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Sampling Terrestrial Arthropod Biodiversity:
A Case Study in Arkansas

A dissertation submitted in partial fulfillment
of the requirements for the degree of
Doctor of Philosophy in Entomology

By

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Abstract

The Interior Highlands is a biodiversity hotspot, with at least 200 known endemic species, but is understudied compared to hotspots, such as the Southern Appalachians. In order to begin to rectify this issue, a nine month study was conducted from mid-March through early December at a 4 ha site at Steel Creek, Buffalo National River, in Newton County, Arkansas. Thirteen collecting methods were employed, including three colors of Lindgren funnel trap, five colors of pan trap, Malaise traps, canopy traps with upper and lower collectors, pitfall traps, and Berlese-Tullgren extraction of leaf litter, which resulted in the collection of 1311 samples during 17 collection events. Target groups, including Formicidae, Carabidae, Cerambycidae, Curculionoidea, Araneae, Isopoda, Mecoptera, Phasmida, Vespidae, Ixodidae, Phalangodidae, and select Diplopoda and Orthoptera were identified. This resulted in 47,481 specimens representing 706 species that were curated and identified, including 18 putatively undescribed species, 56 species that represented new state records, 15 non-native species, and three species of Carabidae endemic to the Interior Highlands, two of which (*Rhadine ozarkensis* and *Scaphinotus infletus*) were previously known only from the original type series. Collection data for four beetle taxa – Buprestidae, Carabidae, Cerambycidae, and Curculionoidea excluding Scolytinae – as well as all taxa combined were analyzed. Pitfall and Malaise traps were the most effective (define here as collecting the most species with fewest samples) combination of collection methods for Carabidae, Curculionoidea, and the combined taxa, while Malaise traps alone and Malaise or canopy traps and green Lindgren funnel traps were the most effective collection methods for Cerambycidae and Buprestidae, respectively. Color of Lindgren funnel traps was important when targeting Buprestidae and some Curculionoidea, but not Carabidae or Cerambycidae. Extrapolated rarefaction curves indicated that 300–600 samples were required

per trap type (1000+ for pitfall traps) before species accumulation is saturated. Finally, four rarely collected specimens or species – a *Temnothorax curvispinosus* gynandromorph, *Orussus minutus*, *Eudociminus mannerheimii*, and *Merope tuber* – are treated individually in detail.

Acknowledgements

I thank Danielle M. Fisher for her assistance with sorting samples and curating thousands of beetles – without her help I would have accomplished much less in the pursuit of my degree; J. Ray Fisher for his assistance identifying random spiders, wasps and other insects and unlimited enthusiasm with respect to things entomological and science in general; all of the many coauthors I've written publications with and who have provided excellent feedback and constructive criticism; the University of Arkansas interlibrary loan department for their assistance in tracking down and scanning the thousands of articles I requested; Kyle Schnepf, David Smith, and Clint Trammel for identifying Buprestidae, “Symphyta”, and Pompilidae, respectively; Buffalo National River for granting the permit that allowed my research to take place; Tim Kring and Rob Wiedenmann for their excellent academic and life advice; my committee; and my adviser Ashley Dowling who let me develop my own project but was always available when problems inevitably arose.

This project and the preparation of this publication was funded in part by the State Wildlife Grants Program (Grant # T39-05) of the U.S. Fish and Wildlife Service through an agreement with the Arkansas Game and Fish Commission.

Dedication

To my wife, Sarah, who has supported me through graduate school, falconry, and every other goal I've set my sights on, and my son, Vaun, who, despite causing many sleepless nights is the best thing to happen to me; my father, for showing me how to collect hibernating insects under bark during winter, thus sparking my love of entomology and for allowing me the freedom to explore the forest and creeks without supervision when I was young; my mother, grandparents and brother for always believing I could accomplish anything; my best friend Jon and the other Purdue Bug Kids Amy and Amber who have travelled this long road to Ph.D.s with me; and Stephen "Steve" Irwin, for his role in the television series *The Crocodile Hunter*, which inspired me as a child.

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List of published works

- Skvarla, M.J., Larson, J.L., Dowling, A.P.G. 2014. Pitfalls and preservatives: A review. *Journal of the Entomological Society of Ontario*, 145: 15–43. Chapter II. Published.
- Skvarla, M.J., Larson, J.L., Fisher, J.R., Dowling, A.P.G. A review of terrestrial and canopy Malaise traps. Chapter III. Prepared for submission.
- Skvarla, M.J., Fisher, D.M., Schnepf, K.E., Dowling, A.P.G. Terrestrial arthropods of Steel Creek, Buffalo National River, Arkansas. I. Select beetles (Coleoptera: Buprestidae, Carabidae, Cerambycidae, Curculionoidea excluding Scolytinae). *Biodiversity Data Journal*. Chapter IV. In press.
- Skvarla, M.J., Dowling, A.P.G. Collecting beetles: Analysis of a single-site data set and comparison of trapping techniques (Coleoptera: Curculionoidea excluding Scolytinae, Carabidae, Buprestidae, Cerambycidae). Chapter V. Prepared for submission
- Skvarla, M.J., Dowling, A.P.G. Sampling terrestrial arthropod diversity: A case study. Chapter VI. Prepared for submission.
- Skvarla, M.J., Dowling, A.P.G. 2014. First report of gynandromorphism in *Temnothorax curvispinosus* (Mayr, 1886) (Hymenoptera: Formicidae). *Proceedings of the Entomological Society of Washington*, 116(3): 349–353. Chapter VII. Published.
- Skvarla, M.J., Tripodi, A.D., Szalanski, A.L., Dowling, A.P.G. 2015. New records of *Orussus minutus* Middlekauff, 1983 (Hymenoptera: Orussidae) represent a significant western range expansion. *Biodiversity Data Journal*, 3: e5793. Chapter VIII. Published.
- Skvarla, M.J., Bertone, M., Fisher, J.R., Dowling, A.P.G. In press. New range and host records and redescription for the cypress weevil, *Eudocimus mannerheimii* (Boheman, 1836) (Coleoptera: Curculionidae). *Coleopterists Bulletin*. Chapter IX. In press
- Skvarla, M.J., Hartshorn, J.A., Dowling, A.P.G. 2014. Report on a large collection of *Merope tuber* Newman, 1838 (Mecoptera: Meropeidae) from Arkansas with notes on collection technique, sex ratio, and male clasper size. *Psyche*, vol. 2014, Article ID 530757, 6 pages, doi:10.1155/2014/530757. Chapter X. Published.

I. Introduction.

Life on Earth is currently experiencing a sixth mass extinction, with species extinction rates 100–10,000 times higher than historic background rates (Pimm et al. 1995; Balmford 1996; Wake & Vredenburg 2008; Barnosky et al. 2011; de Vos et al. 2015). Climate change, globalization, spread of exotic species, and habitat fragmentation have all been implicated as causes. Understanding the causes and developing strategies to avert or minimize this crisis has become a priority among biologists. Vascular plants, vertebrates, and invertebrates are all disproportionately affected by the extinction crisis; however, invertebrates, especially endemic species with limited ranges, are often at the highest risk for extinction (Conrad et al. 2006; Thomas *et al.* 2004).

One of potential solutions to the global extinction crisis is to protect biodiversity hotspots, which are areas of high biodiversity and endemism (Médail & Quézel 1999). Examples of such hotspots are the Mediterranean biome, which comprises 2% of the world's surface but contains 20% of the total floristic richness, and the tropical Andes, which contains nearly 6% and 7% of the world's total vertebrate and plant species, respectively (Médail & Quézel 1997; Meyers et al. 2000). By focusing on protecting these areas instead of individual species, it is possible to protect large percentages of biodiversity in the most spatially- and monetarily-efficient manner (Meyers 1989; Meyers 1990).

The Interior Highlands in the mid-central United States is a biodiversity hotspot with at least 200 endemic species, more than half of which are arthropods (Allen 1990, Robison and Allen 1995, Pringle and Witsell 2005, Zollner et al. 2005, Robison et al. 2008), and at least 58 species that exhibit highly disjunct populations (The Nature Conservancy, Ozarks Ecoregional Assessment Team 2003). It is a mountainous region surrounded by areas of lower elevation that

has remained unsubmerged and unglaciated since the Permian (~290 MYA) and thus acted as a refugia during times of inhospitable climate; additionally, the region was historically connected to the southern Appalachians, though this connection was severed by the early Cenozoic (60 MYA) (Skvarla *et al. in press*). However, the Interior Highlands is under surveyed compared to other similar North American regions of high biodiversity, such as Great Smokey Mountain National Park and the Southern Appalachians more generally.

Efficiently collecting terrestrial arthropods – defined here as collecting the highest number of species with the fewest number of samples – is an important component of survey work as they represent the majority of terrestrial. Much has been written about surveying specific taxa (e.g., epigeal Carabidae: Greenslade 1964, Spence & Niemelä 1994; Formicidae: Andersen 1991, Agosti & Alonso 2000; Araneae: Duffey 1972) or habitats (e.g., dry riverbeds: Corti *et al.* 2013; decaying wood/wood fungi: Kaila 1993, Lachat *et al.* 2006, Ferro & Carlton 2011), comparing a limited number of collection methods (e.g., Juillet 1963, Duelli *et al.* 1999, Wells & Decker 2006, Campbell & Hanula 2007, Lamarre *et al.* 2012, Corti *et al.* 2013), or comparing methods using specimens identified to higher taxonomic units (e.g., order, family, genus) (e.g., Juillet 1963, Lamarre *et al.* 2012). While these studies are laudable, few studies have compared multiple methods using specimens identified to species from a breadth of taxa.

The goals of this dissertation are thus three-fold: 1) intensively survey a single site in order to establish a baseline list of taxa to which future change can be compared; 2) compare collecting methods in order to determine the most efficient combination of traps and the minimum number of samples needed to collect most species so future surveys in similar environments can maximize the return of effort; and 3) report rare and endemic terrestrial

arthropods, as well as species that are new to Arkansas, in order to better understand the arthropods native to the state.

These goals have been addressed in the following manner: Chapters II and III extensively review pitfall and Malaise traps, respectively, which were found to produce the highest number of species and specimens, and exhibited the lowest similarity and overlap in trap catch of the collecting methods considered. Understanding the nuances and issues with both traps is important when implementing them in biodiversity studies. Additionally, the chapters are included in lieu of a more formal literature review.

Chapter IV provides an overview of the geologic history of the Interior Highlands and describes an intensive nine-month survey conducted at Steel Creek, at Buffalo National River. The identity of species in four diverse groups of beetles – Buprestidae, Carabidae, Cerambycidae, and Curculionoidea – were determined and new state records established for 31 species. Additionally, three Interior Highland endemic ground beetles, two of which are known only from the type series, are reported from the site. Chapter V begins with a review of rapid biodiversity assessment techniques and reports analyses of the beetle species reported in Chapter IV, including the most efficient combination of traps for each larger taxon (superfamily/family), role of color in Lindgren funnel traps in attracting different species, and phenology and seasonality of each species. Chapter VI expands upon Chapter V by reporting similar statistics for a much larger dataset that includes 46,146 specimens representing 533 species from a diversity of higher taxa, including beetles, wasps, spiders, mecopterans, millipedes, and others. A workflow for the analyses conducted in Chapters V and VI is presented in Appendix I.

Chapters VII–X are examples of papers and analyses that can be extracted from larger survey efforts and include information about species new to or rare in Arkansas. Chapter VII is

about an individual *Temnothorax curvispinosus* (Formicidae) exhibiting gynandromorphism. This species is common in forests and was previously recorded from the state, but this is the first time gynandromorphism is reported in the species. It also highlights the rarity of finding such a genetic anomaly as this was the only gynandromorph collected out of more than 28,000 ants examined during the study.

Chapter VIII reports *Orussus minutus* species from Arkansas for the first time. The specimens represent a significant western range extension and first report west of the Mississippi River. Collection data for unpublished specimens housed in the United States National Collection was provided by collaborator Dr. David Smith. These specimens more than double the number of specimens reported in the literature and include new state records for Michigan and West Virginia. Additionally, the paper includes data gathered from social media and citizen science websites, as well as a brief note about the future of such websites in natural history and descriptive science.

Chapter IX reports *Eudocimus mannerheimii* (Coleoptera) from Arkansas for the first time. Previously the species had been reported from coastal states from New York south to Florida, west to Louisiana and Mexico. The Arkansas specimens therefore represent the northwestern-most, inland records for the species. *Eudocimus mannerheimii* is reported to feed on various Cupressaceae, including bald cypress (*Taxodium distichum*), pond cypress (*T. ascendens*), and Japanese cedar (*Cryptomeria* sp.); however, eastern red cedar (*Juniperus virginiana*) is the only representative of the family at the collection site, so while I did not observe feeding or oviposition, I hypothesize it to be the host plant. Additional information about specimens collected in North Carolina, including records from arborvitae, was provided by co-author Dr. Matt Bertone.

Chapter X reports the second largest collection of the rarely collected *Merope tuber* (Mecoptera). We reported phenology and male clasper size of the specimens, as well as notes on the collecting technique so the species may be more easily collected in the future.

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II. Pitfalls and preservatives: A review.

Abstract.

An extensive review of the factors that affect the performance of arthropod pitfall traps is given. Liquid preservatives are discussed in a separate section because the choice affects the quality and composition of taxa collected in pitfalls.

Introduction.

Pitfall traps are a popular method for collecting ground beetles, spiders, ants and other epigeal arthropods (Westberg 1977; Niemelä *et al.* 1992; Bestelmeyer *et al.* 2000; Southwood & Henderson 2000; Phillips & Cobb 2005). While many shorter, general overviews exist (e.g., general techniques: Balogh 1958; Duffey 1972; Bestelmeyer *et al.* 2000; Southwood and Henderson 2000; Woodcock 2005; issues with pitfalls: Adis 1979), none have exhaustively examined the published literature recently. Herein we present such a review with the hope it will provide a sound base for those incorporating pitfall traps into their research.

While the choice of preservative will affect the quality of specimens in any type of trap, it is a critical decision in pitfalls for several reasons. Chiefly, preservatives differentially attract and repel select arthropod taxa, which will affect the composition of taxa collected (Weeks & McIntyre 1997). Additionally, pitfalls are often set without covers in open fields, so lose more preservative through evaporation than other traps and are affected to a greater degree by rain and dilution by rainwater (Porter 2005). Therefore, we include a section detailing possible positives and negatives of preservatives used in pitfall traps.

Pitfall Traps

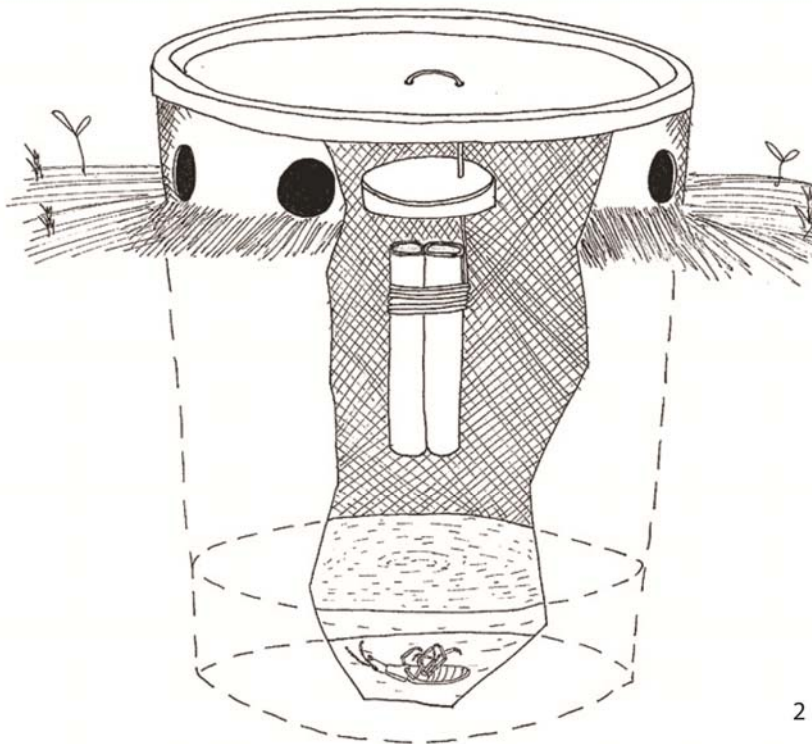
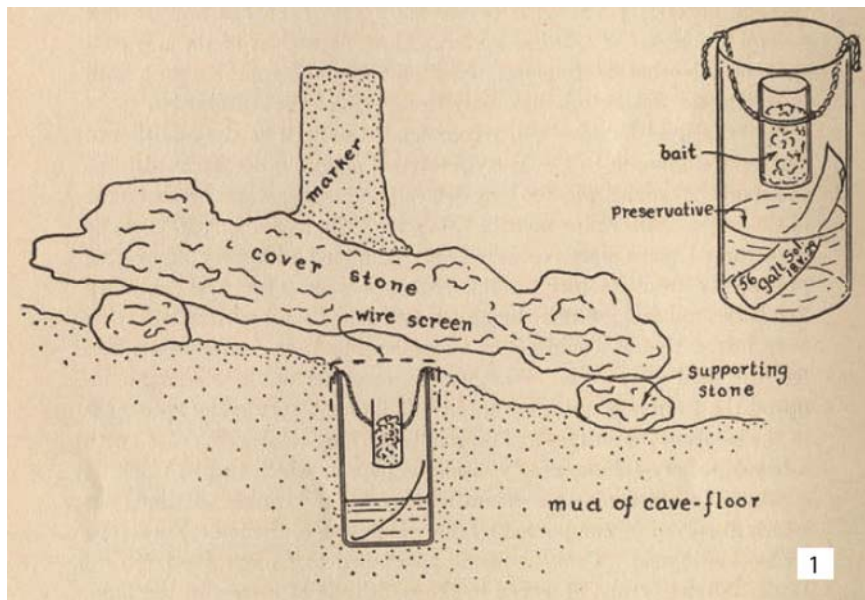
Pitfall traps were first described by Hertz (1927) and shortly thereafter by Barber (1931) (Fig. 1) for collecting cave-inhabiting insects. A pitfall trap is simple in design, consisting of a collecting container buried flush with the ground that passively collects epigeal organisms that accidentally fall into the trap. It may be constructed from any container large enough to hold the target organism, including a large bucket for reptiles or small mammals (Ellis 2013), small plastic cup for larger insects such as Carabidae and large Formicidae (Luff 1975; Abensperg-Traun & Steven 1995), or a glass test tube for small insects such as most Formicidae and small Carabidae (Luff 1968; Abensperg-Traun & Steven 1995). Pitfall traps are widely used in biodiversity surveys as they are cost-effective, ecologically sensitive, collect large numbers of arthropods (Gist & Crossley 1973; Ekschmitt *et al.* 1997; Southwood & Henderson 2000; Work *et al.* 2002), and collect nocturnal species missed by other methods (Törmälä 1982; Samways 1983; Donnelly & Gilme 1985; Huusela-Veistola 1996).

Pitfall traps have been used to sample many arthropod groups, including Scorpionida (Tourtlotte 1974; Margules *et al.* 1994); Isopoda (Hamner *et al.* 1969; Hayes 1970; Paoletti & Hassall 1999; Hornung *et al.* 2007); Diplopoda (Van der Drift 1963; Kurnik 1988; Mesibov *et al.* 1995; Kime 1997; Snyder *et al.* 2006), Chilopoda (Kurnik 1988; Fründ 1990; Adis 1992; Shear & Peck 1992; Voigtlander 2003), and Symphyla (Adis 1992; Shear & Peck 1992; Clark & Greenslade 1996); Araneae (Duffey & Millidge 1954; Muma 1973; Uetz 1977; Corey & Taylor 1988; Bultman 1992; Koponen 1992; Bauchhenss 1995; Buddle *et al.* 2000); Acari (Zacharda 1993; Wickings 2007; Kłosin'ska *et al.* 2009; Mayoral & Barranco 2009; Wohltmann & Mąkol 2009; López-Campos & Vázquez-Rojas 2010; Clark 2013); Collembola (Joosse-van Damme 1965; Pedigo 1966; Budaeva 1993; Cole *et al.* 2001; Frampton *et al.* 2001); Coleoptera

(Backlund & Marrone 1997; Simmons *et al.* 1998; Arbogast *et al.* 2000) including Carabidae (Anderson 1985; Kálás 1985; Cameron & Reeves 1990; Epstein & Kulman 1990; Togashi *et al.* 1990), Tenebrionidae (Ahearn 1971), Staphylinidae (Anderson 1985; Braman & Pendley 1993; Ekschmitt *et al.* 1997), Scarabaeoidea (Young 1981; Peck & Howden 1985; Martínez *et al.* 2009; Anlaş *et al.* 2011; Thakare *et al.* 2011), and certain Latridiidae (Hartley *et al.* 2007); Formicidae (Van der Drift 1963; Greenslade 1973; Anderson 1991; Abensperg-Traun & Steven 1995; Bestelmeyer *et al.* 2000); and even terrestrial Amphipoda (Craig 1973; Margules *et al.* 1994) and Decapoda (Williams *et al.* 1985; Smith *et al.* 1991; Hamr & Richardson 1994; McGrath 1994; McIvor & Smith 1995). Of these taxonomic groups, ground-dwelling Araneae and Coleoptera have been the most studied (Westberg 1977).

Variations on the basic trap have been developed, including more elaborate traps for use under snow (Kronestedt 1968; Steigen 1973); live traps with a layer of gauze that keeps trapped organisms from drowning in rainwater (Duffey 1972); modifications that allow excess rainwater to drain before overflowing the trap (Duffey 1972; Porter 2005); integrated internal funnel and rain cap (Fichter 1941); collecting cup integrated into a larger structure with a base or ramp (Muma 1970); use of holes or slits in the side of a container so an integrated cap can be used (Fig. 2) (Nordlander 1987; Lemieux & Lindgren 1999); modifications to facilitate emptying (Rivard 1962), including automated devices for segregating trap catch over time (Williams 1958; Blumberg & Crossley, 1988; Buchholz 2009); designs to reduce mortality of vertebrate bycatch including floating shelters and wire mesh (Kogut & Padley 1997; Pearce *et al.* 2005); and inexpensive designs using commonly discarded household materials (Morris 1975; Clark & Bloom 1992). Other techniques, such as using an auger bit to drill placement holes for small diameter traps, and equipment, such as a device that can pull traps out of placement holes

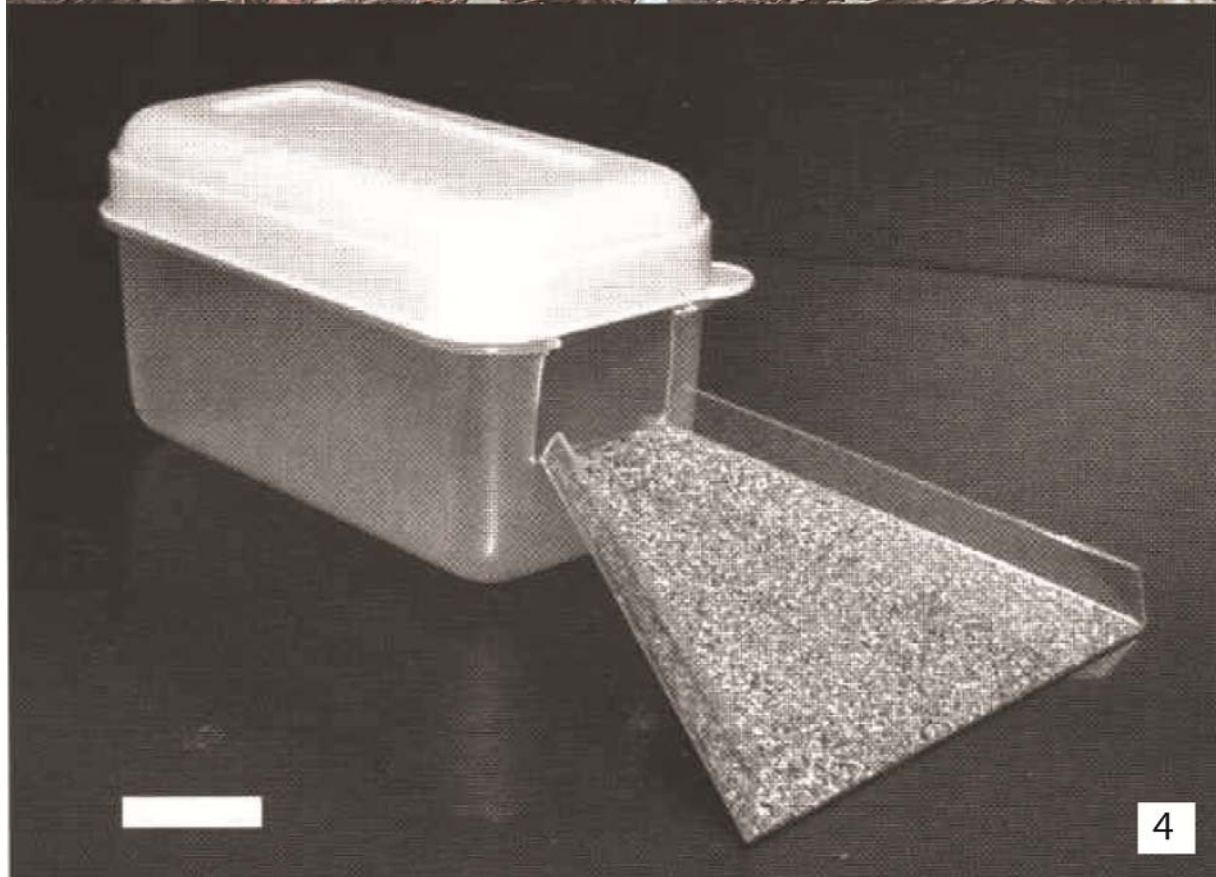
without kneeling or disturbing the surrounding soil, have been developed to make pitfall trapping easier (Vogt & Harsh 2003).



Figures 1–2. Examples of pitfall traps. **Fig. 1.** Pitfall trap described by Barber for collecting cave-inhabiting insects. After Barber (1931). **Fig. 2.** Pitfall trap modified with entrances in the side of the collection cup, which discourages vertebrates from entering the trap and allows the use of an integrated rain cap. Modified from Nordlander (1987) with permission.

Barrier fences have been employed, either with a single pitfall situated in the middle of the fence or with pitfalls at the end of the fence (Fig. 3) (Haeck 1971; Meijer 1971; Reeves 1980; Durkis & Reeves 1982). Linear pitfall traps constructed from house gutters have been employed with success in certain situations, such as investigating the speed and timing of insect populations moving between habitats (Pamanes & Pienkowski 1965; Goulet 1974; Pausch *et al.* 1979).

Ramp traps collect arthropods similarly to pitfall traps, but rather than being sunk into the ground target taxa are directed upwards into the trap via ramps; this allows them to be employed where conventional pitfalls cannot, such as where digging is difficult (e.g., on rocks or in caves) or prohibited by law (Bouchard *et al.* 2000; Campbell *et al.* 2011). Bostanian *et al.* (1983) proposed the first ramp trap design, which is constructed from metal, making it rather bulky and expensive and biased towards large ground beetles. Bouchard *et al.* (2000) proposed a revised design that utilizes plastic sandwich containers and plastic ramps, rendering it light-weight and inexpensive (Fig. 4). Ramp traps have been successfully employed in caves (Campbell *et al.* 2011), areas polluted due to industrial mining (Babin-Fenske & Anand 2010), orchards (Smith *et al.* 2004), and vineyards (Goulet *et al.* 2004). Ramp traps capture a higher abundance and diversity of epigeal spiders than conventional pitfall traps, though when comparing other taxa (e.g., beetles) they collect a different species composition, thus making direct comparison between the trap types difficult or impossible (Pearce *et al.* 2005; Patrick & Hansen 2013). Additionally, ramp traps capture fewer vertebrates than conventional pitfall traps (Pearce *et al.* 2005).



Figures 3–4. Examples of pitfall traps. **Fig. 3.** Pitfall traps (modified from Nordlander 1987) on either side of a barrier fence. **Fig. 4.** Ramp trap. After Bouchard *et al.* (2000). Used with permission.

Colored pan traps, sometimes referred to as water traps, are generally used to collect flying insects via visual response to color cues (e.g. yellow, blue, purple or red) (Kirk 1984; Aguiar & Sharkov 1997; Leong & Thorp 1999; Pucci 2008; Gollan *et al.* 2011). While pan traps are generally set on or above the ground, they may be sunk into it, effectively becoming pitfall traps that also attract and capture flying insects.

Issues with pitfall traps

Objections have been raised to the use of pitfall traps in ecological studies (Adis 1979; Majer 1997; Southwood & Henderson 2000) because they do not evenly catch different taxa for several reasons:

1. Different taxa react differently at the lip of the trap. Gerlach *et al.* (2009) found that millipedes show the most trap-avoidant behavior (20–60%) and carabids show the least (10–25%); overall they found an average of 28% of taxa that encountered a trap were caught, with a range of less than 5% (*Enantiulus nanus* (Latzel, 1884) (Julidae)) to 70% (*Pterostichus burmeisteri* Herr, 1838 (Carabidae)). Luff (1975) found approximately 75% of Carabidae that encounter the edge of a pitfall are collected. In mark-recapture studies, some species become trap-shy if they have been caught previously while other species do not (Benest 1989).

