Final Report

EVALUATION AND MONITORING OF TERRESTRIAL AND AQUATIC INSECT BIODIVERSITY IN FORESTED AND CLEARED WATERSHEDS AT CAMP ATTERBURY, INDIANA

Submitted by:

Steven J. Taylor[1], Andrew V. Suarez[2], Christopher H. Dietrich[1],
Karlene Ramsdell[1] and S. Casey Funderburk[3]

[1]Center for Biodiversity, Illinois Natural History Survey
607 East Peabody Drive, Champaign, IL 61821

[2]Department of Entomology & Department of Animal Biology
University of Illinois at Urbana-Champaign
286 Morrill Hall, 505 S. Goodwin Ave., Urbana, IL 61801

[3]Department of Entomology
University of Illinois at Urbana-Champaign
286 Morrill Hall, 505 S. Goodwin Ave., Urbana, IL 61801

3 February 2005
Illinois Natural History Survey
Center for Biodiversity Technical Report 2005 (1)

Prepared for:

U.S. Army Engineer Research and Development Center
ERDC-CTC, ATTN: Michael L. Denight
2902 Newmark Drive
Champaign, IL 61822-1076

Executive Summary

Camp Atterbury is a 33,132 ha military installation near Edinburgh, Indiana. Construction of a 80 ha (4,550 ha with safety fan) Multi-Purpose Training Range (MPTR) began in 1998, and supports training for military vehicles and dismounted infantry, with a variety of stationary and moving targets. This study provides a baseline for long term monitoring and evaluation of natural communities to assess the impacts of construction of, and training in, the MPTR.

We assessed both aquatic macroinvertebrate and terrestrial insect community diversity, abundance, and richness and similarity at a series of study plots using quantifiable, repeatable and replicated methods. These data provide baseline data facilitating long-term monitoring and assessment as a measure of ecosystem health, and allow evaluation of relationships between community composition and habitat metrics.

Methods

Eight terrestrial study sites, each comprised of a 30 m square plot, were randomly selected, with four of these placed in the cleared portions of the MPTR and four placed in adjacent upland forest. We used several sampling methods, with focus on three groups of taxa (all insect taxa, ants, and leafhoppers and kin) and compared the efficacy of both the methods and the groups as monitoring tools. Sampling methods included: 1) a Malaise trap (mesh tent-like device that captures flying insects) at each site; 2) four sweep sample transects at each site; 3) four leaf litter samples from each site, with invertebrates extracted using the Winkler method; and 4) Nine pitfall traps at each site. Samples were collect during Summer and Fall study periods, and this report gives results from the Summer sample period. Several habitat parameters were recorded, including a vegetation index, canopy cover, ground cover, and leaf litter depth. Dominant plant taxa were collected, and data loggers recorded soil and air temperature during the study.

We sampled aquatic macroinvertebrates at three stream sites draining the MPTR. Invertebrates were collected in replicate samples with a dipnet and these were sorted and subsampled in the laboratory. Canopy cover and basic water chemistry data were collected, and data loggers recorded changes in terrestrial and aquatic temperature. An index of biotic integrity and taxon richness were used to evaluate the aquatic communities.

Results and Discussion

At least 409 taxa and 3776 specimens were collected at terrestrial sample sites during the Summer sampling period. In general, there were some differences among sites, among sampling methods, and among treatments (cleared MPTR versus forested) when we examined taxon richness and species diversity, but these differences could not always be fully resolved. While taxon richness and species diversity differed among treatments, and, in general, plots in the two treatments harbored different insect communities. Species accumulation curves and various estimators of taxon richness were used to evaluate the four sampling methods and the three groups of taxa (all taxa, ants, leafhoppers). Based on the performance of the different taxa (all, ants, leafhoppers) compared across the different methods (malaise sampling, Winkler extracted leaf litter samples, pitfall traps, and sweep samples), the single most effective taxon for monitoring was found to be the ants (Formicidae), and the single best method for monitoring was found to be pitfall trapping.

We collected 818 specimens, primarily aquatic macroinvertebrates, from the three stream sites during Summer sampling. All three streams were dry during the fall sample period, and thus no aquatic macroinvertebrates were collected. Using Hilsenhoff's (1988) family-level index of biotic integrity, water quality was classified as "good" at one site, and "fair" at the other two, although taxon richness was lowest at the site classified as good. In addition to invertebrates, numerous salamanders (*Eurycea cirrigera*, the Two-lined Salamander) were observed in the streams.

For aquatic invertebrates, we found that the small upstream portions that directly drained the MPTR only held water seasonally, and thus were not effective sites for monitoring of stream macroinvertebrates. There was insufficient separation between MPTR-influenced stream sites and control sites, and a lack of replication (few streams flowing away from the MPTR) precluded robust statistical analysis of the data we did obtain. The community of aquatic macroinvertebrates collected during this study appeared similar to the communities reported by Robinson (2004) elsewhere at Camp Atterbury in larger streams, and includes taxa typical of rocky bottom Midwestern forest streams. Fish were largely absent due to the intermittent nature of the streams. Salamanders were abundant in the streams, and because they are top predators in this seasonal habitat, they may be suitable subjects for studies of potential bioaccumulation of toxins.

This study provides a snapshot of insect biodiversity at a point in time, thus providing baseline for any possible future monitoring of insect biodiversity. Sampling methods and analyses developed in this study could easily be implemented at a wide variety of other military installations to facilitate inventory and/or monitoring of insect biodiversity.

Introduction

Camp Atterbury (U.S. Army Atterbury Reserve Force Training Area), near Edinburgh, Indiana, is a 33,132 ha military installation occupying portions of Bartholomew, Brown, and Johnson counties. Approximately 9,000 ha of this installation have been deeded to the Indiana Department of Natural Resources, the Johnson County Department of Parks and Recreation, the Indiana Department of Corrections, and the Indiana Department of Labor. The majority of the non-deeded portions of Camp Atterbury lie in Bartholomew County, in the Scottsburg Lowland physiographic unit (Malott 1922) and the Norman Upland. Camp Atterbury is entirely within the White River Drainage Basin (Schumaker *et al*. 1999). Established in 1942, this facility is used in training National Guard and other reserve forces (including Air National Guard and Air Force Reserve), federal and state law enforcement officers, and state emergency personnel.

The habitat at Camp Atterbury is primarily forested, with unforested land including the cantonment area, training areas, a multipurpose training range, firing ranges, support facilities, and an airport. Our study focuses primarily on monitoring and evaluation of a Multi-Purpose Training Range (MPTR), constructed between 1998 and summer of 2003. The MPTR is divided into three areas for support, firing and targets. The MPTR supports training of tanks, attack helicopters, infantry fighting vehicles, and dismounted infantry. The total area of the MPTR is 80 ha, and with a safety fan total area is 4550 ha. Stationary (Figure 1a) and moving (Figure 1b) targets are located in the northeastern portion of the MPTR, such that trajectories go into the impact area located in the center of Camp Atterbury.

Our protocol is designed to allow monitoring and evaluation of the impacts of construction and military training in the MPTR on natural communities. Much of the upland habitat in this part of Camp Atterbury recently (~1998 and later) has been cleared of forests, and berms have been built to facilitate use of stationary and moving targets. Our study design entails monitoring and evaluation of insect communities in the cleared and adjacent forested uplands, as well as monitoring and evaluation of aquatic macroinvertebrate faunas of three headwater tributaries draining the MPTR.

Degradation of landscapes often results in erosion, water pollution, and loss of habitat for species (Jansen 1997). Military training can have environmental consequences similar to those recorded for military actions during wartime (e.g., Lanier-Graham 1993, Lehman *et a*l. 1999, Austin and Bruch 2000, Milchunas *et al*. 2000, Whitecotton *et al*. 2000, Ehlen and Harmon 2001, Dudley *et al*. 2002, Fang *et al*. 2002). Degredation of landscape in military training areas can result in significant reductions in plant diversity (Dale *et al*. 2002), yet military lands can also function as relatively large and undisturbed (or less fragmented) refugia for sensitive taxa that may be declining as a result of habitat degradation on adjacent lands (e.g., Anders and Dearborn 2004, Carvell 2002, McKee and Berrrens 2001). For example, Pogue and Schnell (2001) found that habitat complexity of prairie-forest ecotones on agricultural lands was less than the habitat complexity on adjacent to a Fort Sill Military Reservation, with implications for positive associations between military land use practices and the native flora and fauna. Furthermore, disturbances associated with military training may maintain certain disturbance-associated communities to the benefit of rare or endangered invertebrate species (e.g., Smith *et al*. 2002).

Restoration ecology attempts to bring the dynamics and diversity of natural ecosystems – including both plants and animals – back towards their original condition by active intervention to repair damage caused by human activities (Atkinson 1988, Jackson *et al*. 1995). Land managers are often concerned with revegetation of land to achieve restoration of disturbed lands, with little consideration of other ecosystem components (Majer 1990). The success of land restoration is commonly evaluated through evaluation of vegetative structure and composition. To approach complete restoration, however, it is necessary to work towards recovering the entire ecosystem (National Research Council 1992).

Cover Photo: Adult male “Wedge-Shaped Beetle” (Coleoptera: Rhipiphoridae) collected in sweep sample at Camp Atterbury, Indiana, July 2004. Scale bar is 5 mm.

A

B

Figure 1. Targets in MPTR at Camp Atterbury, Indiana. A. Stationary target. B. Moving target. Photos by Steve Taylor, 3 November 2003.

The diversity and dynamics of invertebrate communities is typically closely tied to plant communities (Jonas *et al.* 2002). Invertebrates play an important role in the diversity, structure, and dynamics of disturbed ecosystems (Majer and Nichols 1998), and because they 1) function at multiple trophic levels, 2) encompass a variety of functional groups, 3) vary greatly in life history strategies, 4) are sensitive to microclimate, 5) play a critical role in nutrient cycling, and 6) provide an important food source for larger wildlife (Figure 2), insects are reflective of the functioning of the ecosystem as a whole (Jansen 1997, Kim 1993, Kremen *et al.* 1993, Miller 1993, Oliver and Beattie 1993). Furthermore, insects are typically present in large numbers, facilitating statistically meaningful comparative analyses. Insects have excellent potential as indicators of restoration success (Disney 1986, Greenslade and Greenslade 1984, Rosenberg *et al.* 1986). In particular, Landres *et al.* (1988) and Burke and Goulet (1998) have argued that insects are appropriate for assessing biodiversity in smaller areas or in the case of short term disturbances.

Figure 2. Terrestrial invertebrates provide an important food source for games species, such as these wild turkeys on a berm in the Multi-Purpose Training Range (October 2004).

Insects are commonly used for assessment of aquatic systems (Barbour *et al.* 1999; Hellawell 1978; Hilsenhoff 1987, 1988; James and Evison 1979) and in recent years, they also have been used in terrestrial systems (see below). In North America, invertebrate assessment in aquatic systems typically utilizes all macroinvertebrate taxa, and follows methods (Barbour *et al.* 1999) based on Hilsonhoff's pioneering work in Wisconsin (1987, 1988). Assessment of terrestrial systems using invertebrates is less standardized. One approach is to utilize all terrestrial invertebrate taxa (generally identified to family level for insects, and at least order for most other taxa) obtained by one or few sampling methods (e.g., Fay 2003, Jansen 1997, Jonas *et al.* 2002, Longcore 2003, Nakamura *et al.* 2003). The other common approach is to utilize a particular taxon such as butterflies (Kremen 1992), ants (Andersen and Sparling 1997, Bestelmeyer and Wiens. 1996, Majer and Nichols 1998), or beetles (Jonas *et al.* 2002, Rieske and Buss 2001, Villa-Castillo and Wagner 2002, Watts and Gibbs 2002) – again, generally obtained by one or few sampling methods.

New (1998, Chapter 8) reviews the available literature on monitoring and status evaluation of invertebrates. Field implementation of monitoring (modified from Goldsmith 1991) should 1) document long-term environmental change and its ecological effects, 2) document responses of study taxa to changes in management practices, 3) assess effectiveness of a management regime for species assemblages under study, and 4) document changes and rates of change in populations and habitats under study.

Objectives

We here provide a protocol for monitoring and evaluation of the MPTR and present results of preliminary field sampling. This study, when carried out over several sampling periods, will allow us to:

1. Compare the diversity, abundance and richness of invertebrate taxa, primarily insect families, under two land management regimes differing in level of disturbance, focusing on selected terrestrial and aquatic faunas.

2. Provide baseline data for land managers to facilitate quantitative, long-term monitoring and assessment of aquatic and terrestrial invertebrates as a measure of ecosystem health and recovery.

3. Evaluate possible relationships between invertebrate community composition and selected habitat metrics.

Methods

Terrestrial and aquatic sampling in 2004 data were available for only one season for aquatic invertebrates (Summer 5-9 July) because of loss of earlier data (graduate student quit, leaving a fair amount of disorganization) and because the study streams were dry during the fall sample period (10-15 October). For terrestrial invertebrates,

samples were collected in Summer and Fall sample periods. However, sample sorting and identification turned out to comprise a much larger task than anticipated, and thus only samples from the summer sampling period could be sorted and identified in time to include in the analysis for this report.

Aquatic Sampling Sites:

The few streams that are present in the MPTR are small and intermittent. We have selected three sites that drain from the MPTR towards the Driftwood River to the east, and then into the East Fork of the White River near Columbus, Indiana.

1. Site #1 (Figure 3a) is in a headwater tributary on the western border of the firing area – this stream is almost certainly intermittent, and we expect to see little to no water here during longer dry periods. The watershed upstream of the ca.100 m stream reach which we sampled is almost completely impacted by the clearing of the forest. Fallen trees and obvious erosional washouts (Figure 3b) are common. This stream was actively flowing in the Spring and Summer of 2004, but was completely dry during the Fall sampling period.

2. Site #2 is located at a lower elevation in a forested area that appears to partially drain the support area of the MPTR, but also drains adjacent forested lands. It is possible that this stream, somewhat broader than Site #1, dries completely during long dry periods (Figure 4a), and was found to be dry during the Fall sample period.

3. Site #3 is a somewhat larger tributary stream with deeper pools (Figure 4b) and a broader riparian buffer located at a slightly lower elevation than Site #2. This stream appeared to be perennial, but was found to be dry during the Fall sample period. It drains mostly forested lands, but also portions of the firing area and target area.

At each of the three sampling sites, we marked three stream segments, each ~100ft (~30m) in length. Two macroinvertebrate samples were collected in each segment during each trip, one from each of two dominant aquatic habitat types (e.g., riffle, bank, or pool), for a total of six replicate samples at each of the three streams during each sample period. When two habitats were not reasonably available, only one habitat was sampled. Samples were collected using a heavy duty net (Figure 4a, foreground), and larger organic debris and rocks in the sample were washed over a bucket before removal from sample. Field samples were elutriated and decantated using buckets, water, and fine mesh aquarium nets when large quantities of sediment were present. Each replicate sample was preserved in diluted formalin in a whirlpak bag labeled with site date, replicate and habitat data. Samples were preserved in diluted formalin in the field, and were processed through step 2 (below) as soon as possible to replace formalin with ethanol.

In the laboratory, we used a quantitative subsampling method to obtain approximately 100 randomly selected macroinvertebrate specimens from each sample. This procedure has been used successfully for more than 6 years by one of us (SJT) in processing aquatic samples from Midwestern streams.

Laboratory procedure, aquatic samples:

1. Gently dump replicate sample into a 0.5mm (=500 microns [µ]) mesh sieve which has been placed over a funnel with a waste fluid disposal container beneath it, or over a pan. This process should be completed in a sink in case there are spills. Allow excess formalin to drain from sample. Rinse with a small amount of water. Save waste formalin for proper disposal.

A

B

Figure 3. A) Aquatic sampling site #1, note the abundance of downed timber. The sampling reach extends a short distance into the adjacent forested area. B) Erosional washout near aquatic sampling site #1 in the MPTR, Camp Atterbury, Indiana. Riprap in background has been placed, apparently, to control erosion. Photographs by Steve Taylor, 3 November 2003.

A

B

Figure 4. Aquatic samplings at Camp Atterbury, Indiana. A) site #2, B) site #3, Camp Atterbury, Indiana. Photos by Steve Taylor, 3 November 2003.

2. Transfer sample from sieve into a beaker of water, rinsing sieve as needed to transfer material. Gently stir the sample in water in the beaker. This serves to both dilute the formalin further and to randomize the distribution of organisms in the sample. Return sample to the sieve for final draining.

3. Gently remove sample from sieve to gridded (5 grid marks by 3 grid marks) white pan (26 x 35 cm) and evenly distribute the contents across the bottom of the pan.

4. Using a random numbers chart, select a grid coordinate and place a 4.6 cm diameter, (16.62 cm^2) cylindrical "cookie-cutter-like" plastic (Lexan) subsampling insert into the sample, centered at the grid coordinates. Gently work the insert down into the sample.
   - Debris overhanging the grid may be cut with scissors.
   - Organisms which are more than half inside of the insert should be included in the subsample. If it is uncertain whether one half of an organism is within the insert, then it should be included.

5. Pick all specimens from within the insert, preserving them in 80% alcohol. If fewer than 110 animals[1] are obtained, return to step 4. Continue to repeat steps 4 and 5, each time randomly placing of the insert, until 110 animals are obtained. Once sufficient organisms have been obtained, quantitative subsampling of that replicate is done.
   - Do not reuse the same coordinates to subsample more than once in a sample.
   - If all fifteen possible coordinates have been subsampled, yet collectively have yielded fewer than 110 organisms, pick from the remaining sample without using the subsampler until 110 specimens are obtained or the material is completely searched.

6. Samples should be placed in separate vials (70-80% ethanol) from the remainder of the sample. Label samples clearly with collection locality data, site, habitat type, replicate number, taxa that have been included (e.g., annelids, chironomids). Record sorting data on bench sheet.

7. Place the remaining sample (after subsampling is complete) into clearly labeled whirlpak(s), add 80% alcohol as needed, and label with collection locality data, site acronym, habitat type, and replicate number, and clearly indicate on the label that the sample has already been subsampled. Place the whirlpak(s) in a large nalgene jug for long term storage.

Subsamples, now reduced to approximately 100 organisms, were then sorted and identified to family level for most insects (to the most specific taxon reasonable for other taxa such as Annelida, Gastropoda, etc.) - in the laboratory under a dissecting microscope using standard identification guides (e.g., Merritt and Cummins 1997, Smith 2001, Thorp and Covich 2001, Voshell 2002). Numbers of individuals of each taxon were recorded along with the identification, site, habitat, and replicate. These data were then entered into a computer database, where they were analyzed using previously developed data processing procedures in SAS PC (SAS Institute 2001). Metrics produced by this method include taxon richness and a family-level index of biotic integrity (IBI), following Hilsenhoff (1988). These metrics were then compared across sites.

In addition to collecting invertebrates from the streams, we collected basic water parameters during each sampling period. Volume of flow was measured with a flow meter, but only when sufficient flow was present to allow use of the meter (at least 5 cm depth and sufficient velocity are needed). Flow was measured using a Global Flow Probe (FP101, Global Water, Gold River, CA) (Figure 5a). Stream width was measured with a fiberglass open reel tape or meter stick, the width then divided into equal segments, each generally 20-30 cm wide. The water depth was measured in each segment and the average velocity calculated by averaging the velocity at each depth measurement point. Velocity was averaged while slowly moving the probe up and down from just below the water surface to the stream bottom. The flow value within each segment was recorded when a steady average velocity reading was achieved. Volume of flow was calculated using the formula:

$$R=\sum_{s=1}^{i} w_s d_s v_s$$

[1] The 110 animals are all macroinvertebrates (arthropods, mollusks, flatworms, annelids, etc.).

Where **R** is the volume of flow (m3/sec), **s** is the segment number, **i** is the number of segments, **w** is the segment width (m), **d** is the segment depth (m), and **v** is the segment average current velocity (m/sec). Volume of flow data are presented in gallons/minute (gpm), with calculations carried out using computer program (SAS).

Other water parameters measured included water temperature, pH, turbidity, specific conductance, resistivity, total dissolved solids and dissolved oxygen. Turbidty was measured with a turbidity meter (Figure 5b), and most of the remaining parameters were measured with a YSI 556 multiparameter meter (Figure 5c). A Hobo tidbit ® data logger was placed each stream beginning in the summer of 2004 to allow continuous (hourly) logging of water temperature at each site. Air temperature and relative humidity data were also collected at one stream site (site 2) using a HOBO Pro RH/Temp Data Logger mounted on a tree with a white plastic cover for shade and rain protection. Canopy cover was estimated at each stream site during the summer by walking a transect down the middle of the stream segment, and taking 10 canopy photographs in each stream segment, for a total of 30 images at each stream. Photographs were tallied (open/canopy) as described in Taylor (2001) using a computer and an overlaid grid of 100 points to score each image (Figure 6).

Upland Terrestrial Sampling Sites:

Eight sites associated with the MPTR and located in forested and cleared upland areas, were selected (Figure 7). Four sites are in disturbed areas (cleared ca. 1998, or later) and four are in forested areas (Figure 8). Plots are 30 m x 30 m in size, and were randomly placed in designated study area, with the restrictions that each had at least a 10 m buffer of similar vegetation type, and none intersects a road or other man-made structure. Each plot was marked with wire flags, stakes and/or flagging tape.

Our strategy for upland terrestrial sampling was to look at broader taxonomic diversity by examining morphospecies of insects in quantitative samples, and to evaluate two groups ants (Hymenoptera: Formicidae) and leafhoppers (Homoptera: Cicadellidae), which serve different functional roles within the community, by quantifying species-level diversity and abundance. Leafhoppers are strictly herbivores with piercing sucking mouthparts, and feed on a wide variety of vascular plants. The diversity of Midwestern cicadellids is relatively high (DeLong 1948), and they exhibit a wide range of host plant specificity. Ants differ considerably from the leafhoppers in being colonial, and are more diverse in their ecological niches and food sources (e.g., may feed on plant exudates, seeds, fungi, excreted honeydew of aphids, and a variety of animal prey) and interact with organisms at nearly every trophic level. In addition to providing an interesting cross section of the ecosystem, focusing on these two taxonomic groups plays to the taxonomic expertise of two of us (AVS-ants, CHD-leafhoppers).

To further characterize the differences between cleared and forested uplands, we used four sampling methods, each with a different bias in sampling (New 1998, Agosti *et al*. 2000). Pitfall traps will be used to sample ground-dwelling fauna, Winkler samples will be collected for leaf litter fauna, sweep samples will focus on fauna living on grasses, herbs, and smaller woody vegetation, and finally malaise traps will be used to sample flying insects.

Upland Invertebrate Sampling

Four sweep sample transects, each 20 m in length, each comprised a sample at a particular site, and were collected in each upland plot during each season. Each 20 m transect was comprised of 40 sweeps of a heavy duty sweep net (Figure 9). Following Wallner (2004) each transect sample was placed in a “photo tactic optimal insect extractor” (PTOIE), an insect extraction device (Figure 10) developed by one of us (CHD). After about half an hour (mean±standard error: 30.69±1.82 minutes), the whirlpak bag, containing ethanol (70-80%) attached to the bottom of the PTOIE was removed, and the insects in the bag constituted the sweep sample. In addition, the remaining contents of the PTOIE were placed in a white pan or sheet, where no more than 5 minutes were be spent searching for and collecting ants by hand and with an aspirator, ignoring the remaining taxa. The ant sample was preserved separately, but later was pooled with the remaining sweep sample fauna for data analysis.

A single malaise trap (John W. Hock Company, Florida), following the design of Townes (1972, see Figure 11) was placed adjacent to each of the 8 upland plots for approximately 24 hours to collect flying insects. Malaise traps were set up immediately prior to their use, and were removed at the end of each sampling period to limit opportunities for vandalism. The insects obtained by malaise trap accumulated in a reservoir of ethanol in the trap head. The contents of this reservoir constituted the malaise sample for a given plot during a sampling season. Unfortunately, the malaise samples from the summer collection are still not completely sorted and identified because we underestimated the amount of time it would take to process and identify the various samples. Thus the malaise data are excluded from this report.

Figure 5. A) Using flow meter to determine volume of flow, B) placing water sample in turbidity meter, C) using YSI multimeter to collect various water quality parameters – inset shows detail of display.

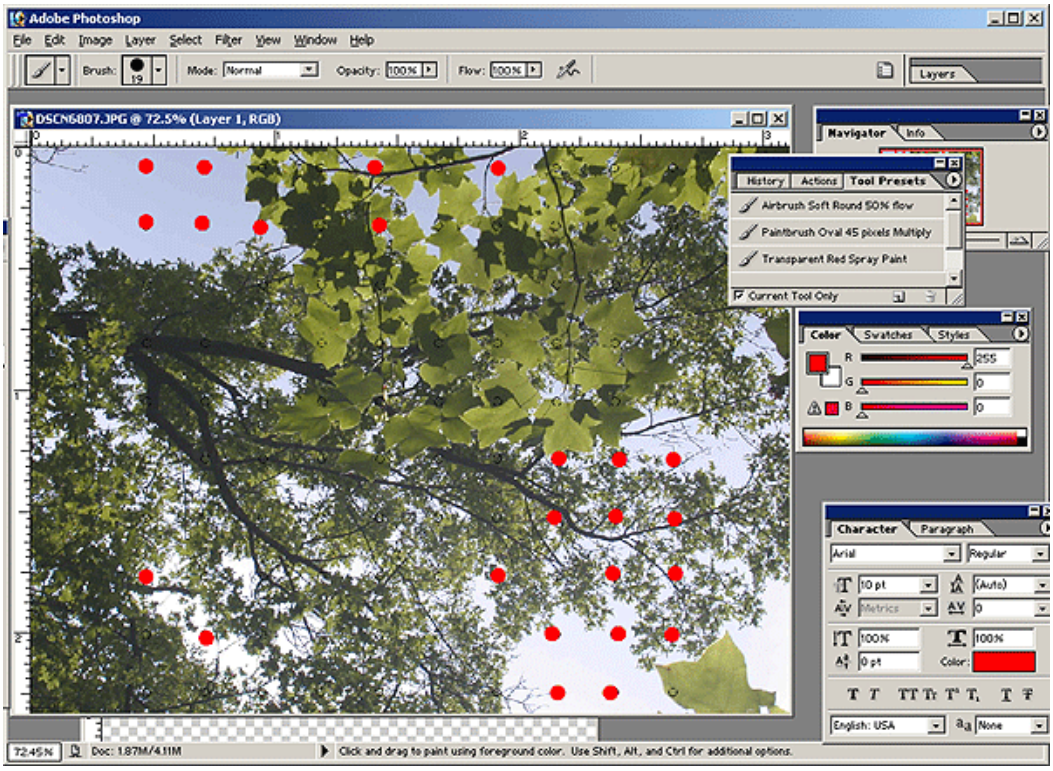


Figure 6. Image analysis of canopy cover photographs using computer. A 10x10 array of points is overlaid on the middle of each photographs and the number of open (here shown as red dots) and canopy grid points is scored to estimate percentage canopy cover. A few of the points scored as canopy can be seen as open black circles, especially on the light colored large leaves in foreground.

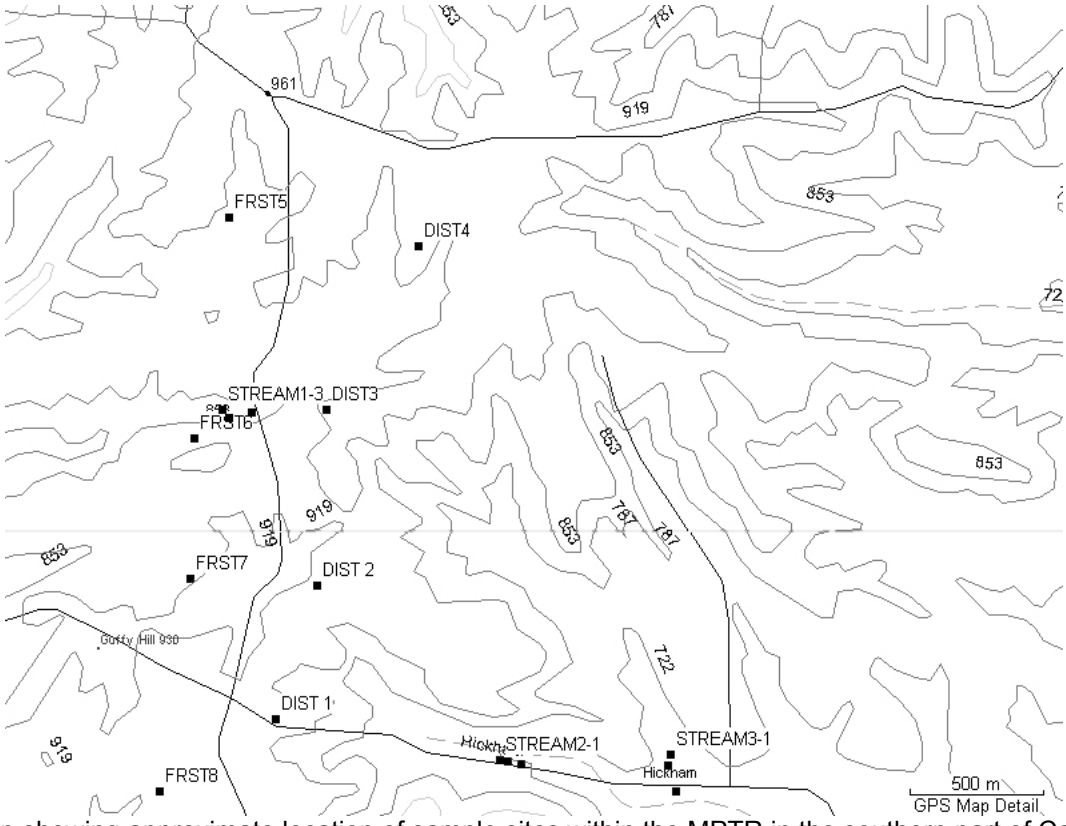


Figure 7. Map showing approximate location of sample sites within the MPTR in the southern part of Camp Atterbury, Indiana. Dist 1 – Dist 4 are upland sites on cleared land, Frst 5 – Frst 8 are upland sample sites on forested land, Stream 1 is labeled “Stream 1-3”, stream 2 “Stream 2-1” and stream 3 “Stream 3-1”.

Figure 8. Cleared (foreground) and forested (background) upland habitat in the MPTR study area at Camp Atterbury, Indiana. Stream 1 is located at the edge of the forest in the low area to the upper left of the photograph. Photo by Steve Taylor 3 November 2003.

Figure 9. Field crew running sweep sample transects at one of the cleared sites in October, 2004. Recent construction work on nearby berm has resulted in removal of surface vegetation.

Figure 7. Four “photo tactic optimal insect extractors” (PTOIEs) in operation. Sweep sample material is placed in darkened (duct-tape covered) chambers. Diurnal, positively phototactic insects are attracted to light coming from hole in lid, and eventually fall into the whirl pack bag, below, where they are preserved.

Figure 11. Malaise trap at site D-4, July 6, 2004. The puddle in the lower right and the soil on right edge of photo are impact craters from military training – these craters were not present at the time the sites were selected.

Figure 12. Field crew placing a pitfall trap at one of the cleared sites, July 2004. Special care is taken to ensure the lip of the trap is just below the soil surface to increase likelihood that invertebrates will be captured. Yellow flag marks pitfall station..

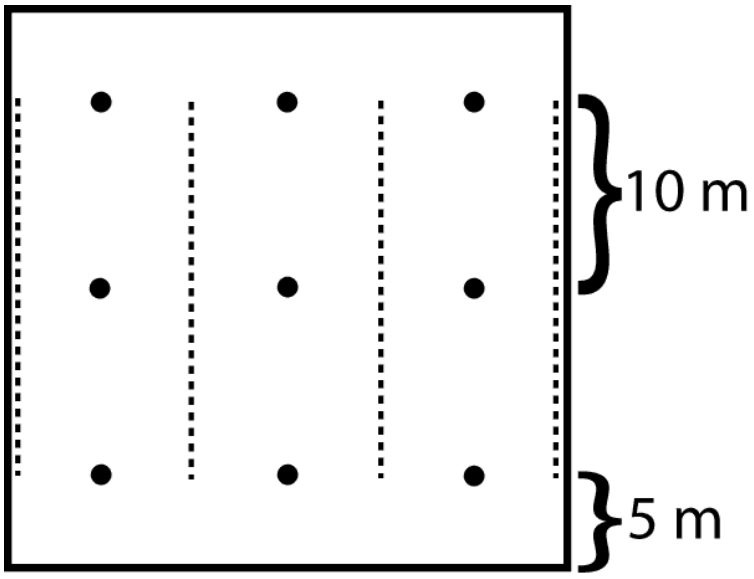


Figure 13. Diagrammatic representation of an upland terrestrial sampling plot. Plot is 30 x 30 m in size. Dashed lines indicate route of the four sweep samples, dots indicate location of pitfall traps. Distances of traps from edge of plot and between traps are indicated.

