavinia himantia (Schwein.) J. Erikss. [‡, NR]
Lentaria byssiseda Corner [‡, NR]
Lentaria micheneri (Berk. & M.A. Curtis) Corner [‡, NR]
Lentaria patouillardii (Bres.) Corner [‡, NR]
Ramaria abietina (Pers.) Quél. [‡, NR]
Ramaria apiculata (Fr.) Donk [‡, NR]
Ramaria aurea (Schaeff.) Quél. [†, ‡]
Ramaria botrytis (Pers.) Ricken [‡, NR]
Ramaria decurrens (Pers.) R.H. Petersen [‡, NR]
Ramaria gracilis (Pers.) Quél. [†]
Ramaria murrillii (Coker) Corner [†]
Ramaria obtusissima (Peck) Corner [‡, NR]
Ramaria stricta (Pers.) Quél. [‡, NR]
Ramaria stricta var. *concolor* Corner [‡, NR]
Ramaria suecica (Fr.) Donk [†]
Ramaricium polyporoideum (Berk. & M.A. Curtis) Ginns [‡, NR]
Ramariopsis kunzei (Fr.) Corner [‡, NR]

Hymenochaetales

Basidirodulum radula (Fr.) Nobles [†]
Coltricia cinnamomea (Jacq.) Murrill [†, ‡]
Coltricia focicola (Berk. & M.A. Curtis) Murrill [†]
Coltricia montagnei (Fr.) Murrill [‡, NR]
Coltricia perennis (L.) Murrill [†, ‡]
Cotylidia pallida (Pers.) Biodin [†]
Fibricium rude (P. Karst.) Jülich [‡, NR]
Fulvifomes johnsonianus (Murrill) Y.C. Dai [‡, NR]
Fuscoporia gilva (Schwein.) T. Wagner & M. Fisch. [†, ‡]
Hydnochaete olivacea (Schwein.) Banker [†, ‡]

Hymenochaete cinnamomea subsp. *spreta* (Peck) Parmasto [†, ‡]
Hymenochaete corrugata (Fr.) Lév. [‡, NR]
Hymenochaete curtisii (Berk.) Morgan [‡, NR]
Hymenochaete fuliginosa (Pers.) Lév. [†, ‡]
Hymenochaete rubiginosa (Dicks.) Lév. [†, ‡]
Hyphodontia arguta (Fr.) J. Erikss. [‡, NR]
Hyphodontia aspera (Fr.) J. Erikss. [‡, NR]
Hyphodontia bugellensis (Ces.) J. Erikss. [‡, NR]
Hyphodontia latitans (Bourdot & Galzin) Ginns & M.N.L. Lefebvre [‡, NR]
Hyphodontia quercina (Pers.) J. Erikss. [‡, NR]
Hyphodontia spathulata (Schrad.) Parmasto [‡, NR]
Inonotus andersonii (Ellis & Everh.) Cerný [‡, NR]
Inonotus cuticularis (Bull.) P. Karst. [†, ‡]
Inonotus dryadeus Fr. [†]
Inonotus glomeratus (Peck) Murrill [‡, NR]
Inonotus hispidus (Bull.) P. Karst. [†, ‡]
Inonotus obliquus (Ach. ex Pers.) Pilát [†]
Inonotus radiatus (Sowerby) P. Karst. [†, ‡]
Onnia tomentosa (Fr.) P. Karst. [‡, NR]
Oxyporus corticola (Fr.) Ryvarden [†, ‡]
Oxyporus latemarginatus (Durieu & Mont.) Donk [†, ‡]
Oxyporus pellicula (Jungh.) Ryvarden [‡, NR]
Oxyporus populinus (Schumach.) Donk [†, ‡]
Phellinus conchatus (Pers.) Quél. [†, ‡]
Phellinus contiguus (Pers.) Pat. [‡, NR]
Phellinus everhartii (Ellis & Galloway) A. Ames [†, ‡]
Phellinus ferrugineofuscus (P. Karst.) Bourdot [‡, NR]
Phellinus ferruginosus (Schrad.) Pat. [‡, NR]
Phellinus gilvus (Schwein.) Pat. [†, ‡]
Phellinus igniarius (L.) Quél. [†, ‡]
Phellinus laevigatus (Fr.) Bourdot & Galzin [‡, NR]
Phellinus nigricans (Fr.) P. Karst. [†, ‡]
Phellinus prunicola (Murrill) Gilb. [‡, NR]
Phellinus punctatus (P. Karst.) Pilát [‡, NR]
Phellinus ribis (Schumach.) Quél. [†, ‡]
Phellinus rimosus (Berk.) Pilát [‡, NR]
Phellinus robiniae (Murrill) A. Ames [‡, NR]
Phellinus robustus (P. Karst.) Bourdot & Galzin [‡, NR]
Phellinus viticola (Schwein.) Donk [‡, NR]
Polystictus hirsutus var. *marginatus* (Bres. ex Schulzer) Sacc. [†, ‡]
Porodaedalea pini (Brot.) Murrill [†]
Pseudochaete tabacina (Sowerby) T. Wagner & M. Fisch. [‡, NR]
Pycnoporus cinnabarinus (Jacq.) P. Karst. [†, ‡]
Rickenella fibula (Bull.) Raithehl. [‡, NR]

Schizopora paradoxa (Schrad.) Donk [†, ‡]
Schizopora radula (Pers.) Hallenb. [‡, NR]
Skvortzovia furfurella (Bres.) Bononi & Hjortstam
 [‡, NR]

Phallales

Lysurus cruciatus (Lepr. & Mont.) Henn. [†, ‡]
Lysurus periphragmoides (Klotzsch) Dring [†, ‡]
Mutinus caninus (Huds.) Fr. [†, ‡]
Mutinus elegans (Mont.) E. Fisch [†]
Mutinus ravenelii (Berk. & M.A. Curtis) E. Fisch.
 [†]
Phallus hadriani Vent. [‡, NR]
Phallus impudicus L. [†, ‡]
Phallus impudicus var. *togatus* (Kalchbr.) Cos-
 tantin & L.M. Dufour [†, ‡]
Phallus ravenelii Berk. & M.A. Curtis [‡, NR]

Polyporales

Abortiporus biennis (Bull.) Singer [†, ‡]
Antrodia albida (Fr.) Donk [†, ‡]
Antrodia albobrunnea (Romell) Ryvar-
 den [‡, NR]
Antrodia juniperina (Murrill) Niemelä & Ryvar-
 den [‡, NR]
Antrodia lenis (P. Karst.) Ryvar-
 den [‡, NR]
Antrodia malicola (Berk. & M.A. Curtis) Donk [‡,
 NR]
Antrodia oleracea (R.W. Davidson & Lombard)
 Ryvar-
 den [‡, NR]
Antrodia sinuosa (Fr.) P. Karst. [‡, NR]
Antrodia xantha (Fr.) Ryvar-
 den [‡, NR]
Antrodiella semisupina (Berk. & M.A. Curtis)
 Ryvar-
 den [‡, NR]
Aurantiporus fissilis (Berk. & M.A. Curtis) H.
 Jahn ex Ryvar-
 den [‡, NR]
Bjerkandera adusta (Willd.) P. Karst. [†, ‡]
Bjerkandera fumosa (Pers.) P. Karst. [†, ‡]
Byssomerulius incarnatus (Schwein.) Gilb. [†, ‡]
Ceriporia purpurea (Fr.) Donk [‡, NR]
Ceriporia spissa (Schwein. ex Fr.) Rajchenb. [‡,
 NR]
Ceriporia tarda (Berk.) Ginns [†, ‡]
Ceriporia viridans (Berk. & Broome) Donk [‡,
 NR]
Ceriporiopsis mucida (Pers.) Gilb. & Ryvar-
 den [‡,
 NR]
Ceriporiopsis subrufa (Ellis & Dearn.) Ginns [‡,
 NR]
Cerrena unicolor (Bull.) Murrill [†, ‡]
Climacocystis borealis (Fr.) Kotl. & Pouzar [‡,
 NR]
Climacodon pulcherrimus (Berk. & M.A. Curtis)
 Nikol. [†, ‡]
Climacodon septentrionalis (Fr.) P. Karst. [†, ‡]

Corioloopsis floccosa (Jungh.) Ryvar-
 den [†, ‡]
Corioloopsis gallica (Fr.) Ryvar-
 den [†, ‡]
Corioloopsis trogii (Berk.) Domanski [‡, NR]
Dacryobolus sudans (Alb. & Schwein.) Fr. [‡, NR]
Daedalea quercina (L.) Pers. [†, ‡]
Daedaleopsis confragosa var. *confragosa* (Bolton)
 J. Schröt. [†, ‡]
Daedaleopsis septentrionalis (P. Karst.) Niemelä
 [‡, NR]
Datronia mollis (Sommerf.) Donk [‡, NR]
Datronia scutellata (Schwein.) Gilb. & Ryvar-
 den [‡, NR]
Diplomitoporus overholtsii (Pilát) Gilb. & Ryvar-
 den [‡, NR]
Erastia salmonicolor (Berk. & M.A. Curtis)
 Niemelä & Kinnunen [‡, NR]
Favolus albidus Masee [‡, NR]
Fibroporia radiculosa (Peck) Parmasto [‡, NR]
Fomes fomentarius (L.) Fr. [†, ‡]
Fomes fulvus (Scop.) Gillet [†, ‡]
Fomes meliae (Underw.) Murrill [‡, NR]
Fomitopsis cajanderi (P. Karst.) Kotl. & Pouzar [†]
Fomitopsis durescens (Overh. ex J. Lowe) Gilb. &
 Ryvar-
 den [‡, NR]
Fomitopsis palustris (Berk. & M.A. Curtis) Gilb.
 & Ryvar-
 den [‡, NR]
Fomitopsis spraguei (Berk. & M.A. Curtis) Gilb. &
 Ryvar-
 den [†, ‡]
Fuscocerrena portoricensis (Spreng. ex Fr.) Ry-
 var-
 den [‡, NR]
Ganoderma australe (Fr.) Pat. [‡, NR]
Ganoderma applanatum (Pers.) Pat. [†, ‡]
Ganoderma cupreolaccatum (Kalchbr.) Z.
 Igmándy [‡, NR]
Ganoderma curtisii (Berk.) Murrill [‡, NR]
Ganoderma lipsiense (Batsch) G.F. Atk. [†, ‡]
Ganoderma lucidum (Curtis) P. Karst. [†, ‡]
Ganoderma resinaceum Boud. [‡, NR]
Ganoderma sessile Murrill [†, ‡]
Ganoderma tsugae Murrill [‡, NR]
Globifomes graveolens (Schwein.) Murrill [†, ‡]
Gloeoporus dichrous (Fr.) Bres. [†, ‡]
Gloeoporus theleporoides (Hook.) G. Cunn. [†, ‡]
Grifola frondosa (Dicks.) Gray [†, ‡]
Hapalopilus croceus (Pers.) Bondartsev & Singer
 [†, ‡]
Hapalopilus nidulans (Fr.) P. Karst. [‡, NR]
Hapalopilus rutilans (Pers.) Murrill [‡, NR]
Haploporus odoratus (Sommerf.) Bondartsev &
 Singer ex Singer [‡, NR]
Heliocybe sulcata (Berk.) Redhead & Ginns [‡,
 NR]
Hjortstamia crassa (Lév.) Boidin & Gilles [‡, NR]

- Hyphoderma litschaueri* (Burt) J. Erikss. & Å. Strid [‡, NR]
Hyphoderma mutatum (Peck) Donk [†, ‡]
Hyphoderma populneum (Peck) Donk [‡, NR]
Hyphoderma puberum (Fr.) Wallr. [‡, NR]
Hyphoderma rubropallens (Schwein.) Ginns [‡, NR]
Hyphoderma setigerum (Fr.) Donk [‡, NR]
Hyphoderma tenue (Pat.) Donk [‡, NR]
Hypochnicium lundellii (Bourdot) J. Erikss. [‡, NR]
Inonotus andersonii (Ellis & Everh.) Cerný [‡, NR]
Inonotus cuticularis (Bull.) P. Karst. [†]
Inonotus hispidus (Bull.) P. Karst. [†]
Inocutis rheades (Pers.) Fiasson & Niemelä [†]
Irpex fuscescens Schwein. [†, ‡]
Irpex lacteus (Fr.) Fr. [†, ‡]
Ischnoderma benzoinum (Wahlenb.) P. Karst. [†, ‡]
Junghuhnia nitida (Pers.) Ryvarden [‡, NR]
Laetiporus cincinnatus (Morgan) Burds. [‡, NR]
Laetiporus sulphureus (Bull.) Murrill [†, ‡]
Lentinus levis (Berk. & M.A. Curtis) Murrill [‡, NR]
Lentinus strigosus Fr. [†, ‡]
Lentinus tigrinus (Bull.) Fr. [†, ‡]
Lenzites betulina (L.) Fr. [†, ‡]
Lenzites elegans (Spreng.) Pat. [†, ‡]
Lenzites flaccida Fr. [†]
Lenzites stereoides (Fr.) Ryvarden [‡, NR]
Lopharia cinerascens (Schwein.) G. Cunn. [†, ‡]
Melanoporia nigra (Berk.) Murrill [†, ‡]
Meripilus giganteus (Pers.) P. Karst. [†, ‡]
Microporellus obovatus (Jungh.) Ryvarden [‡, NR]
Mutatoderma heterocystidiatum (Burt) C.E. Gómez [‡, NR]
Mycoacia aurea (Fr.) J. Erikss. & Ryvarden [‡, NR]
Mycoacia uda (Fr.) Donk [‡, NR]
Mycorrhaphium adustum (Banker) Ryvarden [‡, NR]
Mycorrhaphium adustum (Schwein.) Maas Geest. [†, ‡]
Osteina obducta (Berk.) Donk [‡, NR]
Panus conchatus (Bull.) Fr. [†, ‡]
Perenniporia fraxinea (Bull.) Ryvarden [‡, NR]
Perenniporia fraxinophila (Peck) Ryvarden [‡, NR]
Perenniporia medulla-panis (Jacq.) Donk [†, ‡]
Perenniporia ohioensis (Berk.) Ryvarden [‡, NR]
Perenniporia robiniophila (Murrill) Ryvarden [†, ‡]
Perenniporia subacida (Peck) Donk [‡, NR]
Perenniporia tenuis (Schwein.) Ryvarden [‡, NR]
Perenniporia tenuis var. *pulchella* (Schwein.) Gilb. & Ryvarden [‡, NR]
Perenniporia variegata Ryvarden & Gilb. [‡, NR]
Phaeolus schweinitzii (Fr.) Pat. [‡, NR]
Phanerochaete affinis (Burt) Parmasto [‡, NR]
Phanerochaete avellanea (Bres.) J. Erikss. & Hjortstam [‡, NR]
Phanerochaete burtii (Romell) Parmasto [‡, NR]
Phanerochaete chrysorhiza (Torr.) Budington & Gilb. [†, ‡]
Phanerochaete filamentosa (Berk. & M.A. Curtis) Burds. [‡, NR]
Phanerochaete laevis (Fr.) J. Erikss. & Ryvarden [‡, NR]
Phanerochaete sanguinea (Fr.) Pouzar [‡, NR]
Phanerochaete sordida (P. Karst.) J. Erikss. & Ryvarden [†, ‡]
Phanerochaete velutina (DC.) P. Karst. [‡, NR]
Phlebia albida H. Post [‡, NR]
Phlebia chrysocreas (Berk. & M.A. Curtis) Burds. [‡, NR]
Phlebia cinnabarina Fr. [‡, NR]
Phlebia hydnoidea Schwein. [†]
Phlebia livida (Pers.) Bres. [‡, NR]
Phlebia ludoviciana (Burt) Nakasone & Burds. [‡, NR]
Phlebia queletii (Bourdot & Galzin) M.P. Christ. [‡, NR]
Phlebia radiata Fr. [†, ‡]
Phlebia tremellosa (Schröd.) Nakasone & Burds. [†, ‡]
Phlebiella tulasnelloidea (Höhn. & Litsch.) Ginns & M.N.L. Lefebvre [‡, NR]
Phlebiella vaga (Fr.) P. Karst. [‡, NR]
Phlebiopsis flavidoalba (Cooke) Hjortstam [‡, NR]
Phlebiopsis gigantea (Fr.) Jülich [‡, NR]
Piptoporus betulinus (Bull.) P. Karst. [‡, NR]
Podoscypha ravenelii (Berk. & M.A. Curtis) Pat. [†]
Polyporus admirabilis Peck [†, ‡]
Polyporus albus (Huds.) Fr. [‡, NR]
Polyporus alveolaris (DC.) Bondartsev & Singer [†, ‡]
Polyporus arcularius (Batsch) Fr. [†, ‡]
Polyporus badius (Pers.) Schwein. [†, ‡]
Polyporus brumalis (Pers.) Fr. [†, ‡]
Polyporus epileucus Fr. [†, ‡]
Polyporus incertus Pers. [†]
Polyporus melanopus (Pers.) Fr. [†, ‡]
Polyporus osseus Kalchbr. [†]
Polyporus paragamenus Fr. [†]
Polyporus radicans Schwein. [†, ‡]

- Polyporus rigidus* Lév. [‡, NR]
Polyporus salignus (Fr.) Fr. [‡, NR]
Polyporus squamosus (Huds.) Fr. [†, ‡]
Polyporus varius (Pers.) Fr. [†, ‡]
Poria attenuata (Peck) Sacc. [†]
Poria bresadolae Bourdot & Galzin [‡, NR]
Poria discolor Overh. [‡, NR]
Poria homaema Sacc. [‡, NR]
Poria niphodes (Berk. & Broome) Sacc. [‡, NR]
Poria vitellina (Schwein.) Sacc. [‡, NR]
Poria xantholoma (Schwein.) Cooke [‡, NR]
Poronidulus conchifer (Schwein.) Murrill [†, ‡]
Postia caesia (Schrad.) P. Karst. [‡, NR]
Postia floriformis (Quél.) Jülich [‡, NR]
Postia fragilis (Fr.) Jülich [‡, NR]
Postia guttulata (Peck) Jülich [‡, NR]
Postia tephroleuca (Fr.) Jülich [†, ‡]
Pycnoporellus fulgens (Fr.) Donk [‡, NR]
Pycnoporus cinnabarinus (Jacq.) P. Karst. [†, ‡]
Pycnoporus sanguineus (L.) Murrill [†, ‡]
Rigidoporus crocatus (Pat.) Ryvarden [‡, NR]
Rigidoporus lineatus (Pers.) Ryvarden [‡, NR]
Rigidoporus undatus (Pers.) Donk [†, ‡]
Rigidoporus vitreus (Pers.) Donk [‡, NR]
Sarcodontia setosa (Pers.) Donk [‡, NR]
Scopuloides hydnooides (Cooke & Masee) Hjortstam & Ryvarden [‡, NR]
Skeletocutis alutacea (J. Lowe) Jean Keller [‡, NR]
Skeletocutis nivea (Jungh.) Jean Keller [†, ‡]
Skeletocutis odora (Peck ex Sacc.) Ginns [‡, NR]
Solenia conferta Burt [†]
Sparassis crispa (Wulfen) Fr. [†]
Sparassis laminosa Fr. [‡, NR]
Spongipellis delectans (Peck) Murrill [†, ‡]
Spongipellis pachyodon (Pers.) Kotl. & Pouzar [†, ‡]
Spongipellis spumeus (Sowerby) Pat. [‡, NR]
Spongipellis unicolor (Schwein.) Murrill [†, ‡]
Sporotrichum byssinum Link [‡, NR]
Sporotrichum chryseum Peck [†, ‡]
Steccherinum ciliolatum (Berk. & M.A. Curtis) Gilb. & Budington [‡, NR]
Steccherinum fimbriatum (Pers.) J. Erikss. [‡, NR]
Steccherinum ochraceum (Pers. ex J.F. Gmel.) Gray [†, ‡]
Terana caerulea (Schrad. ex Lam.) Kuntze [†]
Trametes aesculi (Fr.) Justo [‡, NR]
Trametes cubensis (Mont.) Sacc. [‡, NR]
Trametes gibbosa (Pers.) Fr. [‡, NR]
Trametes hirsuta (Wulfen) Pilát [†, ‡]
Trametes ochracea (Pers.) Gilb. & Ryvarden [†, ‡]
Trametes pubescens (Schumach.) Pilát [†, ‡]
Trametes suaveolens (L.) Fr. [‡, NR]
- Trametes versicolor* (L.) Lloyd [†, ‡]
Trametes villosa (Sw.) Kreisel [‡, NR]
Trametopsis cervina (Schwein.) Tomsovský [‡, NR]
Trichaptum abietinum (Pers. ex J.F. Gmel.) Ryvarden [†, ‡]
Trichaptum bifforme (Fr.) Ryvarden [†, ‡]
Trichaptum subchartaceum (Murrill) Ryvarden [‡, NR]
Tyromyces chioneus (Fr.) P. Karst. [†, ‡]
Tyromyces fumidiceps G.F. Atk. [‡, NR]
Tyromyces galactinus (Berk.) J. Lowe [‡, NR]
Wolfiporia dilatohypha Ryvarden & Gilb. [‡, NR]
Wolfiporia extensa (Peck) Ginns [†, ‡]
- Russulales**
- Albatrellus cristatus* (Schaeff.) Kotl. & Pouzar [†, ‡]
Aleurodiscus andinus Núñez & Ryvarden [‡, NR]
Aleurodiscus oakesii (Berk. & M.A. Curtis) Pat. [†, ‡]
Artomyces pyxidatus (Pers.) Jülich [†, ‡]
Asterostroma andinum Pat. [‡, NR]
Auriscalpium vulgare Gray [†, ‡]
Bondarzewia berkeleyi (Fr.) Bondartsev & Singer [†, ‡]
Chondrostereum purpureum (Pers.) Pouzar [‡, NR]
Conferticum ochraceum (Fr.) Hallenb. [‡, NR]
Dichostereum granulosum (Fr.) Boidin & Lanq. [†]
Gloeopeniophorella convolvens (P. Karst.) Boidin, Lanq. & Gilles [‡, NR]
Gloiodon strigosus (Sw.) P. Karst. [†]
Hericium coralloides (Scop.) Pers. [†, ‡]
Hericium erinaceus (Bull.) Pers. [†, ‡]
Heterobasidion annosum (Fr.) Bref. [‡, NR]
Lactarius affinis Peck [†]
Lactarius aquizonatus Kytöv. [‡, NR]
Lactarius atroviridis Peck [‡, NR]
Lactarius camphoratus (Bull.) Fr. [†, ‡]
Lactarius carbonicola A.H. Sm. [‡, NR]
Lactarius chrysorrhoeus Fr. [†, ‡]
Lactarius corrugis Peck [†]
Lactarius deceptivus Peck [†, ‡]
Lactarius deliciosus (L.) Gray [†]
Lactarius distans Peck [†]
Lactarius fragilis (Burl.) Hesler & A.H. Sm. [‡, NR]
Lactarius fuliginellus A.H. Sm. & Hesler [‡, NR]
Lactarius fumosus Peck [‡, NR]
Lactarius gerardii Peck [†, ‡]
Lactarius glaucescens Crossl. [‡, NR]
Lactarius griseus Peck [‡, NR]

- Lactarius hygrophoroides* var. *hygrophoroides* Berk. & M.A. Curtis [†, ‡]
Lactarius hygrophoroides var. *lavandulaceus* Hesler & A.H. Sm. [‡, NR]
Lactarius hyginus (Fr.) Fr. [†]
Lactarius imperceptus Beardslee & Burl. [‡, NR]
Lactarius indigo (Schwein.) Fr. [†, ‡]
Lactarius insulsus (Fr.) Fr. [†, ‡]
Lactarius lignyotus Fr. [†, ‡]
Lactarius maculatipes Burl. [‡, NR]
Lactarius maculatus Peck [‡, NR]
Lactarius mutabilis Peck [‡, NR]
Lactarius piperatus (L.) Pers. [†, ‡]
Lactarius plumbeus (Bull.) Fr. [†]
Lactarius psammicola A.H. Sm. [‡, NR]
Lactarius pubescens Fr. [‡, NR]
Lactarius pyrogalus (Bull.) Fr. [†, ‡]
Lactarius quietus (Fr.) Fr. [‡, NR]
Lactarius quietus var. *incanus* Hesler & A.H. Sm. [‡, NR]
Lactarius rufus (Scop.) Fr. [†]
Lactarius scrobiculatus (Scop.) Fr. [†, ‡]
Lactarius seriffuus (DC.) Fr. [†, ‡]
Lactarius sordidus Peck [†]
Lactarius subplinthogalus Coker [‡, NR]
Lactarius subpurpureus Peck [‡, NR]
Lactarius subseriffuus Longyear [‡, NR]
Lactarius subvernalis var. *cokeri* (A.H. Sm. & Hesler) Hesler & A.H. Sm. [‡, NR]
Lactarius tabidus Fr. [†]
Lactarius trivialis (Fr.) Fr. [†, ‡]
Lactarius uvidus (Fr.) Fr. [†]
Lactarius vellereus var. *vellereus* [†]
Lactarius volemus var. *flavus* Hesler & A.H. Sm. [‡, NR]
Lactarius volemus var. *volemus* (Fr.) Fr. [†, ‡]
Lactarius zonarius (Bull.) Fr. [‡, NR]
Lactifluus subvellereus (Peck) Nuytinck [‡, NR]
Lactifluus vellereus (Fr.) Kuntze [‡, NR]
Laxitextum bicolor (Pers.) Lentz [†, ‡]
Leaia stratosa (Berk.) Banker [‡, NR]
Lentinellus cochleatus (Pers.) P. Karst. [‡, NR]
Lentinellus flabelliformis (Bolton) S. Ito [‡, NR]
Lentinellus micheneri (Berk. & M.A. Curtis) Pegler [‡, NR]
Lentinellus ursinus (Fr.) Kühner [†, ‡]
Lentinellus vulpinus (Sowerby) Kühner & Maire [†]
Peniophora albobadia (Schwein.) Boidin [‡, NR]
Peniophora cinerea (Pers.) Cooke [†, ‡]
Peniophora erikssonii Boidin [‡, NR]
Peniophora erumpens (Burt) Boidin [‡, NR]
Peniophora filamentosa (Berk. & M.A. Curtis) Moffatt [‡, NR]
Peniophora limitata (Chaillet ex Fr.) Cooke [‡, NR]
Peniophora nuda (Fr.) Bres. [‡, NR]
Peniophora rufa (Fr.) Boidin [‡, NR]
Peniophora versiformis (Berk. & M.A. Curtis) Bourdot & Galzin [‡, NR]
Peniophora violaceolivida (Sommerf.) Massee [‡, NR]
Russula adulterina Fr. [‡, NR]
Russula adusta (Pers.) Fr. [‡, NR]
Russula aeruginea Lindblad [†, ‡]
Russula albonigra (Krombh.) Fr. [‡, NR]
Russula alutacea (Pers.) Fr. [†]
Russula amoenolens Romagn. [†, ‡]
Russula anomala Peck [‡, NR]
Russula atropurpurea (Krombh.) Britzelm. [†, ‡]
Russula ballouii Peck [‡, NR]
Russula barlae Quél. [‡, NR]
Russula basifurcata Peck [†]
Russula brevipes var. *brevipes* Peck [†, ‡]
Russula brunneoalba De Marb. [‡, NR]
Russula brunneola Burl. [‡, NR]
Russula brunneoviolacea Crawshay [‡, NR]
Russula chloroides (Krombh.) Bres. [‡, NR]
Russula cinerascens Beardslee [‡, NR]
Russula claroflava Grove [‡, NR]
Russula compacta Frost [†, ‡]
Russula corinthii-rubra Burl. [‡, NR]
Russula cremeirosea Murrill [‡, NR]
Russula crustosa Peck [†, ‡]
Russula cyanoxantha (Schaeff.) Fr. [†, ‡]
Russula cyanoxantha var. *variata* (Banning) Singer [†, ‡]
Russula cystidiosa Murrill [‡, NR]
Russula decolorans (Fr.) Fr. [†]
Russula delica Fr. [†]
Russula densifolia Secr. ex Gillet [†]
Russula disparilis Burl. [‡, NR]
Russula emetica (Schaeff.) Pers. [†, ‡]
Russula flavida Frost [‡, NR]
Russula flavisiccans Bills [‡, NR]
Russula foetens Pers. [†]
Russula font-queri Singer ex Singer [‡, NR]
Russula fragilis Fr. [†]
Russula fragiloides Murrill [‡, NR]
Russula fragrantissima Romagn. [‡, NR]
Russula fucosa Burl. [‡, NR]
Russula furcata (Lam.) Pers. [†]
Russula galochroa (Fr.) Fr. [‡, NR]
Russula granulata Peck [†]
Russula heterophylla (Fr.) Fr. [‡, NR]
Russula humidicola Burl. [‡, NR]
Russula incarnaticeps Murrill [‡, NR]

- Russula ionochlora* Romagn. [‡, NR]
Russula languida Cern. & H. Raab [†]
Russula laurocerasi Melzer [‡, NR]
Russula luteobasis Peck [‡, NR]
Russula luteofolia Fatto [‡, NR]
Russula macropoda Singer [‡, NR]
Russula mariae Peck [†, ‡]
Russula michiganensis Shaffer [‡, NR]
Russula modesta Peck [‡, NR]
Russula nigricans Fr. [†]
Russula ochricompacta Bills & O.K. Mill. [‡, NR]
Russula ochrophylla Peck [†, ‡]
Russula operata Burl. [‡, NR]
Russula ornaticeps Burl. [‡, NR]
Russula paludosa Britzelm. [‡, NR]
Russula pectinata Fr. [†]
Russula pectinatoides Peck [†, ‡]
Russula pseudointegra Arnould & Goris ex R. Maire [‡, NR]
Russula pseudolepida Singer [‡, NR]
Russula puellaris Fr. [‡, NR]
Russula pulchra Burl. [‡, NR]
Russula pulverulenta Peck [‡, NR]
Russula purpurina Quél. & Schulzer [†]
Russula pusilla Peck [‡, NR]
Russula redolens Burl. [‡, NR]
Russula rosea Pers. [†]
Russula rubescens Beardslee [‡, NR]
Russula rubriceps (Kauffman) Singer [‡, NR]
Russula seperina Dupain [‡, NR]
Russula sericeonitens Kauffman [‡, NR]
Russula silvicola Schaffer [‡, NR]
Russula sordida Peck [†]
Russula squalida Peck [†, ‡]
Russula subfoetens W.G. Sm. [†, ‡]
Russula subgraminicolor Murrill [‡, NR]
Russula subpunctata Kauffman [‡, NR]
Russula subsericeonitens Murrill [‡, NR]
Russula subtilis Burl. [‡, NR]
Russula uncialis Peck [‡, NR]
Russula vesca Fr. [†]
Russula veteriosa Fr. [†]
Russula virescens (Schaeff.) Fr. [†, ‡]
Russula xerampelina (Schaeff.) Fr. [†, ‡]
Scytinostroma galactinum (Fr.) Donk [‡, NR]
Scytinostroma portentosum (Berk. & M.A. Curtis) Donk [‡, NR]
Scytinostroma protrusum (Burt) Nakasone [‡, NR]
Scytinostromella arachnoidea (Peck) Hjortstam [†]
Stereum complicatum (Fr.) Fr. [†, ‡]
Stereum gausapatum (Fr.) Fr. [†, ‡]
Stereum hirsutum (Willd.) Pers. [†, ‡]
Stereum ochraceoflavum (Schwein.) Sacc. [‡, NR]
Stereum ostrea (Blume & T. Nees) Fr. [†, ‡]
Stereum striatum (Fr.) Fr. [†, ‡]
Stereum versicolor (Sw.) Fr. [†, ‡]
Vararia investiens (Schwein.) P. Karst. [‡, NR]
Xylobolus frustulatus (Pers.) Boidin [†, ‡]
Xylobolus subpileatus (Berk. & M.A. Curtis) Boidin [‡, NR]
- Septobasidiales**
Septobasidium pteruloides (Mont.) Pat. [‡, NR]
- Thelephorales**
Hydnellum aurantiacum (Batsch) P. Karst. [‡, NR]
Hydnellum caeruleum (Hornem.) P. Karst. [‡, NR]
Hydnellum complicatum Banker [‡, NR]
Hydnellum concrescens (Pers.) Banker [†, ‡]
Hydnellum ferrugineum (Fr.) P. Karst. [‡, NR]
Hydnellum scrobiculatum (Fr.) P. Karst. [‡, NR]
Hydnellum spongiosipes (Peck) Pouzar [†, ‡]
Phellodon atratus K.A. Harrison [‡, NR]
Phellodon confluens (Pers.) Pouzar [‡, NR]
Phellodon melaleucus (Sw. ex Fr.) P. Karst. [‡, NR]
Phellodon niger (Fr.) P. Karst. [†, ‡]
Phellodon tomentosus (L.) Banker [‡, NR]
Sarcodon fennicus (P. Karst.) P. Karst. [‡, NR]
Sarcodon imbricatus (L.) P. Karst. [†, ‡]
Sarcodon scabrosus (Fr.) P. Karst. [‡, NR]
Sarcodon thwaitesii (Berk. & Broome) Maas Geest.[†]
Sarcodon versipellis (Fr.) Nikol. [‡, NR]
Thelephora albidobrunnea Schwein. [‡, NR]
Thelephora anthocephala (Bull.) Fr. [‡, NR]
Thelephora caryophyllea (Schaeff.) Pers. [†, ‡]
Thelephora cuticularis Berk. [‡, NR]
Thelephora intybacea Pers. [‡, NR]
Thelephora multipartita Schwein. [†, ‡]
Thelephora palmata (Scop.) Fr. [†, ‡]
Thelephora penicillata (Pers.) Fr. [‡, NR]
Thelephora regularis Schwein. [†, ‡]
Thelephora schweinitzii Peck [†]
Thelephora spongiosa Schwein. [‡, NR]
Thelephora terrestris Ehrh. [‡, NR]
Thelephora vialis Schwein. [‡, NR]
Tomentella botryoides (Schwein.) Bourdot & Galzin [‡, NR]
Tomentella bryophila (Pers.) M.J. Larsen [†, ‡]
Tomentella crinalis (Fr.) M.J. Larsen [†]
Tomentella fusca (Pers.) J. Schröt. [‡, NR]
Tomentella lapida (Pers.) Stalpers [‡, NR]

Tomentella rubiginosa (Bres.) Maire [‡, NR]

Trechisporales

Subulicystidium brachysporum (P.H.B. Talbot & V.C. Green) Jülich [‡, NR]

Subulicystidium longisporum (Pat.) Parmasto [‡, NR]

Trechispora farinacea (Pers.) Liberta [‡, NR]

Trechispora fastidiosa (Pers.) Liberta [‡, NR]

Trechispora mollusca (Pers.) Liberta [‡, NR]

Trechispora stevensonii (Berk. & Broome) K.H. Larss. [‡, NR]

Tremellales

Basidioidendron caesiocinereum (Höhn. & Litsch.) Luck-Allen [‡, NR]

Basidioidendron eyrei (Wakef.) Luck-Allen [‡, NR]

Ductifera pululahuana (Pat.) Wells [‡, NR]

Eichleriella deglubens (Berk. & Broome) D.A. Reid [‡, NR]

Exidia alba (Lloyd) Burt [‡, NR]

Exidia glandulosa (Bull.) Fr. [†, ‡]

Exidia nucleata (Schwein.) Burt [‡, NR]

Exidia recisa (Ditmar) Fr. [†, ‡]

Gloeotromera alba (Lloyd) Ervin [‡, NR]

Gloeotromera pululahuana (Pat.) Ervin [‡, NR]

Myxarium hyalinum (Pers.) Donk [‡, NR]

Pseudohydnum gelatinosum (Scop.) P. Karst. [†]

Sebacina concrescens (Schwein.) P. Roberts [‡, NR]

Sebacina epigaea (Berk. & Broome) Neuhoff [†]

Sebacina incrustans (Pers.) Tul. & C. Tul. [†, ‡]

Szygospora mycetophila (Peck) Ginns [†]

Tremella albida Huds. [†]

Tremella foliacea Pers. [†, ‡]

Tremella fuciformis Berk. [†, ‡]

Tremella mesenterica (Schaeff.) Retz. [†, ‡]

Tremella reticulata (Berk.) Farl. [‡, NR]

Tremella sparassoidea Lloyd [‡, NR]

Tremella vesicaria Bull. [‡, NR]

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INFRARED SPECTROSCOPIC STUDIES OF C₆₀ AND C₇₀ NANOPARTICLE INTERACTIONS WITH 2-PYRROLIDONE

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ABSTRACT. Fourier transform infrared spectroscopy was used to investigate the interactions between C₆₀ and C₇₀ nanoparticles and 2-pyrrolidone (2-pyrrolidinone, γ -lactam) in a toluene solvent environment. Infrared spectra were collected for 2-pyrrolidone in toluene, 2-pyrrolidone in toluene saturated with C₆₀, and 2-pyrrolidone in toluene saturated with C₇₀ over a concentration range of 200 to 2 microliters (μ L) of 2-pyrrolidone per mL of toluene or toluene saturated with the nanoparticles. At the higher concentrations of this range, small broadenings of the 2-pyrrolidone carbonyl stretching absorptions on the lower wavenumber side were observed when the 2-pyrrolidone was treated with C₆₀ and C₇₀. A significant shift was observed for the carbonyl absorption of 2-pyrrolidone when the 2-pyrrolidone was treated with C₆₀ at 5 and 2 μ L of 2-pyrrolidone per mL of toluene saturated with C₆₀. These observed shifts suggest a 2-pyrrolidone–C₆₀ nanoparticle interaction or complex formation. A small broadening of the carbonyl absorption on the low wavenumber side was observed when the 2-pyrrolidone was treated with C₇₀ at 5 and 2 μ L of 2-pyrrolidone per mL of toluene saturated with C₇₀. Spectral subtraction was used to reveal the carbonyl absorption for the 2-pyrrolidone–C₆₀ complex that exists at the higher concentration range, but is covered up by the intense absorption of the excess 2-pyrrolidone not interacting with C₆₀. The spectrum of 2-pyrrolidone in toluene (100 μ L of 2-pyrrolidone per mL of toluene) was subtracted from the spectrum of 2-pyrrolidone in toluene saturated with C₆₀ (100 μ L of 2-pyrrolidone per mL of toluene saturated with C₆₀). The subtraction process yielded a spectrum containing a spectral absorption very near the shifted absorption for 2-pyrrolidone treated with C₆₀ at the lower concentrations.