2. Activity level (Ekschmitt *et al.* 1997), which is affected by variables such as species-specific behavior (Greenslade 1964; Curtis 1980; Anderson 1991; Topping 1993; Spence & Niemelä 1994; Obrist & Duelli 1996); differences between gender and age (Hayes 1970; Benest 1989; Topping & Sunderland 1992; Thomas *et al.* 1998) including mate-searching (Tretzel 1954), post-copulatory dispersal of females (Merrett 1967) and searching for oviposition sites (Duffey 1956); weather (Williams 1940; Briggs 1961; Greenslade 1961; Juillet 1964; Ericson 1979; Drake 1994); vegetation (Deseo 1959; Greenslade 1964; Novák 1969; Baars 1979), habitat

structure (Melbourne 1999; Melbourne *et al.* 1997; Thomas *et al.* 1998), and habitat type (Melbourne *et al.* 1997); size (Luff 1975; Thiele 1977; den Boer 1981; Franke *et al.* 1988) and speed (Braune 1974; Adis 1976); and hunger and prey density (Grüm 1971; Müller 1984; Henrik & Ekbom 1994), also affect the number of organisms trapped, both within and between taxa (Southwood, & Henderson 2000) and are more influential factors than population size (Briggs 1961) in determining trap catch.

3. Larger species are caught in significantly higher numbers than smaller species (Carabidae: Franke *et al.* 1988; Spence & Niemelä 1994). Several reasons have been suggested for this. Larger, faster beetles are successfully caught a higher percentage of the time than smaller, slower beetles (Braune 1974; Adis 1976) – though some authors have found size and speed do not affect the ability to be caught (Luff 1975; Halsall & Wratten 1988). Smaller beetles may escape more readily from traps because scratches and soil on trap walls may be enough to support their mass as they try to climb out whereas larger beetles fall (Spence & Niemelä 1994).

4. Species-specific morphology can affect escape ability; e.g., *Demetrias atricapillus* (L.) has adhesive setae on the underside of the tarsi that allow it to climb out of pitfalls more easily than other similarly sized carabids (Halsall & Wratten 1988).

5. Pitfall traps do not accurately reflect absolute density of the organisms sampled. This has been demonstrated in the field (Grüm 1959; Briggs 1961; Mitchell 1963; Marsh 1984; Topping & Sunderland 1992) and experimentally in a caged system (Lang 2000) – though caution should be exercised interpreting caged results as they may be skewed by “trap-happy” beetles that prefer dry pitfalls as refugia (Adis 1979, citing Thomas & Sleeper 1977) and may suffer from “Kreb’s effect” (Mac Arthur 1984). However, it should also be noted that some studies have recorded 73–96% capture rates of marked beetles in caged systems (Bonkowska &

Ryszkowski 1975; Dennison & Hodkinson 1984; Desender *et al.* 1985; Desender & Maelfair 1986; Clark *et al.* 1995; Holland & Smith 1999) and one study found no difference between population estimates of millipedes, spiders, and beetles based on hand collecting or pitfalls in a caged system (Gist & Crossley 1973), suggesting such systems may accurately reflect absolute density in certain situations with specific taxa.

In response to these criticisms, various calculations have been proposed to correct for the differences between taxa collected and true population density based on locomotory activity and motility range (Heydemann 1953; Tretzel 1955; Braune 1974; Thomas & Sleeper 1977; Kuschka *et al.* 1987; Stoyan & Kuschka 2001; see also Seifert 1990), though these have been rejected by others (Adis 1979; Müller 1984; Franke *et al.* 1988; Gerlach *et al.* 2009).

Additionally, it has been argued that samples pooled over an entire season correctly represent local species abundance as variations due to weather and other factors that affect activity level are averaged out (Baars 1979; den Boer 1986; Luff 1982). Results of other studies are conflicting, with some showing a large amount of variation between sampling periods in similar habitat when the sampling periods are short (Niemelä *et al.* 1986), and others showing that traps set for short periods caught all species accumulated by longer trapping periods (Niemelä *et al.* 1990; Borgelt & New 2006). In addition, much of the cited research has only examined carabids caught by pitfalls. When collecting other taxa, pitfalls may estimate absolute population density relatively well (ants: Andersen 1991; Vorster *et al.* 1992; Lindsey & Skinner 2001; cursorial spiders: Muma & Muma 1949; Duffey 1962; Huhta 1971; Uetz & Unzicker 1976; tenebrionids: Thomas & Sleeper 1977).

Certain ecological questions, such as comparing taxa along a successional gradient (Bultman & Uetz 1982) or between similar plots (Koivula *et al.* 1999), may be answered as taxa will be equally biased to pitfall traps along the gradient or between plots.

Pitfalls can be used to answer non-ecological questions, such as investigating the phenology (Maelfait & Baert 1975), seasonal and circadian activity (Williams 1959a, b; Williams 1962; Breymeyer 1966a, b; Doane & Dondale 1979), and lifespan (Goulet 1974) of commonly collected taxa, estimating the timing of movement of epigeal species between habitats (Pamanes & Pienkowski 1965; Pausch *et al.* 1979), and estimating dispersal using mark-release-recapture methods (Ericson 1977; Best *et al.* 1981). They also can be employed in taxonomic surveys, though should be paired with other sampling techniques that complement the deficiencies of pitfalls (Majer 1997)

Pitfall trap design

If pitfall traps are to be employed, several considerations must be made as there are many factors that can affect the taxa collected.

Effects of shape, size, and material of receptacle. The shape of the trap affects the composition and number of taxa collected (Cheli & Corley 2010). Pitfalls may be straight-sided or round (Southwood & Henderson 2000), depending on the container used; however, round and straight-edged traps with the same perimeter length catch different numbers of specimens (Braune 1974; Luff 1975; Adis 1976; Spence & Niemelä 1994).

Different diameters of pitfall trap collect different taxa at different rates. When examining ants, larger diameter pitfalls catch more species, though differences are primarily due to differential capture rates of rare species (Abensperg-Traun & Steven 1995). Work *et al.* (2002) compared catch rates and species richness of Carabidae, Staphylinidae, and Araneae

across five diameters (4.5, 6.5, 11, 15, and 20 cm) of pitfall traps; they found that, after standardizing circumference, small traps caught more small carabids and staphylinids and large traps caught more wolf spiders. Luff (1975) found that small traps (2.5 cm dia.) were the most efficient at catching small species of carabids, while large traps (10 cm dia.) caught relatively more large beetles; however, their small traps were made of glass and large traps made of metal, which probably had a confounding effect on the results. Brennan *et al.* (1999) found the largest and second largest traps (17.4 and 11.1 cm dia.) they tested caught the most diverse assemblage of species, though considered the smaller of the two traps more appropriate for sampling spiders as it may decrease the potential of capturing non-target species. One option when using larger traps is to add a funnel to the trap in order to increase trap retention (Vlijm *et al.* 1961).

Another aspect of size is the depth of the trap. Shallow (8 cm) and deeper (15 cm) pitfalls do not effect ant diversity capture (Pendola & New 2007), therefore, when targeting ants, shallow pitfalls are preferred as small vertebrates, such as skinks, may escape more easily from them, thus reducing vertebrate bycatch. However, this has only been demonstrated in ants and may not hold true for large insects, such as some carabids, which are bigger than some small vertebrates.

Pitfall traps used to collect insects have been constructed out of glass (Briggs 1961; Greenslade 1964; Borgelt & New 2006; Pendola & New 2007), plastic (Luff 1973; Morrill 1975; Clark & Blom 1992; Spence & Niemelä 1994), or metal (Ahearn 1971; Hinds & Rickard 1973; Clark & Blom 1992). Choice of material can affect the taxa sampled in live traps as escape rates differ. One study on carabids found 0% escape from glass traps, 4% escape per day from plastic traps, and 10% escape per day from metal traps (Luff 1975). Other studies have also found glass pitfalls retain more arthropods than plastic or metal (Vennila & Rajagopal 2000), though one

found no difference between glass and plastic traps (Waage 1985). Similarly, Topping and Luff (1995) found plastic traps with rough surfaces caught fewer linyphiid spiders than similar traps with smooth surfaces.

Finally, color of the pitfall trap affects the taxa collected: white and yellow traps catch higher numbers of Apidae, Araneae, Carabidae, Diptera, and Formicidae, while brown and green traps catch higher numbers of Isopoda (Buchholz *et al.* 2010).

Effects of trap design, layout, and site selection. Some studies have found that covers do not affect the composition of arthropods trapped by pitfall traps (Work *et al.* 2002; Buchholz & Hanning 2009; Cheli & Corley 2010) while others have found they do (Briggs 1961; Baars 1979; Spence & Niemelä 1994). Some of this may be due to the material used as a cover. Man-made covers, such as metal or ceramic tile, are generally used. Suggestions have been made to use natural material such as bark or rock for covers (van der Berghe 1992), though this has not been systematically investigated.

Pitfall traps that have an integrated cap and circular entrances in the sidewall of the trap (first proposed by Nordlander 1987) caught 80% of the same common carabid species as conventional pitfalls in one study (Lemieux & Lindgren 1999), but otherwise have not been thoroughly investigated and compared to conventional traps.

Pitfall traps must be level with the soil surface as excessive inclination of the soil ringing the traps may direct some arthropods away from the trap (Heydemann 1953). Similarly, a plastic disc surrounding the trap will influence sample size (Adis 1976).

Subterranean pitfall traps have been employed to trap hypogaeic ants (Yamaguchi & Hasegawa 1996; Anderson & Brault 2010; Berghoff *et al.* 2003; Schmidt & Solar 2010), though these perform no better than conventional pitfalls (Pacheco & Vasconcelos 2012).

Use of a barrier fence consistently increases the number of ground beetles collected (Winder *et al.* 2001; Hansen & New 2005). However, the length of the fence influences trap catch (Durkis & Reeves 1982; Morrill *et al.* 1990), with longer fences catching higher diversity of families and species (Brennan *et al.* 2005), making it difficult to compare trap catch between studies. Location and number of the traps along the fence and fence material may also affect trap catch, though these variables have not been specifically investigated.

Spacing between traps is an important consideration as populations, especially of larger taxa such as carabids, can become locally depleted if traps are placed closely together; this can affect trap catch and skew results. Snider and Snider (1986) found no difference in trap catch between pitfalls spaced 0.5, 1, 2, and 4 meters apart. Similarly, Ward *et al.* (2001) found no difference in trap catch between pitfalls spaced 1, 5, and 10 meters apart. However, Digweed *et al.* (1995) found that carabid populations were depleted when pitfalls were placed 10 meters apart but not 25 meters; in addition, traps spaced at 10 meters had the most similar species assemblages and fewest rare species.

The optimum number of pitfall traps depends on the environment of the trapping site. As few as five traps are sufficient in an arid steppe environment (Cheli & Corley 2010), whereas ten to twenty pitfall traps effectively collected the majority of species in temperate areas (Formicidae: Santos *et al.* 2003; Coleoptera: Obrtel 1971; Isopoda Paoletti and Hassall 1999; Araneae: Niemelä *et al.* 1986), and at least twenty five are needed in tropical areas (Vennila & Rajagopal 1999). Various non-parametric estimators have been tested to estimate species richness based on as few as five traps per site (Brose 2002).

Finally, pitfall traps may not be the most efficient method for sampling epigeal arthropods in environments with rugged, steep slopes and a high density of rocks or roots in the

soil where the traps are difficult to set or at high elevation where the mean body size of taxa is generally smaller, and thus more difficult to trap (Nyundo & Yarro 2007). Additionally, some studies have found pitfalls trap more ants in drier areas and seasons (Delsinne *et al.* 2008; Nunes *et al.* 2011), though others have found annual rainfall has no effect (Delsinne *et al.* 2010).

Use of attractants in pitfall traps. The choice of preservative can affect the taxa collected in pitfall traps (Weeks & McIntyre 1997). For instance, bark beetles (Curculionidae: Scolytinae), certain Staphylinidae, and Nitidulidae are caught in higher numbers in pitfalls that use ethanol as the preservative (Drift 1963; Greenslade & Greenslade 1971). In one study, some Carabidae, especially *Bembidion*, were caught in higher numbers in ethylene glycol than water, though the effect varied by sex and time of year (Holopainen 1990, 1992); another study, however, found no difference between ethylene glycol and water when trapping four species of Diplopoda, one species of Chilopoda, and two species of Carabidae (Gerlach *et al.* 2009), suggesting that any effect is species dependent. Formaldehyde has been found to be repellant to Opiliones and Diplopoda and attractive to Carabidae and Staphylinidae (Luff 1968; Pekár 2002; Gerlach *et al.* 2009), though one study found no difference between water and formaldehyde when collecting Carabidae (Waage 1985). Differences have been found between commercially available antifreeze and diluted ethylene glycol (Koivula *et al.* 2003). Efficacy of preservatives can vary with trap size – one study found vinegar to be more effective in large traps but propylene glycol more effective in small traps (Koivula *et al.* 2003). Brine and an ethanol-glycerin mix have lower capture efficiency than other fluids such as pure water, ethanol-water, and ethylene glycol-water, possibly due to the high specific gravities of these fluids, which may allow captured arthropods to float and escape (Schmidt *et al.* 2006). Brine is also attractive to Lepidoptera (Cheli & Corley 2010). Additionally, attraction and repulsion to preservatives can

vary due to sex (Adis 1976), season (Dethier 1947; Adis & Kramer 1975; Adis 1976), and environment (Koivula *et al.* 2003). Thus, careful consideration should thus be used in order to avoid or account for the influence of preservative on the taxa collected.

A drop of detergent is often used to break the surface tension of the preservative in wet pitfalls. This does not seem to affect the rate of capture of most arthropods, though Linyphiidae are caught in higher numbers (up to 1000%) in traps with detergent (Topping & Luff 1995; Pekár 2002), whereas Staphylinidae are caught in higher numbers in traps without detergent (Pekár 2002).

Some Coleoptera naturally aggregate using pheromones to locate conspecifics (Greenslade 1963; Wautier 1970, 1971; Ahearn 1971), which can affect trap catch distribution as the first specimen captured may artificially attract others to the same trap (Luff 1968; Thomas & Sleeper 1977; Luff 1986).

Digging-in effects have been recorded among Formicidae (Greenslade 1973), Carabidae (Digweed *et al.* 1995; Schirmel *et al.* 2010) and other Coleoptera (Schirmel *et al.* 2010), Collembola (Joosse-van Damme 1965; Joosse & Kapteijn 1968), Linyphiidae and other Aranaea (Topping & Luff 1995; Schirmel *et al.* 2010), and Isopoda (Schirmel *et al.* 2010). These effects consist of high capture of certain taxa immediately after pitfall traps are established followed by a subsequent decline. A variety of explanations – such as an increased level of CO₂ (Collembola: Joosse & Kapteijn 1968), decreased barriers to movement (Carabidae: Greenslade 1964), increased number of prey that attract predators (Adis 1979), and decreasing number of foraging Formicidae workers (Romero & Jaffee 1989) – have been suggested, though no consensus has been reached. If digging-in effects are to be avoided, it has been suggested either to place pitfalls inverted for one week before operating them as traps (Greenslade 1973; Schirmel

et al. 2010) or to install a tube or second container in which the pitfall can be placed in order to avoid disturbing the soil when it is serviced (Schirmel *et al.* 2010). Alternatively, if the goal is to catch large numbers of arthropods without regard to comparing between-trap catch, traps may be serviced more frequently in order to take advantage of digging-in effects (Schirmel *et al.* 2010).

Disturbance of leaf litter and vegetation around the traps can cause increased catch of highly mobile taxa, such as Gryllidae (Sperber *et al.* 2007). Areas around active pitfalls should therefore not be visited unless the traps are being serviced. Alternatively, regularly scheduled visits to the trap area will increase the catch of certain mobile taxa, though care should be taken in designing and executing such visits in order to provoke the same disturbance between traps (Sperber *et al.* 2007).

If attraction is desired, baits can be used to purposely affect the taxa collected (Greenslade & Greenslade 1971). Dung and carrion can be used to collect Scarabaeidae, Staphylinidae, Silphidae, Ptiliidae, Histeridae, Hydrophilidae, and Leiodidae. Carnivore and omnivore dung provide good results – with human dung being among the most effective and readily available – while herbivore dung is generally poor (Newton & Peck 1975). Meat, tuna, and honey can be used as baits for ants (Romero & Jaffee 1989). Though not intentional, previously trapped insects may begin to rot in traps in which the preservative is ineffective due to dilution from rain or large numbers of trapped insects, thus attracting carrion feeding taxa (Holland & Reynolds 2005). Vegetable oils have been shown to increase the catch of ants in the tropics (Pacheco & Vasconcelos 2012), especially army ants (Weissflog *et al.* 2000; Berghoff *et al.* 2002; Berghoff *et al.* 2003), although this has not been studied in temperate regions.

Pests of pitfall traps. Occasionally, traps will be regularly disturbed by mammals between collections. Van der Berge (1992) presented three situations with the possible culprits and

associated solutions. For traps where the cup is still in the hole but pushed up “just enough so that the rim is no longer flush with the soil” he suggests moles or voles whose passage has been obstructed are to blame and moving the cup a short distance usually resolves the problem. When one or a few cups, but not the entire trap line, are completely out of the hole, spilled clean, but not chewed on he suggests squirrels are attempting to bury or dig up nuts. Unfortunately, “one is helpless against squirrel disturbance”. The third case is when many, and often the whole line, of cups are out of the hole and chewed or mangled. This, he suggests, is the work of raccoons, opossums or deer that are interested in consuming the preservative. Raccoons are intelligent and will continue to harass a line of pitfall traps if they are reset, so it is best to abandon the line or add a distasteful substance to the preservative. If deer are molesting the traps, it is best to switch from a salt-based preservative which is probably drawing their attention.

Preservatives.

Pitfall traps can be used to collect insects to be kept alive or killed in preservative. If live specimens are required, such as for rearing experiments (as is common in parasitengone mites to correlate life stages) or in cases where the taxon of interest is endangered, e.g. the American burying beetle (*Nicrophorus americanus* (Olivier, 1790)), traps are run dry without preservative. In such cases, traps must be checked at least daily, and often more frequently, so captured individuals do not succumb to heat, desiccate, drown in accumulated rain water, or become predated on by other captured organisms (Mitchell 1963; Luff 1968; Weeks & McIntyre 1997; Bestelmeyer *et al.* 2000; Moreau *et al.* 2013).

When collecting specimens to be killed, the choice of trap preservative is an important consideration as it will affect the quality of specimens, cost of trap maintenance, and how

frequently traps must be serviced. Many authors have investigated the preservation properties of different chemicals and solutions, which are summarized herein.

Ethylene glycol was once used as a preservative, especially in pitfall and pan traps, as it has low volatility compared to ethanol and other alcohols (Martin 1977), is relatively inexpensive, and is readily available as antifreeze. When used in the field it has been reported to not preserve internal organs well and causes specimens to deteriorate to the point of breaking when pinned (Aristophanous 2010), though other studies report sufficient preservation (Sasakawa 2007; Cheli & Corley 2010). Because ethylene glycol is toxic to vertebrates (Thrall *et al.* 1984) and is readily ingested due to its sweet taste (Grauer & Thrall 1982), its use has been discouraged (Hall 1991).

The addition of bitter agents, such as quinine, to ethylene glycol has been suggested as a way to deter vertebrates from drinking the fluid (Hall 1991). Quinine added to ethylene glycol, propylene glycol, and formalin has been shown to have no effect on the number of spiders caught in pitfall traps; in addition, it improves the preservation quality of specimens collected in ethylene glycol (Jud & Schmidt-Entling 2008). Alternatively, a red marking flag placed next to the trap may deter large vertebrates from investigating the trap and drinking the ethylene glycol (Cheli & Corley 2010).

An alternative to ethylene glycol but with similar characteristics is propylene glycol, which is sold as recreational vehicle and boat antifreeze. It also has low volatility and is inexpensive. Propylene glycol is nearly non-toxic as it is metabolized into constituents of the Krebb's cycle and extremely large quantities must be ingested over a short period of time before acute toxicity is reached (Yu 2007). In the field, propylene glycol preserves insects similarly to ethylene glycol (Jud & Schmidt-Engling 2008; Aristophanous 2010). However, Moreau *et al.*

(2013) found no detectable difference in the quality of DNA preservation between propylene glycol and ethanol when undiluted chemicals were used in a lab setting. One reason for the difference between field and lab studies may be due to the fact that ethylene glycol and propylene glycol are hygroscopic; when humidity is moderate to high, both substances will absorb water from the air and dilute naturally (Aristophanous 2010).

Salt brine and saturated borax solution are inexpensive and easy to make as the constituent materials are readily available in grocery stores. The ability of these solutions to preserve insects is extremely poor, however, and not outweighed by cost-savings (Lemieux & Lindgren 1999; Sasakawa 2007; Aristophanous 2010) (though see Schmidt *et al.* 2006 for a counter opinion).

Carnoy's fixative (60% ethanol, 30% chloroform, 10% acetic acid) and white vinegar (10% acetic acid) do not preserve DNA and cause specimens to become brittle, though they generally keep the specimens from rotting (Sasakawa 2007; Aristophanous 2010; Moreau *et al.* 2013). If DNA extraction is not intended, these may be acceptable preservatives.

Methanol and chloroform do not preserve specimens in a way that allows DNA extraction and amplification (Post *et al.* 1993; Fukatsu 1999). In addition, chloroform is difficult to acquire, especially in the large quantities required for use as a trap preservative.

FAACC solution (formaldehyde 4%, acetic acid 5%, calcium chloride 1.3%) and 4% phosphate buffered formaldehyde (4%PBF) both preserve internal organs well, with 4%PBF being the superior of the two (Aristophanous 2010). However, specimens become excessively stiff and although DNA can be extracted from specimens preserved with formaldehyde solutions, DNA amplification is impossible with standard kits (such a Qiagen DNEasy) because formaldehyde causes DNA to cross-link with proteins (Schander & Halanych 2003). Protocols

using prolonged extraction times (up to 7 days) (France & Kocher 1996; Chatigny 2000; Schander & Halanych 2003) and chemical agents (Johnson *et al.* 1995; Chatigny 2000) can be successful.

Amyl acetate is sometimes used in insect jars as the killing agent. This banana-smelling liquid keeps specimens relaxed, unlike other killing agents such as chloroform (Woodward 1951). It is commonly used as a water-removing solvent in industry and can be purchased through specialized suppliers. Amyl acetate has been used for preservation of anatomical dissections (Saunders & Rice 1944) and insects “may be kept stored almost indefinitely between cotton-wool impregnated with this agent” (Woodward 1951), though it has not been tested for DNA preservation (Nagy 2010). Additionally, it has not been tested as a preservative in pitfall traps, can be a skin irritant, and is probably attractive to some insect groups so other, more proven preservatives may be a better choice.

Ethanol is probably the most widely used preservative. It maintains the integrity of internal organs and allows DNA to be easily extracted and amplified (Gurdebeke & Maelfait 2002; Aristophanous 2010; Moreau *et al.* 2013). In the United States, price may be prohibitive for individuals who do not qualify for ethanol tax exemption; however, fuel ethanol has been shown to preserve specimens as well as pure ethanol, so this will provide an alternative source as fuel ethanol becomes more widespread (Szinwelski *et al.* 2012). In addition, ethanol is the most volatile commonly used preservative. In open containers such as pitfall traps ethanol can lose $\frac{3}{4}$ of its volume in fewer than 5 days (Aristophanous 2010). Depending on the trap location this may have implications on how often the traps must be serviced.

Isopropanol, commonly known as rubbing alcohol, is a cheap alternative to ethanol. Similar to ethanol, it preserves DNA well (Rake 1972), so it can be extracted with little

difficulty. One drawback is that isopropanol often discolors specimens, which is a hindrance to identification and morphological studies involving color.

Acetone has shown promise as a preservative. It is relatively inexpensive and readily available as a paint solvent. DNA has been extracted and successfully amplified from acetone-preserved Copepods (Goetze & Jungbluth 2013), pea aphid (*Acyrtosiphon pisum* (Harris, 1776)) (Fukatsu 1999), and Zygoptera (Logan 1999). Additionally, acetone is used to preserve adult Odonata as it dissolves fat, dehydrates the specimen, and reduces decomposition of enzymatic color pigments (Abbott 2008).

Other preservatives require more testing as contradictory results have been reported. Fukatsu (1999) reported DNA amplification after specimens were stored in 2-propanol, ethyl acetate, and diethyl ether, though Post *et al.* (1993) and Reiss *et al.* (1995) reported poor results with 2-propanol and ethyl acetate, respectively.

Summary.

Pitfall traps are often used to sample epigeal arthropods as they are inexpensive and easy to use. However, many factors influence the taxa so collected. Abiotic factors, such as weather, season, slope and aspect, degree of rockiness, and trap characteristics (color and material of the trap, diameter of the opening, spacing between traps, and number of traps at a site) affect the composition of collected taxa, often by affecting behavior of the target arthropods. Biotic factors affecting trap catch include species-specific factors (activity level, size, aggregation to conspecifics, and behavior at the edge of the trap), response to digging-in effects, and habitat structure, including the density of low-growing vegetation. The choice of preservative affects not only the level of preservation of specimens, but also the composition of specimens collected

because various compounds differentially repel and attract different taxa. Taken together, these factors make comparisons between studies difficult.

While there have been calls to standardize pitfall trapping, the design employed in individual studies will continue to be based on the research question and materials available. An effort, however, should be made to report all of the factors that might influence the composition of specimens collected. While this may not be immediately useful, comparisons may be made in the future after further studies elucidate the effects various factors have upon trap catch.

Acknowledgements.

We thank the reviewers for their helpful suggestions; this manuscript is a better product because of them.

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III. A review of terrestrial and canopy Malaise traps

“Since the time of Linnaeus the techniques of catching insects has not improved very much.”

– René Malaise, 1937.

Abstract.

An extensive review of the history and literature concerning Malaise and canopy traps is given. Factors that affect trap catch, including trap design and placement, as well as different uses of the traps are discussed. Both trap styles are compared to each other and other types of arthropod traps.

Introduction.

Malaise traps – which are large, tent-like structures made of fine mesh netting – are one of the most widely-used non-attractant, static insect traps (Muirhead-Thomson 1991). Flying insects, especially Diptera and Hymenoptera, are passively intercepted by the mesh walls; many species, after encountering the mesh wall, climb up and are funneled into a collecting container (Zililhona *et al.* 1998; Achterberg 2009).

Herein we use “Malaise trap” to refer to specifically to terrestrial Malaise traps (e.g., those traps set near, or in contact with the ground or over streams) and “canopy trap” to refer to those traps suspended at considerable height above the ground, generally in the forest canopy. While Malaise and canopy traps are based on the same design and Malaise traps set in different environments (e.g., field, forest, over streams) may sample diversity as different as that sampled by Malaise and canopy traps, we make the distinction between Malaise and canopy traps herein as such a distinction is made in the published literature.

History.

René Malaise (1892–1978) was inspired to invent a new type of insect trap after watching insects fly into a tent and become trapped despite the open flaps. His design consisted of mesh fabric stretched over a wooden box frame open at one end with a collection cylinder at the top (Fig. 1) (Malaise 1937). It revolutionized the collection of flying insects.

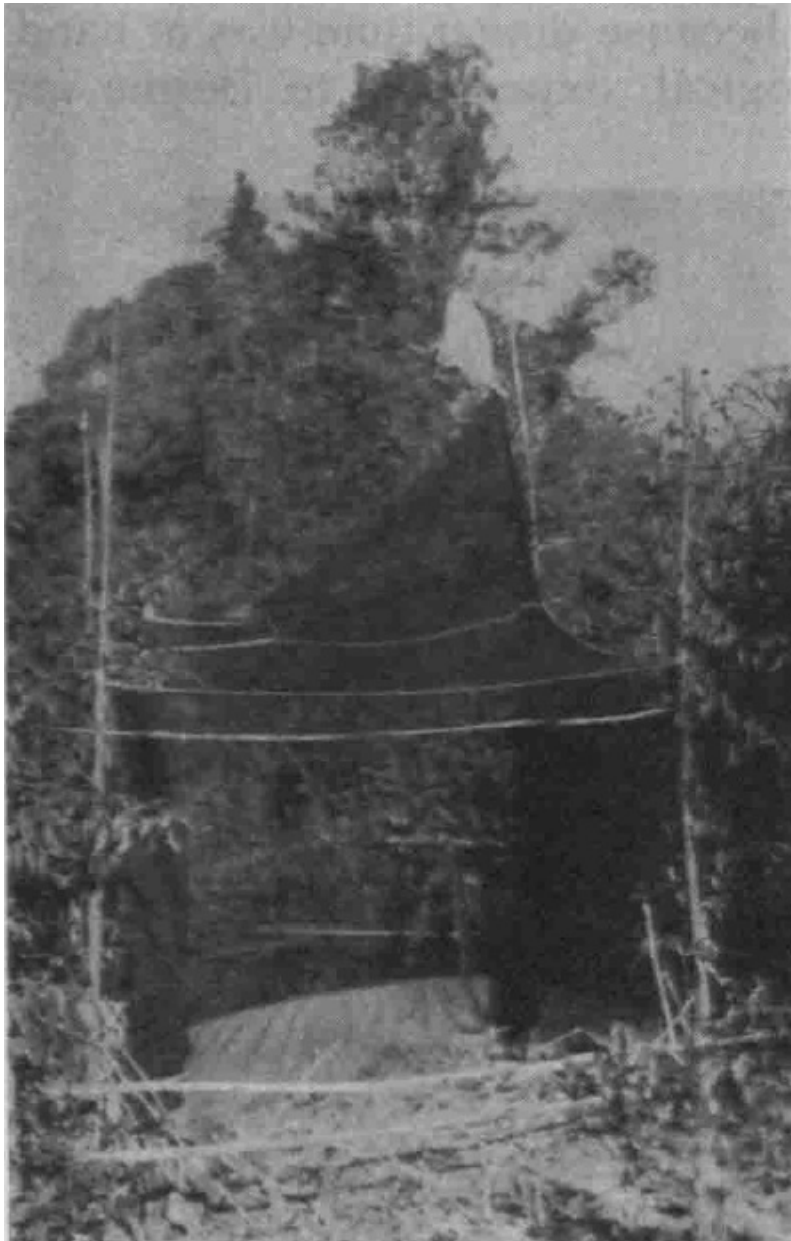


Figure 1. Malaise's original trap. After Malaise (1937).