At the same time the malaise traps were set up, pitfall traps were opened, and ran for a ~24 hour sampling period. Pitfall traps were constructed from 50 ml centrifuge tubes, placed into the ground (Figure 12) such that the upper lip is just below the soil surface (see Bestelmeyer *et al*. 2000). At each 30 x 30 m upland sampling site, a 3 x 3 array of pitfall trap sampling points was established (Figure 9). The first placement of pitfall traps was at least several days in advance of the first sampling period to minimize the "digging-in" effect of settling and disturbance (Greenslade 1973). When pitfall traps were not in use, they were covered tightly but remained in the ground for the course of the study. Pitfall traps which were removed or disturbed by animals, etc., were replaced as needed. When in use, traps were partially filled with propylene glycol as a preservative. Traps (50 ml centriguge tubes with propylene glycol and invertebrates) were recovered and capped, and then were replaced with empty 50 ml centrifuge tubes. In the laboratory, trap contents were gently washed over a fine-mesh screen and transferred to 70-80% ethanol in jars or whirlpak bags, each trap in a separate sample container until specimens could be sorted and identified. The relationship between the location of pitfall traps and the sweep samples is shown in Figure 13.

Finally, we collected four leaf litter samples from each plot, using randomly placed 0.5 m^2 quadrats (0.701 x 0.701 m) to define the sample area (Figure 14 A). Leaf litter samples were processed using standard Winkler sample techniques (Bestelmeyer *et al*. 2000), including use of a sifter (Figure 14 B) and mini-winkler bags for extraction. Leaf litter samples were stored in a large cooler until processing could take place (within 24 hours), and samples were extracted for 2-3 days (Figure 14 C). Winkler leaf litter samples can collect many ant species that typically do not appear in pitfall trap samples (Olson 1991). Litter depth in the vicinity of each leaflitter sample was measured at a series of 10 randomly selected points at a distance of 0.5 m from the center of the quadrat.

Upland Habitat Metrics

Ground Cover class (bare soil, rock, woody plants, grass, dead wood, leaf litter) was tallied in 0.5 m^2 quadrats (0.701 x 0.701 m, Figure 15) quadrats centered on pitfall stations and based on digital images. The digital photos were taken at waist height (ca. 1 m), centered over the quadrat as much as is feasible. In the laboratory, an array of 10 x 10 sample points was electronically overlaid on the digital photos and substrate under each point was tallied as described in Taylor (2001). Similarly, a canopy cover photograph was taken at each pitfall point and tallied (open/canopy) as described in Taylor (2001) (Figure 6). Ground and canopy cover photographs were only taken once per year, during the summer sampling period.

Vegetation structure, especially understory structure which is generally poorly represented in the ground cover and canopy cover photographs, was characterized using a 3 m long rod (made from 1 inch PVC pipe), marked in 20 cm intervals. Number of intercepts of grasses, herbaceous vegetation, shrubs (<= 2 cm DBH), trees intersecting rod in each interval was scored (Figure 16). The rod was placed at 10 points along each of the 4 transects similar to sweep transects but perpendicular (east-west) to minimize interference of two procedures.

These data were then analyzed using midpoints of rod segment heights (10, 30, 50 cm, etc.) to produce a height index:

$$\text{height index} = \frac{\sum_{i=1}^{N} (h_i \times n_i)}{\sum_{i=1}^{N} (n_i)}$$

where N = the number of height classes (for a 3 m rod divided into 20 cm classes, this is 15); h_i = the midpoint of the height of the ith height class (e.g., 10, 30, 50 cm, etc.); and n_i = the number of intersections of vegetation with the rod in the ith height class. The height index was calculated using all vegetation types combined (grasses, herbaceous vegetation, shrubs, and trees), and histograms of vegetation types by height were produced. Similar rod intersect methods have been used by various researchers (e.g., Gibson et al. 1987, Hendrix et al. 1988, Levy and Madden 1933, Longcore 2003, Majer and Nichols 1998) to assess vegetation structure. Vouchers of the ten visually dominant plant species were collected into a plant press and brought back to the laboratory where they were identified by a botanist.

Near the center of each upland sample plot, a HOBO tidbit ® temperature data logger (Figure 17) was buried 2 cm below the surface of the ground, and temperature was logged at 1 hour intervals. One HOBO Pro RH/Temp Data Logger (Figure 18) was mounted in forested habitat and one in cleared habitat at an elevation of 1 m above ground and in the vicinity of the 8 upland sample plots to record temperature and humidity at hourly intervals.

Figure 14. Leaf litter sampling using the Winkler method. A. Collecting leaf litter from 0.5 m^2 quadrat into bag (October 2004). B. Processing litter in field with sifter (July 2004). C. Sifted litter in mini-Winkler extractors in a garage (July 2004), each mini-Winkler represents and individual leaflitter sample, thus 32 mini-Winkler extractors were in use simultaneously. Invertebrates are collected in the whirl-pak bag with ethanol attached to the bottom of each extractor.

Figure 15. A PVC pipe ground cover quadrat with an area of 0.5 m^2, used to characterize the proportions of different ground cover types.

Figure 16. Field crew collecting data for Vegetation Height Index at one of the forested sites, using a 3 m pole marked off in 20 cm increments, July 2004.

For all upland samples, plot number, replicate number, sample number, sample type, and other relevant data were carefully associated with each sample throughout collection, processing, identification, and data analysis. Insects were usually identified to family and to morphospecies. Individual morphospecies were assigned a number that was unique at least within their family, and a reference collection of morphospecies was established to facilitate identification of the same taxa in other samples. Non-insect arthropods to order (except Diplopoda and Chilopoda to Class), and non-arthropod invertebrates to Phylum or Class. Ants (Hymenoptera: Formicidae) and auchenorhynch Homoptera (Cicadellidae and kin) were identified to species level when possible. Identification was facilitated by the used of standard guides to identification (e.g., Borror *et a*l. 1989). Voucher specimens of selected taxa will be deposited in the Illinois Natural History Survey Insect Collection.

Analyses

To compare the diversity, abundance and richness of invertebrate taxa, primarily insect families or morphospecies, under the two land management regimes, we began by compiling taxon lists, descriptive summary data and summary statistics for each sampling method and each plot.

Using the morphospecies, a taxon list was developed for each study site, with taxon occurrence tallied by sample types (e.g., winkler sample, malaise trap, pitfall trap, sweep sample). Total taxon richness, number of individuals and a diversity metric were compared among sites (and within sampling method) using statistical procedures such as *t*-tests, ANOVAs, or, as appropriate, their nonparametric analogs. The diversity metric we used is the Shannon index, H', which is calculated as follows:

$$H' = -\Sigma \mathbf{p}_i \ln \mathbf{p}_i$$

where $\boldsymbol{p}_i$ = proportion of individuals in **i**th species

The Shannon index was chosen because it has been found to be robust when sample size effects are an issue (Leponce et al. 2004).

We also produced taxon accumulation curves for each site and for each sampling method to evaluate the extent to which we have effectively sampled the fauna (Soberón and Llorente 1993, Longino and Colwell 1997). We compared several non-parametric species richness estimators, including Chao1 (Chao 1984, 1987), a second order Jackknife estimate (Jackknife 2, Heltsche and Forrester 1983, Palmer 1991), an incidence-based coverage estimator (ICE, Lee and Chao 1994, Chazdon et al. 1998) and the bootstrap method. The species richness values derived from the above estimators are generally thought to be fairly conservative, with the actual number of species present often being considerably higher than is suggested by these metrics (Longino et al. 2002, Leponce et al. 2004). Richness metrics, and taxon accumulation curves were calculated using the software EstimateS 7.0 (© Colwell 2004, and see Colwell and Coddington 1994), and curves were smoothed with 1000 randomized orderings of the input data, where appropriate, to produce rarefaction curves (Gotelli and Colwell 2001). Shannon diversity (H') for individual samples was calculated using SAS (SAS Institute 2001) in combination with a basic program written by one of us (SJT).

Turning to the focal groups Cicadellidae (and kin) and Formicidae, we repeated portions of the above analyses within each of these two groups to describe our dataset more fully. In addition, available information on the biology and ecology of identified leafhopper and ant species are briefly described for those species which are well known.

Finally, we evaluate possible relationships between invertebrate community composition (various taxon and species metrics) and the habitat metric data collected during our study, including treatment (clear/forested), percent canopy cover, ground cover classes, season, and temperature.

Level of significance for all statistical tests was set *a priori* at p=0.05, and most statistical procedures were carried out using SAS PC (SAS Institute 2001).

Collectively, these data provide a robust baseline dataset that can facilitate quantitative, long-term monitoring and assessment of aquatic and terrestrial invertebrates as a measure of ecosystem health and recovery.

A 

B 

Figure 17. A. Digging up 2 cm soil temperature HOBO Tidbit data logger (in plastic bag). B. Downloading tidbit using Optic Shuttle.

Figure 18. HOBO Pro RH/Temp Data Logger mounted on tree (foreground and inset) at forested site F5 on 6 July 2004. Malaise trap is being set up in background.

Results

Upland Terrestrial Sites

Dominant Plants

We collected visually dominant plants (10 species, more or less) at each site to characterize the vegetation (Table 1). Cleared sites were dominated by typical old field species, with some prairie species among the dominants at D1, D2, and D3. Forested sites were dominated by species characteristic of mesic upland forests. Cluster analysis of species presence/absence data produced a dendrogram reflecting the overall division of sites into forested and cleared lands (Figure 19). In general, the forested sites formed a cluster distinct from the cleared sites, confirming the obvious overall difference in plant composition between the two habitat types. Within the forested sites, F5 and F8 were most similar to one another in dominant plant species. Sites D2 and D3 were more similar to one another than to the other cleared sites.

Vegetative Metrics

Canopy cover differed markedly between disturbed and forested sites, an observation that is readily apparent to even the most casual observer (Figure 20). The presence or absence of a tree canopy was reflected in the composition of the vegetative community beneath (Table 1), the vegetative structure as measured by the height index (Table 2, Figure 21), and the leaf litter depth (Table 2). The ground cover composition appeared to be partly related to canopy cover, especially grasses and leaf litter, but less so for the soil and herbaceous categories (Figure 22). When compared among sample sites (Table 2), significant differences among sites were found for all of the metrics. In most cases, however, post hoc multiple comparisons failed to clearly resolve these differences. Leaf litter was deeper in forested sites (except F8), and canopy cover was greater at forested sites (Table 2). The vegetation height indicies appear to differ among treatments (forested, cleared – see Figure 21, Table 2), but were not well resolved in post hoc multiple comparisons. In general, forested sites had a higher vegetation index for trees and shrubs, while cleared sites had a higher vegetation index for grasses (Table 2). As expected, the overall height index was generally greater at forested sites than at cleared sites (Table 2).

Temperature and Humidity

Data for 2 cm soil temperature were available only for 3 forested sites because one data logger was lost, apparently due to activity of heavy machinery in the study plot between the summer and fall sampling periods. Similarly, these data were available for only three forested sites because one data logger failed to download data (data logger malfunction). Daily cycles of daytime highs and nighttime lows are readily apparent in the data (Figure 23), with the data loggers in the cleared sites averaging consistently higher temperatures than those in the forested sites (Figures 23, 24). The daily minimum and maximum temperatures at cleared sites were higher and lower, respectively, than the same data for forested sites – that is, 2 cm soil temperature daily highs were higher in cleared sites and nightly lows were lower in cleared sites as well (Figure 25). Average relative humidity was measured at one forested site and one cleared site, but the data logger at the cleared site failed to function properly beginning about half way through the data collection period (Figure 26). Nonetheless, sufficient data were collected to allow comparisons to be made. In general, the humidity was higher at the forested site than at the cleared site (Figure 265 [left side of figure only]). When daily minimum and maximum humidities are plotted (Figure 27) it is apparent that, as for the temperature data discussed above, minimum and maximum humidities at the cleared site were more extreme than those at the forested site. Thus, cleared sites represent a more thermally extreme and more variable environment than the forested sites, and the more limited humidity data suggest that the same is true for humidity.

Insect Biodiversity

Large numbers of animals (3,776 specimens of at least 409 unique taxa, Table 3) were collected during our summer sampling period. Because sample identification took longer than anticipated, we do not include results for Malaise samples from the Summer sampling period. As we had previously suspected (see text of research proposal) there was insufficient time to even begin evaluating the collections made in the Fall, thus the analysis will largely focus on Summer data excluding Malaise samples. Photographs of representative species of most families are provided in Appendix 1.

Total numbers of individuals (see bottom of Table 3) varied by sample method (leaf litter, pitfall, or sweep)(X^2=1911.868, df=2, p<0.0001), among sample sites (X^2=1461.157, df=7, p<0.0001), and between treatments (cleared=786, forested=2990 specimens) (X^2=1286.445, df=1, p<0.0001).

Table 1. Visually dominant plants collected at each of the eight upland sites on 5-9 July, 2004. Identifications by Bill Handel, Illinois Natural History Survey.

Scientific name	Common name	D1	D2	D3	D4	F5	F6	F7	F8
Acer rubra	red maple	.	.	.	.	+	.	.	+
Acer saccharum	hard maple	.	.	.	.	.	+	.	.
Achillea millefolium	yarrow	.	.	.	+	.	.	.	.
Ambrosia artemisiifolia	ragweed	+	.	+	.	.	.	.	.
Amphicarpa bracteata	hog peanut	.	.	.	.	+	+	.	+
Andropogon virginicus	broom sedge	.	+	.	+	.	.	.	.
Apocynum cannabinum	dogbane	.	.	+	.	.	.	.	.
Arisaema triphyllum	Jack-in-the-pulpit	.	.	.	.	+	.	.	.
Athyrium angustum	lady fern	.	.	.	.	.	+	.	.
Boehmeria cylindrica	false nettle	.	.	.	.	+	.	.	.
Cercis canadensis	eastern redbud	.	.	.	.	.	.	+	.
Cornus racemosa	gray dogwood	.	.	.	.	.	.	+	.
Desmodium canadense	showy tick trefoil	.	.	.	+	.	.	.	.
Desmodium ciliare	hairy tick trefoil	+	.	.	.	.	.	.	.
Erigeron strigosus	daisy fleabane	.	+	+	.	.	.	.	.
Euthamia graminifolia	grassleaf goldenrod	+	+	.	.	.	.	.	.
Euthamia graminifolia	grassleaf goldenrod	+	.	.	.	.	.	.	.
Fagus grandifolia	American beech	.	.	.	.	+	+	.	+
Festuca pratensis	meadow fescue	+	.	.	.	.	.	.	.
Fragaria virginiana	wild strawberry	.	.	.	.	.	.	.	+
Fraxinus americana	white ash	.	.	.	.	+	.	+	.
Galium triflorum	sweet-scented bedstraw	.	.	.	.	.	+	+	.
Hackelia virginiana	stickseed	.	.	.	.	.	+	.	.
Helianthus strumosus	pale-leaved sunflower	+	.	.	.	.	.	.	.
Kummerowia striata	Japanese lespedeza	+	.	.	.	.	.	.	.
Lespedeza sp.	bush clover	.	.	+	.	.	.	.	.
Lindera benzoin	spice bush	.	.	.	.	.	+	.	.
Lysimachia lanceolata	lance-leaved loosestrife	.	.	.	.	.	.	.	+
Melilotus alba	white sweet clover	+	.	.	.	.	.	.	.
Muhlenbergia sp	muhly grass	.	.	.	.	.	+	.	.
Panicum clandestinum	broad-leaved panic grass	+	.	.	.	.	.	.	.
Panicum sp.	panic grass	.	.	+	.	.	.	.	.
Parthenocissus quinquefolia	Virginia creeper	.	.	.	.	+	+	+	.
Pilea pumila	Canada clearweed	.	.	.	.	.	+	.	.
Plantago lanceolata	buckhorn	+	.	.	+	.	.	.	.
Polystichum acrostichoides	Christmas fern	.	.	.	.	+	.	.	+
Populus deltoides	cottonwood	.	+	.	.	.	.	.	.
Potentilla simplex	common cinquefoil	.	.	+	.	.	.	.	.
Prunus serotina	wild black cherry	.	+	.	.	.	.	.	.
Pycnanthemum tenuifolium	slender mountain mint	.	.	.	+	.	.	.	.
Quercus velutina	black oak	.	.	.	.	.	.	.	+
Robinia pseudoacacia	black locust	+	.	+	.	.	.	.	.
Rhus copallina	dwarf sumac	.	+	+	.	.	.	.	.
Rubus occidentalis	black raspberry	.	+	.	.	.	.	.	.
Rudbeckia hirta	black-eyed susan	.	.	.	+	.	.	.	.
Sabatia angularis	marsh pink	.	+	.	+	.	.	.	.
Sassafras albidum	red sassafras	.	.	.	+	+	.	+	+
Smilax rotundifolia	cat briers	.	.	.	.	.	.	+	+
Solidago canadensis	Canada goldenrod	.	.	+	+	.	.	.	.
Solidago missouriensis	Missouri goldenrod	.	.	.	+	.	.	.	.
Symphoricarpos orbiculatus	buckbrush	.	.	.	.	.	.	+	.
Toxicodendron radicans	posion ivy	.	.	.	.	.	.	+	.
Viburnum acerifolium	maple-leaved arrowwood	.	.	.	.	.	.	.	+
Vitis aestivalis	summer grape	.	.	.	+	.	.	.	.
Unknown grass		.	.	.	+	+	.	.	.
Unkown herb		.	+	.	.	.	.	.	.

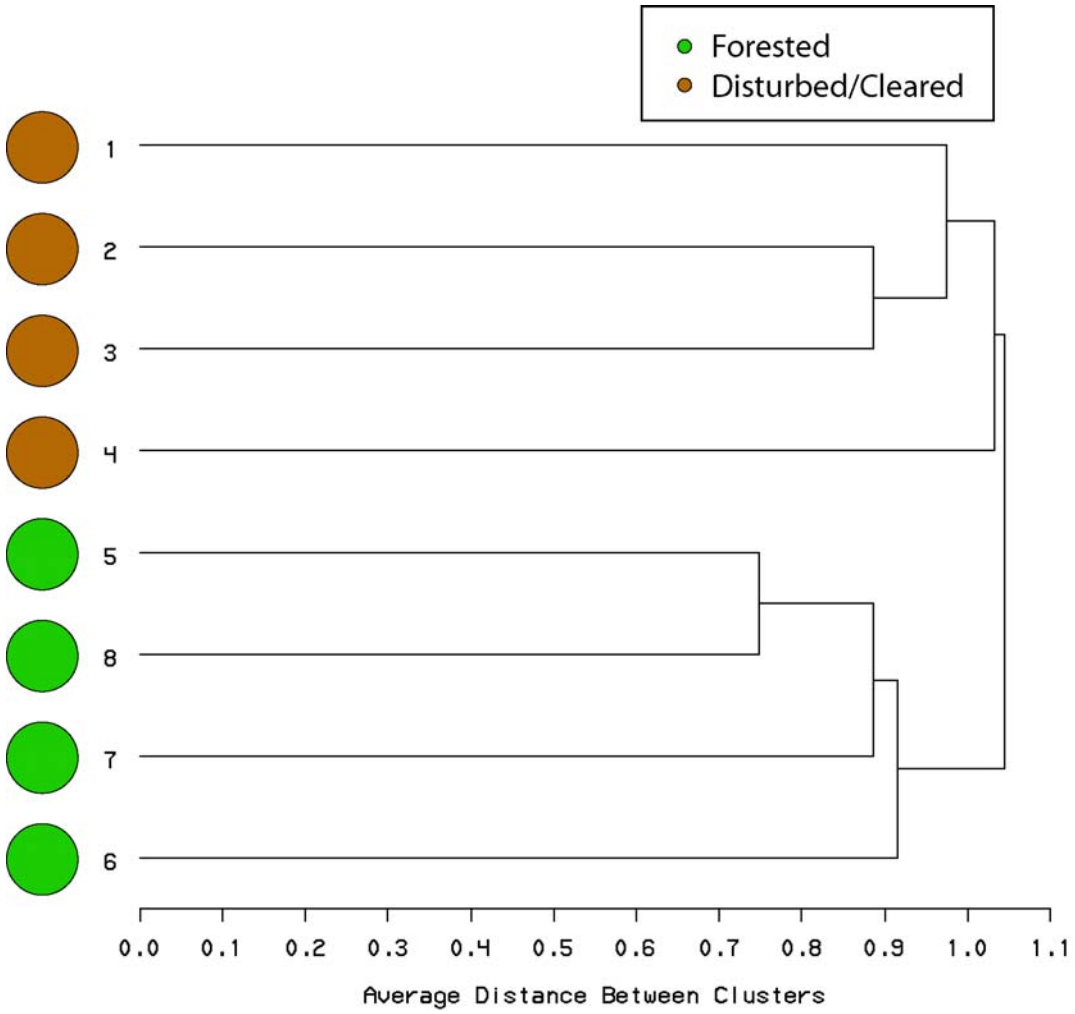


Figure 19. Cluster analysis (UPGMA average linkage analysis, RMS distance) of eight upland study sites based on presence/absence of 10 visually dominant plants collected at each of the sites on 5-9 July, 2004. Identifications by Bill Handel, Illinois Natural History Survey. D1-D4, cleared land; F5-F8, forested land.

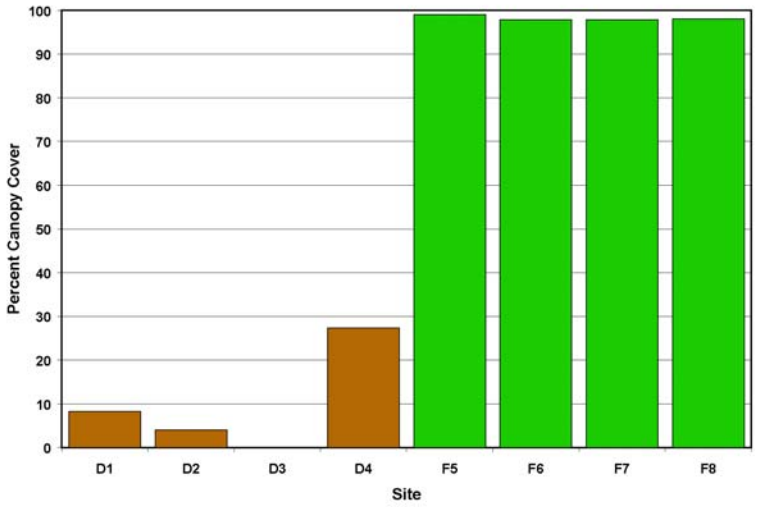


Figure 20. Percentage canopy cover at eight upland sites at Camp Atterbury, Indiana. Based on analysis of 9 replicate digital images at each of the 8 sites. Cleared sites indicated by brown, forested sites by green.

Table 2. One-way ANOVA comparisons between mean values of habitat metrics at eight upland sites at Camp Atterbury, Indiana. Values with the same letter are not significantly different in post-hoc multiple comparisons (Student-Newman-Keuls).

Metric									df	F	p
	A	A	A	A	B	B C	C	A			
Mean Leaf Litter Depth (cm)	0.43 D1	0.45 D2	0.43 D3	0.50 D4	1.65 F5	1.25 F6	1.85 F7	0.73 F8	7,312	16.11	<0.0001
	A	A	A	B	C	C	C	C			
Canopy Cover (%)	8.22 D1	4.00 D2	0.00 D3	27.33 D4	99.0 F5	97.89 F6	97.88 F7	98.0 F8	7,63	61.19	<0.0001
	A	A	A	A	A B	A B	A	B			
Tree Height Index	0.00 D1	0.00 D2	0.00 D3	0.00 D4	24.38 F5	31.03 F6	0.00 F7	47.78 F8	7,312	5.45	<0.0001
	A	A	A	A	A B	B	B	A B			
Shrub Height Index	0.00 D1	0.00 D2	0.00 D3	0.00 D4	15.65 F5	33.66 F6	26.95 F7	19.58 F8	7,312	4.92	<0.0001
	A C	D	A	A C	A C	B C	B	A C			
Herb Height Index	8.49 D1	29.02 D2	7.23 D3	12.23 D4	11.71 F5	15.73 F6	20.30 F7	8.98 F8	7,312	12.51	<0.0001
	A D	B D	B E	A	C	C E	C E	C			
Grass Height Index	14.14 D1	10.43 D2	7.96 D3	16.60 D4	0.00 F5	4.08 F6	3.18 F7	0.00 F8	7,312	15.96	<0.0001
	A	A C	A	A C	C	C	A	B			
Overall Vegetation Height Index	13.13 D1	28.98 D2	9.19 D3	16.89 D4	40.11 F5	43.18 F6	33.37 F7	64.36 F8	7,312	7.30	<0.0001

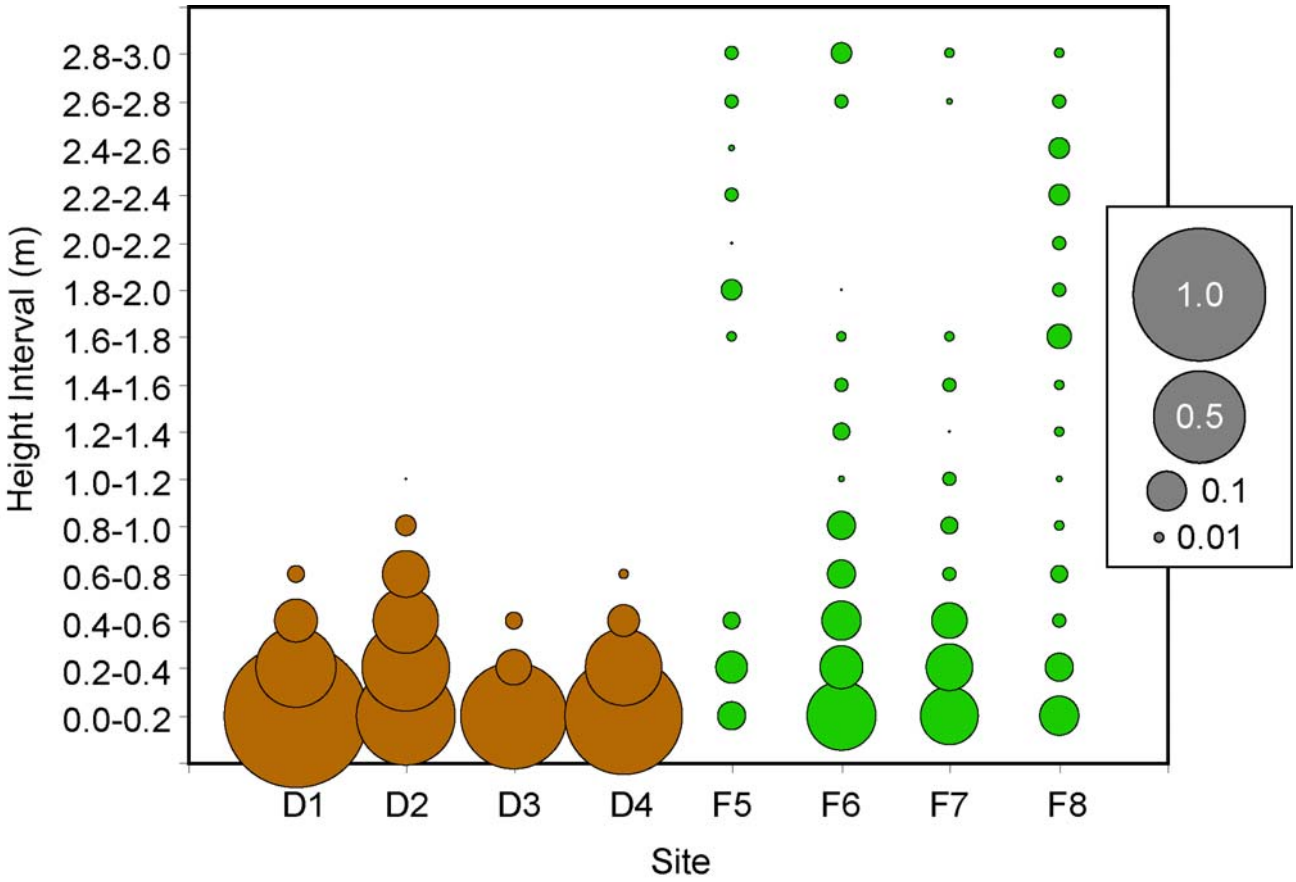


Figure 21. Bubble plot of average height index by height interval (up to 3 meters) for each of the eight upland study sites. Inset legend box shows bubble area in relation to selected height index values. Average height index values are based on average of four transects each with ten sample points for each study site.

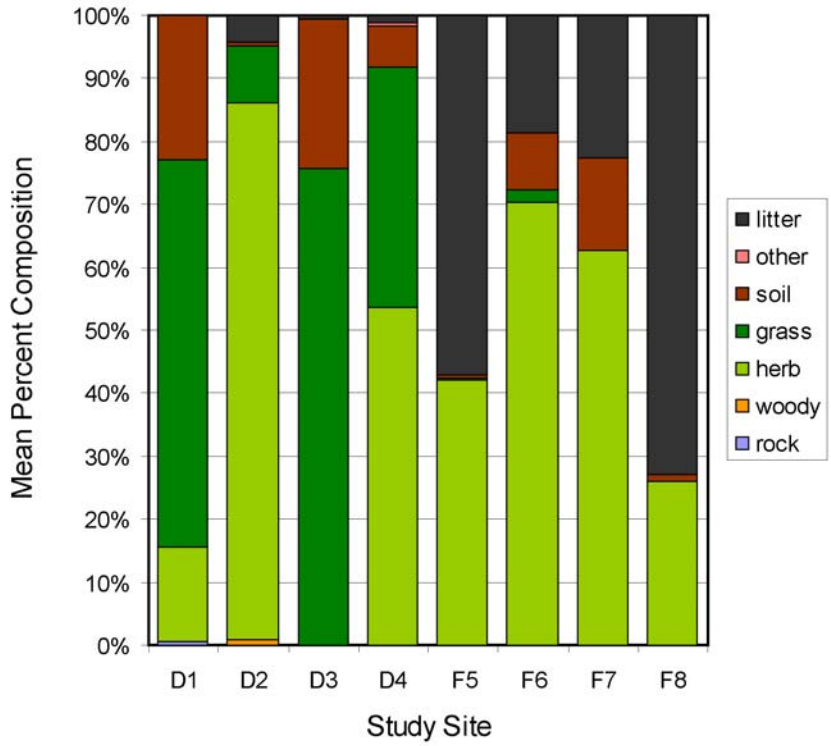


Figure 22. Ground cover type percentages at eight upland sites at Camp Atterbury, Indiana, based on image analysis. Sites 1-4 have been cleared, sites 5-8 are forested. Data are averages based on 9 sample points at each study site.

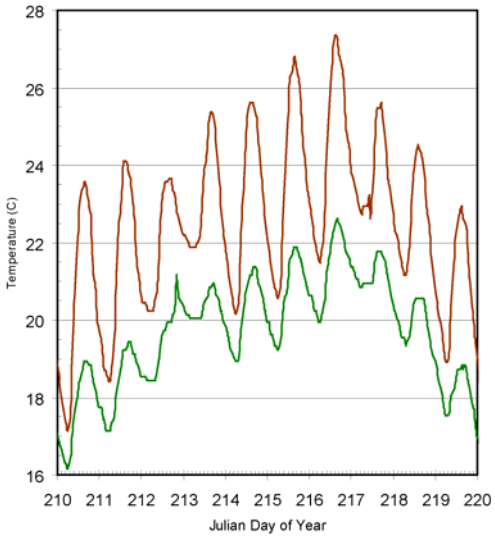


Figure 23. Sample 2 cm soil temperature data from Hobo Tidbit data loggers. Lines represent hourly averages of three data loggers in cleared (brown) and forested (green) plots over 10 days. Full data presented in Figure 24.