Keywords: Infrared spectroscopy, nanoparticles, C₆₀ and C₇₀, fullerenes, fullerene/nanoparticle interactions, 2-pyrrolidone, 2-pyrrolidinone, γ -lactam

INTRODUCTION

The interest in the interactions of C₆₀ and C₇₀ nanoparticles with a variety of molecular systems has motivated a number of studies using a variety of analysis methods (Holleman et al. 1999; Kyzyma et al. 2008; Jurow et al. 2012; King et al. 2012; Kyrey et al. 2012; Tropin et al. 2013; Bowles et al. 2014; Kirsch et al. 2015). This paper reports the results of an infrared spectroscopic study of the interactions between C₆₀ and C₇₀ nanoparticles and 2-pyrrolidone using toluene as a solvent environment (Aksenova et al. 2013).

The compound 2-pyrrolidone is a five-membered, cyclic amide sometimes referred to as gamma-lactam (γ -lactam) or 2-pyrrolidinone. There are a number of pharmaceutical and biological applications related to the 2-pyrrolidone cyclic structure (Midgley et al. 1992; Harreus et al. 2011). The C₆₀ and C₇₀ nanoparticles

dissolve in aromatic solvents such as toluene to form colored solutions; C₆₀ in toluene is blue, while C₇₀ in toluene is red (Ruoff et al. 1993). Infrared spectroscopy was used in this study to seek shifts in the carbonyl absorption of the 2-pyrrolidone as an indication of its interactions with the C₆₀ and C₇₀ nanoparticles in a toluene solvent environment.

EXPERIMENTAL METHODS

A Digilab FTS 7000 infrared spectrometer and a circle cell fitted with a ZnSe ATR rod were used to collect spectra for the investigation by averaging 500 scans at a spectral resolution of two wavenumbers. The empty circle cell was used as the background, single-beam spectrum for spectral collection. The spectrometer was purged with dry air for an hour prior to spectral collection to remove potentially interfering atmospheric water vapor. Since the 2-pyrrolidone interaction with the nanoparticles is likely an equilibrium process involving both interacting and non-interacting 2-pyrrolidone, spectral collection was carried out over a broad concentration range (200 to 2

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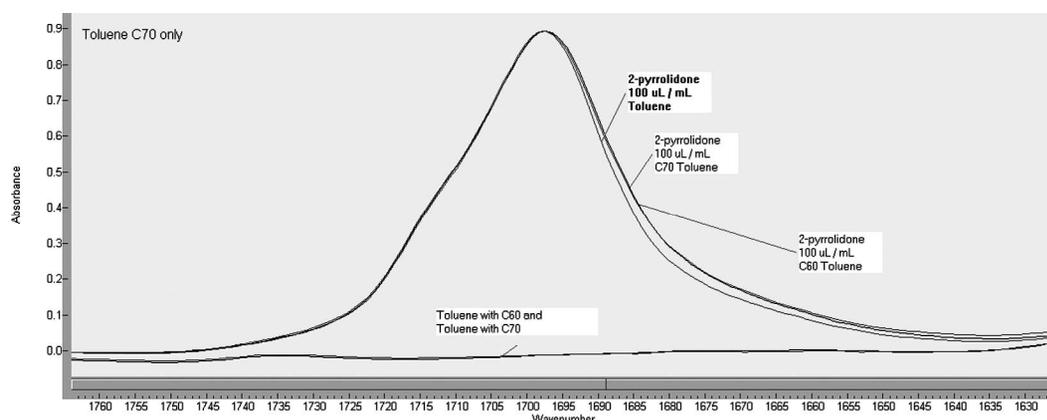


Figure 1.—The spectra of 100 μL of 2-pyrrolidone per mL of toluene, per mL of toluene saturated with C_{60} , and per mL of toluene saturated with C_{70} are shown. In addition, the spectra of toluene saturated with C_{60} and toluene saturated with C_{70} are included.

microliters (μL) of 2-pyrrolidone per mL of toluene or toluene saturated with the nanoparticles) to determine the optimal concentrations to observe infrared spectral changes.

INVESTIGATION RESULTS

Figure 1 contains the spectrum of 100 μL of 2-pyrrolidone per mL of toluene, the spectrum 100 μL of 2-pyrrolidone per mL of toluene saturated with C_{60} , and the spectrum of 100 μL of 2-pyrrolidone per mL of toluene saturated with C_{70} . In addition, the spectrum of toluene saturated with C_{60} and the spectrum of toluene saturated with C_{70} are shown. The spectra of toluene and toluene saturated with nanoparticles do not show any spectral absorption in the region of the 2-pyrrolidone carbonyl absorption. The spectra of the 2-pyrrolidone solutions containing C_{60} and C_{70} at these concentrations show a very small broadening of the carbonyl absorption on the low wavenumber side. While these broadenings are small, the broadenings were consistently observed in repeated experiments. Spectra collected at higher concentrations (200 μL of 2-pyrrolidone per mL of toluene and toluene saturated with C_{60} or C_{70}) also show consistent, small broadening on the lower wavenumbers side of the carbonyl absorption of 2-pyrrolidone resulting from its nanoparticle treatment.

Complex formation, resulting from the interactions of the nanoparticles with the 2-pyrrolidone, is likely an equilibrium process involving both 2-pyrrolidone interacting and not interacting with the nanoparticles. Of course, shifting the

equilibrium toward the 2-pyrrolidone–nanoparticle complex could result from increasing the concentration of either the nanoparticles or the 2-pyrrolidone. Changing the concentrations of the nanoparticles is limited by their low solubility in the toluene solvent, i.e., 2.8 mg of C_{60} per mL of toluene (Ruoff et al. 1993). Increasing the concentration of 2-pyrrolidone would shift the equilibrium toward any 2-pyrrolidone–nanoparticle complex; however, it would also increase the amount of the 2-pyrrolidone not interacting with the nanoparticles which also contains the infrared probe, the carbonyl bond. The higher concentrations of the non-interacting 2-pyrrolidone could cover up the carbonyl absorption of any 2-pyrrolidone–nanoparticle complex and interfere with its observation.

Concentration studies were carried out to find an optimum concentration level of 2-pyrrolidone to generate a 2-pyrrolidone–nanoparticle complex and also allow the observation of the carbonyl absorption of the complex. Figure 2 shows spectra collected at 10 μL of 2-pyrrolidone per mL of toluene and toluene saturated with C_{60} or C_{70} . The spectra show significant broadening of the carbonyl absorption of the 2-pyrrolidone treated with C_{60} , but only very slight broadening of the carbonyl absorption of 2-pyrrolidone treated with C_{70} (Fig. 2).

Spectra collected at 5 and 2 μL of 2-pyrrolidone per mL of toluene and toluene saturated with C_{60} or C_{70} are shown in Fig. 3. Intensities of the absorptions have been adjusted to help clarify positioning along the wavenumber axis and do

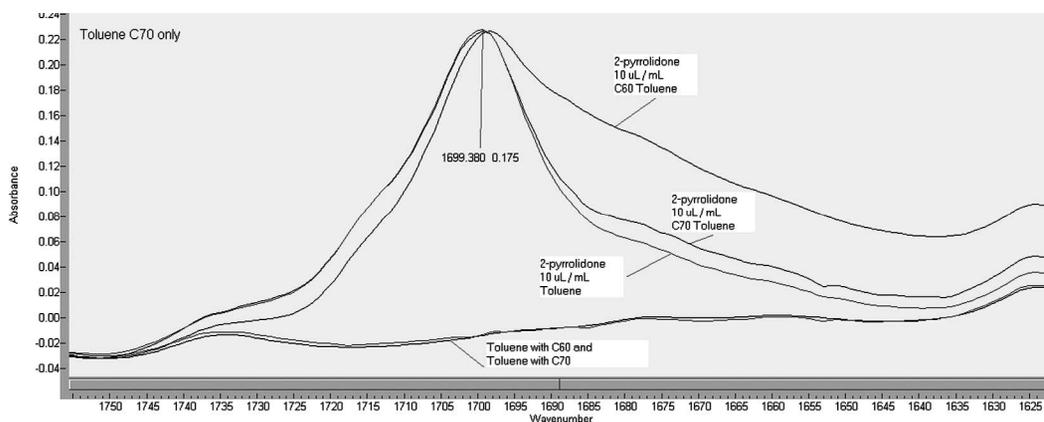


Figure 2.—The spectra of 10 μL of 2-pyrrolidone per mL of toluene, per mL of toluene saturated with C_{60} , and per mL of toluene saturated with C_{70} are shown. In addition, the spectra of toluene saturated with C_{60} and toluene saturated with C_{70} are included.

not reflect relative concentrations of 2-pyrrolidone. Figure 3 shows spectra of 2-pyrrolidone with a significant shift of the whole carbonyl absorption from about 1700 cm^{-1} to about 1687 cm^{-1} resulting from its treatment with C_{60} at 5 and 2 μL of 2-pyrrolidone per mL of toluene saturated with C_{60} . The significant shift of the 2-pyrrolidone, carbonyl absorption clearly supports a complex formation or interaction between the 2-pyrrolidone and C_{60} nanoparticles. The spectrum of 2-pyrrolidone treated with C_{70} shows only a very slight broadening of the carbonyl absorption on the low wavenumber side at 5 μL of 2-pyrrolidone per mL of toluene saturated with C_{70} .

Figure 4 shows four spectra collected over the concentration range of 10 to 5 μL of 2-pyrrolidone per mL of toluene saturated with C_{60} . The spectra show a clear transition of the absorption position of 2-pyrrolidone treated with the C_{60} at 10 μL per mL (located near 1700 cm^{-1}) to its absorption at 5 μL per mL (located near 1687 cm^{-1}). Again, intensities of absorptions are adjusted to aid in seeing spectral absorption shifts along the wavenumber axis. The spectra suggests an absorption near 1700 cm^{-1} for 2-pyrrolidone not interacting with C_{60} and an absorption for 2-pyrrolidone interacting with C_{60} near 1687 cm^{-1} (Fig. 4).

The infrared absorption observed near 1687 cm^{-1} for the complex formed between 2-pyrroli-

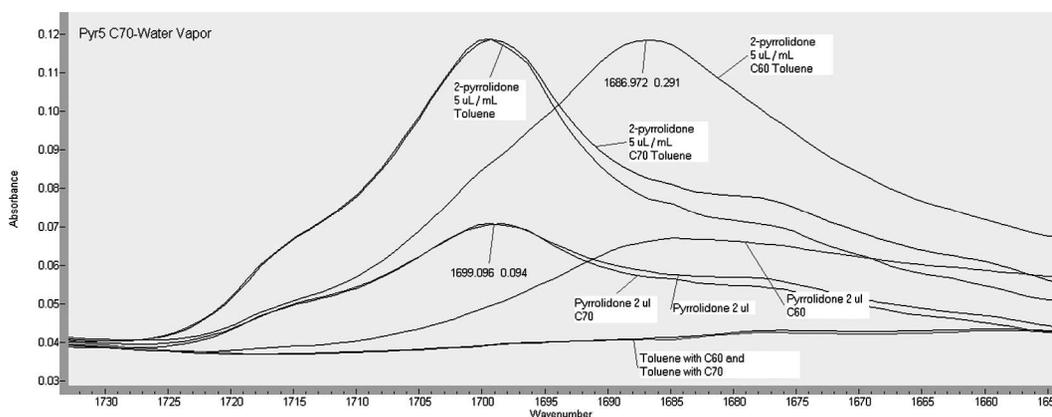


Figure 3.—The spectra of 5 and 2 μL of 2-pyrrolidone per mL of toluene, per mL of toluene saturated with C_{60} , and per mL of toluene saturated with C_{70} are shown. In addition, the spectra of toluene saturated with C_{60} and toluene saturated with C_{70} are shown.

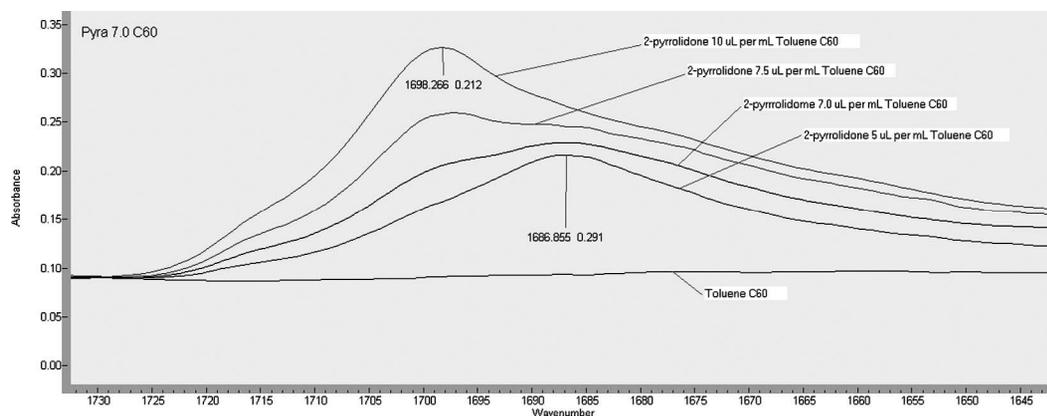


Figure 4.—The spectra of 10.0 μL , 7.5 μL , 7.0 μL , & 5.0 μL of 2-pyrrolidone per mL of toluene saturated with C_{60} are shown to clarify the carbonyl shift from near 1698 cm^{-1} to near 1687 cm^{-1} .

done and the C_{60} nanoparticles at the lower concentrations (5 and 2 μL per mL) very likely exists at the higher concentrations of 2-pyrrolidone (200 and 100 μL per mL); however, this infrared absorption may be buried under the intense carbonyl absorption of the excess, non-interacting 2-pyrrolidone.

Spectral subtraction has been shown to be a useful tool in the separation of overlapping spectral absorptions resulting from a mixture of absorbing species (Gillette & Koenig 1984; Honigs et al. 1985; Charles 1994; Siyuan et al. 2010). Figure 5 contains again the spectrum of 100 and 5 μL of 2-pyrrolidone per mL of

toluene and toluene saturated with C_{60} . The spectrum that results from subtracting the spectrum of 100 μL of 2-pyrrolidone per mL of toluene times 0.95 (subtraction factor) from the spectrum of 100 μL of 2-pyrrolidone per mL of toluene saturated with C_{60} is also given in Fig. 5. The goal of the subtraction process is to remove the intense carbonyl absorption caused by the high concentration of the non-interacting 2-pyrrolidone at this high concentration level and thus to reveal the underlying carbonyl absorption of the relatively low concentration 2-pyrrolidone- C_{60} complex. The result of the subtraction process yields a

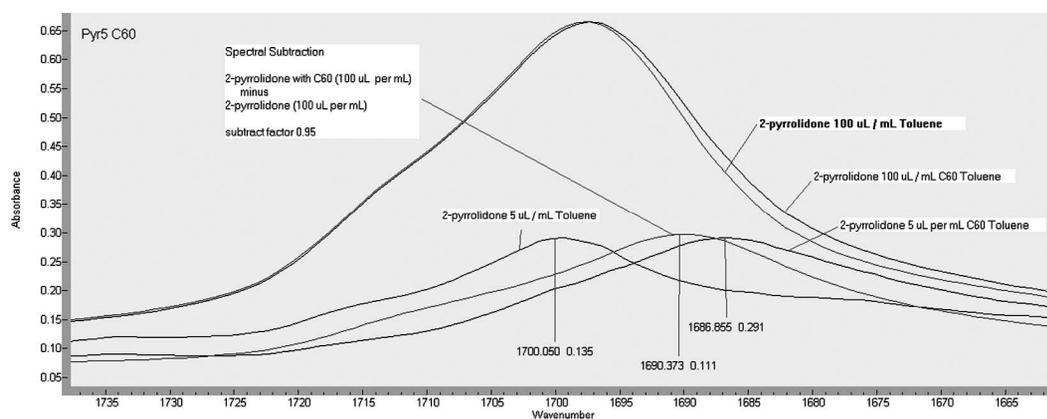


Figure 5.—The spectra of 100 μL of 2-pyrrolidone per mL of toluene, 100 μL of 2-pyrrolidone per mL of toluene saturated with C_{60} , 5 μL of 2-pyrrolidone per mL of toluene, and 5 μL of 2-pyrrolidone per mL of toluene saturated with C_{60} are shown. Also shown is the spectrum that result from subtracting the spectrum of 100 μL of 2-pyrrolidone per mL of toluene from the spectrum of 100 μL of 2-pyrrolidone per mL of toluene saturated with C_{60} (subtraction factor of 0.95).

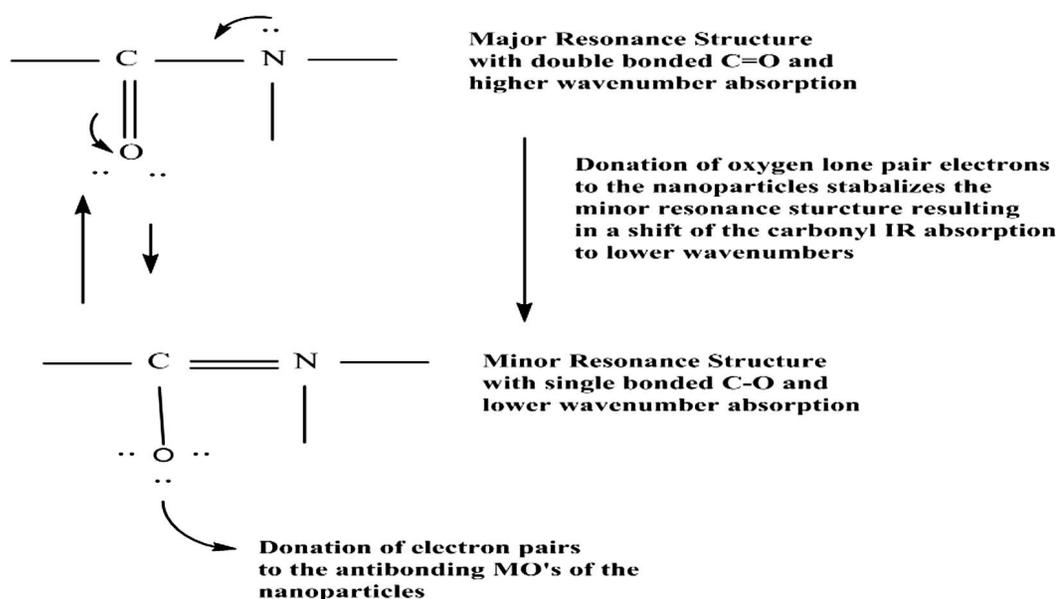


Figure 6.—A resonance structure model suggesting oxygen, lone pair donation to the nanoparticles (C_{60}) that shifts stability toward the resonance structure with the single bonded carbonyl and supports a shift of the carbonyl absorption to lower wavenumbers.

spectrum with a spectral absorption at 1690 cm^{-1} which is in good agreement with the 1687 cm^{-1} location of the absorption for the 2-pyrrolidone- C_{60} complex observed in the spectrum at the lower concentration levels. Again, intensities of the absorptions have been adjusted to clarify positioning along the wavenumber axis.

DISCUSSION AND CONCLUSIONS

The carbonyl infrared absorption in amide functionality is characterized by an absorption peak below 1700 cm^{-1} compared to “normal” carbonyl absorption peaks observed above 1700 cm^{-1} . The traditional explanation for the carbonyl-absorptions of amides being below 1700 cm^{-1} is that the lone-pair electrons on the nitrogen are delocalized into the amide carbon–nitrogen chemical bond generating a minor resonance structure with a carbon–oxygen single bond. This minor resonance structure adds single bond character to the carbonyl bond and contributes to the lower wavenumbers observed for the carbonyl infrared absorption in amide functionality (Avram & Mateescu 1970).

The observed shifts to lower wavenumbers of the carbonyl absorption in 2-pyrrolidone resulting from its interactions with C_{60} could

suggest that its interaction with the C_{60} occurs through the lone-pair electrons of the oxygen part of its amide structure. A number of studies indicate that C_{60} are good electron acceptors for molecular systems (Charvet et al. 2012; Schubert et al. 2013; Stranius et al. 2014). A recent study of the interactions of formamide with C_{60} and C_{70} showed a shift of the carbonyl absorption to higher wavenumbers suggesting an interaction through the nitrogen lone-pair electrons of the amide structure (Kirsch et al. 2015). The ring structure of the 2-pyrrolidone could make the nitrogen lone-pair electrons less accessible. If the lone pair electrons of the oxygen on the 2-pyrrolidone are donated into the antibonding molecular orbitals of the C_{60} (Feng et al. 2008), the resonance structure containing the single bonded CO is stabilized yielding a lower wavenumber infrared absorption of the carbonyl group. Figure 6 shows this oxygen lone-pair donation process, and describes its impact on the carbonyl absorption.

In summary, the infrared spectra collected in this study suggest that interactions occur between 2-pyrrolidone and C_{60} resulting from possible donation of the electron pairs from the oxygen part of its amide functionality into the antibond-

ing MO's of the C₆₀. At lower concentrations of 2-pyrrolidone (5 and 2 μL of 2-pyrrolidone per mL of toluene and toluene saturated with C₆₀), a significant shift of its amide carbonyl absorption (~1700 cm⁻¹ to ~1687 cm⁻¹) is observed supporting this kind of interaction. A very slight broadening of the carbonyl absorption of 2-pyrrolidone when treated with C₇₀ in toluene suggests possible interactions, but further studies are needed to clarify. Spectral subtraction of 2-pyrrolidone from 2-pyrrolidone treated with C₆₀ at higher concentration levels of 2-pyrrolidone reveals an absorption in good agreement (1690 cm⁻¹ vs 1687 cm⁻¹) with that observed in the spectrum of the 2-pyrrolidone treated with C₆₀ at lower concentration and also supports complex formation with C₆₀.

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OPEN-LOOP GEOTHERMAL DISCHARGE STREAM DESIGN AFFECTS THE PRECIPITATION OF CALCIUM

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ABSTRACT. Open-loop geothermal outflows have the potential for altering the chemical composition of local freshwater bodies by infusing them with spent ground water from geothermal heating/cooling systems (geothermal systems). A recently installed geothermal system and outflow on the campus of Taylor University has been investigated as a model for understanding the importance of certain stream design features in the removal of hardness ions. It has been previously reported that aerating streambed features preferentially removed iron but had little impact on the removal of dissolved calcium. This study describes the evaluation of dissolved calcium concentrations along the entirety of the stream (540 m from the geothermal system discharge to a nearby lake) by analyzing stream samples using atomic absorption spectroscopy. A slight gradual decrease in calcium concentration was observed along the 19 sampling sites with the exception of a 30% decrease between sites 12 and 13, the location of a large settling basin. When a lake-fed pool/fountain, designed to supplement the stream flow was turned on, only a 7% decrease in calcium concentration was observed. It was determined that while rapid moving water through rocky environments maximized the precipitation of iron; soil beds, slow moving water, and settling basins tended to maximize the precipitation of calcium. Additionally, a stream design that incorporates supplemental fresh water being added to the stream, often used for aesthetic reasons, caused a greater mobility of calcium ions and dramatically decreased its precipitation.

Keywords: Geothermal, stream design, calcium, deposition, atomic absorption spectroscopy

INTRODUCTION

In fall of 2012, Taylor University finished the construction of a new science building complete with many sustainable features. The features included wind turbines, solar panels, a heliostat, and an open-loop geothermal heating and cooling system, resulting in a Gold LEED (Leadership in Energy and Environmental Design) building certification. An outflow, which winds through campus to a nearby lake, was created to receive the discharge water from the geothermal system. Through numerous student projects and lab activities, the Department of Chemistry and Biochemistry has been studying the chemistry of Taylor Lake for years. Consequently, the recent construction has created a prime opportunity to study the impact of the stream design on the chemistry of the outflow and the lake.

Geothermal systems come in many forms and are one of the most widely used “green” heating

and cooling systems (Spitler 2005). In the United States, it is estimated that 50,000 systems are installed annually in homes, schools, and commercial buildings (Curtis et al. 2005). These systems offer clear environmental and economic benefits. Geothermal heating and cooling systems have higher initial costs than conventional systems but lower operating costs due to their efficiency (Self et al. 2013). If properly maintained, most systems retain their efficiency for 25 to 30 years (Bloomquist 2000). These systems produce half of the CO₂ emissions of conventional systems and therefore are expected to help cut global CO₂ emissions by over 6% (Curtis et al. 2005; Chua et al. 2010).

Several geothermal heating and cooling system designs have been created; each catering to a different geographical region with their varying soil and rock type as well as water availability and composition (Curtis et al. 2005). A geothermal system that uses ground or surface water to absorb and reject heat from the building and then discharge the used water either back into the ground or out to surface is considered an open-

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loop system. A system in which the heat exchange occurs when building system water is circulated through pipes buried in the ground or submerged in surface water is considered a closed-loop system. It is recommended that for small systems, such as the one featured here, discharge be released to surrounding surface water (Rafferty 2003).

The calcium concentration within surface water bodies, typically less than 15 mg/L (Chapman 1996), is primarily affected by substrate rock type, and their fluctuations can be due to changes in flow rates, litter fall (Webster & Patten 1979), and changes in turbidity – which facilitates nucleation (Mercer et al. 2005). When calcium levels are so high that intervention is required, typically in industrial wastewater situations, approaches utilizing cation exchange, chemical precipitation, and bioreactors have been employed (Hammes et al. 2003a, b; Nomanbhay & Palanisamy 2005). However, for geothermal outflows with only moderately elevated hardness levels, a carefully designed geothermal discharge stream may allow the discharge to naturally soften before it reaches its endpoint. If the stream design features do not effectively soften the discharge, then the remaining mineral content could cause a disruption in the ecosystem receiving it. Rocky streambeds, waterfalls, and high flow rates, among other features, encourage mineral deposition along the stream bottom through aeration. Previous investigation of the discharge outflow (Griffiths et al. 2013) demonstrated this to be true for iron. Atomic absorption spectroscopy (AAS) measurements along the first 90 m of the stream showed dramatic decrease in iron concentration that correlated strongly to the location of a rocky streambed and a small waterfall – both oxygenating features. However, the study found only a 16% gradual decrease in dissolved calcium concentration over that same distance suggesting that aeration does not significantly facilitate the precipitation of calcium. It is, therefore, believed that any decrease in calcium concentration occurs anaerobically, through precipitation with anions which are insoluble with calcium (e.g., carbonate or bicarbonate) found along the streambed. This paper outlines a study of the entire length of the stream, 540 m, in the hopes of finding a stream feature that preferentially facilitates a decrease in calcium concentrations more readily. Additionally, a new design feature was added to the stream between the earlier study and the present one that needed to be

investigated. A fresh water lake-fed pool was connected 30 m above the source of the geothermal discharge to supplement the stream flow.

METHODS

Description of the geothermal discharge stream.—The stream is 540 m long and runs downhill from an elevation of about 284 m at its source to an elevation of about 271 m where it enters Taylor Lake (Fig. 1). This gradient supports fast flowing water along the majority of the stream. The streambed is narrow from its source to the lake, never widening more than 2 m except for in several settling basins. Before reaching the lake, the water travels through four settling basins where the flow is slowed. The basins, located at 90 m, 330 m, 390 m, and 420 m from the source, have approximate diameters of 5 m, 10 m, 25 m, and 30 m respectively. Upstream from the first settling basin, the streambed is composed of mostly rocks, but downstream from the basin the streambed becomes mostly soil, sediment, and plant matter. Additionally, the stream flow is supplemented by fresh water pumped from the lake to a small pool (and fountain) just above the geothermal discharge. When ambient temperatures are nearer the desired building temperature and the geothermal discharge is consequently less, the flow from this lake-fed pool, located 30 m above the source of the geothermal outflow, is turned on allowing fresh lake water to mix with the outflow water to supplement the stream flow. Under more extreme temperature conditions, and when the geothermal discharge is consequently higher, the lake-fed pool flow is sometimes off.

Sampling.—Samples were collected in a similar fashion as in the previous study (Griffiths et al. 2013). Each of the sample sites was marked with a flag, photographed, and denoted on a map of the stream so that they could be revisited. At each site, 300 mL samples were collected in triplicate. Sample sites spanned the length of the stream from 0 to 540 m from the source, in 30 m increments. As to not disturb the streambed and skew results, the samples were collected initially from the sampling site farthest downstream, near the lake, and then upstream toward the source. This sampling procedure was followed once while the lake-fed pool flow was off and once again while it was on. Samples were preserved with dilute hydrochloric acid (0.1 M) to

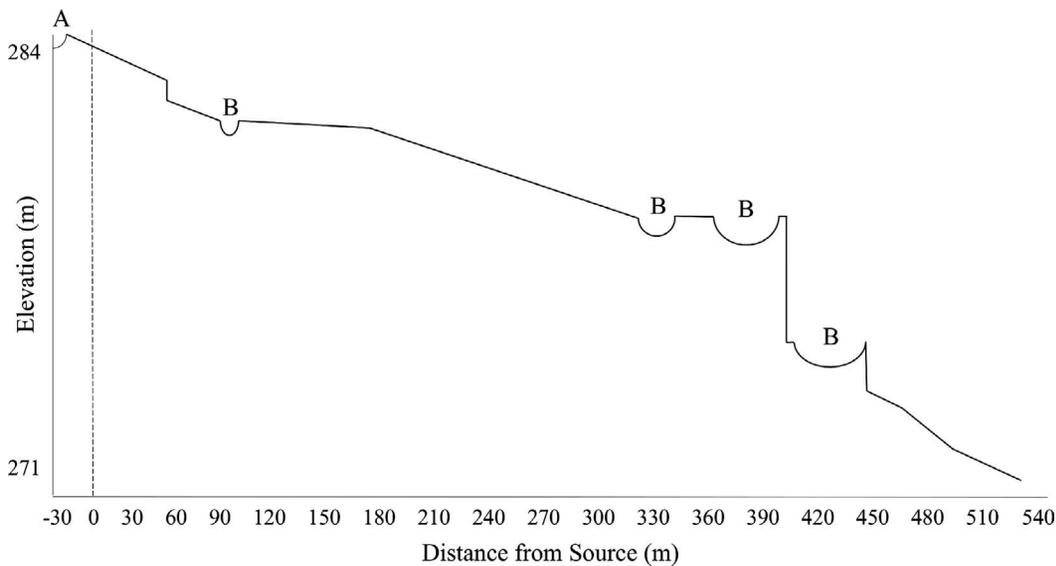


Figure 1.—Elevation change from geothermal discharge source (dashed line) to lake. The lake-fed pool location (A) at -30 m above the source and settling basins (B) at 90 m, 330 m, 390 m, and 420 m are also shown.

minimize precipitation and to avoid the potential interference during analysis (also added to standards) (EPA method 215.1) (Chapman 1996).

Analysis.—Dissolved calcium concentrations were evaluated using an iCE 3000 Series AAS (Thermo Scientific) equipped with an ASX-520 autosampler (CETAC) following the EPA method 215.1. Absorbance was monitored at 422.7 nm using an air/acetylene flame with a fuel flow rate of 1.4 L/min. One mL of HCl was added to samples to reduce precipitation prior to analysis. The addition of HCl also has the benefit of reducing the interference effects from phosphate and sulfate, the two most likely interferences for calcium in this study. For analysis, 10 mL of each sample was transferred to an autosampler tube and labeled based on its sample location. Calibration of the instrument was done using 1000 mg/L standard stock calcium solution from Fisher Scientific. Since calcium concentrations were in the 20 to 35 mg/L range in the past survey of the stream, 1 mg/L, 5 mg/L, 15 mg/L, 30 mg/L, and 40 mg/L standards were prepared for quantitation. Each triplicate sample was analyzed in triplicate to evaluate instrumental and sampling precision. Values reported in this paper are averages of all nine measurements corresponding to each location and each sampling event, so that one

value and standard deviation is given to represent each site. The time intensive method of standard addition, which is capable of accommodating potential matrix effects resulting from the differences among sample solutions and standard solutions (Ito & Tsukada 2002), was not employed because of the large number of samples analyzed during this study. The addition of HCl should help to minimize matrix effects, but the concern should not be ignored. The data are presented as absolute concentrations, but may be more cautiously considered as relative values. It is the relative values and not the absolute values that are critical to the present investigation. The statistical analysis of the data was performed using the OriginPro 9.0 software package. Two-way ANOVA (analysis of variance) was performed to evaluate the statistical interactions between distance from outflow source and pool flow status (on or off).