Vecht (1939) translated Malaise's design into Dutch after successfully testing it in Burma.

Henry Townes (1913–1990) elaborated upon Malaise's design and presented a modified version of the trap at the 1959 Annual Meeting of the Entomological Society of America. After “many requests for plans of the trap” he published schematics for his design, though was worried the publication might “freeze the model at this stage of development rather than encourage further experimentation with it” (Townes 1962). His fear may be correct in part: the most widely-available and commonly used Malaise traps are only slightly modified from Townes' design.

Marston (1965) proposed improvements to Townes' design, including the use of a prefabricated tubular aluminum frame instead of using a wooden frame (Fig. 2). Móczár (1967) proposed additional modifications to Marston's to lighten Townes' design. Townes (1972), after possibly reading of this improvement and realizing that a 13.5 pound trap was much too heavy, designed a light-weight Malaise trap (Fig. 3).

Others continued to experiment with the design of Townes' Malaise trap. Schroeder *et al.* (1975) proposed a more durable design that used a metal frame instead of wood and bronze screen funnel instead of plastic for use on windswept rangelands. Masner and Goulet (1981) noticed some Hymenoptera do not readily climb up the mesh into the collector, so designed a Malaise-type trap impregnated with fast-acting insecticide and a collecting trough underneath. Hutcheson (1991) suggested a modified collection jar to facilitate easy servicing. Achterberg (2009) suggested further improvements, including angling the entrance of the collection jar at 45° instead of horizontally as is the case in commercial designs; he also provided an excellent overview of various Malaise trap designs.



Figure 2. Large Malaise trap utilizing a prefabricated aluminum frame. After Marston (1965). Used with permission.



Figure 3. Townes' light-weight Malaise trap. After Townes (1972). Used with permission.

Gressit and Gressit (1962) introduced a variation of the Malaise trap that consists of a large (7m long by 3.6m high) sheet of fabric with collectors at either end. The fabric is supported between two poles or trees (Fig. 4). Townes (1962) commented on Gressit & Gressit's trap, saying their "design is basically a good one and merits further development" and that compared to his trap their design "is much more portable and easier to make, but is possibly less efficient for some kinds of insects." This is perhaps less true now that collapsible fiberglass poles are used in commercial Townes-style Malaise traps; however, the Gressit and Gressit design is reported to be effective and warrants further study.

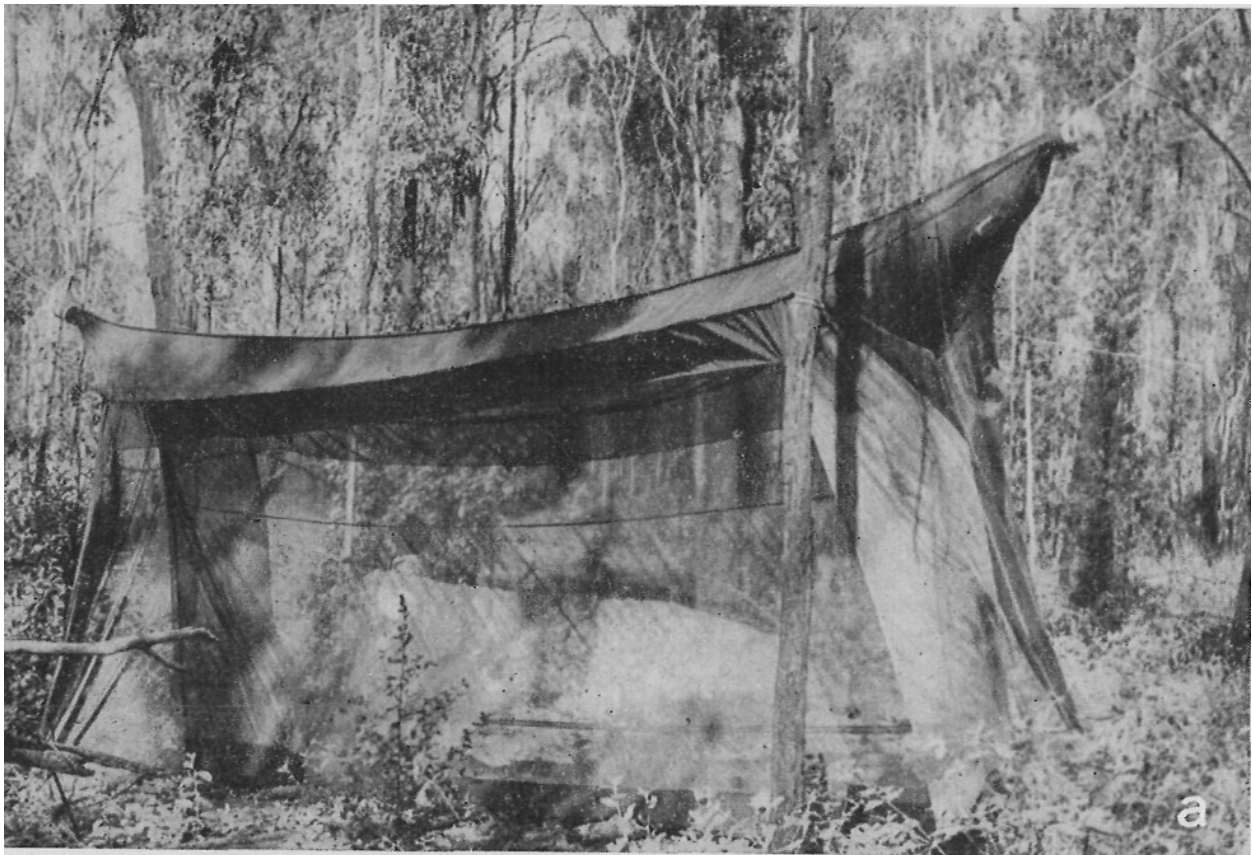


Figure 4. Gressit and Gressit-style Malaise trap. After Gressit and Gressit (1962). Used with permission courtesy of the Bishop Museum.

Butler (1965) proposed a design which consists of a bed net with a hole cut in the roof and a hole cut in the side of one wall. A collecting trap consisting of a metal cylinder and

polythene bag is placed in the hole in the roof. Butler (1966) modified this design for use in crop areas and reported a trapping rate of 370-450 insects/trap/day.

Blotzhofer and Riggs (1998) suggested changes to the standard Townes lightweight Malaise trap for trapping live Odonata.

Malaise traps with four fabric panes open 360° were originally proposed for use in rice paddies (Nishida & Torii 1970; Yano *et al.* 1975).

Various methods have been described for using Malaise traps to sample canopy arthropods. Some researchers have attached standard Malaise traps to tall scaffolding or platforms constructed in the canopy (Coulson *et al.* 1971; Crossley *et al.* 1973; Southwood *et al.* 1979). Others tied Townes-style Malaise traps off to a wooden frame and used ropes and pulleys in order to raise the structure into the canopy (Hammond 1990; Faulds and Crabtree 1995; Basset *et al.* 1997).

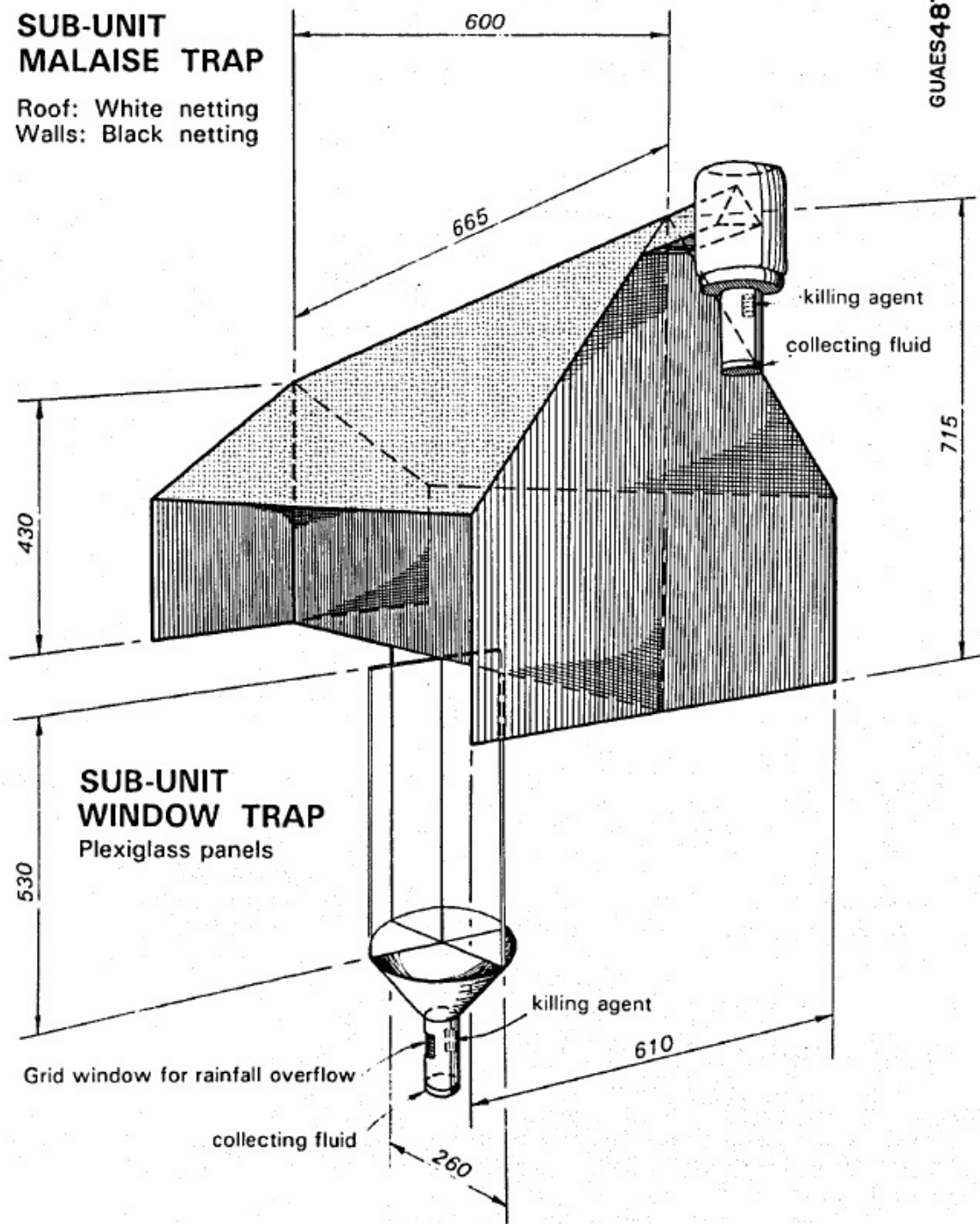
Murchie *et al.* (2001) described a rotary device that segregates Malaise trap catch into two-hour time intervals.

Malaise traps have been combined with other traps, including light traps (Dufour 1980), window traps (Basset 1988) (Fig. 5), and intercept traps, including colored pan traps, in order to modify taxa collected or increase efficiency. The addition of yellow pan traps beneath Malaise traps increases the trap effectiveness in catching Diptera, Hymenoptera, Heteroptera, and Thysanoptera (Darling & Packer 1988; Campos *et al.* 2000).

Steyskal (1981) provided an extensive bibliography on Malaise trap research.

SUB-UNIT MALAISE TRAP

Roof: White netting
Walls: Black netting



GUAES487090

Figure 5. Diagram of a combined Malaise/window trap. After Basset (1988). Used with permission.

Taxa collected.

Diptera and Hymenoptera are generally the numerically dominant taxa in Malaise traps, with Diptera often representing the largest percentage (Table 1). Because of this, Malaise traps are often used to survey diversity of and collect Diptera in general; however, they have also been used to specifically collect many taxa, including “Nematocera” Tabanidae, Syrphidae, Tachinidae, Oestridae, and Tephritidae (Table 2). Hymenoptera are generally the second most-collected taxa, though usually represent a much smaller percentage of the total catch than flies. As in Diptera, Malaise traps are often used to survey hymenopteran diversity in general, but have also been used to collect specific taxa (Table 3). Besides Diptera and Hymenoptera, Malaise traps have been used to collect a variety of Arachnida, Odonata, Coleoptera, Lepidoptera, and other insects (Table 4).

Canopy traps have been used to collect Psocoptera, Thysanoptera, Coleoptera, Diptera, Lepidoptera, Neuroptera, and Hymenoptera (Table 5).

Malaise traps have been shown to be some of the most consistent traps in terms of the composition of higher taxa collected, giving credence to the confidence hymenopterists and dipterists have that they will invariably collect those taxa (Kitching *et al.* 2001). While such consistency has not been studied in canopy traps, there is no reason to believe they do not sample similar groups irrespective of site locality.

Citation	Diptera (%)	Hymenoptera (%)	Lepidoptera (%)	Coleoptera (%)	Hemiptera (%)	Plecoptera (%)	Collembola (%)	"Other" (%)
Marston 1965	66.3	12.3	4.8	5.6	6.8	-	3.4	4.2
Geijskes 1968	58	19	14	4.6	2.3	-	-	2.1
Matthews & Matthews 1969	52.2	16.9	7.1	3.1	9.4	8.3	1.7	11.3
Matthews & Matthews 1969	43.1	22.5	10	2	3.9	16.8	1.2	18.5
Matthews & Matthews 1969	14.7	1.2	0.8	0.3	0	76.9	0.7	83
Matthews & Matthews 1969	54.4	13	15.5	2.6	10.9	0.3	0.6	3.6
Yano <i>et al.</i> 1975	69.7	4.6	4.6	0.8	13.6	0	3.2	6.7
Rose 1978	84.2	5.85	3.52	0.11	3.74	-	-	2.58
Cooksey & Barton 1981	57	15	17	-	8	-	-	3
Moeed & Meads 1987	84.2	4	1.8	2.3	-	-	4.9	2.8
Basset 1988	47.8	8.1	3.7	10.3	19.5	-	-	10.7
Basset & Arthington 1992	-	-	-	-	-	-	-	-
Dutra & Marinoni 1994	85.1	3.2	5.7	1.7	2.5	<0.01	0.9	2
Dutra & Marinoni 1994	72	3.3	13.5	2.6	1.4	0	5.3	7
Campos <i>et al.</i> 2000	84.4	7.8	3.2	0.8	2.9	-	-	0.9
Campos <i>et al.</i> 2000	64.3	10	1.3	7	10	-	-	7.4
Campos <i>et al.</i> 2000	57.4	16.5	0.9	5.3	12.2	-	-	7.7
Hughes <i>et al.</i> 2000	69.2	14.3	-	-	-	-	-	16.5
Kowk & Corlett 2002	80.6	3.9	5.3	6.3	-	-	-	3.9
Brown 2005	84	-	-	-	-	-	-	-
Brown 2005	81	-	-	-	-	-	-	-
Brown 2005	64	-	-	-	-	-	-	-
Horn <i>et al.</i> 2005	68.5	4.1	9.5	7.6	-	-	-	10.2
Horn <i>et al.</i> 2005	26.6	7.3	30.8	18.1	-	-	-	17.2

Table 1. Summary of Malaise trapping studies and taxa collected.

Citation	Number of specimens collected	Duration per trap (days)	Number of traps	Specimens/ trap/day	Locality	Trap type
Marston 1965	2927	7	1	418.14	Kansas, USA	Malaise
Geijskes 1968	90,182	-	-	-	Suriname	Malaise
Matthews & Matthews 1969	23,722	91	1	260.68	New York, USA	Malaise
Matthews & Matthews 1969	6138	91	1	67.45	New York, USA	Malaise
Matthews & Matthews 1969	7008	91	1	77.01	New York, USA	Malaise
Matthews & Matthews 1969	3480	91	1	38.24	New York, USA	Malaise
Yano <i>et al.</i> 1975	13,709	3.34	19	216.03	Thailand/China	Malaise
Rose 1978	37,198	127	6	48.82	Malaysia	Malaise/canopy
Cooksey & Barton 1981	10,830	12	1	902.50	Arkansas, USA	Malaise
Moeed & Meads 1987	45,965	365	1	125.93	New Zealand	Malaise
Basset 1988	14,597	365	5	8.00	Queensland, Australia	Composite
Basset & Arthington 1992	46,019	730	5	12.61	Queensland, Australia	Composite
Dutra & Marinoni 1994	62,924	365	1	172.39	Parana, Brazil	Malaise
Dutra & Marinoni 1994	38,868	365	1	106.49	Parana, Brazil	Malaise
Campos <i>et al.</i> 2000	6,120	14	4	109.29	Minas Gerais, Brazil	Malaise
Campos <i>et al.</i> 2000	2,436	14	4	43.50	Minas Gerais, Brazil	Malaise
Campos <i>et al.</i> 2000	4,816	14	4	86.00	Minas Gerais, Brazil	Malaise
Hughes <i>et al.</i> 2000	12,776	60	3	70.98	Colorado, USA	Malaise
Kowk & Corlett 2002	53,897	940	4	14.33	Hong Kong, China	Malaise
Brown 2005	4,646	5	1	929.20	Tambopata, Peru	Malaise
Brown 2005	905	4	1	226.25	Tambopata, Peru	Malaise
Brown 2005	1,064	3	1	354.67	Puntarenas, Costa Rica	Malaise
Horn <i>et al.</i> 2005	-	-	5	-	South Carolina, USA	Canopy
Horn <i>et al.</i> 2005	-	-	5	-	South Carolina, USA	Canopy

Table 1 (Cont.). Summary of Malaise trapping studies and taxa collected.

Taxon	Reference
General Diptera	Kitching <i>et al.</i> 2004; Roháček & Ševčík 2009
"Nematocera"	Salmela <i>et al.</i> 2007
Tipulidae	Dufour 1980; Toft & Beggs 1995; Peterson <i>et al.</i> 2004
Sciaridae	Steffan 1972; Vilkamaa <i>et al.</i> 2007
Mycetophilidae	Økland 1994; Toft <i>et al.</i> 2001; Toft & Chandler 2004; Jakovlev & Penttinen 2007
Culicidae	Graham 1969; Witter <i>et al.</i> 2012
Simuliidae	Adler <i>et al.</i> 1983; Currie & Adler 2000; Witter <i>et al.</i> 2012
Psychodidae	Quate 1999; Alexander 2000; Alexander <i>et al.</i> 2001
Tabanidae	Strickler & Walker 1993
Syrphidae	Burgio & Sommaggio 2002; Thompson & Zumbado 2002; Krčmar <i>et al.</i> 2005; Gittings <i>et al.</i> 2006; Nol <i>et al.</i> 2006; Smith <i>et al.</i> 2008; Whitemore <i>et al.</i> 2008; Birtele & Hardersen 2012
Dolichopodidae	Pollet <i>et al.</i> 1989
Agromyzidae	Scheirs <i>et al.</i> 1997
Stratiomyiidae	Hauser 2008; Whitemore <i>et al.</i> 2008; Birtele & Hardersen 2012
Calliphoridae	Rosati & VanLaerhoven 2007
Sarcophagidae	Dahlem & Downes 1996; Whitemore <i>et al.</i> 2008
Tachinidae	Cerretti <i>et al.</i> 2004; Stireman <i>et al.</i> 2012
Oestridae	Capelle 1970; Cogley & Cogley 2000; Fleenor & Taber 2007; Witter <i>et al.</i> 2012
Axiniidae	Colless 1994
Tephritidae	Asquith & Kido 1994
Pipunculidae	Skevington 2001

Table 2. Diptera families collected in Malaise traps.

Taxon	Reference
General Hymenoptera "Symphyta"	Darling & Packer 1988; Noyes 1989; Campos <i>et al.</i> 2000; Shlyakhtenok 2000; Sobek <i>et al.</i> 2009 Holuša 2002; Brand <i>et al.</i> 2003; Harris 2006
Apocrita	Karem <i>et al.</i> 2006
Evaniidae	Deans & Kawada 2008
Embolemidae	Amarante <i>et al.</i> 1999
Plumariidae	Penteado-Dias & Scatolini 2003
Chrysididae	Shlyakhtenok 2000; Strumia 2003
Diapriidae	Masner 1976a,b
Ichneumonidae	Noyes 1989; Bartlett <i>et al.</i> 1999 Sääksjärvi <i>et al.</i> 2004; Sperber <i>et al.</i> 2004; Sääksjärvi <i>et al.</i> 2006; Ulber & Nitzsche 2006; Veijalainen <i>et al.</i> 2013
Braconidae	Shimbori & Shaw 2014
Mymaridae	Vance <i>et al.</i> 2007
Platygastridae	Stevens & Austin 2007; Burks <i>et al.</i> 2013
Formicidae (especially alates)	Collingwood 1981; Deyrup & Trager 1986; Longino & Colwell 1997; Delabie & Reis 2000; Kaspari <i>et al.</i> 2001; Collingwood & van Harten 2005; Colby & Prowell 2006; Framenau & Thomas 2008; Fisher <i>et al.</i> 2009; Guerrero <i>et al.</i> 2010
Pompilidae	Shlyakhtenok 2000
Vespidae	Archer 1990; Beggs <i>et al.</i> 1998; Beggs & Rees 1999; Shlyakhtenok 2000; Sackmann <i>et al.</i> 2001
Mutillidae (especially males)	Pitts <i>et al.</i> 2004; Pilgrim & Pitts 2006

Table 3. Hymenoptera families collected in Malaise traps

Taxon	Reference
Araeneae	Wilkinson <i>et al.</i> 1980; Hauge & Midtgaard 1986; Jennings & Hilburn 1988; Oxbrough <i>et al.</i> 2010; Vedel <i>et al.</i> 2011
Acari	Bo-yi 1996; Clark 2004; Behan-Pelletier & Winchester 2008; Bo-yi 2008 a, b; Ripka & Szabó 2010; Skvarla <i>et al.</i> 2014
Opiliones	Hicks <i>et al.</i> 2003
Pseudoscorpiones	Aguiar & Buhrnheim 1998
Collembola	Fjellberg 1992
Odonata	Muzón & Spinelli 1995; Roble 1995; Flint 1996; Glotzhober & Riggs 1998
Ephemeroptera	Peterson <i>et al.</i> 2004
Orthoptera	Samways & Moore 1991; Quinn <i>et al.</i> 1993; Donnelly 1995; Johnson <i>et al.</i> 1995; Muzon & Spinelli 1995; Bomar 2001
Plecoptera	Peterson <i>et al.</i> 2004; Winterbourn 2005
Hemiptera	Cancelado & Yonke 1970; Hodgkinson & Casson 1991
Thysanoptera	Olsen & Midtgaard 1996
Coleoptera	Hosking 1979; Hutcheson 1999; Hutcheson & Kimberley 1999; Harris <i>et al.</i> 2000; Toft <i>et al.</i> 2001; Grimbacher & Stork 2007; Stork <i>et al.</i> 2008; Ohsawa 2010
Carabidae	Liebherr & Mahar 1979; Ulyshen <i>et al.</i> 2005; Ulyshen <i>et al.</i> 2006; Cassola 2009; Meng <i>et al.</i> 2012
Eucnemidae	Hoffman <i>et al.</i> 2009
Elateridae	Steiner 2000; Nol <i>et al.</i> 2006
Lampyridae	Barrows <i>et al.</i> 2008
Mordellidae	Jackman & Nelson 1995
Buprestidae	Curletti & van Harten 2002
Cerambycidae	Noguera <i>et al.</i> 2002; Warriner <i>et al.</i> 2002; Vance <i>et al.</i> 2003; Schiefer & Newell 2010
Chrysomelidae	Schiefer 1998; Spencer <i>et al.</i> 1998; Spencer <i>et al.</i> 1999; Furth <i>et al.</i> 2003; Aslan <i>et al.</i> 2012
Scirtidae	Ruta 2011
Curculionidae	Dutcher <i>et al.</i> 1986; Deyrup & Atkinson 1987
Scolyinae	Atkinson <i>et al.</i> 1991
Platypodinae	
Staphylinidae	
Pselaphinae	Chandler 1987
Scarabaeoidea	Kriska & Young 2002

Table 4. Arthropod taxa, excluding Diptera and Hymenoptera, collected in Malaise traps.

Taxon	Reference
Neuroptera	Hollier & Belshaw 1992, 1993; Vas <i>et al.</i> 2001; Abraham <i>et al.</i> 2003
Mecoptera	Byers 1973
Lepidoptera	Owen 1969; Butler <i>et al.</i> 1999; Harris <i>et al.</i> 2004; Campbell 2007
Tortricidae	Eveleigh <i>et al.</i> 2007
Sesiidae	Steinbauer <i>et al.</i> 2000
Gracillariidae	Steinbauer <i>et al.</i> 2000
Trichoptera	Jones & Resh 1988; Sode & Wiberg-Larsen 1993; Peterson <i>et al.</i> 2004; Winterbourn 2007; Winterbourn <i>et al.</i> 2007

Table 4 (Cont.). Arthropod taxa, excluding Diptera and Hymenoptera, collected in Malaise traps.

Taxon	Reference
Psocoptera	Santos <i>et al.</i> 2007; Sokolova <i>et al.</i> 2010
Thysanoptera	Santos <i>et al.</i> 2007
	Tangmitcharoen <i>et al.</i> 2006; Hardersen <i>et al.</i> 2014
Coleoptera	
Cerambycidae	Vance <i>et al.</i> 2003; Dodds <i>et al.</i> 2010
Buprestidae	
Coccinellidae	Santos <i>et al.</i> 2007
Curculionidae,	
Scolytinae	Dodds <i>et al.</i> 2010
Diptera	Tangmitcharoen <i>et al.</i> 2006
Stratiomyidae	Whitemore <i>et al.</i> 2008
Syrphidae	Whitemore <i>et al.</i> 2008
Tachinidae	Cerretti <i>et al.</i> 2004
Sarcophagidae	Whitemore <i>et al.</i> 2008
Tephritidae	Asquith & Kido 1994
Lepidoptera	Tangmitcharoen <i>et al.</i> 2006
Tortricidae	Eveleigh <i>et al.</i> 2007
Zygaenidae	Hoddle 2006
Neuroptera	Hollier & Belshaw 1993
Hymenoptera	Tangmitcharoen <i>et al.</i> 2006

Table 5. Insect taxa collected in canopy traps.

Uses.

Malaise traps are excellent tools for surveying biodiversity, especially when used in conjunction with traps that collect non-overlapping assemblages of arthropods, such as pitfall traps (e.g., Peck 1989; Benton 1995; Winchester & Ring 1996; Handler 2007; Missa *et al.* 2009). They can also be used to sample a specific subset of biodiversity, such as natural enemies (Nishida & Torii 1970), or monitor specific species, such as pests or agents released for biological control (Steinbauer *et al.* 2000; Toft & Changler 2004).