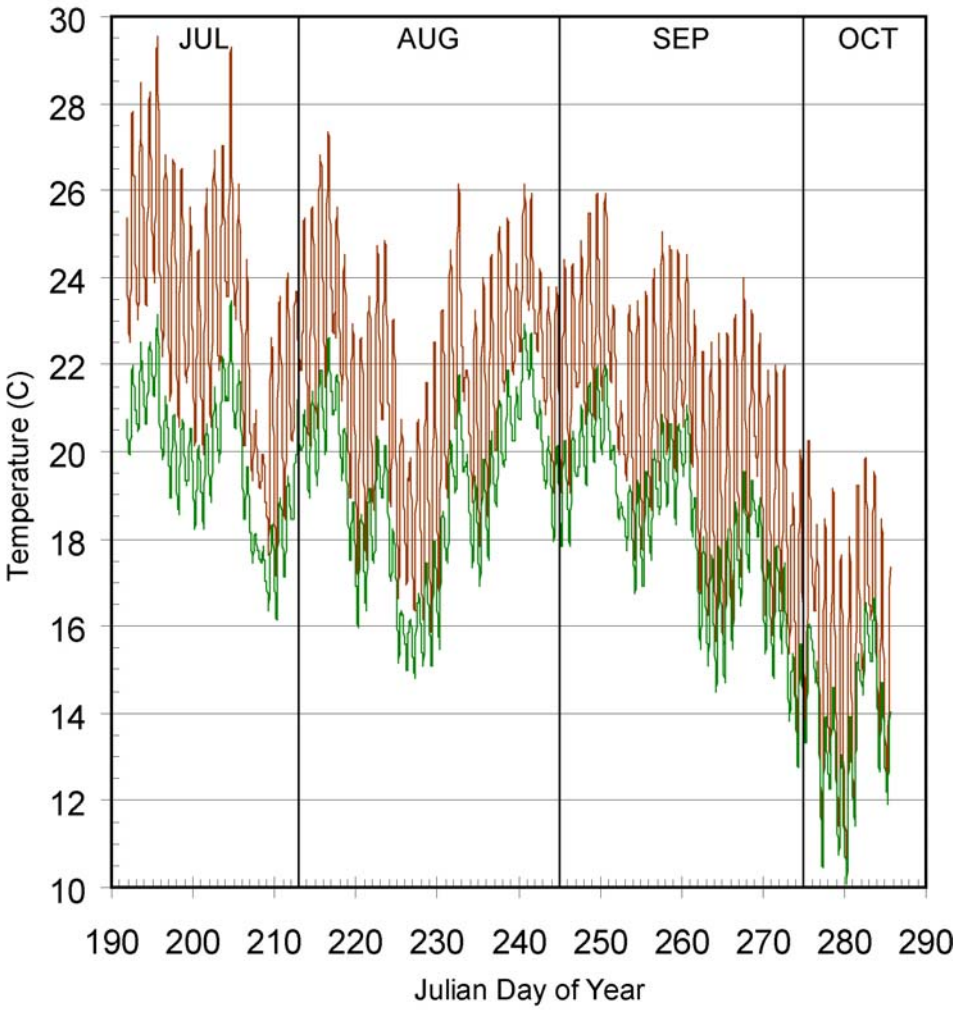


Figure 24. 2 cm soil temperature at cleared (brown line) and forested (green line) sample sites. Data are Hobo Tidbit ® hourly temperatures averaged across three sites in each treatment (cleared sites 2, 3, and 4; forested sites 5, 6, and 7). Data logger at site 1 was lost and data logger at site 8 was faulty (failed attempts to download data). Data presented span the approximately three month period from 9 July to 11 October, 2004. A detail of a portion of this data is shown in Figure 23.

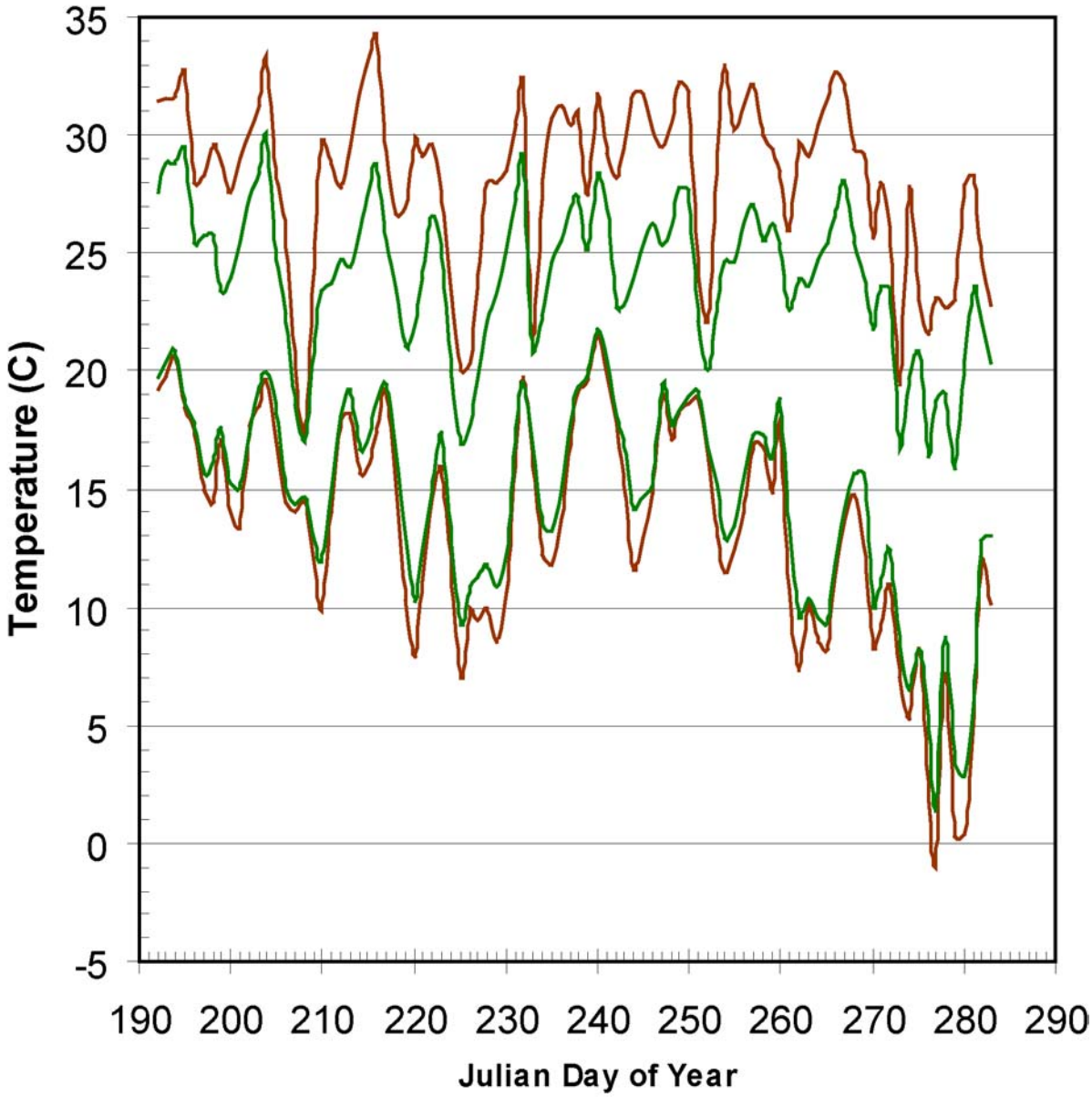


Figure 25. Minimum and maximum daily temperatures at cleared (brown line) and forested (green line) habitats in the upland study area. Data are from Hobo Temp/RH Pro ® hourly temperatures, from which daily minima and maxima were derived. Data presented span the approximately three month period from 10 July to 9 October, 2004.

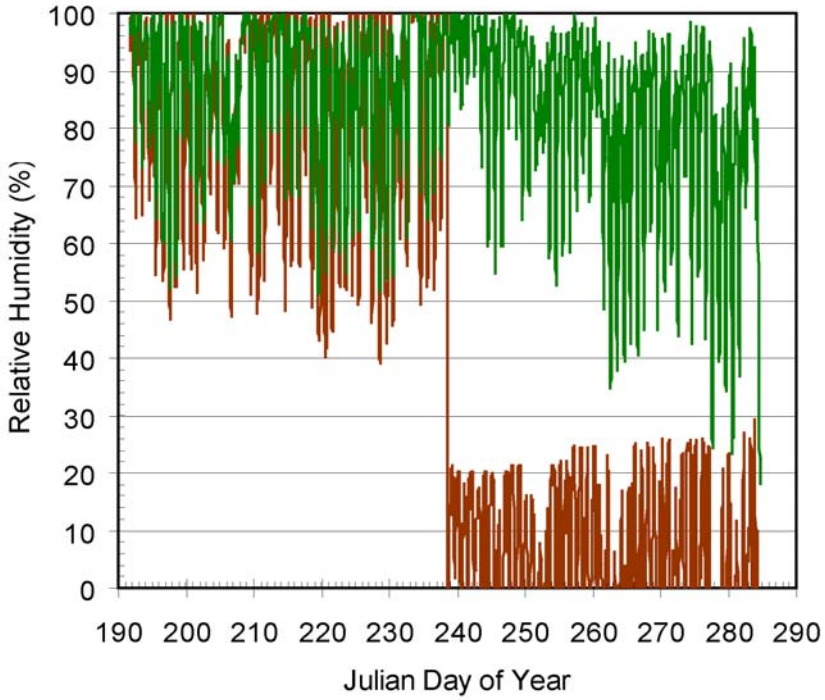


Figure 26. Relative humidity at cleared (brown line) and forested (green line) habitats in the upland study area. Hourly data are from Hobo Temp/RH Pro ® hourly temperatures. Data presented span the approximately three month period from 10 July to 9 October, 2004. The sudden drop in the cleared habitat humidity around day 239 is not real, but instead indicates the failure of the data logger.

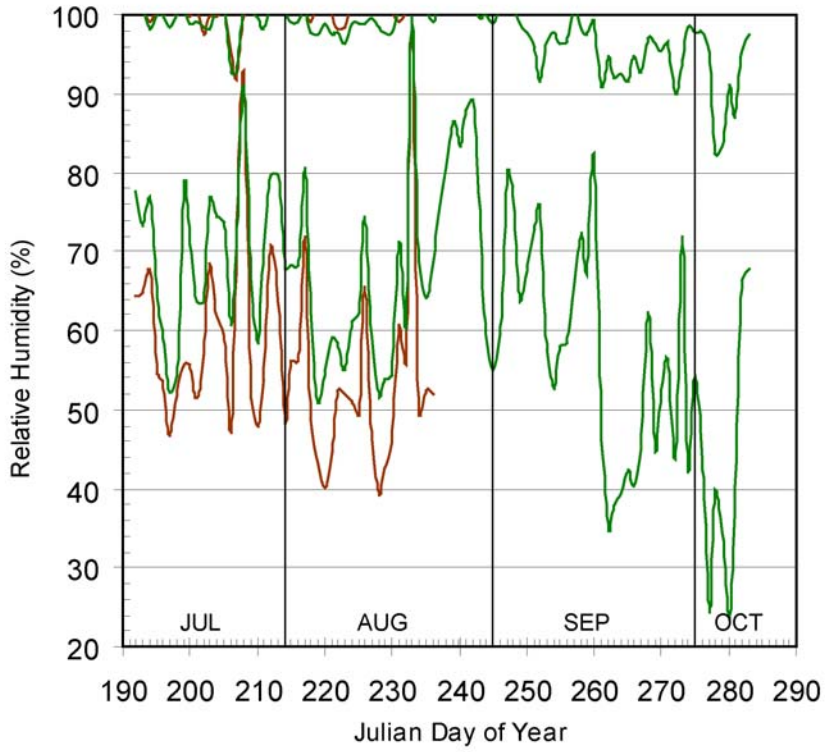


Figure 27. Minimum and maximum relative humidity (%) at cleared (brown line) and forested (green line) habitats in the upland study area. Data are from Hobo Temp/RH Pro ® hourly humidity, from which daily minima and maxima were derived. Data presented span the approximately three month period from 10 July to 9 October, 2004, but because the cleared area data logger provided only erroneous data after day 239, data after that date are omitted.

Table 3. List of 3,776 individuals of at least 409 taxa collected by three methods at eight sites in the MPTR at Camp Atterbury, Indiana July 5-9, 2004. Numbers under taxon heading are unique (within family) identifiers for morphospecies. Higher taxonomic levels indicated by bold text. Appendix 1 contains photographs of selected taxa.

Order Family taxon	Leaf	Pit	Sweep	D1	D2	D3	D4	F5	F6	F7	F8
Mollusca:Gastropoda	1	2	0	0	0	0	0	1	1	1	0
Annelida	0	1	0	0	0	0	0	0	1	0	0
Crustacea:Isopoda	0	1	0	0	0	0	1	0	0	0	0
Arthropoda:Arachnida											
Pseudoscorpiones	21	1	0	0	0	0	0	5	8	2	7
Opiliones	1	0	0	0	0	0	0	0	0	0	1
Acari											
Undetermined											
117	0	0	1	1	0	0	0	0	0	0	0
6	0	1	0	0	0	0	0	0	1	0	0
92	1	0	0	0	0	0	0	1	0	0	0
undetermined	1534	9	11	2	20	2	9	278	328	294	621
Araneae	205	29	21	11	13	5	12	47	29	120	18
Arthropoda:Chilopoda											
Lithobiomorpha	2	0	0	0	2	0	0	0	0	0	0
Undetermined	31	0	0	0	2	0	0	11	9	6	3
Arthropoda:Diplopoda	47	4	0	0	1	0	0	8	26	7	9
Arthropoda:Insecta											
Collembola											
Entomobryidae											
241	11	1	1	0	0	0	0	2	5	2	4
242	11	0	0	0	0	0	0	7	1	0	3
248	61	41	0	0	15	1	1	34	0	17	34
251	1	0	0	0	0	0	0	1	0	0	0
252	1	0	0	0	0	0	0	1	0	0	0
257	1	0	0	0	0	0	0	0	1	0	0
259	3	1	0	0	0	0	0	1	1	0	2
262	4	1	0	0	0	0	0	0	1	1	3
263	0	24	0	21	3	0	0	0	0	0	0
264	0	18	0	3	0	5	10	0	0	0	0
Hydrogastruridae											
267	0	4	0	0	2	1	1	0	0	0	0
255	9	1	0	0	0	0	0	0	6	0	4
256	17	0	0	0	0	0	0	0	4	1	12
267	0	5	0	0	5	0	0	0	0	0	0

continued on following page

Table 3. Continued.

Order Family taxon	Leaf	Pit	Sweep	D1	D2	D3	D4	F5	F6	F7	F8
Isotomidae											
265	0	3	0	2	0	0	1	0	0	0	0
Makenziellidae											
247	2	0	0	0	0	0	0	2	0	0	0
260	1	0	0	0	0	0	0	0	1	0	0
Smithinthuridae											
243	0	0	2	2	0	0	0	0	0	0	0
250	3	0	0	0	0	0	0	2	0	0	1
266	0	5	0	0	1	0	1	2	0	0	1
Tomoceridae											
249	48	76	0	5	12	0	2	46	15	8	36
254	22	1	0	0	0	0	0	8	4	2	9
258	1	0	0	0	0	0	0	0	1	0	0
268	0	1	0	0	0	0	0	0	0	0	1
Undetermined											
243	0	0	2	2	0	0	0	0	0	0	0
undetermined	0	10	1	1	0	0	1	9	0	0	0
Blattaria											
Blattidae											
205	1	0	0	0	0	0	0	1	0	0	0
Orthoptera											
Acrididae											
227	0	0	1	0	0	0	1	0	0	0	0
236	0	0	2	0	0	0	2	0	0	0	0
237	0	0	1	0	0	0	1	0	0	0	0
239	0	0	1	0	0	0	1	0	0	0	0
Gryllidae											
217	0	1	0	0	1	0	0	0	0	0	0
218	0	2	0	0	2	0	0	0	0	0	0
219	0	12	0	2	1	1	8	0	0	0	0
220	0	1	0	0	0	1	0	0	0	0	0
234	0	0	3	2	0	1	0	0	0	0	0
Rhaphidophoridae											
216	1	2	0	0	0	0	0	1	0	2	0
Tettigoniidae											
238	0	0	1	0	0	0	1	0	0	0	0
Psocoptera											
Undetermined											
230	0	0	1	0	0	0	0	1	0	0	0
231	0	0	1	0	0	0	0	1	0	0	0
261	1	0	0	0	0	0	0	0	1	0	0
Thysanoptera											
Aelothripidae											
228	0	0	1	0	0	0	1	0	0	0	0
Phlaeothripidae											
206	14	0	0	0	5	1	0	6	1	1	0

continued on following page

Table 3. Continued.

Order / Family / taxon	Leaf	Pit	Sweep	D1	D2	D3	D4	F5	F6	F7	F8
207	5	0	0	0	1	2	0	2	0	0	0
208	1	0	0	0	1	0	0	0	0	0	0
209	2	0	0	0	0	2	0	0	0	0	0
210	6	0	0	0	0	0	0	5	0	0	1
211	6	0	0	0	0	0	0	4	0	0	2
212	1	0	0	0	0	0	0	1	0	0	0
213	1	0	0	0	0	0	0	1	0	0	0
214	1	0	0	0	0	0	0	0	0	1	0
215	2	0	0	0	0	0	0	0	0	0	2
235	0	0	1	0	1	0	0	0	0	0	0
Thripidae											
226	0	0	1	0	0	1	0	0	0	0	0
229	0	0	11	8	0	1	0	1	0	1	0
232	0	0	2	0	0	0	0	0	2	0	0
233	0	0	1	1	0	0	0	0	0	0	0
Hemiptera											
Anthocoridae											
123	0	0	1	0	0	1	0	0	0	0	0
Coreidae											
125	0	0	2	0	0	0	2	0	0	0	0
Dipsocoridae											
134	3	0	0	0	0	0	0	2	0	0	1
Lygaeidae											
108	2	0	0	0	0	0	0	0	2	0	0
109	3	0	0	0	0	0	0	1	2	0	0
11	0	3	0	2	1	0	0	0	0	0	0
111	0	2	0	1	0	0	1	0	0	0	0
112	0	3	0	1	2	0	0	0	0	0	0
120	0	0	2	1	1	0	0	0	0	0	0
122	0	0	1	0	0	1	0	0	0	0	0
Miridae											
116	0	0	10	3	1	3	3	0	0	0	0
Pentatomidae											
119	0	0	1	1	0	0	0	0	0	0	0
121	0	0	1	0	1	0	0	0	0	0	0
Reduviidae											
107	1	0	0	0	1	0	0	0	0	0	0
110	1	0	0	0	0	0	0	0	1	0	0
124	0	0	1	0	0	0	1	0	0	0	0
13	0	0	1	0	0	1	0	0	0	0	0
Thyreocoridae											
115	0	0	2	1	1	0	0	0	0	0	0
Undetermined											
11	0	1	0	1	0	0	0	0	0	0	0
118	0	0	3	3	0	0	0	0	0	0	0
126	0	0	1	0	0	0	0	0	1	0	0

continued on following page

Table 3. Continued.

Order Family taxon	Leaf	Pit	Sweep	D1	D2	D3	D4	F5	F6	F7	F8
Homoptera											
Aphididae											
18	0	0	1	0	0	0	1	0	0	0	0
203	2	0	0	0	0	0	0	0	0	2	0
204	3	0	0	0	0	0	0	0	0	3	0
undetermined	0	1	0	1	0	0	0	0	0	0	0
Aphrophoridae											
Aphrophora spumarius	0	1	6	6	0	0	1	0	0	0	0
Cicadellidae											
Aceratagallia accola	0	0	3	2	1	0	0	0	0	0	0
Agallia constricta	0	2	11	12	1	0	0	0	0	0	0
Balclutha abdominalis	0	0	1	0	0	1	0	0	0	0	0
Chlorotettix spatulatus	0	0	2	1	0	0	0	0	0	0	1
Cuerna costalis	0	1	1	0	0	0	2	0	0	0	0
Draeculacephala antica	1	0	2	1	0	0	2	0	0	0	0
Empoasca sp. 1	0	0	1	0	0	0	1	0	0	0	0
Laevicephalus unicoloratus	0	0	1	1	0	0	0	0	0	0	0
Osbornellus consors	1	2	0	1	0	0	1	0	0	1	0
Scaphytopius acutus	1	0	0	0	0	0	0	0	0	1	0
Stirellus bicolor	0	1	0	0	0	0	0	1	0	0	0
undetermined	0	1	0	1	0	0	0	0	0	0	0
Delphacidae											
Delphacodes sp. 1	0	0	1	0	0	0	1	0	0	0	0
Derbidae											
Cedusa sp.	0	0	1	1	0	0	0	0	0	0	0
Issidae											
Bruchomorpha occulata	2	0	0	0	0	0	0	0	0	2	0
Membracidae											
Campylenchia latipes	0	0	1	0	0	0	0	0	0	0	1
Entylia carinata	0	0	1	0	0	0	1	0	0	0	0
Psyllidae											
undetermined	0	0	4	1	0	2	1	0	0	0	0
Coleoptera											
Anthribidae											
10	0	0	1	0	1	0	0	0	0	0	0
11	0	0	1	0	1	0	0	0	0	0	0
5	0	1	0	0	0	0	1	0	0	0	0
55	1	0	0	0	0	0	0	0	1	0	0
Cantharidae											
30	0	0	1	0	0	0	0	0	1	0	0
Carabidae											
4	0	1	0	1	0	0	0	0	0	0	0
6	0	4	0	0	0	0	0	2	1	1	0
74	1	0	0	0	0	0	1	0	0	0	0
81	1	0	0	0	0	0	0	1	0	0	0
9	3	0	0	0	0	0	0	3	0	0	0
Chrysomelidae											
129	2	0	0	0	0	0	0	0	0	2	0
14	0	0	1	0	0	1	0	0	0	0	0

continued on following page

Table 3. Continued.

Order Family taxon	Leaf	Pit	Sweep	D1	D2	D3	D4	F5	F6	F7	F8
3	0	0	2	1	0	1	0	0	0	0	0
5	0	2	0	0	0	1	1	0	0	0	0
7	0	0	2	1	1	0	0	0	0	0	0
70	1	0	0	0	1	0	0	0	0	0	0
72	1	0	0	0	1	0	0	0	0	0	0
9	0	0	1	0	1	0	0	0	0	0	0
Cicindelidae											
3	0	2	0	2	0	0	0	0	0	0	0
36	0	2	0	1	0	0	1	0	0	0	0
36	0	0	0	0	0	0	0	0	0	0	0
Coccinellidae											
17	0	0	1	0	0	0	1	0	0	0	0
Corylophidae											
5	0	0	1	1	0	0	0	0	0	0	0
Cryptophagidae											
100	2	0	0	0	0	0	0	1	1	0	0
Cucujidae											
73	1	0	0	0	1	0	0	0	0	0	0
Curculionidae											
105	1	0	0	0	0	0	0	1	0	0	0
130	3	0	0	0	0	0	0	0	0	2	1
132	1	0	0	0	0	0	0	0	0	0	1
2	0	0	1	1	0	0	0	0	0	0	0
4	0	1	0	0	1	0	0	0	0	0	0
69	1	0	0	0	1	0	0	0	0	0	0
8	0	0	2	0	2	0	0	0	0	0	0
Dascillidae											
155	3	0	0	0	0	0	0	0	0	3	0
Elateridae											
3	0	3	0	3	0	0	0	0	0	0	0
39	0	2	0	0	0	2	0	0	0	0	0
4	0	1	0	1	0	0	0	0	0	0	0
5	0	1	0	0	0	1	0	0	0	0	0
Histeridae											
47	0	1	0	0	1	0	0	0	0	0	0
5	0	4	0	0	0	0	4	0	0	0	0
Lampyridae											
89	1	0	0	0	0	0	0	1	0	0	0
Leiodidae											
104	5	0	0	0	0	0	0	2	0	0	3
156	1	0	0	0	0	0	0	0	0	1	0
49	0	6	0	0	4	0	0	2	0	0	0
87	1	0	0	0	0	0	0	1	0	0	0
Mordellidae											
1	0	0	14	12	0	1	0	0	1	0	0
21	0	0	2	0	0	0	0	1	0	0	1
29	0	0	1	0	0	0	0	0	1	0	0
32	0	0	1	0	0	0	0	0	1	0	0

continued on following page

Table 3. Continued.

Order Family taxon	Leaf	Pit	Sweep	D1	D2	D3	D4	F5	F6	F7	F8
4	0	1	0	1	0	0	0	0	0	0	0
Mycetophagidae											
27	0	0	1	0	0	0	0	0	1	0	0
Nitidulidae											
71	1	0	0	0	1	0	0	0	0	0	0
Nitidulidae											
85	3	0	0	0	0	0	0	2	0	1	0
86	2	0	0	0	0	0	0	2	0	0	0
Pselaphidae											
128	1	0	0	0	0	0	0	0	1	0	0
139	1	0	0	0	0	0	0	0	0	0	1
Ptillidae											
101	1	0	0	0	0	0	0	0	1	0	0
22	0	0	1	0	0	0	0	1	0	0	0
4	0	0	1	1	0	0	0	0	0	0	0
Ptilodactylidae											
98	3	0	0	0	0	0	0	0	2	1	0
Rhipiphoridae											
1	0	0	1	0	0	1	0	0	0	0	0
15	0	0	1	0	0	1	0	0	0	0	0
Scarabaeidae											
4	0	1	0	0	1	0	0	0	0	0	0
5	0	1	0	0	0	0	1	0	0	0	0
6	0	4	0	0	0	0	0	1	3	0	0
60	0	1	0	0	0	0	0	0	1	0	0
96	1	0	0	0	0	0	0	0	1	0	0
Scydmaenidae											
127	4	0	0	0	0	0	0	0	1	3	0
Staphylinidae											
102	4	0	0	0	0	0	0	0	4	0	0
103	1	0	0	0	0	0	0	0	1	0	0
131	1	0	0	0	0	0	0	0	0	0	1
157	1	0	0	0	0	0	0	0	0	1	0
26	1	0	1	0	0	0	0	1	1	0	0
3	0	1	1	1	0	0	0	0	0	1	0
35	1	1	0	0	0	0	0	0	0	1	1
38	0	1	0	0	0	0	0	0	0	1	0
4	0	6	0	1	5	0	0	0	0	0	0
40	1	0	0	0	0	0	0	0	1	0	0
44	0	112	0	0	16	0	16	1	4	3	72
45	0	1	0	0	0	0	0	0	0	1	0
46	0	7	0	0	2	0	3	1	0	1	0
5	0	1	0	0	1	0	0	0	0	0	0
62	1	4	0	0	0	0	0	2	0	1	2
88	1	0	0	0	0	0	0	1	0	0	0
9	2	0	0	0	0	0	0	2	0	0	0
93	8	0	0	0	0	0	0	1	6	0	1
95	1	0	0	0	0	0	0	0	1	0	0
99	4	0	0	0	0	0	0	0	2	0	2

continued on following page

Table 3. Continued.

Order Family taxon	Leaf	Pit	Sweep	D1	D2	D3	D4	F5	F6	F7	F8
Tenebrionidae											
97	6	0	0	0	0	0	0	2	4	0	0
Undetermined											
106	1	0	0	0	0	0	0	0	0	0	1
5	0	3	0	0	0	0	3	0	0	0	0
9	1	0	0	0	0	0	0	1	0	0	0
94	1	0	0	0	0	0	0	0	1	0	0
Diptera											
Suborder Nematocera											
Undetermined											
555	0	0	3	1	0	1	1	0	0	0	0
572	0	0	2	0	0	0	0	2	0	0	0
Suborder Brachycera											
Undetermined											
563	0	0	1	0	0	1	0	0	0	0	0
587	0	0	1	0	0	0	0	1	0	0	0
619	0	0	1	0	0	0	0	0	0	1	0
620	0	0	1	0	0	0	0	0	0	0	1
Agromyzidae											
609	0	0	1	0	0	0	0	0	0	1	0
617	0	0	1	0	0	0	0	0	0	1	0
Bibionidae											
517	1	0	0	0	0	0	0	0	1	0	0
Cecidomyiidae											
516	1	0	0	0	0	0	0	0	1	0	0
521	1	0	0	0	0	0	0	0	1	0	0
522	1	0	0	0	0	0	0	0	1	0	0
523	1	0	0	0	0	0	0	0	1	0	0
534	1	0	0	0	0	0	0	0	1	0	0
535	2	0	0	0	0	0	0	0	2	0	0
588	0	0	1	0	0	0	0	1	0	0	0
Ceratopogonidae											
556	0	0	2	2	0	0	0	0	0	0	0
559	0	0	1	1	0	0	0	0	0	0	0
584	0	0	1	0	0	0	0	1	0	0	0
Chironomidae											
515	1	0	2	1	0	0	0	2	0	0	0
518	1	0	0	0	0	0	0	0	0	0	1
530	1	0	0	0	0	0	0	1	0	0	0
531	1	0	0	0	0	0	0	1	0	0	0
564	0	0	2	0	0	2	0	0	0	0	0
Chloropidae											
505	0	2	0	0	2	0	0	0	0	0	0
557	0	0	1	1	0	0	0	0	0	0	0
558	0	0	7	5	1	1	0	0	0	0	0
561	0	0	1	0	1	0	0	0	0	0	0
Culicidae											
592	0	0	1	0	0	0	0	1	0	0	0

continued on following page

Table 3. Continued.

Order Family taxon	Leaf	Pit	Sweep	D1	D2	D3	D4	F5	F6	F7	F8
604	0	0	1	0	0	0	0	0	1	0	0
623	0	0	1	0	0	0	0	0	0	0	1
624	0	0	1	0	0	0	0	0	0	0	1
625	0	0	1	0	0	0	0	0	1	0	0
Diastatidae											
503	0	1	0	0	1	0	0	0	0	0	0
Dolichopodidae											
565	0	0	2	0	0	1	1	0	0	0	0
567	0	0	1	0	0	0	1	0	0	0	0
569	0	0	1	0	0	0	1	0	0	0	0
582	0	0	10	0	0	0	0	3	4	0	3
586	0	0	2	0	0	0	0	2	0	0	0
607	0	0	1	0	0	0	0	0	1	0	0
Heleomyzidae											
601	0	0	1	0	0	0	0	0	1	0	0
Muscidae											
511	0	1	0	0	1	0	0	0	0	0	0
566	0	0	2	0	0	0	2	0	0	0	0
608	0	0	1	0	0	0	0	0	1	0	0
Mycetophilidae											
589	0	0	1	0	0	0	0	1	0	0	0
602	0	0	3	0	0	0	0	1	2	0	0
622	0	0	1	0	0	0	0	0	1	0	0
Phoridae											
507	0	1	0	0	0	0	0	0	1	0	0
508	0	1	0	0	0	0	0	0	0	0	1
509	0	2	0	0	0	0	1	0	1	0	0
514	1	0	1	0	0	0	0	0	0	1	1
532	1	0	0	0	0	0	0	1	0	0	0
533	1	0	2	0	0	0	0	2	1	0	0
536	2	0	3	0	0	0	0	2	2	0	1
562	0	0	3	0	0	1	0	0	2	0	0
570	0	0	1	0	0	0	1	0	0	0	0
591	0	0	1	0	0	0	0	1	0	0	0
602	0	0	1	0	0	0	0	0	1	0	0
603	0	0	8	0	0	0	0	3	5	0	0
605	0	0	1	0	0	0	0	0	1	0	0
606	0	0	2	0	0	0	0	0	2	0	0
614	0	0	1	0	0	0	0	0	0	1	0
621	0	0	1	0	0	0	0	0	1	0	0
627	12	0	0	0	0	0	0	0	12	0	0
Piophilidae											
568	0	0	1	0	0	0	1	0	0	0	0
Psychodidae											
590	0	0	1	0	0	0	0	1	0	0	0
Rhinophoridae?											
618	0	0	1	0	0	0	0	0	0	0	1
Sarcophagidae											
502	0	1	0	0	1	0	0	0	0	0	0

continued on following page

Table 3. Continued.

Order / Family / taxon	Leaf	Pit	Sweep	D1	D2	D3	D4	F5	F6	F7	F8
Scathophagidae											
571	0	0	1	0	0	0	1	0	0	0	0
Sciaridae											
522	2	0	0	0	2	0	0	0	0	0	0
504	0	1	0	0	0	0	0	0	1	0	0
519	2	0	0	0	0	0	0	0	2	0	0
Sphaeroceridae											
510	0	1	0	0	1	0	0	0	0	0	0
506	0	1	0	0	1	0	0	0	0	0	0
510	0	5	0	0	0	2	3	0	0	0	0
512	0	1	0	1	0	0	0	0	0	0	0
513	0	1	0	0	1	0	0	0	0	0	0
Tabanidae											
501	0	1	0	1	0	0	0	0	0	0	0
Tephritidae											
560	0	0	2	0	2	0	0	0	0	0	0
Undetermined											
520	2	0	0	0	0	0	0	0	2	0	0
undetermined	1	1	0	0	0	1	0	1	0	0	0
Hymenoptera											
Braconidae											
150	3	0	0	0	0	0	0	1	0	1	1
178	0	0	1	1	0	0	0	0	0	0	0
19	0	0	1	0	0	0	0	0	1	0	0
191	0	0	1	0	0	0	0	1	0	0	0
201	0	0	1	0	0	0	0	0	0	0	1
Ceraphronidae											
14	0	1	0	0	1	0	0	0	0	0	0
149	1	0	0	0	0	1	0	0	0	0	0
152	3	0	0	0	0	0	0	3	0	0	0
154	1	0	0	0	0	0	0	1	0	0	0
163	2	0	0	0	0	0	0	0	0	2	0
173	1	0	0	0	0	0	0	0	0	0	1
24	0	0	1	0	0	0	0	0	1	0	0
28	0	0	1	0	0	0	0	0	1	0	0
Colletidae											
Hylaeinae											
184	0	0	1	0	1	0	0	0	0	0	0
Cynipidae											
172	1	0	0	0	0	0	0	0	0	1	0
Diapriidae											
15	1	0	0	0	0	0	0	1	0	0	0
151	2	0	0	0	0	0	0	0	0	2	0
159	1	0	0	0	0	0	0	0	1	0	0
164	2	0	0	0	0	0	0	0	0	1	1
165	2	0	0	0	0	0	0	0	0	1	1
166	1	0	0	0	0	0	0	0	0	1	0
169	1	0	0	0	0	0	0	0	0	1	0
170	3	0	0	0	0	0	0	0	0	2	1

continued on following page

Table 3. Continued.