RESULTS AND DISCUSSION

Previous investigation into the geothermal discharge outflow showed a dramatic decrease in iron concentration but only minimal decrease in calcium concentration along the initial aerating portion of the stream, 0–90 m (Griffiths et al. 2013). Consequently, the present study involved analyzing dissolved calcium concentration in

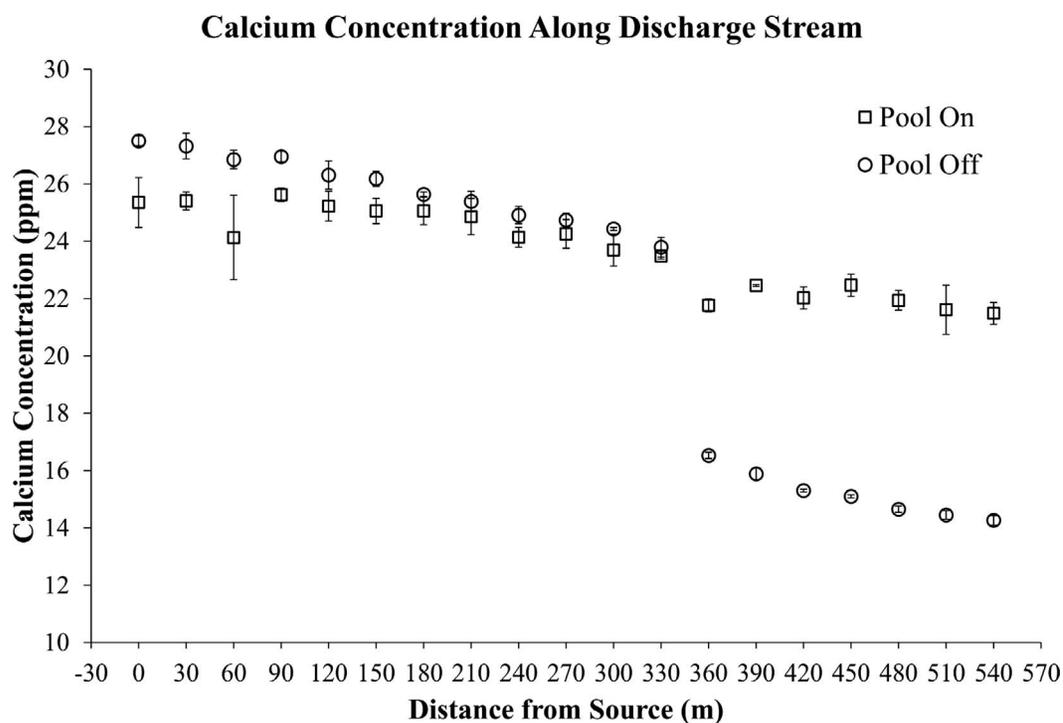


Figure 2.—Dissolved calcium concentration along the stream when the lake-fed pool flow was on (squares) and off (circles). Error bars represent the standard deviation among the nine analyses for each site. The relative standard deviation of the repeated measurements was determined to be approximately 0.1%. Therefore, the primary contribution to the sample site standard deviations (error bars) is due to the variation among samples.

samples collected along the entire stream, once while the lake-fed pool flow was on, and once while it was off. Prior to this study, other parameters that may influence the behavior of dissolved calcium had been investigated (unpublished) to a limited extent at a few locations and, although not directly applicable, the typical values observed provide a general description of the water system (pH \sim 7.1, temperature \sim 22.3°C, alkalinity \sim 16 mg/L CaCO₃ (EPA method 310.1), sulfate \sim 320 mg/L (EPA method 375.4), conductivity \sim 900 μ S/cm (EPA method 120.1). An ANOVA test determined that, at the 0.05 level, the rate of the decrease in calcium concentration was statistically different between when the lake-fed pool flow was on and when it was off.

Lake-fed pool flow off.—When the outflow stream carries only geothermal discharge (i.e., without dilution by lake water) the calcium concentration decreased by about 48% from source to lake. The data from the sample sites along the stream were grouped into two segments (Fig. 2). These segments extended

from the source (0 m) to 330 m and from 360 m to 540 m. In both segments, there was an obvious decrease in calcium concentration as the distance from the outflow source increased (Segment 1: linear $R^2 = 0.985$, with a total drop in concentration of -3.71 mg/L; Segment 2: linear $R^2 = 0.952$, with a total drop in concentration of -2.27 mg/L). The two segments are divided by a 7.26 mg/L decrease in calcium concentration (30%) between 330 m and 360 m along the stream. This transitional region of the stream (330 m to 360 m) corresponds to the location of a settling basin (Fig. 1) that, consequently, is suspected to aid in the deposition of calcium to the stream bed. Given this rapid decrease in calcium concentration, it is not surprising that the population means of Segment 1 and Segment 2 were significantly different at a 0.05 level according to an ANOVA test.

Lake-fed pool flow on.—When the pool flow was turned on, lake water is added to the discharge stream, presumably resulting in

higher flow rates and dilution of dissolved ions. The calcium concentration in the lake and pool were determined to be similar, ~ 13 mg/L, and notably lower than at the discharge source, ~ 25 mg/L. At the discharge source the dissolved calcium concentration was lower than the “pool flow off” value by 2.15 mg/L, presumably as a result of dilution (Fig. 2). Although the population means of the two segments (0 m – 330 m and 360 m – 540 m) were still determined to be significantly different at a 0.05 level, the dramatic drop in concentration between the two segments of the stream, as noted when the pool was on, did not exist (Fig. 2). For pool-on data, there was a clear decrease in calcium concentration as the distance from the stream source increased (linear $R^2 = 0.859$), but only a total drop in concentration of -3.86 mg/L. When the lake-fed pool flow was on, calcium concentration decreased by only 15% from source to outflow. Additionally, a two-way ANOVA analysis found a strong statistical interaction between the sample site location and the pool flow status. In other words, whether the pool was on or off had a significant impact on calcium concentration. Consequently, despite the potential dilution caused by the surface water being added to the stream (seen early in the stream), the calcium concentration was dramatically higher in the water flowing into the lake when the lake-fed pool is on (Fig. 2).

In summary, the decrease in calcium concentration likely occurs through precipitation as insoluble salts with anions within the water or streambed rather than through oxidation. This may be a slower process. When the lake-fed pool flow was on, calcium concentration decreases by only 15% between the source and the lake. However, when the lake-fed pool flow was off, calcium concentration decreases by 48% between the source and the lake. The addition of the lake-fed pool water to the stream causes an increase in flow rate. The higher flow rate leaves less time for the precipitation process to occur in the settling basins. This explanation accounts for the difference in percent calcium concentration decrease when the lake-fed pool flow was on as opposed to when it is off. In addition, when the pool flow is off, 98% of the decrease in dissolved calcium occurs beyond 90 m where the streambed design changes dramatically, suggesting that stream design features occurring later in the stream dissipate calcium more effectively.

The design of this stream is important for lessening the impact that high mineral content geothermal discharge has on the downstream lake ecosystem. It must contain features that naturally soften the water on its way to the lake. In this study, settling basins may be the design feature that dissipate calcium concentrations from the stream, just as the rocky streambed and small waterfall did for iron in the previous study. Interestingly, the strategies that ought to be employed to remove two of the most abundant hardness ions are somewhat opposed to one another. Consequently, the ideal stream design must be long enough to incorporate both an aerating environment for iron and a settling environment for calcium.

Unfortunately, with the present data it is impossible to determine if the rapid decrease in calcium concentration within the third settling basin is primarily driven by the chemistry of the basin sediment through cation-exchange or precipitation or driven by the low flowrate within the basin. The geothermal outflow design generates a variable flow within the stream throughout the day and between days, as a function of the difference between ambient and building temperatures, which make it difficult to exhaustively study all sites within the same sampling conditions. However, now that the key location has been identified, a subsequent comprehensive analysis of this third settling basin can be performed at various flowrates.

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THE INFLUENCE OF DAMS ON FISH COMMUNITIES AND ASSOCIATED HABITAT IN THE ST. JOSEPH RIVER WATERSHED, INDIANA

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ABSTRACT. We evaluated the influence of dams on fish communities and river habitat using data gathered on the St. Joseph River and Elkhart River in north-central Indiana from 1998 to 2016. Comparisons of Index of Biotic Integrity (IBI) and Qualitative Habitat Evaluation Index (QHEI) values above and below dams on these two rivers generally indicate moderate impairment to fish communities and habitat upstream of dams and in impounded areas of both rivers. Across all sites a comparison of mean index scores for the IBI, QHEI, and metrics within both indices indicate significantly higher quality fish communities and habitats outside of impounded areas on the St. Joseph River.

Keywords: Dams, fish community, IBI, QHEI, Saint Joseph River

INTRODUCTION

The St. Joseph River in north-central Indiana, a tributary to Lake Michigan, drains 12,134 square kilometers. Along its 338 kilometer course from Hillsdale County, Michigan to its confluence at Lake Michigan, are 17 dams with an estimated 190 additional dams within the watershed as a whole (Wesley & Duffy 1999). In the Indiana section of the river, four dams remain; two active hydropower dams and two former hydropower dams.

Dams are known to have detrimental effects on the ecology of a river. They alter the physical conditions of a stream. Dams are generally constructed in high gradient areas where fish spawning habitat is often located. Fish spawning migrations and the associated movement of native mussel species upstream are blocked because of dams (Eads et al. 2015). Fish communities are separated and populations become isolated, thus limiting genetic dispersal and diversity (Zhao et al. 2016). Lentic conditions upstream of a dam reduce flow, causing a buildup of fine sediment and a change in river habitat. Downstream of a dam, streams are starved of sediment resulting in loss of habitat and negative impacts to stream banks and floodplains (Marren et al. 2014). Chemical changes also occur, often resulting in low dissolved oxygen concentrations above dams,

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limiting the streams ability to support aquatic life (Santucci et al. 2005).

Biological community monitoring is generally performed to evaluate the health of a stream due to impacts from pollution or immediate habitat destruction (Karr et al. 1986). In an effort to understand effects of dams on St. Joseph River Watershed fish communities, we compared fish community structure and habitat conditions above and below dams on the St. Joseph and Elkhart Rivers in Elkhart and St. Joseph Counties, Indiana.

METHODS

Fish community data collected from the period 1998 to 2016 by the City of Elkhart Public Works and Utilities during routine fish surveys in the Saint Joseph River Watershed were analyzed. These data were from 23 stations on the St. Joseph River and nine stations on the Elkhart River. Fish communities were sampled annually at each station for three consecutive years, with follow up sampling events on a three year rotational basis. All stations were sampled twice during spring and summer months in a given year to account for seasonal variation in fish community assemblages. Fish were collected using a pulsed-DC boat mounted electrofishing unit, with a current output range of 12 to 16 amps, a voltage range of zero to 500 volts, and a pulse frequency setting of 120 pulses per second. All available

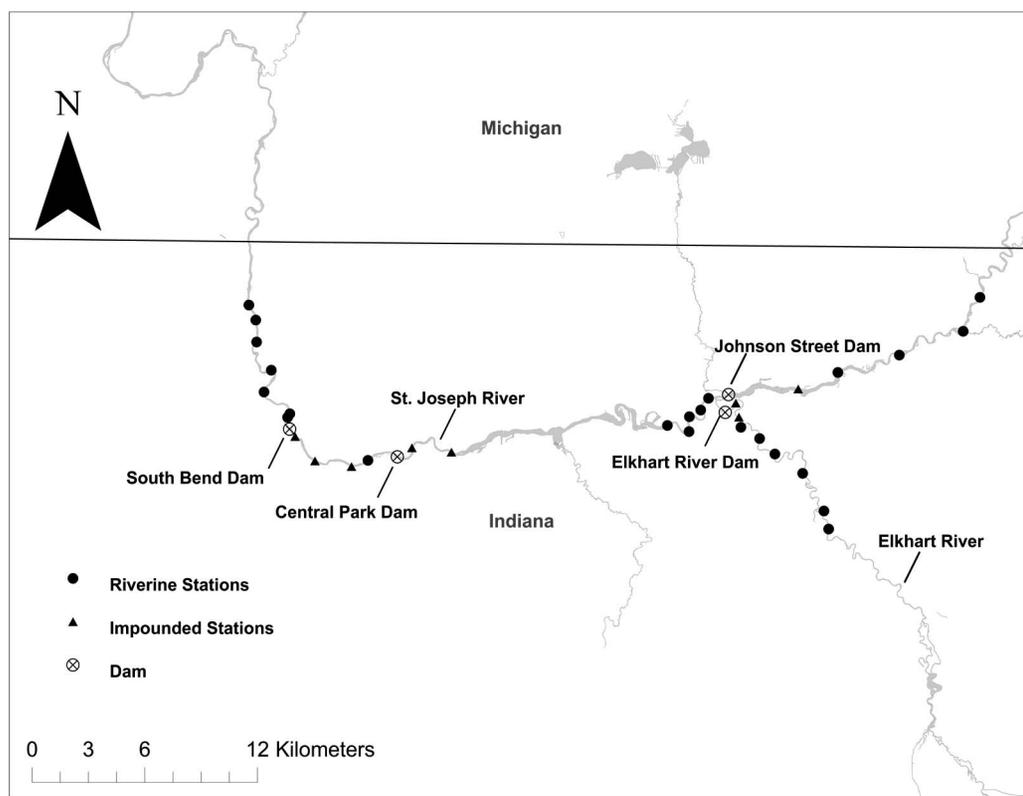


Figure 1.—A Map of the St. Joseph and Elkhart Rivers in Elkhart and St. Joseph Counties, showing dam locations and stations sampled from 1998–2016.

habitat types within a 500 m section of river were sampled at each station.

Fish communities were evaluated using the Index of Biotic Integrity (IBI) (Karr 1981) calibrated for use in warm water streams of the Northern Indiana Till Plain (Simon 1997). Habitat was evaluated using the Qualitative Habitat Evaluation Index (QHEI) (Rankin 1989) following fish community surveys. Data analysis focused on sampling stations above and below three major dams on the St. Joseph River (Fig. 1): the Johnson Street Dam in Elkhart, the Central Park Dam in Mishawaka, and the South Bend Dam in South Bend. A general evaluation of IBI scores from stations above one low head dam on the Elkhart River in Elkhart was also performed (Fig. 1).

General comparisons of riverine versus impounded stations were made by averaging IBI and QHEI scores from all sampling events at a given station. Mean individual metric scores from the IBI and QHEI also were calculated. For the QHEI, individual metrics included substrate,

cover, channel, riparian, riffle, and pool metrics. For the IBI, the individual metrics included number of species (species richness), total fish abundance, and % of lithophilic spawner metrics. Simple lithophilic spawners are species that require clean gravel or cobble for reproduction habitat. Statistical analyses used SigmaPlot® software, Systat Software, Inc., San Jose, California, USA. Two sample T-tests were used to compare means of variables with the exception of channel and riparian metrics of the QHEI, and the total abundance of fish. The Mann-Whitney, non-parametric rank sum test (U-test) was used to compare the channel, riparian, and total fish abundance metrics given that variances between groups were not equal. To account for possible type I errors with multiple tests, significance levels were adjusted to 0.005 using the Bonferroni correction.

RESULTS

South Bend Dam.—Mean IBI scores were lower at stations in the impoundment of the

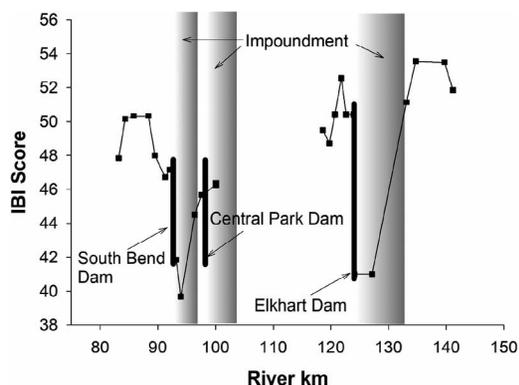


Figure 2.—Longitudinal depiction of mean IBI scores for stations along the St. Joseph River. Black dots represent sampling stations. Impounded areas are shaded. IBI scores may range from 12 to 60. All mean values are above 36 indicating that no stations are considered “impaired” (Simon 1997).

South Bend Dam than those downstream of the South Bend Dam (Fig. 2). A mean IBI score of 41 occurred at the station directly above the South Bend dam. At the next station, approximately 0.8 km upstream, the mean IBI score was lower at 39. Scores increased upstream with respective mean IBI scores of 44.5 and 45.7 at river km stations 96.4 and 97.5. At the station immediately below the South Bend Dam, the mean IBI score was 47.1. At the next station, approximately 0.8 km downstream, the mean IBI score was slightly lower at 46.7. However, the mean IBI scores increased to 50 farther downstream of the dam.

Central Park Dam.—IBI scores above and below the Central Park Dam were relatively similar. The station downstream of the dam at river km 97.5 had a mean IBI score of 45.7, while the station above the dam had an IBI score of 46.7. The IBI score dropped slightly at the second most upstream station above the dam to 46.2. The station below the Central Park Dam is located just on the upstream edge of the impoundment caused by the South Bend Dam.

Johnson Street Dam.—Mean IBI scores around the Johnson Street Dam resulted in a similar pattern as the South Bend Dam (Fig. 2). The mean IBI score at the station immediately above the dam was 41, lower than the score of 50 at the station immediately below the dam. At the next station above the Johnson Street Dam, which is upstream of the impoundment,

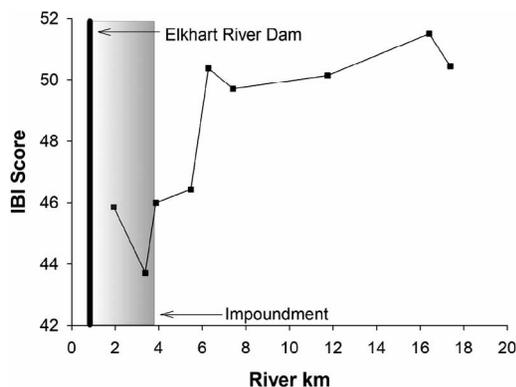


Figure 3.—Longitudinal depiction of mean IBI scores for stations along the Elkhart River. Black dots represent sampling stations. Impounded areas are shaded. IBI scores may range from 12 to 60. All mean values are above 36 indicating that no stations are considered “impaired” (Simon 1997).

the mean IBI score was higher at 51. Mean IBI scores increased with upstream distance from the Johnson Street impoundment. Below the dam, the mean IBI score increased to 52.6 at river km 122, but decreased below 50 farther downstream.

Elkhart River Dam.—The Elkhart River dam is approximately 0.8 km upstream of the St. Joseph River confluence, thus no sampling stations were located below the dam. However, upstream of the dam, our results had similar IBI trends as above the South Bend Dam and the Johnson Street Dam (Fig. 3). At the station immediately upstream of the dam, the mean IBI score was 46, dropped slightly to 44 at the next most upstream station, and increased to 46 at the third station upstream of the dam. This third station was located just upstream of the impoundment. Further upstream, with distance from the impoundment, the mean IBI scores were nearly 50.

Comparison of Indices.—A general comparison of several parameters, including the number of fish species, IBI scores, QHEI scores, and individual QHEI metrics, revealed significant differences in fish community and habitat integrity in impounded versus riverine sections of the St. Joseph River (Table 1, Fig. 4). Values were significantly lower for most parameters in impounded sections of the river with the exception of the riparian, cover, and pool metrics in the QHEI and total fish abundance (Table 1, Fig. 5).

Table 1.—Fish community and habitat parameters for stations above and below dams on the St. Joseph River (1998 to 2016). Mean values are presented for all parameters except for channel and riffle/run metrics of the QHEI and for total abundance, which are presented as median values. Significance was determined at *P* values less than 0.005.

	Impounded stations	(n=6) Std. dev.	SEM	Riverine stations	(n=17) Std. dev.	SEM	<i>t</i> or <i>u</i> <i>value</i>	<i>P</i> value
# of Species	21.30	1.59	0.65	25.60	2.75	0.67	t 4.343	0.002
Total Abundance	508	(median)		461	(median)		u 33.00	0.220
% Simple Lithophils	11.75	5.51	2.25	27.17	8.88	2.15	t 3.959	<0.001
IBI	43.30	2.90	1.20	49.90	2.30	0.56	t 5.603	<0.001
QHEI	61.10	3.30	1.40	74.30	5.10	1.20	t 5.876	<0.001
QHEI Metrics								
Substrate	11.12	1.99	0.81	14.81	1.50	0.36	t 3.691	<0.001
Cover	12.24	1.20	0.50	13.44	1.20	0.29	t 0.209	0.049
Channel	12.40	(median)		15.15	(median)		u 3.000	<0.001
Riparian	5.64	0.89	0.36	6.65	1.35	0.30	t 1.707	0.103
Riffle/Run	0.98	(median)		4.71	(median)		u 0.000	<0.001
Pool/Glide	8.89	0.29	0.12	9.58	0.50	0.13	t 2.990	0.007

The mean species richness at stations in the impounded areas of the St. Joseph River was 21.3. At riverine stations, the mean species richness was 25.6. IBI scores averaged 43.3 at impounded stations versus 49.9 at riverine stations. Median fish abundance at riverine stations (508) was higher than impounded stations (461), however, the difference was not statistically significant (Table 1). Mean total fish abundance was highly variable among the riverine stations ranging from 354 to 952. Mean total fish abundance at impounded stations was not as variable, ranging from 327 to 532. Several impounded stations had higher mean total abundance than several riverine

stations. The mean % of simple lithophils collected at each station was significantly higher at riverine stations (27.2) than at impounded stations (11.8) (Table 1).

Average QHEI scores were very low at impounded stations (61.1) relative to riverine stations (74.3) (Table 1). The highest variation in individual QHEI metrics was for the riffle metric (Table 1). For impounded stations, the mean riffle QHEI score was 0.98, while the riverine stations had a mean score of 4.71 (Table 1). Mean riparian, cover, and pool QHEI metric scores were similar for the impounded stations and riverine stations (Table 1).

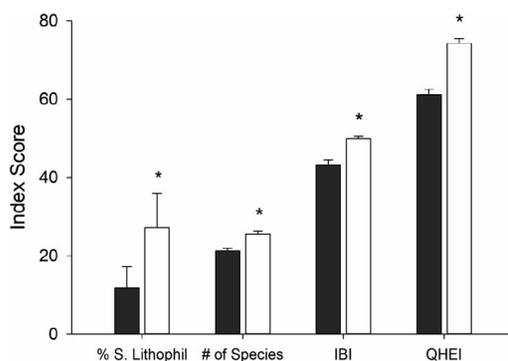


Figure 4.—Percent simple lithophils, number of species, IBI scores, and QHEI scores for stations on the St. Joseph River. Dark bars represent values for stations in impounded areas, while white bars represent values for stations in riverine areas. Data are means \pm SEM. Asterisks denote a significantly higher value.

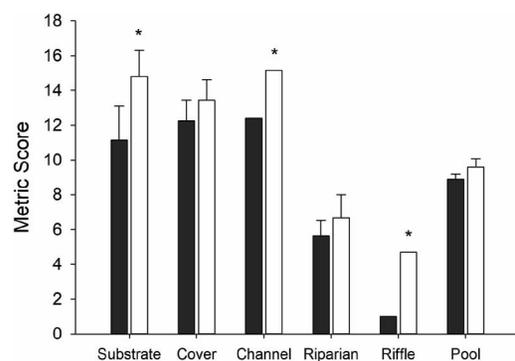


Figure 5.—QHEI metric scores for stations on the St. Joseph River. Dark bars represent mean values for stations in impounded areas, while white bars represent mean values for stations in riverine areas. Channel and riffle metric data are medians. Substrate, cover, riparian and pool data are means \pm SEM. Asterisks denote a significantly higher value.

DISCUSSION

Species richness is a parameter that demonstrates the negative effects of dams (Agostinho et al. 2008; Gardner et al. 2011; Liermann et al. 2012; Yun-zhi et al. 2013; Magilligan et al. 2016). Similar to these studies, we found that mean species richness was significantly higher in riverine sections of the St. Joseph River. Our QHEI scores and QHEI metrics suggest that fish community integrity is linked to habitat variation, which is limited in impounded sections of the St. Joseph River. While poor habitat may contribute to decreased species richness above a dam, the presence of a dam as a barrier can inflate species diversity and abundance on the downstream side of the dam (Dodd et al. 2003). Gardner et al. (2011) found that fish species richness and abundance decreased significantly at the downstream site following dam removal on the Sedgeunkedunk in Maine, USA. Catalano et al. (2007) reported similar findings on the Bamboo River, Wisconsin, USA following dam removals, but also found that species richness recovered to pre-removal levels after two years at two dam removal sites. The presence of dams on the St. Joseph River undoubtedly interferes with the natural migration of fish species and may inflate species richness below the dams. However, the lack of suitable habitat above the dam also limits species richness. We observed a reduced number of species with limited habitat variability. For example, substrate scores (QHEI metric) were significantly lower in impounded areas, corresponding with a significantly lower abundance of simple lithophilic spawners that require high quality coarse substrate for reproduction.

Our study found that fish abundance did not differ significantly in impounded versus riverine sections of the St. Joseph River. These findings are not consistent with other studies. Santucci et al. (2005) found significantly higher fish abundance in free flowing sections of the Fox River, Illinois, than in impounded areas. Yun-zhi et al. (2013) also reported that fish abundance was lower in impoundments caused by lowhead dams. We found that mean total abundance of fish in riverine stations varied significantly among riverine stations with the lowest of 354 at a station below the South Bend dam and the highest at 952 below the Johnson Street Dam. Beyond the presence of dams, it is likely that other factors, such as pollutants from point and non-point sources, urban habitat disturbance, and unsteady

stream discharge may have influenced the fish communities in the St. Joseph River (Deegan 2012). Baldwin et al. (2016) found several organic compounds in the St. Joseph River at levels with the potential to cause adverse effects on aquatic organisms.

Several impounded stations also had higher mean total abundance than several riverine stations; the highest mean total abundance for an impounded station was 532, which was 178 more than mean total abundance at a riverine station below the South Bend Dam. Agostinho et al. (2008) suggested that the upper third of a reservoir can have high relative abundance due to an increase in primary productivity in the transitional area between the impoundment and free flowing river. We speculate that impoundment to riverine transitional areas may have inflated IBI scores and IBI metrics given the presence of both lentic and lotic factors and the associated increase in habitat diversity.

Mean IBI and QHEI scores demonstrated significantly lower fish community integrity and habitat in impounded sections of the St. Joseph River. IBI scores on the Elkhart River also increased upstream of the Elkhart River dam impoundment. Santucci et al. (2005) recorded significantly higher IBI and QHEI scores in fish communities in free flowing sections of the Fox River, Illinois, USA than those in impounded sections. In the riverine sections of the St. Joseph River, substrate diversity and quality was superior, contributing to higher QHEI scores. The channel metric of the QHEI was also significantly lower in the impounded sections of the St. Joseph River, a result of floodplains being underwater, decreased sinuosity, and a decrease in the quality and quantity of pool, riffle, and run complexes.

Mean IBI scores in the Mishawaka section of the St. Joseph River, where riverine habitat is most limited, were inconsistent with our findings for the others sections of the St. Joseph River. At the Central Park Dam, the mean IBI score above the dam is slightly higher than the score below the dam. Within 2 km downstream of the Central Park dam, the river transitions into the impoundment caused by the South Bend Dam. The Central Park Dam also creates an impoundment that is approximately 5 km long and extends upstream to another St. Joseph River dam. Rolls et al. (2013) suggested that there is a spatial complexity on rivers with several dams, with interacting effects from fragmentation and altered habitat. With impoundment influences upstream and down-

stream of the Central Park Dam, it is likely that there are negative interacting effects of multiple dams on the fish communities present.

The riparian QHEI metric was one QHEI parameter that did not differ significantly between riverine and impounded stations. The predominant land use surrounding the St. Joseph River in northern Indiana is urban, with heavy urban land use in the downtown areas of South Bend, Mishawaka, and Elkhart, and residential land use on the river segments between these cities. The three main St. Joseph River dams in this study are located in the downtown areas of each city with significant urban impacts to the riparian zones on the upstream and downstream side of each dam.

Given the above dam versus below dam data, the negative influence of dams on the St. Joseph River are apparent. Within the context of the South Bend-Mishawaka-Elkhart municipal area, however, we acknowledge additional influences beyond the presence of dams. Other major influences include non-point sources of pollution, urban habitat disturbance, and unsteady stream discharge. The spatial complexity associated with the presence of multiple dams likely has interacting effects on fish communities in the St. Joseph River, particularly in the Mishawaka section of the St. Joseph River.

ACKNOWLEDGMENTS

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FOREST PLANT COMPOSITION IN PATCHES OF VARYING AGES IN AN INTENSIVELY FARMED LANDSCAPE

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ABSTRACT. The stability of plant community diversity in post-agricultural forests is an important ecological question throughout the Midwestern United States. Since forests in this region are often islands in a matrix of annual croplands, a better understanding of the persistence of native plant diversity and the potential for incursions of exotic invasive species is crucial for maintaining biodiversity in these landscapes. This study examined plant communities in several post-agricultural forests in northern Indiana that varied by age since agricultural abandonment, and investigated differences in invasive species presence across several canopy layers (canopy, midstory, understory). Vegetation was sampled in circular plots across three distinct forest parcels. In addition to forest age, several other conditions hypothesized to affect plant diversity, such as tree canopy cover, litter depth, underlying soil type, and adjacent land uses, were also measured. Overall herbaceous diversity and integrity, as measured by the Floristic Quality Index (FQI), were relatively high in these forests. Invasive species, e.g., *Alliaria petiolata* and *Rosa multiflora*, were also generally uncommon in these forests, even in areas affected by local disturbances such as deer trails, trash dumps, and canopy gaps, although older plots generally had lower invasive presence than younger ones. This study highlights the importance of maintaining existing older-growth forests in this landscape and avoiding disturbances to the interior of such forests to minimize further expansion of invasive species.

Keywords: plant invasion, land use legacies, edge effects, herbaceous diversity, FQI

INTRODUCTION

A diversity of variables influences the composition of contemporary plant communities and their vulnerability to invasive species. But particularly in landscapes marked by intensive human disturbance, the proximity of native habitats to human-dominated land uses can be a key determinant of both biotic and abiotic conditions in the habitat (Vilà & Ibáñez 2011; With 2002). Further, previous land use often leaves a persistent ecological legacy, even as those portions of the landscape return to a less-disturbed state (Medley & Krisko 2007; Flory & Clay 2009; Kuhman et al. 2010). Our ability to predict areas of natural habitat with high native biodiversity, and contrarily low invasive presence, depends on our understanding of the current and prior arrangement of land uses in an area.

Even in regions with substantial mature forest cover, ongoing agricultural and urban land uses contribute to fragmentation of those habitats,

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with concomitant increases in edge effects (Gavier-Pizarro et al. 2010; Harper et al. 2005; McDonald & Urban 2006; Tomasetto et al. 2013). A previous study in old-growth forest islands in rural central Indiana found that alien species richness and frequency dropped sharply inward from forest edges, and that forest interiors were relatively free of aliens (Brothers & Spingarn 1992). The primary factors believed to limit invasion were low light and disturbance levels in interiors, combined with limited dispersal of invasive propagules into the interior. In light of the many changes on this landscape since the work of Brothers & Spingarn (including increasing landscape saturation by invasive propagules, higher populations of herbivores, and altered climate regimes), part of the motivation for this study was to ascertain whether their patterns still held in this region. Gignac & Dale (2007) also observed that habitat fragment size and shape affected native and alien plant species richness and abundance in agro-environments, with native abundance highest in larger, less-disturbed patches, and overall richness highest in edges. Invasion processes are also strongly influenced by distur-

bances, including the passages caused by roads and other transportation routes, such as trails. Increased soil moisture, soil disturbance, nutrient runoff, sun exposure, and temperature along the road edges in the upper Midwest promote plant invasion and growth of exotic species such as *Elaeagnus umbellata* Thunb., *Ligustrum obtusifolium* Siebold & Zucc., and *Lonicera maackii* (Rupr.) Herder (Flory & Clay 2009; Watkins et al. 2003).

Since the remaining forest fragments in agricultural landscapes are likely under high invasion pressure, it is crucial that efficient methods for identifying and controlling invasions in these habitats be developed. This study was designed to provide a limited snapshot of current herbaceous diversity and invasion extent in mixed-hardwood forests of the agricultural upper Midwest. Specifically, we questioned whether the abundance of invasive plant species varied significantly with the age of the forest fragments, and how it was related to other patterns of plant diversity and environmental variables.

METHODS

Study location.—The study was conducted at the Merry Lea Environmental Learning Center (MLELC), in Wolf Lake, Indiana (approx. 41.3180° N, 85.5140° W), in the summer of 2013 (June – July). MLELC is an area consisting of diverse ecosystems and landscapes, including prairies, forests, marshes, wetland, and savanna. This high landscape heterogeneity means that many of the habitats have high perimeter: area ratios, although the natural habitat patches are still generally much larger than in the surrounding agricultural landscape. The 481 ha preserve is not only a host for much of the region's biodiversity but also incorporates many ongoing restoration projects, including recreation of wetland habitats by altering hydrology and reestablishing prairies through controlled burning. The landscape bears numerous prominent features that are remnants of the last glaciation, including eskers, kames, and kettle ponds.

Plot selection and structure.—Sampled plots were in three forests within MLELC, designated Thomas, Wysong, and Byer after the original landowners (Fig. 1). These forests were chosen to represent as much of the range of upland forest types present on the MELEC property as possible. Each one has complete canopy cover, but the tree species richness

varies across the three forests, as does the specific land use history. None are primary, old-growth forest, although Byer (formerly managed woodlot) has been closed canopy since at least the 1930's (decade of the earliest aerial photographs of the region), while Thomas and Wysong (formerly pastures and crop fields; Fig. 2) were released from agriculture 50–60 years ago.

Each circular plot was 78.5 m² in area (5 m radii) and 50 m from neighboring plots (measured from center-to-center). To assure sampling covered the breadth of sub-habitats present in each forest, plots were laid out along transects radiating out from the approximate center of the forest, as spokes from a hub. The factors assessed for each plot are detailed in Table 1. In addition to field data, each plot's coordinates were collected using GPS, and geographic information systems (ArcMap, ESRI) were then used to calculate distances to the nearest non-forested land cover, as well as plot slope, plot aspect (both from the State of Indiana Geographic Information Office), and USDA-SSURGO (Soil Survey Geographic Database) soil series.

Thomas: This woodlot was relatively open in the understory (Fig. 2A) with housing, old fields, and active farm land surrounding the edges. Dominant tree species in these plots included *Acer saccharum* Marshall, *Ulmus americana* L., *Quercus alba* L., *Q. velutina* Lam., *Q. rubra* L. and *Ostrya virginiana* (Mill.) K. Koch. Shrubs were generally sparse in this woodlot but included scattered populations of *Xanthoxylum americanum* Mill. and the invasive species *Elaeagnus umbellata* and *Lonicera maackii*. The range of litter depth in Thomas was 1.8 mm to 34 mm (mean 16.6 mm). The average canopy cover was 94%.