Malaise and canopy traps have been used to investigate the arthropod community associated with specific habitats, such as specific tree species (Basset & Arthington 1992) tree fall gaps (Horn *et al.* 2005; Ozanne 2005; Nol *et al.* 2006; Ulyshen *et al.* 2006; Richard & Windsor 2007; Hiaro *et al.* 2008) and dead wood (Hutcheson & Jones 1999; Ozanne 2005). They have also been used to investigate differences between patches of similar habitat (Hutcheson & Jones 1999; Choi *et al.* 2010; Fraser *et al.* 2007; Fraser *et al.* 2008), differently treated patches of similar habitat (burning: Cancelado & Yonke 1970; Campbell *et al.* 2007; harvesting: Dean *et al.* 2005; Newell & King 2009; insecticide treatment: Dilling 2007; Dilling *et al.* 2007; Santos *et al.* 2007) and different habitats (Coulson *et al.* 1971; Crossley *et al.* 1973; Greiler & Tschamtk 1993; Bomar 2001; Hicks *et al.* 2003; Gittings *et al.* 2006; Tangmitcharoen *et al.* 2006; Cunningham & Murray 2007; Vance *et al.* 2007; Rohr *et al.* 2007; Smith *et al.* 2008; Rohr *et al.* 2009; Banks *et al.* 2010); community differences in monospecific and highly diverse tree canopies in agroforestry (Sperber *et al.* 2004); how arthropod communities change during plant succession (Hollier & Belshaw 1992; Hutcheson 1999; Shlyakhtenok & Agunovich 2001; Nol *et al.* 2006; Missa *et al.* 2009; Rohr *et al.* 2009) or stand growth (Hutcheson & Jones 1999), invasion by foreign plant species (Toft *et al.* 2001), or along an environmental (Harris *et al.* 2000; Vas *et al.*

2001; Lynch *et al.* 2002; Kato *et al.* 2004; Kitching *et al.* 2004; Karen *et al.* 2006; Hirao *et al.* 2008x; Carr 2010) or latitudinal gradient (Kitching *et al.* 2004; Veijalainen *et al.* 2013); differences between arthropod communities associated with different tree species (Basset *et al.* 1996); vertical stratification within a habitat (Roberts 1976a; Rose 1978; Hollier & Belshaw 1993; Asquith & Kido 1994; Hammon *et al.* 1997; Preisser *et al.* 1998; Charles & Basset 2005; Grimbacher & Stork 2007; Sobek *et al.* 2009; Ulyshen 2011; Birtele & Hardersen 2012); and attractiveness of flowers (Rohrig *et al.* 2008).

When operated for long periods of time (e.g., weeks to years), Malaise and canopy traps can be used to investigate meteorological variables affecting flight activity (Matthews & Matthews 1969; Burnett & Hays 1974; Nyrop & Simmons 1986; Isard *et al.* 1999; Briers & Cariss 2003; Witter *et al.* 2012) and diel (Rickleps 1975; Hammond 1990; Basset & Springate 1992; Springate & Basset 1996; Spencer *et al.* 1998; Isard *et al.* 2000; Murchie *et al.* 2001; Shlyakhtenok & Agunovich 2001) and seasonal or phenological cycles (Evans & Owen 1965; Rickleps 1975; Denlinger 1980; Wright *et al.* 1984; Elliott 1986; Hammond 1990; Hollier & Belshaw 1993; Dutra & Marinoni 1994; Ellis & Simor; Thomas 1994; Jackman & Nelson 1995; Toft & Beggs 1995; Flint 1996; Tereshkin 1996; Spencer *et al.* 1998; Kaspari *et al.* 2001; Shlyakhtenok & Agunovich 2001; Noguera *et al.* 2002; Hicks *et al.* 2003; Sperber *et al.* 2004; Maleque *et al.* 2006; Whitemore *et al.* 2008; Winterbourn 2005; Ulber & Nitzsche 2006; Eveleigh *et al.* 2007; Pinheiro *et al.* 2008; Choi *et al.* 2010).

Malaise and canopy traps can also be used to investigate insect movement, such as movement within and between habitat patches (Naranjo 1991; Spencer *et al.* 1999; Hossain *et al.* 2002; Briers *et al.* 2004; Gangurde 2007; Williams *et al.* 2007a; Macfadyen & Muller 2013), including into agricultural areas (Dutcher *et al.* 1986; Dyer & Landis 1997; Spencer *et al.* 1998;

Isard *et al.* 1999; Irwin *et al.* 2000; Nicholls *et al.* 2001; Ulber & Nitzsche 2006); flight patterns in relation to wind direction (Pruess & Pruess 1966; Isard *et al.* 1999) and mating (Abbott 2006); and movement and dispersal (Cooksey & Wright 1987), especially of adult aquatic insects (Buskirk 1975; Mendl & Müller 1979; Müller 1982; Jones & Resh 1988; Sode & Wiberg-Larsen 1993; Williams & Williams 1993; Griffith *et al.* 1998; Briers *et al.* 2004; Peterson *et al.* 2004; Winterbourn 2005; Solem & Bongard 2007; Winterbourn *et al.* 2007). However, caution should be used as mark-recapture studies have shown that instantaneous direction, which is indicated by the side of the trap insects are collected on, may not always be a reliable way to determine overall direction of movement between habitats or along gradients (Macneale *et al.* 2004).

Malaise and canopy traps can be used to estimate abundance individual species (Beggs *et al.* 1998) and establish damage thresholds (Beggs & Rees 1999) or create an index of abundance, availability, and biomass of aerial prey available to predators (Lynch *et al.* 2002; Araneae: Buskirk 1975; Kato *et al.* 2003; Kato *et al.* 2004; Odonata: Kirkton & Schultz 2001; Anura: Horn *et al.* 2005; Chiroptera: Jong & Ahlen 1991; Fukui *et al.* 2006; Aves: Poulin *et al.* 1992; Rodenhouse & Holmes 1992; Duguay *et al.* 1997; Duguay *et al.* 2000; Johnson & Sherry 2001; Kwok & Corlett 2002; Murakami & Nakano 2002; Iwata *et al.* 2003). Collected taxa can also be associated with specific habitats and used as habitat indicators (Fraser *et al.* 2007).

Malaise traps can also be used to collect specific guilds of insects, such as those attracted to corpses and potentially useful in forensic studies (De Jong 2010) and medically important species (Roberts 1971, 1972; Alexander 2000).

Trap setup.

Location of a trap affects the taxa collected (Ozanne 2005). Insects often follow specific flight paths through vegetation and a trap located along a flight path will catch more specimens

than one that is not (Matthews & Matthews 1983; Hutcheson 1990; Southwood & Henderson 2000). Traps set in sunny, exposed areas collect more insects than those in sheltered, shaded areas (Noyes 1989; Irvine & Woods 2007). Topography, wind, water, light, and other abiotic conditions should also be taken into consideration (Gressitt & Gressitt 1962; Richards & Windsor 2007). Additionally, some researchers have suggested setting traps in a north-south orientation with the trap head facing the sun's zenith (Noyes 1989).

While environmental factors have been little studied, Matthews and Matthews (1969) reported temperature and precipitation had a strong influence on trap catch, with the largest catches happening on hot, sunny days following rain.

Few studies have investigated how many traps are required to effectively sample a given area. Two that focused on parasitoid wasps found that species accumulation curves failed to reach an asymptote even after sixteen and twenty seven traps were operated after multiple months (Sääksjärvi *et al.* 2004; Fraser *et al.* 2008).

The addition of a bottom collector to canopy traps is important as some taxa are preferentially caught in the top or bottom collector depending on whether the trap is set in the understory or canopy (Vance *et al.* 2007).

Wet or dry killing agents may be used in the collecting head. Both have advantages depending on the taxa targeted. Wet killing agents – such as 70-90% ethanol or propylene glycol – also function as a preservative, which is needed if traps are serviced on a weekly or longer basis. Delicate specimens, especially Lepidoptera, may be damaged by wet killing agents and unidentifiable beyond higher taxonomic levels (e.g., family or genus). Dry killing agents – such as naphthalene, insecticide-permeated strips, or urinal cakes – help alleviate this but require traps

be serviced more often, potentially daily, as specimens may damage themselves before succumbing to the agent if an excess of specimens builds up in the trap head.

Factors influencing catch.

Various aspects of trap design affect the taxa collected. Matthews and Matthews (1983) found Towne's style Malaise traps caught ten times as many specimens as Cornell-style Malaise traps. Mesh size is an important consideration when collecting Hymenoptera as coarse mesh is more effective in collecting Aculeata, fine mesh is more effective in collecting microhymenoptera, and both coarse and fine mesh are effective in collecting Ichneumonoidea (Darling & Packer 1988). The color of the mesh panels has been shown to affect the catch of Tabanidae and Culicidae (Roberts 1970, 1972); black, in particular, increases the overall number of specimens and species collected (Hansen 1988). Disney *et al.* (1982) found that slightly altering the position of the collecting container from the peak of a Malaise trap to just below the peak significantly reduced the overall number of certain Diptera species collected and somewhat reduced the number of species collected. The age of Malaise traps has also been shown to significantly alter trap catch, possibly changes in color due to exposure to sunlight (Roberts 1975; Duarte *et al.* 2010).

Species-specific factors of target taxa such as behavior, habitat preference, and activity level influence trap catch. For instance, many species of Syrphidae are readily collected in Malaise traps, though some abundant species avoid the trap altogether (Burgio & Sommaggio 2007). Collections of mosquitos are biased towards *Aedes* and *Culex* (Acuff 1976) while collections of Agromyzidae are female-biased (Scheirs *et al.* 1997). When used to collect spiders, Malaise traps sample a greater proportion of arboreal, web-building species compared to terrestrial, active-hunting species (Jenning & Hilburn 1988; Oxbrough 2010).

The addition of various lures can increase the catch of specific taxa. For example, the addition of carbon dioxide in the form of dry ice or compressed gas released over time increases the trap catch of hematophagous Diptera and mammalian parasites (Easton *et al.* 1968; Smith *et al.* 1965; Geijskes 1968; Witter *et al.* 2012; Tabanidae: Roberts 1971; Anderson & Hoy 1972; Blume *et al.* 1972; Roberts 1976b; Hollander & Wright 1980; Strickler & Walker 1993; Leprince *et al.* 1994; Culicidae: Breeland & Pickard 1965; Graham 1969; Oestridae: Capelle 1970; Wright *et al.* 1984; Cogley & Cogley 2000; Fleenor & Taber 2007; Witter *et al.* 2012). Most studies have found 1-octen-3-ol (French & Kline 1989; Schreck *et al.* 1993; Krčmar *et al.* 2005; Krčmar *et al.* 2010), ammonia (Hribar *et al.* 1992; Krčmar *et al.* 2010), acetone (Krčmar *et al.* 2010), lactic acid (Krčmar *et al.* 2010), and aged animal urine (Krčmar *et al.* 2005; Krčmar *et al.* 2006; Krčmar *et al.* 2010), as well as the addition of a large, round, black object (such as an inflated beach ball covered in black cloth) (Catts 1970; Schreck *et al.* 1993) increase the number of Tabanidae caught in Malaise and canopy traps, though some have not (Leprince *et al.* 1994). 1-octen-3-ol is also attractive to Culicidae (Nilssen 1998). 2,4-hexadlenyl butyrate and heptyl butyrate are highly potent, specific lures attractive to *Vespula* yellowjackets; Malaise traps baited with these chemicals can be used to control yellowjacket populations over small areas such as fruit orchards (Davis *et al.* 1973). Methyl eugenol is attractive to some species of Hawaiian Drosophilidae and Muscidae (Asquith & Kido 1994).

Terrestrial / canopy trap comparison.

Within temperate forests, some studies have found that Malaise traps, when compared to canopy traps, catch more insect specimens and sample a higher diversity at both the family (Preisser *et al.* 1998; Rohr *et al.* 2007; Barkley 2009) and species level (Syrphidae: Birtele & Hardersen 2012; Tachinidae: Cerretti *et al.* 2004; Stireman *et al.* 2012). However, other studies

have found that Malaise and canopy traps have similar observed species richness, though capture significantly different species assemblages (Cerambycidae: Vance *et al.* 2003; Hardersen *et al.* 2014 Neuroptera: Hollier & Belshaw 1993), while others have found no difference in species composition (Stork & Grimbacher 2006). Additionally, while observed species richness is equivalent or higher in Malaise traps, expected species richness, which is based on various species richness estimators, may be higher in canopy traps (Vance *et al.* 2003; Stireman *et al.* 2012). Finally, canopy and Malaise traps collect similar feeding-guild assemblages, at least when considering Coleoptera (Grimbacher & Stork 2007).

When both styles of trap collect the same taxon, relative abundance in per trap may vary significantly depending on the taxonomic level analyzed (family: Barkley 2009; genus: Roberts 1976b; species: Eveleigh *et al.* 2007).

Comparisons to other traps and collecting methods.

Specimens collected by Malaise traps, including delicate Culicidae, are preserved in better condition than those taken in other traps (Graham 1969).

Malaise traps are more frequently in forests while window traps are preferred in open landscapes; both traps, however, can be used in either situation (Duelli *et al.* 1999).

Malaise traps, when compared to glass-barrier, window, and sticky, collect more specimens of Diptera, Hymenoptera, and Hemiptera but fewer specimens of Coleoptera (Juillet 1963; Lamarre *et al.* 2012). However, other studies that used finer taxonomic units found that Malaise traps collect more specimens of certain beetle families (e.g., Cleridae, Curculionidae, Elateridae) than light, window, and sticky traps (Hosking 1979).

Malaise traps are less efficient than colored pan traps when targeting pollenating insects, including bees, though the addition of colored fabric to Malaise traps increases the number of pollinators collected (Bartholomew & Prowell 2005; Campbell & Hanula 2007).

Möricke/yellow pan and Malaise traps collect significantly different assemblages: one study found only 12% overlap in the Hymenoptera species collected by either method (Finnamore *et al.* 2012). Yellow pan traps generally collect more specimens but are dominated by a few species while Malaise traps collect fewer specimens representing more species with a more even distribution of species (Wells & Decker 2006). When considering Ichneumonidae specifically, Möricke traps collect more Orthocentrinae and Cryptini (Mazón & Bordera 2008; Aguiar & Santos 2010). Within a species, sexes may be preferentially collected by each method: Malaise traps catch collect more male Ichneumonidae and female Agromyzidae while Möricke or yellow pan traps collect more female Ichneumonidae and male Agromyzidae (Scheirs *et al.* 1997; Aguiar & Santos 2010).

Malaise and white pan traps are more or less efficient depending on the family of Diptera considered (Disney *et al.* 1982).

Malaise traps are more efficient than hand rearing when collecting Ichneumonoidea (Bartlett 2000) but less effective when hand collecting Cerambycidae (Noguera *et al.* 2002).

Malaise traps and canopy fogging collect similar assemblages of Formicidae, which is significantly different than the assemblage sampled by collecting leaf litter (Longino & Colwell 1997). Furth *et al.* (2003) found broad overlap between the flea beetle (Chrysomelidae: Alticinae) taxa collected by Malaise traps and canopy fogging in Costa Rica; they also found fogging to be more efficient than Malaise traps on a per sample basis but Malaise traps were more efficient on a per individual basis, so Malaise traps are more efficient over long time spans.

While effectiveness is reduced, Malaise traps continue to function in damp conditions, so may be preferable to vacuum-sampling understory vegetation that is constantly wet (Noyes 1989).

Suction traps are more efficient at collecting Culicidae than Malaise traps (Lothrop *et al.* 2002).

Social wasps are collected more efficiently with watered-down honey bait (Noll & Gomes 2009) and hand collecting (Silveira 2002) than with Malaise traps.

Malaise traps and sweep netting differentially sample genera when collecting Tabanidae: the majority of *Tabanus* and *Hybomitra* are collected in Malaise traps while the majority of *Chrysops* are collected by netting (Tallamy *et al.* 1976; Strickler & Walker 1993).

Malaise traps undersample Neuropteroidea, with the exception of Raphidioptera, when compared to light and suction traps (Abraham *et al.* 2003).

When sampling pecan weevil (*Curculio caryae*), Malaise traps situated in the first crotch of pecan trees collect more beetles than cone emergence traps (Dutcher *et al.* 1986).

Summary.

Malaise traps revolutionized the collection of flying insects. Many iterations and refinement in design have been proposed since their inception.

An array of insects are collected by the traps. Trap catch is generally dominated by Diptera and Hymenoptera, with actively flying species of other orders also commonly represented. The traps can be used for a number of purposes, including general collecting and biodiversity surveys, investigating insect movement, vertical stratification, and diel and seasonal patterns of abundance.

Many factors influence the taxa collected. Abiotic factors, such as weather, season, and trap design, orientation, and placement can variously affect the behavior of target taxa and influence the species trapped. Biotic factors affecting trap catch include the type and density of surrounding vegetation and species-specific behavior. The addition of various lures increases number of certain species; this has been best studied in hematophagous Diptera and other pest species.

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IV. Terrestrial arthropods of Steel Creek, Buffalo National River, Arkansas. I. Select beetles (Coleoptera: Buprestidae, Carabidae, Cerambycidae, Curculionoidea excluding Scolytinae).

Abstract.

Background

The Ozark Mountains are a region with high endemism and biodiversity, yet few invertebrate inventories have been made and few sites extensively studied. We surveyed a site near Steel Creek Campground, along the Buffalo National River in Arkansas, using twelve trap types – Malaise traps, canopy traps (upper and lower collector), Lindgren multifunnel traps (black, green, and purple), pan traps (blue, purple, red, white, and yellow), and pitfall traps – and Berlese-Tullgren extraction for eight and half months.

New information

We provide collection records of beetle species belonging to eight families collected at the site. Thirty one species represent new state records: (Buprestidae) *Actenodes acornis*, *Agrilus cephalicus*, *Agrilus ohioensis*, *Agrilus paracelti*, *Taphrocerus nicolayi*; (Carabidae) *Agonum punctiforme*, *Synuchus impunctatus*; (Curculionidae) *Acalles clavatus*, *Acalles minutissimus*, *Acoptus suturalis*, *Anthonomus juniperinus*, *Anametis granulata*, *Idiostethus subcalvus*, *Eudociminus mannerheimii*, *Madarellus undulates*, *Magdalis armicollis*, *Magdalis barbata*, *Mecinus pascuorum*, *Myrmex chevrolatii*, *Myrmex myrmex*, *Nicentrus lecontei*, *Otiorhynchus rugostriatus*, *Piazorhinus pictus*, *Phyllotrox ferrugineus*, *Plocamus hispidulus*, *Pseudobaris nigrina*, *Pseudopentarthrum simplex*, *Rhinoncus pericarpus*, *Sitona lineatus*, *Stenoscelis brevis*, *Tomolips quericola*. Additionally, three endemic carabids, two of which are known only from the type series, were collected.

Introduction.

The Interior Highlands is a mountainous physiogeographic division in the central United States and the only significant topographic relief between the Appalachian and Rocky Mountains (Fig. 1). The area is known to harbor high biodiversity and many endemic species but remains grossly understudied. It is comprised of two regions with different geological histories: the Ouachita Mountains, which occupy west-central Arkansas and southeastern Oklahoma, and the Ozarks, which occupy southern Missouri, northern Arkansas, and extreme southeastern Kansas (Fig. 2).



Figure 1. The Buffalo River from an overlook on the Buffalo River Trail near Steel Creek. Photo © Jasari. Used under Creative Commons license Attribution-ShareAlike 3.0 (CC BY-SA 3.0) (Creative Commons 2015).

The Ouachita Mountains are east-west trending fold mountains approximately 100 km wide and 190 km long (3,237,600 ha), with elevations up to 818 m (Robison and Allen 1995). They are the largest exposure of the Ouachita orogeny, which formed during the assembly of Pangea (by ~270 Ma); other exposures of the orogeny include the Marathon Mountains in Mexico and the base of the Sierra del Carmen in Coahuila, Mexico (Flawn 1968, Spearing 1991,

U.S. Geological Survey 2014). Historically, the Ouachitas were connected to the Marathon Mountains to the west and Appalachian Mountains to the east. However, the break-up of Pangea and subsequent expansion of the Western Interior Seaway during the Cretaceous eroded and covered the mountains to the west while the formation of the Mississippi embayment, which resulted from the uplifting, rapid erosion, and subsequent subsidence of the area between the Ouachita and Appalachian Mountains from the mid-Cretaceous through early Cenozoic, severed the connection to the Appalachians (Carlton and Cox 1990, Spearing 1991, Cox and Van Arsdale 2002, Poole et al. 2005, U.S. Geological Survey 2014).

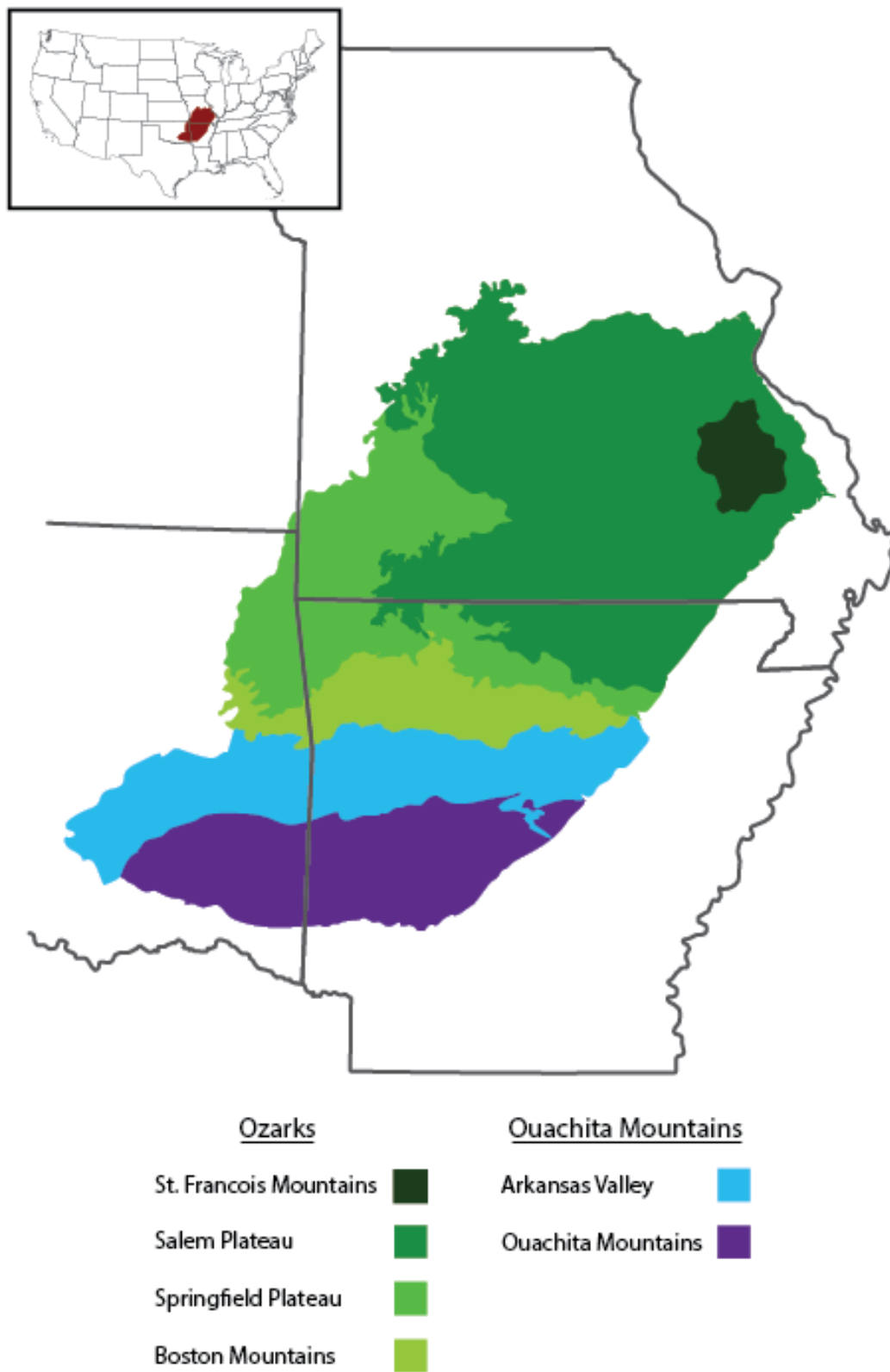


Figure 2. Geologic subregions of the Interior Highlands. Inset shows the region in context of the entire United States.

Prior to European settlement, the Ouachita Mountains were dominated by shortleaf pine (*Pinus echinata* Mill.), pine-hardwood, and mixed oak (*Quercus* L.) forests, with diverse, fire-dependent forb and grass understories (Hedrick et al. 1999); fire return intervals averaged 10 years and tree densities averaged 420 trees per ha with a mean diameter of 29 cm (Kreiter 1992, Masters et al. 1995). However, most virgin forest was heavily logged between 1910 and 1940 (Smith 1986) and presently tens of thousands of hectares have been converted to loblolly pine (*Pinus taeda* L.) plantations (Hedrick et al. 1999). The understory is dominated by woody vegetation and tree density has increased to 494–618 trees per ha while the mean diameter has decreased to 23 cm and average fire return intervals range from 40 to 1,200 years (Kreiter 1992, Masters et al. 1995).

The Ozarks, also referred to as the Ozark Mountains or Ozark Plateau, is divided into four geologic subdivisions. The Saint Francois Mountains, the oldest subdivision, is the exposed remains of a Proterozoic mountain range that formed through volcanic and intrusive activity 1485 Ma (Denison et al. 1984); it is also the smallest subdivision, covering approximately 180 square kilometers (Bretz 1965). The Salem Plateau, Springfield Plateau, and Boston Mountains are younger (Ordovician, Mississippian, and early Pennsylvanian age, respectively) plateaus that formed as the result of sedimentation and deposition along the edge of Laurentia. The Salem and Springfield Plateaus are composed largely of limestone and dolomite and are typified by karst topography, with thousands of caves and hundreds of springs documented in the region, while the Boston Mountains are composed largely of sandstone and shale (Bretz 1965, Arkansas Geological Survey 2015, Missouri Department of Natural Resources 2015, National Park Service 2015). The plateaus have been repeatedly uplifted and weathered, with the final uplift of the

Ozarks occurring during the formation of the Ouachita orogeny; the region has remained exposed for the last 270 million years (Bretz 1965, Robison and Allen 1995, Guccione 2008, U.S. Geological Survey 2014).

The Salem and Springfield Plateaus rise to elevations of 450 m and 550 m, respectively, and are characterized by relatively flat plateau surfaces that form extensive plains cut into rolling, level-topped hills around rivers and other flowing water (Foti 2014). Oak/hickory forests and open woodlands are typical for the region, though extensive rocky, open glades can be common; additionally, the Springfield Plateau historically had extensive prairies, though these have largely been converted to agriculture (Foti 2014). The Boston Mountains is a highly dissected plateau, due to differential weathering of the relatively soft shale and harder sandstone, and the most rugged subdivision of the Ozarks, with an average elevation around 500 m and peaks up to 780 m. Oak/hickory forests predominate in most of the region, though drier south-facing slopes with extensive sandstone support short-leaf pine forests and moist, protected ravines support beech and sugar maple, which are uncommon elsewhere in the Ozarks (Foti 2014). For more information about the regions as they occur in Arkansas see Anderson 2006.

The Ouachita Mountains and Ozarks have never been connected as the Arkansas Valley (also called the Arkansas River Valley), which is part of the Arkoma Basin, formed as a foreland basin through downwarping along the Ouachita orogeny when the Ouachita Mountains were uplifted (Morris 1974, Wickham et al. 1976). The Arkansas River and its tributaries have increased the disconnection by eroding thousands of feet of sediment from the valley floor, which currently has an elevation of 90–150 m, and act as a physical barrier to poor-dispersing species (Carlton and Cox 1990, Foti and Bukenhofer 1998, Foti 2011). Differential erosion throughout the valley has left a few steep-sided, sandstone capped plateaus: Mount Magazine,

Petit Jean Mountain, and Mount Nebo, which rise to elevations of 839 m, 741 m, and 411 m respectively (Higgins 2015, Peakery 2015).

The Interior Highlands can also be divided by ecoregion. Ecoregions, as defined by the Commission for Environmental Cooperation, are divided into three levels: Level I is the most inclusive and places the region "in context at global or intercontinental scales"; Level II regions are subdivisions of Level I regions and are "intended to provide a more detailed description of the large ecological areas nested within the level I regions"; finally, Level III has the smallest subdivisions that "enhance regional environmental monitoring, assessment and reporting, as well as decision-making" and "allow locally defining characteristics to be identified, and more specifically oriented management strategies to be formulated" (Commission for Environmental Cooperation 1997, Environmental Protection Agency 2015). At Level I, the Interior Highlands are included in the Eastern Temperate Forests, along with much of Eastern United States. At Level II the Interior Highlands are included in the Ozark, Ouachita-Appalachian Forests division, which also includes mountainous forests in the Appalachians. At Level III the Saint Francois Mountains, Salem and Springfield Plateaus are considered together as one subdivision – the Ozark Highlands – while the Boston Mountains, Arkansas Valley, and Ouachita Mountains are each considered separate subdivisions.

As may be expected with the regions inclusion in the Level I Eastern Temperate Forests ecoregion, many species found in the Interior Highlands are typical of eastern North America. However, some western species reach their eastern range limit in the Interior Highlands (e.g., Texas brown tarantula [*Aphonopelma hentzi* (Jean-Étienne Girard, 1852)], eastern collared lizard [*Crotaphytus collaris* (Say, 1823)], western diamondback rattlesnake [*Crotalus atrox* Baird & Girard, 1853]); these species likely colonized the Interior Highlands during the post-glacial

Xerothermic Interval (6,000-4,000 b.p.), during which time prairies and xeric habitat similar to that in the west expanded into the Interior Highlands, and remained after the climate became more moist (Dowling 1956, Smith 1965, Trauth 1989, Trauth and Cochran 1992). Additionally, many species exhibit highly disjunct populations or are endemic to the region due to a number of factors: the abundance of caves and karst habitat support numerous localized cavernicolous species (Crandal 1998, Culver et al. 2000, Graening et al. 2003, Sarver and Lister 2004, Graening et al. 2006); rare habitats, such as xeric limestone prairies and glades, support specialized species assemblages (Baskin and Baskin 1988, Heikens 1999, Baskin and Baskin 2000, Ware 2002, Lawless 2005); previous connections to similar habitat (e.g., the Ouachitas and Appalachians, the River Valley plateaus and higher elevation habitat) have been severed for millions of years, allowing isolated populations of poor-dispersing organisms to speciate (e.g., Carlton and Cox 1990); and the Interior Highlands served as a refugia during periods of high sea levels and glaciation due to the unique geographic history discussed above (Redfearn 1986, The Nature Conservancy, Ozarks Ecoregional Assessment Team 2003).