Order Family taxon	Leaf	Pit	Sweep	D1	D2	D3	D4	F5	F6	F7	F8
171	2	0	0	0	0	0	0	0	0	1	1
174	1	0	0	0	0	0	0	0	0	0	1
19	0	0	1	0	0	0	0	1	0	0	0
193	0	0	1	0	0	0	0	0	1	0	0
196	0	0	1	0	0	0	0	0	1	0	0
20	0	0	1	0	0	0	0	1	0	0	0
23	0	0	1	0	0	0	0	1	0	0	0
25	1	0	1	0	0	0	0	0	2	0	0
31	0	0	1	0	0	0	0	0	1	0	0
33	0	0	1	0	0	0	0	0	1	0	0
34	0	0	1	0	0	0	0	0	1	0	0
Encyrtidae											
182	0	0	1	1	0	0	0	0	0	0	0
Eucoilidae											
183	0	0	1	0	1	0	0	0	0	0	0
Eulophidae											
153	1	0	0	0	0	0	0	1	0	0	0
176	0	0	2	1	0	0	1	0	0	0	0
Eurytomidae											
177	0	0	1	1	0	0	0	0	0	0	0
Formicidae											
Aphaenogaster cf *rudis*	62	2	1	0	1	0	3	48	6	6	1
Aphaenogaster rudis ?	8	39	1	1	11	0	0	14	4	8	10
Aphaenogaster sp.	5	0	1	0	0	0	0	3	2	0	1
Aphaenogaster treatae	1	6	0	0	0	0	6	0	0	1	0
Brachymyrmex depelis	17	0	3	1	0	0	0	0	19	0	0
Camponotus pennsylvanicus ?	0	0	1	0	0	1	0	0	0	0	0
Camponotus sp. A	0	1	1	0	0	0	0	0	1	1	0
Camponotus subbarbatus	2	0	19	0	0	0	0	9	0	8	4
Crematogaster lineolata	1	13	14	1	27	0	0	0	0	0	0
Crematogaster lineolata ?	0	2	10	3	7	0	2	0	0	0	0
Crematogaster punctulata	0	6	0	2	0	0	4	0	0	0	0
Dolichoderus plagiatus	0	0	2	0	0	0	2	0	0	0	0
Formica fusca ?	0	0	1	0	0	0	1	0	0	0	0
Formica pallidefulva	0	1	9	5	4	0	1	0	0	0	0
Formica rubicunda	0	1	0	0	0	0	1	0	0	0	0
Formica rufa group	0	3	0	1	0	0	2	0	0	0	0
Formica schaufussi ?	0	1	0	0	0	0	1	0	0	0	0
Lasius alienus	28	22	15	1	31	0	7	6	4	16	0
Lasius alienus ?	0	0	1	0	0	0	0	0	0	1	0
Lasius neoniger	1	14	0	0	1	7	7	0	0	0	0
Lasius umbratus	0	1	0	0	0	0	1	0	0	0	0
Leptothorax sp.	2	0	0	0	0	0	0	1	1	0	0
Leptothorax ambiguus	2	0	0	0	0	0	0	0	1	0	1
Leptothorax curvispinosus	61	0	137	0	5	0	0	122	46	17	8
Monomorium minimum	1	13	46	11	6	15	21	0	0	0	7
Myrmecina americana	4	0	1	0	0	0	1	2	0	2	0
Myrmica sp. A	2	11	0	0	0	0	12	0	0	1	0
Myrmica sp. B	0	7	0	1	0	1	5	0	0	0	0

continued on following page

Table 3. Continued.

Order Family taxon	Leaf	Pit	Sweep	D1	D2	D3	D4	F5	F6	F7	F8
Paratrechina parvula	1	0	0	0	0	0	0	0	0	1	0
Pheidole bicarinata	0	4	0	0	0	0	4	0	0	0	0
Pheidole pillifera	0	0	1	0	0	0	1	0	0	0	0
Ponera pennsylvanica	22	1	0	0	1	0	1	5	16	0	0
Prenolepis imparis	1	0	0	0	0	0	0	0	1	0	0
Pyramica sp. A	8	0	1	0	0	0	1	0	0	8	0
Solenopsis molesta	8	26	1	2	26	0	7	0	0	0	0
Stenamma impar	8	5	0	0	0	0	4	5	3	1	0
Stenamma schmittii	1	0	0	0	0	0	0	0	0	0	1
Tapinoma sessile	1	6	3	2	3	0	2	2	0	0	1
Halictidae											
12	0	0	1	0	0	1	0	0	0	0	0
6	0	0	3	2	0	1	0	0	0	0	0
Ichneumonidae											
158	1	0	0	0	0	0	0	0	1	0	0
194	0	0	1	0	0	0	0	0	1	0	0
199	0	0	1	0	0	0	0	0	0	0	1
200	0	0	1	0	0	0	0	0	0	0	1
Megaspilidae											
161	1	0	0	0	0	0	0	0	1	0	0
162	1	0	0	0	0	0	0	0	0	1	0
Mymaridae											
181	0	0	1	1	0	0	0	0	0	0	0
Platygastridae											
160	1	0	0	0	0	0	0	0	0	0	1
185	0	0	1	0	0	0	1	0	0	0	0
187	0	0	2	0	0	0	2	0	0	0	0
188	0	0	2	0	0	0	1	0	1	0	0
189	0	0	1	0	0	0	1	0	0	0	0
Platygastridae Sceliotrachelinae											
160	1	0	0	0	0	0	0	0	1	0	0
Proctotrupidae											
198	0	0	1	0	0	0	0	0	0	1	0
Pteromalidae											
13	0	1	0	1	0	0	0	0	0	0	0
175	0	0	1	1	0	0	0	0	0	0	0
Rhopalosomatidae											
14	0	1	0	0	0	0	0	0	0	0	1
Scelionidae											
13	0	4	0	4	0	0	0	0	0	0	0
135	3	2	0	1	0	0	1	0	0	3	0
137	1	3	0	1	0	1	0	0	0	0	2
14	0	4	0	1	1	2	0	0	0	0	0
144	0	0	1	0	0	0	0	1	0	0	0
148	1	0	0	0	1	0	0	0	0	0	0
152	5	0	0	0	0	0	0	2	0	3	0
167	1	0	0	0	0	0	0	0	0	1	0
168	1	0	0	0	0	0	0	0	0	1	0
180	0	0	1	1	0	0	0	0	0	0	0

continued on following page

Table 3. Continued.

Order Family taxon	Leaf	Pit	Sweep	D1	D2	D3	D4	F5	F6	F7	F8
186	0	0	1	0	0	0	1	0	0	0	0
19	0	0	1	0	0	0	0	0	0	1	0
190	0	0	1	0	0	0	1	0	0	0	0
Trichogrammatidae											
179	0	0	1	1	0	0	0	0	0	0	0
195	0	0	1	0	0	0	0	0	1	0	0
Undetermined	0	1	0	0	1	0	0	0	0	0	0
Lepidoptera											
Gelechiidae											
244	0	0	1	0	0	0	0	1	0	0	0
245	0	0	1	0	0	0	0	0	1	0	0
Noctuidae											
221	2	0	0	0	1	0	0	0	0	0	1
222	2	0	0	0	0	0	0	1	1	0	0
240	0	0	1	0	0	0	1	0	0	0	0
Pterophoridae											
223	1	0	0	0	0	0	0	1	0	0	0
225	1	0	0	0	0	0	0	0	0	0	1
Pyralidae											
undetermined	1	0	0	0	0	0	0	0	1	0	0
Undetermined											
246	0	0	1	1	0	0	0	0	0	0	0
undetermined	3	0	0	0	0	0	0	3	0	0	0
Totals	2524	675	577	196	284	82	224	792	674	599	925

Richness, Diversity and Evenness: differences among sites

When mean overall taxon richness per sample was compared among study sites, significant differences were found for all three sample methods (pitfall, leaf litter, sweep) (Table 4). *Post hoc* multiple comparisons were unable to fully resolve these differences for pitfall and sweep samples (Table 4). Pitfall sample richness was highest at sites 2 and 4 in the cleared area, but some forested sites had higher richness, on the average than did the remaining two cleared sites (Table 4). The highest mean richness was found at site 1 (cleared) but this value was not significantly different from one of the forested sites and another cleared site (Table 4). Leaf litter taxon richness showed a much more obvious trend, with significantly higher taxon richness at the forested sites than at the cleared sites (Table 4).

When mean taxon richness per sample among sites was evaluated for ants (Formicidae) only, significant differences among sites were found for pitfall and leaf litter samples, but not for sweep samples (Table 5). *Post hoc* multiple comparisons tended to show lower taxon richness in pitfall traps from the forested sites than at some of the cleared sites, but two of the cleared sites (1 and 3) were not significantly different from the forested sites in taxon richness of Formicidae (Table 5). Multiple comparisons for leaf litter taxon richness of ants were even less well resolved, though the highest average richness values tended to occur at forested sites (Table 5).

For the Homoptera (leafhoppers and kin), mean taxon richness per sample among sites did not differ among sites for pitfall and for leaflitter (Table 6), where numbers of leafhoppers were generally quite low (Table 3). Sweep sample mean taxon richness for Homoptera differed among sites, and *post hoc* multiple comparisons indicate that ricness was highest at site 1, and did not differ significantly among the remaining 7 sites (Table 6).

When mean overall species diversity (H') per sample was compared among study sites, significant differences were found for all three sample methods (pitfall, leaf litter, sweep) (Table 4). *Post hoc* multiple comparisons were unable to fully resolve these differences, with no obvious pattern to the groups of sites which were found to be not different from one another for each type of sample (Table 4).

For the ants along, mean H' per sample among study sites differed significantly for pitfall and leaf litter samples but not for sweep samples (Table 5). Pitfall H' was highest in cleared sites, except that site 3 was not significantly different from the forested sites (Table 5). Leaf litter ant diversity was generally higher at forested sites, but these differences were not fully resolved in *post hoc* multiple comparisons (Table 5).

For Homoptera, mean H' per sample among study sites was not different for pitfall and leaf litter samples, but did differ among sweep samples (Table 6). Note that this is the opposite of the condition for ants. *Post hoc* multiple comparisons show the highest diversity of leafhoppers was in sweep samples at site 1 (Table 6), the site that also had the highest Homopteran taxon richness.

Overall mean evenness of taxa per sample did not differ among sites for pitfall or leaf litter samples (Table 4), but did differ among sites for sweep samples, but *post hoc* multiple comparisons were unable to fully resolve these differences (Table 4).

Mean evenness of Formicidae per sample differed among sites for all three sample methods (pitfall, leaf litter and sweep) (Table 5). Evenness was higher in ant pitfall samples at sites 1, 2 and 4 than at the remaining sites (Table 5), and tended to be higher for leaflitter samples at forested sites, although site 8 did not differ from three of the cleared sites in evenness and site 2 did not differ from the forested sites (Table 5). No differences among sites could be detected in *post hoc* multiple comparisons of sweep sample ants (Table 5).

For Homoptera, mean evenness per sample did not differ among sites for pitfall and leaflitter samples (where Homoptera were less frequently recorded [Table 3]), but Homopteran evenness differed among sites for sweep samples (Table 6). *Post hoc* multiple comparisons showed that mean Homopteran evenness was higher at sites 1 and 4 than at the remaing 6 sites (Table 6).

Table 4. One-way ANOVA comparisons between mean values for Species Diversity, Richness and Evenness of all 409 invertebrate taxa at the eight upland sample sites at Camp Atterbury, Indiana, 5-9 July, 2005. Values with the same letter are not significantly different in *post hoc* multiple comparisons (Student-Newman-Keuls), where p was not less than 0.05, *post hoc* comparisons are not evaluated. Sample size is N=9/site for pitfall samples and N=4/site for both sweep and leaf litter samples.

Metric	Site										
Sample Type	1	2	3	4	5	6	7	8	df	F	p
Taxon Richness											
Pitfall	5.67 A	9.0 B	2.67 C D	10.11 B	3.67 C D	2.78 C D	2.22 C	4.78 A D	7,64	26.80	<0.0001
Leaf litter	0.0 A	8.5 A	1.75 A	1.5 A	25.0 B	24.0 B	20.5 B	17.5 B	7,24	22.15	<0.0001
Sweep	19.0 A	8.5 B	7.0 B	12.0 A B	10.0 B	12.5 A B	4.75 B	6.5 B	7,24	4.44	0.0027
Species Diversity (H')											
Pitfall	1.5457 A	1.948 B	0.811 C D	2.172 B	1.107 A C D	0.854 C D	0.609 D	1.194 A C	7,64	15.82	<0.0001
Leaf litter	0.0 A	1.586 B C	0.506 A	0.268 A	2.028 B	1.799 B C	1.601 B C	0.908 A C	7,24	8.30	<0.0001
Sweep	2.740 A	1.705 A B	1.662 A B	2.188 A B	1.426 B	1.976 A B	1.354 B	1.698 A B	7,24	2.92	0.0233
Evenness											
Pitfall	0.288	0.223	0.257	0.218	0.320	0.284	0.182	0.252	7,64	1.97	0.0728
Leaf litter	0.0	0.152	0.170	0.067	0.083	0.077	0.088	0.052	7,24	1.30	0.2923
Sweep	0.152 A D	0.216 A C D	0.258 B C D	0.195 A C	0.142 A	0.173 A C	0.308 B D	0.272 B	7,24	6.60	0.0002

Table 5. One-way ANOVA comparisons between mean values for Species Diversity, Richness and Evenness of 39 ant taxa (Hymenoptera:Formicidae) at the eight upland sample sites at Camp Atterbury, Indiana, 5-9 July, 2005. Values with the same letter are not significantly different in *post hoc* multiple comparisons (Student-Newman-Keuls), where p was not less than 0.05, *post hoc* comparisons are not evaluated. Sample size is N=9/site for pitfall samples and N=4/site for both sweep and leaf litter samples.

Metric	Site										
Sample Type	1	2	3	4	5	6	7	8	df	F	p
Taxon Richness											
Pitfall	1.0 A	2.33 B	0.56 A	4.44 C	0.67 A	0.33 A	0.44 A	0.67 A	7,64	28.47	<0.0001
Leaf litter	0.0 A	3.0 B C	0.0 A	0.75 A B	4.5 C	4.5 C	4.0 C	1.75 A B C	7,24	7.30	<0.0001
Sweep	2.75	2.75	1.0	2.25	2.5	2.0	2.5	2.25	7,24	1.23	0.3233
Species Diversity (H')											
Pitfall	0.219 A	0.653 B	0.000 A	1.334 C	0.000 A	0.000 A	0.000 A	0.000 A	7,64	51.36	<0.0001
Leaf litter	0.000 A	0.789 B	0.000 A	0.000 A	0.921 B	1.240 B	1.081 B	0.520 A B	7,24	7.87	<0.0001
Sweep	0.834	0.692	0.125	0.599	0.415	0.350	0.723	0.673	7,24	1.69	0.1580
Evenness											
Pitfall	0.109 A	0.227 B	0.000 C	0.309 D	0.000 C	0.000 C	0.000 C	0.000 C	7,64	23.77	<0.0001
Leaf litter	0.000 A	0.204 B	0.000 A	0.000 A	0.228 B	0.275 B	0.277 B	0.173 A B	7,24	6.98	0.0001
Sweep	0.304 A	0.204 A	0.063 A	0.271 A	0.166 A	0.153 A	0.242 A	0.297 A	7,24	2.48	0.0457

Table 6. One-way ANOVA comparisons between mean values for Species Diversity, Richness and Evenness of 20 Homopteran taxa (excluding aphids) at the eight upland sample sites at Camp Atterbury, Indiana, 5-9 July, 2005. Values with the same letter are not significantly different in *post hoc* multiple comparisons (Student-Newman-Keuls), where p was not less than 0.05, *post hoc* comparisons are not evaluated. Sample size is N=9/site for pitfall samples and N=4/site for both sweep and leaf litter samples.

Metric	Site										
Sample Type	1	2	3	4	5	6	7	8	df	F	p
Taxon Richness											
Pitfall	0.44	0.0	0.0	0.33	0.11	0.0	0.0	0.0	7,64	2.01	0.0678
Leaf litter	0.0	0.0	0.0	0.25	0.0	0.0	0.75	0.0	7,24	1.96	0.1038
Sweep	3.25 A	0.5 B	0.5 B	1.5 B	0.0 B	0.0 B	0.0 B	0.5 B	7,24	11.07	<0.0001
Species Diversity (H')											
Pitfall	0.077	0.000	0.000	0.077	0.000	0.000	0.000	0.000	7,64	0.86	0.5450
Leaf litter	0.000	0.000	0.000	0.000	0.000	0.000	0.159	0.000	7,24	1.00	0.4553
Sweep	1.063 A	0.000 B	0.000 B	0.448 B	0.000 B	0.000 B	0.000 B	0.000 B	7,24	11.98	<0.0001
Evenness											
Pitfall	0.039	0.000	0.000	0.039	0.000	0.000	0.000	0.000	7,64	0.86	0.5450
Leaf litter	0.000	0.000	0.000	0.000	0.000	0.000	0.080	0.000	7,24	1.0	0.4553
Sweep	0.328 A	0.000 B	0.000 B	0.178 C	0.000 B	0.000 B	0.000 B	0.000 B	7,24	11.42	<0.0001

When we examine species accumulation curves for the three sampling methods (Figure 28), several trends become apparent. One factor to consider is the shape of the curves, by habitat. If the shapes of the curves are very similar, then we might expect the habitats to be similar in diversity, richness and evenness – as can be seen from Figure 28, this is almost always not the case. For pitfall and sweep samples, total species of all taxa (Figure 28a,b,c) accumulate more quickly in cleared than in forested treatements, as the numbers of samples increases. That portion of the fauna sampled by pitfall is richer no matter whether we look at all invertebrates (Figure 28b), or ants (Figure 28e), or only Homopterans (Figure 28h). This is still the case, but less distinctly so, for sweep samples where differences between habitat treatments are less clear for total taxa (Figure 28c) and Formicidae (Figure 28f) than for Homopterans (Figure 28i), the latter group being poorly represented in forested samples. However, in all cases for pitfall and sweep samples, richness was higher in cleared than in forested plots (Figure 28b,c,e,f,h,i). Leaf litter samples tended to show the opposite trend, with richness increasing more rapidly in forested than in cleared treatments for all taxa (Figure 28a), ants (Figure 28b) and Homoptera (Figure 28c).

The extent to which a fauna is thoroughly sampled – or the extent to which sufficient replicate samples were collected to thoroughly sample the fauna – can be evaluated by examining the shape of the species accumulation curve produced by repeated randomization and resampling of the dataset (e.g., Figure 28). This accumulation curve should be steepest towards the left, as each new sample, on the average, results in the addition of new species. For relatively well sampled faunas, the curve should begin to level out towards the right-hand side of the graph, as additional samples tend not to increase the species richness. This latter case is evident for the Formicidae in leaf litter and in pitfall traps when all samples are considered, in leaf litter when only forest samples are considered, and in the pitfall samples when only the cleared treatment is considered (Figure 28d,e). In general, then, the sampling protocol was sufficient to obtain the majority of the ant taxa from leaf litter in the forested habitat and from pitfalls in the cleared habitat. The Hompotera (Figure 28g,h,i) were generally undersampled by our protocol – that is, there were too few samples to obtain the majority of the species, and adding additional replicate samples likely would increase the species richness of Homoptera. In general, our sampling protocol was sufficient to collect the majority of ant taxa (except perhaps in sweep samples), and, especially for the homoptera and for all taxa considered together, additional sample replicates should result in obtaining higher species richness. Thus, as a monitoring tool, focusing exclusively on the ants might provide the most cost-effective and data-rich approach if resources for monitoring are limited. Furthermore, if sampling is restricted to Formicidae, it might make sense to restrict sampling methods to leaf litter and pitfall samples to obtain the most data for the least effort.

Total species richness in samples is never the total richness that actually exists in a community, and Figure 29 presents some estimates of what total species richness might be. The observed species richness, S_{obs}, in Figure 29 represents the same data as the solid line in Figure 28. The other lines are the estimators of total richness (see methods for a more complete discussion). In general, the bootstrap method provided the most conservative (lowest) estimate of richness (Figure 29). Second to the bootstrap method, the Jackknife 2 estimator was the next most consistant metric, increasing gradually with increasing S_{obs}. The ICE and Chao 2 estimators were less consistent (Figure 29). When all taxa are considered (Figure 29a,b,c) the metrics varied rather widely in their final estimates of richness for leaflitter and sweep samples (Figure 29a,c), ranging from around 200 to about 475 taxa for leaflitter samples and from over 200 to just under 700 taxa for sweep samples. For pitfall samples most of the metrics converged at an estimate of a little over 200 species when all taxa are considered (Figure 29b). For the ants in pitfall traps, most of the estimators were not much higher than S_{obs}, suggesting that we captured the majority of the taxa and the the total richness of ants in the summer is close to 25-30 taxa in upland habitats (forested and cleared) at Camp Atterbury (Figure 29e). Leaf litter richness estimators for ants were somewhat less consistant, and placed the total richness in the range of 20 to 35 taxa (Figure 29d). Richness estimators for ants in sweeps samples were less consistant (Figure 29f), but if the Chao 1 estimator is considered an outlier, the total richness is perhaps in the range of 20 to just over 30 taxa. For Homoptera, it appears that the number of samples was not high enough to obtain a good estimate of richness. Notice that the ICE estimator for all taxa (Figure 29a,b,c) and Formicidae (Figure 29d,e,f) tends to have a hump on the left side of the x axis (number of samples) before settling down to a generally increasing curve. This hump is not present for Homoptera in leaf litter samples (Figure 29g) and is on the right (Figure 29h) or towards the middle (Figure 29i) for pitfall and sweep samples, respectively, suggesting that there is inadequate data for at least this estimator.

When we examine the rank order abundance of species within each sample type, the sweep samples stand out as distinct – 70.3 % of the sweep sample taxa are represented by a single specimen, whereas leaf litter and pitfall samples have only 54.4 and 49.1 %, respectively, of their taxa represented by a single specimen (Figure 30). Numbers of taxa represented by two specimens are similar among sample methods, with 15.8, 13.4, and 14.6 % of the taxa represented by two specimens for leaf litter, pitfall and sweep samples, respectively (Figure 30). At the other extreme of abundance, 10.5, 13.4, and 7.0 % of the species are represented by ten or specimens for leaf litter, pitfall, and sweep samples, respectively (Figure 30).

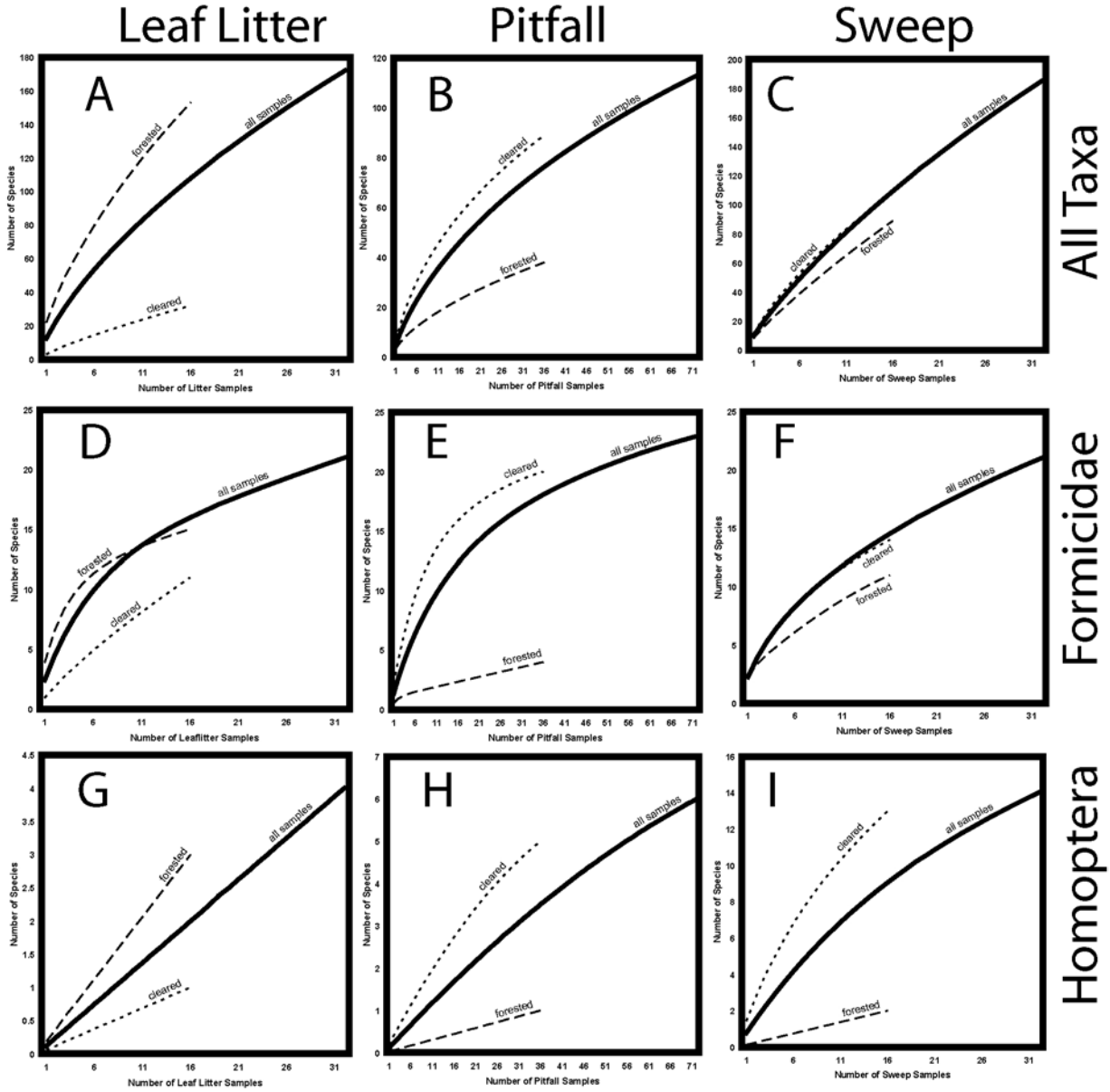


Figure 28. Species accumulation curves for leaf litter (A,D,G), pitfall (B,E,H), and sweep(C,F,I) samples with all taxa (A,B,C), ants only (D,E,F), and leafhoppers and kin only (G,H,I). Solid line represents the combined species accumulation curve using all data from both habitat types. Curves based on random resampling of each data set 1000 times.

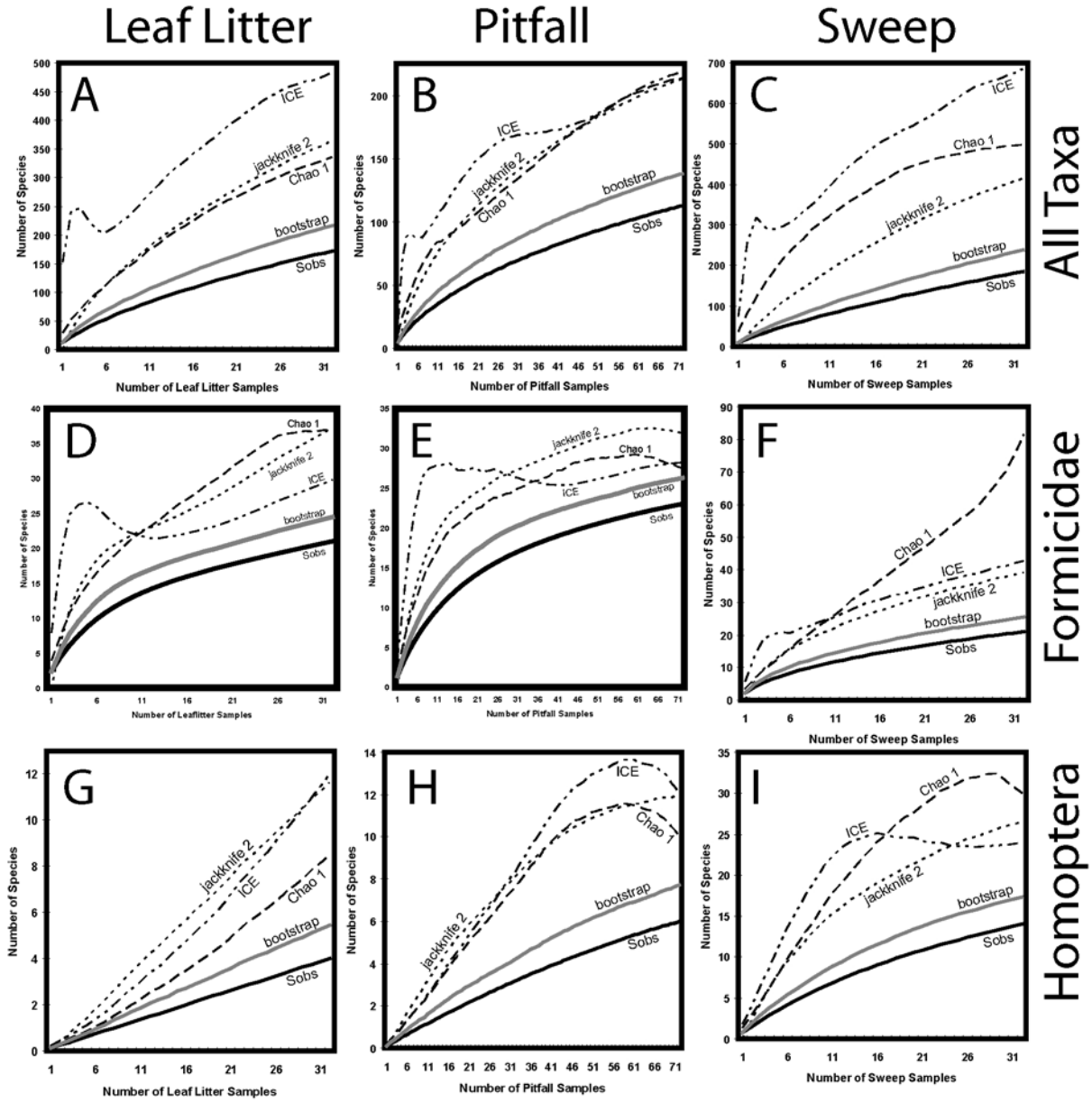


Figure 29. Comparison of various estimates of total species richness for leaf litter (A,D,G), pitfall (B,E,H), and sweep(C,F,I) samples with all taxa (A,B,C), ants only (D,E,F), and leafhoppers and kin only (G,H,I). Curves based on random resampling of each data set 1000 times.

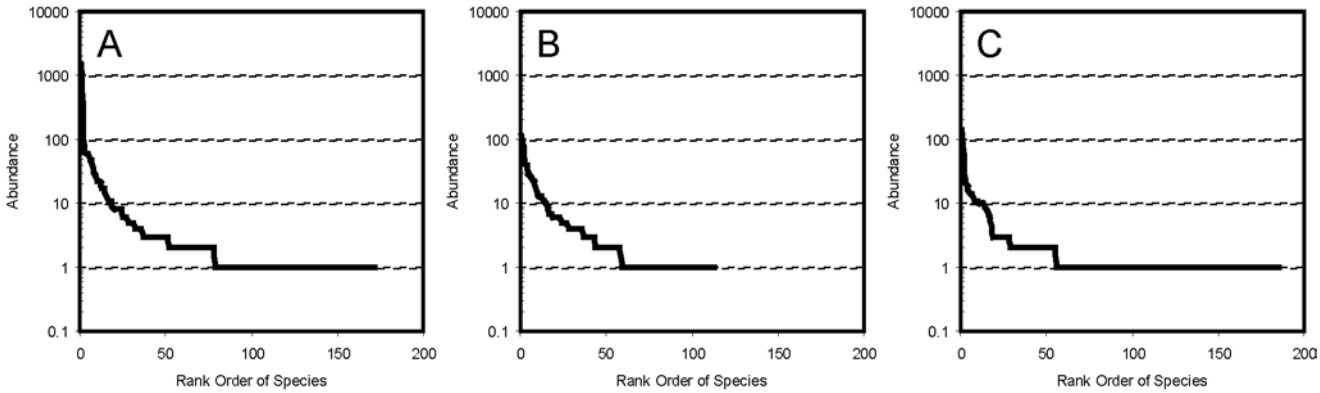


Figure 30. Abundance of all taxa, sorted by rank order of abundance. A – leaf litter, B – pitfall, C – sweep.

Insect Orders

To evaluate relative abundance of taxa and specimens across sample methods and among study sites, we lumped lesser orders of insects and other taxa (myriopods, arachnids, an earthworm and snail) into “other” and retained the orders Collembola, Homoptera, Hemiptera, Coleoptera, Hymenoptera, and Diptera as major orders for analysis.