Wysong: The edges and some of the interior featured dense thickets of shrubs, including natives such as *X. americanum*, *Rubus occidentalis* L., *Lindera benzoin* L., and occasional clusters of invasives including *Rosa multiflora* Thunb., and *E. umbellata*. The tree community was quite diverse, but dominant species included *Q. alba*, *Ulmus rubra* Muhl., *Juglans nigra* L., *A. saccharum*, and *Crataegus* spp. (Fig. 2B). Along one of the transect lines the distance between plots was larger due to interspersed vernal pools that were not sampled because of their dramatically different hydrology and edaphic conditions. The range of litter depth

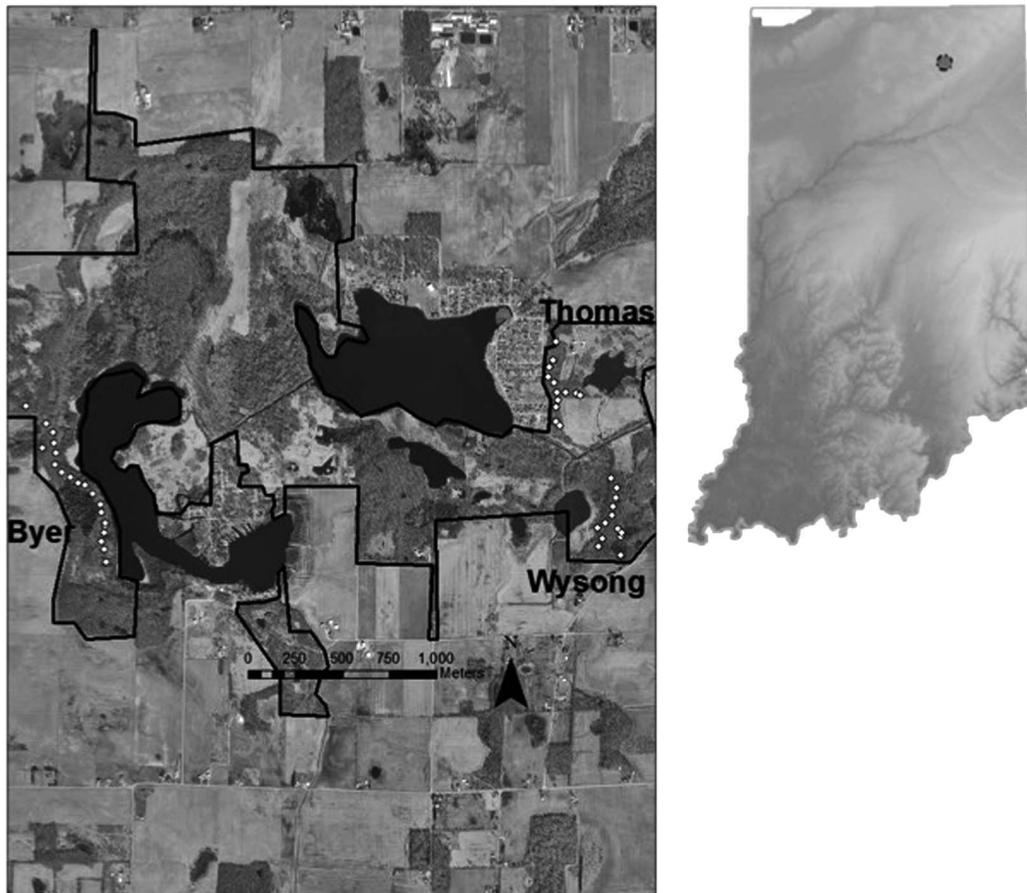


Figure 1.—Aerial photograph of the Merry Lea Environmental Learning Center (MLELC; border shown with solid dark line), with the Thomas, Wysong, and Byer woodlots labeled. Plot locations in each woodlot are denoted by gray dots. Highlighted dot on the Indiana map indicates the location of MLELC in Noble County. (Source data from the Indiana Geological Survey, Spatial Data Portal, at: <http://gis.iu.edu/downloadData/index.php>)

in Wysong was 2.2 mm to 34 mm (mean 9.9 mm). The average canopy cover was 95%.

Byer: This woodland surmounts a gravelly esker, with more mesic forest communities on either side of this drier upland. It has many deer trails and a great diversity of plot settings; for example, dense populations of *Prunus serotina* Ehrh. could be found away from the esker but *U. rubra* was well represented throughout. Important tree species in this woodlot included *A. saccharum*, *Celtis occidentalis* L., *Fraxinus americana* L., *J. nigra*, *O. virginiana*, *P. serotina*, and *U. rubra*. Significant shrub species included *L. benzoin*, *R. occidentalis*, and *R. multiflora*, as well as saplings of many of the above tree species. The majority of

these plots were located along a narrow section of the woodland with frequent canopy gaps due to fallen oak and elm trees. In addition, patches of marshy soil and accompanying fern cover were present within or adjacent to several plots. The range of litter depth in Byer was 10.8 mm to 27.4 mm and the average was 17.3 mm. The average canopy cover was 96%.

Statistical analysis.—Data were analyzed using PC-ORD (v5.1) and R (v3.0.2). PC-ORD was used to run non-metric multidimensional scaling (NMS) ordination of the understory community data (autopilot mode with Sorensen distance measure; 50 runs with real data, and 50 with randomized; maximum iterations set at 200; stability criterion over last



Figure 2.—Typical plot structure of Thomas (A) and Wysong (B). Note the relatively open understory in the former as compared to the latter. Both forests are on former agricultural land (pastures) within a half-mile of each other.

10 iterations set to 0.00001). R was used to run stepwise multiple linear regressions (functions ‘lm’ and ‘step’) to better describe which of the numerous plot level variables most influenced the richness and abundance of non-native species in the plots. R was also used to generate one-way ANOVAs (function ‘aov’) to compare means for biotic and environmental variables between the three woodlots ($\alpha < 0.05$), with Tukey’s pairwise test used post-hoc to identify significantly different forests.

The floristic quality index (FQI) value was calculated for each plot using the formula below, which was developed by Swink & Wilhelm (1979, 1994), and subsequently modified for use in Indiana (Rothrock 2004). This index weights the species present in a community by the relative coefficient of conservatism (CC) of each species, where lower values are assigned to more wide-ranging, disturbance-tolerant species and higher

Table 1.—Measurements, observations, and spatial data collected for each plot.

Data category	Variables measured or calculated
Overstory (Trees)	<ul style="list-style-type: none"> - species identification - diameter at breast height (DBH; all individuals > 2 m tall)
Midstory (Shrubs and Saplings)	<ul style="list-style-type: none"> - species identification - measured stem count and area covered by each individual (m²) - sapling trees shorter than 2 m were recorded in this layer
Understory (Herbs and Seedlings)	<ul style="list-style-type: none"> - species identification (including tree and shrub seedlings) - % cover for each species in four randomly-distributed 1 m² quadrats per plot, one quadrat per quarter-section of the circle (cover classes: one small plant [0.01], several small plants [0.05], 0.25-0.75, 0.75-1.25, 1.25-5, 5-10, 10-15, 15-20, 20-30, 30-50, 50-90, and > 90%) - floristic quality index value (FQI) calculated for each plot (see text)
Environmental Variables	<ul style="list-style-type: none"> - % canopy cover (mean of concave spherical densiometer readings at 5 locations in plot: center and 1 m from center on each of four cardinal direction bearings) - mean leaf litter depth-to-duff, cm (measured at same five locations) - presence/absence of natural (deer trails, windthrown trees) and anthropogenic (foot trails, farm trash dumps) disturbances within the plot borders
Geospatial Data	<ul style="list-style-type: none"> - distance of each plot center to forest edge in 2012 (using orthophotography) and in 1938 (Byer only; using georectified aerial photography; Thomas and Wysong have regenerated since 1938) - underlying SSURGO soil series (then simplified to two main series – muck and loam) and permeability class (high, medium, and low) - mean slope of plot (%) and plot aspect (azimuth categorized into appropriate cardinal direction [e.g., $315 \leq x < 45 = N$, $45 \leq x < 135 = E$, etc.]; both calculated based on $5 \times 5'$ digital elevation models from the State of Indiana)

Table 2.—Summary of the species richness, densities (cover), and selected physical characteristics of plots in this study. All numerical values are means of plots in that category (standard error values follow in parentheses); qualitative values are the most common value for that variable in that woodlot. Variables with a significant difference in means among the three forests are bold-faced, with the *p*-value of their one-way ANOVA test indicated.

Woodlot	Thomas (<i>n</i> = 13)	Wysong (<i>n</i> = 13)	Byer (<i>n</i> = 18)	Overall (<i>n</i> = 44)
Species Richness				
Trees	3.9 (1.1)	2.4 (0.7)	3.0 (0.7)	3.1 (0.2) <i>p</i> = 0.011
Shrubs	1.6 (0.4)	3.1 (0.9)	2.9 (0.7)	2.6 (0.3) <i>p</i> = 0.039
Herbs	10.8 (3.0)	12.7 (3.5)	13.2 (3.1)	12.3 (0.7)
Invasive	0.8 (0.2)	1.2 (0.3)	1.2 (0.3)	1.1 (0.1)
Species Densities				
Shrub Cover (m ²)	4.1 (1.1)	11.7 (3.2)	3.8 (0.9)	6.2 (1.0) <i>p</i> < 0.001
Herbs (% cover)	30.7 (8.5)	45.3(12.6)	50.8 (12.0)	43.2 (4.2)
Invasive Herbs (% cover)	2.1 (0.6)	7.4 (2.0)	3.6 (0.8)	4.3 (1.7)
Physical Characteristics				
Primary Soil Type	Loam	Loam	Loam	Loam
Soil Permeability	High	High	Medium	High
Mean Litter Depth (mm)	13.8 (3.8)	9.8 (2.7)	17.3 (4.1)	14.1 (1.3)
Mean Canopy Cover (%)	94 (1.1)	95 (1.5)	96 (0.4)	95.4 (0.6)

coefficients (up to 10) to more disturbance-sensitive species. Together with the overall richness of native species in the community, this index differentiates communities based not just on richness, but also the likelihood that the species present represent the native suite for such habitats. The FQI values were then used in the regression analyses with R. The formula below describes calculation of the standard FQI value, where CC_i is the CC value for every species in the sample and $N_{\text{native species}}$ is the richness of native species found in the sample.

$$FQI = \frac{\sum(CC_i)}{\sqrt{N_{\text{native species}}}}$$

RESULTS

General forest descriptions.—In general the plots were similar across all three forests in terms of structural variables such as soil series and permeability, litter layer thickness, and canopy coverage (Table 2). Herbaceous species richness and overall cover were also similar across all plots, although the species composition varied from plot to plot. On average, plots in these forests contained about 12 total species in their mid-summer herbaceous layers, with one of those being a non-native invasive species (from among four principal species: *Alliaria petiolata* (M. Bieb.) Cavara & Grande, *Euonymus alatus* (Thunb.) Siebold, *Rosa multiflora*,

and *E. umbellata*). Similarly, herbaceous plants covered 43% of the forest floor in these plots, with invasives comprising one-tenth of that total coverage. The three forests, however, did have differences between them in terms of the woody plants (Table 2). Thomas had significantly higher richness of tree species and significantly lower shrub species richness per plot. In terms of total shrub cover, though, Wysong had substantially more shrubs than either Thomas or Byer (Table 2).

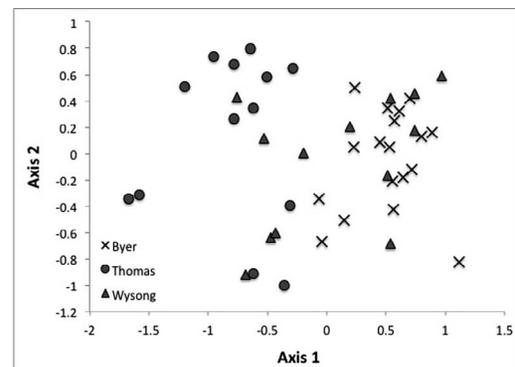


Figure 3.—NMS ordination results for three woodlots in Noble County, Indiana. For clarity, only two dimensions of the ordination are depicted, although a three-dimensional solution was more parsimonious. Plots from different woodlots are depicted with different symbols.

Table 3.—Correlations of key environmental variables to the three axes from the most parsimonious NMS solution.

	Axis 1		Axis 2		Axis 3	
	<i>r</i>	<i>r</i> ²	<i>r</i>	<i>r</i> ²	<i>r</i>	<i>r</i> ²
Canopy Density	0.405	0.164	0.128	0.016	0.09	0.008
Litter Depth	-0.075	0.006	0.162	0.026	0.195	0.038
Disturbance Presence	-0.172	0.029	-0.318	0.101	-0.175	0.031
Distance-to-Edge	0.404	0.163	-0.036	0.001	-0.295	0.087
Adjacent Land Use	0.431	0.186	0.362	0.131	0.014	0.000
Soil Category	-0.126	0.016	0.320	0.103	-0.280	0.078

Invasion characteristics by plot.—Invasion was also related to plot-scale disturbance factors such as deer trails, fallen trees, and farm trash dumps. There was an inverse relationship between the herb and shrub layers in such plots. In plots with higher shrub abundance, whether native or not, there was less herbaceous cover overall. Invasion also seemed to vary with soil type; muck soils were observed to have more total herb abundance and richness but loam soils had higher invasion. In our plots greater herbaceous species richness and cover of invasive herbs occurred on plots with loamy soils of medium-high permeability.

Composition in relation to environmental variables.—The ordination graph (Fig. 3) illustrates the relative dissimilarities between the different plots according to the understory species present. NMS analysis illustrated the dissimilarity between Thomas and Byer woodlots, and the high variability of the plots in Wysong. Mean final stress for a two-dimensional solution was 24.706 (28.03 in the Monte Carlo test), for a 3-dimensional solution was 17.476 (18.97 in Monte Carlo) and 14.011 for a 4-dimensional solution (14.5 in Monte Carlo). Axis 1 explained 31.8% of the variation, Axis 2 12.0%, and Axis 3 an additional 16.0%, meaning the three-dimensional solution accounted for 60% ($r^2 = 0.598$) of the total variation. The environmental factors correlated to the plot ordination axes include canopy cover, litter depth, present distance from edge, and adjacent land use type (Table 3).

Richness of the understory was correlated with several variables that influenced the abiotic conditions of the forest floor, as seen in the multiple regression model results (Table 4). The richness of invasive species alone was not strongly connected to the environmental variables mea-

sured in this study, except that it increased with increasing overall herbaceous richness and decreased with increasing presence of more ecologically ‘conservative’ native species (i.e., FQI). Not surprisingly, the cover of invasive species was positively related to the richness of invasives present (where one species was found more typically were), although their abundance also increased on loamy soils and decreased where a rich shrub community was present.

DISCUSSION

This study, although limited in scope, underlines the crucial importance of forest longevity on limiting the spread of non-native invasive species, even though numerous other variables can

Table 4.—Summary of forward-selected linear regression models examining the relationships between invasive species richness and abundance and a suite of environmental variables collected at the plot level. Estimated coefficients of each relationship are shown, with significant relationships in **bold** (* = $p < 0.05$, ** = $p < 0.01$, *** = $p < 0.001$). Overall model results: Invasive richness ($F = 8.137$ on 5 and 37 df ($p = 0.00003$), final AIC of -56.96 vs null model AIC of -18.35); Invasive abundance ($F = 5.877$ on 5 and 37 df ($p = 0.00043$), final AIC of 112.5 vs null model AIC of 127.61).

Variable	Invasive richness	Invasive abundance
(Intercept)	0.4303	2.9861
Invasive Richness		2.6701***
Shrub Spp. Richness	-0.1003	-0.8035*
Muck Soils		1.5040
Loam Soils		3.0702*
Tree Spp. Richness		-0.7359
Invasive Abundance	0.0364	
FQAI	-0.1371**	
Herb Richness	0.1583***	
Slope	-0.0150	

likewise affect the composition of forest plant communities. Although invasive species, such as *A. petiolata* and *R. multiflora*, were observed in many of the plots in this study, in very few of those plots were invasive species common, even those species that have been present on the surrounding landscape for decades (Brothers & Spingarn 1992). This same pattern held across relatively large variations in the structure (mid- and understory cover) and richness of the plant community, with the particular species composition being less important than the age and structural development of the surrounding forest (Table 4). Surprisingly, local disturbances of any type tended to correlate to lower cover of invasives in the understory, with many of these also associated with higher understory FQI values. This hints that local (plot-level/microsite) disturbance may be important in promoting diversity and even limiting invasion in these forests, by creating reservoirs of plant propagules necessary to maintain landscape (beta) diversity and support community resistance to invasion (Hobbs & Huenneke 1992; Holle 2013).

Although invasive species were sparse in many of the plots, the type of plots that they were preferentially found in was telling, namely those with recent (i.e., 50–60 years before present) agricultural activity (often loamy soil sites), and adjacent to ongoing agricultural disturbances. Although these factors were not all statistically significant in our study, they align with other studies that identified recent soil disturbance, elevated soil nutrient levels (especially nitrogen), and propagule dispersal through wildlife such as birds as vital for the spread of invasive species (Flinn & Vellend 2005; Knight et al. 2009; Medley & Krisko 2007). They also are often brighter, warmer, and drier than older, more structurally complex forests in the region, and thus may be preferred habitats for many of the common invasive species in the area, which are well-adapted for droughty, disturbed, higher-light conditions (Brothers & Spingarn 1992; McDonald et al. 2008).

These findings have several useful implications for management regimes regarding invasive plants in this region. Concentrating invasive species management in forests with recent agricultural histories, rich soils, and close to agricultural land will more clearly reduce the population densities of many invasive plants. By contrast native-dominated forest interiors will likely continue to support that diversity, provided canopies

remain largely intact outside of natural tree fall or careful selective logging. Another corollary is that awareness and careful monitoring of existing disturbed areas in forest interiors is an important way to prevent incipient invasions from becoming larger ones.

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REMEDICATION OF AN INDIANA BROWNFIELD: A STUDENT IMMERSIVE LEARNING EXPERIENCE

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ABSTRACT. Over five years a former auto salvage yard in Indiana was transformed from an unsightly and hazardous brownfield to an urban green space by student teams from Ball State University, Muncie, IN. The project assessed the feasibility of selected turf species for recolonization of barren and potentially toxic soil. This was followed by a phased design and implementation plan for the space, providing different stages of rehabilitation. Subsequently, a range of activities was conducted in the field including debris removal, eradication of invasive vegetation, installation of a clay cap and soil cover to several acres, planting turf grass and ornamental trees on capped areas, creation of small prairies, and installation of a hiking trail. Students gained academic credit for an ‘immersive learning activity’ that incorporated site assessment, data analysis, site planning, and map preparation using computer-aided design, as well as intensive field work. Similar applications of the reported restoration project may be applied to brownfields in other cities, potentially creating a series of spatially- or thematically-connected park spaces out of formerly unusable land.

Keywords: Brownfields, immersive learning, remediation, restoration

INTRODUCTION

Hundreds of thousands of brownfield sites have been documented throughout the United States (US EPA 2015), and an estimated 2250 occur in Indiana (IDEM 2017). A brownfield is defined as an ‘abandoned, idled, or underused industrial or commercial site where expansion or redevelopment is complicated by real or perceived environmental contamination’ (Edwards 2009). Such sites are often infertile and may be contaminated with heavy metals or other hazardous substances. The degree of contamination may pose a public health risk and is, at a minimum, detrimental to plant growth.

Remediation of brownfields can be carried out via several techniques, such as soil vapor extraction, soil washing, and chemical treatment. Many such technologies, however, permanently alter soil properties, employ costly systems, and require the use of hazardous materials (Li no date). Less invasive methods include capping with geomembranes and/or clay followed by placement of a topsoil layer.

Plant-based remediation technologies are considered a low-cost and effective treatment for many brownfields. Certain plant species have been found to remove potentially toxic metals

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from soil. In other cases, revegetation is preferred due to its benefits in preventing erosion by the action of water or wind, limiting runoff of metallic debris, and in creating an aesthetically pleasing site (US EPA 2006).

Brownfield soils commonly possess traits such as infertility, poor drainage, low organic matter content, and limited populations of indigenous microorganisms that cycle nutrients. Consequently, numerous studies have been carried out using municipal and industrial wastes as soil substitutes on drastically disturbed and/or contaminated soils (Pichtel et al. 1994; Halofsky & McCormick 2005; Pichtel & Bradway 2007).

In decades past Muncie, IN was home to numerous heavy industries; their subsequent closing and/or departure resulted in the presence of many derelict sites. One of these brownfields was the focus of a remediation project carried out by Ball State University (Muncie, IN) student teams over five years (2012 to 2017). The ultimate intent of the project was to redevelop an abandoned auto scrapyards to a community green space. Restoration of the property offered an opportunity for community enhancement. Easily accessible by public transportation and therefore available for public use, the site lies adjacent to the Cardinal Greenway, a rails-to-trails project, which provides standard recreational amenities.

A number of local organizations (e.g., neighborhood associations; the Red Tail Conservancy

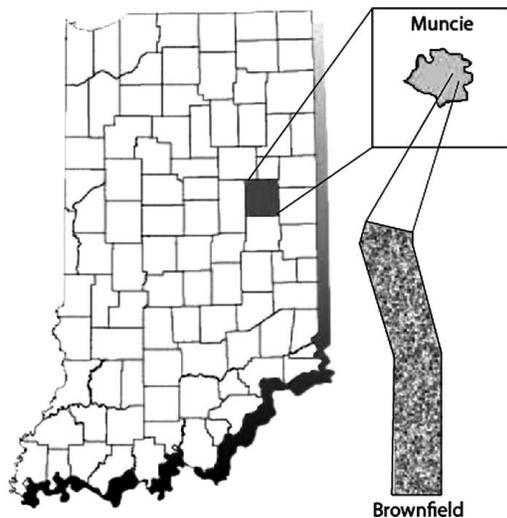


Figure 1.—Regional and local maps showing the study area.

Land Trust) expressed enthusiasm in creating green spaces for the neighborhood, and in converting this blighted brownfield site to beneficial use. Ultimately, the community surrounding the site will benefit from the transformed property by experiencing enhanced aesthetics, creation of valuable green space, increased property values, and by local citizens ‘owning’ the site.

Specific goals of the remediation were to (1) assess the impact of soil amendments and selected turf species in colonizing the infertile site; (2) remove automotive debris and other solid waste for proper disposal; (3) eradicate noxious and invasive vegetation; (4) establish selected tree and prairie species; (5) encourage wildlife by building appropriate structures; and (6) construct park amenities for future users. Actualization of these goals depended upon students involved in immersive learning who earned university-level credit for their contribution in transforming the site to an urban green space.

Immersive learning.—Ball State University’s Immersive Learning program goes beyond school-sponsored service through internships. Distinctive characteristics, including student-driven projects, lasting outcomes, and community partnerships, set immersive learning apart from other experiential learning opportunities at the University. To be considered immersive, a project must:

- carry academic credit and focus on student learning outcomes

- engage participants in an active learning process that is student-driven but guided by a faculty mentor
- produce a tangible outcome or product, such as a business plan or policy recommendations
- involve at least one team of students, working on a project that is interdisciplinary in nature
- include community partners and create an impact on the larger community as well as on the student participants.

MATERIALS AND METHODS

The site.—The Indiana Brownfields List (IDEM 2012) was reviewed for potential sites within Delaware County, IN, that would be feasible for restoration. Using GIS mapping, prospective sites were ranked via the following criteria: (1) a registered brownfield located adjacent to areas considered disadvantaged and of high density; (2) near existing residential areas; and (3) accessible by public transportation. The site considered most suitable for the project was a former auto salvage yard in Muncie, IN (LAT/LONG: 40.1881°N / 85.3628°W). The site is located approximately 1.6 km southeast of downtown Muncie (Fig. 1).

The property had been used as a commercial and industrial facility since 1934. From 1934 to 1971 a bulk oil and gas storage facility, a refinery, and other businesses were active; two oil warehouses, two pump houses, two other buildings, ten gasoline tanks, and a large ammonia storage tank were previously housed on-site. From 1976 to 2002 the property was used as an automotive salvage yard. Following closure in 2002, substantial waste was left behind. A spill was reported on the site in 2003, a result of vandalized drums releasing grease and automotive fluids into soil. All contaminants along with affected soil and materials were excavated and removed from the site under orders of the Indiana Department of Environmental Management.

During an environmental site assessment (Symbiont 2008, 2009), tires, automotive parts, construction and demolition debris, two trailers, and general refuse were identified throughout the 2.1 ha site. There was evidence that unknown materials had been dumped and residues from oil and gasoline releases were detected. No above-ground or underground storage tanks were observed during the site investigation. Several

soil/fill piles with unknown contents were located on the north side of the property and a wrecked office trailer was located on the southeast corner of the site.

Wetlands occur 1.3 km northeast of the site. No wetland delineation was carried out on the property. The site is not located in a flood zone; it is flat with sparse vegetation and exposed soil. Areas of standing water have been observed, likely from a high water table combined with poor drainage. Groundwater movement is to the north/northeast toward the White River. Soil type is classified as UfuA—Urban land-Millgrove complex, 0 to 1 percent slopes (USDA-NRCS, 2016).

Residential neighborhoods composed of standard single-family homes border the east and west of the site. A number of businesses occur directly to the south. This is primarily a family-based neighborhood that is showing signs of economic decline. Green spaces (e.g., parks) are severely limited in the area.

Environmental site assessment.—Student teams engaged in an extensive review of known site conditions, initially referring to phase I and phase II environmental site assessments (Symbiont 2008, 2009). The teams additionally referred to USDA-NRCS soil survey maps, geologic maps, Sanborn Fire Insurance maps, and other published data before going into the field. Teams subsequently engaged in extensive site reconnaissance. During site recon, soil material was collected and analyzed for various chemical parameters (i.e., plant nutrients, heavy metals; see below).

Prior to entry to the site, all students were trained in relevant aspects of site safety, including characteristics of hazardous chemicals, use of personal protective equipment, and potential site hazards. Students were required to read and sign a detailed Health and Safety Plan.

Plant tolerance and metal uptake.—The ability of several plant species, native to the Midwestern United States and showing promise in earlier studies to contribute to metal uptake and/or stabilization (Pichtel & Bradway 2007), were evaluated at the site. Plots measuring 2 × 3 m were established, and perennial ryegrass (*Lolium perenne* L.) was seeded at a 190 kg/ha rate (IN DOT 2015) and red clover (*Trifolium pretense* L.) at 100 kg/ha. Plots were amended with composted organic material which was derived from leaves and grass clippings, at a rate of 100 metric tons/ha. Soil was sampled from plots at the end of the

growing season from the upper 20 cm and returned to the laboratory, where it was air-dried and sieved through a 2 mm mesh sieve.

Determination of soil K, Cd, Cr, Cu, Ni, Pb, and Zn was carried out after shaking 5 g of soil with 25 ml of 5 mM diethylenetriaminetriacetic acid (DTPA) on an oscillating shaker (120 osc./min) and measurement using flame atomic absorption spectrophotometry (FAAS) (Perkin-Elmer Analyst 2000, Norwalk, CT).

A subsample of soil from the plots was analyzed for particle size distribution using the hydrometer method (Day 1965). Organic carbon content was determined by loss on ignition at 360° C (Nelson & Sommers 1982) and pH in a 1:2 (w:v) soil:deionized water slurry. Soils were tested for soluble NO₃⁻ and NH₄⁺ by the microplate method (GEN 5 Microplate Spectrophotometer Powerwave X5/X52 by BioTek). The organic compost was also tested for the above parameters.

Above-ground plant tissue was sampled 90 d after seeding. Tissue was cut ~ 1–2 cm above ground surface and placed into paper bags where it was dried for 48 h at 105° C. A total of 0.5 g tissue was mixed with 10 ml of 75–80% concentrated HNO₃ and microwave-digested (MARS microwave digester, CEM Corp., Matthews, NC). The method used for plant digestion was as follows: vessel temperatures were ramped to 190° C for 15 min with a holding period of 15 min at 800 W. The cooled digestates were diluted with 40 ml DI water and analyzed for Cd, Cr, Cu, Ni, Pb, and Zn using FAAS. Potassium concentrations were analyzed using ion chromatography (Dionex ICS 5000). A total of 4.9 µl of the tissue HNO₃ digestate was mixed with 100 µl of methanesulfonic acid and passed through a CS-19 cation exchange column prior to injection into the instrument.

Site restoration activities.—Since 2012 teams of Ball State University students with backgrounds in environmental management were recruited for the site restoration activity. Students were brought together in a so-called ‘immersive learning’ course through the university in order to gain academic credit from the experience. Five student teams, addressing different aspects of site remediation, have operated at the site over five years. Approximately 75 students have been involved in the project. Learning objectives for the immersive learning activity appear in Table 1.

A phased design and implementation plan for the space was formulated with different stages of

Table 1.—Learning objectives for the immersive learning activity.

Number	Outcome
1	Demonstrates a working understanding of core concepts (including principles, relationships, theories, and relevant laws of nature) applicable to the management of natural and man-made environments.
2	Respects and uses critical thinking skills and skeptical inquiry to assess potential environmental hazards and propose suitable responses.
3	Understands and applies valid and reliable scientific methods in the conduct of research.
4	Demonstrates proficiency and attention to safety in the use of analytical instrumentation.
5	Demonstrates proficiency in computer applications, including the compilation and analysis of data and the presentation of both data and analytical results.
6	Demonstrates a knowledge of professional ethics in the student's chosen field.
7	Demonstrates a command of written English to communicate technical information to scientific and lay audiences.
8	Understands and applies the relevant regulatory and policy requirements in the management of natural resources and the environment.

rehabilitation. The ultimate intent was to enhance local environmental quality as students worked together to redevelop the site. Activities included (1) debris removal; (2) eradication of invasive and nuisance plant species; (3) fence removal and replacement; (4) capping and installation of topsoil cover; (5) prairie establishment and encouraging wildlife; and (6) trail construction.

RESULTS AND DISCUSSION

Characterization of soil and compost.—Soil pH was 7.6 and soil total organic content (TOC) ranged from 0.6% to 13.1% with an average of 3.8% (Table 2). The study site is highly variable in terms of soil chemical and physical properties due to a wide range of industrial and commercial uses over almost a century (Symbiont 2009). It is suspected that TOC levels > 2% are due to anthropogenic inputs, e.g., improper release or disposal of fuels and lubricating oils (Cornelissen et al.

2005). Soil NO_3^- and NH_4^+ were 1.4 and 3.1 mg/kg, respectively, and K concentration was 1072.8 mg/kg (Table 2). Soil texture was a sandy loam. Compost TOC measured 36.8%. Compost concentrations of NO_3^- , NH_4^+ , and K^+ were somewhat lower than those of soil (Table 2).

Soil Cd concentration was 6.3 mg/kg (Table 2). The Cd values are considered evidence of anthropogenic contamination (Kabata-Pendias 2001) since Cd concentrations of non-contaminated soils range from 0.1–0.5 mg/kg. Cadmium concentrations of contaminated soils can measure as high as >14,000 mg/kg (Gohre & Paszkowski 2006). At an Indiana Superfund site, Pichtel et al. (2000) measured 52 mg/kg soil Cd. Concentrations of soil Cu, Cr, Ni, Pb, and Zn were commensurate with those of local agricultural soils (Table 2).

Plant analyses.—Red clover did not become well established at the field plots. This may

Table 2.—Selected soil and compost chemical and physical properties. ($n = 4$.)

	pH	TOC	NO_3^-	NH_4^+	K	
	%		mg/kg			
Soil	7.6 ± 0.1	$3.8 \pm 1.2^*$	1.4 ± 0.9	3.1 ± 2.4	1072.8 ± 52.3	
Compost	7.5 ± 0.01	36.8 ± 5.8	0.9 ± 0.1	2.5 ± 0.1	716.5 ± 51.0	
	Cd	Cu	Cr	Ni	Pb	Zn
	mg/kg					
Soil	6.3 ± 2.3	131.4 ± 17.7	15.4 ± 8.6	23.2 ± 3.7	55.0 ± 39.0	334.5 ± 78.5
Compost	1.3 ± 1.0	70.1 ± 7.8	3.8 ± 3.6	3.5 ± 1.5	15.7 ± 12.5	76.5 ± 52.2

* Range 0.6 - 13.3%.

Table 3.—Metal concentrations, ryegrass tissue. BDL* = below detectable limit.

Cd	Cu	Cr	Ni	Pb	Zn
mg/kg					
BDL*	124.5 ± 33.3	33.3 ± 17.8	72.8 ± 20.5	226.4 ± 217.9	303.5 ± 80.1

have been due, in part, to a significant drought occurring during part of the growing season. Furthermore, invasive species became established on many plots and may have suppressed clover growth.

Ryegrass tissue Cd concentration was below detectable limits (Table 3). Tissue Cu and Cr measured 124.5 and 33.3 mg/kg, respectively. Tissue Ni, Pb, and Zn were 72.8, 226.4, and 303.5 mg/kg, respectively. The quantity of tissue Pb is a promising indicator for phytoextraction. This species, however, does not qualify as a hyperaccumulator of soil Pb. A hyperaccumulator is defined as a plant that has the ability to accumulate extraordinarily high amounts of heavy metals in the aerial organs, far in excess of the levels found in the majority of species, without suffering phytotoxic effects (Rascio & Navari-Izzo 2010). Previous studies have demonstrated that perennial ryegrass has the potential for the rehabilitation of Cd- or Pb-contaminated soil (Arienzo et al. 2004; Si et al. 2006).

The ryegrass had poor coverage on infertile soil material; however, average coverage percentage on plots amended with compost was 75% (data not tabulated). Ye et al. (2000) found that Italian ryegrass (*Lolium multiflorum* Lam.) experienced a higher percent coverage of test plots when grown with a soil amendment barrier of fly ash or combusted coal residue as compared to bare soil.

Site restoration.— *Debris removal:* Removal of automobile debris and other solid waste was carried out by Ball State University students; on one occasion, several persons required to engage in community service were brought to the site, courtesy of Delaware County Community Corrections. Work teams transported waste to roll-off dumpsters provided by the City of Muncie. At least 20 large roll-off dumpsters (38 m³ volume each) were filled and removed for disposal. In 2013 the abandoned office trailer was demolished by city crews using conventional heavy equipment.

Eradication of invasive and nuisance plant species: The site was overgrown with invasive species of perennial shrubs and trees including Asian bush honeysuckle (*Lonicera maackii*

(Rupr.) Maxim.), multiflora rose (*Rosa multiflora* Thunb.), stinkweed (*Thlaspi arvense* L.), Tree of Heaven (*Ailanthus altissima* (Mill.) Swingle), Siberian elm (*Ulmus pumila* L.), cottonwood (*Populus deltoides* Marshall), poison ivy (*Toxicodendron radicans* (L.) Kuntze), among others. As many invasive plants occurred alongside fences, they could not be effectively removed by a Bobcat® tractor; hence, they were cut using chainsaws and/or loppers. Student teams decided that chemical treatments were not to be used for plant removal.

Fence removal and replacement: A 3 m tall corrugated steel perimeter fence along portions of the property was dismantled manually. This process was preferred rather than using heavy equipment in order to support recycling of useful components such as heavy wooden beams and sheet metal, and to keep costs under control. On the south end of the property a split rail fence was installed. A split rail gate for possible future pedestrian traffic was included. Due to the extreme density of the underlying soil material, a gas-powered auger was necessary to drill holes for fence posts.

Capping and topsoil cover: The surface of the site was restored sequentially in 0.4 hectare (1 acre) sections. First, the surface was scraped using a Bobcat tractor in order to (1) remove debris occurring within the top 0.5 m for eventual removal; (2) create a level or contoured surface (depending on location); and (3) remove additional unwanted vegetation. Following scraping, local clay material was delivered. Clay piles were spread using a Bobcat to a depth of 0.5 m and subsequently compacted. The clay cap was covered with topsoil, which was spread to a depth of 0.5 m using the Bobcat. Straw was immediately placed over the exposed soil to prevent erosion by water or wind.

Plant establishment and support of wildlife: Immediately following placement of straw onto soil, turf grass seed was applied. Perennial ryegrass in combination with red clover was selected. Seed was distributed using a mechan-

Table 4.—Species of grasses and flowering plants installed in the four prairies.

Common name	Scientific name
Big bluestem	<i>Andropogon gerardii</i> Vitman
Switchgrass	<i>Panicum virgatum</i> L.
Prairie oval sedge	<i>Carex bicknellii</i> Britton
Side-oats grama	<i>Bouteloua curtipendula</i> (Michx.) Torr.
Nodding wild onion	<i>Allium cernuum</i> Roth.
Heath aster	<i>Symphotrichum ericoides</i> (L.) G.L. Nesom var. <i>ericoides</i>
White false indigo	<i>Baptisia alba</i> (L.) Vent. var. <i>macrophylla</i> (Larisey) Isely
Purple coneflower	<i>Echinacea purpurea</i> (L.) Moench
Rattlesnake master	<i>Eryngium yuccifolium</i> Michx.
Downy sunflower	<i>Helianthus mollis</i> Lam.
False sunflower	<i>Heliopsis helianthoides</i> (L.) Sweet
Foxglove beardtongue	<i>Penstemon digitalis</i> Nutt. ex Sims
Purple prairie clover	<i>Dalea purpurea</i> Vent.
Yellow coneflower	<i>Ratibida pinnata</i> (Vent.) Barnhart
Butterflyweed	<i>Asclepias tuberosa</i> L.
Rosinweed	<i>Silphium integrifolium</i> Michx.
Compass plant	<i>Silphium laciniatum</i> L.
Sky blue aster	<i>Symphotrichum oolentangiense</i> (Riddell) G.L. Nesom var. <i>oolentangiense</i>
Riddell's goldenrod	<i>Solidago riddellii</i> Frank ex Riddell
Rough blazingstar	<i>Liatris aspera</i> Michx.
Round-headed bushclover	<i>Lespedeza capitata</i> Michx.

ical seed spreader. The newly-established vegetation was allowed to grow to about 20 cm tall, which encouraged formation of a dense root mat. Following initial mowing, the grass was mowed on an as-needed basis. Turf was watered by rainfall only.