The Nature Conservancy, Ozarks Ecoregional Assessment Team 2003 reported 58 species with highly disjunct populations in the Ozarks and a number of authors have discussed the disjunct populations of taxa in the region (birds: Selander 1965; fish: Bailey and Allum 1962; amphibians: Blair 1965; reptiles: Trauth et al. 2004; aquatic insects: Ross 1965; plants: Steyermark 1959, Redfearn 1986, Hemmerly 2002). While a comprehensive list of Interior Highland endemics is lacking, various authors have worked on geographic or taxonomic subsets: e.g., Pringle and Witsell 2005 stated that at least 20 species of plants are endemic to the Ouachita Mountains and Zollner et al. 2005 listed 36 plants endemic to the Interior Highlands; Allen 1990 reported 68 species of endemic insects and suggested there are at least 200 endemic plant and

animal species in the Interior Highlands overall; Robison and Allen 1995 recorded 117 species endemic to Arkansas, most of which were found in the highland regions, though Robison et al. 2008 later reduced the number of Arkansas endemics to 100; and The Nature Conservancy, Ozarks Ecoregional Assessment Team 2003 reported 159 endemic species in the Ozarks. Additional disjunct and endemic species continue to be found and described (Table 1), so the number of such species is likely to continue to increase for the foreseeable future.

Range status	Taxonomic category	Select references
Disjunct	lichens	Lendemer & Harris 2007, Harris & Ladd 2008, Harris & Lendemer 2009, Barton & Lendemer 2014, Lendemer & Harris 2014
	plants	Simurda & Knox 2000, Rimmer & Summers 2006, Peck 2011
	molluscs	Nekola & Coles 2001
	arthropods	Carlton & Robison 1998
	fish	Berendzen et al. 2008
Endemic	lichens	Knudsen & Lendemer 2009
	plants	Rothrock & Reznicek 2001, Pringle & Witsell 2005, Campbell 2006, Nelson 2008, Floden et al 2009, Yatskievych et al. 2013
	arthropods	Wolfe & Harp 2003, Sokolov et al 2004, Holsinger et al 2006, Dillmann et al 2010, Hildebrandy & Maddison 2011, Radwell et al 2011
	fish	Kinzinger & Wood 2010, Adams et al. 2013

Table 1. Select references to recently discovered and described species with disjunct and endemic distributions in the Interior Highlands.

Aquatic insects and crayfish have been relatively well surveyed within the Interior Highlands (Table 2). Terrestrial insects and other arthropods, however, have been poorly surveyed and represent an excellent opportunity to find new endemic and disjunct species (though see Carlton and Robison 1998 concerning litter-dwelling beetles in the Ouachitas). This manuscript is the first in a series examining the arthropod fauna at a single site at Steel Creek along the Buffalo National River in the Boston Mountains of Arkansas. In addition to the new

species records and other notes included below, it is intended to serve as an in-depth introduction and reference for future papers based on data collected during the study and other surveys in the Interior Highlands.

Taxon	Select references
Ephemeroptera	McCafferty & Provonsha 1978, Sarver & Kondratieff 1997, Baumgardner & Kennedy 1999, Ferro & Sites 2007
Plecoptera	Ernst et al 1986, Poulton & Steward 1991, Ferro & Sites 2007
Trichoptera	Bowles & Mathis 1989, Mathis & Bowles 1992, Moulton & Stewart 1996, Ferro & Sites 2007, Etnier 2010
Astacoidea	Williams 1954

Table 2. Select references for well-sampled aquatic arthropods in the Interior Highlands.

Sampling Methods.

Sampling description: The following traps were maintained within the site: five Malaise traps (MegaView Science Co., Ltd., Taichung, Taiwan), twenty-five pan traps (five of each color: blue, purple, red, yellow, white) which were randomly arranged under the Malaise traps (one of each color per Malaise trap) so as to also act as intercept traps; fifteen Lindgren multi-funnel traps (ChemTich International, S.A., Heredia, Costa Rica) (five of each color: black, green, purple); four SLAM (Sea, Land, and Air Malaise) traps (MegaView Science Co., Ltd., Taichung, Taiwan) with top and bottom collectors that acted as canopy traps; and seventeen pitfall trap sets. Sixteen of the seventeen pitfall sets were placed in two transects of sets spaced every five meters centered on two Malaise traps while the final set was placed away from other traps. Additionally, ten leaf litter samples were collected for Berlese extraction when traps were serviced.

Pitfall traps were based on a design proposed by Nordlander 1987; they were made using plastic soup containers and modified from the original design by cutting three slots into the side of each container instead of circular entrances. The slots were cut 2 cm under the rim and

measured 2 cm tall x 9.3 cm wide, resulting in three equidistant 1.5 cm posts and a 28 cm collecting surface. The diameter at the base of the slots is approximately 10.5 cm and the cups are 10.5 cm deep below the slots, resulting in a collecting volume of 2,988 cm³. This design allowed the matching lids to be used as rain covers instead of using separate covers, such as ceramic tiles or bent metal sheeting. Each pitfall trap set was made by burying a single cup on either side of a 30.5 cm x 15.5 cm aluminum fence; trap catch from both cups was combined and treated as a single sample.

Berlese-Tullgren samples were collected from a variety of habitats, including thin leaf litter away from objects; thick leaf litter accumulated along logs and rocks; moss; tree holes; bark from fallen, partially decayed trees; and bark and leaf litter accumulated at the base of standing, dead trees. An attempt was made to collect moist, non-desiccated litter in order to increase the number of specimens collected; this resulted in fewer samples being taken from thin leaf litter, moss, and tree bark during the hot, dry summer months. Tree holes were only collected from once each so as not to totally destroy them as potential habitat; as the number of tree holes within the site was limited, this resulted in only a handful of collections from this habitat type. Leaf litter samples were processed for four to seven days until the litter was thoroughly dry using modified Berlese-Tullgren funnels.

Trap placement began on 8 March 2013 and all traps were set by 13 March 2013, except Lindgren funnels, which were set on 1 April 2013. Traps set earlier than 13 March were reset on that date in order to standardize trap catch between traps. Traps were serviced approximately every two weeks (Table 3). The last collection of pitfall traps and pan traps occurred on 6 November 2013; Malaise, SLAM, and Lindgren funnel traps were run for an additional month, with the final collection on 4 December 2013. Berlese-Tullgren samples were not collected on

13 April, 15 May, and 6 November due to heavy rain that began during trap servicing and precluded sample collection. Berlese-Tullgren samples collected on 28 June were lost due to evaporation of ethanol in the funnel collecting cups after sample processing began. Pitfall cups were dislodged on 13 April (one set), 15 May (one set), 28 June (four sets), 17 July (five sets) due to unknown circumstances, though the pattern of litter and debris around the cups on two occasions suggested heavy rainfall and water accumulation forced the cups from the holes. In total, 1311 samples were collected (Table 4).

Collection period
13 March 2013 – 1 April 2013
1 April 2013 – 13 April 2013
30 April 2013 – 15 May 2013
15 May 2013 – 29 May 2013
29 May 2013 – 12 June 2013
12 June 2013 – 28 June 2013
28 June 2013 – 17 July 2013
17 July 2013 – 30 July 2013
30 July 2013 – 13 August 2013
13 August 2013 – 28 August 2013
28 August 2013 – 11 September 2013
11 September 2013 – 25 September 2013
25 September 2013 – 8 October 2013
8 October 2013 – 23 October 2013
23 October 2013 – 6 November 2013
6 November 2013 – 20 November 2013
20 November 2013 – 4 December 2013

Table 3. Collection periods.

Propylene glycol (Peak RV & Marine Antifreeze) (Old World Industries, LLC, Northbrook, IL) was used as the preservative in all traps as it is non-toxic and generally preserves specimens well (Skvarla et al. 2014). Insect escape was impeded by the addition of a squirt of unscented, hypoallergenic dish detergent to the propylene glycol to act as a surfactant.

Trap catch was sieved in the field and stored in Whirl-Pak bags (Nasco, Fort Atkinson, WI) in 90% ethanol until sorting.

Trap type	Number of traps or collections	Number of samples
Berlese-Tullgren	10	140
Canopy trap (lower)	4	72
Canopy trap (upper)	4	72
Lindgren funnel (black)	5	85
Lindgren funnel (green)	5	85
Lindgren funnel (purple)	5	82
Malaise trap	5	95
Pan trap (blue)	5	82
Pan trap (purple)	5	81
Pan trap (red)	5	83
Pan trap (white)	5	83
Pan trap (yellow)	5	83
Pitfall	17	268

Table 4. Maximum number of traps collected (canopy, Lindgren funnel, Malaise, pan, and pitfall traps) or collections made (Berlese-Tullgren) per collecting period and total number of samples per sampling type; traps were occasionally destroyed or otherwise lost during the 2-week sampling period.

Quality control: Samples were coarse-sorted using a Leica MZ16 stereomicroscope illuminated with a Leica KL1500 LCD light source and a Wild M38 stereomicroscope illuminated with an Applied Scientific Devices Corp. Eco-light 20 fiber optic light source. After sorting, specimens were stored individually or by family in 2 mL microtubes (VWR International, LLC, Randor, PA) in 70% ethanol. Hard-bodied specimens (e.g., Carabidae, Curculionidae) were pinned or pointed as appropriate.

Specimens were identified with the use of published keys (Table 5). In some cases, difficult to key specimens were photographed through the eye piece of the stereomicroscope

using the camera on an HTC Droid Incredible 4G LTE cell phone or Samsung Galaxy S5 cell phone; the photographs were uploaded to Bugguide (Iowa State University 2015b) and identifications were proposed by Bugguide members. Proposed identifications were then double checked using published sources and either confirmed or corrected on the website.

The sole representative of *Lymanthes* collected keys to *L. sandersoni* in Sleeper 1965. However, the character that separates *L. sandersoni* and *L. arkansasensis* is dubious, especially given that the two species are described from one and two specimens, respectively, from areas that are geographically similar and not widely separated (less than 300 km). Furthermore, R. S. Anderson, who is currently revising the genus, believes that all *Lymanthes* in the eastern United States (excluding Texas) belong to a single species, *L. scrobicollis* (Paquin and Anderson 2009). Considering this, we identify the specimen collected as *L. sandersoni* with the caveat that it is likely that both *L. sandersoni* and *L. arkansasensis* will be synonymized with *L. scrobicollis* in the future.

Ormiscus consists of 14 described and approximately 30 undescribed species in North America north of Mexico (Valentine 2002). Species are most easily identified by the male secondary sexual features (e.g., characters on the mid and hind tibiae), however some species appear to be parthenogenetic (B. Valentine, pers. comm., via Iowa State University 2015b). In summary, this genus is in need of a major revision. As two-thirds or more of the North American species remain undescribed, we have declined to assign the single specimen collected to species.

Two weevil species, *Auleutes nebulosus* and *Laemosaccus nephele*, are thought to be complexes of multiple cryptic species that are in need of revision (Anderson 2002, Ciegler 2010). As a limited number of specimens (2 and 4 per species complex, respectively) were

collected, it is unlikely that multiple species were collected; additionally, modern revisions are lacking and identification of putative species is impossible. Specimens were therefore identified as the nominative species with the caveat that future studies may break the species complexes up and assign specimens collected in this study to other species.

The males of nine of 17 species of *Cercopeus* in the United States, including the widespread species *C. chrysorrhoeus*, are undescribed (O'Brien et al. 2010). All female *Cercopeus* collected in this study were identified as *C. chrysorrhoeus*; we therefore assumed that the males collected, which do not conform to the nine described males, are also *C. chrysorrhoeus*.

The *Chrysobothris femorata* species group consists of a dozen species that are difficult to separate (with the exception of *C. adelpha*) as the characters used to distinguish species, including genitalia, are variable and often intermediate between species (Paiero et al. 2012). Further revision of the group is needed to positively identify species so, except for *C. adelpha*, we have chosen not to assign specimens to individual species.

All specimens have been deposited in the University of Arkansas Arthropod Museum (UAAM), with the following exceptions: 1) 1–5 exemplars of each species have been deposited in the Dowling Lab Collection at the University of Arkansas; 2) the following specimens were sent to Peter Messer for identification confirmation and have been deposited in the P. W. Messer Collection: *Agonum striatopunctatum* (MS 13-0529-072, #136215; MS 13-0612-022, #139663), *Cicindela rufiventris* (MS 13-0717-001, #134492), *Cyclotrachelus incisus* (MS 13-0413-023, #139591; MS 13-0413-019, #139592; MS 13-0413-006, #139594; MS 13-1008-075, #139596), *Cyclotrachelus parasodalis* (MS 13-0430-019, #131983; MS 13-0529-037, #135057; MS 13-1106-002, #138280), *Lophoglossus haldemanni* (MS 13-0529-066, #135053),

Pterostichus punctiventris (MS 13-0401-018, #135065; MS 13-1023-021, # 136216), *Rhadine ozarkensis* (MS 13-0925-027, #134547), *Scaphinotus fissicollis* (MS 13-1106-037, #137830), *Selenophorus ellipticus* (MS 13-0925-005, #136223), *Selenophorus opalinus* (MS 13-0813-034, # 136217), *Trichotichus autumnalis* (MS 13-0730-005, #136226), *Trichotichnus vulpeculus* (MS 13-0911-027, #136218).

New Arkansas state records for Buprestidae are based on the range data given by Paiero et al. 2012; for Carabidae are based on range data given by Bousquet 2012b; and for Attelabidae and Curculionidae are based on O'Brien and Wibmer 1982 and supplemented by more recent literature (see individual species notes for specific citations). No attempt was made to assess the state record status of Cerambycidae as recent checklists and keys (e.g., Linsley 1962a, Linsley 1962b, Linsley 1963, Linsley 1964, Linsley and Chemsak 1972, Linsley and Chemsak 1976, Chemsak and Linsley 1982, Linsley and Chemsak 1984, Linsley and Chemsak 1995, Yanega 1996, Lingafelter 2007, Bezark and Monné 2013) report regional presence rather than presence by state and/or contain range maps for a few species with a limited number of records and J. A. Chemsak sadly passed before completing his "Illustrated Revision of the Cerambycidae of North America" series, which includes detailed range maps for the species treated (though see Chemsak 1996 for Parandrinae, Spondylidinae, Aseminae, and Prioninae and Chemsak 2007 for Lepturinae).

Family	Genus	Reference
Anthribidae		Valentine 1960, Valentine 1998
Attelabidae		Hamilyon 1971, Hamilton 1989, Hamilton 2002
Brentidae		Anderson and Kissinger 2002
Buprestidae		Nelson et al. 2008, Paiero et al. 2012
Carabidae		Ball 1959, Lindroth 1969, Ciegler 2000, Arnett and Ivie 2001, Ball and Bousquet 2001, Pearson et al. 2006
Carabidae	<i>Abacidus</i>	Lindroth 1969, Sadek 1982
Carabidae	<i>Agonum</i>	Liebherr 1994
Carabidae	<i>Anisodactylus</i>	Noonan 1973
Carabidae	<i>Brachinus</i>	Erwin 1970
Carabidae	<i>Calathus</i>	Ball and Negre 1972
Carabidae	<i>Carabus</i>	Haldeman 1852
Carabidae	<i>Chlaenius</i>	Bell 1960
Carabidae	<i>Clinidium</i>	Bell and Bell 1975, Bell 1999
Carabidae	<i>Clivina</i>	Ball 2001, Bousquet 2009
Carabidae	<i>Cychrus</i>	Gidaspow 1973
Carabidae	<i>Cymindis</i>	Hunting 2013
Carabidae	<i>Dicheirus</i>	Noonan 1973
Carabidae	<i>Harpalus</i>	Noonan 1991
Carabidae	<i>Lebia</i>	Madge 1967
Carabidae	<i>Notiophilus</i>	Larochelle and Lariviere 1990
Carabidae	<i>Notobia</i>	Noonan 1973
Carabidae	<i>Platynus</i>	Liebherr and Will 1996, Bousquet 2012b
Carabidae	<i>Progalertina</i>	Ball and Nimmo 1983
Carabidae	<i>Pseudophonus</i>	Ball and Anderson 1962
Carabidae	<i>Pterostichus</i>	Bousquet 1992
Carabidae	<i>Rhadinae</i>	Barr 1974
Carabidae	<i>Scaphinotus</i>	Van Dyke 1938, Allen and Carlton 1988
Carabidae	<i>Stenolophus</i>	Bousquet and Messer 2010
Carabidae	<i>Tachyta</i>	Erwin 1975
Cerambycidae		Yanega 1996, Lingafelter 2007
Cerambycidae	<i>Astylopsis</i>	Schiefer 2000
Cerambycidae	<i>Purpuricenus</i>	MacRae 2000
Cerambycidae	<i>Saperda</i>	Schiefer and Newell 2010
Curculionidae		Schaeffer 1907, Blatchley and Leng 1916, Anderson 2002, Hespenheide 2002, Ciegler 2010, Lyal 2010, WTaxa et al. 2012
Curculionidae	<i>Cercopus</i>	O'Brien et al. 2010
Curculionidae	<i>Conotrachelus</i>	Schoof 1942
Curculionidae	<i>Cossonus</i>	Van Dyke 1915
Curculionidae	<i>Curculio</i>	Gibson 1969

Table 5. References used for specimen identification.

Family	Genus	Reference
Curculionidae	<i>Dichoxenus</i>	Sleeper 1956
Curculionidae	<i>Eubulus</i>	Anderson 2008
Curculionidae	<i>Geraeus</i>	Prena 2009
Curculionidae	<i>Lechriops</i>	Hespenheide 2003
Curculionidae	<i>Linogeraeus</i>	Prena 2009
Curculionidae	<i>Lissorhoptrus</i>	O'Brien and Haseeb 2014
Curculionidae	<i>Lymantes</i>	Sleeper 1965, Paquin and Anderson 2009
Curculionidae	<i>Notiodes</i>	Board 1972
Curculionidae	<i>Oopterinus</i>	O'Brien 1985
Curculionidae	<i>Otiorhynchus</i>	Warner and Negley 1976
Curculionidae	<i>Pandeletius</i>	Howden 1959
Curculionidae	<i>Rhinoncus</i>	Hoebeke and Whitehead 1980
Curculionidae	<i>Tychius</i>	Clark 1971
Curculionidae	<i>Tyloderma</i>	Wibmer 1918

Table 5 (cont.). References used for specimen identification.

Geographic Coverage.

Description: The survey was conducted at 4 hectare plot established at Steel Creek along the Buffalo National River in Newton County, Arkansas, centered at approximately N 36°02.269', W 93°20.434'. The site is primarily 80–100 year old mature second-growth Eastern mixed deciduous forest dominated by oak (*Quercus*) and hickory (*Carya*), though American beech (*Fagus grandifolia*) and eastern red cedar (*Juniperus virginiana*) are also abundant. A small (14 m x 30 m), fishless pond and glade (10 m x 30 m) with sparse grasses are present within the boundaries of the site.

Coordinates: 36.0367 and 36.0397 Latitude; -93.3917 and -93.3397 Longitude.

Taxonomic Coverage.

Description: All specimens of Anthribidae, Attelabidae, Brachyceridae, Brentidae, Bupresidae, Carabidae, Cerambycidae, Curculionidae excluding Scolytinae were identified to species.

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Data Resources.

Data package title: Steel Creek survey

Resource link: <http://dx.doi.org/10.5061/dryad.4h40n>

Number of data sets: 1

Data set name: Steel Creek beetles

Data format: Darwin Core Archive

Data format version: 1.0

See Table A1 for explanations of column headings in the data set spreadsheet.

Additional Information.**Analysis**

8,048 specimens representing 251 species and 188 genera were collected during this study (Table 6), with the following totals by family: Anthribidae: 15 specimens, 4 species, 4 genera; Attelabidae: 19 specimens, 3 species, 3 genera; Brachyceridae: 1 specimen, 1 species, 1 genus; Brentidae: 6 specimens, 1 species, 1 genus; Buprestidae: 375 specimens, 27 species, 9 genera; Carabidae: 1970 specimens, 62 species, 36 genera; Cerambycidae: 1885 specimens, 82 species, 57 genera; Curculionidae: 3777 specimens, 71 species, 52 genera.

Family	Genus	Species	Total specimens collected
Anthribidae	<i>Euparius</i>	<i>Euparius marmoreus</i>	11
Anthribidae	<i>Eurymycter</i>	<i>Eurymycter fasciatus</i>	2
Anthribidae	<i>Ormiscus</i>	<i>Ormiscus</i>	1
Anthribidae	<i>Toxonotus</i>	<i>Toxonotus cornutus</i>	1
Attelabidae	<i>Eugnamptus</i>	<i>Eugnamptus angustatus</i>	12
Attelabidae	<i>Synolabus</i>	<i>Synolabus bipustulatus</i>	1
Attelabidae	<i>Temnocerus</i>	<i>Temnocerus aeratus</i>	6
Brachyceridae	<i>Notiodes</i>	<i>Notiodes limatulus</i>	1
Brentidae	<i>Arrhenodes</i>	<i>Arrhenodes minutus</i>	6
Buprestidae	<i>Acmaeodera</i>	<i>Acmaeodera tubulus</i>	70
Buprestidae	<i>Acmaeodera</i>	<i>Acmaeodera pulchella</i>	1
Buprestidae	<i>Actenodes</i>	<i>Actenodes acornis*</i>	1
Buprestidae	<i>Agrilus</i>	<i>Agrilus arcuatus complex</i>	1
Buprestidae	<i>Agrilus</i>	<i>Agrilus bilineatus</i>	35
Buprestidae	<i>Agrilus</i>	<i>Agrilus cephalicus*</i>	18
Buprestidae	<i>Agrilus</i>	<i>Agrilus defectus</i>	1
Buprestidae	<i>Agrilus</i>	<i>Agrilus fallax</i>	1
Buprestidae	<i>Agrilus</i>	<i>Agrilus geminatus</i>	1
Buprestidae	<i>Agrilus</i>	<i>Agrilus lecontei</i>	4
Buprestidae	<i>Agrilus</i>	<i>Agrilus masculinus</i>	1
Buprestidae	<i>Agrilus</i>	<i>Agrilus ohioensis*</i>	1
Buprestidae	<i>Agrilus</i>	<i>Agrilus olentangyi</i>	1
Buprestidae	<i>Agrilus</i>	<i>Agrilus obsoletoguttatus</i>	12
Buprestidae	<i>Agrilus</i>	<i>Agrilus paracelti*</i>	3
Buprestidae	<i>Anthaxia</i>	<i>Anthaxia viridifrons</i>	6
Buprestidae	<i>Brachys</i>	<i>Brachys aerosus</i>	1
Buprestidae	<i>Chrysobothris</i>	<i>Chrysobothris adelpha</i>	60
Buprestidae	<i>Chrysobothris</i>	<i>Chrysobothris femorata complex</i>	70
Buprestidae	<i>Chrysobothris</i>	<i>Chrysobothris sexsignata</i>	7
Buprestidae	<i>Dicerca</i>	<i>Dicerca divaricata*</i>	3
Buprestidae	<i>Dicerca</i>	<i>Dicerca lurida</i>	58
Buprestidae	<i>Dicerca</i>	<i>Dicerca obscura</i>	8
Buprestidae	<i>Dicerca</i>	<i>Dicerca spreta</i>	1
Buprestidae	<i>Ptosima</i>	<i>Ptosima gibbicollis</i>	5
Buprestidae	<i>Taphrocercus</i>	<i>Taphrocercus gracilis</i>	3
Buprestidae	<i>Taphrocercus</i>	<i>Taphrocercus nicolayi*</i>	2

Table 6. Species collected, including total number of specimens. New state records are indicated by an asterisk (*).

Family	Genus	Species	Total specimens collected
Carabidae	<i>Agonoleptus</i>	<i>Agonoleptus conjunctus</i>	17
Carabidae	<i>Agonum</i>	<i>Agonum punctiforme</i> *	2
Carabidae	<i>Agonum</i>	<i>Agonum striatopunctatum</i>	3
Carabidae	<i>Amara</i>	<i>Amara aenea</i>	3
Carabidae	<i>Amara</i>	<i>Amara cupreolata</i>	14
Carabidae	<i>Amara</i>	<i>Amara musculus</i>	30
Carabidae	<i>Anisodactylus</i>	<i>Anisodactylus rusticus</i>	33
Carabidae	<i>Apenes</i>	<i>Apenes sinuata</i>	8
Carabidae	<i>Badister</i>	<i>Badister notatus</i>	3
Carabidae	<i>Bembidion</i>	<i>Bembidion affine</i>	6
Carabidae	<i>Bembidion</i>	<i>Bembidion rapidum</i>	2
Carabidae	<i>Brachinus</i>	<i>Brachinus americanus</i>	91
Carabidae	<i>Calathus</i>	<i>Calathus opaculus</i>	14
Carabidae	<i>Calleida</i>	<i>Calleida viridipennis</i>	8
Carabidae	<i>Carabus</i>	<i>Carabus sylvosus</i>	20
Carabidae	<i>Chlaenius</i>	<i>Chlaenius platyderus</i>	1
Carabidae	<i>Chlaenius</i>	<i>Chlaenius tomentosus</i>	3
Carabidae	<i>Cicindela</i>	<i>Cicindela rufiventris</i>	3
Carabidae	<i>Cicindela</i>	<i>Cicindela sexguttata</i>	32
Carabidae	<i>Clinidium</i>	<i>Clinidium sculptile</i>	1
Carabidae	<i>Clivina</i>	<i>Clivina pallida</i>	1
Carabidae	<i>Cyclotrachelus</i>	<i>Cyclotrachelus incisus</i>	797
Carabidae	<i>Cyclotrachelus</i>	<i>Cyclotrachelus parasodalis</i>	33
Carabidae	<i>Cymindis</i>	<i>Cymindis americana</i>	9
Carabidae	<i>Cymindis</i>	<i>Cymindis limbata</i>	203
Carabidae	<i>Cymindis</i>	<i>Cymindis platycollis</i>	8
Carabidae	<i>Dicaelus</i>	<i>Dicaelus ambiguus</i>	22
Carabidae	<i>Dicaelus</i>	<i>Dicaelus elongatus</i>	11
Carabidae	<i>Dicaelus</i>	<i>Dicaelus sculptilis</i>	78
Carabidae	<i>Dromius</i>	<i>Dromius piceus</i>	1
Carabidae	<i>Elaphropus</i>	<i>Elaphropus granarius</i>	1
Carabidae	<i>Galerita</i>	<i>Galerita bicolor</i>	19
Carabidae	<i>Galerita</i>	<i>Galerita janus</i>	2
Carabidae	<i>Harpalus</i>	<i>Harpalus faunus</i>	1
Carabidae	<i>Harpalus</i>	<i>Harpalus katiae</i>	1
Carabidae	<i>Harpalus</i>	<i>Harpalus pensylvanicus</i>	5

Table 6 (cont.). Species collected, including total number of specimens. New state records are indicated by an asterisk (*).