Leaf litter samples were dominated, in terms of numbers of specimens, by the “other” group (75.1 % of the specimens), largely because 1535 of the 2524 specimens were mites (Acarina) (Figure 31, Table 3). Nonetheless, Hymenoptera (11.6%) and Diptera (7.8%) comprised a large number of the specimens collected. When we examine the data in terms of number of taxa (Figure 32), the overwhelming impact of the Acarina is eliminated because we generally did not identify non-insect invertebrates, thus most mites are considered a single taxon. We can see that in addition to the Hymenoptera and Diptera, which make up 30.8% and 11.6 %, respectively, of the taxa present in leaf litter samples, the Coleoptera and Collembola (25.6%, 9.3%, respectively) are important components of the leaf litter fauna (Figure 32), as are “other” groups such as the thrips (Thysanoptera, Table 3).

Dominant orders, in terms of numbers of specimens, in the pitfall samples were Hymenoptera (29.9%), Collembola (28.4%) and Coleoptera (26.1%) (Figure 31), and when the data in Table 3 are evaluated in terms of numbers of taxa in the major orders, these three orders are still among the most abundant taxa (26.5, 13.3, and 25.7 %, respectively) but the Diptera (13 % of the taxa) are also important contributors to the taxonomic diversity of the pitfall samples (Figure 32).

In terms of numbers of specimens, sweep samples were dominated by Hymenoptera (54.2% of the specimens), but Diptera, Coleoptera, Homoptera and other were also abundant (15.8, 6.8, 6.4, and 11.3%, respectively) (Figure 31). Hymenoptera are also the most diverse in terms of number of taxa in the sweep samples (32.4%), followed by Diptera, Coleoptera, Homoptera, Hemiptera, and other (27.6, 11.9, 8.1, 6.5, and 11.4 %, respectively) (Figure 32).

When we examine major insect orders across sites, pooling all sample methods, Coleoptera and Hymenoptera, in almost all cases, comprised a larger portion of the total numbers of individuals at each site than did the other orders (Figure 33), except for lower numbers of Hymenoptera at site 8 (5.2% of the specimens at F8) and relatively high numbers of Homoptera (16.3% of the specimens) at site 1. Coleoptera were clearly the dominant order of insects at forested sites (46.1-78.4% of the specimens), followed by Hymenoptera (5.2-29.0%), Diptera, and Collembola (Figure 33). At cleared sites, Hymenoptera were generally the numerically dominant order, comprising 26.5 to 48.7% of the specimens (Figure 33). Coleoptera (19.5-27.8 % of the specimens) were also abundant at cleared sites (Figure 33).

Hymenoptera, and to a lesser extent Coleoptera, tended to be represented by the greatest number of taxa relative to other major orders (21.3-42.2%, and 13.3-29.3% of the taxa, respectively) when we look at the diversity of taxa within sites (Figure 34). Both Homoptera and Hemiptera were comprised a greater portion of taxa present at cleared sites (6.8-13.3 % and 3.6-9.3% of the taxa, respectively) than at forested sites (0.0-2.9%, and 1.4-4.5% of the taxa, respectively) (Figure 34). Finally, Diptera comprised a greater portion of the taxa present at most forested sites (except site 7) than they did at the cleared sites (Figure 34).

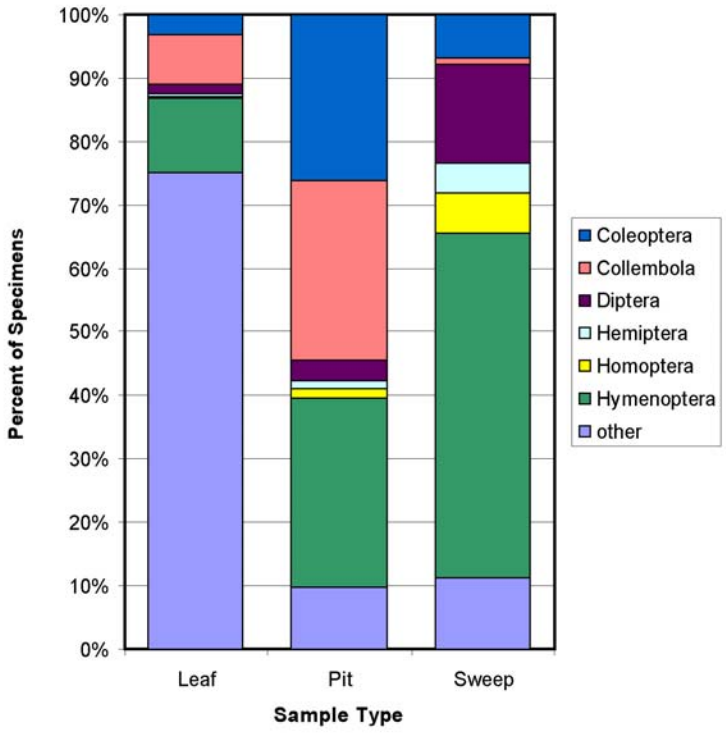


Figure 31. Numbers of specimens in major orders of insects, as percentages, for leaf litter samples (2524 specimens), pitfall samples (675 specimens) and sweep samples (577 specimens) based on July 5-9, 2004 collections from Camp Atterbury, Indiana.

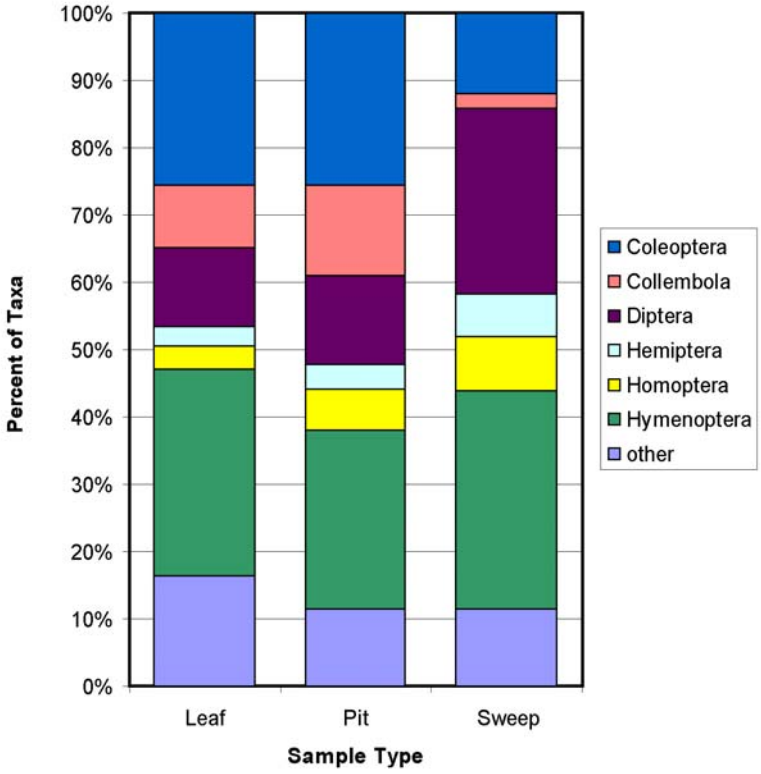


Figure 32. Numbers of morphotaxa in major orders of insects, as percentages, for leaf litter samples (172 taxa), pitfall samples (113 taxa) and sweep samples (185 taxa) based on July 5-9, 2004 collections from Camp Atterbury, Indiana.

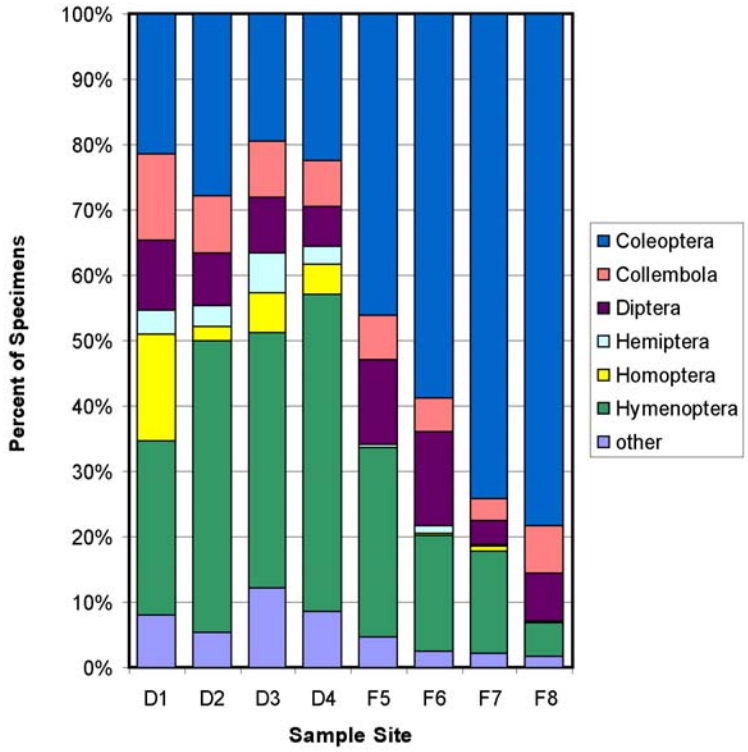


Figure 33. Numbers of specimens in major orders of insects, as percentages, for sites 1-8 (196, 284, 82, 224, 792, 674, 599, and 925 specimens, respectively) based on July 5-9, 2004 collections from Camp Atterbury, Indiana.

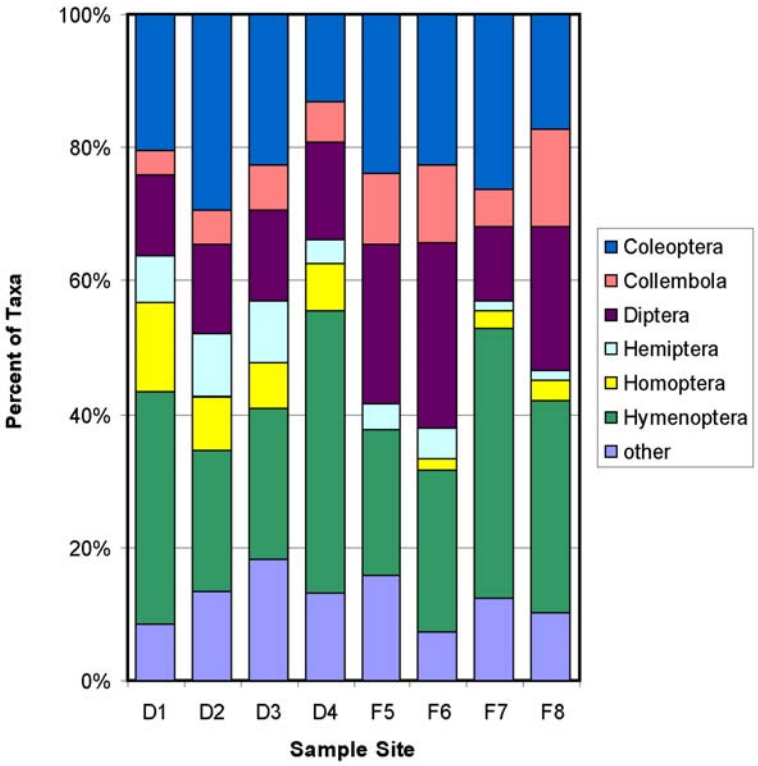


Figure 34. Numbers of morphotaxa in major orders of insects, as percentages, for sample sites 1-8 (83, 75, 44, 83, 101, 111, 72, and 69 taxa, respectively) based on July 5-9, 2004 collections from Camp Atterbury, Indiana.

Similarity among sites and samples based on morphospecies presence/absence

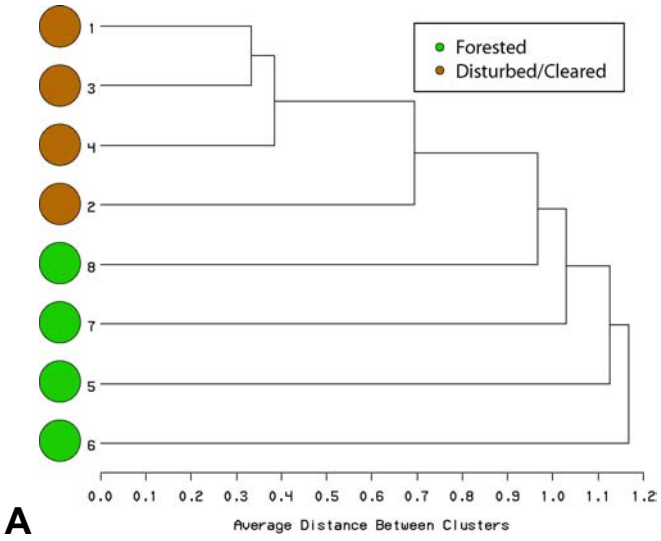
Cluster analysis of invertebrate species presence/absence data produced a dendrograms reflecting the overall division of sites into forested and cleared lands (Figure 35). Cleared sites were distinctly more similar to one another than to forested sites for leaf litter (Figure 35a), forested sites were more similar to one another than to cleared sites for pitfall data (Figure 35b), and the two treatments (forest/cleared) were less clearly distinct when morphospecies presence absence data are examined for sweep data (Figure 35b). At the level of individual samples, morphospecies presence/absence in leaf litter samples still showed almost complete separation between treatments, with nearly all cleared sites being more similar to one another than to forested sites (Figure 36), a trend that is somewhat exaggerated by the complete lack of leaf litter invertebrates in several of the samples for which almost no litter was present. Individual pitfall traps also showed a strong tendency to group by treatment (forest/cleared), with the morphospecies presence/absence in most forest pitfall samples being more similar to one another than to most cleared pitfall samples (Figure 37). Individual sweep samples from forested areas tended to be somewhat more similar to one another than to sweep samples from the cleared areas (Figure 38), although the two treatments are less well separated at the level of individual samples than are leaf litter and pitfall samples.

Homoptera (Leafhoppers and kin)

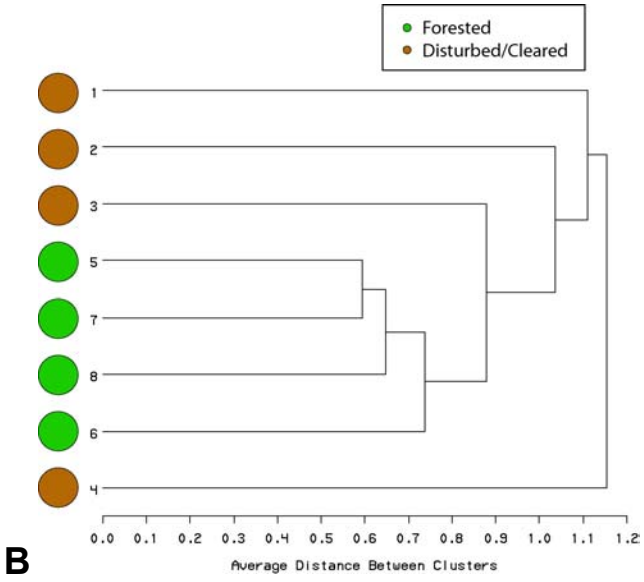
This group is the only one for which we have fairly complete data from malaise traps (Table 7). The absence of Homoptera from the mailaise trap at site 8 appears to be the result of misplaced specimens (the rest of the site 8 malaise sample is intact). Species richness of Homoptera was higher in malaise samples than in sweep, leaf litter, or pitfall samples. In fact, all species of Homoptera, excluding Aphididae, were obtained in the malaise samples. Numbers of specismens varied among sites, but no pattern in overall abundance could be discerned (Table 7). Examination of Table 7, however, suggests that even with this limited malaise sampling, it is readily apparent that some taxa are restricted primarily to the forested sites, and others to the cleared sites. These observations are supported by the cluster analysis (Figure 39), in which the forested sites are grouped together as more similar to one another than to the cleared sites.

Most of the homopteran species encountered are common in woodland habitats throughout the eastern U.S. and none of the species encountered are considered rare. In general, samples from relatively undisturbed woods are expected to contain several species of the following genera: *Arboridia, Erythroneura, Eratoneura, Erasmoneura, Osbornellus,* and *Scaphoideus*. Many species of these genera appear to have very narrow host preferences, but unfortunately the host plants remain poorly documented. Most appear to be restricted to a single genus or species of tree. The leafhopper *Jikradia olitoria* (Figure 40) and the flatid *Metcalfa pruinosa* (Figure 41) are common in woodlands, *Agalia constricta* (Figure 42) is common in old fields and forest edges, and species of *Chlorotettix, Gyponana, Ponana*, and *Scaphytopius* are typically indicative of forest edges. *Chlorotettix* species are often associated with wetlands, where many species specialize on sedges. The delphacid genus *Delphacodes* (Figure 43) also contains sedge-feeding species. Species of the leafhopper genera *Cuerna, Laevicephalus*, and *Stirellus*, the spittlebug *Philaenus spumarius* (Figure 44), the treehopper genera *Campylenchia* (Figure 45a) and *Entylia* (Figure 45b), and the planthopper genus *Bruchomorpha* are typical of more open habitats. Most of these occur in pristine as well as highly disturbed habitats (e.g., old fields), while some such as *Myndus* sp. (Figure 46) (Homoptera:Cixiidae) are more typically found on native prairie grasses. Several of the homopterans which are commonly encountered are introduced species, including *Philaenus spumarius* and *Aphrodes bicincta* (Figure 47).

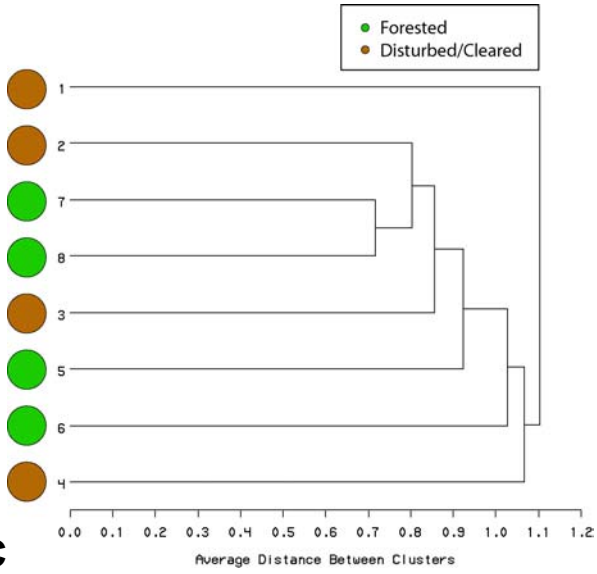
The life histories of these species are similar overall. Females insert eggs into the living tissue of their host plant. The eggs hatch and the immatures (nymphs) feed on plant sap, undergoing five molts before reaching the adult stage. Most large species (e.g., *Osbornellus, Scaphoideus, Chlorotettix*) have only one generation per year; smaller species (e.g., *Arboridia, Erythroneura*) usually have two. Some species overwinter as adults in leaf litter (e.g., *Aboridia*, *Erythroneura*) but others (e.g., *Osbornellus, Scaphoideus*) overwinter in the egg stage.



A



B



C

Figure 35. Cluster analysis (UPGMA average linkage analysis, RMS distance) of eight upland study sites based on presence/absence of all invertebrate taxa (pooled across all samples within sample type) for leaf litter (A), pitfall (B), and sweep (C) samples.

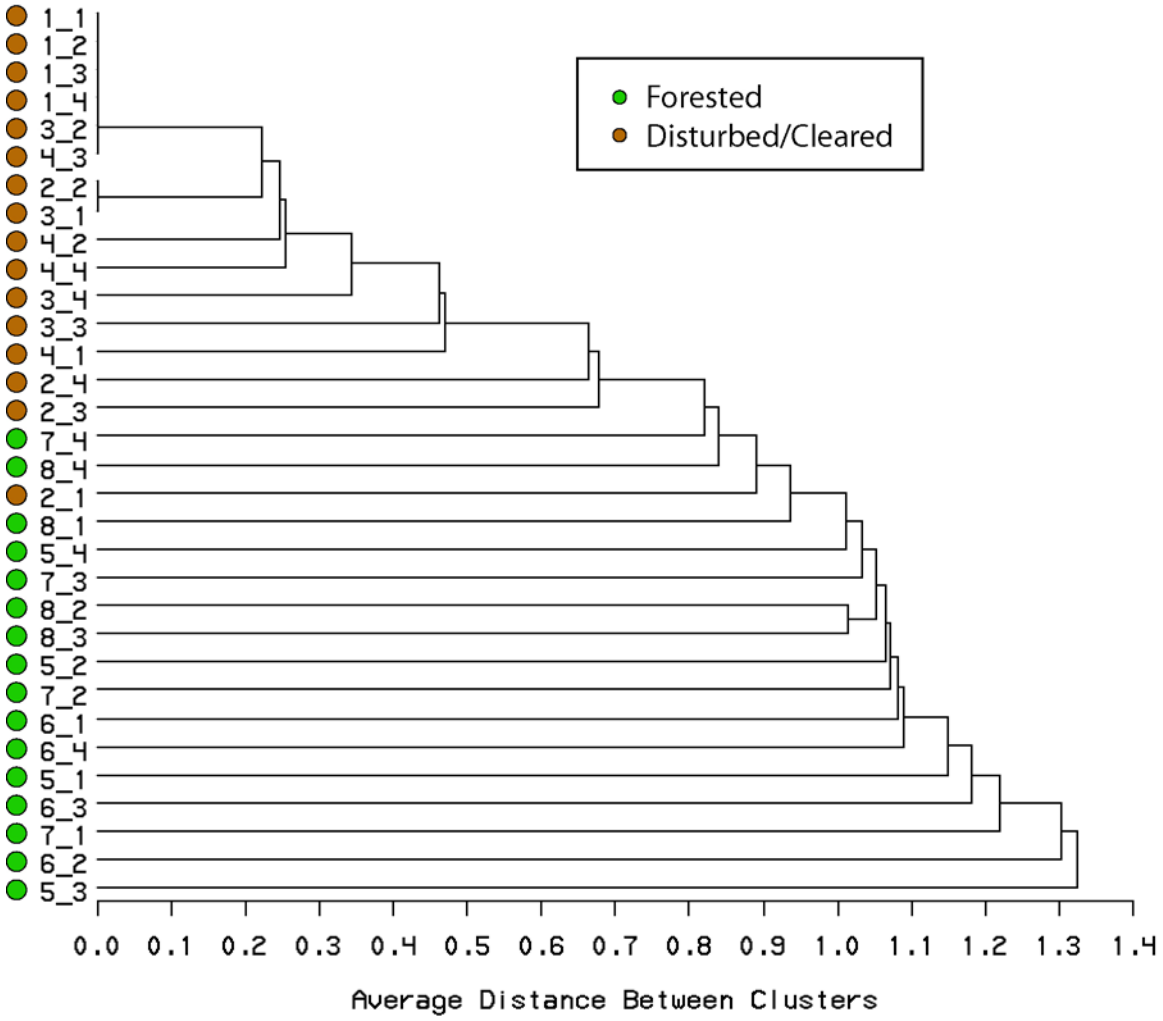


Figure 36. Cluster analysis (UPGMA average linkage analysis, RMS distance) of leaf litter samples for all sites based on presence/absence of all taxa. Two number sample code represents site number and sample replicate, respectively.

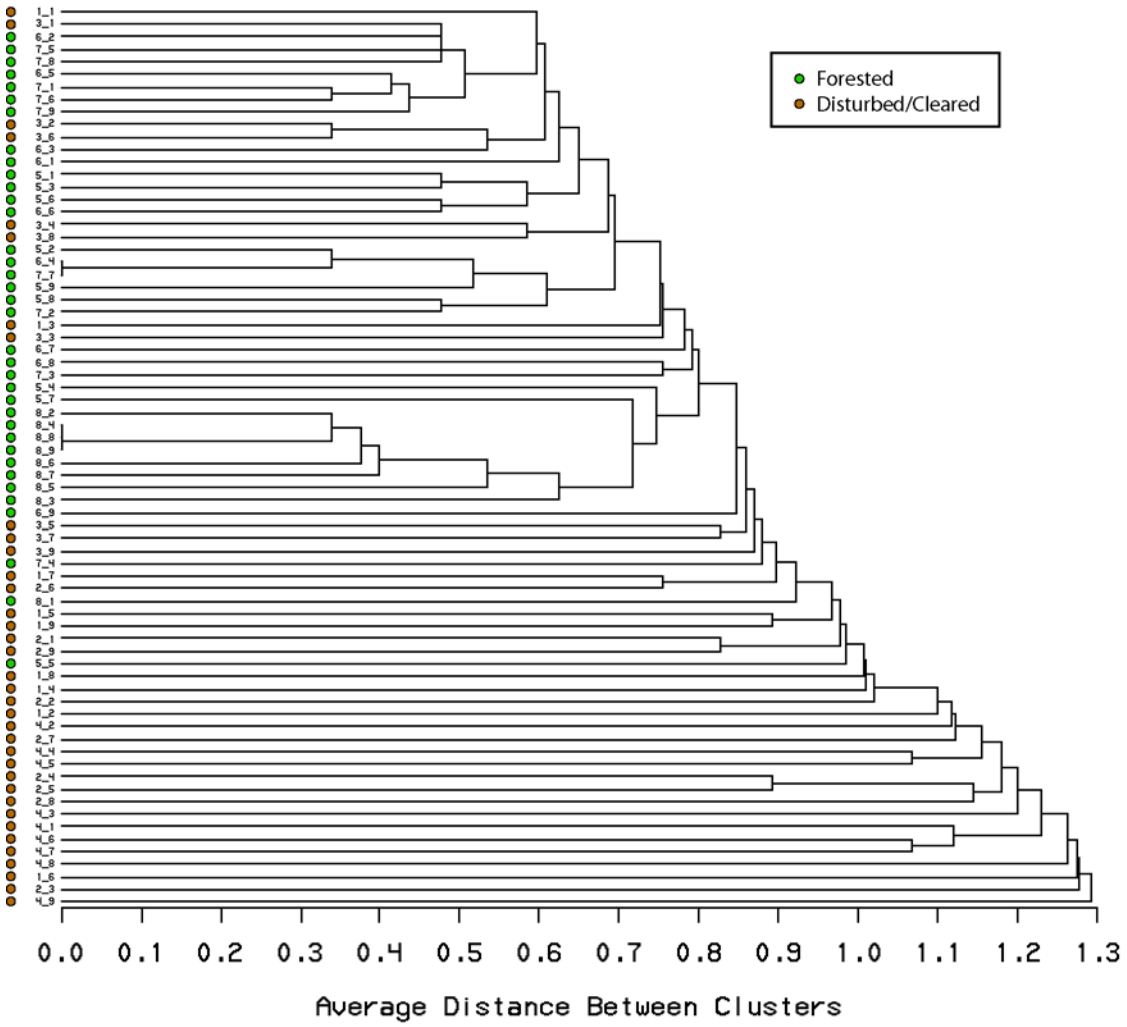


Figure 37. Cluster analysis (UPGMA average linkage analysis, RMS distance) of pitfall samples for all sites based on presence/absence of all taxa. Two number sample code represents site number and sample replicate, respectively.

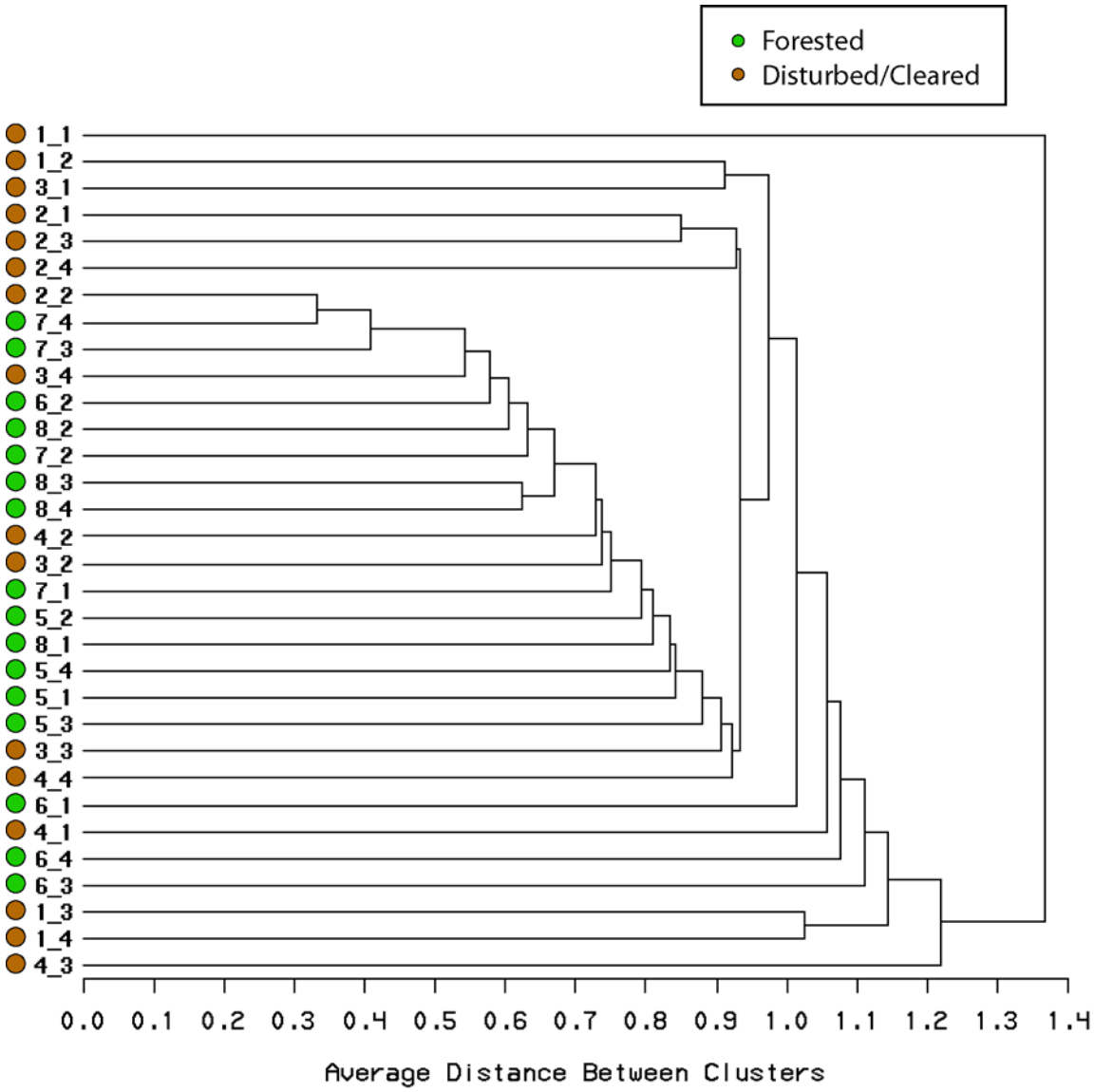


Figure 38. Cluster analysis (UPGMA average linkage analysis, RMS distance) of sweep samples for all sites based on presence absence of all taxa. Two number sample code represents site number and sample replicate, respectively.

Table 7. Homoptera collected from malaise trap samples at Camp Atterbury, Indiana, July 2004. Sites 1-4 are cleared, 5-7 are forested.