Several ornamental and fruit trees were purchased locally for planting. Species included flowering dogwood (*Cornus florida* L.), sweetgum (*Liquidambar styraciflua* L.), Yoshino flowering cherry (*Prunus × yedoensis* Matsum.), quaking aspen (*Populus tremuloides* Michx.), and Shumard oak (*Quercus shumardii* Buckley).

Students identified four areas for establishment of tall grass prairies. The seedbed was prepared both manually and with a rototiller. Seeds and live seedlings (Table 4) were purchased from local sources.

Students constructed several bird, bat, and butterfly houses that were subsequently installed around the property.

Trail construction: Hiking trails were installed throughout the site. The trails begin at the south entrance and extend through the woodlot. Initially, teams manually cleared small trees and stumps using chain saws and loppers. This was followed by a Harley® rake attached to a Bobcat tractor to clear the smaller vegetation across a width of 2 m. The cleared area was covered with hardwood chips,

provided by Ball State University, to a depth of 20 cm. The layout of the property is shown in Fig. 2.

CONCLUSIONS

A former brownfield in Muncie, Indiana has undergone extensive improvement in terms of environmental quality, safety, and aesthetics. Surface debris has been removed; barren soil has been capped by a thick clay and soil cover; and lawn, prairie and ornamental plantings now predominate on much of the property. In essence, the restored site now serves as an asset, rather than a liability, for the local community.

The project relates to the Ball State University Strategic Plan as follows:

1. Ball State University has pledged to offer “action-oriented learning, including immersive out-of-class experiences”. In this project student teams actively participated in the product by designing, planning, and constructing the new facility. A significant input of time and expertise has come together to formulate an attractive, community-friendly property.
2. The university is to “Embrace and support partnerships and collaborations across the institution and with the greater external community”. This goal was achieved via

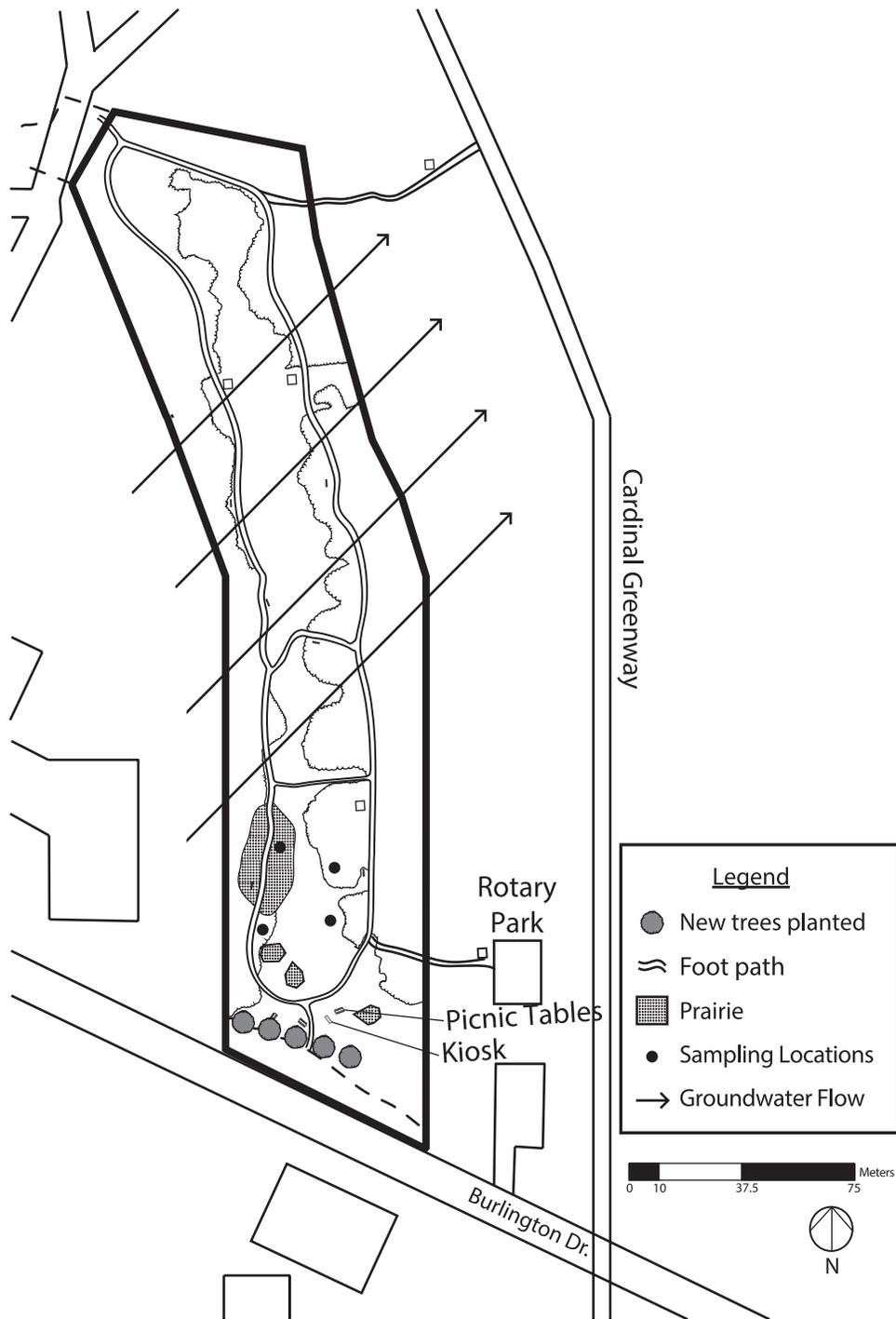


Figure 2.—Current layout of the site showing various improvements.

regular interaction with community organizations regarding site planning and implementation. Long-term collaboration is encouraged for on-going site maintenance and enhancement. Avenues will be sought for redevelopment work at nearby sites.

3. The Ball State University Strategic Plan for 2012–2017 offers the goal: “Continue to position the university as a steward of the environment by building on the university’s expertise and success in sustainability.” The redevelopment work clearly aligns with the concept of sustainability. According to US EPA (2014), sustainability “creates and maintains the conditions under which humans and nature can exist in productive harmony, that permit fulfilling the social, economic and other requirements of present and future generations”.

The reported project has served as a highly effective learning experience for Ball State University students. Knowledge and skills were gained in computer-aided design, environmental planning, plant ecology, soil science, and site remediation. Both team and communication skills were enhanced among students. Throughout this experience, students described their immersive learning experience as ‘a valuable way to understand underlying concepts’, ‘more interesting’, and ‘a better way to retain facts and data’... as compared with classroom-based instruction. Additionally, students appreciated working on their own in planning the future of the site.

The study demonstrates that a former brownfield is capable of being revegetated by common turf species. It is emphasized, however, that each infertile and/or toxic site must be assessed for revegetation species on a case-by-case basis.

Dedicated work goes on at the site; Ball State students continue to augment professional skills as well as gain academic credit while enhancing the quality of the property.

ACKNOWLEDGMENTS

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FLORISTIC INVENTORY OF THE COOPER WOODS–SKINNER WOODS COMPLEX, BALL STATE UNIVERSITY, DELAWARE COUNTY, INDIANA

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ABSTRACT. Owned by Ball State University, both Cooper Woods (CW) and Skinner Woods (SK) are located in northwest Muncie, Indiana. Together the two woodlands are ~19 ha. A floristic survey of both sites revealed 356 taxa representing 225 genera in 90 families. The 12 families with the highest number of taxa in descending order were Poaceae, Asteraceae, Cyperaceae, Rosaceae, Fabaceae, Brassicaceae, Fagaceae, Lamiaceae, Apiaceae, Asparagaceae, Polygonaceae, and Ranunculaceae; these 12 families included 197 (55.3%) of the species documented. Of the 356 documented species, 276 (77.5%) were native and 80 (22.5%) were non-native. A physiognomic analysis for the combined sites revealed the flora consisted of 77 (21.6%) woody species, 202 (56.8%) herbaceous forbs and vine species, 74 (20.8%) graminoid species (grasses and sedges), and three (0.8%) ferns and allies. The FQI and mean C for the native flora were 55.0 and 3.4, respectively, and for all species were 48.6 and 2.6, respectively. The native FQI and mean C imply that the complex is of nature preserve quality, thus possessing sufficient conservatism and species richness to be of importance from a regional perspective. However, the difference between the mean C values (native vs. all species) suggests that exotics are having a negative impact on the native flora, an observation especially true for CW. No state endangered, threatened or rare species were reported, but two species, *Rudbeckia fulgida* var. *fulgida* and *Spiranthes ovalis* var. *erostellata*, are on the state watch list. Five Delaware County records were reported, three native species (*Cardamine parviflora* var. *arenicola*, *Carex squarrosa*, *Rudbeckia fulgida* var. *fulgida*) and two non-native species (*Chaiturus marrubiastrum* and *Lathyrus latifolius*).

Keywords: Floristic quality index (FQI), county records, vascular plants, flora-Indiana, physiognomic analysis, Delaware County IN

INTRODUCTION

Located in central Delaware County, Indiana, the Cooper-Skinner Area is owned by Ball State University and managed by the Ball State University Field Station and Environmental Education Center (FSEEC; Fig. 1). According to FSEEC documents, the Cooper-Skinner Area is comprised of many distinct microenvironments with each habitat rich in diversity for field research and environmental education purposes. Some of the areas have undergone restoration (i.e., planting with native wetland, forest, or prairie species), while others are left to develop naturally (John Taylor, Land Manager of FSEEC, Pers. Comm.). Despite the heavy use of the two property complex in both teaching and research, there

have been no publications concerning the flora and fauna.

An inventory is the simplest method to document species diversity and is necessary in monitoring changes in species composition of a site over time. Measures of species diversity can be used as indicators of the well-being of ecological systems (Magurran 1988; Bourdaghs 2006). For this study, ecological status was determined by calculating the Floristic Quality Index and average Coefficient of Conservatism (mean C) (see Swink & Wilhelm 1994 and the methods section). Although a list of tree species occurring at Cooper Woods exists (Badger, Pers. Comm.), no such list exists for Skinner Woods. In addition, there are no complete inventories of the flora occurring at either site. Consequently, the overall goal of this project was to produce a vouchered list of the flora within Cooper Woods and Skinner Woods. The specific objectives of this research were: (1) to identify and voucher all vascular plant

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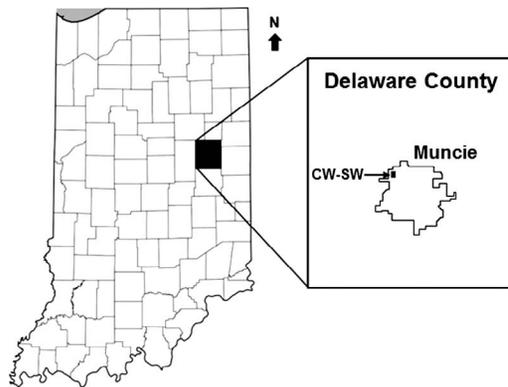


Figure 1.—Map of Indiana showing the location of Delaware County (left) and the Cooper Woods – Skinner Woods complex within the city of Muncie (right).

taxa within each woodland; and (2) to use the species lists to calculate the Floristic Quality Index (FQI) and the average Coefficient of Conservatism (mean C) for the entire site and for the two separate woodlands (Swink & Wilhelm 1994; Rothrock 2004). These results will allow Cooper-Skinner to be compared to other nature preserves studied in Indiana, especially east-central Indiana. They also allow identification of areas of special concern that support rare or threatened plants, if any, and communities sensitive to disturbance.

SITE DESCRIPTION

The study site.—The Cooper-Skinner Field Area is located in northwest Muncie, Delaware County, Indiana (Fig. 1). The Cooper-Skinner Field Area is comprised of three adjacent properties: the Cooper Natural Area (not included in this study), the Cooper Woodland Area, and the Skinner Field Area containing Skinner Woods (Fig. 2). The following information comes from documents maintained by the FSEEC and John Taylor, Land Manager of FSEEC (Pers. Comm.); these documents are currently unavailable to the public pending update of the website.

Cooper Natural Area: The Cooper Natural Area, 23 ha (57 acres), covers the southern portion of the Cooper Property, i.e., from the southern edge of Cooper Woodland south to Bethel Pike (Fig. 2). Prior to 2002 this land was used to grow row crops and hay. Since 2002 most of the land was redirected to the establishment of native prairie vegetation. This

section of the Cooper Property was not included in the current study.

Cooper Woodland Area: This woodland has two units that differ in land use history, current legal ownership, and dates of acquisition. Ball State University (for the Department of Biology) acquired ownership of the Esther L. Copper and Dr. Robert H. Copper's Memorial Woodland Area in 1969. The area covers 13 ha (31.5 acres), divided into Cooper Woods (7 ha; east side), an early succession area (6 ha) containing wildlife plantings and natural vegetation (west side; Fig. 2). Eagle Branch, a tributary of Jakes Creek, begins on the eastern edge of this woodland and flows west and north through the woodland.

Skinner Field Area: This land was donated to Ball State University by William (Bill) Skinner in 2002. Lying north of the Cooper property, the Skinner Field Area is composed of 19 ha (47 acres; Fig. 2). The Skinner Woodland Area, which contains both a woodland and a 6 ha (15 acres) old-field, is contiguous with the Cooper Woodland Area (Fig. 2). The woodland at Skinner appears to have a similar structure to Cooper Woodland, having both a more mature area and an earlier successional area. The successional area includes a small woodland and a larger old-field. Overall, the Skinner woodland is drier than the Cooper woodland.

Site characteristics.—The Cooper-Skinner Field Area drains into Eagle Branch, a tributary of Jakes Creek. Jakes Creek flows west to Killbuck Creek, which flows south eventually emptying into the White River. Thus, the Cooper-Skinner Field Area lies within the Upper White River Watershed (USGS Cataloging Unit: 05120201; USGS 2017).

Cooper-Skinner Field Area lies near the northern border of the Tipton Till Plain Section or the New Castle Till Plains and Drainageways (IUPUI 2013), of the Central Till Plain Region of Indiana (Homoya et al. 1985; Hedge et al. 1997). The Tipton Till Plain is a flat to gently rolling surface produced as a result of the continental glaciation during the Ice Age, particularly the Wisconsin glacial age (Hedge et al. 1997). As the ice sheets retreated, a thick layer of till and outwash filled the bedrock valleys and covered the bedrock hills producing a flat to gently rolling landscape (Hedge et al. 1997; Hill 2015), as seen in the study site.

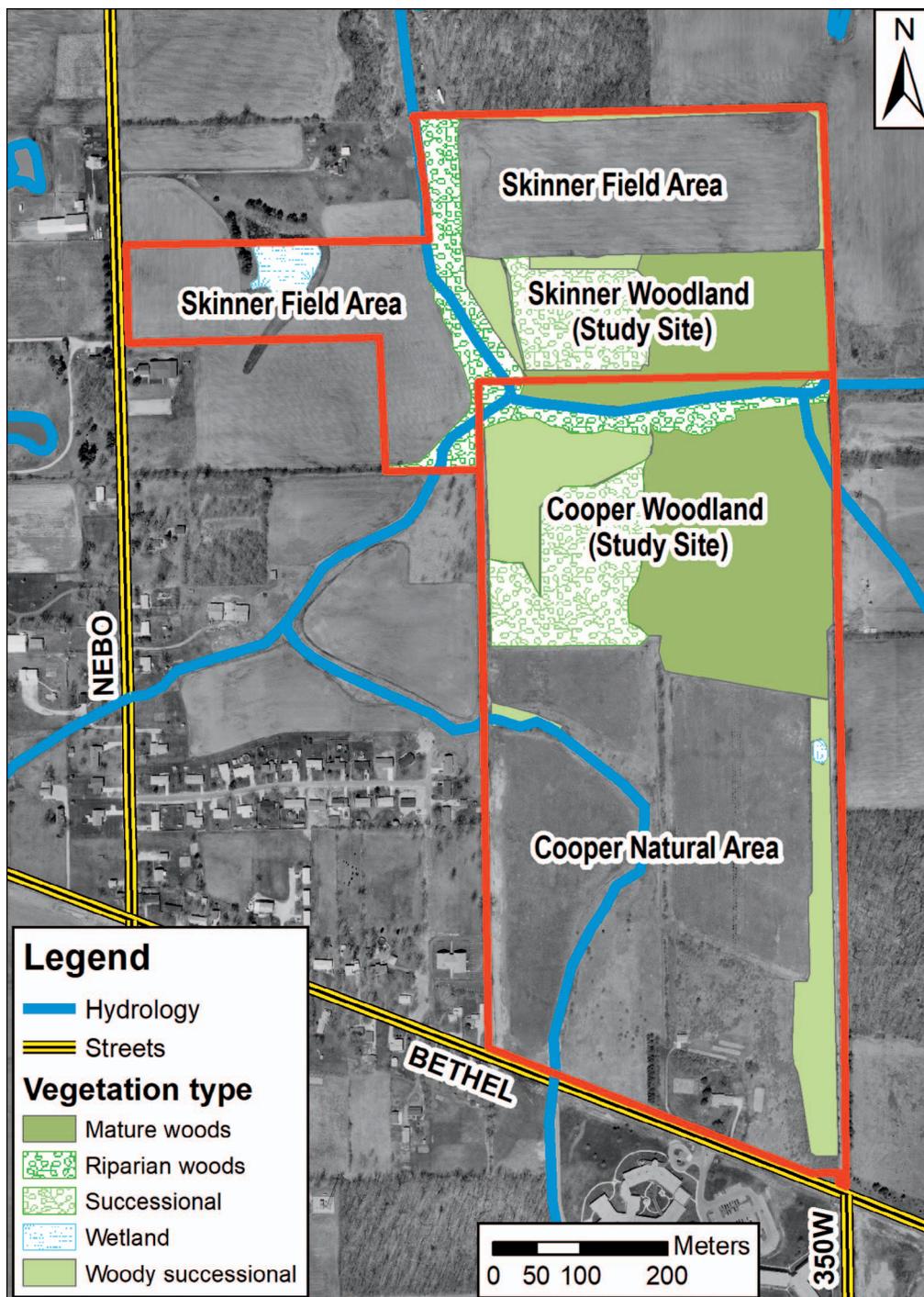


Figure 2.—Modified aerial overview of the Cooper and Skinner Field Areas with emphasis on vegetational communities. Cooper Field Area includes the Cooper Natural Area, not included in this study, and the Cooper Woodland highlighted in color. The Skinner Field Area includes the Skinner Woodland highlighted in color. (Figure courtesy of John Taylor, Land Manager of FSEEC.)

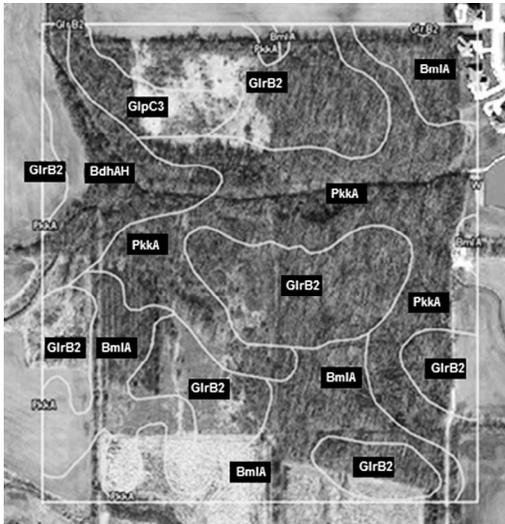


Figure 3.—Map illustrating the distribution of the major soil types at the Cooper Woods – Skinner Woods complex. BdH AH = Bellcreek silty clay loam; BmlA = Blount-Del Rey silty loams; GlpC3 = Glynwood clay loam; GlrB2 = Glynwood silty loam; PkkA = Pewamo silty clay loam. (From WWS 2017)

The soil at Cooper-Skinner consists primarily of silt loams, clay loams, and silty clay loam (WSS 2017). The three groups comprising approximately 89% of the soil at the site are Glynwood silt loam, Pewamo silty clay loam, and Blount-Del Rey silt loam (Fig. 3). Glynwood silt loam has a 1–4 % slope, is moderately well drained, and is considered prime farmland. Pewamo silty clay loam has a 0–1 % slope, is poorly drained, and is considered prime farmland if drained. Blount-Del Rey silt loam has 0–1 % slope, is somewhat poorly drained, and is considered prime farmland if drained.

HISTORY OF THE SITE

The Cooper Property.—In 1951, the Cooper's purchased the land comprising the current Cooper Woodland and Cooper Natural Area as part of a 50 ha (123.3 acre) farm. In 1957 at the request of the Cooper family, the USDA Soil Conservation Service (SCS) designed a management plan to help drain water from the farm. In 1958 an SCS-engineered waterway (drainage ditch) was constructed along the west edge of the property that drained water into Eagle Branch. Beginning in 1959, the northern part of the Cooper property, including the woodland, was placed in the USDA Soil

Reserve Program for a period of five years. The Coopers cleared 610 m of footpaths in the woods in 1960. Between 1961 and 1962, timber, especially white oak (*Quercus alba*), was harvested from the woods, but a number of large mature trees remained. In addition, wildlife plantings were established in the springs of 1971, 1972, and 1974 in the area designated as the successional area today. The wildlife plantings included many exotic and invasive herbs and shrub species, such as autumn olive (*Elaeagnus umbellata*), sericea lespedeza (*Lespedeza cuneata*), and reed canary grass (*Phalaris arundinacea*). In 1993, trail clearing in the wooded section ended and in 2004, most of the exotic wildlife plantings were removed. Lastly, the establishment of a native prairie began in the old fields of the Cooper property that comprise today's Cooper Natural Reserve Program.

The Skinner Field Area.—Ball State University received this parcel of land as a donation from William (Bill) Skinner in 2002. The Skinner Field Area was mainly used for environmental education by Ball State students taking Natural Resources and Environmental Management 101. Currently, there are limited access points to the Skinner Field Area from the Cooper Property.

MATERIALS AND METHODS

During the 2015 growing season [March through October], thirty-two forays were made into the study area; forays were made into the three major habitat types: old-fields, successional woodlands, and mature woodlands (Fig. 2). In addition an effort was made to cover the various microhabitats within these major habitats.

Voucher specimens for each species were collected and deposited in the Ball State University Herbarium (BSUH). Notes on vegetation consisted of a species list with visual estimates of distribution and relative abundance (see catalog of vascular plants, Appendix 1). Relative abundance for species is defined as follows: rare = < 5 sites although a species may be abundant at one site; infrequent = occasional, not widespread throughout its potential habitats, but may be locally abundant at a site; frequent = common throughout its potential habitats and may be locally abundant at one or more sites; and

abundant = common and numerous throughout its potential habitats.

Nomenclature follows the Angiosperm Phylogeny Group (FNA 2008; Angiosperm Phylogeny Group 2009; Stevens 2016). List preparation and sources used to identify plant taxa included Deam (1940), Gleason & Cronquist (1991), Swink & Wilhelm (1994), Yatskievych 2000, Jackson (2004), Weeks et al. (2005), Voss & Reznicek (2012), Weakley et al. (2012), BONAP (2014), and USDA (2017).

Using the program developed by the Conservation Design Forum in conjunction with Rothrock (2004), the Floristic Quality Index (FQI) and the mean Coefficient of Conservatism (mean C) were determined not only for Cooper Woods and Skinner Woods individually, but for the two sites combined. The program also generates a detailed physiognomic analysis of the flora, both native and exotic. For a detailed description of FQI and an explanation of C-values, see Swink & Wilhelm (1994), Rothrock (2004), and Rothrock & Homoya (2005).

Potential Delaware County plant species records were determined from the following sources: Indiana Natural Heritage Data Center's records for Delaware County (available from the Indiana Division of Nature Preserves), Deam (1940), Overlease & Overlease (2007), BONAP (2014), USDA Plant Database (USDA 2017), and the list of species published for Ginn Woods (Ruch et al. 1998, 2004), Munsee Woods Nature Preserve (Prast et al. 2013), and Dutro Woods Nature Preserve (Ruch et al. 2014b). State listed species, i.e., endangered, rare threatened, or of special concern, were determined from the list provided by the Indiana Division of Nature Preserves (IDNR Nature Preserves 2016).

RESULTS

Floristic quality of Cooper Woods and Skinner Woods combined.—The vascular flora of Cooper Woods-Skinner Woods (CW-SW) combined consisted of 356 taxa representing 225 genera in 90 families. Thirty-nine families had only one species and 14 families had only two species. The 12 families with the highest number of taxa were Poaceae (45 spp., 12.6%), Asteraceae (38 spp., 10.6%), Cyperaceae (28 spp., 7.9%), Rosaceae (20 spp., 5.6%), Fabaceae (11 spp., 3.1%), Brassicaceae (9 spp., 2.5%), Fagaceae (9 spp., 2.5%), Lamiaceae (9 spp., 2.5%), Apiaceae (7 spp., 2.0%), Asparagaceae (7 spp., 2.0%), Polygonaceae (7 spp.,

Table 1.—Physiognomic analysis of the vascular flora documented in Cooper Woods and Skinner Woods combined, Delaware County, Indiana. A = annual, B = biennial, H = herbaceous, P = perennial, W = woody. * = % of total = native plus non-native species.

	Native species summary		Exotic species summary	
	Number	% of Total*	Number	% of Total*
# of species	276	77.5%	80	22.5%
Tree	43	12.1%	6	1.7%
Shrub	14	3.9%	7	2.0%
W-Vine	6	1.7%	1	0.3%
H-Vine	3	0.8%	0	0.0%
P-Forbs	115	32.3%	23	6.5%
B-Forbs	5	1.4%	9	2.5%
A-Forbs	34	9.6%	13	3.7%
P-Grass	19	5.3%	9	2.5%
A-Grass	5	1.4%	12	3.4%
P-Sedge	28	7.9%	0	0.0%
A-Sedge	1	0.3%	0	0.0%
Ferns & Allies	3	0.8%	0	0.0%

2.0%), and Ranunculaceae (7 spp., 2.0%); these 12 families contained 197 (55.3%) of the 356 taxa documented (Appendix 1).

The physiognomic summary of the flora in CW-SW is presented in Table 1. Of the 356 taxa documented, 276 (77.5%) were native species and 80 (22.5%) were non-native species. Of the 356 total taxa, 77 (21.6) species were woody (e.g., trees, shrub, woody vines), 202 (56.8%) species were forbs and herbaceous vine, 74 (20.8) species were graminoid (e.g., grasses and sedges), and three (0.8%) species were ferns or fern allies (Table 1).

The Floristic Quality Index (FQI) and mean C for the combined CW-SW complex are listed in Table 2. For the combined native flora in both sites, the FQI and mean C was 55.0 and 3.4, respectively, and the FQI and mean C for total species (native and non-native) was 48.6 and 2.6, respectively.

Floristic quality Cooper Woods.—The vascular flora of Cooper Woods (CW) consisted of 332 taxa representing 214 genera in 85 families. The ten families with the highest number of taxa were Poaceae (42 spp., 12.7%), Asteraceae (38 spp., 11.5%), Cyperaceae (25 spp., 7.5%), Rosaceae (19 spp., 5.7%), Fabaceae (11 spp., 3.3%), Fagaceae (9 spp., 2.7%), Lamiaceae (9

Table 2.—Floristic quality matrices for Cooper Woods and Skinner Woods combined and separate, Delaware County, Indiana. All species = native species + non-native species.

Site	Number of species	FQI	Mean C
Cooper + Skinner Combined			
Native species	276	55.0	3.4
All species	356	48.6	2.6
Cooper Woods Only			
Native species	255	51.9	3.2
All species	332	45.4	2.5
Skinner Woods Only			
Native species	222	47.6	3.2
All species	272	43.0	2.6

spp., 2.7%), Apiaceae (7 spp., 2.1%), Polygonaceae (7 spp., 2.1%), and Ranunculaceae (7 spp., 2.1%). These ten families contained 174 (52.4%) of the 332 taxa documented (Appendix 1).

The physiognomic summary of the flora in CW is listed in Table 3. Of the 332 taxa documented, 255 (76.8%) were native species and 77 (23.2%) were non-native species. Of the 332 total taxa, 73 (22.0%) species were woody (e.g., trees, shrub, woody vines), 190 (57.2%) species were forbs and herbaceous vine, 67 (20.2%) species were grami-

Table 3.—Physiognomic analysis of the vascular flora documented in Cooper Woods, Delaware County, Indiana. A = annual, B = biennial, H = herbaceous, P = perennial, W = woody. * = % of total = native plus non-native species.

	Native species summary		Exotic species summary	
	Number	% of Total*	Number	% of Total*
# of species	255	76.8%	77	23.2%
Tree	41	12.3%	6	1.8%
Shrub	12	3.6%	7	2.1%
W-Vine	6	1.8%	1	0.3%
H-Vine	2	0.6%	0	0.0%
P-Forbs	108	32.5%	23	6.9%
B-Forbs	5	1.5%	9	2.7%
A-Forbs	30	9.0%	13	3.9%
P-Grass	19	5.7%	9	2.7%
A-Grass	5	1.5%	9	2.7%
P-Sedge	24	7.2%	0	0.0%
A-Sedge	1	0.3%	0	0.0%
Ferns & Allies	2	0.6%	0	0.0%

Table 4.—Physiognomic analysis of the vascular flora documented in Skinner Woods, Delaware County, Indiana. A = annual, B = biennial, H = herbaceous, P = perennial, W = woody. * = % of total = native plus non-native species.

	Native species summary		Exotic species summary	
	Number	% of Total*	Number	% of Total*
# of species	222	81.6%	50	18.4%
Tree	34	12.5%	2	0.7%
Shrub	13	4.8%	5	1.8%
W-Vine	6	2.2%	0	0.0%
H-Vine	1	0.4%	0	0.0%
P-Forbs	91	33.5%	14	5.1%
B-Forbs	5	1.8%	6	2.2%
A-Forbs	26	9.6%	9	3.3%
P-Grass	16	5.9%	7	2.6%
A-Grass	4	1.5%	7	2.6%
P-Sedge	23	8.5%	0	0.0%
A-Sedge	1	0.4%	0	0.0%
Ferns & Allies	2	0.7%	0	0.0%

noid (e.g., grasses and sedges), and two species (0.6%) were ferns or fern allies (Table 3).

The Floristic Quality Index (FQI) and mean C for CW are recorded in Table 2. For the native flora in CW, the FQI and mean C was 51.9 and 3.2, respectively, and the FQI and mean C for total species (native and non-native) was 45.4 and 2.5, respectively.

Floristic quality Skinner Woods.—The vascular flora of Skinner Woods (SW) consisted of 272 taxa representing 174 genera in 78 families. The ten families with the highest number of taxa were Poaceae (32 spp., 11.8%), Asteraceae (26 spp., 9.6%), Cyperaceae (23 spp., 8.5%), Rosaceae (19 spp., 7.0%), Fabaceae (10 spp., 3.7%), Brassicaceae (9 spp., 3.3%), Fagaceae (7 spp., 2.6%), Lamiaceae (6 spp., 2.2%), Polygonaceae (6 spp., 2.2%), and Juglandaceae (4 spp., 1.5%). These ten families contained 142 (52.2%) of the 272 taxa documented (Appendix 1).

The physiognomic summary of the flora in SW is recorded in Table 4. Of the 272 taxa documented, 222 (81.6%) were native species and 50 (18.4%) were non-native species. Of the 272 total taxa (native and non-native), 60 (22.1%) species were woody (e.g., trees, shrub, woody vines), 152 (55.9%) species were herbaceous forbs or vines, 58 (21.3%) species were graminoid (e.g., grasses

and sedges), and two species (0.7%) were ferns or fern allies.

The FQI and mean C for SW are documented in Table 2. For the native flora in SW, the FQI and mean C was 47.6 and 3.2, respectively, and the FQI and mean C for total species (native and non-native) was 43.0 and 2.6, respectively.

DISCUSSION

Inventory and floristic quality index.—The vascular plant taxa documented in CW and SW were typical of other floristic inventories of vegetation in east-central Indiana (Prast et al. 2013; Rothrock et al. 1993; Rothrock 1997; Ruch et al. 1998, 2002, 2004, 2007, 2008a, b, 2009, 2012, 2014a, b, 2015; Stonehouse et al. 2003, Tungesvick 2011). The twelve most dominant plant families, which include approximately 55% of the taxa reported at the CW-SW complex (e.g., 197 of 356 documented taxa (Appendix 1)) and 53–65% of taxa reported in the studies cited above, were the Apiaceae, Asteraceae, Brassicaceae, Caryophyllaceae, Cyperaceae, Fabaceae, Lamiaceae, Plantaginaceae, Poaceae, Polygonaceae, Ranunculaceae, and Rosaceae.

For the combined woodland complex, the floristic quality index (FQI) for the native flora was 55.0 and the mean C (mean coefficient of conservatism) was 3.4 (Table 2). The FQI and mean C for all plants, native plus non-native, was 48.6 and 2.6, respectively (Table 2). Analysis of these matrices provides an understanding regarding the quality of these sites. Swink & Wilhelm (1994) have suggested that sites with FQIs higher than 35 possess “sufficient conservatism and richness to be of profound importance from a regional perspective.” Ruch and colleagues have been using a native FQI ≥ 45 for designating a site as nature preserve quality. Thus, having a native FQI = 55.0 suggests that the CW-SW woodland complex is of nature preserve quality. However, does the low native mean C contradict this conclusion?

When comparing the FQI and mean C for 28 reference sites in Indiana, Rothrock & Homoya (2005) noted that the best quality woodland sites in the Central Till Plain, whether for historical or for innate biological reasons, have a limited number of species from the highest fidelity categories. They noted that for the best sites the native mean C is in the low 4 range, most sites fall between 3.8–4.1 (Rothrock & Homoya 2005). Our research team has conducted complete

inventories for more than 25 sites in east-central Indiana, and approximately a quarter of these have FQIs above 50 and native mean C's below 3.8, such as Wapi-nipi State Nature Preserve, formerly known as Coffman Woods Nature Preserve, (native FQI = 58.4, mean C = 3.5) in Wayne County (Ruch et al. 2014a), Munsee Woods State Nature Preserve (native FQI = 55.0, mean C = 3.2) in Delaware County (Prast et al. 2014), and McVey Memorial Forest (native FQI = 60.0, mean C = 3.5) in Randolph County (unpublished data). It was noted in these studies that the probable reason for the low native mean C was due to the limited number of species from the highest fidelity categories. For example, in the current study of the 276 native taxa documented from the CW-SW complex, only 22 species (8.0%) had C-values ≥ 7 , while 146 native species (52.9%) had C-values ≤ 3 and 108 native species (39.1%) had C-values of 4–6 (Appendix 1). Thus, it is not uncommon for sites in east-central Indiana (such as the CW-SW woodland complex, native mean C = 3.4) with FQIs above 50 to have native mean C values from 3.2–3.8 (see Ruch et al. 2014a).

What explains the pattern of reduced number of species in the highest fidelity categories and is it a historical as well as a contemporary pattern? In support of a historical pattern one might note the rather homogenous substrate and topography of the Tipton Till Plain forests. In the absence of a fen or other wetland supporting conservative species (e.g., Mounds State Park; see Rothrock et al. 1993) or the canyon walls seen in west-central Indiana (Hedge 1997), these woodlands have a limited suite of soil types and only modest differences in microclimates that might encourage a greater richness of conservative species. In the contemporary setting, floristically significant parcels may have fewer conservative species due to the size and setting. Unlike the large woodlands in the southern half of the state, the highest quality woodlands in east-central Indiana are typically small, isolated, and with urbanization, especially agriculture, contiguous with their borders. The small size results in woodlands having limited core as opposed to edge areas, altering patterns of species distributions and abundances (Whitney & Runkle 1981; Kupfer 1997). At the same time urbanization and agriculture create constant anthropogenic pressures resulting in changes in abiotic factors such as hydrological and nutrient cycles and biotic impact from herbivory and

introduced species (McKnight 1993; Honnay et al. 2002; Faulkner 2004; McKinney 2008).