Family	Genus	Species	Total specimens collected
Carabidae	<i>Lebia</i>	<i>Lebia analis</i>	1
Carabidae	<i>Lebia</i>	<i>Lebia marginicollis</i>	1
Carabidae	<i>Lebia</i>	<i>Lebia viridis</i>	37
Carabidae	<i>Lophoglossus</i>	<i>Lophoglossus haldemanni</i>	1
Carabidae	<i>Mioptachys</i>	<i>Mioptachys flavicauda</i>	12
Carabidae	<i>Notiophilus</i>	<i>Notiophilus novemstriatus</i>	67
Carabidae	<i>Platynus</i>	<i>Platynus decentis</i>	9
Carabidae	<i>Platynus</i>	<i>Platynus parmarginatus</i>	2
Carabidae	<i>Plochionus</i>	<i>Plochionus timidus</i>	2
Carabidae	<i>Pterostichus</i>	<i>Pterostichus permundus</i>	105
Carabidae	<i>Pterostichus</i>	<i>Pterostichus punctiventris</i>	11
Carabidae	<i>Rhadine</i>	<i>Rhadine ozarkensis</i>	1
Carabidae	<i>Scaphinotus</i>	<i>Scaphinotus unicolor</i>	4
Carabidae	<i>Scaphinotus</i>	<i>Scaphinotus fissicollis</i>	12
Carabidae	<i>Scaphinotus</i>	<i>Scaphinotus infletus</i>	1
Carabidae	<i>Selenophorus</i>	<i>Selenophorus ellipticus</i>	4
Carabidae	<i>Selenophorus</i>	<i>Selenophorus gagatinus</i>	8
Carabidae	<i>Selenophorus</i>	<i>Selenophorus opalinus</i>	1
Carabidae	<i>Stenolophus</i>	<i>Stenolophus ochropezus</i>	5
Carabidae	<i>Synuchus</i>	<i>Synuchus impunctatus*</i>	3
Carabidae	<i>Tachyta</i>	<i>Tachyta parvicornis</i>	3
Carabidae	<i>Tachys</i>	<i>Tachys columbiensis</i>	4
Carabidae	<i>Tachys</i>	<i>Tachys oblitus</i>	2
Carabidae	<i>Trichotichnus</i>	<i>Trichotichnus autumnalis</i>	176
Carabidae	<i>Trichotichnus</i>	<i>Trichotichnus fulgens</i>	11
Carabidae	<i>Trichotichnus</i>	<i>Trichotichnus vulpeculus</i>	1
Cerambycidae	<i>Aegomorphus</i>	<i>Aegomorphus modestus</i>	8
Cerambycidae	<i>Aegormorphus</i>	<i>Aegormorphus quadrigibbus</i>	1
Cerambycidae	<i>Anelaphus</i>	<i>Anelaphus parallelus</i>	162
Cerambycidae	<i>Anelaphus</i>	<i>Anelaphus pumilus</i>	4
Cerambycidae	<i>Astyleiopus</i>	<i>Astyleiopus variegatus</i>	1
Cerambycidae	<i>Astylidius</i>	<i>Astylidius parvus</i>	2
Cerambycidae	<i>Astylopsis</i>	<i>Astylopsis macula</i>	4
Cerambycidae	<i>Astylopsis</i>	<i>Astylopsis sexguttata</i>	1
Cerambycidae	<i>Bellamira</i>	<i>Bellamira scalaris</i>	2
Cerambycidae	<i>Brachyleptura</i>	<i>Brachyleptura champlaini</i>	5
Cerambycidae	<i>Callimoxys</i>	<i>Callimoxys sanguinicollis</i>	4

Table 6 (cont.). Species collected, including total number of specimens. New state records are indicated by an asterisk (*).

Family	Genus	Species	Total specimens collected
Cerambycidae	<i>Centrodera</i>	<i>Centrodera sublineata</i>	1
Cerambycidae	<i>Clytoleptus</i>	<i>Clytoleptus albofasciatus</i>	6
Cerambycidae	<i>Cyrtinus</i>	<i>Cyrtinus pygmaeus</i>	5
Cerambycidae	<i>Cyrtophorus</i>	<i>Cyrtophorus verrucosus</i>	17
Cerambycidae	<i>Dorcaschema</i>	<i>Dorcaschema alternatum</i>	2
Cerambycidae	<i>Dorcaschema</i>	<i>Dorcaschema cinereum</i>	15
Cerambycidae	<i>Dorcaschema</i>	<i>Dorcaschema nigrum</i>	2
Cerambycidae	<i>Dorcaschema</i>	<i>Dorcaschema wildii</i>	2
Cerambycidae	<i>Eburia</i>	<i>Eburia quadrigeminata</i>	7
Cerambycidae	<i>Ecyrus</i>	<i>Ecyrus dasycerus</i>	1
Cerambycidae	<i>Elytrimitatrix</i>	<i>Elytrimitatrix undata</i>	30
Cerambycidae	<i>Elaphidion</i>	<i>Elaphidion mucronatum</i>	196
Cerambycidae	<i>Enaphalodes</i>	<i>Enaphalodes rufulus</i>	1
Cerambycidae	<i>Euderces</i>	<i>Euderces reichei</i>	1
Cerambycidae	<i>Euderces</i>	<i>Euderces picipes</i>	5
Cerambycidae	<i>Euderces</i>	<i>Euderces pini</i>	3
Cerambycidae	<i>Eupogonius</i>	<i>Eupogonius pauper</i>	2
Cerambycidae	<i>Gaurotes</i>	<i>Gaurotes cyanipennis</i>	1
Cerambycidae	<i>Graphisurus</i>	<i>Graphisurus despectus</i>	8
Cerambycidae	<i>Graphisurus</i>	<i>Graphisurus fasciatus</i>	10
Cerambycidae	<i>Heterachthes</i>	<i>Heterachthes quadrimaculatus</i>	18
Cerambycidae	<i>Hyperplatys</i>	<i>Hyperplatys maculata</i>	1
Cerambycidae	<i>Knuliana</i>	<i>Knuliana cincta</i>	10
Cerambycidae	<i>Leptostylus</i>	<i>Leptostylus transversus</i>	18
Cerambycidae	<i>Leptura</i>	<i>Leptura emarginata</i>	2
Cerambycidae	<i>Lepturges</i>	<i>Lepturges angulatus</i>	1
Cerambycidae	<i>Lepturges</i>	<i>Lepturges confluens</i>	9
Cerambycidae	<i>Micranoplium</i>	<i>Micranoplium unicolor</i>	3
Cerambycidae	<i>Molorchus</i>	<i>Molorchus bimaculatus</i>	65
Cerambycidae	<i>Monochamus</i>	<i>Monochamus titillator</i>	2
Cerambycidae	<i>Neoclytus</i>	<i>Neoclytus acuminatus</i>	60
Cerambycidae	<i>Neoclytus</i>	<i>Neoclytus caprea</i>	2
Cerambycidae	<i>Neoclytus</i>	<i>Neoclytus horridus</i>	2
Cerambycidae	<i>Neoclytus</i>	<i>Neoclytus jouteli</i>	1
Cerambycidae	<i>Neoclytus</i>	<i>Neoclytus mucronatus</i>	133
Cerambycidae	<i>Neoclytus</i>	<i>Neoclytus scutellaris</i>	129
Cerambycidae	<i>Necydalis</i>	<i>Necydalis mellita</i>	2

Table 6 (cont.). Species collected, including total number of specimens. New state records are indicated by an asterisk (*).

Family	Genus	Species	Total specimens collected
Cerambycidae	<i>Oberea</i>	<i>Oberea ulmicola</i>	1
Cerambycidae	<i>Obrium</i>	<i>Obrium maculatum</i>	10
Cerambycidae	<i>Oncideres</i>	<i>Oncideres cingulata</i>	2
Cerambycidae	<i>Orthosoma</i>	<i>Orthosoma brunneum</i>	7
Cerambycidae	<i>Parelaphidion</i>	<i>Parelaphidion aspersum</i>	7
Cerambycidae	<i>Phymatodes</i>	<i>Phymatodes amoenus</i>	2
Cerambycidae	<i>Phymatodes</i>	<i>Phymatodes testaceus</i>	8
Cerambycidae	<i>Phymatodes</i>	<i>Phymatodes varius</i>	4
Cerambycidae	<i>Physocnemum</i>	<i>Physocnemum brevilineum</i>	1
Cerambycidae	<i>Prionus</i>	<i>Prionus imbricornis</i>	1
Cerambycidae	<i>Purpuricenus</i>	<i>Purpuricenus humeralis</i>	1
Cerambycidae	<i>Purpuricenus</i>	<i>Purpuricenus paraxillaris</i>	13
Cerambycidae	<i>Saperda</i>	<i>Saperda discoidea</i>	9
Cerambycidae	<i>Saperda</i>	<i>Saperda imitans</i>	29
Cerambycidae	<i>Saperda</i>	<i>Saperda lateralis</i>	9
Cerambycidae	<i>Saperda</i>	<i>Saperda tridentata</i>	3
Cerambycidae	<i>Sarosesthes</i>	<i>Sarosesthes fulminans</i>	5
Cerambycidae	<i>Stenocorus</i>	<i>Stenocorus cinnamopterus</i>	7
Cerambycidae	<i>Stenosphenus</i>	<i>Stenosphenus notatus</i>	73
Cerambycidae	<i>Sternidius</i>	<i>Sternidius alpha</i>	6
Cerambycidae	<i>Strangalepta</i>	<i>Strangalepta abbreviata</i>	1
Cerambycidae	<i>Strangalia</i>	<i>Strangalia bicolor</i>	31
Cerambycidae	<i>Strangalia</i>	<i>Strangalia luteicornis</i>	205
Cerambycidae	<i>Strophiona</i>	<i>Strophiona nitens</i>	24
Cerambycidae	<i>Tilloclytus</i>	<i>Tilloclytus geminatus</i>	2
Cerambycidae	<i>Trachysida</i>	<i>Trachysida mutabilis</i>	2
Cerambycidae	<i>Trigonarthris</i>	<i>Trigonarthris minnesotana</i>	2
Cerambycidae	<i>Trigonarthris</i>	<i>Trigonarthris proxima</i>	3
Cerambycidae	<i>Typocerus</i>	<i>Typocerus lugubris</i>	2
Cerambycidae	<i>Typocerus</i>	<i>Typocerus velutinus</i>	46
Cerambycidae	<i>Typocerus</i>	<i>Typocerus zebra</i>	5
Cerambycidae	<i>Urgleptes</i>	<i>Urgleptes querci</i>	28
Cerambycidae	<i>Urgleptes</i>	<i>Urgleptes signatus</i>	9
Cerambycidae	<i>Xylotrechus</i>	<i>Xylotrechus colonus</i>	360

Table 6 (cont.). Species collected, including total number of specimens. New state records are indicated by an asterisk (*).

Family	Genus	Species	Total specimens collected
Curculionidae	<i>Acalles</i>	<i>Acalles carinatus</i>	11
Curculionidae	<i>Acalles</i>	<i>Acalles clavatus</i> *	5
Curculionidae	<i>Acalles</i>	<i>Acalles minutissimus</i> *	5
Curculionidae	<i>Acoptus</i>	<i>Acoptus suturalis</i> *	1
Curculionidae	<i>Anthonomus</i>	<i>Anthonomus juniperinus</i> *	1
Curculionidae	<i>Anthonomus</i>	<i>Anthonomus nigrinus</i>	3
Curculionidae	<i>Anthonomus</i>	<i>Anthonomus rufipennis</i>	5
Curculionidae	<i>Anthonomus</i>	<i>Anthonomus suturalis</i>	22
Curculionidae	<i>Aphanommata</i>	<i>Aphanommata tenuis</i>	9
Curculionidae	<i>Apteromechus</i>	<i>Apteromechus ferratus</i>	600
Curculionidae	<i>Anametis</i>	<i>Anametis granulata</i> *	5
Curculionidae	<i>Auleutes</i>	<i>Auleutes nebulosus complex</i>	2
Curculionidae	<i>Buchananius</i>	<i>Buchananius sulcatus</i>	4
Curculionidae	<i>Canistes</i>	<i>Canistes schusteri</i>	26
Curculionidae	<i>Caulophilus</i>	<i>Caulophilus dubius</i>	1
Curculionidae	<i>Cercopeus</i>	<i>Cercopeus chrysorrhoeus</i>	560
Curculionidae	<i>Chalcodermus</i>	<i>Chalcodermus inaequicollis</i>	1
Curculionidae	<i>Conotrachelus</i>	<i>Conotrachelus affinis</i>	9
Curculionidae	<i>Conotrachelus</i>	<i>Conotrachelus anaglypticus</i>	39
Curculionidae	<i>Conotrachelus</i>	<i>Conotrachelus aratus</i>	162
Curculionidae	<i>Conotrachelus</i>	<i>Conotrachelus carinifer</i>	56
Curculionidae	<i>Conotrachelus</i>	<i>Conotrachelus elegans</i>	44
Curculionidae	<i>Conotrachelus</i>	<i>Conotrachelus naso</i>	130
Curculionidae	<i>Conotrachelus</i>	<i>Conotrachelus posticatus</i>	979
Curculionidae	<i>Cophes</i>	<i>Cophes fallax</i>	73
Curculionidae	<i>Cophes</i>	<i>Cophes obtentus</i>	1
Curculionidae	<i>Cossonus</i>	<i>Cossonus impressifrons</i>	12
Curculionidae	<i>Craponius</i>	<i>Craponius inaequalis</i>	1
Curculionidae	<i>Cryptorhynchus</i>	<i>Cryptorhynchus fuscatus</i>	6
Curculionidae	<i>Cryptorhynchus</i>	<i>Cryptorhynchus tristis</i>	168
Curculionidae	<i>Curculio</i>	<i>Curculio othorhynchus</i>	1
Curculionidae	<i>Cyrtepistomus</i>	<i>Cyrtepistomus castaneus</i>	133
Curculionidae	<i>Dichoxenus</i>	<i>Dichoxenus setiger</i>	76
Curculionidae	<i>Dietzella</i>	<i>Dietzella zimmermanni</i>	1
Curculionidae	<i>Dryophthorus</i>	<i>Dryophthorus americanus</i>	30
Curculionidae	<i>Epacalles</i>	<i>Epacalles inflatus</i>	65
Curculionidae	<i>Eubulus</i>	<i>Eubulus bisignatus</i>	28
Curculionidae	<i>Eubulus</i>	<i>Eubulus obliquefasciatus</i>	193

Table 6 (cont.). Species collected, including total number of specimens. New state records are indicated by an asterisk (*).

Family	Genus	Species	Total specimens collected
Curculionidae	<i>Eudociminus</i>	<i>Eudociminus mannerheimii</i>	1
Curculionidae	<i>Eurhoptus</i>	<i>Eurhoptus</i> sp. 1	28
Curculionidae	<i>Eurhoptus</i>	<i>Eurhoptus pyriformis</i>	15
Curculionidae	<i>Geraeus</i>	<i>Geraeus penicillus</i>	1
Curculionidae	<i>Hypera</i>	<i>Hypera compta</i>	4
Curculionidae	<i>Hypera</i>	<i>Hypera meles</i>	19
Curculionidae	<i>Hypera</i>	<i>Hypera nigrirostris</i>	1
Curculionidae	<i>Hypera</i>	<i>Hypera postica</i>	1
Curculionidae	<i>Idiostethus</i>	<i>Idiostethus subcalvus</i> *	1
Curculionidae	<i>Laemosaccus</i>	<i>Laemosaccus nephele</i> group	3
Curculionidae	<i>Leichrops</i>	<i>Lechriops oculatus</i>	30
Curculionidae	<i>Lymantes</i>	<i>Lymantes sandersoni</i>	1
Curculionidae	<i>Madarellus</i>	<i>Madarellus undulatus</i> *	9
Curculionidae	<i>Magdalis</i>	<i>Magdalis armicollis</i> *	3
Curculionidae	<i>Magdalis</i>	<i>Magdalis barbata</i> *	5
Curculionidae	<i>Mecinus</i>	<i>Mecinus pascuorum</i> *	2
Curculionidae	<i>Myrmex</i>	<i>Myrmex chevrolatii</i> *	7
Curculionidae	<i>Myrmex</i>	<i>Myrmex myrmex</i> *	1
Curculionidae	<i>Nicentrus</i>	<i>Nicentrus lecontei</i> *	1
Curculionidae	<i>Oopterinus</i>	<i>Oopterinus perforatus</i>	17
Curculionidae	<i>Otiorhynchus</i>	<i>Otiorhynchus rugosostriatus</i> *	46
Curculionidae	<i>Pandeletius</i>	<i>Pandeletius hilaris</i>	51
Curculionidae	<i>Piazorhinus</i>	<i>Piazorhinus pictus</i> *	2
Curculionidae	<i>Phyllotrox</i>	<i>Phyllotrox ferrugineus</i> *	20
Curculionidae	<i>Plocamus</i>	<i>Plocamus hispidulus</i> *	1
Curculionidae	<i>Pseudobaris</i>	<i>Pseudobaris nigrina</i> *	9
Curculionidae	<i>Pseudopentarthrum</i>	<i>Pseudopentarthrum simplex</i> *	13
Curculionidae	<i>Rhinoncus</i>	<i>Rhinoncus pericarpus</i> *	1
Curculionidae	<i>Sitona</i>	<i>Sitona lineatus</i> *	1
Curculionidae	<i>Stenoscelis</i>	<i>Stenoscelis brevis</i> *	4
Curculionidae	<i>Tachyerges</i>	<i>Tachyerges niger</i>	1
Curculionidae	<i>Tomolips</i>	<i>Tomolips quercicola</i> *	2
Curculionidae	<i>Tychius</i>	<i>Tychius prolixus</i>	7
Curculionidae	<i>Tyloderma</i>	<i>Tyloderma foveolatum</i>	1

Table 6 (cont.). Species collected, including total number of specimens. New state records are indicated by an asterisk (*).

Thirty one species (12%) collected during this study represent new Arkansas state records: (Buprestidae) *Actenodes acornis*, *Agrilus cephalicus*, *Agrilus ohioensis*, *Agrilus*

paracelti, *Taphrocerus nicolayi*; (Carabidae) *Agonum punctiforme*, *Synuchus impunctatus*; (Curculionidae) *Acalles clavatus*, *Acalles minutissimus*, *Acoptus suturalis*, *Anthonomus juniperinus*, *Anametis granulata*, *Eudociminus mannerheimii*, *Idiostethus subcalvus*, *Madarellus undulatus*, *Magdalis armicollis*, *Magdalis barbata*, *Mecinus pascuorum*, *Myrmex chevrolatii*, *Myrmex myrmex*, *Nicentrus lecontei*, *Otiorhynchus rugostriatus*, *Piazorhinus pictus*, *Phyllotrox ferrugineus*, *Plocamus hispidulus*, *Pseudobaris nigrina*, *Pseudopentarthrum simplex*, *Rhinoncus pericarpus*, *Sitona lineatus*, *Stenoscelis brevis*, *Tomolips quericola*.

Three endemic carabids – *Cyclotrachelus parasodalis*, *Rhadine ozarkensis*, *Scaphinotus infletus* – were also collected.

Notes on select species

Agrilus ohioensis has been recorded from many eastern states, but is rarely collected. Larvae have been reported from American hornbeam, *Carpinus caroliniana* Walter, (Nelson and MacRae 1990, Wellso and Jackman 2006) and winged elm, *Ulmus alata* Michx., (Nelson et al. 1981), both of which are present at the site. One reason for their apparent rarity may be from a lack of specialized collecting. Collecting small branches of hosts and rearing specimens is a specialized technique frequently used by wood borer enthusiasts. More work of this nature with these and other hosts should yield a wider distribution for this species and many other "rare" buprestids, including *Agrilus cephalicus*.

Agonum punctiforme occurs from North Carolina to southeastern Texas, with a record from Missouri that "needs confirmed", and *Amara cupreolata* has been previously recorded in Arkansas but "the record needs confirmation" (Bousquet 2012a), so it is unsurprising the species were collected in Arkansas.

Cyclotrachelus parasodalis is an Arkansas endemic which has only been reported in the literature a handful of times, including the original description and description of the larvae (Freitag 1969, Allen and Thompson 1977, Thompson 1979, Hamilton 2015). Approximately 3,000 specimens are housed in the UAAM collection, most of which coincide with the collection localities and dates given by Allen and Thompson 1977, though the authors did not provide specific label data or the number of specimens collected per site in the publication (Fig. 3). Given the abundance of specimens and apparently wide range within the state, it is surprising the species has not been recorded in Missouri or Oklahoma sections of the Interior Highlands. Additionally, two specimens collected in cotton fields in the Mississippi Alluvial Plain indicate the species is not restricted entirely to the Interior Highlands, though it may be endemic to the region immediately surrounding the Interior Highlands.

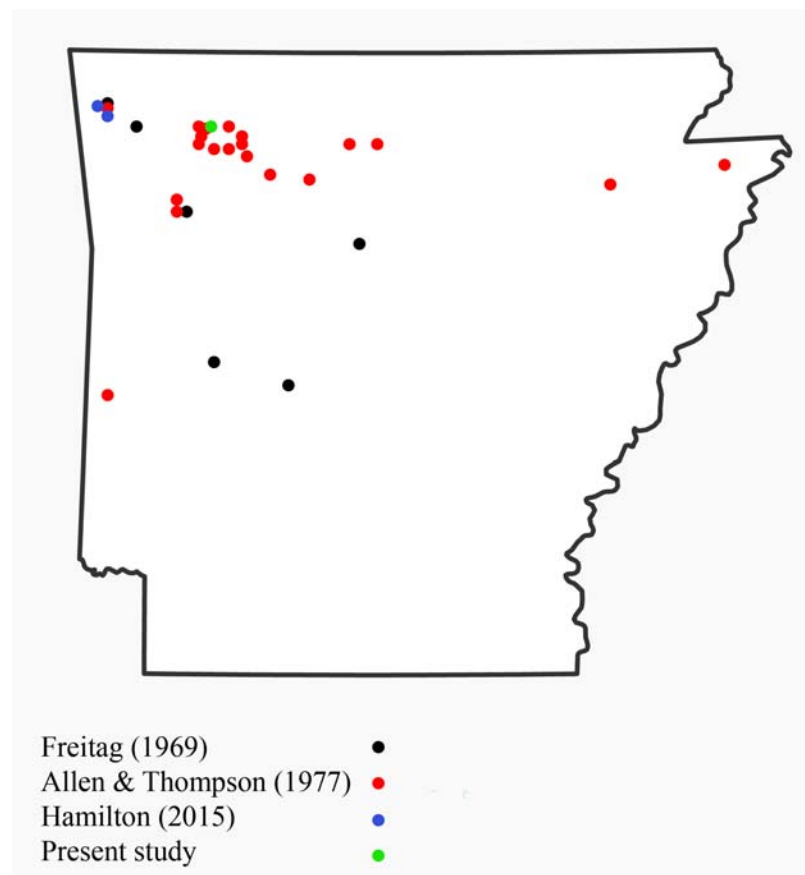


Figure 3. Known collection localities of *Cyclotrachelus parasodalis*.

Rhadine ozarkensis is previously known only from the type series collected in Fincher's Cave, near Black Oak, Arkansas (Washington County, not Craighead County) (Barr 1960, Bousquet 2012b). This specimen represents a range expansion of over 65 km. That it was collected in a pitfall trap on the surface suggests that the species may not be restricted to caves or can move between suitable cave habitat using the karst topography of the region.

Pterostichus punctiventris ranges from northern Georgia south to Alabama west to east-central Missouri, eastern Oklahoma, and Texas (Bousquet 2012b). It is apparently known from a limited number of specimens and localities; in Arkansas, it has only been collected previously in Blanchard Springs State Park in Stone County (Bousquet 1992).

Scaphinotus infletus is known from only three specimens collected from three localities within 30 km of the study site (Allen and Carlton 1988, Bousquet 2012b). This specimen represents a new locality for the species and confirms its presence in the area after nearly thirty years without being collected.

Synuchus impunctatus is known from Missouri and Kansas, but has not previously been recorded from Arkansas (Bousquet 2012b).

Tachys columbiensis was thought to be confined to the Coastal Plain and Piedmont Plateau, ranging from southeastern Pennsylvania to southern Florida west to Mississippi and eastern Texas, though it has also been recorded from central Arkansas (Pulaski and Garland Counties) (Bousquet 2012b). These specimens represent a new northwestern range limit and a new physiogeographic region (Ozark Mountains) for the species.

Trichotichnus vulpeculus is recorded from western New Brunswick south to eastern Georgia, west to Wisconsin and northern Arkansas (Bousquet 2012b). These specimens are therefore likely near the southwestern range limit for this species.

Acalles clavatus was previously known from Florida, South Carolina and Louisiana (Ciegler 2010, O'Brien and Wibmer 1982); it has been reared from small twigs of *Quercus falcata* Michaux (Ferro et al. 2009).

Acoptus suturalis is known from northeastern North America, from Quebec south to North Carolina and Illinois and Iowa; additional records are known from Georgia and Mexico (O'Brien and Wibmer 1982). It has been raised from the branch of an American elm (*Ulmus americana* L.) and may be a vector of butternut canker virus (*Sirococcus clavigignenti-juglandacearum*) in butternut (*Juglans cinerea* L.) (Hoffman 1942, Halik and Bergdahl 2002).

Anametis granulata is found in northern and eastern North America, from Newfoundland and Quebec, south to New Jersey, west to Missouri, Wyoming and Montana; additional specimens are known from Texas, New Mexico, and Mexico (O'Brien and Wibmer 1982, Ocaña 1996).

Anthonomus juniperinus is known from the eastern United States, from Massachusetts south to Florida, west to West Virginia, as well as Texas, Oregon, and Paget, Bermuda (O'Brien and Wibmer 1982, Clark and Burke 2010). It feeds on *Gymnosporangium juniperi-virginianae* Schwein., a fungus parasitic on *Juniperus* L., and juniper berries (Ciegler 2010, Clark and Burke 2010).

Buchananianus sulcatus is widely distributed in the eastern and southeastern United States (O'Brien and Wibmer 1982). It has been reared from the fruiting bodies of the ascomycete

fungus *Trichoderma peltatum* (Berk.) Samuels, Jaklitsch, and Voglmayr (Prena et al. 2014) and adults have been collected in leaf litter and under branches (Kissinger 1957).

Caulophilus dubius is known from Quebec and New York south to Georgia, west to Illinois and Mississippi, as well as Texas (O'Brien and Wibmer 1982, Douglas et al. 2013). Adults are found beneath dead tree bark and in tree holes (Blatchley and Leng 1916, Ciegler 2010).

Eubulus bisignatus is widespread in eastern and southern North America, ranging from Ontario south to Florida, west to Nebraska, Texas, Arizona, and California; it is also recorded from Mexico and Guatemala. It was not recorded from Arkansas by O'Brien and Wibmer 1982 but was reported by Anderson 2008. Adults are frequently collected at lights and in Malaise and flight-intercept traps and have been collected from a number of hardwood species including *Quercus* L., *Castanea* Mill., *Fagus* L., *Betula* L., *Carya* Nutt., and *Acer* L. (Anderson 2008).

Eubulus obliquefasciatus is commonly collected in flight-intercept traps and at lights. Adults have been collected on dead oak and sweetgum; otherwise, nothing is known about their biology (Anderson 2008).

The *Eudociminus mannerheimii* specimen collected during this study was included with other specimens collected near the field site in a forthcoming publication (Skvarla et al. in press [Chapter IX]) that suggests eastern red cedar (*Juniperus virginiana* L.) as a possible host as it is the only species of Cupressaceae present at the site. Additionally, the specimens represented a new state record and northwestern range expansion from previous records.

Idiostethus subcalvus is found from Pennsylvania south to South Carolina, west to Illinois and Missouri (O'Brien and Wibmer 1982, Ciegler 2010). Downie 1958 reported it is "very abundant" in April and May in Indiana. It been taken on *Caulophyllum thalictroides* (L.)

Michaux, *Hydrophyllum appendiculatum* Michx., *Phacelia* Juss. and *Ranunculus hispidus* Michx. var. *nitidus* (Chapm.) T. Duncan (Robertson 1929, Ciegler 2010, Graham et al. 2012).

Madarellus undulatus is found in eastern North America, from Quebec and Connecticut south to Florida, west to South Dakota, Kansas, and Missouri (O'Brien and Wibmer 1982). It has been collected with black pyramid traps (Bloem et al. 2002), Malaise traps, fogging (Werle 2002) and at lights (Ciegler 2010). Larvae have been reported to feed on *Vitis* L., *Toxicodendron radicans* (L.) Kuntze and *Parthenocissus quinquefolia* (L.) Planch. (Blatchley and Leng 1916, Bouchard et al. 2005).

Magdalis armicollis is found in the eastern United States from Connecticut south to Georgia, west to North Dakota, Montana, Nebraska, and Texas (O'Brien and Wibmer 1982, Quinn 2000). Larvae mine galleries in stressed, dying, and dead *Ulmus* L. and adults feed on the leaves (Blatchley and Leng 1916, Hoffman 1942, Majka et al. 2007). Larval feeding is generally confined to branches smaller than 7.5 cm; however, in large numbers, larval and adult feeding can cause significant damage that may result in tree death (Baker 1941, Booth and Johnson 2009). *Magdalis armicollis* is not a vector of Dutch elm disease (Goeden and Norris 1963).

Magdalis barbata is found in North America from Connecticut and Ontario south to Georgia, west to Montana, Texas, Nevada, and California (O'Brien and Wibmer 1982). Larvae mine galleries in the branches of dead and dying *Quercus*, *Ulmus*, and *Carya* and adults feed on the leaves of *Ulmus* (Blatchley and Leng 1916, Hoffman 1942, Majka et al. 2007). *Magdalis barbata* is not a vector of Dutch elm disease (Goeden and Norris 1963).

Myrmex myrmex is native to the eastern United States, from Connecticut south to Florida, west to Indiana and Iowa (O'Brien and Wibmer 1982). It develops in the dead and dying wood of sycamore (Burke et al. 1975), which was present in small numbers at the site.

Notiodes limatulus is widespread in North America, ranging from New York south to Georgia, west to Idaho, Texas, and California, and into Mexico. It was not recorded in Arkansas by O'Brien and Wibmer 1982 but was reported in the state by O'Brien and Anderson 1996.

Otiorhynchus rugostriatus is adventive from Europe and has been established in North America since 1876; it is now widespread through the United States and Canada (O'Brien and Wibmer 1982, Mattson et al. 1994). Larvae feed on roots of Rosaceae and other plants (Mattson et al. 1994).