Superfamily	Family	Species	1	2	3	4	5	6	7	8	Totals
Aleyrodoidea	Aleyrodidae	unidentified	0	0	5	0	0	0	0	0	5
Psylloidea	Psyllidae	unidentified	0	0	2	0	0	0	0	0	2
Cercopoidea	Aphrophoridae	*Aphrophora cribrata*	1	0	0	0	0	0	0	0	1
		Philaenus spumarius	1	0	0	0	0	0	0	0	1
	Cercopidae	*Prosapia bicincta*	0	1	0	0	0	0	0	0	1
Fulgoroidea	Cixiidae	*Myndus* sp.	0	1	0	0	0	0	0	0	1
	Delphacidae	*Delphacodes basivitta*	0	0	1	0	0	0	0	0	1
		Liburniella ornata	0	0	1	0	0	0	0	0	1
	Derbidae	*Apache degeerii*	0	0	0	0	0	0	1	0	1
		Cedusa sp.	0	0	0	0	0	1	2	0	3
Membracoidea	Cicadellidae	*Agallia constricta*	1	0	0	1	0	0	0	0	2
		Arboridia sp.	1	0	5	1	12	1	7	0	27
		Balclutha impicta	1	0	0	0	0	0	0	0	1
		Balclutha abdominalis	0	0	2	0	0	0	0	0	2
		Chlorotettix lusorius	0	0	0	0	0	0	1	0	1
		Chlorotettix galbanatus	13	0	1	1	0	0	0	0	15
		Chlorotettix viridius	2	0	0	0	0	0	0	0	2
		Chlorotettix vacunus	3	2	0	1	0	0	0	0	6
		Cuerna costalis	3	0	2	0	0	0	0	0	5
		Dikraneura angustata	0	0	1	1	2	1	0	0	5
		Dikrella sp.	1	0	1	0	0	1	1	0	4
		Draeculacephala antica	1	0	0	0	0	1	0	0	2
		Empoasca sp.1	17	4	13	4	1	2	5	0	46
		Erasmoneura vulnerata	0	0	0	0	17	0	0	0	17
		Erasmoneura sp.	1	1	0	0	0	1	2	0	5
		Eratoneura sp.1	5	1	3	0	14	1	13	0	37
		Eratoneura sp.2	1	0	1	0	0	0	0	0	2
		Eratoneura sp.3	1	0	0	0	0	0	0	0	1
		Eratoneura sp.4	1	0	0	0	0	0	0	0	1
		Erythroneura sp.1	0	1	1	0	1	13	2	0	18
		Erythroneura sp.2	0	0	0	0	1	2	2	0	5
		Erythroneura sp.3	0	0	0	0	3	1	2	0	6
		Erythroneura sp.4	0	0	0	0	5	1	1	0	7
		Graphocephala versuta	0	3	3	0	0	0	0	0	6
		Gyponana angula	1	0	0	0	0	0	0	0	1
		Gyponana sp.	2	0	0	1	0	1	1	0	5
		Idiodonus kennicotti	1	0	0	0	0	0	0	0	1
		Idiocerus moniliferae	1	0	0	0	0	0	0	0	1
		Osbornellus clarus	7	0	0	0	0	0	0	0	7
		Osbornellus consors	0	3	0	2	0	0	1	0	6
		Paraphlepsius irroratus	20	0	0	2	0	0	0	0	22
		Ponana limbatipennis	0	0	0	0	0	0	1	0	1
		Scaphoideus angustatus	3	0	2	0	0	0	0	0	5
		Scaphoideus cinereus	1	0	0	0	1	0	0	0	2
		Scaphoideus major	0	0	0	0	3	0	0	0	3
		Scaphoideus torquus	0	1	0	0	0	0	0	0	1
		Scaphoideus sp.1	0	0	2	0	0	1	0	0	3
		Scaphytopius acutus	2	0	0	0	0	0	0	0	2
		Scaphytopius verecundus	2	0	0	0	0	0	0	0	2
		Typhlocyba sp.	2	0	0	0	0	0	0	0	2
	Membracidae	*Campylenchia latipes*	0	1	0	0	0	0	0	0	1
		Entylia carinata	0	1	1	0	0	0	0	0	2
	TOTALS		96	20	47	14	60	28	42	0	307

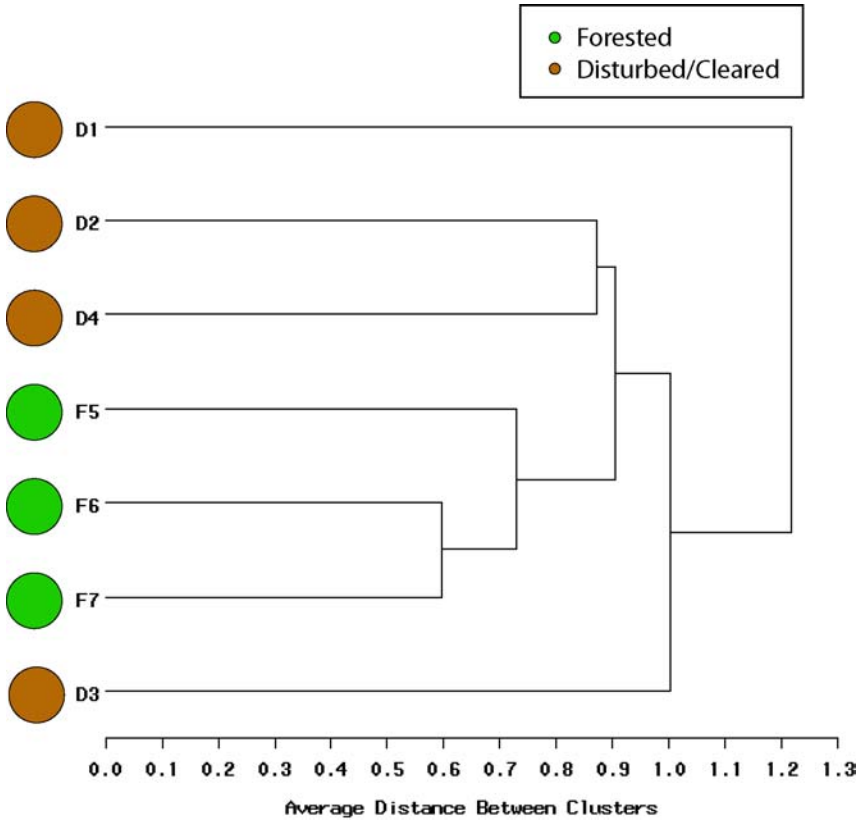


Figure 39. Cluster analysis (UPGMA average linkage analysis, RMS distance) of seven upland study sites based on presence/absence of Homoptera taxa in malaise trap samples. Site 8 is excluded, as apparently the sample was lost or misplaced prior to identification.

Figure 40. *Jikradia olitoria* (Homoptera: Cicadellidae), a common woodland leafhopper. Photograph by Chris Dietrich.

Figure 41. *Metcalfa pruinosa* (Homoptera: Flatidae) common woodland flatid planthopper. Photograph by Chris Dietrich.

Figure 42. *Agallia constricta* (Homoptera: Cicadellidae), a native generalist leafhopper common in old fields and forest edges. Photograph by Chris Dietrich.

Figure 43. *Delphacodes* sp. (Homoptera: Delphacidae), a sedge-feeding planthopper. Photograph by Chris Dietrich.

Figure 44. *Philaenus spumarius* – a spittle mass made by nymph of common introduced European spittlebug (Homoptera: Aphrophoridae). Photograph by Chris Dietrich.

A

B

Figure 45. A) *Campylenchia latipes* (Homoptera: Membracidae) nymphs and B) *Entylia carinata* (Homoptera: Membracidae) common composite-feeding treehoppers often found in old fields and prairies. Photograph by Chris Dietrich.

Figure 46. *Myndus* sp. (Homoptera:Cixiidae) is a cixiid planthopper that feeds on native prairie grasses. Photograph by Chris Dietrich.

Figure 47. *Aphrodes bicincta*, a common grass-feeding leafhopper introduced from Europe. Photograph by Chris Dietrich.

Subfamily Formicinae

Brachymyrmex depelis

This subterranean species (Figure 48) is rarely seen except when releasing alates that appear to surface from every crevice in areas where it occurs. The smallest North American ant, it occurs from coast to coast and is often found in highly disturbed areas including entirely urban environments (Creighton 1950). It can be easily identified by its small size and 10 segmented antenna (most ants have 12). Its subterranean nature also appears to promote its coexistence with invasive ants.

Camponotus sp.

The genus *Camponotus* includes the "carpenter ants". While many live in damaged wood (giving them a bad name to home owners) many live only in live wood or in soil. There are many species in North America. The most common species in our sampling was *C. subbarbatus*, but *C. pennsylvanicus* (Figure 49) also occurred in sweep samples at one cleared site.

Formica spp.

A large genus of temperate (Nearctic, Palearctic) species, including a many generalized foragers, and many species which tend homopterans. Our samples include *Formica fusca* (Figure 50) and *Formica nitideventris* (Figure 51).

Lasius spp.

The genus Lasius contains about 75+ species broadly distributed across the Palearctic, Nearctic, and temperatue portions of the Oriental region. Most are generalized foragers, and some tend homopterans. Our samples include three species, *Lasius alienus* (Figure 52), *Lasius neoniger* (Figure 53), and *Lasius umbratus* (Figure 54).

Prenolepis imparis (the winter ant):

Unlike most native above-ground foraging ant species, *P. imparis* (Figure 55) is most active in the late fall and winter months (Tschinkel 1987, Suarez et al. 1998). This species has very deep nests (up to 3 meters) and can store huge amount of liquid resources in the "crop" of workers which can live up to two years (Tschinkel 1987). It is one of the more common North American ants occurring from Mexico into Canada, from coast to coast. Interestingly, *P. imparis* appears to be able to withstand the presence of invasive ants throughout its range. Temporal niche partitioning may allow long-term coexistence between the winter ant and introduced ant species (Ward 1987).

Subfamily Myrmicinae

Aphaenogaster rudis complex

This group of species is one of the most common in temperate eastern forest. It undoubtedly consists of 3 or more species, but until the group is revised we will be unable to tell them apart.

Crematogaster sp.

Commonly known as "acrobat ants" (Figure 56), species in the genus *Crematogaster* can nest in either the soil or in branches. Their name comes from the incredible ability to walk on almost any surface. The characteristic heart shaped abdomen easily tells members of this genus apart from other ants.

Leptothorax curvispinosus

This is a small yellow ant (Figure 57) with a characteristic 11 segmented antenna. It is very common in the eastern United States extending as far west as central Texas and the Dakotas. They nest primarily in plant cavities including stems and acorns but can be found nesting in moist soil in forested areas (MacKay 2000).

Monomorium minimum

This (Figure 58) is another common species in eastern North America. It occurs in almost any habitat including highly disturbed areas. They can have multiple queens per nest.

Myrmecina americana

This is the only species (Figure 59) of the genus and occurs widely throughout North America (from coast to coast). It is generally found in forested areas with well-developed litter. Due to its cryptic habits, very little is known about the biology of this widely occurring species.

Figure 48. *Brachymyrmex depelis* (Hymenoptera: Formicidae), photograph by Alex Wild, used by permission.

Figure 49. *Camponotus pennsylvanicus* (Hymenoptera: Formicidae), photograph by Alex Wild, used by permission.

Figure 50. *Formica fusca* (Hymenoptera: Formicidae), photograph by Alex Wild, used by permission.

Figure 51. *Formica nitideventris* (Hymenoptera: Formicidae), photograph by Alex Wild, used by permission.

A

B

Figure 52. *Lasius alienus* (Hymenoptera: Formicidae), A) worker with larvae, B) worker. Photographs by Alex Wild, used by permission.

A

B

Figure 53. *Lasius neoniger* (Hymenoptera: Formicidae), A) worker, B) nest. Photographs by Alex Wild, used by permission.

Figure 54. *Lasius umbratus* (Hymenoptera: Formicidae), photograph by Alex Wild, used by permission.

A

B

Figure 55. *Prenolepis impairs* (Hymenoptera: Formicidae). A) Worker with brood, B) Worker. Photographs by Alex Wild, used by permission.

Figure 56. *Crematogaster lineolata* (Hymenoptera: Formicidae), photograph by Alex Wild, used by permission.

Figure 57. *Leptothorax curvispoinosus* (Hymenoptera: Formicidae), photograph by Alex Wild, used by permission.

Figure 58. *Monomorium minimum* (Hymenoptera: Formicidae), photograph by Alex Wild, used by permission.

Figure 59. *Myrmecina americana* (Hymenoptera: Formicidae), photograph by Alex Wild, used by permission.

Pheidole bicarinata
This relatively common species of *Phiedole* (Figure 60) occurs from the east coast to Colorado, from Florida to New Jersey. It is one of the few north temperate species of this genus (see also Figure 61), which is highly speciose in tropical and subtropical regions. It will nest in logs or in the soil under stones. It will often be found in open disturbed grassy areas including lawns (Wilson 2003).

Pyramica sp.
These members of the Dacetini group are small, rarely seen litter ants. They have special glands below their petiole which are unique to this group and whose purpose is still unknown. They have elongate jaws that snap shut very quickly and are thought to be specialized predators on Collembola.

Solenopsis molesta (the thief ant):
This species (Figure 62, in the *Solenopsis* subgenus *Diplorhoptrum*) is one of the most common North American ant species. It forages both above and below ground and may be the least vulnerable of all ant species to habitat loss and fragmentation (Suarez et al. 1998). It has been reported to forage only up to 10 m from the nest (Human and Gordon 1996) and may not need large areas to support large populations. *Solenopsis molesta* has also been reported to behave parasitically upon many other ant species (Wheeler and Wheeler 1973, Snelling and George 1977) including one invasive species (Suarez et al. 1998). It will frequently nest next to host ant species and raid their nests for food and eggs.

Subfamily Ponerinae
Ponera pennsylvanica
This primitive ant (Figure 63) lives almost entirely underground although stray workers are often collected under rocks or in litter samples. It occurs primarily in forested habitats and can be quite common in eastern deciduous forests.

Subfamily Dolichoderinae
Tapinoma sessile
This (Figure 64) is one of North America's most widespread ants occurring in a wide variety of habitats from coast to coast. Its pungent odor has given it the common name "odorous house ant". It is one of the most common native pest ants (along with carpenter ants of the genus *Camponotus*) (Knight and Rust 1990). It is unusual among ants in that it does have a formal nest but will relocate its nest regularly in a somewhat nomadic fashion.

Abundance of ants, both in number of specimens and number of taxa, was positively correlated with leaflitter depth for winkler samples (Figure 65a,b).

Other Hymenoptera

Of the major insect orders, the Hymenoptera dominated in numbers of species and individuals in the sweep, pitfall and leaf litter samples. The majority of collected hymenopterans were from a single family, the ants (Table 3; 701 individuals belonging to 38 morphospecies). However, the parasitic hymenoptera (=Parasitica) showed greater diversity (102 individuals from 69 morphospecies and 17 families). In addition to the Formicidae, three other aculeate families were collected: Halictidae (2 morphospecies, 4 individuals), Colletidae (1 individual), and Rhopalosomatidae (1 individual, Figure 66), the latter being a brachypterous female. Rhopalosomatidae are infrequently collected, and the North American contingent consists of only 3 species, each belonging to a different genus (Borror et al., 1989; Goulet & Huber, 1993), at least some of these are known to attack crickets as larvae. The individual in our collection is probably *Olioxon banksi*l. Although *O. banksii* is known from numerous states in the eastern US (Stange 1991), our collection is apparently the first record for Indiana.

Most of the Parasitica with known host associations belonged to families that are predominantly or exclusively egg parasitoids (Table 3; 42 individuals; Encyrtidae, Mymaridae, Platygastridae, Scelionidae, Trichogrammatidae). Next in importance are the larval and pupal fly parasitoids which are mainly represented by diapriids (26 individuals). The morphospecies belonging to Pteromalidae, Proctotrupidae and Eucoilidae, might attack flies, but other hosts are also utilized by species in these families. The remaining morphospecies in the collection are primarily braconids, ichneumonids or ceraphoronids. The braconids and ichneumonids are closely related and parasitize a variety of life strategies and holometabolous hosts, including flies. Little is known about the life histories of the ceraphoronids.

The typical life history of a parasitic wasp is: a female oviposits on, in, or near a host; her progeny feeds on the host thereby killing it; pupation occurs on, in, or near the host. Because her young will develop on a single host, females of some species base the number of eggs laid and/or sex of progeny on one or more host factors. Larval success depends on overcoming a host's defenses. Egg parasitoids are at an advantage because insect eggs lack immune

Figure 60. *Pheidole bicarinata*, major (top) and minor workers (Hymenoptera: Formicidae), photograph by Alex Wild, used by permission.

Figure 61. *Pheidole pillifera* with larvae (Hymenoptera: Formicidae), photograph by Alex Wild, used by permission.

Figure 62. *Solenopsis molesta* (Hymenoptera: Formicidae), photograph by Alex Wild, used by permission.

Figure 63. *Ponera pennsylvanica* (Hymenoptera: Formicidae), photograph by Alex Wild, used by permission.

A

B

Figure 64. *Tapinoma sessile* (Hymenoptera: Formicidae). A) workers with brood, B) worker. Photographs by Alex Wild, used by permission.

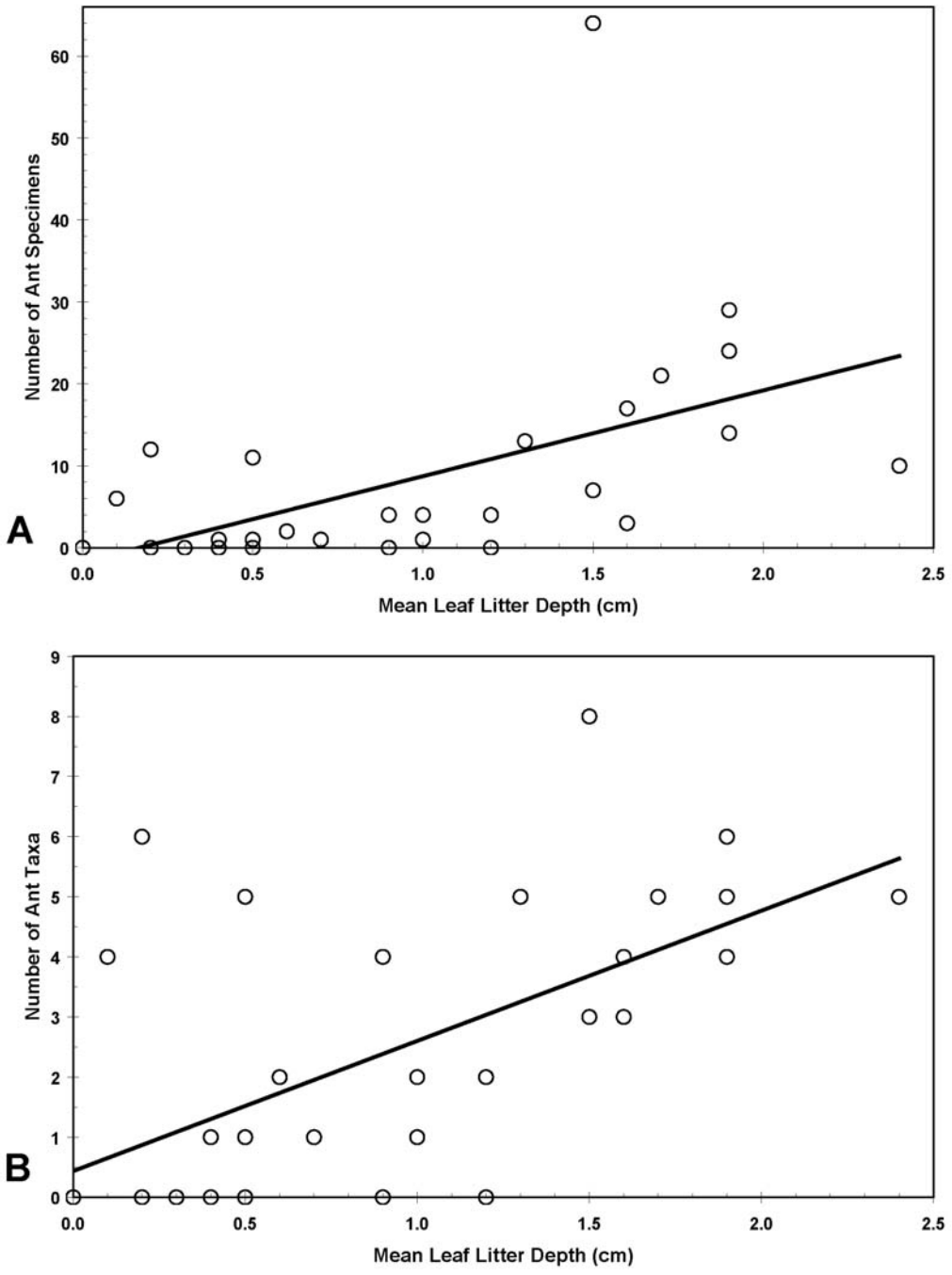


Figure 65. Number of ant specimens (A) and number of ant taxa (B) in winkler samples in relation to mean leaflitter depth (n=10 measurements per 0.5 m^2 winkler sample). Line of best fit is shown: A: y = 10.476x - 1.7455, R^2 = 0.2842; B: y = 2.1656x + 0.4369, R^2 = 0.3625.

Figure 66. Brachypterous adult female Rhopalosomatidae (Hymenoptera) collected in pitfall trap at site 8, July 2004. Scale bar = 5 mm.

systems. Thus, an egg parasitoid has little if any, need for specializations to overcome its host; and these species tend to be polyphagous, sometimes attacking hosts from different orders (see Strand, 1986; Hirose, 1994). Because of this tendency and the paucity of information on host specificity for most species, egg parasitoids would be poor choices as indicators of insect biodiversity. Host egg size and quality has been shown to determine the adult morphology of at least one trichogrammatid species. Species of lepidopteran host affected wing length and, thereby, flight abilities. The individual parasitioid morphology ranged from fully winged to apterous (David Orr, pers. comm.). Because of their tendency to polyphagy and host-dependent morphology, it is possible that the number of morphospecies found at Camp Atterbury would overestimate actual egg parasitoid diversity.

The likely choice of a parasitic hymenoptera taxon to be used as an indicator of overall insect biodiversity is the Ichneumonidae. There are more known ichneumonid species than in any other hymenopteran family, and they are the best-studied group in Parasitica. Additionally, ichneumonids are usually highly specialized. It is thought that the majority of North American ichneumonids use 3 or fewer host species (Townes & Townes, 1951: in Strand, 1986). An additional factor favoring the use of ichneumonids is that they tend be large for parasitoids, therefore, they are easier to identify using keys. Although few individuals were found in the analyzed sweep, leaf litter and pitfall samples, Ichneumonidae will likely be the most speciose of the Parasitica in Malaise samples (based on preliminary sorting of 2 malaise samples). If the ichneumonids prove useful as bioindicators, the sampling method could consist exclusively of malaise trapping. This would simplify the overall experimental design and reduce time spent in the field, but would require use of an appropriate taxonomic expert.

Teratological Anomolies

Among the thousands of insects we examined, we found two beetles, both in the family Scarabaeidae, with morphological anomalies. The first of these was a beetle (Figure 67c) which has a small, misshapen right antenna (Figure 67a) and right rear leg (Figure 67d), when compared with the normal left side (Figure 67b,e). These anomalies may be attributable to damage during the larval or pupal stage of the beetle. The other individual, however, was unusual in that the left rear leg is double, beginning with the femur, giving the appearance of a seven legged beetle (Figure 68a,b). The origins of this second leg are certainly unknown, but similar anomalies have be reported before for beetles, such as multiple tarsae on *Meloe corallifer* (Ortuño and Hernández 1993), an extra leg on the carabid beetle *Psalidognathus* sp. (Osuna 1992), and the “godzilla” mutant of *Tribolium castaneum* (Sulston and Anderson 1996), which has its origins in the embryonic segmentation process during development. Of particular concern, however, is the possibility that the sort of mutation observed in Figure Y could be induced by environmental contaminants. While such a link between environmental contaminants and teratological anomalies has been questioned (for example Ankley et al. [2004] suggest parasitism by tremetodes may play a role in hind leg anomalies of anurans), it certainly cannot excluded from consideration. Indeed, Migliorini et al (2004) examined heavy metal effects on soil arthropod communities in a shooting range, and found that lead (Pb) from spent pellets bioaccumulates in soil organisms, thus entering the trophic network.

Parasitic Coleoptera

Perhaps the most interesting insect collected was the beetle family Rhipiphoridae (see photo on cover of this report). Rhipiphorids are widely distributed but uncommonly collected, especially adult males. The larvae of this family are parasitic on wasps and bees. In some species, perhaps including the species we collected, the female lays eggs on flower buds, and the eggs hatch as the flowers begin to bloom. First instar larvae attach themselves to the host bee when it visits the flower, then drop off the adult bee when it returns to its nest, when eggs of the host bee hatch, the rhipiphorid larva feeds on the larva. Eventually the rhipiphorid pupates in the bee’s nest and emerges later for a brief adult life – adults in some species may live about two weeks. We collected an adult male in a sweep sample at site 1 (Figure 69a) and, in another sweep sample at the same site on the same date (6 July 2004), we collected a halictid bee with a first instar larval rhipiphorid attached to its wing (Figure 69b,c).

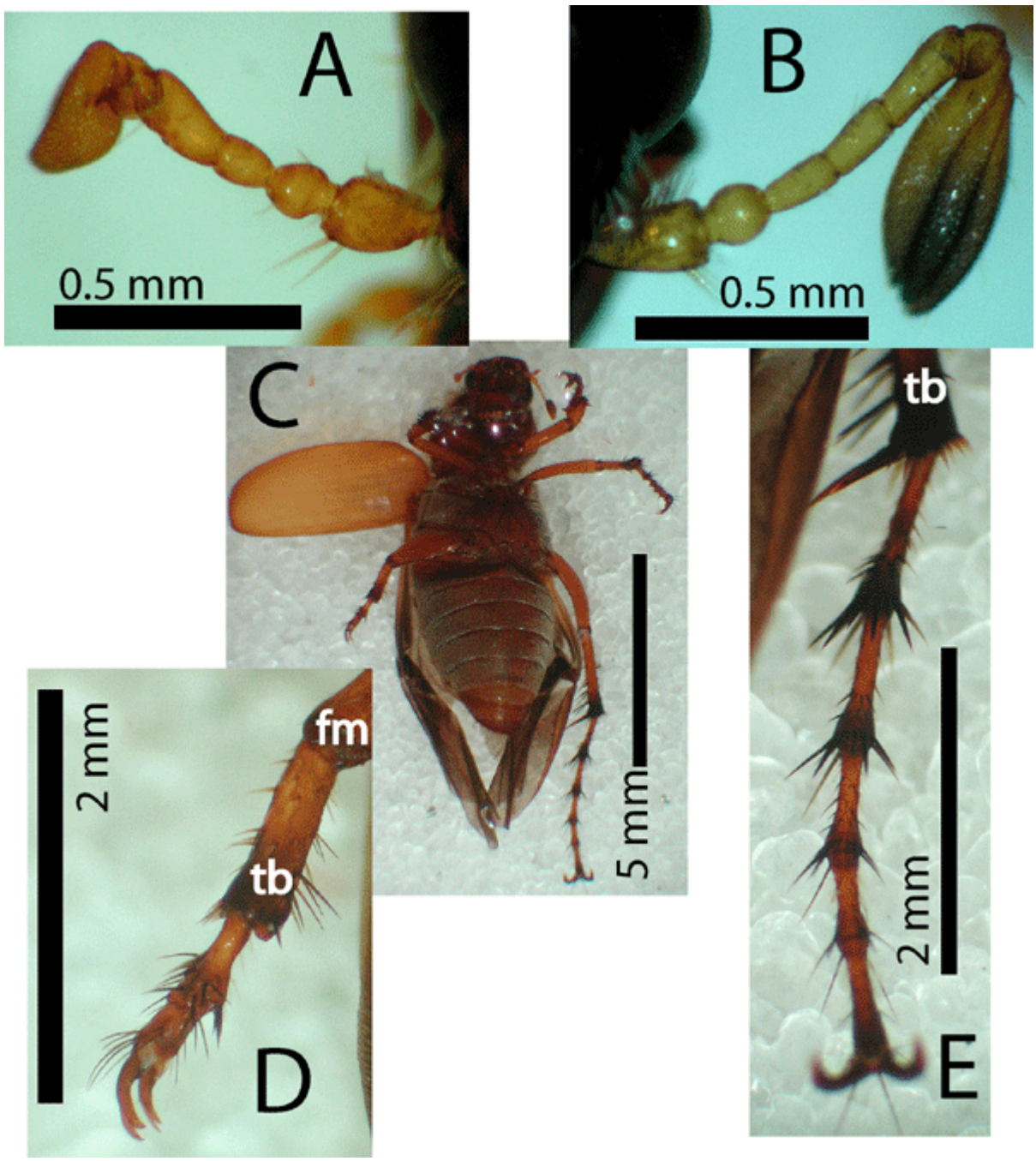


Figure 67. Scarab beetle with teratological anomalies of right antenna and right rear leg. A. reduced right antenna, B. normal left antenna, C. whole animal – abnormal leg and antenna are visible, D. reduced right rear leg (fm=femur, tb=tibia), E. tarsus of normal left rear leg.

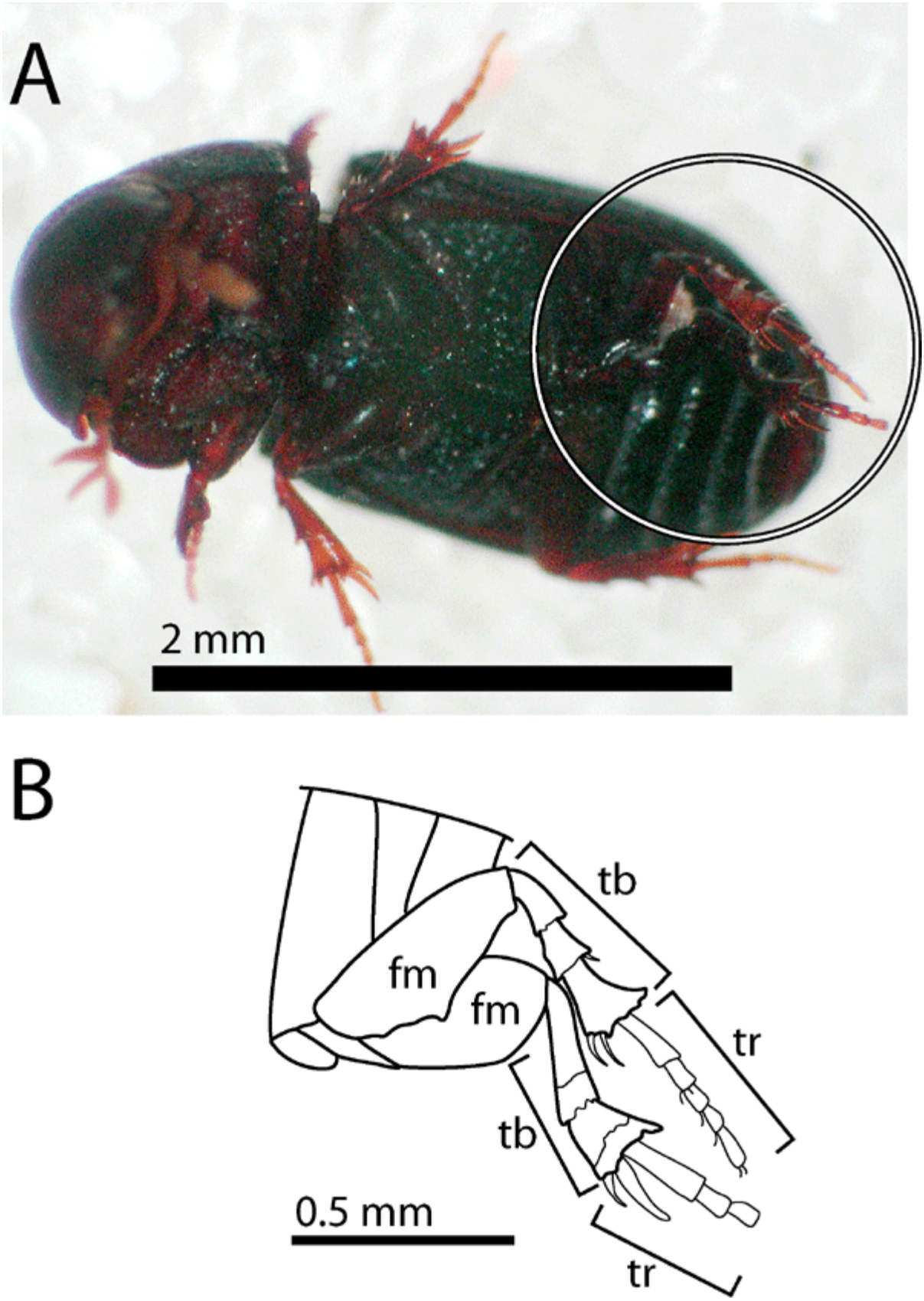


Figure 68. Sacarb beetle from pitfall trap at site D2 with two left rear legs. A. Photograph of ventral view of the beetle, leg anomaly circled. B. Line drawing of leg anomaly: fm-femur, tb-tibia, tr-tarsus.