In summary based on the native FQI and mean C, the CW-SW woodland complex is of nature preserve quality and does possess “sufficient conservatism and richness to be of profound importance from a regional perspective” as described by Swink & Wilhelm (1994). This woodland complex is an example of the highest quality woodland in central Delaware County.

Determining the floristic quality of a site typically entails first calculating the site’s metrics with only native species and then with all species, native and non-native. The difference between these can be used to document the impact that exotic species are having upon the site. Rothrock & Homoya (2005) have suggested that the natural quality of a site has been compromised when non-native diversity lowers the mean C by more than 0.7 units. In the CW-SW complex, the mean C was lowered 0.8 units. Thus, it can be presumed that the non-native flora is having a negative impact on the native flora. However, it should be pointed out that one cannot rely completely on the difference between the mean C-values when assessing the affect of the non-native species. In addition visual assessments must be included. A visual examination of the CW-SW complex revealed that the diversity of non-native species was concentrated on the margins of the site or in successional communities, such as old fields, that were only a part of the overall property. The interior woodlands at both sites contained very few species, or even total numbers, of non-natives. Similar observations were reported previously at Botany Glen (Stonehouse et al. 2003), Fall Creek Gorge (Rothrock & Homoya (2005), Ginn Woods (Ruch et al. 1998, 2004), Mounds State Park (Rothrock et al. 1993; Tungsveik 2011), and Lick Creek Summit Nature Preserve (Ruch et al. 2008a). In all these studies, even though the non-native species lowered mean C by at least 0.8 units, all reported that the integrity of the sites was not compromised. It is best summarized by Rothrock & Homoya (2005), “this amount of ‘exotics load,’ while of concern, may not be sufficient to detract from the preservation potential of a site.”

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APPENDIX 1

CATALOG OF VASCULAR FLORA AT COOPER WOODS AND SKINNER WOODS, DELAWARE COUNTY, INDIANA

(Arranged alphabetically by family in each phylum and class)

Species are listed alphabetically by family, then genera, under major plant groups. Non-native (exotic) species are capitalized. Nomenclature follows the Angiosperm Phylogeny Group III (APG 2009; Stevens 2015). Each species report contains the following information: (1) current scientific name; (2) location (CW = Cooper Woods, SW = Skinner Woods); (3) vegetation association (CB = creek bank, MWL = mature woodland, OF = old-field, SUCWL = early successional woodland, VP = vernal pool, and WE = woodland edge); (4) a visual estimate of its relative abundance (see below); (5) the Indiana Coefficient of Conservation, C-value (Rothrock 2004); and (6) the Ball State University Herbarium (BSUH) number(s). The relative abundance for species is defined as follows; rare = ≤ 5 sites although a species may be abundant at one site; infrequent = occasional, not widespread throughout its potential habitats, but may be locally abundant at a site; common = frequent throughout its potential habitats and may be locally abundant at one or more sites; and abundant = common and numerous throughout its potential habitats. Potential Delaware County records are indicated by a pound-symbol in parentheses (#) immediately preceding a species. There are five Delaware County records. Species were deemed unreported for Delaware County, and hence considered a county record, if they did not appear in the Indiana Natural Heritage Data Center’s records for Delaware County [this is the same plant list in the computer database of Keller et al. (1984)], the USDA Plant Database (2017), The Biota of North America Program (BONAP): Maps by States and Provinces (2014), Overlease & Overlease (2007), Deam (1940), the species listed at Dutro Woods Nature Preserve (Ruch et al.

2014b), Ginn Woods (Ruch et al. 1998, 2004), and Munsee Woods Nature Preserve (Prast et al. 2013). Lastly, no species documented at CW-SW occur on the Indiana Department of Natural Resources list of endangered, threatened or rare plants, but two species, *Rudbeckia fulgida* var. *fulgida* and *Spiranthes ovalis* var. *erostellata*, are on the state watch list (IDNR Nature Preserves 2016).

MONILOPHYTA

CYSTOPTERIDACEAE

Cystopteris protrusa (Weath.) Blasdel – CW, SW; MWL, SUCWL; Common; C = 4; BSUH 19775, 19667.

EQUISETACEAE (Horsetail Family)

Equisetum arvense L. – CW; MWL; Rare; C = 1; BSUH 20050.

OPHIOGLOSSACEAE

(Adder's-tongue Family)

Sceptridium dissectum (Spreng.) Lyon forma *obliquum* – SW; MWL- near trash pile; Rare; C = 3; BSUH 19990.

CONIFEROPHYTA

CUPRESSACEAE

(Redwood or Cypress Family)

Juniperus virginiana L. var. *virginiana* – CW, SW; OF; Rare; C = 2; BSUH 19549.

PINACEAE (Pine Family)

PINUS RESINOSA Aiton – CW; SUCWL; Rare; C = 0; BSUH 19953.

Pinus strobus L. – CW; WE, SUCWL; Rare; C = 5; BSUH 19703.

PINUS SYLVESTRIS L. – CW; WE; Rare; C = 0; BSUH 19944, 19484.

MAGNOLIOPHYTA

MAGNOLIOPSIDA (Dicotyledons)

ACANTHACEAE (Acanthus Family)

Ruellia strepens L. – CW, SW; MWL, WE; Common; C = 4; BSUH 20037.

ADOXACEAE (Moschatel Family)

Sambucus canadensis L. – CW, SW; MWL; Infrequent; C = 2; BSUH 20035.

AMARANTHACEAE

(Goosefoot or Pigweed Family)

AMARANTHUS RETROFLEXUS L. – CW; WE/OF; Infrequent but locally common; C = 0; BSUH 20068.

Amaranthus tuberculatus (Moquin-Tandon) J.D. Sauer – CW; CB, WE/OF; Infrequent; C = 1; BSUH 19884.

CHENOPODIUM ALBUM L. – CW, SW; OF, WE, SUCWL, VP; Infrequent; C = 0; BSUH 20054.

ANACARDIACEAE (Cashew Family)

Toxicodendron radicans (L.) Kuntze var. *negundo* (Greene) Reveal – CW, SW; All habitats; Common; C = 1; BSUH 20049.

ANNONACEAE (Custard apple Family)

Asimina triloba (L.) Dunal – CW; SUCWL; Rare; C = 6; BSUH 19989.

APIACEAE (Carrot or Parsley Family)

Cryptotaenia canadensis (L.) DC. – CW, SW; MWL; Abundant; C = 3; BSUH 20046.

DAUCUS CAROTA L. – CW, SW; WE, OF; Rare; C = 0; BSUH 19733.

Erigenia bulbosa (Michx.) Nutt. – CW, SW; MWL; Infrequent; C = 5; BSUH 19742.

Osmorhiza claytonii (Michx.) C.B. Clarke – CW, SW; MWL; Common; C = 3; BSUH 20009.

Sanicula odorata (Raf.) K.M. Pryer & L.R. Phillippe – CW, SW; MWL; Common and locally abundant; C = 2; BSUH 19995.

Thaspium trifoliatum (L.) A. Gray var. *aureum* (L.) Britton – CW, SW; MWL; C = 5; BSUH 20019.

Zizia aurea (L.) W.D.J. Koch – CW; WE; C = 7; BSUH 20010.

APOCYNACEAE

(Dogbane and Milkweed Family)

Apocynum cannabinum L. – CW, SW; OF, WE; Infrequent; C = 2; BSUH 19772.

Asclepias incarnata L. var. *incarnata* – CW, SW; CB near pond; Rare; C = 4; BSUH 19734.

- Asclepias syriaca* L. – CW, SW; WE/OF; C = 1; BSUH 19751.
- ARISTOLOCHIACEAE (Birthwort Family)
Aristolochia serpentaria L. (= *Endodeca serpentaria* (L.) Raf.) – CW; MWL; Rare; C = 8; BSUH 19786.
- ASTERACEAE (Aster or Sunflower Family)
Ageratina altissima (L.) R.M. King & H. Rob. var. *altissima* – CW; MWL; Rare; C=2; BSUH 19074, 19718.
- Ambrosia artemisiifolia* L. var. *elatior* (L.) Descourt. – CW, SW; OF, SUCWL; Infrequent ; C = 0; BSUH 19708.
- Ambrosia trifida* L. – CW, SW; MWL; Infrequent but locally common; C = 0; BSUH 20061.
- Bidens bipinnata* L. – CW; WE/OF; Rare; C = 0; BSUH 19722, 19821.
- Bidens frondosa* L. – CW, SW; MWL; Infrequent; C = 1; BSUH 20065.
- Bidens vulgata* Greene – CW; MWL, SUCWL; Common and locally abundant; C = 0; BSUH 19832.
- CICHORIUM INTYBUS L. – CW, SW; WE, OF; Rare; C = 0; BSUH 19767.
- CIRSIIUM ARVENSE (L.) Scop. – CW, SW; OF, WE; C = 0; BSUH 19770.
- Cirsium discolor* (Muhl. ex Willd.) Spreng. – CW, SW; OF; Infrequent; C = 3; BSUH 20064.
- Conyza canadensis* (L.) Cronquist var. *canadensis* – CW, SW; OF, WE; Common; C = 0; BSUH 19706.
- Erechtites hieraciifolius* (L.) Raf. ex DC. – CW; SUCWL; Rare; C = 2; BSUH 19817.
- Erigeron annuus* (L.) Pers. – CW, SW; MWL; Infrequent; C = 0; BSUH 20047.
- Erigeron philadelphicus* L. var. *philadelphicus* – CW, SW; MWL; Infrequent; C = 3; BSUH 19997.
- Eupatorium altissimum* L. – CW, SW; OF; Common; C = 1; BSUH 19714.
- Eupatorium perfoliatum* L. – CW; SUCWL; Rare; C = 4; BSUH 20059.
- Eupatorium serotinum* Michx. – CW, SW; WE, OF; Common; C = 0; BSUH 19723.
- Euthamia graminifolia* (L.) Nutt. – CW, SW; OF; Common; C = 3; BSUH 20058.
- Helianthus decapetalus* L. – CW; WE, MWL; Infrequent; C = 5; BSUH 19827, 19874.
- Helianthus tuberosus* L. – CW; WE, MWL; Rare but locally common; C = 2; BSUH 19826.
- Lactuca floridana* (L.) Gaerth. – CW, SW; MWL, SUCWL; Infrequent; C = 5; BSUH 20062.
- LACTUCA SERRIOLA L. – CW; WE; Rare; C = 0; BSUH 19715.
- Packera glabella* (Poir.) C. Jeffrey – CW, SW; MWL, WE; Infrequent; C = 0; BSUH 19994.
- Packera obovata* (Willd.) W.A. Weber & Á. Löve – CW, SW; MWL; Common; C = 7; BSUH 19803.
- Ratibida pinnata* (Vent.) Barnhart – CW; OF; Rare; C = 5; BSUH 19798.
- (#) *Rudbeckia fulgida* Aiton var. *fulgida* – CW; MWL; Infrequent but locally common; C = 5; BSUH 19843. (State Watch List; S2=imperiled in state)
- Rudbeckia hirta* L. – CW, SW; OF, SUCWL; Common; C = 2; BSUH 19795.
- Rudbeckia triloba* L. var. *triloba* – CW; WE; Rare; C = 3; BSUH 19878.
- Solidago altissima* L. – CW, SW; OF, SUCWL, WE; Abundant; C = 0; BSUH 19719.
- Symphyotrichum cordifolium* (L.) G.L. Nesom – CW, SW; MWL, WE; Common; C = 5; BSUH 19823.
- Symphyotrichum lanceolatum* (Willd.) G.L. Nesom var. *lanceolatum* – CW; OF (west ditch bank); Rare; C = 3; BSUH 20067.

Symphytotrichum lateriflorum (L.) Á. Löve & D. Löve var. *lateriflorum* – CW, SW; All habitats; Abundant; C = 3; BSUH 19717.

Symphytotrichum novae-angliae (L.) G.L. Nesom – CW, SW; OF; Common; C = 3; BSUH 19738.

Symphytotrichum pilosum (Willd.) G.L. Nesom var. *pilosum* – CW, SW; OF; Common; C = 0; BSUH 19716.

TARAXACUM OFFICINALE G.H. Weber ex F.H. Wiggers – CW, SW; CB, OF, MWL, WE; Infrequent; C = 0; BSUH 19690.

TRAGOPOGON PRATENSIS L. – CW, MWL, Rare C = 0; BSUH 20025.

Verbesina alternifolia (L.) Britton ex Kearney – CW, SW; SUCWL, WE; Infrequent; C = 3; BSUH 19986.

Vernonia gigantea (Walter) Trel. – CW, SW; OF; Common; C = 2; BSUH 19701.

Xanthium strumarium L. – CW, SW; OF, WE; Rare; C = 0; BSUH 19837.

BALSAMINACEAE (Touch-me-not Family)
Impatiens capensis Meerb. – CW, SW; MWL, SUCWL; Abundant; C = 2; BSUH 19756.

BERBERIDACEAE (Barberry Family)
Jeffersonia diphylla (L.) Pers. – CW; MWL; Rare but locally abundant; C = 7; BSUH 19688.

Podophyllum peltatum L. – CW, SW; MWL; Abundant; C = 3; BSUH 19765.

BETULACEAE (Birch Family)
Carpinus caroliniana Walter ssp. *virginiana* (Marshall) Furlow – CW, SW; MWL; Infrequent; C = 5; BSUH 20041.

Corylus americana Walter – SW; MWL; Rare; C = 4; BSUH 19813.

Ostrya virginiana (Mill.) K. Koch – CW, SW; MWL; Common; C = 0; BSUH 20042.

BIGNONIACEAE (Trumpet-Creeper Family)
Catalpa speciosa (Warder) Engelm. – CW; SUCWL; Rare; C = 0; BSUH 19984.

BORAGINACEAE (Borage Family)
Hackelia virginiana (L.) I.M. Johnst. – CW, SW; MWL; Rare; C = 0; BSUH 19791.

Mertensia virginica (L.) Link – CW, SW; MWL; Rare; C = 6; BSUH 19678.

BRASSICACEAE (Mustard Family)
ALLIARIA PETIOLATA (M. Bieb.) Cavara & Grande – CW, SW; MWL; Infrequent; C = 0; BSUH 19692.

BARBAREA VULGARIS W.T. Aiton – CW, SW; OF, SUCWL; Infrequent; C = 0; BSUH 19806.

Cardamine bulbosa (Schreb. ex Muhl.) Britton, Sterns & Poggenb. – CW, SW; MWL, SUCWL; Common; C = 4; BSUH 19693.

Cardamine concatenata (Michx.) O. Schwarz – CW, SW; MWL; Abundant; C = 4; BSUH 19750.

Cardamine douglassii Britton – CW, SW; MWL; Common; C = 5; BSUH 19694.

(#) *Cardamine parviflora* L. var. *arenicola* (Britton) O.E. Schulz – SW; WE/OF; Common; C = 4; BSUH 19863.

Cardamine pensylvanica Willd. – CW, SW; MWL; Rare; C = 2; BSUH 19777.

Rorippa palustris (L.) Besser ssp. *fernaldiana* (Butters & Abbe) Jonsell – CW, SW; OF, WE; Rare but locally frequent; C = 2; BSUH 19920.

Rorippa sessiliflora (Nutt.) A.S. Hitchc. – SW; WE/OF; Infrequent; C = 3; BSUH 19864.

CAMPANULACEAE (Bellflower Family)
Campanula americana L. (= *Campanulastrum americanum* (L.) Small) – CW; MWL; Rare; C = 4; BSUH 19957.

Lobelia inflata L. – CW, SW; MWL, OF, WE; Common and locally abundant; C = 3; BSUH 19705, 19861.

Lobelia siphilitica L. – CW, SW; WE; Common; C = 3; BSUH 19705.

- CAPRIFOLIACEAE (Honeysuckle Family)
LONICERA MAACKII (Rupr.) Maxim. – CW, SW; MWL; Abundant; C = 0; BSUH 19675.
- LONICERA MORROWII* A. Gray – CW, SW; OF; Common; C = 0; BSUH 19941.
- Triosteum aurantiacum* E.P. Bicknell var. *aurantiacum* – SW; MWL; Rare; C = 5; BSUH 19812.
- VIBURNUM OPULUS* var. *OPULUS* – CW, SW; MWL; Infrequent; C = 0; BSUH 19993.
- Viburnum prunifolium* L. – CW, SW; MWL; Common and locally abundant; C = 4; BSUH 19800, 19663.
- CARYOPHYLLACEAE (Pink Family)
CERASTIUM FONTANUM Baumg. ssp. *VULGARE* (Hartm.) Greuter & Burdet – CW, SW; OF; Abundant; C = 0; BSUH 19938, 20005.
- DIANTHUS ARMERIA* L. – CW; MWL; Rare; C = 0; BSUH 20052.
- Silene stellata* (L.) W.T. Aiton – CW, SW; MWL; Rare but locally common; C = 5; BSUH 19959.
- Silene virginica* L. – CW, SW; MWL; Rare; C = 7; BSUH 19980.
- STELLARIA MEDIA* (L.) Vill. – CW; SUCWL, WE; C = 0; BSUH 20069.
- CELASTRACEAE (Bittersweet Family)
CELASTRUS ORBICULATUS Thunb. – CW; MWL; Infrequent [to rare]; C = 0; BSUH 20036.
- Euonymus atropurpureus* Jacq. var. *atropurpureus* – CW, SW; MWL; Infrequent; C = 5; BSUH 19696.
- Euonymus obovata* Nutt. – CW, SW; MWL; Common; C = 0; BSUH 19797.
- CONVOLVULACEAE
 (Morning-Glory Family)
Calystegia sepium (L.) R. Br. – CW, SW; MWL, WE; Infrequent but locally common; C = 1; BSUH 19769.
- IPOMOEA HEDERACEA* Jacq. – CW; WE; Rare; C = 0; BSUH 20063.
- CORNACEAE (Dogwood Family)
Cornus drummondii C.A. Mey. – CW, SW; MWL/WE; Common; C = 2; BSUH 19983.
- Cornus obliqua* Raf. – CW; OF, SUCWL; Infrequent; C = 5; BSUH 19882.
- DIPSACACEAE
DIPSACUS FULLONUM L. – CW, SW; OF, SUCWL, WLE; Infrequent; C = 0; BSUH 19965.
- ELAEAGNACEAE (Oleaster Family)
ELEAGNUS UMBELLATA Thunb. – CW, SW; OF, WE; Infrequent; C = 0; BSUH 20004.
- EUPHORBIACEAE (Spurge Family)
Acalypha rhomboidea Raf. – CW, SW; All habitats; Abundant; C = 0; BSUH 19828.
- Euphorbia dentata* Michx. – CW; WE; Rare; C = 0; BSUH 19815.
- Euphorbia maculata* L. – CW; OF, WE; Rare; C = 0; BSUH 19699.
- Euphorbia nutans* Lag. – CW, SW; OF; Infrequent; C = 0; BSUH 19814.
- FABACEAE (Legume Family)
Cercis canadensis L. – CW, SW; MWL, SUCWL; Infrequent; C = 3; BSUH 19684, 19808.
- Desmodium canadense* (L.) DC. – CW, SW; OF; Infrequent but locally common; C = 3; BSUH 19834.
- Desmodium paniculatum* (L.) DC. var. *paniculatum* – CW, SW; OF; Infrequent to common in the fields; C = 2; BSUH 19713.
- Gleditsia triacanthos* L. – CW, SW; MWL; Rare to infrequent; C = 1; BSUH 19937.
- (#) *LATHYRUS LATIFOLIUS* L. – CW; OF; Rare; C = 0; BSUH 19793.
- MEDICAGO LUPULINAL* L. – CW, SW; MWL, SUCWL; Rare; C = 0; BSUH 20039.
- MELILOTUS OFFICINALIS* (L.) Lam. – CW, SW; MWL, WE; Infrequent but locally common; C = 0; BSUH 19782.

SECURIGERA VARIA (L.) Lassen – CW; OF; Rare; C = 0; BSUH 20057.

TRIFOLIUM HYBRIDUM L. – CW, SW; OF, SUCWL, WE; Common; C = 0; BSUH 20038.

TRIFOLIUM PRATENSE L. – CW, SW; OF, SUCWL, WE; Common; C = 0; BSUH 19763.

TRIFOLIUM REPENS L. – CW, SW; OF; Infrequent; C = 0; BSUH 19982.

FAGACEAE (Beech Family)

Fagus grandifolia Ehrh. – CW, SW; MWL; Infrequent; C = 8; BSUH 20028.

Quercus bicolor Willd. – CW, SW; MWL; Infrequent; C = 7; BSUH 19730, 19973.

Quercus imbricaria Michx. – CW; MWL, WE; Rare; C = 3; BSUH 19726.

Quercus macrocarpa Michx. var. *macrocarpa* – CW, SW; MWL, WLE; Abundant; C = 5; BSUH 19663.

Quercus muehlenbergii Engelm. – CW, SW; SUCWL; Infrequent; C = 4; BSUH 20012.

Quercus palustris Münchh. – CW, SW; MWL; Rare; C = 3; BSUH 20027.

Quercus rubra L. – CW, SW; MWL; Abundant; C = 4; BSUH 19974.

Quercus shumardii Buckley – CW, SW; MWL; Abundant; C = 7; BSUH 19975.

Quercus velutina Lam. – CW; MWL; Rare; C = 4; BSUH 19819.

FUMARIACEAE (Fumitory Family)

Dicentra cucullaria (L.) Bernh. – CW, SW; MWL; Infrequent; C = 6; BSUH 19744.

GERANIACEAE (Geranium Family)

Geranium maculatum L. – CW, SW; MWL; Common; C = 4; BSUH 19781.

GROSSULARIACEAE (Gooseberry Family)

Ribes cynosbati L. – CW, SW; MWL; Infrequent; C = 4; BSUH 19801.

HYPERICACEAE (St. John's-wort Family)
Hypericum mutilum L. – SW; WE/OF; Infrequent but locally common; C = 4; BSUH 19709.

HYPERICUM PERFORATUM L. – CW, SW; MWL; Rare C = 0; BSUH 19784.

Hypericum punctatum Lam. – CW, SW; OF; Common; C = 3; BSUH 19729.

HYDROPHYLLACEAE (Waterleaf family)
Hydrophyllum macrophyllum Nutt. – CW, SW; MWL; Rare; C = 7; BSUH 19996.

Hydrophyllum virginianum L. – CW, SW; MWL; Abundant; C = 4; BSUH 19999.

JUGLANDACEAE (Walnut Family)

Carya cordiformis (Wangenh.) K. Koch – CW, SW; MWL; Infrequent; C = 5; BSUH 19787.

Carya glabra (P. Miller) Sweet – CW, SW; MWL; Infrequent; C = 4; BSUH 19788.

Carya laciniosa (Michx. f.) G. Don – CW, SW; MWL; Abundant; C = 8; BSUH 19846.

Carya ovata (P. Miller) K. Koch – CW, SW; MWL, SUCWL; Common; C = 4; BSUH 19960.

Juglans nigra L. – CW, SW; MWL, SUCWL, WE; Infrequent to common C = 2; BSUH 20013.

LAMIACEAE (Mint Family)

Blephilia hirsuta (Pursh) Benth. – CW, SW; MWL; Rare but locally common; C = 5; BSUH 19905, 19724.

(#) *CHAITURUS MARRUBIASTRUM* (L.) Rchb. – CW; WE; Rare but locally common; C = 0; BSUH 19721, 19876.

GLECHOMA HEDERACEA L. – CW, SW; MWL; Rare; C = 0; BSUH 19761.

Lycopus americanus Muhl. ex W.P.C. Barton – CW, SW; CB, OF; Common to abundant; C = 3; BSUH 19704.

Monarda fistulosa L. var. *fistulosa* – CW; OF; Infrequent; C = 3; BSUH 19792.

PRUNELLA VULGARIS L. – CW, SW; OF, SUCWL; Common; C = 0; BSUH 19736.

Scutellaria lateriflora L. – CW, SW; MWL, VP; Common; C = 4; BSUH 19789.

Stachys tenuifolia Willd. – CW; MWL, VP; Common; C = 4; BSUH 19880, 19954.

Teucrium canadense L. – CW, SW; MWL, WE; Infrequent but locally common; C = 3; BSUH 19735.

LAURACEAE (Laurel Family)

Lindera benzoin (L.) Blume var. *benzoin* – CW, SW; WE, VP; Rare; C = 5; BSUH 19683, 19711.

LIMNANTHACEAE (False Mermaid Family)
Floerkea proserpinacoides Willd. – CW; MWL; Common; C = 5; BSUH 19691.

LINDERNIACEAE (False Pimpernel Family)

Lindernia dubia (L.) Pennell var. *anagallidea* (Michx.) Cooperr. – CW, SW; OF; Rare but locally frequent; C = 3; BSUH 19727.

MAGNOLIACEAE (Magnolia Family)

Liriodendron tulipifera L. – CW; OF, SUCWL; Rare; C = 4; BSUH 19697.

MALVACEAE (Mallow Family)

ABUTILON THEOPHRASTI Medik. – CW, SW; OF (disturbed soil near soil pit), WE; Rare; C = 0; BSUH 19702, 19720.

HIBISCUS TRIONUM L. – CW, SW; WE, CB near pond; Rare; C = 0; BSUH 19707, 19839.

SIDA SPINOSA L. – CW, SW; OF, WE; Rare; C = 0; BSUH 19879.

Tilia americana L. var. *americana* – CW; MWL; Rare but locally frequent; C = 5; BSUH 19820.

MENISPERMACEAE (Moonseed Family)

Menispermum canadense L. – CW, SW; MWL; Common; C = 3; BSUH 19670.

MONTIACEAE (Blinks Family)

Claytonia virginica L. var. *virginica* – CW, SW; MWL; Abundant; C = 2; BSUH 19749.

MORACEAE (Mulberry Family)

MORUS ALBA L. – CW, SW; MWL, SUCWL, WE; C = 0; BSUH 20031.

Morus rubra L. – SW; MWL; Rare; C = 4; BSUH 19732.

MYRSINACEAE

Lysimachia ciliata L. – CW, SW; OF; Rare but locally common; C = 4; BSUH 19966.

OLEACEAE (Olive Family)

Fraxinus americana L. – SW; WE; Rare; C = 4; BSUH 19886.

Fraxinus nigra Marshall – CW, SW; MWL around vernal pools; Infrequent; C = 7; BSUH 19731.

Fraxinus pennsylvanica Marshall – CW, SW; MWL, SUCWL; Infrequent; C = 1; BSUH 19662, 19961. [NOTE: prior to the emerald ash bore invasion, this was the most abundant tree species in both woodlands]

LIGUSTRUM OBTUSIFOLIUM Siebold & Zucc. – CW, SW; MWL; Infrequent; C = 0; BSUH 20040.

ONAGRACEAE (Evening Primrose Family)
Circaea lutetiana L. ssp. *canadensis* (L.) Asch. & Magnus – CW, SW; MWL; Abundant; C = 2; BSUH 19783.

Epilobium coloratum Biehler – CW, SW; CB near pond, WE; Infrequent; C = 3; BSUH 19829.

Ludwigia palustris (L.) Elliott – SW; VP; Rare but locally abundant; C = 3; BSUH 19867, 19971.

Oenothera biennis L. – CW, SW; OF, WE; Rare; C = 0; BSUH 19773.

OXALIDACEAE (Wood Sorrel Family)

Oxalis dillenii Jacq. – CW, SW; MWL; Abundant; C = 0; BSUH 19955.

Oxalis stricta L. – CW, SW; MWL; Infrequent; C = 0; BSUH 19728.

PAPAVERACEAE (Poppy Family)

Sanguinaria canadensis L. – CW, SW; MWL; Common; C = 5; BSUH 19677, 19762.

- Stylophorum diphyllum* (Michx.) Nutt. – CW, SW; MWL; Rare; C = 7; BSUH 19810.
- PENTHORACEAE (Ditch stonecrop Family)
Penthorum sedoides L. – CW, SW; SUCWL, VP; Rare but locally common; C = 8; BSUH 19831.
- PHRYMACEAE (Lopseed Family)
Mimulus alatus Aiton – CW, SW; MWL, VP; Infrequent but locally common; C = 4; BSUH 19700.
- Phryma leptostachya* L. – CW, SW; MWL; Infrequent; C = 4; BSUH 20053.
- PLANTAGINACEAE (Plantain Family)
Gratiola neglecta Torr. – CW, SW; SUCWL, WE/OF; Infrequent but locally common; C = 4; BSUH 19757.
- Penstemon calycosus* Small – CW, SW; MWL; Infrequent; C = 4; BSUH 20023.
- PLANTAGO LANCEOLATA L. – CW, SW; OF, WE; Common; C = 0; BSUH 19753.
- Plantago rugelii* Decne. – CW, SW; MWL, SUCWL, WE; Abundant; C = 0; BSUH 19768.
- Veronica peregrina* L. var. *peregrina* – SW; WE/OF; Infrequent but locally common; C = 0; BSUH 19848, 19862.
- PLATANACEAE (Plane-Tree Family)
Platanus occidentalis L. – CW; MWL, SUCWL; Rare; C = 3; BSUH 20030.
- POLEMONIACEAE (Phlox Family)
Phlox divaricata L. ssp. *divaricata* – CW, SW; MWL; Abundant; C = 5; BSUH 19802.
- Polemonium reptans* L. var. *reptans* – CW, SW; MWL; Infrequent; C = 5; BSUH 19804.
- POLYGONACEAE (Smartweed Family)
Fallopia scandens (L.) Holub – CW, SW; MWL/CB; Rare; C = 0; BSUH 19740.
- PERSICARIA LONGISETA (de Bruijn) Kitag. – CW, SW; MWL, SUCWL; Common; C = 0; BSUH 20060.
- PERSICARIA MACULOSA Gray – CW, SW; MWL; Infrequent; C = 0; BSUH 19836.
- Persicaria punctata* (Elliott) Small – CW, SW; SUCWL, VP; Infrequent; C = 3; BSUH 19847.
- Persicaria virginiana* (L.) Gaertn. – CW, SW; MWL; Abundant; C = 3; BSUH 19790.
- POLYGONUM AVICULARE L. – CW, SW; WE; Common; C = 0; BSUH 19962.
- RUMEX CRISPUS L. – CW, SW; MWL, SUCWL; Common; C = 0; BSUH 20045.
- PORTULACACEAE (Purslane Family)
 PORTULACA OLERACEA L. – CW; MWL; Rare; C = 0; BSUH 19838.
- RANUNCULACEAE (Buttercup Family)
 ERANTHIS HYEMALIS (L.) Salisb. – CW; MWL [planted and naturalized]; Rare but locally abundant; C = 0; BSUH 19747.
- Hepatica acutiloba* D.C. – CW; MWL near vernal pool; Rare; C = 8; BSUH 19746.
- Ranunculus abortivus* L. – CW, SW; MWL; Infrequent; C = 0; BSUH 19669, 19796.
- Ranunculus hispidus* Michx. var. *caricetorum* (Greene) T. Duncan – CW, SW; VP; Abundant; C = 10; BSUH 19902, 19807.
- Ranunculus hispidus* Michx. var. *hispidus* – CW; MWL; Infrequent; C = 7; BSUH 19875.
- Thalictrum revolutum* DC. – CW; WE; Rare; C = 5; BSUH 19741.
- Thalictrum thalictroides* (L.) Eames & B. Boivin – CW; MWL; Infrequent; C = 7; BSUH 19664.
- RHAMNACEAE (Buckthorn Family)
 RHAMNUS CATHARTICA L. – CW; MWL, SUCWL, WE; Infrequent; C = 0; BSUH 20014.
- ROSACEAE (Rose Family)
Agrimonia parviflora Aiton – CW, SW; OF; Infrequent but locally abundant; C = 4; BSUH 19794.
- Agrimonia pubescens* Wallr. – CW, SW; MWL; Infrequent; C = 5; BSUH 19956.
- Crataegus crus-galli* L. – CW, SW; OF; Rare; C = 4; BSUH 19943.

- Crataegus mollis* (Torr. & A. Gray) Scheele – CW, SW; MWL; Infrequent; C = 2; BSUH 19991, 20020.
- Crataegus phaenopyrum* (L. f.) Medik. – SW; OF; Rare (one tree); C = 0; BSUH 19856.
- Crataegus punctata* Jacq. – CW, SW; MWL; Rare; C = 2; BSUH 20044.
- Fragaria virginiana* P. Miller – CW, SW; OF; Infrequent but locally common; C = 2; BSUH 19940.
- Geum canadense* Jacq. var. *canadense* – CW, SW; MWL; Abundant; C = 1; BSUH 19764.
- Geum laciniatum* Murray – CW, SW; CB, OF, MWL; Abundant; C = 3; BSUH 20055.
- Geum vernum* (Raf.) Torr. & A. Gray – CW, SW; CB; OF, MWL, SUCWL; Abundant; C = 1; BSUH 19766, 19809, 19758.
- MALUS PUMILA* P. Miller – CW, SW; OF; Rare; C = 0; BSUH 19811.
- Potentilla norvegica* L. – CW, SW; OF; Rare; C = 0; BSUH 19833.
- Potentilla simplex* Michx. – CW, SW; MWL; Common but locally abundant; C = 2; BSUH 19666.
- Prunus americana* Marshall – CW, SW; MWL; Rare; C = 4; BSUH 20029.
- Prunus serotina* Ehrh. – CW, SW; MWL; Infrequent; C = 1; BSUH 19936, 20001.
- PYRUS CALLERYANA* Decne. – CW; OF, SUCWL; Infrequent; C = 0; BSUH 20033.
- ROSA MULTIFLORA* Thunb. ex Murray – CW, SW; OF, SUCWL; C = 0; BSUH 20018.
- Rosa setigera* Michx. – CW, SW; OF, SUCWL, WE; Infrequent; C = 4; BSUH 19752.
- Rubus occidentalis* L. – CW, SW; CB, OF; Infrequent; C = 1; BSUH 19942, 20003.
- Rubus pensilvanicus* Poir. – CW, SW; CB; SUCWL; Common; C = 5; BSUH 20011.
- RUBIACEAE (Madder Family)
- Galium aparine* L. – CW, SW; CB; MWL, SUCWL; Common; C = 1; BSUH 19779.
- Galium circaezans* Michx. – CW, SW; CB, MWL; Common; C = 7; BSUH 20022.
- Galium concinnum* Torr. & A. Gray – CW, SW; MWL; Common; C = 5; BSUH 20048.
- Galium obtusum* Bigelow var. *obtusum* – CW, SW; MWL around vernal pools; Common; C = 5; BSUH 19903.
- RUTACEAE (Citrus Family)
- Zanthoxylum americanum* P. Miller – CW, SW; SUCWL; Infrequent but locally common; C = 3; BSUH 20015.
- SALICACEAE (Willow Family)
- Populus deltoides* Marshall – CW, SW; SUCWL; Infrequent; C = 1; BSUH 20017.
- SAPINDACEAE (Soapberry Family)
- Acer negundo* L. var. *negundo* – CW, SW; MWL near vernal pools; Rare; C = 1; BSUH 19890.
- Acer rubrum* L. var. *rubrum* – CW; MWL; Infrequent; C = 5; BSUH 19976.
- Acer saccharum* Marshall – CW, SW; MWL; Abundant; C = 4; BSUH 19661, 19776.
- Acer saccharinum* L. – CW, SW; Common; C = 1; BSUH 19998.
- Aesculus glabra* Willd. var. *glabra* – CW, SW; MWL; Abundant; C = 5; BSUH 19799.
- SOLANACEAE (Nightshade Family)
- DATURA STRAMONIUM* L. – CW; OF - distured area; Rare; C = 0; BSUH 19698.
- Solanum carolinense* L. var. *carolinense* – CW, SW; OF; Infrequent; C = 0; BSUH 19710.
- Solanum ptycanthum* Dunal – CW, SW; MWL; Rare but locally common; C = 0; BSUH 20056.
- STAPHYLEACEAE (Bladdernut Family)
- Staphylea trifolia* L. – SW; MWL; Rare but locally common; C = 5; BSUH 19737.