Rhinoncus pericarpus is adventive from the Palaearctic (Majka et al. 2007). It was first recorded in northeastern North America in 1895 and the Pacific Northwest in 1913; in the east it is known from Nova Scotia south through Georgia, west to Illinois (O'Brien and Wibmer 1982, Majka et al. 2007). *Rhinoncus pericarpus* is reported to feed on *Rumex* L. and *Cannabis* L. and have been collected from *Rheum* L. and *Medicago sativa* L. (Harada 1930, Hoebeke and Whitehead 1980).

Stenoscelis brevis is widespread in eastern North America, from Ontario and Quebec south to Florida, west to Wisconsin, Kansas, and Mississippi (O'Brien and Wibmer 1982). Larvae bore under the bark of dead hardwood (O'Brien 1997). Adults have been collected in Lindgren multifunnel traps baited with manuka oil, from leaf litter using Berlese extraction and under the bark of dead trees (Johnson et al. 2014, Ferro et al. 2012).

Tachyerges niger was not reported from Arkansas by O'Brien and Wibmer 1982 but was recorded from the state by Sweeney et al. 2012; it is associated with *Salix* L.

Tychius picirostris is adventive from Europe and widely established in North America (Anderson and Howden 1994).

Discussion

It is unsurprising that few Carabidae represented new state records as carabid workers formerly associated with the University of Arkansas (e.g., R. T. Allen, C. E. Carlton, R. G. Thompson) have heavily sampled the region. Conversely, nearly one in five Buprestidae (19%) and one in three Curculionidae (32%) collected during this study represent new state records. Such high percentages of unrecorded species in charismatic and diverse taxa highlights how little attention many groups have received in the state and how much basic science and natural history is left to be done in 'The Natural State'.

Buprestids are capable of flying between habitat patches and rapidly colonizing new areas, so it is unlikely that new species will be discovered even though buprestids are understudied in the Interior Highlands. However, considering the high number of endemic species that are restricted to leaf litter habitats or are poor dispersers, how relatively understudied leaf litter weevils are, and that known but undescribed species were collected during this study, it is likely that the Interior Highlands is a fruitful area for finding new and disjunct weevil species.

Acknowledgements.

We thank Peter Messer for confirming the identity of *Rhadine ozarkensis* and other carabids; Robert Anderson for confirming the identity of *Eurhoptus* species; Ted MacRae for confirming new buprestid state records; and Hailey Higgins for curating and identifying cerambycid specimens. This project and the preparation of this publication was funded in part by the State Wildlife Grants Program (Grant # T39-05) of the U.S. Fish and Wildlife Service through an agreement with the Arkansas Game and Fish Commission.

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Appendix I. Dataset description.

Column label	Column description
typeStatus	Nomenclatural type applied to the record
catalogNumber	Unique within-project and within-lab number applied to the record
recordedBy	Who recorded the record information
individualCount	The number of specimens contained within the record
lifeStage	Life stage of the specimens contained within the record
kingdom	Kingdom name
phylum	Phylum name
class	Class name
order	Order name
family	Family name
genus	Genus name
specificEpithet	Specific epithet
scientificNameAuthorship	Name of the author of the lowest taxon rank included in the record
scientificName	Complete scientific name including author and year
taxonRank	Lowest taxonomic rank of the record
country	Country in which the record was collected
countryCode	Two-letter country code
stateProvince	State in which the record was collected
county	County in which the record was collected
municipality	Closest municipality to where the record was collected
locality	Description of the specific locality where the record was collected
verbatimElevation	Average elevation of the field site in meters
verbatimCoordinates	Approximate center point coordinates of the field site in GPS coordinates
verbatimLatitude	Approximate center point latitude of the field site in GPS coordinates
verbatimLongitude	Approximate center point longitude of the field site in GPS coordinates
decimalLatitude	Approximate center point latitude of the field site in decimal degrees
decimalLongitude	Approximate center point longitude of the field site in decimal degrees

Table A1. Column headings and description of column data of the data set.

Column label	Column description
georeferenceProtocol	Protocol by which the coordinates were taken
identifiedBy	Who identified the record
eventDate	Date or date range the record was collected
habitat	Description of the habitat
language	Two-letter abbreviation of the language in which the data and labels are recorded
institutionCode	Name of the institution where the specimens are deposited
basisofRecord	The specific nature of the record

Table A1 (cont.). Column headings and description of column data of the data set.

V. Collecting beetles: Analysis of a single-site data set and comparison of trapping techniques (Coleoptera: Carabidae, Buprestidae, Cerambycidae, Curculionoidea excluding Scolytinae)

Abstract.

Beetles (Coleoptera) are a charismatic group of insects targeted by collectors and often used in biodiversity surveys. As part of a larger project, we surveyed a small (4 hectare) plot in the Boston Mountains of Arkansas using 70 traps of 12 trap types and Berlese-Tullgren extraction of leaf litter and identified all Buprestidae, Carabidae, Cerambycidae, and Curculionoidea excluding Scolytinae to species. This resulted in the collection of 7973 specimens representing 242 species arranged in 8 families. The combination of pitfall and Malaise traps effectively collected Carabidae, Cerambycidae, and Curculionoidea while Buprestidae were most effectively collected by Malaise and green Lindgren funnel traps. Species accumulation curves based on the data did not become asymptotic and extrapolated rarefaction curves did not become asymptotic until 350–1000 samples, suggesting that much more effort is required to completely inventory even a small site. Additionally, seasonal activity is presented for each species and the similarity and overlap between collecting dates and seasons is discussed for each family.

Introduction.

We are currently in the midst of a global extinction crisis as species are becoming extinct at rates 100-10,000 times greater than historic background rates, with some suggesting it is the beginning of the sixth mass extinction (Pimm et al. 1995; Balmford 1996; Wake & Vredenburg 2008; Barnosky et al. 2011; Voss et al. 2015). Vascular plants, vertebrates, and invertebrates are

all experiencing higher-than-average extinction rates, with invertebrates experiencing some of the most rapid declines (Conrad et al. 2006; Thomas *et al.* 2004). Averting this crisis has become a priority among biologists.

One of many proposed ways of solving the global extinction crisis is to protect biodiversity hotspots, which are those areas of high biodiversity and endemism (Médail & Quézel 1999). Examples of such hotspots are the Mediterranean biome, which comprises 2% of the world's surface but contains 20% of the total floristic richness, and the tropical Andes, which contains 6.7% and 5.7% of the world's total plant and vertebrate species, respectively (Médail & Quézel 1997; Meyers et al. 2000). By focusing on protecting these areas instead of individual species it is possible to protect large percentages of biodiversity in the most spatially- and monetarily-efficient manner (Meyers 1989; Meyers 1990).

Known hotspots in the U.S. include the southern Appalachians, temperate rainforests of the Northwest, and southern California. The Interior Highlands (Fig. 1) comprise some of the oldest continuously exposed land worldwide and have been proposed to be a hotspot on par with these (The Nature Conservancy, Ozarks Ecoregion Assessment Team 2003; Skvarla et al. 2015 [Chapter VIII]). Many species found in the Interior Highlands are characteristic of other refugia, such as the southern Appalachians and the Sierra Madre in Mexico and over 200 species are known to be endemic to the region (Allen 1990; Robison & Allen 1995; Redfearn 1986; Skvarla et al. 2015 [Chapter VIII]). Still, in comparison to other regions of hyperdiversity, the Interior Highlands remain understudied, especially with regards to terrestrial invertebrates, which are vital components of biodiversity and play important roles in pollination, decomposition, soil formation and fertility, nutrient turnover, and population regulation of other organisms through parasitism and predation (Daily et al. 1997; Yen & Butcher 1997; Wickings & Grandy 2011).

They have also been shown to be important indicators of environmental change and can be used to assess conservation and biodiversity (Ward & Larivière 2004).



Figure 1. View of the Ozarks from an overlook on the Buffalo River Trail near Steel Creek.

Arthropods, however, are often ignored because they are considered too difficult to deal with: sampling arthropods produces thousands to millions of specimens that must be curated and identified; many species are still undescribed; and there are few useable keys and fewer experts to consult about identification (Ward & Larivière 2004). Rapid biodiversity assessment (RBA) approaches, which aim to reduce cost and effort, have been suggested to circumvent these problems. RBA approaches fall into four categories: (1) restricted sampling in place of intensive sampling (sampling surrogacy); (2) use of higher taxonomic levels other than species (species surrogacy); (3) the use of morphospecies (otherwise known as recognizable taxonomic units or

parataxonomic unit) identified by non-specialists (taxonomic surrogacy); and (4) the use of surrogate taxa in place of all taxa (taxon-focusing) (Ward & Larivière 2004).

Sampling surrogacy involves some manner of reduced sampling, including but not limited to shorter sampling duration, reduced number of sampling methods, and sub-sampling existing material. A few limited studies have shown that such reduced sampling, if done correctly, can be used in place of more intensive sampling (e.g., Niemelä et al. 1990; Hammond 1994; Sparrow et al. 1994; Samu & Lövei 1995). Care must be taken, however, as sampling methods do not evenly collect species (e.g., pitfall traps: Skvarla et al. 2014 [Chapter II]) and some species are active for very brief periods of time.

Species surrogacy involves using higher taxonomic levels, such as genus or family, instead of species. This method has the benefits of being less time- and resource-intensive as these levels are generally much easier to identify. Species surrogacy can be used confidently in areas where the relationship between genera and species is near 1:1; for example, Pik et al. (1999) and Neville and New (1999) demonstrated such a relationship within ants in forested areas of Australia. In areas that such a relationship does not hold species surrogacy can severely skew any estimate of species richness. Depending on the level chosen species surrogacy can also mask various qualities, such as differences in feeding types and trophic levels.

Taxonomic surrogacy is the use of morphospecies in place of species. The benefit of using morphospecies is that large quantities of prepared material can be processed by parataxonomists who do not possess extensive formal training in identification. Abadie et al. (2008) compared the accuracy of parataxonomic identifications of plants by volunteers with identifications performed by taxonomic experts and found that morphotype identification varied significantly between and within volunteers; morphotype identification was sensitive to

differences among habitats but less sensitive than species identification; and that the number of morphotypes identified by volunteers was strongly correlated with species-richness. Derrai et al. (2002) similarly found that volunteers correctly separated as species 91% of Lepidoptera but only 63% and 50% of Coleoptera and Araneae.

Taxonomic surrogacy also falls short because less can be done with the data generated outside of the project that generated the identifications. Reporting Carabidae sp.1, Carabidae sp. 2, and Carabidae sp. 3 is sufficient for generating a biodiversity index based on the number of species or for generating a species accumulation curve but is useless when trying to assess beta diversity between different habitats, identifying biodiversity hotspots and endemic species, or any other meaningful comparisons between the study site and other areas.

Taxon focusing includes a number of techniques that involve identifying a species or group of species in place of a wider range of species. These approaches assume that data and patterns from the identified species can be used to inform and protect the larger group of species. Few guidelines for choosing focal taxa have been suggested; as a result, focal taxa are generally chosen for practical reasons, such as ease of identification, personal interest, and prior use in similar hypotheses (New 1998; 1999). In addition, there is little evidence that the patterns of a handful of species can accurately predict or reflect larger biodiversity patterns (Prendergast et al. 1993; Lawton et al. 1998; Lindenmayer et al. 2002). Some authors have tried to work around this by analyzing many diverse organisms. For example, Kotze and Samways (1999) used Carabidae, Staphylinidae, and Formicidae and Lawton et al. (1998) examined Aves, Papilionoidea, Coleoptera, Formicidae, termites, and soil Nematodes.

As RBA approaches often cannot fully capture or predict the arthropod biodiversity of an area, more intensive surveys must be done in order to find and confirm biodiversity hotspots.

Although optimum sampling methods have been extensively tested for a few groups (e.g., ants: Agosti et al. 2000) for others groups they have not. Additionally, while certain collecting techniques are assumed to collect high diversity (e.g., Malaise and pitfall traps), few studies have actually tested those assumptions.

As part of a larger project examining the efficiency and overlap of various collecting techniques, we identified the Buprestidae, Cerambycidae, Carabidae, and Curculionoidea excluding Scolytinae. These families were chosen because, at least in the Nearctic where this study was conducted, they are generally easy to identified to family, have an abundance of material such as keys and checklists available to aid in identification, and we assumed it is easier for non-experts to switch between groups with similar morphology (e.g., different beetle families) than between groups with disparate morphology (e.g., beetles and flies or millipedes). The collection data and new state records of species in those families were reported in Chapter IV. Herein we analyze the data in order to compare and contrast the different collecting techniques within and between families and suggest the most efficient single and combined collection techniques.

Materials and Methods.

Site description

A 4 ha plot was established at Steel Creek along the Buffalo National River in Newton County, Arkansas, centered at approximately N 36°02.269', W 93°20.434'. The site is primarily mature second-growth deciduous forest dominated by oak (*Quercus* L. [Fagaceae]) and hickory (*Carya* Nutt. [Juglandaceae]), although American beech (*Fagus grandifolia* Ehrh. [Fagaceae]) and eastern red cedar (*Juniperus virginiana* L. [Cupressaceae]) are also abundant.

Sampling methods

The methods used were covered in detail in Chapter IV, so we provide the following summary: The following traps were maintained within the site: five Malaise traps (MegaView Science Co., Ltd., Taichung, Taiwan), twenty-five pan traps (five of each color: blue, purple, red, yellow, white) which were randomly arranged under the Malaise traps (one of each color) so as to also act as intercept traps; four SLAM (Sea, Land, and Air Malaise) traps (MegaView Science Co., Ltd., Taichung, Taiwan) with top and bottom collectors (Fig. 2); fifteen Lindgren multi-funnel traps (ChemTich International, S.A., Heredia, Costa Rica) (five of each color: black, green, purple) (Fig. 3); and seventeen pitfall trap sets (Fig. 4). Sixteen of the seventeen pitfall sets were placed in two transects of sets spaced every five meters centered on two Malaise traps while the final set was placed away from other traps (a third transect was planned, but eliminated due to the added collection time. The set placed away from the others is a remnant of that third transect). Additionally, ten leaf litter samples were collected for Berlese extraction when traps were serviced.

Pitfall traps were made using plastic soup containers based on a modified design proposed by Nordlander (1987). The pitfall traps had three slots measuring 2 cm tall x 9.3 cm wide cut 2 cm under the rim, resulting in three equidistant 1.5 cm posts and a 28 cm collecting surface. The diameter at the base of the slots was approximately 10.5 cm and the cups were 10.5 cm deep below the slots, resulting in a collecting volume of 2,988 cm³. The container lids were used as rain covers. Each pitfall trap set was made by burying a single cup on either side of a 30.5 cm x 15.5 cm aluminum fence; trap catch from both cups was combined and treated as a single sample.

Traps were placed non-randomly within the plot in order to maximize the efficiency of each trap, though an attempt was made to evenly space like-traps in order to decrease the chance of interference between traps. Malaise traps were placed in perceived flight paths. SLAM and Lindgren funnel traps were suspended from the branches of large trees 4–10 meters above the ground in the lower canopy; the location of appropriately sized trees dictated trap placement within each block.

Berlese-Tullgren samples were collected from a variety of habitats, including thin leaf litter on open ground; thick leaf litter accumulated along logs and rocks; moss; tree holes; bark from fallen, partially decayed trees; and bark and leaf litter accumulated at the base of standing, dead trees. Tree holes were only collected from once each so as not to totally destroy them as potential habitat; as the number of tree holes within the site was limited, this resulted in only a handful of collections from this habitat type. Litter was processed in the field using a litter reducer until approximately one gallon of processed litter was collected; this was stored in one gallon self-sealing bags during transport. Litter samples were collected after all traps had been serviced in order to reduce exposure to heat and reduce mortality of collected specimens. Leaf litter samples were processed for four to seven days until the litter was thoroughly dry using modified Berlese-Tullgren funnels.



Figures 2–4. **Fig. 2.** Malaise trap with pan traps underneath acting as intercept traps and a S.L.A.M. canopy trap. **Fig. 3.** Black Lindgren funnel trap. **Fig. 4.** Pitfall set. The canopy trap and Lindgren funnel trap were lowered from the canopy for the photographs.

All traps were set by 13 March 2013, except Lindgren funnels, which were set on 1 April 2013. Traps were serviced approximately every two weeks (14 days \pm 3 days). The final collection of pitfall traps and pan traps occurred on 6 November 2013 and the final collection of Malaise, SLAM, and Lindgren funnel traps occurred on 4 December 2013. Berlese-Tullgren samples from 13 April, 15 May, 28 June and 6 November were not taken or were lost. Pitfall sets were lost on 13 April (one set), 15 May (one set), 28 June (four sets), 17 July (five sets). In total, 1311 samples were collected.

Propylene glycol (Peak RV & Marine Antifreeze) (Old World Industries, LLC, Northbrook, IL) was used as the preservative in all traps as it is non-toxic and generally preserves specimens well (Skvarla et al. 2014 [Chapter II]). Trap catch was sieved in the field and stored in Whirl-Pak bags (Nasco, Fort Atkinson, WI) in 90% ethanol until sorting.

Sample preparation and identification

Samples were coarse-sorted using a Leica MZ16 stereomicroscope illuminated with a Leica KL1500 LCD light source and a Wild M38 stereomicroscope illuminated with an Applied Scientific Devices Corp. Eco-light 20 fiber optic light source. After sorting, specimens were stored individually or by family in 2 ml microtubes (VWR International, LLC, Randor, PA) in 70% ethanol. Hard-bodied specimens (e.g., Carabidae, Curculionidae) were pinned or pointed as appropriate.

Carabidae, Cerambycidae, and Curculionidae were identified with the use of published keys. In some cases, difficult-to-key specimens were photographed through the eye piece of the stereomicroscope using the camera on an HTC Droid Incredible 4G LTE cell phone or Samsung Galaxy S5 cell phone; the photographs were uploaded to Bugguide (Iowa State University 2015) and identifications were proposed by Bugguide members. Proposed identifications were then

checked using published sources and either confirmed or corrected on the website. Buprestidae were sent to Kyle Schnepf at the Florida State Collection of Arthropods for identification. Cerambycidae were identified by Hailey Higgins (University of Arkansas) as part of an undergraduate research project; identifications confirmed by the lead author.

One to five voucher specimens of each species have been retained in the Dowling Lab collection at the University of Arkansas while the remaining species have been submitted to the University of Arkansas Arthropod Museum (UAAM).

Statistical Analysis

Specimen abundance per trap per date was recorded in Excel (Microsoft 2013). For each family analyzed, the following procedures were followed:

A one-way analysis of variance (ANOVA) test ($\alpha = 0.05$) was performed in Excel to compare the effect of trap type on number of species and specimens. Due to uneven trapping effort and because traps were randomly lost due to rain and animal disturbance, we compared the average number of species and specimens collected per trap type per date after correcting for the number of traps per type (Eqs. 1, 2).

$$\frac{\sum \text{species per trap}}{\text{Number of traps}} \quad (1)$$

$$\frac{\sum \text{specimens per trap}}{\text{Number of traps}} \quad (2)$$

If a significant difference was detected, the means were separated using a Tukey-Kramer test ($\alpha = 0.05$) performed in Excel using the Real Statistics Resource Pack add-in (Zaiontz 2015). We chose to use ANOVA and Tukey-Kramer rather than their non-parametric equivalents as

both tests are relatively robust with respect to violations of the normality assumption (Kirk 1995; Samuels & Witmer 2003) and easily performed within Excel.

EstimateS (Colwell 2013) was used to calculate species accumulation estimators for each trap type using all samples collected per trap type: abundance coverage-based estimator of species richness (ACE) (Chao et al. 2000); incidence coverage-based estimator of species richness (ICE) (Chao et al. 2000); Chao 1 richness estimator (Chao1) (Chao 1984); Chao 2 richness estimator (Chao2) (Chao 1984, 1987); first-order Jackknife richness estimator (Jack1) (Burnham & Overton 1978, 1979); second-order Jackknife richness estimator (Jack2) (Burnham & Overton 1978, 1979) (see Gotelli & Colwell [2010] for a synopsis of each estimator).

Additionally, the sample-based rarefaction curve (S_{est}) (Colwell et al. 2004), which is the expected number of species in t pooled samples given the reference sample, was also calculated. EstimateS was run on default settings except that classic Chao1 and Chao2 estimators were used instead of the default bias-corrected Chao1 and Chao2 as suggested by the program. One hundred randomizations of sample order were performed in order to smooth the curves. As the various estimators generally calculated similar trends, we report only Chao1 estimators for each trap type per family in a single graph rather than all estimators per trap type in separate graphs for clarity and include graphs of all of the estimators in Appendix I. Because uneven sampling effort between trap types does not allow the number of species collected by each trap type to be directly compared, EstimateS was used to extrapolate the number of samples per trap type to 1000 samples, at which point the number of estimated species collected per trap type were compared. Samples were randomized across traps within a trap type and across dates. Error bars were excluded from accumulation and rarefaction graphs in order to enhance clarity.

Species similarity between trap types and seasonality was investigated by calculating shared species indices using EstimateS. EstimateS output was organized in Excel and final graphs were constructed in Adobe Illustrator (Adobe 2012). EstimateS calculates a number of different shared species estimators; herein we report the Sørensen similarity index, an incidence-based (i.e., presence/absence) index, and Chao's Sørensen similarity index, an abundance-based index (Chao et al. 2005). These indices indicate the similarity of the compared samples, which varies between 0 and 1 and indicate no to complete similarity. The statistical significance of similarity cannot be determined from these indices; therefore, when discussing the estimated similarity, we use the terms low (0–0.24), medium (0.25–0.49), high (0.50–0.74) and very high (0.75–1.0).

Shared species indices for trap types were calculated based on the total number of specimens per species collected per trap type. Shared species indices for collection dates were calculated based on the total specimens collected per species per date; the four trap types that collected the most species per family are reported.

The effect of Lindgren funnel trap color was investigated per species by performing a one-way ANOVA test ($\alpha = 0.05$) as described above on the total number of specimens collected per date by each color of Lindgren funnel when more than five specimens of a species were collected by any color of Lindgren funnel trap. Collection periods in which no beetles were collected by any trap were excluded from the analyses.

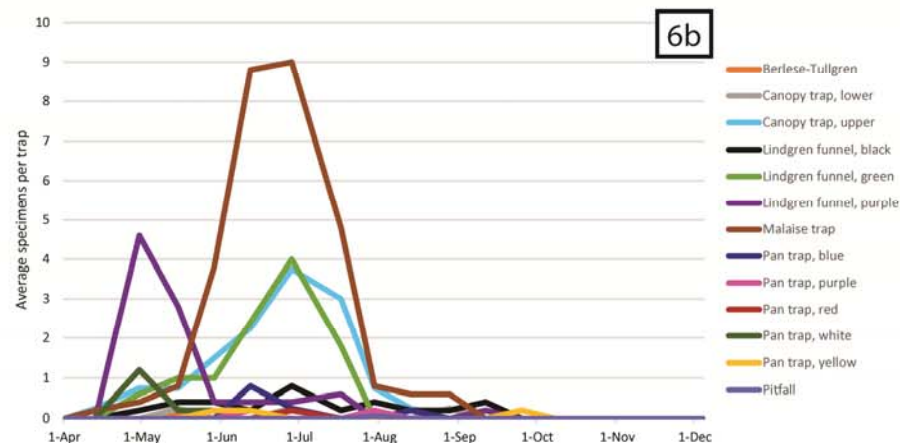
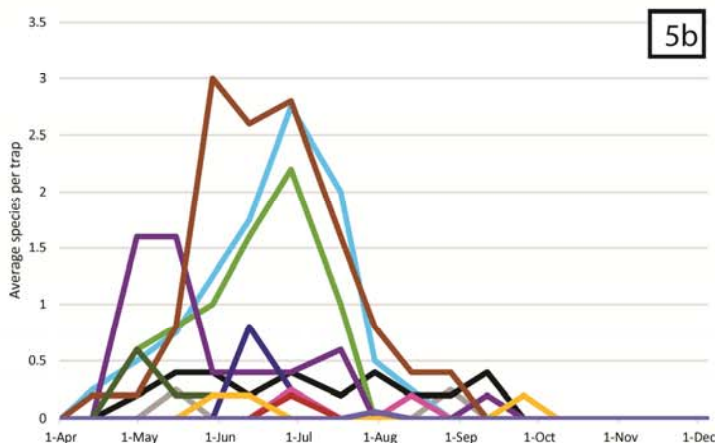
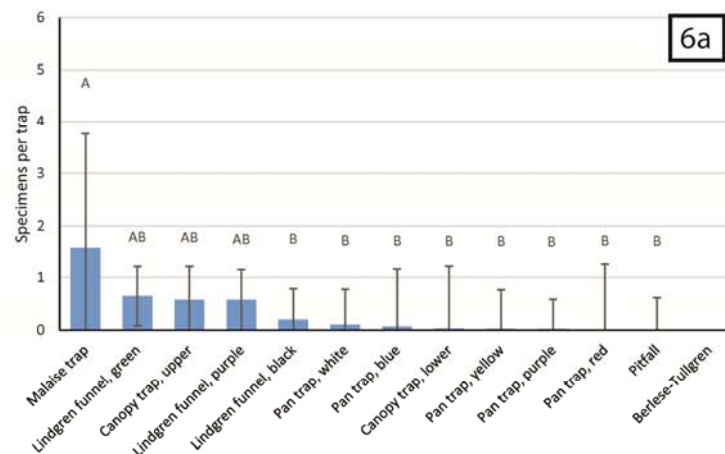
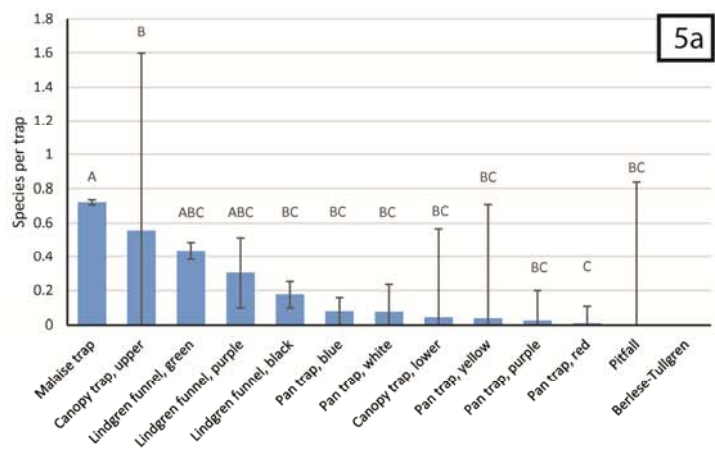
Results.

Buprestidae

Collection efforts resulted in 347 specimens representing 27 species. Malaise traps generally caught the most species (Figs. 5a,b) and specimens (Figs. 6a,b). Berlese-Tullgren

extraction of leaf litter produced no buprestids and was not considered in the analyses. Most species were represented by fewer than 20 specimens, with 11 species (41%) being represented by singletons (Fig. 7).

There was a significant ($p < 0.05$) effect of trap type on the number of species collected for the twelve trap types ($F(11,189) = 4.61$, $p = 1.40 \times 10^{-6}$). The mean number of species collected by Malaise traps ($M = 0.72$, $SD = 1.04$) was not significantly different from green Lindgren funnel traps ($M = 0.44$, $SD = 0.67$) and purple Lindgren funnel traps ($M = 0.31$, $SD = 0.52$) but was significantly different than all other trap types ($p > 0.05$, Tukey-Kramer). The mean number of species in upper canopy traps ($M = 0.56$, $SD = 0.83$) were significantly different from red pan traps and pitfall traps. All other trap types were not significantly different from each other: lower canopy trap ($M = 0.04$, $SD = 0.10$), black Lindgren funnel trap ($M = 0.18$, $SD = 0.17$), blue pan trap ($M = 0.08$, $SD = 0.21$), purple pan trap ($M = 0.03$, $SD = 0.08$), red pan trap ($M = 0.02$, $SD = 0.05$), white pan trap ($M = 0.08$, $SD = 0.16$), yellow pan trap ($M = 0.04$, $SD = 0.08$) ($p > 0.05$) (Fig. 5a).



Figures 5,6. Average number of buprestid species and specimens collected per trap. **Fig 5a.** Average number of species/trap. **Fig. 5b.** Average number of species/trap/date. **Fig. 6a.** Average number of specimens/trap. **Fig. 6b.** Average number of specimens/trap/date. **Figs. 5a,6a.** Bars indicate one standard deviation, letters indicate mean separation as determined by Tukey-Kramer test. **Figs. 5b, 6b.** Trap type indicated by the same color.

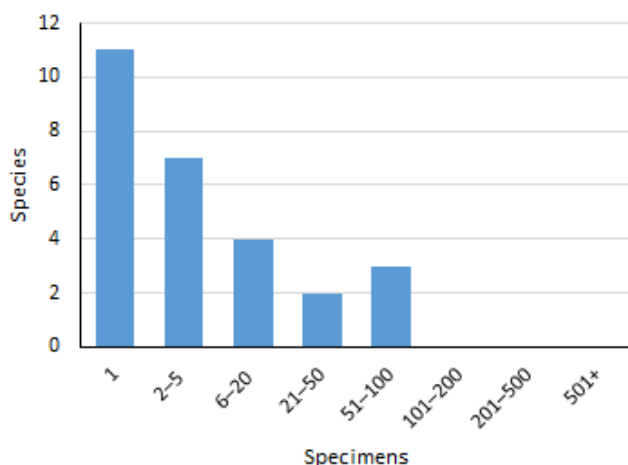


Figure 7. Total number of buprestid specimens/species collected across all traps.