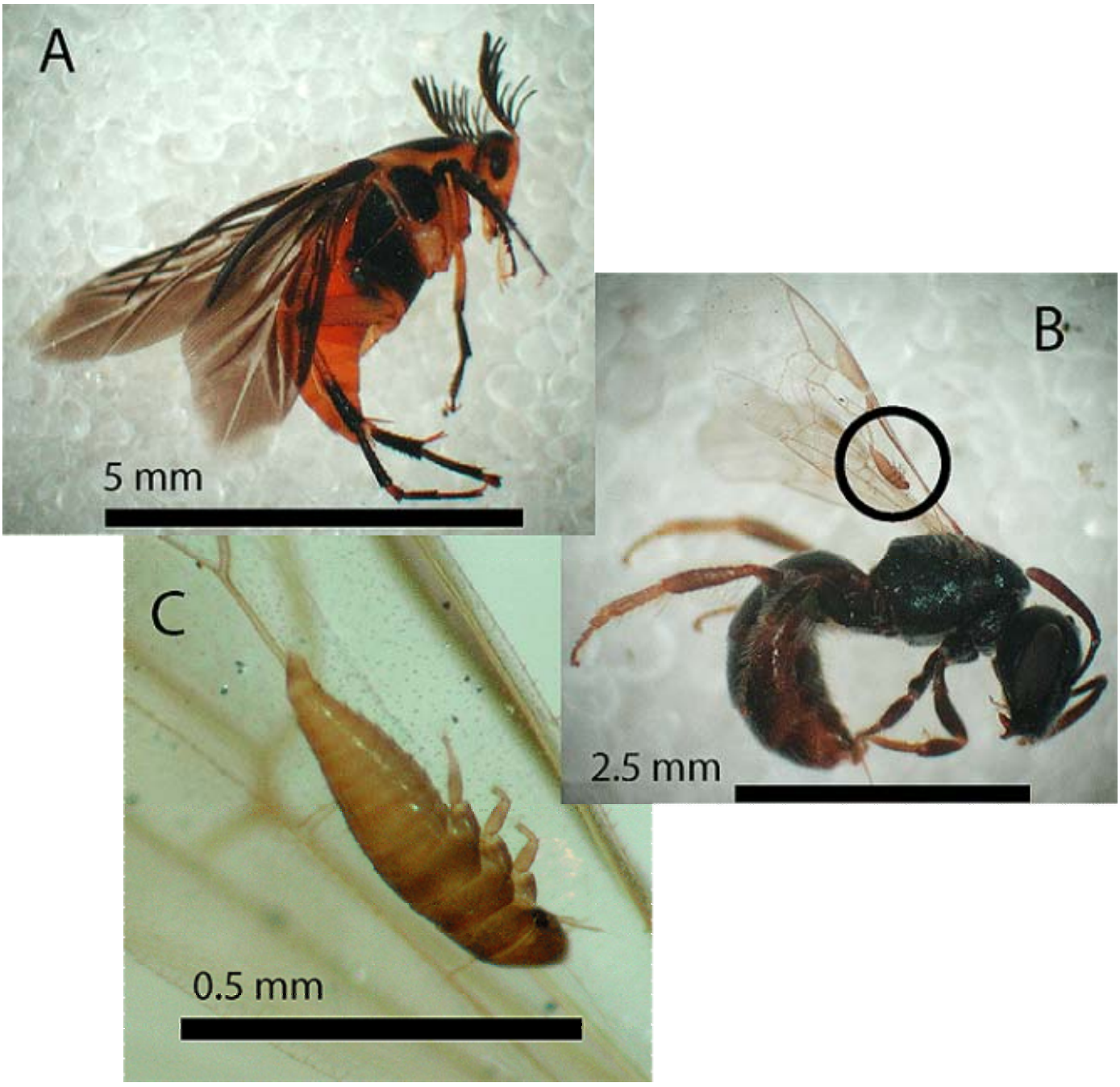


Figure 69. Rhipiphorid beetles from site D1 sweep samples. A. Adult male, B. Halictid bee with larval rhipiphorid on wing (in circle), C. Close-up of larval rhipophorid on bee's wing.

Usefulness of Ants and Homoptera as indicators of overall invertebrate community structure

While leaflitter depth was a good indicator of richness and diversity of invertebrates in litter sample (Figure 70c,d), the Homoptera and Formicidae in these samples appear to differ in their value as indicator taxa. The richness and diversity of ants is fairly well correlated with overall diversity and richness (Figure 70k,l), whereas the richness and diversity of Homoptera in leaflitter provided little information about overall invertebrate diversity in these samples (Figure 70h,i). For pitfall samples, the percentage ground cover that was grasses or herbaceous vegetation (Figure 71b) was positively correlated with the overall species diversity, while percentage ground cover that was leaflitter (Figure 71d) and the percentage canopy cover (Figure 71a) were negatively correlated with overall species diversity. Diversity and richness of Homoptera in pitfall samples were poor indicators of overall richness and diversity (Figure 71e,f), while diversity and richness of Formicidae (Figure 71j,k) were better indicators of overall richness and diversity, although when overall diversity was low, ant diversity was not a useful indicator of diversity (Figure 71k). For sweep samples, Homopteran species richness was a good indicator of overall richness in samples (Figure 72a), and the same is generally true for species diversity (Figure 72b), although Homopteran species diversity was not a useful predictor of overall diversity when diversity was low (Figure 72b). Formicidae species richness in sweep samples (Figure 72c) was positively correlated with overall species richness. While data are incomplete for Malaise samples, the abundance of Homoptera in these samples (Table 7) suggests that they would serve as good indicators of overall diversity and richness in Malaise samples.

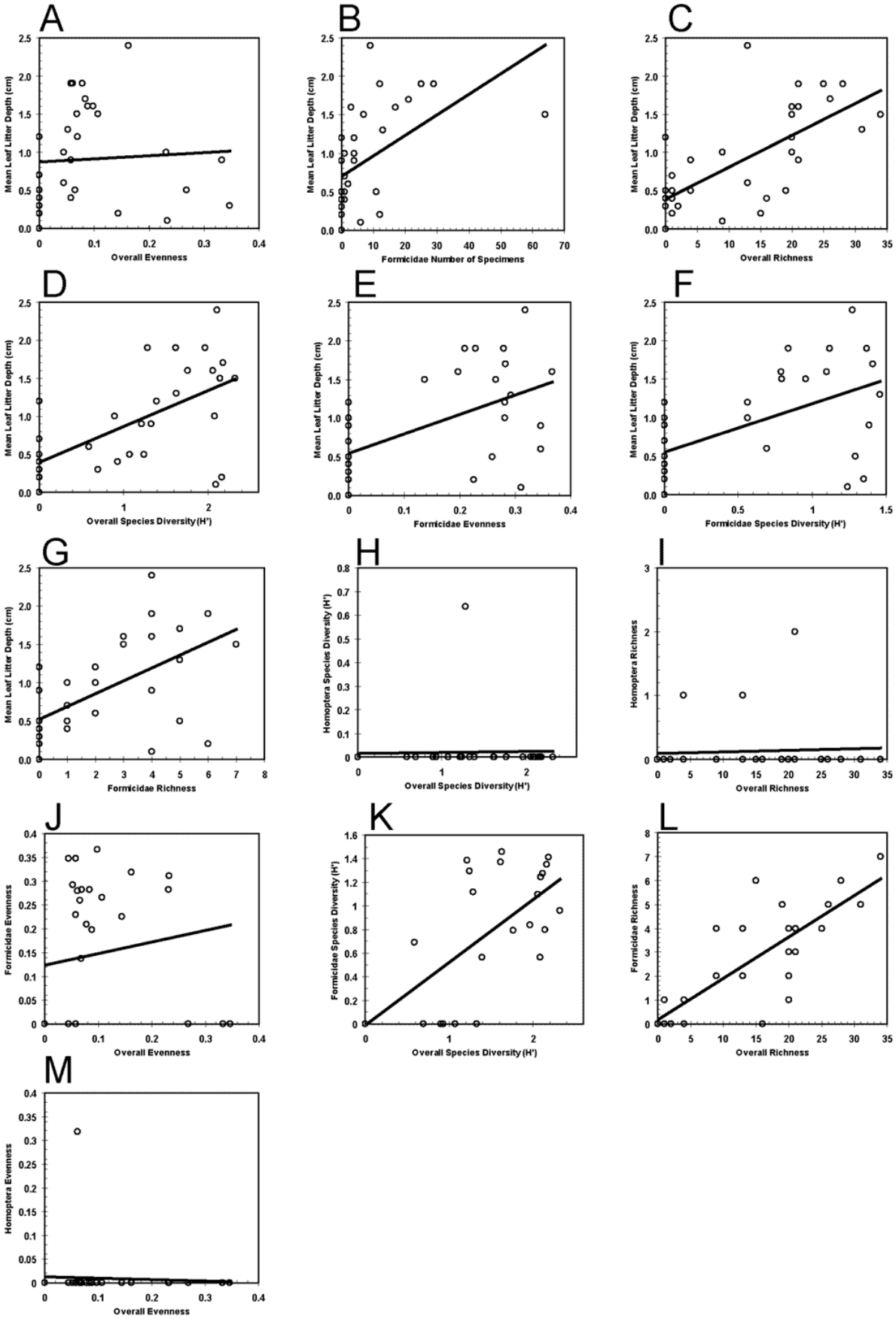


Figure 70. Leaf litter sample data. Comparison of richness, evenness and diversity for overall invertebrate taxa, ants (Formicidae), and homoptera (including Aphididae), and relationships of these metrics to leaf litter depth. Based on 4 leaf litter samples from each of 8 sample sites at Camp Atterbury, Indiana, July 2004.

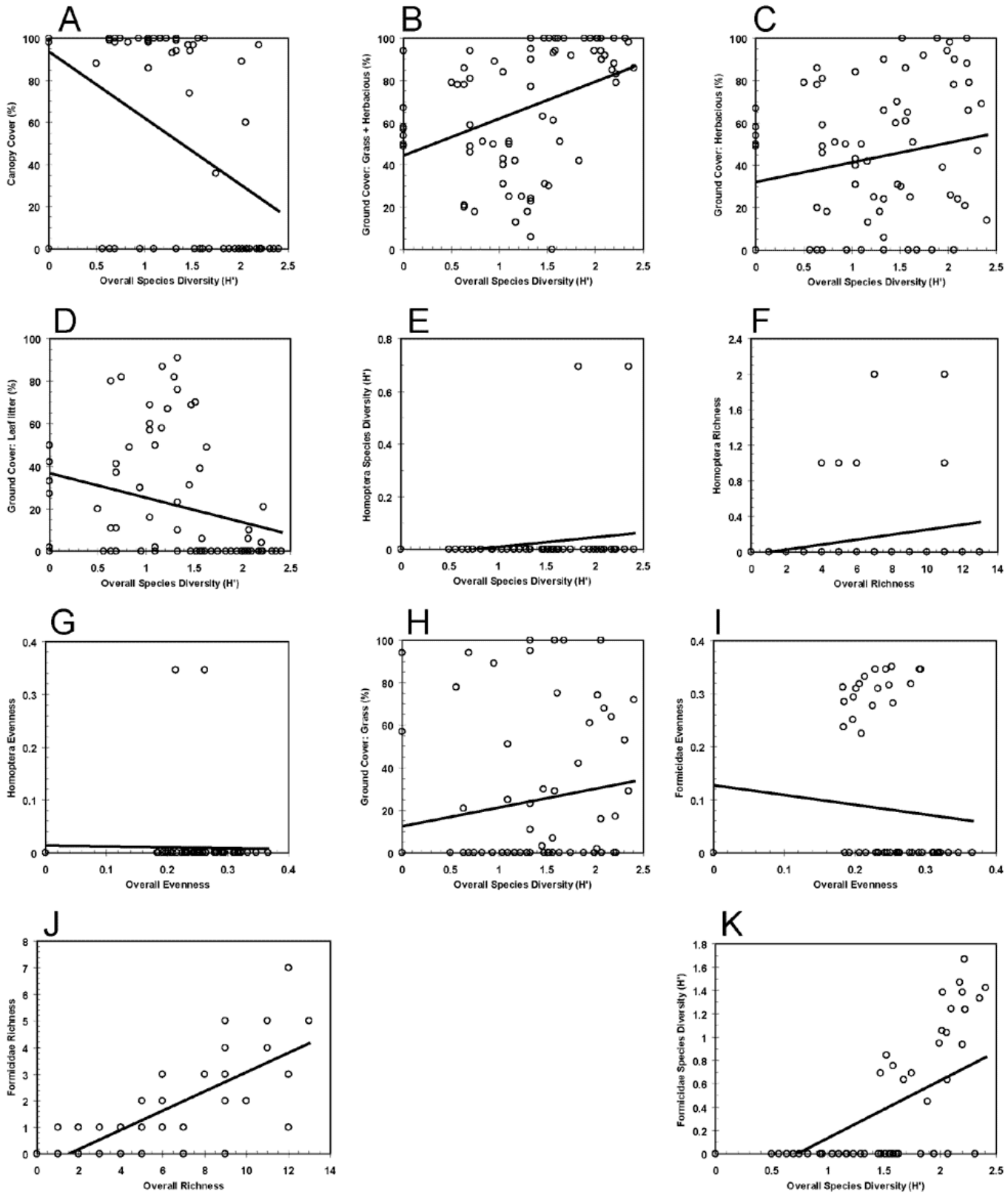


Figure 71. Pitfall trap sample data. Comparison of richness, evenness and diversity for overall invertebrate taxa, ants (Formicidae), and homoptera (including Aphididae), and relationships of these metrics to selected ground cover and canopy cover metrics. Based on 9 pitfall trap samples from each of 8 sample sites at Camp Atterbury, Indiana, July 2004.

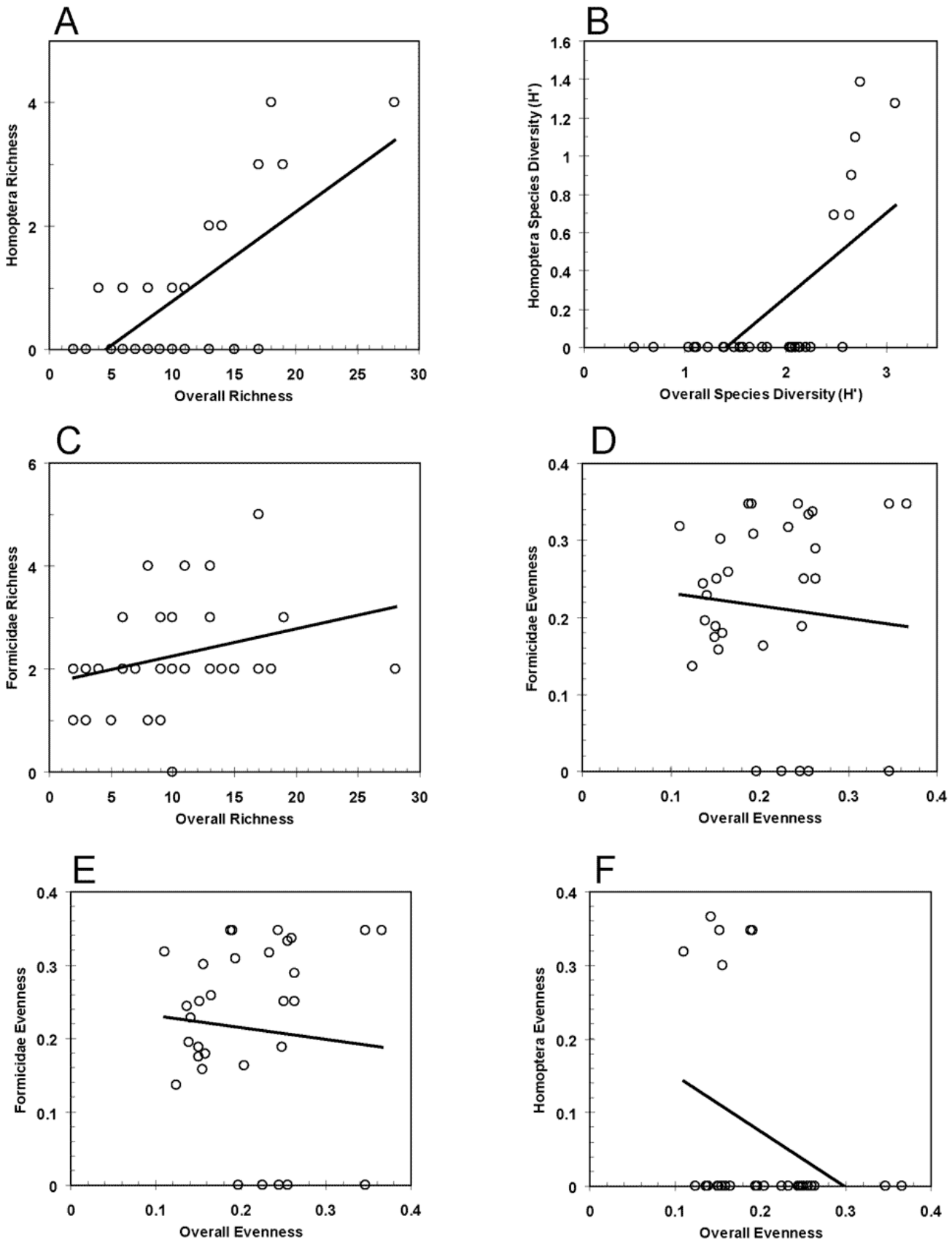


Figure 72. Sweep sample data. Comparison of richness, evenness and diversity for overall invertebrate taxa, ants (Formicidae), and homoptera (including Aphididae). Based on 4 sweep samples from each of 8 sample sites at Camp Atterbury, Indiana, July 2004.

Aquatic Sites

Aquatic samples, and associated water quality data, from 24 November 2003 and 1 April 2004 aquatic collection trips were the responsibility of a graduate student who dropped out of school, and although data were provided by the student to the PI of this project, the data are unintelligible and hopelessly disorganized and thus are not included in the results of this report. The aquatic data summarized herein come almost exclusively from July 27-28, 2004 collections. During the 11-15 October, 2004 field visit, all three streams were completely dry, thus no aquatic sampling was conducted during the fall.

Water quality parameters

Water temperature, as measured by the YSI meter, varied among sites and within site across sample periods, but no pattern was discernable (Table 8). Dissolved oxygen was generally higher when water was flowing (2 April 2004) than when water was moving slowly or restricted to isolated pools (27-28 July 2004) (Table 8). The pH was generally highest at site 1 and lowest at site 3, but the data are not sufficient to determine whether or not these differences are significant (Table 8). Turbidity was lower at each site when streams were flowing (2 April 2004) than when water was moving slowly or restricted to isolated pools (27-28 July 2004), and there appeared to be generally higher turbidity at site 1 than at the other two sites, but the data are insufficient to determine whether or not these differences are significant (Table 8). Specific conductivity was variable within and among sites, but no trend is apparent in the limited data (Table 8). Measurable flow was present in November 2003 and April 2004, and the highest volume of flow was at site 3, with the lowest volume of flow at site 1 (Table 8). In July 2004, water at site 3 was reduced to isolated pools with no flow, and by October 2004, all three streams were completely dry.

In-stream temperature was monitored hourly using a Hobo Tidbit data logger (Figure 73). These data showed similar trends for all three streams (Figure 73), with water temperatures generally cooling from an early July average of around 24 °C to 12-14 °C by mid October (Figure 73), with Stream 1 having temperatures 0.5 to 2.0 °C cooler than the other two streams. These data, however, are less complete than they appear. When the data were recovered from the data loggers in mid October, all three streams were found to be completely dry (Figure 74). A closer evaluation of the logged data (Figure 75), shows that through much of July, daily water temperatures fluctuated on a daily basis , but that near the end of July these fluctuations were somewhat reduced. It may be that the reduction in amplitude of daily fluctuations corresponds to a point where the stream ceased flowing and the logger began to record only fluctuations in pooled water. About 8-15 days into September (Figure 75) a second change becomes apparent, when the amplitude of daily fluctuations increases to a level exceeding that range found in early July, when flowing water was present. It seems plausible that this second change corresponds to the point when the streams went completely dry – thus the data loggers began to record air temperatures instead of water temperatures. The single air/humidity data logger in place at Stream 2 recorded fluctuations in temperature and humidity (Figure 76) comparable to those recorded by the upland forest air/humidity logger (Figures 25, 27). The range of temperatures was also comparable to stream temperature data recorded by the Hobo tidbit data loggers (Figure 73).

Aquatic Macroinvertebrates

We obtained 818 organisms in the stream samples collected on 27-28 July, 2004 (Table 9). Most of the taxa were typical of small, intermittent, rocky bottomed streams, though some taxa (e.g., Formicidae, Histeridae) are clearly accidentals. Others, including crayfish (Cambaridae), isopods (Asellidae), certain caddisflies (*Cheumatopsyche* sp.), and several groups of beetles (Dryopidae, Elmidae) and flies (Ceratopogonidae, Chironomidae, Tipulidae) are typical of small rocky-bottom streams. Much of the stream fauna must be able to survive periods of drying by living in the hyporheos (water filled spaces between graves in the stream bed) during annual stream drying. When individual sample values (Table 10) were averaged within each site (Table 11), the water quality was classified as “Good” (Some organic pollution probable) at site 1 and “Fair” (Fairly substantial pollution likely) at sites 2 and 3, based on Hilsenhoff’s (1988) family-level index of biotic integrity. These scores are typical for small Midwestern streams. Robinson (2004) used the genus-level Hilsenhoff Biotic Index to assess multiple streams at Camp Atterbury, and found values ranging from “good” to “poor” – comparisons between Robison’s (2004) data and ours are not possible because he used a higher level of taxonomic resolution, and generally sampled larger, more permanent streams. While site 1 scored better on the family-level index of biotic integrity than the other two sites, family and taxon richness were notably lower at Site 1 than at sites 2 or 3. The small number of sites and sample periods precludes rigorous statistical analysis, and the aquatic data fail to provide much meaningful information regarding the effects of establishment of the MPTR at Camp Atterbury.

Table 8. Water quality data collected at the three stream sites near the MPTR at Camp Atterbury, Indiana. See map (Figure 7) for specific locations.

Stream Site	Date	Water Temperature (° C)	Dissolved Oxygen (mg/L or % saturation)	pH	Turbidity (FTU)	Specific Conductivity (mS/cm)	Volume of Flow (gpm)
1	2Apr04	11.70	100.7%sat	7.41	13.26	0.158	84.19
	28Jul04	15.46	65.3 %sat	7.10	20.86	0.263	0.00[1]
	12Oct04						0.00[2]
2	3Nov03	15.6	9.28 mg/L				96.39
	2Apr04	12.21	97.2 %sat	7.05	10.64	0.264	154.07
	27Jul04	17.52	77.4 %sat	7.30	5.83	0.437	0.00[1]
	12Oct04						0.00[2]
3	3Nov03	16.5	7.76 mg/L				154.41
	1Apr04	9.93	96.6 %sat	6.55	8.33	0.209	323.89
	28Jul04	16.36	52.4 %sat	6.37	13.83	0.176	0.00[3]
	12Oct04						0.00[2]

[1] Flowing water present, but rate of flow too low to register on flow meter
[2] No water in stream
[3] Water reduced to isolated pools, no discernable flow

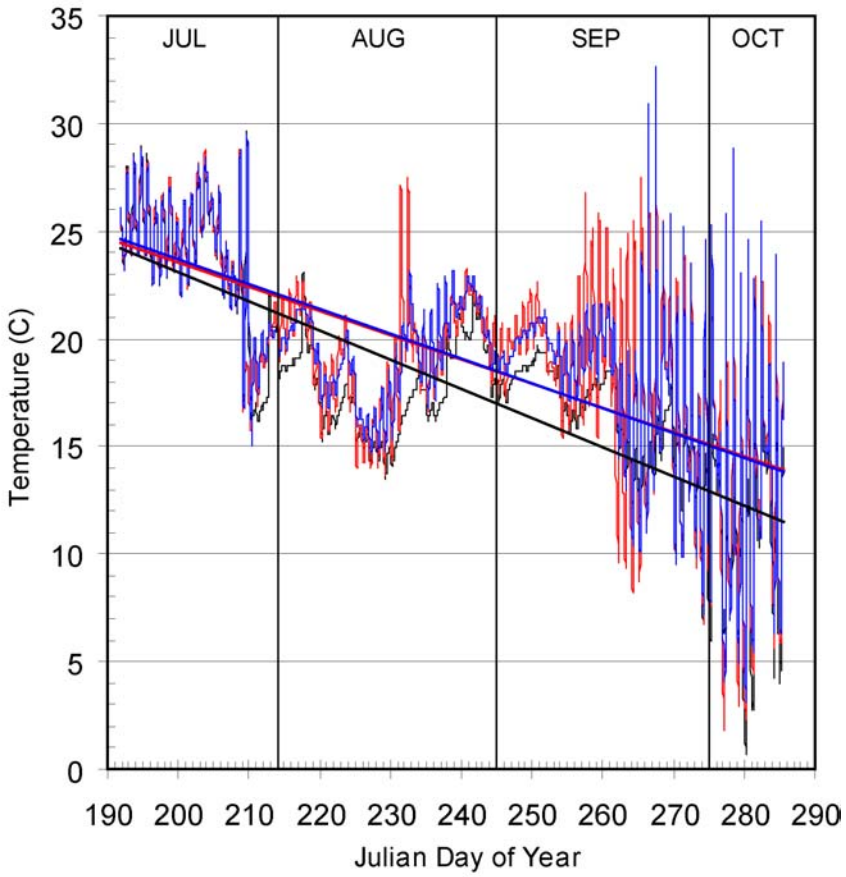


Figure 73. Data recorded by stream temperature loggers at Stream 1 (black line), Stream 2 (red line) and Stream 3 (blue line). Data are Hobo Tidbit ® hourly temperatures. Straight lines are best fit linear regressions, color corresponding to stream. Data presented span the approximately three month period from 9 July to 11 October, 2004.

A

B

C

Figure 74. Stream beds at study sites 10-15 October, 2004. A. Stream 1, B. Stream 2 (note Hobo Tidbit data logger at tip of blue arrow), C. Stream 3. Compare to Figures 3,4.

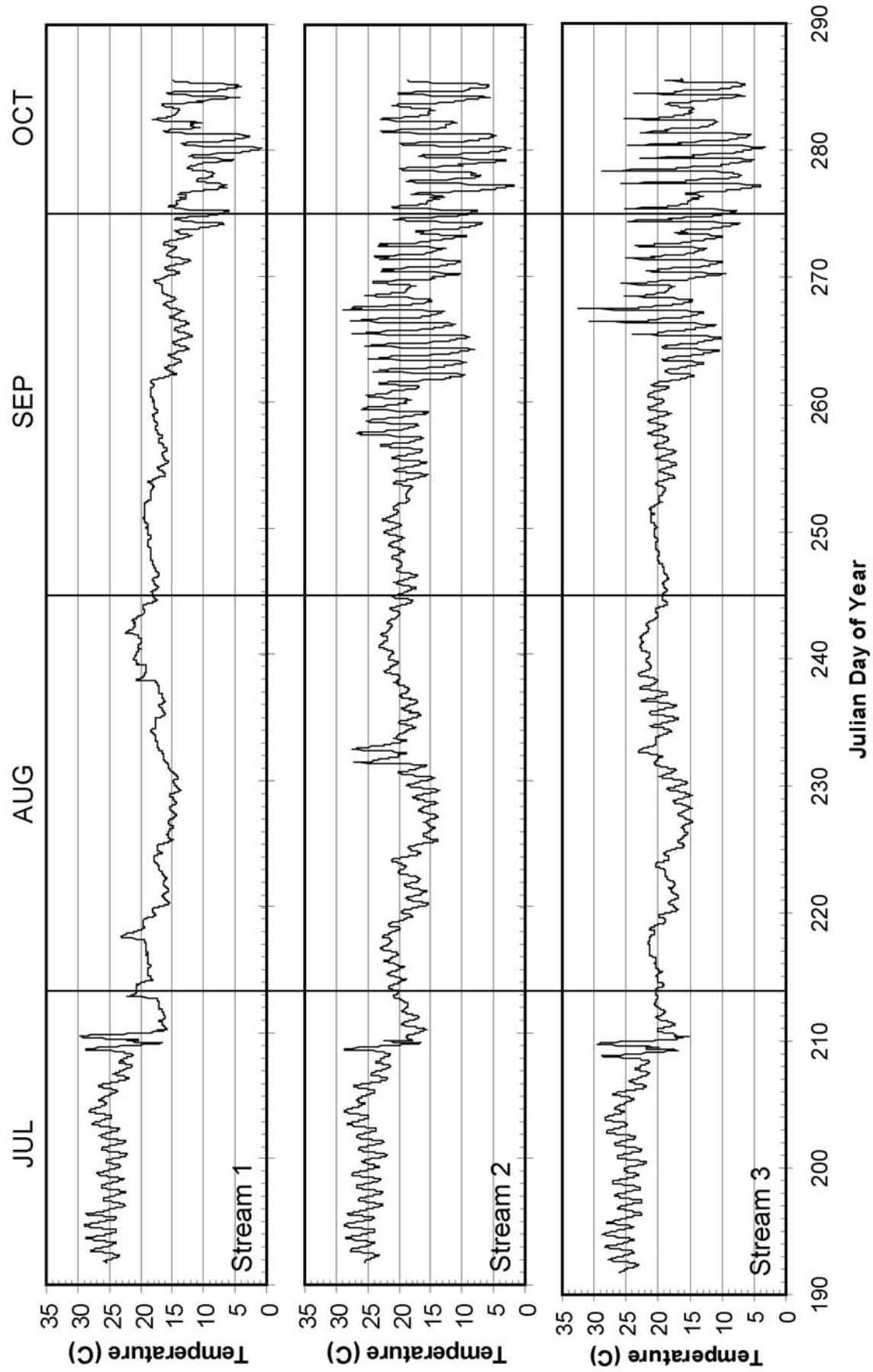


Figure 75. Data recorded by stream temperature loggers at stream sites. Data are Hobo Tidbit ® hourly temperatures. Data presented span the approximately three month period from 9 July to 11 October, 2004.

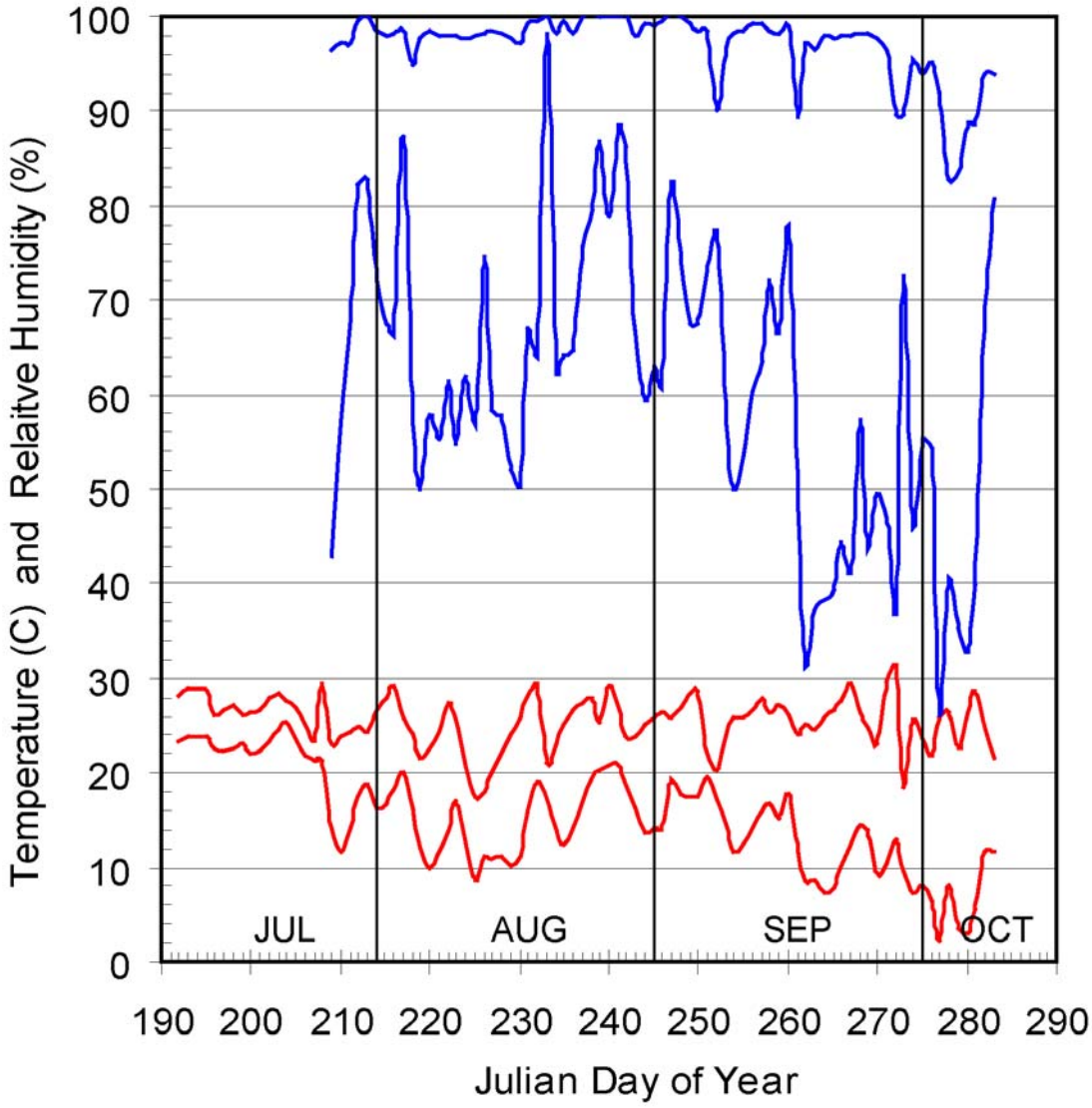


Figure 76. Daily minima and maxima for air temperature (red lines) and relative humidity (blue lines), derived from Hobo Temp/RH Pro ® hourly humidity and temperature data logger at stream site 2. Data presented span the approximately three month period from 10 July to 9 October, 2004, questionable humidity data (readings out of normal range) from portions of July have been excluded.

Table 9. Summary of 818 aquatic specimens collected from three streams at Camp Atterbury, Indiana on July 27-28, 2004.