THEOPHRASTACEAE

Samolus parviflorus Raf. – CW, SW; SUCWL, WE/OF; Rare but locally abundant; C = 5; BSUH 19981.

ULMACEAE (Elm Family)

Celtis occidentalis L. – CW, SW; MWL; Common; C = 3; BSUH 20032.

Ulmus americana L. – CW, SW; MWL; Common; C = 3; BSUH 19816, 19668.

ULMUS PUMILA L. – CW, SW; OF, SUCWL; Infrequent; C = 0; BSUH 19755.

Ulmus rubra Muhl. – CW, SW; MWL; Infrequent; C = 3; BSUH 20026.

URTICACEAE (Nettle Family)

Boehmeria cylindrica (L.) Sw. – CW, SW; VP; Common; C = 3; BSUH 19774, 19877.

Laportea canadensis (L.) Wedd. – CW, SW; MWL; Infrequent but locally common; C = 2; BSUH 19958.

Pilea pumila (L.) A. Gray var. *pumila* – CW, SW; MWL, SUCWL; Infrequent but locally common; C = 2; BSUH 19739.

VALERIANACEAE

Valerianella umbilicata (Sull.) Alph. Wood – CW, SW; OF; Infrequent; C = 5; BSUH 19939.

VERBENACEAE (Vervain Family)

Verbena urticifolia L. – CW, SW; MWL; Infrequent; C = 3; BSUH 19785.

VIOLACEAE (Violet Family)

Viola sororia Willd. – CW, SW; SUCWL; Infrequent; C = 1; BSUH 19780.

Viola striata Aiton – CW, SW; MWL; Infrequent; C = 4; BSUH 19689.

VITACEAE (Grape Family)

Parthenocissus quinquefolia (L.) Planch. – CW, SW; MWL; Abundant; C = 2; BSUH 19671.

Vitis riparia Michx. – CW, SW; SUCWL; Common; C = 1; BSUH 19934.

Vitis vulpina L. – CW, SW; WE Infrequent; C = 3; BSUH 19695.

MAGNOLIOPHYTA

LILIOPSIDA (Monocotyledons)

ALISMATACEAE (Water-plantain Family)

Alisma subcordatum Raf. – SW; VP; Rare; C = 2; BSUH 19712.

AMARYLLIDACEAE (Amaryllis Family)

Allium canadense L. var. *canadense* – CW, SW; MWL; Rare but locally abundant; C = 1; BSUH 19665.

Allium tricoccum Aiton [SYN: *Allium burdickii* (Hanes) A.G. Jones] – CW; MWL; Infrequent but locally common; C = 6; BSUH 19835.

ALLIUM VINEALE L. – CW, SW; MWL; Rare; C = 0; BSUH 19686.

GALANTHUS NIVALIS L. – CW; MWL; Rare but locally abundant [naturalized]; C = 0; BSUH 19676.

NARCISSUS POETICUS L. – CW; MWL; Infrequent but local abundant [naturalized]; C = 0; BSUH 19681.

NARCISSUS PSEUDONARCISSUS L. – CW; MWL; Infrequent but locally common [naturalized]; C = 0; BSUH 19748.

ARACEAE (Arum Family)

Arisaema dracontium (L.) Schott – CW, SW; MWL; Common, especially in Skinner; C = 5; BSUH 19674.

Arisaema triphyllum (L.) Schott ssp. *triphyllum* – CW, SW; MWL; Infrequent; C = 4; BSUH 19805.

ASPARAGACEAE (Asparagus Family)

ASPARAGUS OFFICINALIS L. – CW; WE/OF; Rare; C = 0; BSUH 19771.

Camassia scilloides (Raf.) Cory – CW, SW; MWL; Infrequent but locally common; C = 5; BSUH 19673.

CHIONODOXA LUCILIAE Boiss. – CW; MWL; Rare but locally abundant; naturalized; C = 0; BSUH 19745.

- Maianthemum racemosum* (L.) Link ssp. *racemosum* – CW; MWL; Infrequent but locally common; C = 4; BSUH 20024.
- ORNITHOGALUM UMBELLATUM* L. – CW; MWL; Rare; C = 0; BSUH 19664.
- Polygonatum biflorum* (Walter) Elliott var. *biflorum* – CW, SW; MWL; Infrequent; C = 4; BSUH 19778.
- Polygonatum biflorum* (Walter) Elliott var. *commutatum* (Schult. & Schult. f.) Morong – CW; MWL; Rare; C = 4; BSUH 20034.
- COMMELINACEAE (Spiderwort Family)
- Tradescantia subaspera* Ker Gawl. – SW; MWL; Common; C = 4; BSUH 19754.
- Tradescantia virginiana* L. – SW; WE; Rare; C = 7; BSUH 20043.
- CYPERACEAE (Sedge Family)
- Carex blanda* Dewey – CW, SW; MWL; Abundant; C = 1; BSUH 19948.
- Carex cephalophora* Muhl. ex Willd. – CW, SW; MWL; Infrequent; C = 3; BSUH 19929.
- Carex cristatella* Britton – CW; MWL; Rare but locally abundant; C = 3; BSUH 19851.
- Carex davisii* Schwein. & Torr. – CW, SW; MWL; Abundant; C = 3; BSUH 19931.
- Carex gracillima* Schwein. – SW; MWL; Infrequent; C = 7; BSUH 19910.
- Carex granularis* Muhl. ex Willd. – CW, SW; MWL; Abundant; C = 2; BSUH 19912, 19927.
- Carex grayi* Carey – CW, SW; SUCWL; Rare; C = 5; BSUH 19915.
- Carex grisea* Wahlenb. – CW, SW; MWL; Abundant; C = 3; BSUH 19853, 19947.
- Carex hirtifolia* Mack. – CW, SW; SUCWL; Infrequent; C = 5; BSUH 19899, 19917.
- Carex hystericina* Muhl. ex Willd. – CW; CB near pond; Rare; C = 5; BSUH 19913.
- Carex jamesii* Schwein. – CW, SW; MWL; Common; C = 4; BSUH 19945.
- Carex lacustris* Willd. – CW, SW; VP; Common; C = 7; BSUH 19946.
- Carex laxiculmis* Schwein. var. *laxiculmis* – CW, SW; MWL; Infrequent but locally common; C = 7; BSUH 19911.
- Carex leavenworthii* Dewey – CW, SW; MWL; Infrequent; C = 1; BSUH 19852.
- Carex lupulina* Muhl. ex Willd. – CW; MWL around vernal pools; C = 4; BSUH 19892.
- Carex molesta* Mack. ex Bright – CW, SW; OF; Abundant; C = 2; BSUH 19922.
- Carex radiata* (Wahlenb.) Small – CW, SW; MWL; Abundant; C = 4; BSUH 19930, 20008, 19901.
- Carex shortiana* Dewey – CW, SW; MWL; Infrequent; C = 3; BSUH 19894, 19924.
- Carex sparganioides* Muhl. ex Willd. – SW; MWL; Rare; C = 4; BSUH 19916.
- (#) *Carex squarrosa* L. – CW; VP; Rare; C = 4; BSUH 19979.
- Carex stipata* Muhl. ex Willd. var. *stipata* – CW, SW; MWL; Infrequent; C = 2; BSUH 19928.
- Carex tribuloides* Wahlenb. var. *tribuloides* – CW, SW; MWL around vernal pools; Abundant; C = 5; BSUH 19896, 19978, 19988.
- Carex vulpinoidea* Michx. – CW, SW; SUCWL; Infrequent; C = 2; BSUH 19895, 19923.
- Cyperus esculentus* L. var. *leptostachyus* Böckler – CW, SW; OF; Infrequent; C = 0; BSUH 19949.
- Cyperus strigosus* L. – CW; MWL, SUCWL; Infrequent; C = 0; BSUH 19859.
- Eleocharis obtusa* (Willd.) Schult. – CW, SW; OF, WE/OF; Infrequent but locally abundant; C = 1; BSUH 19333.
- Scirpus atrovirens* Willd. – SW; WE/OF; Rare; C = 4; BSUH 19969.

Scirpus pendulus Muhl. – CW, SW; CW; MWL; Infrequent but locally common; C = 2; BSUH 19893, 19985.

DIOSCOREACEAE (Yam Family)

Dioscorea villosa L. – SW; MWL; Infrequent; C = 4; BSUH 19914.

IRIDACEAE (Iris Family)

Sisyrinchium angustifolium Mill. – CW, SW; CW; MWL; Infrequent but widespread; C = 3; BSUH 20051.

JUNCACEAE (Rush Family)

Luzula multiflora (Ehrh.) Lej. var. *multiflora* – CW; SUCWL; Rare but locally abundant; C = 6; BSUH 19672.

Juncus tenuis Willd. – CW, SW; MWL, SUCWL; Common; C = 0; BSUH 19925.

Juncus torreyi Coville – CW, SW; WE near creek; Rare; C = 3; BSUH 19967.

LEMNACEAE (Duckweed Family)

Lemna minor L. – SW; VP; Common; C = 3; BSUH 19972.

LILIACEAE (Lily Family)

Erythronium albidum Nutt. – CW, SW; MWL; Common and locally abundant; C = 3; BSUH 19743.

Erythronium americanum Ker Gawl. ssp. *americanum* – CW; MWL; Rare; C = 5; BSUH 19687.

Lilium michiganense Farw. – SW; MWL; Rare; C = 5; BSUH 20016.

MELANTHIACEAE (Bunchflower Family)

Trillium recurvatum Beck – CW, SW; MWL; Abundant; C = 4; BSUH 19680, 19759.

Trillium sessile L. – CW, SW; MWL; Abundant; C = 4; BSUH 19679, 19760.

ORCHIDACEAE (Orchid Family)

Spiranthes ovalis Lindl. var. *erostellata* Catling – CW; OF; Rare; C = 3; BSUH 20066. (State Watch List)

POACEAE (Grass Family)

AGROSTIS GIGANTEA Roth – CW, SW; OF; Common; C = 0; BSUH 19849.

Agrostis perennans (Walter) Tuck. – CW, SW; MWL; Infrequent but locally abundant; C = 2; BSUH 19871.

Alopecurus carolinianus Walter – CW, SW; WE/OF; Rare but locally common; C = 0; BSUH 19926.

Andropogon gerardii Vitman – CW, SW; OF; Rare but locally common; C = 5; BSUH 19881.

BROMUS COMMUTATUS Schrad. – CW, SW; SUCWL; Rare; C = 0; BSUH 19909.

BROMUS INERMIS Leyss. – CW, SW; MWL; Rare; C = 0; BSUH 19897, 19908.

BROMUS JAPONICUS Murray – SW; WE; Infrequent; C = 0; BSUH 19951.

Cinna arundinacea L. CW, SW; MWL; Abundant; C = 4; BSUH 19845.

DACTYLIS GLOMERATA L. – CW, SW; OF, WE; Infrequent; C = 0; BSUH 20007.

Dichanthelium acuminatum (Sw.) Gould & Clark var. *fasciculatum* (Torr.) Freckman – CW, SW; Infrequent but widespread; C = 2; BSUH 19857, 19866, 19891.

DIGITARIA ISCHAEMUM (Schreber) Muhl. – CW; SUCWL, WE; Infrequent but locally common; C = 0; BSUH 19873.

DIGITARIA SANGUINALIS (L.) Scop. – CW; SUCWL, WE; Infrequent but locally common; C = 0; BSUH 19872.

ECHINOCHLOA CRUSGALLI (L.) P. Beauv. – CW; MWL, SUCWL; Infrequent; C = 0; BSUH 19841.

Echinochloa muricata (P. Beauv.) Fernald var. *muricata* – CW, SW; SUCWL; Common; C = 1; BSUH 19870.

ELEUSINE INDICA (L.) Gaertn. – CW; WE; Infrequent; C = 0; BSUH 19885.

Elymus hystrix L. – CW, SW; MWL, SUCWL; Common, locally abundant; C = 5; BSUH 19888.

Elymus macgregorii R. Brooks & J.J.N. Campb. – CW, SW; MWL; Infrequent; C = 3; BSUH 19854.

Elymus villosus Muhl. ex Willd. – CW, SW; MWL; Common; C = 4; BSUH 19887.

- Elymus virginicus* L. – CW, SW; MWL; Common; C = 3; BSUH 19977.
- ERAGROSTIS PILOSA* (L.) P. Beauv. var. *PILOSA* – CW, SW; WE; Rare; C = 0; BSUH 19883.
- Festuca arundinacea* Schreb. (= *SCHEDONORUS ARUNDINACEUS* (Schreb.) Dumort) – CW, SW; OF; Abundant; C = 0; BSUH 19824, 19970.
- Festuca subverticillata* (Pers.) Alexeev – CW, SW; MWL; Common; C = 4; BSUH 19919.
- Glyceria striata* (Lam.) Hitchc. – CW, SW; MWL; Common; C = 4; BSUH 19907.
- HORDEUM JUBATUM* L. ssp. *JUBATUM* – CW; WE; Rare; C = 0; BSUH 19850.
- Leersia oryzoides* (L.) Sw. – CW, SW; MWL, SUCWL; C = 2; BSUH 19952.
- Leersia virginica* Willd. – CW, SW; MWL, SUCWL; Abundant; C = 4; BSUH 19830.
- Panicum capillare* L. – CW; OF; Rare; C = 0; BSUH 19869.
- Panicum dichotomiflorum* Michx. var. *dichotomiflorum* – CW, SW; OF, WE; Infrequent but locally common; C = 0; BSUH 19825.
- Panicum virgatum* L. var. *virgatum* – CW; OF; Rare but locally common; C = 4; BSUH 19844.
- Phalaris arundinacea* L. – CW; OF; Rare; C = 0; BSUH 19906. (Native, K. Yatskievych, Pers. Comm.).
- PHLEUM PRATENSE* L. – CW, SW; OF, WE; Infrequent; C = 0; BSUH 19889.
- POA ANNUA* L. – CW, SW; OF, SUCWL, WE; Infrequent; C = 0; BSUH 19921.
- POA COMPRESSA* L. – CW, SW; MWL; Common; C = 0; BSUH 19898.
- POA PRATENSIS* L. ssp. *PRATENSIS* – CW, SW; OF, SUCWL, WE; Common; C = 0; BSUH 19992.
- Poa sylvestris* A. Gray – CW, SW; MWL; Common; C = 5; BSUH 19918.
- POA TRIVIALIS* L. – CW; MWL especially around vernal pools; Infrequent but locally abundant; C = 0; BSUH 19900.
- Schizachyrium scoparium* (Michx.) Nash var. *scoparium* – CW; OF; Infrequent but locally abundant; C = 4; BSUH 19818.
- SETARIA FABERI* Herrm. – CW, SW; OF, WE; Common; C = 0; BSUH 19860.
- SETARIA PUMILA* (Poir.) Roem. & Schult. – CW, SW; OF, WE; Common; C = 0; BSUH 19858.
- SETARIA VIRIDIS* (L.) P. Beauv. var. *VIRIDIS* – SW; WE/OF; Infrequent; C = 0; BSUH 19950.
- Sorghastrum nutans* (L.) Nash – CW; OF; Infrequent but locally common; C = 4; BSUH 19868.
- SORGHUM BICOLOR* (L.) Moench – SW; OF/WE; Rare; C = 0; BSUH 19822.
- Sphenopholis intermedia* (Rydb.) Rydb. – CW, SW; Infrequent but locally common; C = 3; BSUH 19932.
- Tridens flavus* (L.) Hitchc. – CW, SW; OF, WE; Infrequent; C = 1; BSUH 19865.
- TRITICUM AESTIVUM* L. – CW; CB near the pond; Rare; C = 0; BSUH 19968.
- SMILACACEAE (Greenbrier Family)
- Smilax ecirrhata* (Engelm. ex Kunth) S. Watson – CW, SW; MWL; Common; C = 5; BSUH 19855.
- Smilax hispida* Raf. (= *Smilax tamnoides* L.) – CW, SW; MWL; Common; C = 3; BSUH 20021.
- Smilax lasioneura* Hook. – CW; MWL; Rare; C = 4; BSUH 19935, 20000.
- TYPHACEAE (Cattail Family)
- TYPHA ANGUSTIFOLIA* L. – CW; OF in soil pit; Rare; C = 0; BSUH 19760.

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MEDICAL STUDENTS SHOW LIMITED USE OF COMPUTER-AIDED INSTRUCTION IN STUDYING GROSS ANATOMY

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ABSTRACT. Within the last few decades, there has been increased interest in computer-aided instruction (CAI) as a supplement to, or replacement for, cadaver dissection. With the multitude of resources now available, it is important to collect information regarding students' use of anatomical resources. The purpose of this study was to assess the use of computer-aided instruction (CAI) by gross anatomy students at the Indiana University School of Medicine (IUSM). A survey was developed to assess how frequently students used a variety of resources. The gross anatomy resources were chosen based on their 1) prominence in the field of anatomy; 2) high level of quality; and 3) appropriateness for medical students. Students reported using general computer applications, such as PowerPoint, web browsing, and email most frequently to study gross anatomy. Instructor-made resources were also popular. The three most frequently used commercially available gross anatomy software programs were 1) *The Imaging Atlas of Human Anatomy*; 2) *Netter Interactive CD*; and 3) *The Visible Human Dissector*. However, a majority of students did not use, or were unaware of, the commercially available anatomy software. Students used resources that held the most potential for improving their grades, as was illustrated by the use of instructor-made CD-ROMs/DVD-ROMs at IUSM-Northwest and by the use of *the Visible Human Dissector* and *The Imaging Atlas of Human Anatomy* at IUSM-Lafayette. A number of exam questions came directly from these resources at these campuses. Because students' use of CAI was limited, adding or creating additional resources should be carefully considered.

Keywords: Gross anatomy, computer-aided instruction (CAI), anatomy education, science education

INTRODUCTION

Medical education as a whole, and gross anatomy (GA) specifically, has been undergoing a massive transition towards a prominent electronic presence. The primary means of studying and learning anatomy has been through the use of cadaver dissections, textbooks, and lectures. Within the last few decades, there has been increased interest in alternative methods, including the use of computer-aided instruction (CAI), to supplement or replace cadaver dissection (McNulty et al. 2000; Kesner & Linzey 2005; Kish et al. 2013; Saltarelli et al. 2014). CAI can take many forms including image databases, dissection videos, websites, computer animations, smart phone or tablet applications (apps), and podcasts (Lonn & Teasley 2009; Richardson et al.

2011; Jaffar 2012; Leggate 2012; Baheerathan & Selvaskandan 2014).

The transition towards a technology-centered curriculum has resulted in many research studies aimed at evaluating the effectiveness of different multimedia resources or developing new CAI programs to improve student learning of anatomical structures (McNulty et al. 2000, 2009; Nieder et al. 2000; Van Sint Jan et al. 2003; Elisndo-Omana et al. 2004; Hariri et al. 2004; Hudson 2004; Jastrow & Hollinderbaumer 2004; Lei et al. 2005; Linton et al. 2005; Levinson et al. 2007; Gould et al. 2008; Cook et al. 2010; Rich & Guy 2013). These studies have predominantly involved the evaluation of CAI resources that are instructor-made and course-specific. Results from these studies have been mixed and dependent upon the format of the CAI resource being evaluated.

The development and implementation of any new instructional resource should be driven by well-defined educational objectives that consider

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the needs of the students (Jastrow & Hollinderbaumer 2004; Mayfield et al. 2013). Evidence-based instructional design standards and guidelines for creating and implementing CAI are especially important in a medical curriculum (Han et al. 2014). However, in reality, CAI resources tend to be provided as supplemental learning materials for medical students to use at their own discretion. For CAI to realize its full potential, it must be incorporated into the curriculum and assume a more prominent role. Towards this purpose, it is necessary to examine the extent of CAI use and its effectiveness in learning outcomes. Several different terms have been used to describe this process. Educational and instructional designers use the term 'needs assessment' (Bacro et al. 2013), while others refer to it as 'implementation profiling' (Ellaway et al. 2014).

The use and effectiveness of CAI can be most easily evaluated through the collection of computer usage statistics and student surveys. Survey data can provide additional insights into what resources students feel are most useful in their studies (Jastrow & Hollinderbaumer 2004). For example, Mayfield et al. (2013) created a survey to assess students' use of iPads versus traditional dissection atlases "and the nature of [the students'] participation" in the GA laboratory; while Rich & Guy (2013) focused their survey on rating the value of an online teaching module versus lectures and laboratory sessions "as a source of understanding the course and its contents". Surveys may also reveal discrepancies between students' preferences for using electronic resources and students' actual use of electronic resources. Jastrow & Hollinderbaumer (2004) found that students wanted CAI applications in anatomy and stated that they would use them; however, in practice a third of students reported that they accessed the available CAI materials less than once a month.

When considering the inclusion or development of CAI resources for teaching GA, one of the first steps should be to determine which resources students are actively using. However, there is a noticeable lack of information about what learning resources students actually use under normal circumstances. Ellaway et al. (2014) conducted a similar study to explore how students made use of mobile devices. Although their experiment focused on the use of hardware, they did find that student use was heavily dependent upon context. With the

variety of available resources, it is more important to make a concerted effort to collect information regarding students' use of anatomical resources on a regular basis. The purpose of this study was to assess the use of computer-aided instructional (CAI) resources used to study gross anatomy by first-year medical students at Indiana University School of Medicine (IUSM).

MATERIALS AND METHODS

This study was conducted at IUSM. IUSM is one of the largest medical schools in the United States. It consists of a central campus in Indianapolis, Indiana and eight regional centers across the state of Indiana. Four of the nine campuses granted permission to conduct this research project. The campuses that participated were the main campus in Indianapolis, IUSM-Northwest (Gary), IUSM-Lafayette, and IUSM-Terre Haute.

At the time of this study, the curriculum at IUSM was equivalent across the campuses, but not identical. Teaching pedagogies at each campus varied. However, there was a common core curriculum, statewide discipline exams, and common assessment of competencies for standardization across the nine campuses. The main Indianapolis campus and IUSM-Lafayette had discipline-based curricula with multiple courses in each semester. At both campuses GA was a stand-alone course in the fall semester and was taught through didactic lectures and full cadaver dissection. IUSM-Northwest (Gary) had an integrated, problem-based learning (PBL) block schedule. GA was integrated with cell biology, histology, embryology, and radiology and was taught through prosected cadavers. IUSM-Terre Haute had an integrated discipline-based curriculum with multiple courses in each semester. GA was taught through a combination of team-based learning (TBL), lecture, and full cadaver dissection during the fall semester. CAI was used as a supplement to dissection laboratory sessions.

A pencil-and-paper questionnaire assessing the use of multimedia, specifically computer-aided instruction, in gross anatomy courses was developed to evaluate how frequently students used a variety of resources ranging from instructor-made to commercially available anatomy software. Several previously published surveys evaluating medical students' use of CAI and their computer literacy were used as a guideline for the questionnaire in the current study (Magid et al. 1988; Lang

Table 1.—Response rate from participating IUSM Campuses.

Campus	%	N
IUSM-Northwest	87.5	21
IUSM-Indianapolis	71.0	98
IUSM-Lafayette	100.0	16
IUSM-Terre Haute	95.8	23

1995; Lynch et al. 2000; Dørup 2004; Jastrow & Hollinderbaumer 2004; Link & Marz 2006; Forman & Pomerantz 2006). The questionnaire contained items designed to 1) assess general computer-usage; 2) determine the frequency and usefulness of various resources, including the ones made by course instructors; and 3) gauge the use of commercially available software packages designed specifically for anatomy. The GA resources included in this survey were chosen based on the following factors: 1) their prominence in the field of anatomy; 2) their high level of quality; and 3) their appropriateness for first-year medical students. The questionnaire was pilot tested on a sample of fourteen second-year medical students at IUSM-Lafayette in order to obtain feedback regarding the clarity of the instructions and appropriateness of the questions. A cover letter attached to each survey explained the purpose of the study and addressed concerns regarding anonymity and informed consent. This anonymous survey study was deemed exempt by the Institutional Review Board (Protocol #: 081007481).

Descriptive statistical analyses were conducted using SAS 9.1. Percentages were calculated based on the total number of respondents who answered each item. Chi-Square Goodness of Fit Tests and Chi-Square Tests of Association were conducted using SPSS version 24.

Table 2.—Demographic information for first-year medical students at IUSM.

Age	
Mean	24
Range	20–44
Gender	
Male	56%
Female	44%
Ethnicity	
Caucasian	68%
Asian/Pacific Islander	18%
African American	5%
Hispanic	3%

Table 3.—Basic computer usage of first-year medical students at IUSM.

	%	N
Use or Own PC	63.3	105
Use or Own Mac	33.1	55
Cable Broadband Internet	57.3	90
DSL Internet	28.0	44
Use Wireless Internet	95.4	147
Access Email Daily	96.8	152
Browse Internet Daily	90.6	144

RESULTS

The overall response rate for participating IUSM campuses was 78% (n=158). The response rates at the four participating campuses are reported in Table 1. Demographic information for all surveyed students is provided in Table 2. Data on basic computer usage, provided in Table 3, were used as a base-line comparison to the data obtained regarding CAI used to study GA.

Overall, students reported using Internet browsing (44.6%), PowerPoint presentation software (47.1%), and email (37.6%) on a daily basis to study GA (Table 4). There was a statistically significant difference ($p < 0.001$) in the proportion of students reporting daily use of the following computer applications to study GA: Internet browsing, word processing, spreadsheets, CD-ROMs/ DVD-ROMs, presentation software, and email. Word processing, spreadsheets, and CD-ROMs/ DVD-ROMs were used less frequently, with spreadsheets being the least used resource. Not surprisingly, instructor-made resources were quite popular with the students. There was a statistically significant difference ($p < 0.001$) in the proportion of students reporting the use of different types of instructor-made resources. Instructor-made handouts and PowerPoint presentations were used by a majority of students across all four participating campuses. In a typical didactic lecture, PowerPoint presentations are the predominant method of delivering anatomy content. GA course packets, consisting of detailed notes or outlines for each lecture, are typically provided to students at several of the IUSM campuses. Additional handouts could have been in the form of print-outs of the PowerPoint lectures. The use of CAI at one particular campus, IUSM-Northwest, stood out from the others. Here 81% of surveyed students reported using instructor-made CD-ROMs or DVD-ROMs to study GA. This campus utilized a problem-based

Table 4.—Frequency of use of resources to study gross anatomy. * = There was a statistically significant difference ($p < 0.001$) in the proportion of students reporting daily use of the following computer applications: internet browsing, word processing, spreadsheets, CD/DVD-ROMs, presentation software, and email.

Resource	Daily* %	Several times a week %	Several times a month %	Several times a year %	Less often %	Never %
Internet Browsing	44.6	35.0	14.0	1.3	1.3	3.8
Word Processing (Word, Wordperfect)	15.9	26.1	23.6	6.4	10.2	17.8
Spreadsheets (Excel)	5.1	16.4	19.0	3.8	19.6	36.1
CD-ROMs/ DVD-ROMs	1.3	8.4	13.0	4.6	13.6	59.1
Presentation Software (Powerpoint, Corel)	47.1	26.8	10.2	1.9	3.8	10.2
Email	37.6	29.9	10.8	0.6	9.6	11.5

learning (PBL) curriculum in a block schedule. Instructor made CD-ROMs and DVD-ROMs may have been used to provide students with electronic versions of handouts, PowerPoint lectures, or provide additional anatomy content to students as they work through PBL cases.

Not only was it beneficial to determine how often students used CAI resources, it was also important to discover whether or not the students found them useful. A majority of students reported that course websites, learning management systems (such as Blackboard), and PowerPoint presentation software were useful to study GA (by 80.9%, 89.2%, 89.7% of student, respectively). Email and reference websites (such as Wikipedia) were also reported as useful (by 75.8% and 69.8% of students, respectively). Although the overall use of CD-ROMs/DVD-ROMs was rather infrequent, they were seen as useful by 20.7% of participating students. There was a statistically significant difference ($p < 0.001$) in the proportion of students reporting that they found the following computer applications useful to study GA: Internet browsing, word processing,

spreadsheets, CD-ROMs/DVD-ROMs, presentation software, and email.

The surveys indicated that the overall use of CD-ROMs/DVD-ROMs was dependent upon location (Table 5). Students at the IUSM-Northwest campus used CD-ROMs/DVD-ROMs more frequently (1/3 of students using them several times a week) than at the other reporting campuses. The sample size was too low to calculate the statistical significance of the proportion of students reporting frequent use (daily or several times a week) of CD-ROMs/DVD-ROMs to study GA by campus at the four IUSM campuses. However, there was a statistically significant difference ($p < 0.001$) in the proportion of students reporting that they never used CD-ROMs/ DVD-ROMs to study GA. A majority of students at both the IUSM-Indianapolis and the IUSM-Terre Haute campuses reported never using any CD-ROMs/DVD-ROMs to study GA.

The perceived usefulness of CD-ROMs/DVD-ROMs in general was also variable across the campuses (Table 6), which is most likely due to the differences in how often students at each campus

Table 5.—Frequency of use of CD-ROMs and DVD-ROMs to study gross anatomy at each IUSM Campus. * = There was a statistically significant difference ($p < 0.001$) in the proportion of students reporting that they never used CD-ROMs/ DVD-ROMs to study GA.

Campus	Daily %	Several times a week %	Several times a month %	Several times a year %	Less often %	Never* %
IUSM – Northwest	0	33.3	38.1	9.5	14.3	4.8
IUSM – Indianapolis	1.1	4.2	4.2	3.2	12.8	74.5
IUSM – Lafayette	6.3	6.3	37.5	6.3	12.5	31.2
IUSM –Terre Haute	0	4.4	8.7	4.4	17.4	65.2

Table 6.—Frequency of reported usefulness of CD-ROMs and DVD-ROMs to study gross anatomy at each IUSM Campus.

Campus	Useful %	Undecided %	Not useful %	N/A – Did Not Use %
IUSM-Northwest	61.9	23.8	14.3	0
IUSM-Indianapolis	10.5	27.4	3.2	58.9
IUSM-Lafayette	31.2	37.5	18.8	12.5
IUSM-Terre Haute	17.4	17.4	4.3	60.9

used them. At IUSM-Northwest, 61.9% of surveyed students found CD-ROMs/DVD-ROMs useful to study GA compared with only 31.3% of students at IUSM-Lafayette. However, there was no statistically significant difference ($p=0.08$) in the proportion of students reporting that they found CD-ROMs/ DVD-ROMs useful for studying GA by campus.

We were particularly interested in determining if students used commercially available software for learning GA. Examples included *ADAM Interactive*, *Netter's Interactive Atlas*, *The Imaging Atlas of Human Anatomy*, *Primal 3D*, *The Dynamic Human*, CD-ROMs/DVD-ROMs accompanying textbooks of anatomy, *Thieme Image Collection*, *Acland's DVD Atlas*, and *The Visible Human Dissector* (Fig. 1). There was a statistically significant difference ($p < 0.001$) in the proportion of students reporting frequent use (daily or several times a week) of these commercially-available CAI resources. The three most

used commercially available software programs were: 1) *The Imaging Atlas of Human Anatomy* (27.3%); 2) *Netter Interactive CD* (23.2%); and 3) *The Visible Human Dissector* (16.2%). However, they were not used frequently by many students. Instead, a majority of students did not use or were unaware of these commercially available anatomy instructional software packages (Table 7). Of the three most used programs mentioned above, *The Imaging Atlas of Human Anatomy* was used the most frequently, with 13.6% of students reporting usage daily or several times a week (Table 7).

To further investigate the use of these three resources, the usage frequencies were determined for each campus. Of the four participating campuses, IUSM-Lafayette was the only campus that showed a distinct pattern of use for these commercially available anatomy software programs. At IUSM-Lafayette, *The Visible Human Dissector* was the most frequently used software program. In fact, just over 75% of the students

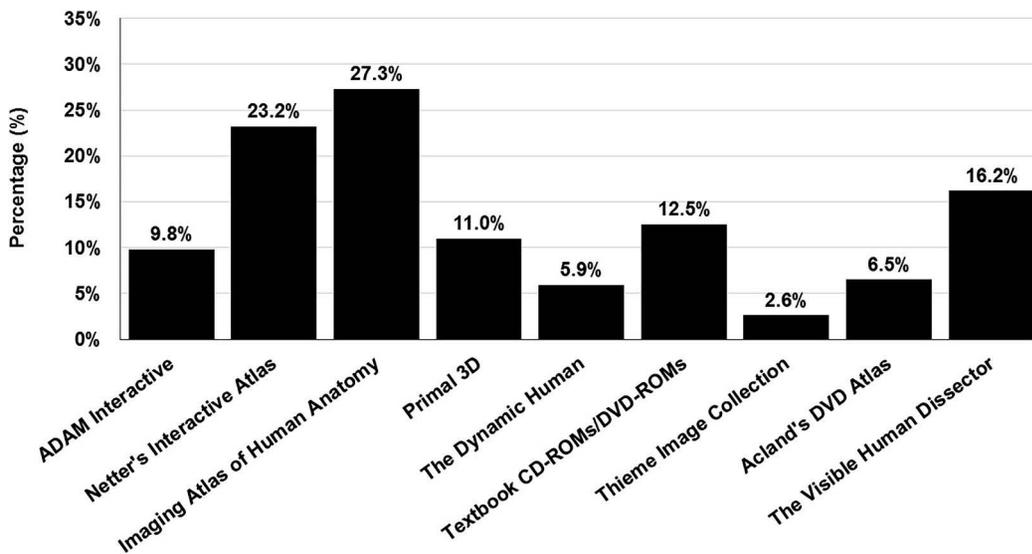


Figure 1.—Percentage of students at IUSM reporting use of commercially available anatomy software to study gross anatomy.

Table 7.—Frequency of use of commercially available anatomy software to study gross anatomy at each IUSM Campus.

Resource	Daily %	Several times a week %	Several times a month %	Several times a year %	Less often %	Never %	Have not Heard Of %
Netter's Interactive Atlas CD-ROM	0.7	5.8	7.1	1.9	7.7	65.8	11.0
Imaging Atlas of Human Anatomy	6.5	7.1	10.4	0.7	2.6	45.4	27.3
The Visible Human Dissector	0.7	6.5	3.2	4.5	1.3	42.9	40.9

used *The Visible Human Dissector* between several times a week and several times a month (Fig. 2).

In addition to using *The Visible Human Dissector*, over half (56.3%) of the students at IUSM-Lafayette made use of *The Imaging Atlas of Human Anatomy*, with 12.5% indicating daily use of this program (Fig. 3). These two resources were made available to students at IUSM-Lafayette in CD-ROM/DVD-ROM format; and a number of written exam questions came directly from the content of these resources. From the data obtained from this study, it is unclear whether the other IUSM campuses made use of these, or similar, resources.

From these observations it can be concluded that students at IUSM-Northwest use instructor-made CD-ROMs/DVD-ROMs frequently to study GA, and they generally found them useful. Data also suggest that students at IUSM-

Lafayette used commercially available anatomy CD-ROMs/DVD-ROMs, and generally found them useful as well.