Phylum:Class Order	Family	Taxon	Stream 1	Stream 2	Stream 3
Mollusca:Gastropoda			1	1	0
Annelida:Oligochaeta			0	9	0
Arthropoda:Arachnida			4	10	1
Arthropoda:Crustacea					
Decapoda					
	Cambaridae		10	23	44
Isopoda					
	Asellidae		8	22	2
Arthropoda:Insecta					
Collembola			5	2	1
Thysanoptera					
	Phlaeothripidae		1	0	0
Megaloptera					
	Sialidae	*Sialis* sp.	10	4	3
Odonata					
	Aeshnidae	*Boyeria* sp.	0	0	2
	Calopterygidae	*Hetaerina* sp.	0	4	3
	Corduliidae		0	1	12
	Undetermined		0	1	0
Ephemeroptera					
	Baetidae		8	6	4
	Caenidae		0	2	0
	Heptageniidae	*Stenonema* sp.	62	33	3
	Undetermined		0	6	0
Hemiptera					
	Gerridae	*Aquarius* sp.	2	6	3
		Undetermined	4	1	3
	Hydrometridae	*Hydrometra* sp.	0	0	3
	Notonectidae	*Notonecta* sp.	0	0	1
	Veliidae	*Microvelia* sp.	0	2	0
		Undetermined	0	2	0
	Undetermined		0	0	3
Homoptera					
	Cicadellidae		0	0	1

Continued on next page

Table 9. Continued.

Phylum:Class Order	Family	Taxon	Stream 1	Stream 2	Stream 3
Trichoptera					
	Hydropsychidae	*Cheumatopsyche* sp.	0	18	0
		Undetermined	0	6	0
	Undetermined		0	2	1
Coleoptera					
	Dryopidae	*Helichus* sp.	0	3	10
		Undetermined	1	14	0
	Dytiscidae	*Hydroporus* sp.	1	2	3
		Undetermined	0	2	0
	Elmidae	*Stenelmis* sp.	0	1	0
		Undetermined	0	7	0
	Histeridae		1	0	0
	Hydrophilidae		0	0	1
	Lampyridae		2	0	0
	Psephenidae	*Psephenus* sp.	0	10	0
		Undetermined	1	13	0
	Staphylinidae		3	2	1
	Undetermined		1	1	0
Diptera					
	Ceratopogonidae		1	11	2
	Chironomidae		26	82	41
	Culicidae		0	1	1
	Dixidae		9	21	8
	Simuliidae		0	0	2
	Syrphidae		0	0	1
	Tabanidae		0	9	3
	Tipulidae		0	6	0
	Undetermined		3	9	3
Hymenoptera					
	Formicidae		0	1	0
	Undetermined		3	1	0
Chordata:					
Caudata					
	Plethodontidae				
		Eurycea cirrigera (Two-lined Salamander)	3	5	3
Anura			0	0	1
Osteichthyes			5	60	54
		TOTALS	175	420	223

Table 10. Summary of aquatic sample data, by site, habitat and replicate, with Family-Level Index of Biotic Integrity (IBI). Based on July 2004 sampling.

Stream	habitat	rep	Family IBI	Taxon Richness	Family Richness
1	bank	2	5.8000	17	13
1	bank	3	4.0000	9	9
1	riffle	2	4.0588	5	5
1	riffle	3	5.8000	11	10
2	bank	1	5.5000	18	15
2	bank	2	4.8571	15	11
2	bank	3	5.3333	8	8
2	riffle	1	4.9250	19	14
2	riffle	2	5.1605	30	19
2	riffle	3	6.0000	17	15
3	bank	1	5.0000	12	11
3	bank	2	5.5000	17	15
3	bank	3	5.5500	29	22

Table 11. Summary of Family-Level Index of Biotic Integrity (IBI) and richness metrics in relation to canopy cover at each site. Based on July 2004 sampling.

Site	Family Level IBI	Water Quality[1]	Taxon Richness	Family Richness	Percent Canopy Cover
1	4.915	Good	10.5	9.0	57.67
2	5.296	Fair	17.83	14.0	83.07
3	5.35	Fair	19.33	16.0	87.90

[1]Cutoff points for Hilsenhoff's (1988) family level biotic index are: 0.00-3.75, Excellent, Organic pollution unlikely; 3.76-4.25, Very good, Possible slight organic pollution; 4.26-5.00, Good, Some organic pollution probable; 5.01-5.75, Fair, Fairly substantial pollution likely; 5.76-6.50, Fairly poor, Substantial pollution likely; 6.51-7.25, Poor, Very substantial pollution likely; 7.26-10.00, Very poor, Severe organic pollution likely.

Discussion and Recommendations

While it is obviously not practical to take into account each species individually when developing protocols for management of military lands, community-wide surveys such as ours can broadly examine anthropogenic habitat alteration and its affects on biodiversity. Hughes et al. (2000) found that the richness was correlated with habitat type, a finding corroborated by our study. The upland terrestrial protocol could be refined and simplified by only focusing on target taxa (e.g., ants and Homoptera) while retaining the same invertebrate sampling protocol. Our data suggest that the Formicidae (especially for leaf litter and pitfall samples), and to a lesser extent Homoptera (for sweep and perhaps Malaise samples), can function as useful indicator taxa reflecting the overall structure of the community. Such a simplified protocol will achieve its greatest effectiveness if it is implemented for several years (3-5 years, ideally 10 or more), allowing documentation of possible changes in community structure over time, and also allowing yearly fluctuations in community structure to not have an undue influence over the results.

Monitoring aquatic sites adjacent to the MPTR is probably best done in late spring or early summer, when water is still present in these small, intermittent streams. A family-level IBI at three stream sites provides insufficient information for meaningful analysis, but continued sampling over several years may yield a larger dataset wherein trends may be detected, and thus these data may have value for longer-term monitoring. The list of aquatic taxa generated by our study (Table 9) is consistent with the taxon list of Robinson (2004), although it provides significantly less taxonomic resolution than Robinson's study. The Two-lined Salamander, *Eurycea cirrigera*, is relatively abundant in these streams, and as a top-level predator, it may be prone to bioaccumulation of some contaminants which could be present. We suggest a tissue bioassay study of a few salamanders from each of the three stream sites, and perhaps other streams at Camp Atterbury, which could be carried out every few years to monitor for potential bioaccumulated pollutants.

Summary

We evaluated the hypothesis that habitat degradation alters the abundance and diversity of aquatic and terrestrial arthropods (which in turn has consequences for plant and vertebrate animal communities), and found obvious changes in terrestrial arthropod communities. We also examined habitat characteristics that are correlated with arthropod communities to quantify a baseline for future efforts aimed at managing or restoring aquatic and terrestrial habitats and communities at Camp Atterbury. Our study design measured both within and among plot variation in two aquatic (riffle, bank) and four terrestrial (flying, leaf litter, vegetative and ground dwelling) ecotypes. These data provide a robust index of invertebrate responses to disturbance. While the clearing of the MPTR appears to have negatively affected the abundance and diversity of some groups, especially leaf litter dwelling taxa, other taxa are more abundant and diverse in the cleared area. These data can be used to facilitate future monitoring, management and restoration efforts.

Acknowledgments

We thank Rob Meyers, Carla Guthrie and Jean Krejca (all Zara Environmental, Buda, Texas) for help with aquatic insect identification. Nick Dolce, Erin Raboin, Courtney McCusker (all University of Illinois), Rob Meyers (Zara Environmental) and Vanessa R. Block (Illinois Natural History Survey) put in long days helping complete the field work for this study. JoAnn Jacoby (University of Illinois) helped with Winkler sample extraction and equipment maintenence. Amélie Perin (University of Illinois) did most of the data entry for the project. Bill Handel (Illinois Natural History Survey) identified vouchers of dominant plant species. We thank Dr. David B. Orr (Department of Entomology, North Carolina State University, Raleigh, NC) for useful discussions on the parasitic Hymenoptera.

Literature Cited

Agosti, D., J. D. Majer, L. E. Alonso, and T. R. Schultz (eds.). 2000. Ants: standard methods for measuring and monitoring biodiversity. Smithsonian Institution Press, Washington, DC. 280 pp.

Aksnes, D. L. and J. Giske. 1993. A theoretical model of aquatic visual feeding. Ecological Modeling 67:233-250.

Anders, A. D. and D. C. Dearborn. 2004. Population trends of the endangered Golden-cheeked Warbler at Fort Hood, Texas, from 1992-2001. The Southwestern Naturalist 49(1):39-47.

Andersen, A. N. and G. P. Sparling. 1997. Ants as indicators of restoration success: relationship with soil microbial biomass in Australian seasonal tropics. Restoration Ecology 5(2):109-114.

Ankley, G. T., S. J. Degitz, S. A. Diamond and J. E. Tietge. 2004. Assessment of environmental stressors potentially responsible for malformations in North American anuran amphibians. Ecotoxicology and Environmental Safety 58(1):7-16.

Atkinson, I. A. E. 1988. Presidential address: opportunities for ecological restoration. New Zealand Journal of Ecology 11:1-12.

Austin, Jay E. and Carl E. Bruch (eds.). 2000. The environmental consequences of war: legal, economic, and scientific perspectives. Cambridge University Press, New York. 691 pp.

Barbour, M. T., J. Gerritsen, B. D. Snyder, and J. B. Stribling. 1999. Rapid bioassessment protocols for use in wadeable streams and rivers: periphyton, benthic macroinvertebrates and fish, 2nd ed. EPA 841-B-99-002. U.S. Environmental Protection Agency, Office of Water, Washington, DC.

Bestelmeyer, B. T. and J. A. Wiens. 1996. The effects of land use on the structure of ground-foraging ant communities in the Argentine Chaco. Ecological Applications 6(4):1225-1240.

Bestelmeyer, B. T., D. Agosti, L. E. Alonso, C. R. F. Brandão, W. L. Brown, Jr., J. H. C. Delabie, and R. Silvestre. 2000. Field techniques for the study of ground-dwelling ants. Pages 122-144 *in:* Agosti, D., J. D. Majer, L. E. Alonso, and T. R. Schultz (eds). Ants: standard methods for measuring and monitoring biodiversity. Smithsonian Intitution Press, Washington, DC. 280 pp.

Borror, D. J., C. A. Triplehorn, and N. F. Johnson. 1989. Introduction to the Study of Insects (6th ed.). International Thomson Publ., 800 pp.

Burke, E. and H. Goulet. 1998. Landscape and area effects on beetle assemblages in Ontario. Ecography 21:472-479.

Carvell, C. 2002. Habitat use and conservation of bumblebees (*Bombus* spp.) under different grassland management regimes. Biological Conservation 103:33-49.

Chao, A. 1984. Non-parametric estimation of the number of classes in a population. Scandanavian Journal of Statistics 11:265-270.

Chao, A. 1987. Estimating the population size for capture-recapture data with unequal catchability. Biometrics 43:783-791.

Chazdon, R. L. et al. 1998. Statistical method for estimating species richness of woody regeneration in primary and secondary rain forests of NE Costa Rica. In: Dallmeier, F. and J. A. Comiskey (eds.). Forest biodiversity research, monitoring and modeling: conceptual background and Old World case studies. Parthenon Publishing, Paris, pp 285-309.

Colwell, R. K. 2004. EstimateS: Statistical estimation of species richness and shared species from samples. Version 7. User's Guide and application published at: http://purl.oclc.org/estimates.

Creighton, W. S. 1950. The ants of North America. Bulletin of the Museum of Comparative Zoology 104.

Dale, V. H., S. C. Beyeler, and B. Jackson. 2002. Understory vegetation indicators of anthropogenic disturbance in longleaf pine forests at Fort Benning, George, USA. Ecological Indicators 1:155–170.

DeLong, D. M. 1948. The leafhoppers, or Cicadellidae, of Illinois. INHS Bulletin 24: 97-376

Disney, R. H. L. 1986. Assessments using invertebrates: posing the problem. Pages 271-293 in Usher, M. B. (ed.). Wildlife conservation evaluation. Chapman and Hall, London.

Dudley, J. P., J. R. Ginsberg, A. J. Plumptre, J. A. Hart, and L. C. Campos. 2002. Effects of war and civil strife on wildlife and wildlife habitats. Conservation Biology 16:319-329.

Ehlen, Judy and Russell S. Harmon (eds.). 2001. The environmental legacy of military operations. Geological Society of America, Reviews in Engineering Geology Volume 14. Boulder, Colorado. 228 pp.

Fang, S., S. Wente, G. Z. Gertner, G. Wang, and A. Anderson. 2002. Uncertainty analysis of predicted disturbance from off-road vehicular traffic in complex landscapes at Fort Hood. Environmental Management 30(2):199–208.

Fay, P. A. 2003. Insect Diversity in Two Burned and Grazed Grasslands. Environmental Entomology 32(5):1099-1104.

Gibson, C. W. D., H. C. Dawkins, V. K. Brown and M. Jepson. 1987. Spring grazing by sheep: effects on seasonal changes during old field succession. Vegetatio 70:33-43.

Goldsmith, B. (ed.). 1991. Monitoring for Conservation and Ecology. Chapman & Hall, London. 275 pp.

Gotelli, N. J. and R. K. Colwell. 2001. Quantifying biodiversity: procedures and pitfalls in the measurement and comparison of species richness. Ecology Letters 4:379-391.

Goulet, H., and J.T. Huber (eds.). 1993. Hymenoptera of the world: An identification guide to families. Canada Communications Group-Publishing, Ottawa, Canada. 668 pp.

Greenslade, P. J. M. 1973. Sampling ants with pitfall traps: Digging-in effects. Insectes Sociaux 20:343-353.

Greenslade, P. J. M. and P. Greenslade. 1984. Invertebrates and environmental assessment. Environment and Planning 3:13-15.

Hecht, T. and van der Lingen, C. D. 1992. Turbidity induced changes in the foraging strategies in estuaries. S. Afr. J. Zool. 27:95-107.

Hellawell, J. M. 1978. Biological surveillance of rivers: a biological monitoring handbook. Water Research Centre, Stevenage, United Kingdom.

Heltsche, J. F. and Forrester, N. E. 1983. Estimating species richness using the jackknife procedure. Biometrics 39:1-11.

Hendrix, S. D., V. K. Brown, and H. Dingle. 1988. Arthropod guild structure during early old field succession in a New World and Old World site. Journal of Animal Ecology 18:301-310.

Hilsenhoff, W. L. 1987. An improved bioindicator of organic stream pollution. Great Lakes Entomologist 20:31-39.

Hilsenhoff, W. L. 1988. Rapid field assessment of organic pollution with a family-level biotic index. Journal of the North American Benthological Society 7:65-68.

Hirose, Y. 1994. Determination of species richness and composition in egg parasitoid assemblages of Lepidoptera. Pages 19-29 *in*: Hawkins, B.A., and W. Sheehan (eds.). Parasitoid community ecology. Oxford University Press Inc., New York. 516 pp.

Hughes, J. B., G. C. Daily, P. R. Ehrlich. 2000. Conservation of insect diversity: a habitat approach. Conservation Biology 14(6):1788-1797.

Human, K. G., and D. M. Gordon. 1996. Exploitation and interference competition between the invasive Argentine ant, *Linepithema humile*, and native ant species. Oecologia 105:405–412.

Jackson, L. L., N. Lopoukhine, and D. Hillyard. 1995. Ecological restoration: a definition and comments. Restoration Ecology 3:71-75.

James, A. and L. Evison, ed. 1979. Biological indicators of water quality. Wiley, Chichester, United Kingdom.

Jansen, A. 1997. Terrestrial invertebrate community structure as an indicator of the success of a tropical rainforest restoration project. Restoratioin Ecology 5(2):115-124.

Jonas, J. L., M. R. Whiles, and R. E. Charlton. 2002. Aboveground invertebrate responses to land management differences in a central Kansas grassland. Environmental Entomology 31(6):1142-1152.

Kim, K. C. 2993. Biodiversity, conservation and inventory: why insects matter. Biodiveristy and Conservation 2:191-214.

Knight, R. L., and M. K. Rust. 1990. The urban ants of California with distributional notes of imported species. Southwestern Entomologist 15:167–178.

Kremen, C., R. K. Colwell, T. L. Erwin, D. D. Murphy, R. F. Noss, and M. A. Sanjayan. 1993. Terrestrial arthropod assemblages: their use in conservation planning. Conservation Biology 7:796-808.

Landres, P. B., J. Verner, and J. W. Thomas. 1988. Ecological uses of vertebrate indicator species: a critique. Conservation Biology 2:316-328.

Lanier-Graham, Susan D. 1993. The Ecology of War: Environmental Impacts of Weaponry and Warfare. Walker S. Co., New York. 185 pp.

Lee, S.-M. and A. Chao. 1994. Estimating population size via sample coverage for closed capture-recapture models. Biometrics 50:88-97.

Lehman, R. N., K. Steenhof, M. N. Kochert, and L. B. Carpenter. 1999. Effects of military training activities on shrub-steppe raptors in southwestern Idaho. Environmental Management 23:409-417.

Leponce, M. L. Theunis, J. H. C. Delabie and Y. Roisin. 2004. Scale dependence of diversity measures in a leaf-litter ant assemblage. Ecography 27:253-267.

Levy, E. B. and E. A. Madden. 1933. The point method of pasture analysis. New Zealand Journal of Agriculture 46:267-279.

Longcore, T. 2003. Terrestrial arthropods as indicators of ecological restoration success in coastal sage scrub (California, U.S.A.). Restoration Ecology 11(4):397–409.

Longino, J. T. and R. K. Colwell. 1997. Biodiversity assessment using structured inventory: Capturing the ant fauna of lowland tropical rainforest. Ecological Applications 7:1263-1277.

Longino, J. T., J. Coddington, and R. K. Colwell. 2002. The ant fauna of a tropical rain forest: estimating species richness three different ways. Ecology 689-702.

Ludwig, J. A. and J. F. Reynolds. 1988. Statistical Ecology: a primer on methods and computing. John Wiley and Sons, New York.

MacKay, W.P. 2000. Review of the new world ants of the subgenus *Myrafant* (Genus *Leptothorax*). Sociobioogy 36:265-444.

Majer, J. D. 1990. Rehabilitation of disturbed land: long-term prospects for the recolonisation of fauna. Proceedings of the Ecological Society of Australia 16:509-519.

Majer, J. D. and O. G. Nichols. 1998. Long-term recolonization patterns of ants in western Australian rehabilitated bauxite mines with reference to their use as indicators of restoration success. Journal of Applied Ecology 35:161-182.

Malott, C. A. 1922. The physiography of Indiana. *In*: Handbook of Indiana Geology, Part 2. Indiana Department of Conservation Publication 21, pp 59-256.

McKee, M. and R. P. Berrens. 2001. Balancing Army and endangered species concerns: green vs. green. Environmental Management 27(1):123-133.

Merritt, R. W. and K. W. Cummins (eds.). 1997. An Introduction to the Aquatic Insects of North America (3rd edition). Kendall/Hunt Publishing Company. 862 pp.

Migliorini, M, G. Pigino, N. Bianchi, F. Bernini, and C. Leonzio. 2004. The effects of heavy metal contamination on the soil arthropod community of a shooting range. Environmental Pollution 129(2):331-340.

Milchunas, D. G., K. A. Schulz, and R. B. Shaw. 2000. Plant community structure in relation to long-term disturbance by mechanized military maneuvers in a semiarid region. Environmental Management 25(5):525–539.

Miller, J. C. 1993. Insect natural history, multi-species interactions and biodiversity in ecosystems. Biodiversity and Conservation 2:233-241.

Nakamura, A., H. Proctor and C. P. Catterall. 2003. Using soil and litter arthropods to assess the state of rainforest restoration. Ecological Management & Restoration 4(supplement):S20-S28.

National Research Council. 1992. Restoration of aquatic ecosystems: science, technology, and public policy. National Research Council, Washington, DC.

New, T. R. 1998. Invertebrate Surveys for Conservation. Oxford University Press, Oxford. 240 pp.

Oliver, I., and A. J. Beattie. 1993. A possible method for the rapid assessment of biodiversity. Conservation Biology 7:562-568.

Olson, D. M. 1991. A comparison of the efficacy of the litter sifting and pitfall traps for sampling leaf litter ants (Hymenoptera: Formicidae) in a tropical wet forest, Costa Rica. Biotropica 23: 166-172.

Ortuño, V. M. and J. M. Hernández. 1993. Diversos casos teratológicos en Coleoptera. Bol. R. Soc. Esp. Hist. Nat. (Sec. Biol.) 89:163-179.

Osuna, E. 1992. Fenomeno teratologico en *Psalidognathus* sp. (Coleoptera: Cerambycidae). Bol. Entomol. Venez. N.S. 7(2):145-156.

Palmer, M. W. 1991. Estimating species richness: the second-order jackknife reconsidered. Ecology 72:1512-1513.

Pielou, E. C. 1984. The interpretation of ecological data: a primer on classification and ordination. John Wiley and Sons, New York.

Pogue, D. W. and G. D. Schnell. 2001. Effects of agriculture on habitat complexity in a prairie-forest ecotone in the southern Great Plains of North America. Agriculture, Ecosystems and Environment 87: 287-298.

Rieske, L. K. and L. J. Buss. 2001. Influence of site on diversity and abundance of ground- and litter-dwelling Coleoptera in Appalachian oak–hickory forests. Environmental Entomology 30(3):484-494.

Robinson, B. A. 2004. An inventory of aquatic macroinvertebrates and calculation of selected biotic indices for the U.S. Army Atterbury Reserve Forces Training Area near Edinburgh, Indiana, September 2000-August 2002. Scientific Investigations Report 2004-5010. U.S. Department of the Interior, U.S. Geological Survey. iv + 19 pages.

Rosenberg, D. M., H. V. Danks, and D. M. Lehmkuhl. 1986. Importance of insects in environmental impact assessment. Environmental Management 10:773-783.

SAS Institute. 2001. The SAS system for Windows, Release 8.2. SAS Institute, Inc., Cary, NC.

Schumaker, D.J., J. M. Fenelon, N. T. Baker, J. D. Martin, E. R. Bayless, D. V. Jacques, and C. G. Crawfored. 1999. Environmental setting and natural factors and human influences affecting water quality in the White River Basin, Indiana. Indianapolis, Indiana, Natal Water-Quality Assessment Program Water-Resources Investigations Report 97-4260. 66 pp.

Smith, D. G. 2001. Pennak's Freshwater Invertebrates of the United States: Porifera to Crustacea, 4th Edition. John Wiley & Sons. 648 pp.

Smith, M. A., M. G. Turner, and D. H. Rusch. 2002. The effect of military training activity on Eastern Lupine and the Karner Blue Butterfly at Fort McCoy, Wisconsin, USA. Environmental Management 29(1):102–115.

Snelling, R. R., and C. George. 1979. The taxonomy, distribution, and ecology of California desert ants. Report to Bureau of Land Management, United States Department of the Interior, Riverside, California, USA.

Soberón, M. J. and J. Llorente. 1993, The use of species accumulation functions for the prediction of species richness. Conservation Biology 7:480-488.

Stange, L.A. 1991. The Rhopalosomatidae of Florida. Fla. Dept. Agric. and Consumer Services, Div. of Plant Industry Entomology Circular No. 341.

Strand, M. R. 1986. The physiological interactions of parasitoids with their hosts and their influence on reproductive strategies. Pages 97-136 *in*: Waage, J., and D. Greathead (eds). Insect parasitoids. Academic Press, New York. 389 pp.

Suarez, A.V., D. T. Bolger, and T.J. Case. 1998. Effects of fragmentation and invasion on native ant communities in coastal southern California. Ecology 79: 2041-2056

Sulston, I. A. and K. V. Anderson. 1996. Embryonic patterning mutants in *Tribolium castaneum*. Development 122:805-814.

Taylor, S. J. 2001. Investigation of the potential for Red Imported Fire Ant (*Solenopsis invicta*) impacts on rare karst invertebrates at Fort Hood, Texas: Literature survey and study design. Illinois Natural History Survey, Center for Biodiversity Technical Report 2001(12):1-49.

Thorp, J. and A. Covich. 2001. Ecology and Classification of North American Freshwater Invertebrates (2nd Edition). Academic Press. 950 pp.

Townes, H. 1972. A light-weight malaise trape. Entomological News 83(9):239-247.

Townes, H., and M. Townes. 1951. Family Ichneumonidae. Pages 184-409 *in*: Muesebeck, C.F.W., Krombein, K.V., and H.K. Townes (eds). Hymenoptera north of Mexico. U.S. Department of Agriculture Monograph 2.

Tschinkel, W. R. 1987. Seasonal life history and nest architecture of a winter-active ant, *Prenolepis imparis*. Insectes Sociaux 34:143–164.

Villa-Castillo, J. and M. R. Wagner. 2002. Ground beetle (Coleoptera: Carabidae) species assemblage as an indicator of forest condition in northern Arizona ponderosa pine forests. Environmental Entomology 31(2):242-252.

Voshell, J. R., Jr. 2002. A Guide to Common Freshwater Invertebrates of North America. McDonald & Woodward Pub. Co., 456 pp.

Wallner, A. 2004. Importance of monitoring terrestrial arthropod biodiversity in Illinois ecosystems, with special reference to Auchenorrhyncha. Critical Trend Assessment Program Report (*in preparation*).

Ward, P. S. 1987. Distribution of the introduced Argentine ant (*Iridomyrmex humilis*) in natural habitats of the lower Sacramento Valley and its effects on the indigenous ant fauna. Hilgardia 55:1–16.

Watts, C. H. and G. W. Gibbs. 2002. Revegetation and its effect on the ground-dwelling beetle fauna of Matiu-Somes Island, New Zealand. Restoration Ecology 10(1):96-106.

Wheeler, G. C., and J. Wheeler. 1973. Ants of Deep Canyon. University of California Press, Berkely, California, USA.

Whitecotton, Capt. R. C. A., M. B. David, R. G. Darmody, and D. L. Price. Impact of foot traffic from military training on soil and vegetation properties. Environmental Management 26(6):697-706.

Wilson, E.O. 2003. *Pheidole* in the New World. Harvard University Press.

Appendix 1. Photographs of selected taxa in many of the families of terrestrial insects collected at Camp Atterbury, July, 2004. Scale bar length indicated in parentheses. Taxa present in this appendix but not in Table 3 generally are from Malaise samples. Ants, most Homoptera, and several beetles are pictured in the body of the text of this report.

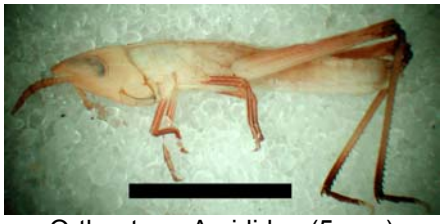
Collembola: Tomoceridae (2 mm)

Collembola: Mackenziellidae (0.5 mm)

Collembola: Hypogastruridae (0.5 mm)

Collembola: Entomobryiidae (2 mm)

Orthoptera: Rhaphidophoridae (5 mm)



Orthoptera: Acrididae (5 mm)

Blatteria: Blattidae (5 mm)

Neuroptera: Hemerobiidae (5 mm)

Homoptera: Aphididae (0.5 mm)

Hemiptera: Anthocoridae (1 mm)

Hemiptera: Lygaeidae (5 mm)

Hemiptera: MIridae (5 mm)

Hemiptera: Pendatomidae (5 mm)

Hemiptera: Reduviidae (5 mm)

Hemiptera: Theriocoridae (2.5 mm)

Thysanoptera: Thripidae (0.5 mm)

Thysanoptera: Phlaeothripidae

Thysanoptera: Aeolothripidae (2 mm)

Lepidoptera: Pterophoridae (5 mm)

Lepidoptera: Noctuidae (5 mm)

Lepidoptera: Gelechiidae (5 mm)

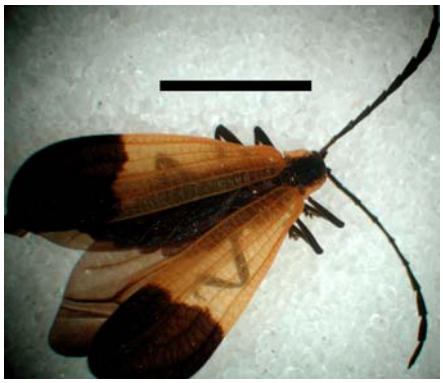
Lepidoptera: Blastobasidae (1 mm)

Coleoptera: Ptilidae (0.5 mm)

Coleoptera: Pselaphidae (0.5 mm)

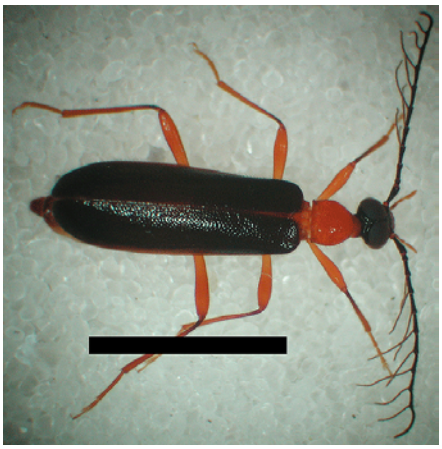
Coleoptera: Ptilodactylidae (2 mm)

Coleoptera: Mordellidae (2 mm)

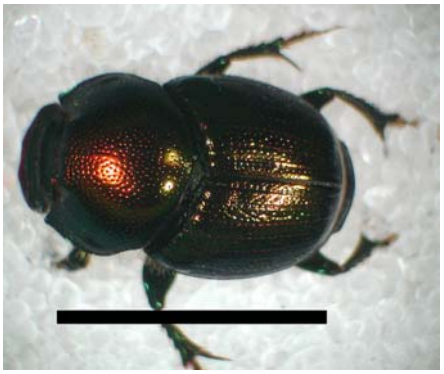


Coleoptera: Lycaenidae (5 mm)

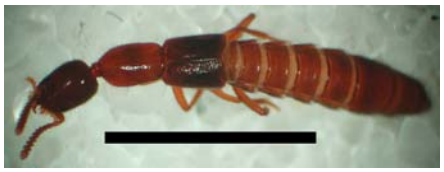
Coleoptera: Nitidulidae (2 mm)



Coleoptera: Pyrrhochoridae (5 mm)



Coleoptera: Scarabaeidae (5 mm)



Coleoptera: Staphylinidae (2.5 mm)

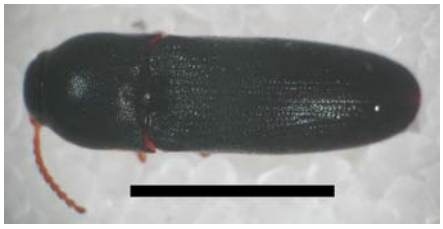
Coleoptera: Chrysomelidae (5 mm)

Coleoptera: Dascillidae (5 mm)

Coleoptera: Lampyridae (5 mm)

Coleoptera: Lampyridae (5 mm)

Coleoptera: Histeridae (5 mm)

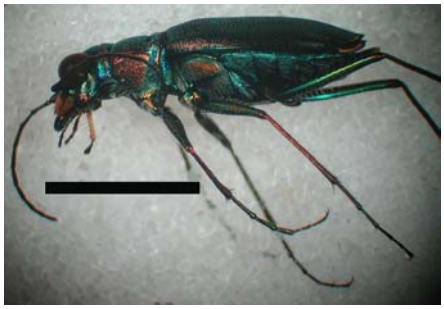


Coleoptera: Elateridae (2.5 mm)

Coleoptera: Cucujidae (5 mm)

Coleoptera: Curculionidae (2.5 mm)

Coleoptera: Cryptophagidae (1 mm)



Coleoptera: Cicindellidae (5 mm)

Coleoptera: Cantheridae (5 mm)

Coleoptera: Anthribidae (5 mm)

Hymenoptera: Rhopalosomatidae (5 mm)

Hymenoptera: Platygastridae (0.5 mm)

Hymenoptera: Scelionidae (0.5 mm)

Hymenoptera: Pteromalidae (1 mm)

Hymenoptera: Perilampidae (1 mm)

Hymenoptera: Trichogrammatidae (0.5 mm)

Hymenoptera: Proctotrupidae (5 mm)

Hymenoptera: Ormyridae (1 mm)

Hymenoptera: Mymaridae (0.5 mm)

Hymenoptera: Eurytomidae (2 mm)

Hymenoptera: Eulophidae (1 mm)

Hymenoptera: Encyrtidae (0.5 mm)

Hymenoptera: Cynipidae (1 mm)

Hymenoptera: Ceratophoronidae (0.5 mm)

Hymenoptera: Colletidae (2.5 mm)

Hymenoptera: Ichneumonidae (5 mm)

Hymenoptera: Halictidae (5 mm)

Hymenoptera: Pompilidae (5 mm)

Hymenoptera: Sphecidae (5 mm)

Hymenoptera: Tenthredinidae (5 mm)

Hymenoptera: Braconidae (5 mm)

Diptera: Syrphidae (5 mm)

Diptera: Muscidae (5mm)

Diptera: Bibionidae (5 mm)

Diptera: Cicidomyiidae (1 mm)

Diptera: Spheroceridae (1 mm)

Diptera: Phoridae (1 mm)

Diptera: Chloropidae (1 mm)

Diptera: Sciaridae (2.5 mm)

Diptera: Diastatidae (2 mm)

Diptera: Sarcophagidae (5 mm)

Diptera (5 mm)