DISCUSSION

Overall, students reported using general computer applications, such as PowerPoint, web browsing, and email, most frequently to study GA. Email as a study method was an interesting and surprising finding. New technologies are changing the learning environment for professional students and studying is no longer limited to reading the textbook, attending class, and taking notes. Email as a study method could include submitting questions to the course instructor, or sharing notes, references, and websites with other classmates. Several studies have used email as an integral part of instructional

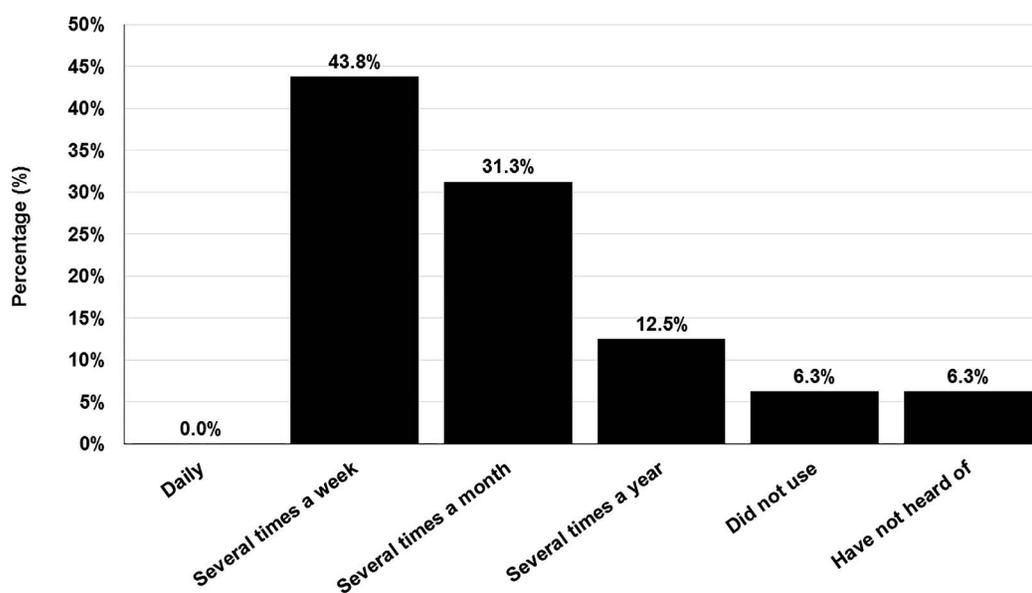


Figure 2.—Frequency of use of *The Visible Human Dissector* to study gross anatomy at the IUSM-Lafayette Campus.

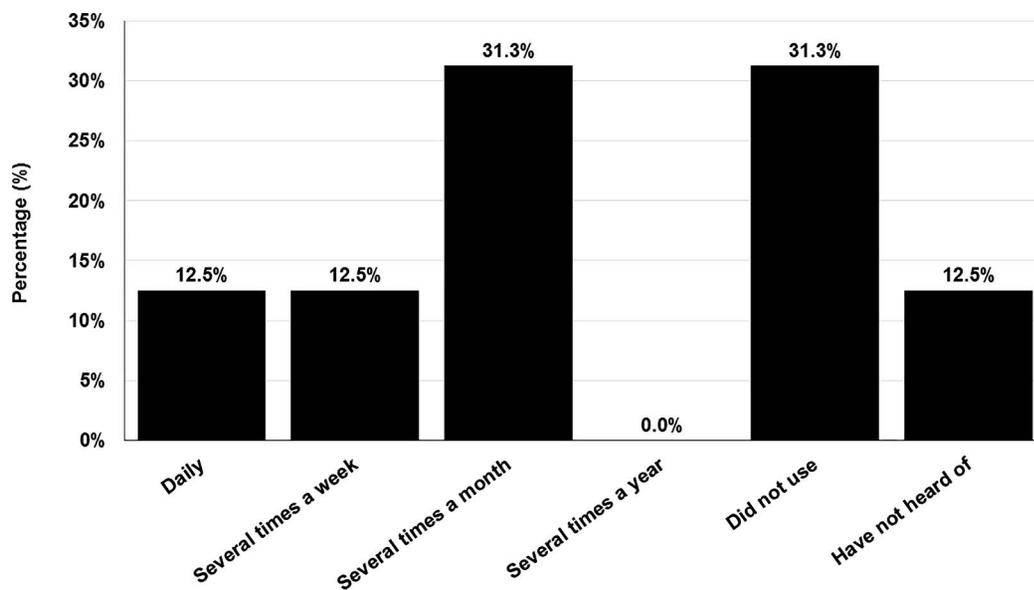


Figure 3.—Frequency of use of *The Imaging Atlas of Human Anatomy* to study gross anatomy at the IUSM-Lafayette Campus.

intervention activities (Kerfoot et al. 2012; Bow et al. 2013).

The distinction between technology as an educational tool versus technology as an integral part of instruction is blurring. Technologically-savvy students may have different perceptions of what qualifies as ‘studying’. For example, Han et al. (2014) found that students perceived Google Docs, wikis, podcasts, YouTube, Google Calendar, Skype, and Learning Management Software (LMS) useful for learning. The addition of technology such as iPad apps, *Second Life*, *Twitter*[™], and *Facebook* to the domain of instructional methods has further changed the way people study and learn (Lonn & Teasley 2009; Richardson et al. 2011; Richardson-Hatcher et al. 2013, 2014; Jaffar 2014; Lee & Gould 2014). Most of these computer applications do not explicitly fall within the category of CAI. A major drawback to these applications is that they may not provide relevant, correct, and up-to-date information. Raikos & Waidyasekara (2014) evaluated YouTube videos of the human heart and discovered that most were of poor quality and were not of sufficient detail for medical students. Vilensky & Steenberg (2015) had similar findings from their investigation of anatomy pages on Wikipedia. Lewis et al. (2014) performed a systematic search for iPad apps relating to anatomy. With no guidelines or standards for

the content of apps, the concerns remain not just on the availability of recent and up-to-date information on the content in these learning apps, but also on the validity and appropriateness of the content.

The current study indicated that the use of CAI to study GA was limited. Students used instructor-made resources, but most students either did not use, or were unaware of, the commercially available anatomy software programs. Possible reasons for this are: 1) the programs were not available for the students; 2) the programs were available but the students were not explicitly informed about them; or 3) the students were informed that programs were available as supplemental study material, but the programs were not incorporated directly into the course. The availability of commercially prepared CAI programs for students may be challenging to address, requiring financial investment on the part of universities and/or students. However, textbooks like Moore’s *Clinically Oriented Anatomy*, Netter’s *Atlas of Anatomy*, and Gilroy’s *Atlas of Anatomy* are now available in eBook format, making access to enhanced online study tools even easier. There are also many anatomy applications (apps) available for download onto tablets or smartphones.

This highly technology-driven generation of students with access to educational resources

literally at their fingertips may be expected to find useful online resources for their studies on their own. However, the time-intensive demands of medical school education along with the compressed design of anatomy taught in an integrated curriculum may prevent students from seeking out and identifying the best and most appropriate resources to aid their studies. In fact, Johnson et al. (2013) found that students expect the faculty to be able to direct them to appropriate resources. As experts in the field, anatomy faculty could advise students on the best CAI resources available.

Students tend to use resources that hold the most potential for directly improving their grades, as was illustrated by the frequency of use of instructor-made CD-ROMs/DVD-ROMs at IUSM- Northwest and by the frequency of use of *the Visible Human Dissector* and *The Imaging Atlas of Human Anatomy* at IUSM-Lafayette. The information from these programs was included as part of the GA exams at these campuses. Jastrow & Hollinderbaumer (2004), in their study of web-based resources and CD-ROMs in GA, found that students' "focus of interest was on material with relevance to current courses and examinations", while Jaffar (2012) found that "students preferred resources based on the expected benefits in examinations". Both of these results support the findings from the current study.

CAI is most effective when it is fully incorporated into a course rather than solely used as an optional resource (Jastrow & Hollinderbaumer 2004; Vivekananda-Schmidt et al. 2004; Kish et al. 2013). When CAI is provided as supplementary resources for GA courses, students' use of these resources is often inconsistent (McNulty et al. 2009). When asked why they did not use the CAI resources, students' common responses included: "1) technical difficulties with their computers; 2) lack of sufficient time; 3) other resources were more useful to their study; and 4) specific CAI did not fit their learning style" (McNulty et al. 2009). In light of the results of McNulty et al. (2009), the findings from the current study may not be unique to IUSM, and may have broader implications for developing and incorporating CAI into GA courses at medical schools across the country.

If, as in this study, students' use of CAI is limited mostly to instructor-made resources and materials directly incorporated into exams, the purpose of adding or creating additional resources should be carefully considered. Prior to

spending countless hours creating and testing new CAI resources, instructors might consider tailoring one of the many programs already in existences to fit their course design, course objectives, and needs of the students (Doubleday et al. 2011; Robin et al. 2011). Along these similar lines, Attardi & Rogers (2015) critically evaluated ten commercial software programs prior to choosing Netter's 3D Interactive Anatomy as the program that would fit their needs best (Netter 2012). CAI can be a great tool to aid teaching and enhance the learning experience when used appropriately and in conjunction with other teaching tools. The focus of educators should be how CAI could address specific learning objectives within the anatomy curriculum (Tworek et al. 2013).

Although this study took place during a transitional period in CAI from CD-ROM/DVD-ROMs to Internet-based resources, the impact of the findings remains relevant. There is now a plethora of available resources online that are accessible via any mobile device (laptop, tablet, smart phone, ebook reader, etc.). Smart phone applications and YouTube videos can be created by just about anyone. So it is important to evaluate these resources critically, both for consistency with specified learning objectives and for potential use among students. CAI may enhance and maximize student learning when these resources are matched to course and curricular learning objectives. One of the most important findings from this study was that students were largely unaware of the commercially available GA resources. We are still in the midst of a technology revolution in medical education, so instructors should stay abreast of new technologies and actively evaluate how students choose to use these technologies as they study anatomy.

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IDENTIFYING AND MITIGATING PRACTICES THAT INDUCE STEREOTYPE THREAT AT VALPARAISO UNIVERSITY

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ABSTRACT. This paper describes the implementation, assessment, and impact of a professional development project to address stereotype threat at Valparaiso University (VU). Stereotype threat is a psychological phenomenon that has been shown to cause disadvantaged groups to underperform on a wide range of tasks. Additionally, it is recognized as a key contributor to the underrepresentation of women and minorities in science, technology, engineering, and mathematics (STEM) fields. This project sought to assess the extent of stereotype threat and execute an intervention to reduce stereotype threat on VU's campus. Supported by a grant from the American Association of University Women (AAUW), VU hosted lectures and workshops by Dr. Catherine Good, a stereotype threat expert. Several follow-up discussion events were held over the following months to increase the impact of Dr. Good's visit. Through these activities faculty, staff, and students learned about stereotype threat and its influence on learning. Assessments of each event and the subsequent implications for the mitigation of practices that induce stereotype threat are discussed in this paper. The results provide significant hope for future reduction of stereotype threat at VU. However, the results also highlight a gap between faculty/staff self-perceptions and student experiences with this issue.

Keywords: Stereotype threat, implicit bias, faculty, professional development, STEM education

INTRODUCTION AND PROJECT SUMMARY

Women and ethnic minorities are unequally represented in many science, technology, engineering, and mathematics (STEM) disciplines (National Science Board 2016, Chapter 2). One known barrier to the participation of underrepresented groups in STEM is the psychological phenomena known as stereotype threat (ST). As defined by Steele & Aronson (1995), "stereotype threat is a fear of confirming a negative stereotype about your group." ST is a well-established phenomenon in psychology research literature, with studied groups ranging from women to African Americans and even Caucasian males (Nguyen & Ryan 2008). Inspired by the research report *Solving the Equation* from the American Association of University Women (AAUW), Valparaiso University (VU) undertook efforts to

raise awareness of ST among faculty, staff, and students (Corbett & Hill 2015). Our work presents a case-study on a campus-wide professional development project about ST. In this paper we detail the project's effect on ST awareness in faculty, staff, and students as well as faculty/staff's intent to change current mentoring and teaching practices.

The goal of our project was to raise awareness about ST and provide strategies to reduce or eliminate it in classrooms, informal mentoring, and the workplace both on and off VU's campus. To achieve this goal, two different audiences were targeted: (1) faculty and staff (hereafter referred to as 'employees'), and (2) students. For employees a professional lecture, pedagogical development workshops, and follow-up discussions were sponsored. For students a general lecture, including an invitation to the general public, with follow-up discussions were held.

In planning these activities, the need for an expert on both the phenomenon of ST and techniques for reducing it was identified. To this

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end, Dr. Catherine Good, Associate Professor of Psychology at the City University of New York, was invited as the primary lecturer and to lead the workshop sessions. A nationally recognized expert in this field, Dr. Good's work focuses on how negative stereotypes contribute to women's underachievement and underrepresentation in math and science disciplines (Aronson et al. 1999; Good et al. 2007, 2008, 2012). She has presented at many national conferences as a keynote or workshop leader including the Joint Mathematics Meetings, the American Physical Society's March Meeting, and the National Center for Women & Information Technology's Summit. Dr. Good also studies methods of helping women overcome vulnerability to ST and alternative techniques for faculty to avoid causing ST (Aronson et al. 2002; Good et al. 2003; Dweck et al. 2004; Inzlicht et al. 2006).

The remainder of this paper is laid out as follows. The *Background and Motivation* section explains in more depth the effects of ST as well as the institutional context which sparked this project. The *Methods and Approach* section details our actual implementation and marketing for the intervention activities. The *Assessment Plan* outlines how we measured the impact of our interventions on knowledge and intent. The *Results* contain two subsections, one on employees, one on students, which report the survey responses and basic interpretations of them. Finally, the *Discussion* summarizes the overall intervention outcome and shares lessons learned.

BACKGROUND AND MOTIVATION

Stereotype threat background.—Stereotype threat (ST) has been explicitly studied since the publication of Steele & Aronson's seminal article in 1995 where the term first appeared (Steele & Aronson 1995). Since then over 850 articles have been published examining ST (based on a Google-Scholar search for "Stereotype Threat" in titles). Here we will highlight three of the several meta-analyses that exist on these works. First, Walton & Cohen (2003) perform a meta-analysis that highlights how the "in-group" can actually achieve a performance boost. Second, Nguyen & Ryan (2008) introduce three levels of ST: blatant, moderately explicit, and indirect/subtle. They also review several articles that attempted to remove ST through either explicit or subtle interventions. Most importantly, their meta-analysis found that the experiments statistically

supported an effect on women, with a greater impact on minorities. Nguyen and Ryan also provided a ranking of which type of interventions were most effective for each group experiencing ST. Third, and more recently, Pennington et al. (2016) performed a meta-analysis of the various psychological mediators that can create or influence the actual performance in individuals exposed to ST. Three broad categories of mediators were examined: affective/subjective, cognitive, and motivational mechanisms. Each category had detailed examples of the mediators and highlighted experiments discussed in the literature. These meta-analyses clearly indicate that ST is a valid psychological phenomenon applicable to a broad range of groups, with a variety of causes and negative outcomes.

This project was specifically concerned with increasing awareness of the impact that ST can have on women and underrepresented minorities (URMs) as well as introducing ways to mitigate ST. As a concrete summary of the impacts above, Corbett & Hill (2015) state that when someone experiences ST it reduces working memory capacity, increases stress and anxiety, and may lead to disengagement from domains in which a person feels stereotyped (Pennington et al. 2016). Many researchers are investigating how to alleviate the psychological impact of ST. Proven strategies include addressing women and URM's sense of belonging by creating a community of equality and welcome. For example, regardless of the task or degree program difficulty, establishing that everyone (men and women) must work hard has been shown to increase women's sense of belonging (Smith et al. 2013). Similarly, Carol Dweck's work on "growth mindsets" has also been employed by researchers to increase a sense of belonging by making students aware that difficulties, challenges, and failures are a normal part of earning a degree (Dweck et al. 2004; Walton & Cohen 2007; Dweck 2008). Finally, simply reducing the disparities between men and women (or other groups) can help prevent feelings of non-belonging (London et al. 2014).

Institutional motivation.—Within the gender imbalanced (with male prevalence) environments of STEM, we acknowledge that the College of Engineering (CoE) faculty, science professors, and other technical professors often unknowingly commit stereotype threats on a regular basis. Even though VU's CoE has an percentage of women above many undergrad-

uate engineering schools (National Science Board 2016), its gender mix is still heavily skewed with only about 20% female students. Of greater concern, Computer and Information Sciences (CIS) has had a very low average enrollment of women (12.5%), well below the national average. The CIS department also had a 14% lower retention rate for women than men in 2009–2013. Women, as a clear minority in these disciplines may easily experience a more limited sense of community. This makes imperative the creation of a welcoming and inclusive environment free from ST and other negative psychological influences.

METHODS AND ASSESSMENT PLAN

Methods.—To create a community of acceptance, the employees must be able to talk and teach in ways that do not generate ST. Moreover, they must be convinced that they need to change. Together these are the primary goals for this project, the education and impact on practices surrounding ST for employees and especially within the STEM faculty. These goals suggested the two-stage intervention created: an educational lecture and a skills workshop.

By initially offering the lecture, employees were presented with research data demonstrating the existence of ST and making evident the need for change. Research focusing on a variety of at risk groups was shown. Even more compelling was the research that ST could be induced in white males, a traditionally privileged group. Appealing to a campus culture that highly values effective teaching and mentoring, the lecture focused on the negative effect that ST has on learning. It concluded with a description of three effective interventions – encouraging a growth mindset, encouraging belonging based on effort or engagement, and re-attribution for difficulty. All of these serve as effective practices for improving student learning, regardless of the state of ST.

The workshop, presented as a professional development activity, provided employees a venue for learning important skills as well as possible interventions to reduce ST within their classrooms. To increase long-term effectiveness, follow-up discussions were scheduled two months after the lecture and workshops. This allowed all participants to reflect on what they had learned and to plan future activities for themselves or the general university.

An additional component in creating a ST free environment is helping students themselves be alert to experiencing or creating ST. Research has shown that students who are aware of ST are less likely to suffer from the negative effects (Gujardo 2005; Johns et al. 2005; Dar-Nimrod & Heine 2006). Moreover, by making students aware of ST, they will be able to share that knowledge with other students and avoid creating it themselves. These reasons led to the inclusion of a general lecture for the student body. Similar to the interventions for employees, follow-up discussions with students were scheduled over the next two months to increase the long-term efficacy of this intervention.

Faculty and staff were invited to attend the lecture and workshops through a wide range of messaging. The Assistant Provost for Inclusion, the Deans of their respective colleges, and advertising by VU's Institute for Teaching and Learning provided official invitations. More informally, student groups on campus, such as Athena Society (a gender equality group) and the Society of Women Engineers (SWE), used real-life examples and personal invitations to inspire faculty attendance.

Students were invited to the general lecture through student organizational leaders, posters, and targeted emails. Specifically, the event was shared with the university student leadership network and the Engineering Leadership Student Association Committee. This led to at least two professional societies including the event as part of their membership education requirements.

Assessment plan.—As stated above, the primary goal of our project was to educate and modify practices of employees, especially STEM faculty. The secondary goals of our project were to educate the general student body about ST and promote general awareness. To evaluate the results of our project, we established four measures of success: (1) lecture and workshop attendance, (2) understanding of ST, (3) short- and long-term behavior change, and, for employees only, (4) inspiring intent to change current mentoring or pedagogical practices.

Faculty and staff assessment: We assessed (1) by recording attendance numbers, especially of STEM faculty, at the lectures and workshop. For (2) and (4) we administered a post-lecture and post-workshop survey to participants to evaluate their learning and future plans. For (3), short-term impact was assessed by informal

Table 1.—Faculty and staff understanding of stereotype threat.

I believe...	Strongly disagree (1)	Disagree	Neutral	Agree	Strongly agree (5)	Average	Std. dev.
From this event [lecture] I learned a lot of new things about stereotype threat	0	1	0	4	8	4.46	0.88
I learned a lot from this workshop.	0	0	1	5	7	4.31	0.87
that stereotype threat can negatively impact performance of underrepresented groups (e.g. gender, ethnicity, etc.)	0	0	0	1	12	4.92	0.28
that stereotype threat can negatively impact performance of any group or person.	0	1	2	3	7	4.23	1.01
my teaching practices or pedagogy should change to reduce stereotype threats	0	1	2	3	6	4.17	1.03
that my teaching practices currently introduce stereotype threat to at least one group	0	4	1	7	0	3.25	0.97

interviews with SWE members and other female STEM students and follow-up conversations with faculty. Additionally, part of a second round of post-event surveys asked employees to reflect on any changes they instituted at the end of spring semester. Finally, the long-term impact of (3) will be evaluated through institutional retention data and graduation/exit interviews, which are already being analyzed for similar outcomes in another project for the National Center for Women in Information & Technology.

Students and community assessment: For goal (1) we used an identical assessment to the employee's. We assessed (2) by creating and administering a short survey at the end of the public lecture, accessible via smartphone. Included were a few questions to gauge understanding followed by brief demographic questions. Goal (3) or short/long term impact was assessed by attendance at the follow-up events and subsequent engagement of students in various activities to reduce ST on campus.

RESULTS

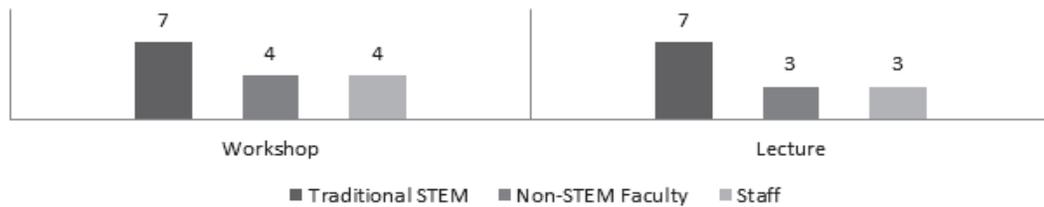
Results from faculty and staff.—With regards to the primary goals of our project, to educate and influence practices of employees, we achieved notable success. For measure (1),

there was a total attendance of 22 employees between both of Dr. Good's lectures. Additionally, there were over 30 attendees at the workshops held specifically for employees. Overall, between 35 and 40 individual employees interacted directly with Dr. Good during the lectures and workshops. While the initial goal for attendance of more than 50 at the lecture(s) was not achieved, the workshop attendance goal of more than 30 was achieved and through several follow-up activities managed to reach at least 50 employees.

To understand the effectiveness of various activities (Goals 2–4), several follow-up surveys were administered. Thirteen responses to the lecture and 16 responses to the workshops were collected, giving an average response rate of 58%. The demographic data from the surveys shown in Fig. 1 indicate that the target audience was reached, including a focus on STEM faculty.

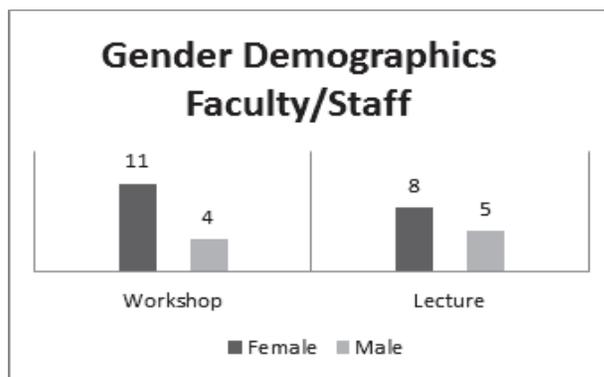
To assess goal (2), employee understanding of ST, we asked four questions on a Likert scale (Table 1). We acknowledge that these questions were both subjective and affective; however, these results were strongly positive. They show that the employees absorbed the information from Dr. Good's lecture about the pervasive effect ST can have on learning.

Affiliation Demographics Faculty/Staff



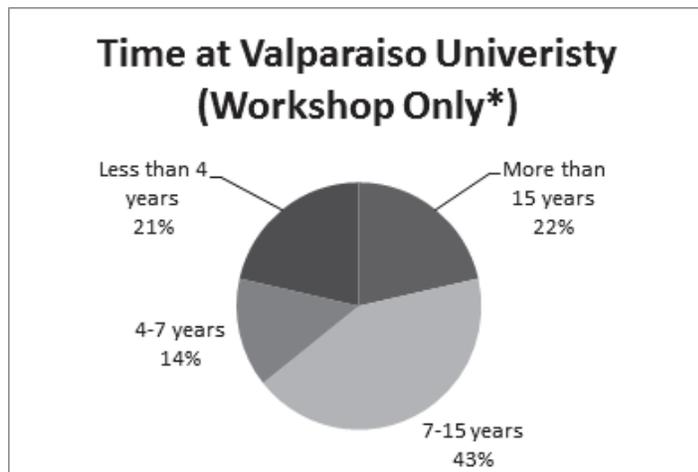
(a)

Gender Demographics Faculty/Staff



(b)

Time at Valparaiso Univeristy (Workshop Only*)



(c)

Figure 1.—Demographic data for attendees of the workshops and lecture. Similar data were not collected in the post-lecture survey.

For measure (3), employee intent or inspiration to change, two questions were asked during the initial post-workshop survey (Table 2, with response counts). Additionally, some inference

can be made based on the attendance at follow-up meetings. While the project did not originally include any follow-up discussions, the opportunity arose to schedule more events specifically for

Table 2.—Faculty and staff intent to change.

	Very unlikely (1)	Unlikely	Neutral	Likely	Very likely (5)	Average	Std. dev.
Based on this workshop, how likely are you to change your teaching practices?	0	0	1	8	4	4.25	0.58
Based on this workshop, how prepared do you feel to create classroom activities which reduce or avoid stereotype threat?	0	1	2	8	2	3.94	0.77

employees. These follow-up discussions were held during finals week, nearly two months after Dr. Good's visit. These events attracted more than 40 participants, including several new attendees. While no formal assessment was made of these events, notes were taken about possible future activities and actions for campus (summarized in the Appendix 1). Based on these informal discussions several faculty members and staff members intend to implement alternative practices on campus to reduce ST.

While planning changes was a desirable outcome, it was important to know if the workshops had actually equipped employees to implement change. Thus, in the post-workshop survey two paired questions (Table 3) were asked about employee skills in avoiding ST during classroom and mentoring activities. Although survey scores indicate that VU employees feel fairly skilled at avoiding ST in both classroom (3.81/5) and

informal settings (4.19/5), they thought that the workshops still improved their abilities. It should be noted that the standard deviation on improving their avoidance of informal ST was higher when compared to the standard deviation for improving classroom skills. This is because the distribution of responses was skewed towards '5', with more employees feeling as though they improved greatly in this area.

Based on the formal and informal assessment collected so far, our intervention has been successful in making employees both aware of and desiring to reduce ST on the VU campus. Because the events occurring during the middle of the spring semester, it is still too early to ascertain whether Dr. Good's workshops and lectures will have a lasting influence on campus culture or faculty instruction. However, based on the special follow-up discussions, an optimistic view is certainly justified. This was strongly supported

Table 3.—Faculty and staff skills.

	Very low (1)	Low	Neutral	High	Very high (5)	Average	Std. dev.
Please rate your ability to engage with students in a classroom setting without introducing stereotype threat	0	0	5	7	1	3.81	0.66
How did the workshop impact [your ability in the classroom]?	0	0	0	7	6	4.38	0.62
Please rate your ability to engage with students in an informal setting (mentoring, office hours, etc.) without introducing stereotype threat	0	0	1	9	3	4.19	0.54
How did the workshop impact [your ability to informally mentor]?	0	0	2	3	8	4.38	0.81

Table 4.—Faculty and staff perceptions of administration.

I believe...	Strongly disagree (1)	Disagree	Neutral	Agree	Strongly agree (5)	Average	Std. dev.
VU's administration is working to address issues of stereotype threat	1	5	3	2	2	2.92	1.26
VU's administration is working to address issues of equality (gender, ethnicity, etc.)	0	3	2	5	3	3.62	1.12
I believe...	Significantly decrease (1)	Decrease	Not Chnage	Increase	Significantly increase (5)	Average	Std. dev.
that the effort Valparaiso University expends towards reducing stereotype threat should ...	0	1	2	7	3	3.92	0.86
that the effort Valparaiso University expends toward reducing inequality (of any sort) should ...	0	0	4	4	5	4.08	0.86

by the four survey questions (Table 4) about current and future efforts of VU's administration.

Results from students and community.—The secondary goal of educating the student body was carried out explicitly through the open lecture by Dr. Good. Similar questions to those for the employees about general understanding of the effects of ST were asked (Table 5). As with VU employees, these showed strong and

positive results. In addition to helping students understand ST, it was important to discover what students currently felt they were experiencing. Table 6 displays the two questions asked – one about their experiences and one about their perceptions of the administration. These results were disappointing since most respondents felt they in fact had experienced ST and were not persuaded that the adminis-

Table 5.—Student and community understanding of stereotype threat.

	Strongly disagree (1)	Disagree	Neutral	Agree	Strongly agree (5)	Average	Std. dev.
From this event I learned a lot of new things about stereotype threat	0	0	4	5	15	4.46	0.78
Based on this event, I feel more able to identify when I am experiencing stereotype threat	0	0	2	10	12	4.42	0.65
I believe that stereotype threat can negatively impact performance of underrepresented groups (e.g. gender, ethnicity, etc.)	0	0	0	8	16	4.67	0.48
I believe that stereotype threat can negatively impact performance of any group or person.	0	0	2	8	14	4.50	0.66
I believe that stereotype threat represents a real, measurable phenomenon	0	0	1	6	16	4.65	0.57

Table 6.—Student and community experiences.

	Strongly disagree (1)	Disagree	Neutral	Agree	Strongly agree (5)	Average	Std. dev.
I feel that I've experienced stereotype threat in a course or activity related to Valparaiso University	4	3	3	2	9	3.43	1.63
I believe VU's administration is working to address issues of equality (gender, ethnicity, etc.)	0	3	9	6	4	3.50	0.96

tration was addressing equality issues. Finally, while only a tertiary goal, it was hoped the students would be better equipped to deal with ST personally. Here we succeeded admirably (Table 7).

DISCUSSION

Impact of results.—The overall responses to the survey questions show that faculty, staff, and students have benefited from this project. Furthermore, in the surveys immediately following the events, the faculty and staff respondents overwhelmingly (92%) indicated a plan to change their personal practices. A slightly smaller portion actually felt prepared to implement changes (77%). Only one participant indicated s/he did not actually feel prepared to implement changes. However, these survey responses and attendance numbers serve only as indirect measures of change.

During the two months following the lectures and workshops, we held several follow-up discussions for faculty, staff, and students. These discussions focused on changes for Valparaiso's campus and generated many ideas for reducing ST. We have generalized some of these ideas and included them in the Appendix 1 to spark further conversations. The discussions were well attended (over 40 employees and 30 students) and even attracted new participants because of the initial

attendees' enthusiasm. This enthusiasm has had important, tangible outcomes such as the welcome packet for new women engineers developed by current female students. Perhaps the most promising and important outcome was the participants' interest in continuing the process of addressing ST into the next academic year.

More directly, our longitudinal data collected almost a year after Dr. Good's visit show that participants generally followed through on their intent to change. Specifically, Fig. 2 shows that 71% of respondents made a change within a year. In addition, 93% were interested in seeing the same or a similar workshop offered again. Overall, the survey had a 28% response rate out of the employees who participated in the lectures, workshops, and follow-up discussions. In addition to these results, 15 faculty members have been participating this year in a learning community focused on growth mindset, one of the acknowledged ways to address ST.

Generally, these results indicated a positive change in campus culture. Yet, comparing the employees' responses to the students' experiences raises an important concern. Faculty indicated they were able to avoid ST in the classroom and informal mentoring (Question 1 & 3, Table 3), while some students clearly indicated that they had experienced ST (Question 1, Table 6). We are concerned about this discrepancy but acknowl-

Table 7.—Student and community preparation.

	Strongly disagree (1)	Disagree	Neutral	Agree	Strongly agree (5)	Average	Std. dev.
Based on this event, I feel more able to withstand or otherwise combat stereotype threat (against myself or others)	0	2	2	8	12	4.25	0.94

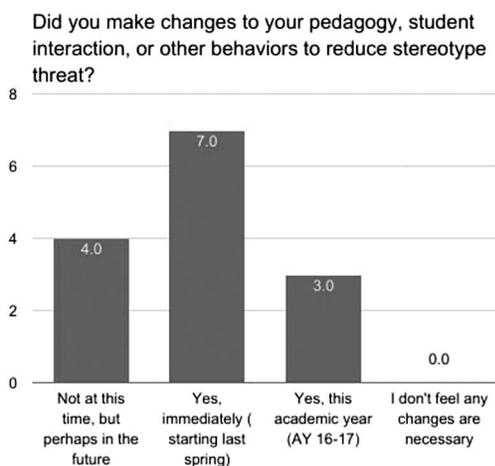


Figure 2.—Survey results from 1 year after events.

edge that the students perspectives may not refer to the specific faculty surveyed. However, it would not be entirely surprising for faculty to report not performing ST while students actually did report experiencing ST. Previous work has shown a measurable failure of faculty to correctly assess student's thoughts and views (Schmitt et al. 2015).

Transferable lessons.—There are two lessons from the project that are transferable to other institutions and professional development activities. First, even though the primary target of our program was faculty and staff, the student leadership proved immensely valuable in persuading the faculty and staff to participate. If the subject of the event is relevant to students, consider including student leaders or advocates, even if they are not the intended audience.

Second, the greater lesson comes from the value of originally unplanned follow-up activities related to the speaker. By having a series of lectures,

workshops, and then subsequent discussions with students, faculty, and staff, we were able to deepen engagement with the topic. The lecture, with workshops on the following day, while akin to a traditional conference event, did not sufficiently deepen our discussions. Instead, by having structured conversations nearly two months later, participants were able to return to the idea and better process what they had learned about ST. This strategy fits well with the pedagogical idea of mastery-based learning and, more generally, the power of repetition (Kulik et al. 1990). This extended engagement model is replicable for any high-profile speaker visit or event on a campus.

Future work.—When someone experiencing or creating stereotype threat is able to identify and explain the psychological problem, s/he has made the first step towards overcoming it. The education conveyed through our project has made that identification possible for many students, faculty, and staff. More broadly, the administration is reviewing the recommendations generated in the discussions (see Appendix 1) for feasibility and implementation. Overall, this project has served as an initial call to action for students, faculty, and administrators to address situations of stereotype threat. With a broader population aware of the challenge, Valparaiso University has made the first steps towards identifying and mitigating stereotype threat on our campus.

ACKNOWLEDGMENTS

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APPENDIX 1

Specific Classroom/Pedagogical Thoughts on Reducing ST:

- When doing assessments (especially pre/posttests, or scholarship of teaching work) be sure to avoid saying or implying that the tests reflect student abilities, that is, “intrinsic” traits, rather than “current state”
- When returning grades, be careful of attributions. A student's *work* receives a grade, not the student. Example: “Your paper has received a grade of XX”
- Hide or remove names from online classes to help avoid gender bias

- Include a “quality of failure” grade (See articles by Edward Burger, 2012)
- Include discussions of “growth mindsets” throughout the semester, not just at the beginning or before exams.
- Find ways to encourage “communities of belonging” within academic disciplines
 - Departmental social events
 - Discipline-based study tables
 - RA led/dorm-based study tables

Ways to Promote/Spread knowledge about stereotype threat (ST)/growth mindsets (GM)

- Hold a faculty learning community on Stereotype Threat or Growth Mindsets
- Include programming in required freshman courses
- Organize Residential or Greek Life programming
- Relate to faculty the importance of these issues to Freshman/Sophomore retention
- Provide more knowledge to faculty about student backgrounds and experiences, possibly through presentations by recruitment/admissions staff
- Blog Posts or emails from the university's Teaching and Learning Center/Staff

- Provide fliers, printable websites, or other resources to faculty and advisors about ST/GM
 - A "Top 10 Things Faculty Say" about inducing ST, and how to avoid them
 - General information about the effects and how to avoid ST
- Groups of People who may need training in Stereotype Threat/Growth Mindsets
- Professional Advisors
- Faculty, especially those teaching:
 - Freshman courses
 - General Education/Study Skills courses
- Peer Tutors (and tutoring center directors)

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