There was a significant ($p < 0.05$) effect of trap type on the number of specimens collected for the twelve trap types ($F(11,189) = 3.79$, $p = 3.3 \times 10^{-5}$). The mean number of specimens collected by Malaise traps ($M = 1.66$, $SD = 2.95$) was not significantly different from green Lindgren funnel traps ($M = 0.65$, $SD = 1.13$), purple Lindgren funnel traps ($M = 0.55$, $SD = 1.24$), and upper canopy traps ($M = 0.74$, $SD = 1.15$) but was significantly different ($p < 0.05$) than all other trap types. All other trap types were not significantly different from each other: lower canopy trap ($M = 0.04$, $SD = 0.10$), black Lindgren funnel trap ($M = 0.1820$, $SD = 0.22$), blue pan trap ($M = 0.08$, $SD = 0.21$), purple pan trap ($M = 0.03$, $SD = 0.07$), red pan trap ($M = 0.01$, $SD = 0.05$), white pan trap ($M = 0.12$, $SD = 0.30$), yellow pan trap ($M = 0.04$, $SD = 0.08$) ($p < 0.05$, Tukey-Kramer) (Fig. 6a).

The effects of the color of Lindgren funnel traps was tested for seven species. Color had a significant ($p < 0.05$, Tukey-Kramer) effect on the number of specimens collected for six species; the mean number of specimens was significantly higher in green traps for three species, significantly higher in black and purple traps for one species each, and significantly higher in both green and purple traps for one species (Table 1).

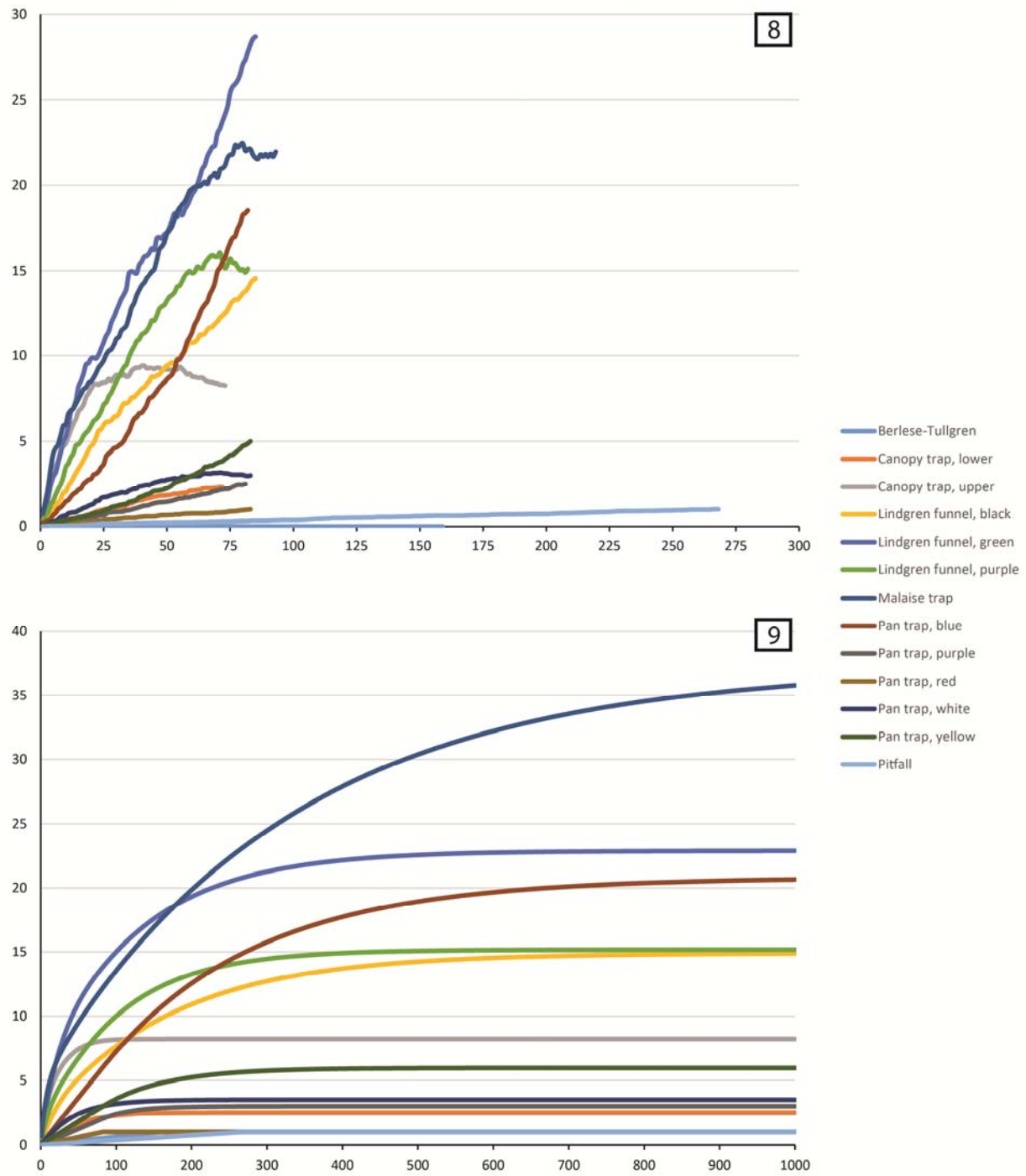
Species	ANOVA					Tukey-Kramer			
		df	ss	F	p-value	trap color	mean	sd	Separation of means
<i>Agrilus bilineatus</i>	Between groups	2	2.11	1.38	0.283	black	1.17	0.98	-
	Within groups	15	11.5			green	0.67	1.03	-
	Total	17	13.61			purple	0.33	0.52	-
<i>Agrilus cephalicus</i>	Between groups	2	3735	19.29	<0.001*	black	0	0	b
	Within groups	9	8.75			green	3.75	1.71	a
	Total	11	46.25			purple	0	0	b
<i>Agrilus lecontei</i>	Between groups	2	3.56	16	0.004*	black	0	0	b
	Within groups	6	0.67			green	1.33	0.58	a
	Total	8	4.22			purple	0	0	b
<i>Agrilus obsolettoguttatus</i>	Between groups	2	20.17	7.12	0.014*	black	0	0	b
	Within groups	9	12.75			green	2.75	2.06	a
	Total	11	32.92			purple	0	0	b
<i>Dicerca lurida</i>	Between groups	2	13.5	4.26	0.007*	black	0	0	b
	Within groups	9	6.75			green	0	0	b
	Total	11	20.25			purple	2.25	1.5	a
<i>Dicerca obscura</i>	Between groups	2	2.17	13	0.002*	black	1	0	a
	Within groups	9	0.75			green	0	0	b
	Total	11	2.92			purple	0.25	0.5	b
<i>Ptosima gibbicollis</i>	Between groups	2	2.89	6.5	0.031*	black	0	0	b
	Within groups	6	1.33			green	1.33	0.58	a
	Total	8	4.22			purple	0.33	0.58	a,b

Table 1. Results of ANOVA tests comparing the effect of color on the number of specimens of different species of Buprestidae collected in Lindgren funnel traps. $P < 0.05$ is considered significant. Significant values are indicated by as asterisk (*).

Species accumulation estimator curves for six of the thirteen trap types (Berlese-Tullgren, upper and lower canopy traps, purple, red, and white pan, and pitfall traps) became asymptotic and coalesced with the actual number of species collected (Figs. 8, A1a–m). However, those trap types collected the fewest buprestids. Malaise and green Lindgren funnel traps are estimated to collect the most species after 1000 samples, with green Lindgren funnels collecting the most species for the first 150 samples and Malaise traps collecting more species thereafter (Fig. 9).

Green and purple Lindgren funnel and Malaise traps exhibit, with a single exception, medium similarity with each other and medium to very high similarity with canopy traps (Fig. 10). All four trap types exhibit medium to very high similarity with black Lindgren funnel and blue pan traps and generally exhibit low similarity with yellow, purple, and red pan and lower canopy traps, though all pan traps, excepting blue, collected relatively few species.

Buprestidae exhibited distinct seasonal trends, which is reflected in the number of species and specimens collected per trap type (Figs. 5b, 6b). Eleven of twelve species that were only sampled during one trapping period and five of six species that exhibited population increases did so during the same time period; additionally, only seven species were collected after 17 July, all of which were collected before that date. When comparing trap collection dates using similarity indices, Malaise traps (Fig. 11a) typically exhibit high to very high similarity between trap dates within 6 weeks of each other. Conversely, green Lindgren funnel traps, with a few exceptions, exhibited low to medium similarity regardless of the trapping periods compared (Fig. 11b). Overall, collections made within four to six weeks of each other typically have high to very high similarity, while collections made beyond six weeks apart show low to medium similarity (Fig. 11c) and most species and specimens were collected from late spring through early summer (early June–mid July) (Fig. 12).



Figures 8, 9. Species rarefaction curves. **Fig. 8.** Chao 1 rarefaction curves based on the data. **Fig. 9.** Estimated rarefaction curves ($S(\text{est})$) extrapolated to 1000 samples.

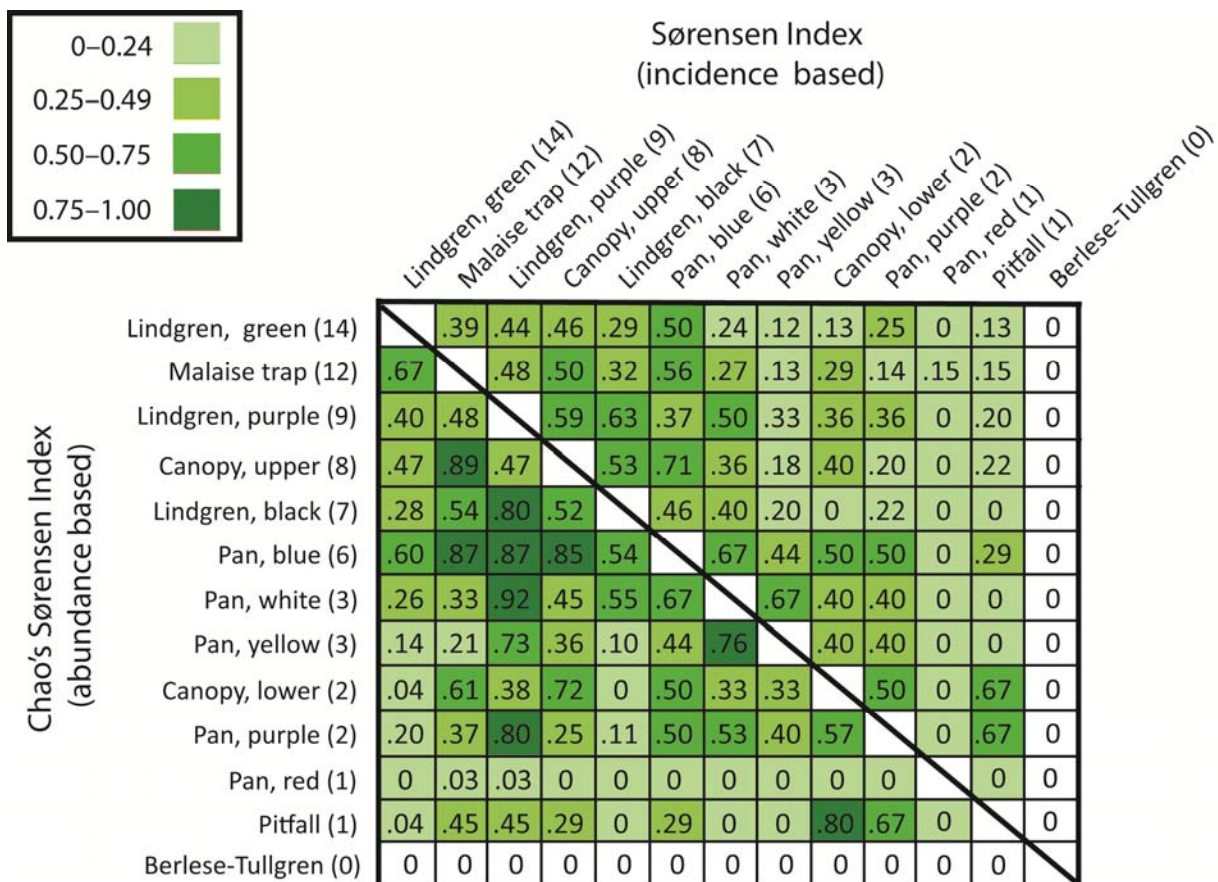
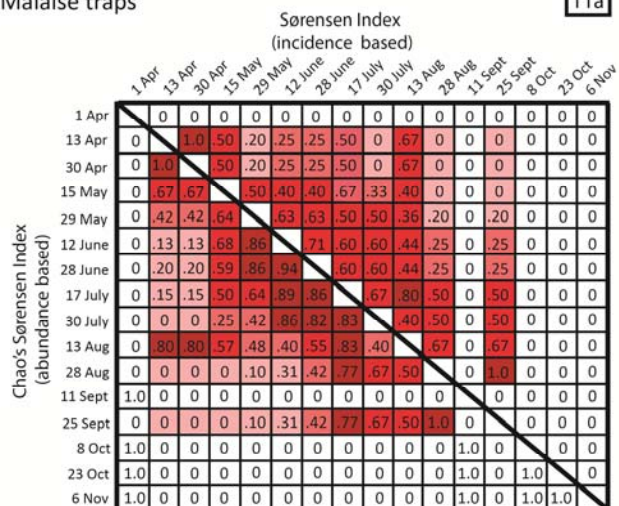


Figure 10. Similarity of trap catch as determined by Sørensen and Chao's Sørensen Indices.
Number of species collected per trap type is indicated parenthetically after each trap type.

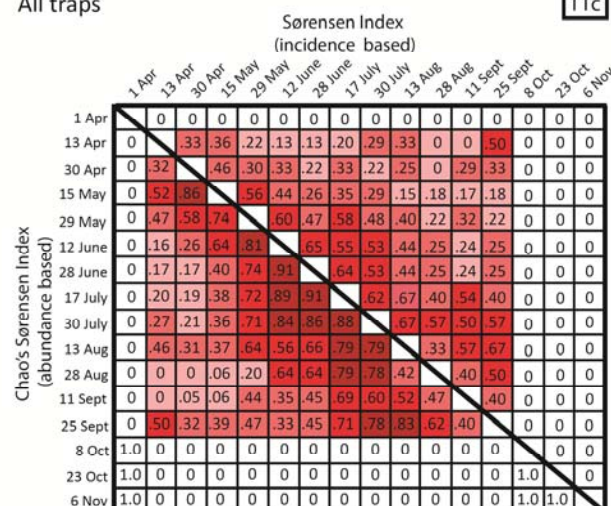
Malaise traps

11a



All traps

11c



Lindgren funnel, green

11b

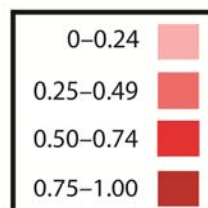
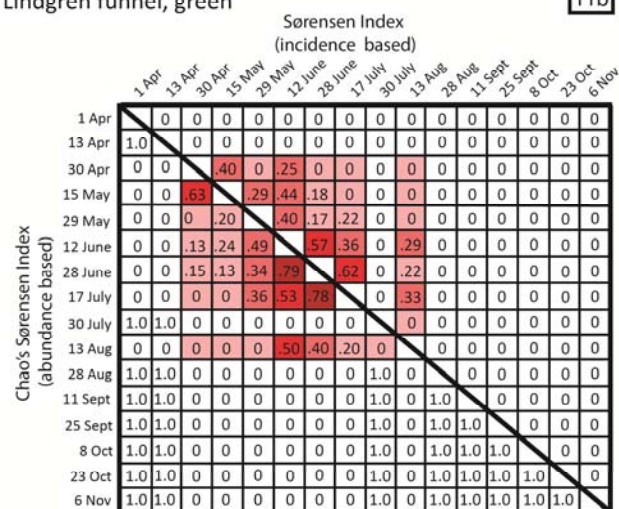


Figure 11. Sørensen and Chao's Sørensen Indices comparing similarity of trap catch by date in Malaise and green Lindgren funnel traps and all trap catch combined.

Fig. 12

Buprestidae

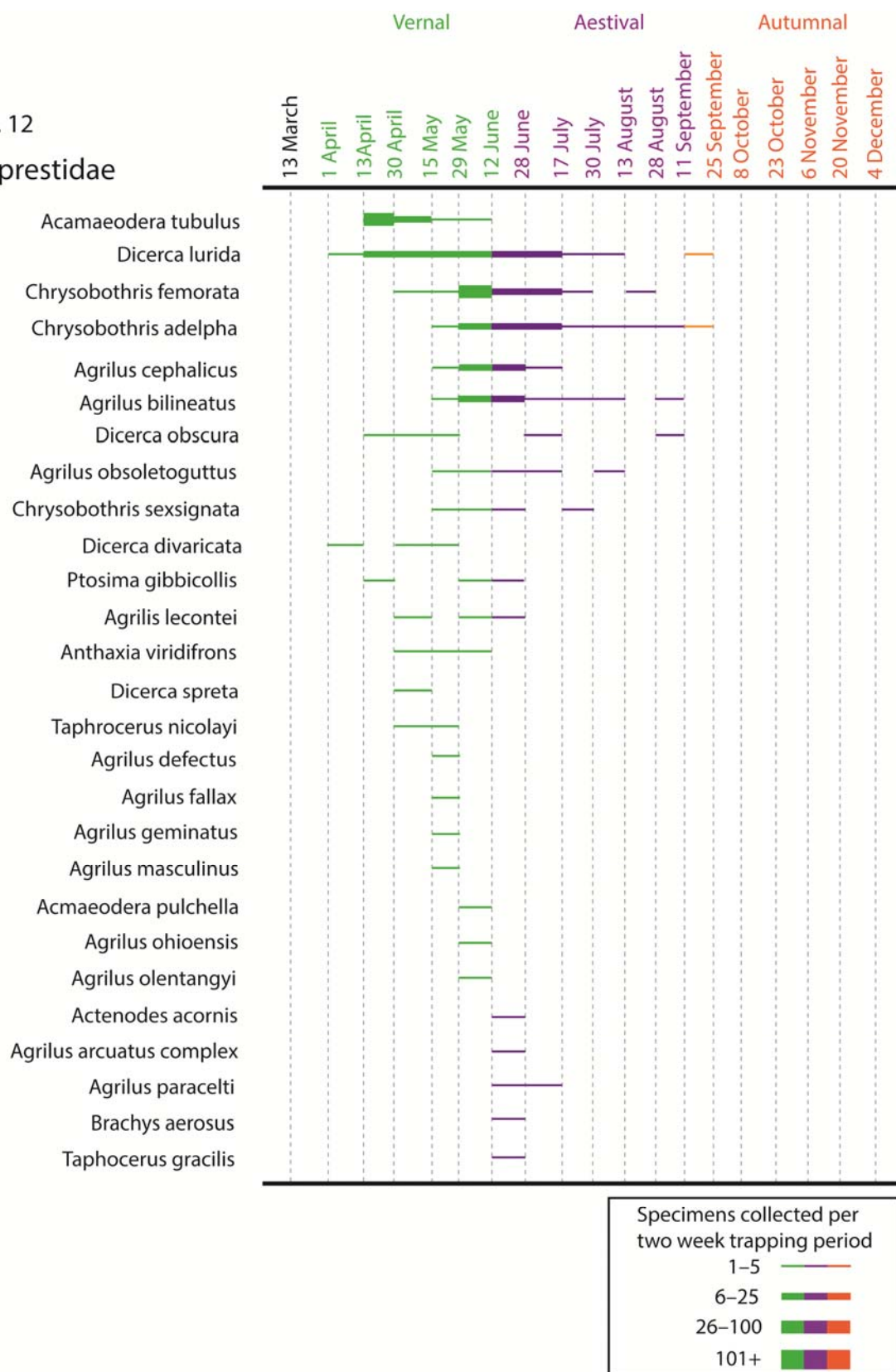
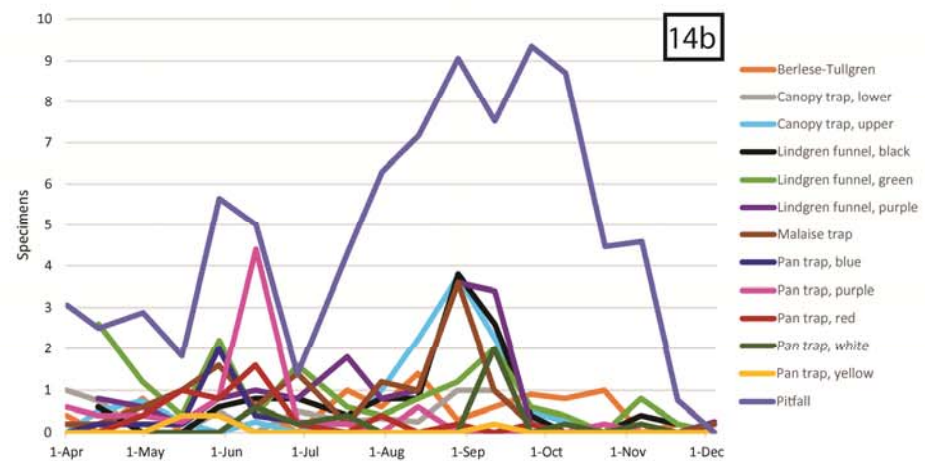
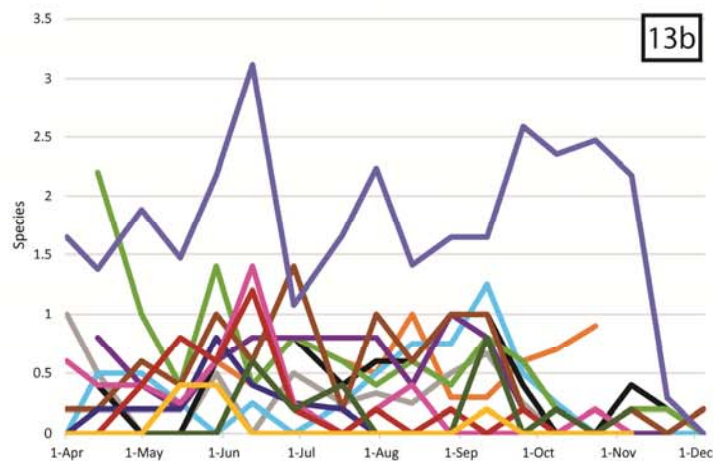
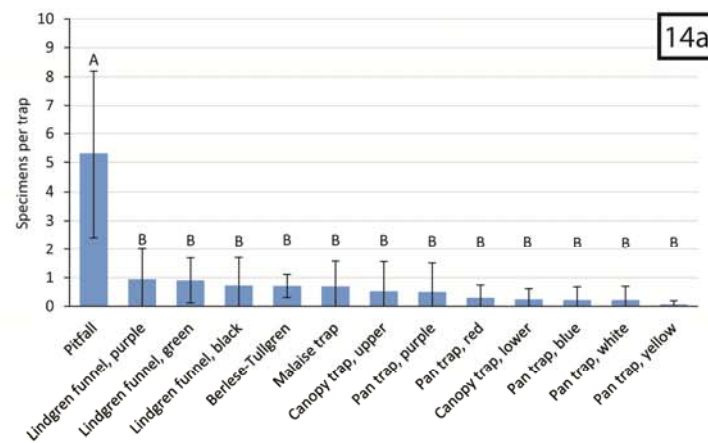
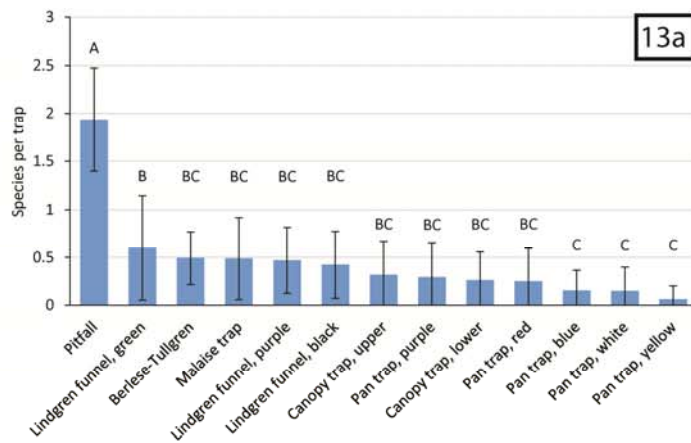


Figure 12. Phenology of buprestids collected during this study summed across all trap types.

Carabidae

Collection efforts resulted in 1964 specimens representing 62 species. Pitfall traps caught the most species (Figs. 13a,b) and specimens (Figs. 14a,b). Most species were represented by fewer than 20 specimens, with 17 species (27%) being represented by singletons (Fig. 15).

There was a significant ($p < 0.05$) effect of trap type on the number of species collected for the thirteen trap types ($F(12,203) = 23.55$, $p = 2.60 \times 10^{-32}$). The mean number of species collected by pitfall traps ($M = 1.84$, $SD = 0.66$) was significantly different than all other trap types; green Lindgren funnel ($M = 0.60$, $SD = 0.55$) was significantly different from blue, white, and yellow pan traps but not other trap types ($p < 0.05$, Tukey-Kramer); the remaining trap types were not significantly different from each other: Berlese-Tullgren ($M = 0.49$, $SD = 0.28$), lower canopy trap ($M = 0.26$, $SD = 0.30$), upper canopy trap ($M = 0.32$, $SD = 0.35$), black Lindgren funnel ($M = 0.42$, $SD = 0.35$), purple Lindgren funnel ($M = 0.47$, $SD = 0.35$), Malaise trap ($M = 0.49$, $SD = 0.43$), blue pan trap ($M = 0.15$, $SD = 0.21$), purple pan trap ($M = 0.29$, $SD = 0.36$), red pan trap ($M = 0.25$, $SD = 0.35$), white pan trap ($M = 0.15$, $SD = 0.25$), and yellow pan trap ($M = 0.06$, $SD = 0.14$) ($p > 0.05$) (Fig. 13a).



Figures 13,14. Average number of carabid species and specimens collected per trap. **Fig 13a.** Average number of species/trap. **Fig. 13b.** Average number of species/trap/date. **Fig. 14a.** Average number of specimens/trap. **Fig. 14b.** Average number of specimens/trap/date. **Figs. 13a,14a.** Bars indicate one standard deviation, letters indicate mean separation as determined by Tukey-Kramer test. **Figs. 13b, 14b.** Trap type indicated by the same color.

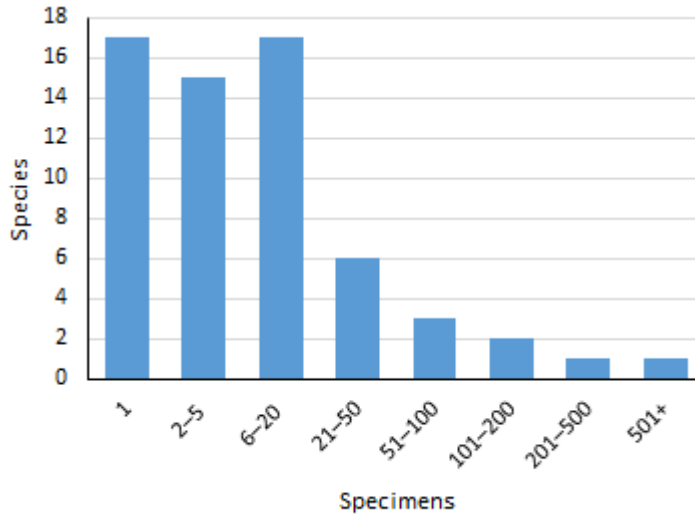


Figure 15. Total number of carabid specimens/species collected across all traps.

There was a significant effect ($p < 0.05$) of trap type on the number of specimens collected for the thirteen trap types ($F(12,203) = 24.03$, $p = 8.05 \times 10^{-33}$). The mean number of specimens collected by pitfall traps ($M = 4.69$, $SD = 2.89$) was significantly different than all other trap types and that all other trap types were not significantly different from each other: Berlese-Tullgren ($M = 0.71$, $SD = 0.40$), lower canopy trap ($M = 0.32$, $SD = 0.38$), upper canopy trap ($M = 0.67$, $SD = 1.04$), black Lindgren funnel ($M = 0.68$, $SD = 0.99$), green Lindgren funnel ($M = 0.85$, $SD = 0.79$), purple Lindgren funnel ($M = 0.86$, $SD = 1.10$), Malaise trap ($M = 0.73$, $SD = 0.88$), blue pan trap ($M = 0.20$, $SD = 0.47$), purple pan trap ($M = 0.45$, $SD = 1.0$), red pan trap ($M = 0.28$, $SD = 0.44$), white pan trap ($M = 0.20$, $SD = 0.48$), and yellow pan trap ($M = 0.06$, $SD = 0.13$) ($p < 0.05$, Tukey-Kramer) (Fig. 14a).

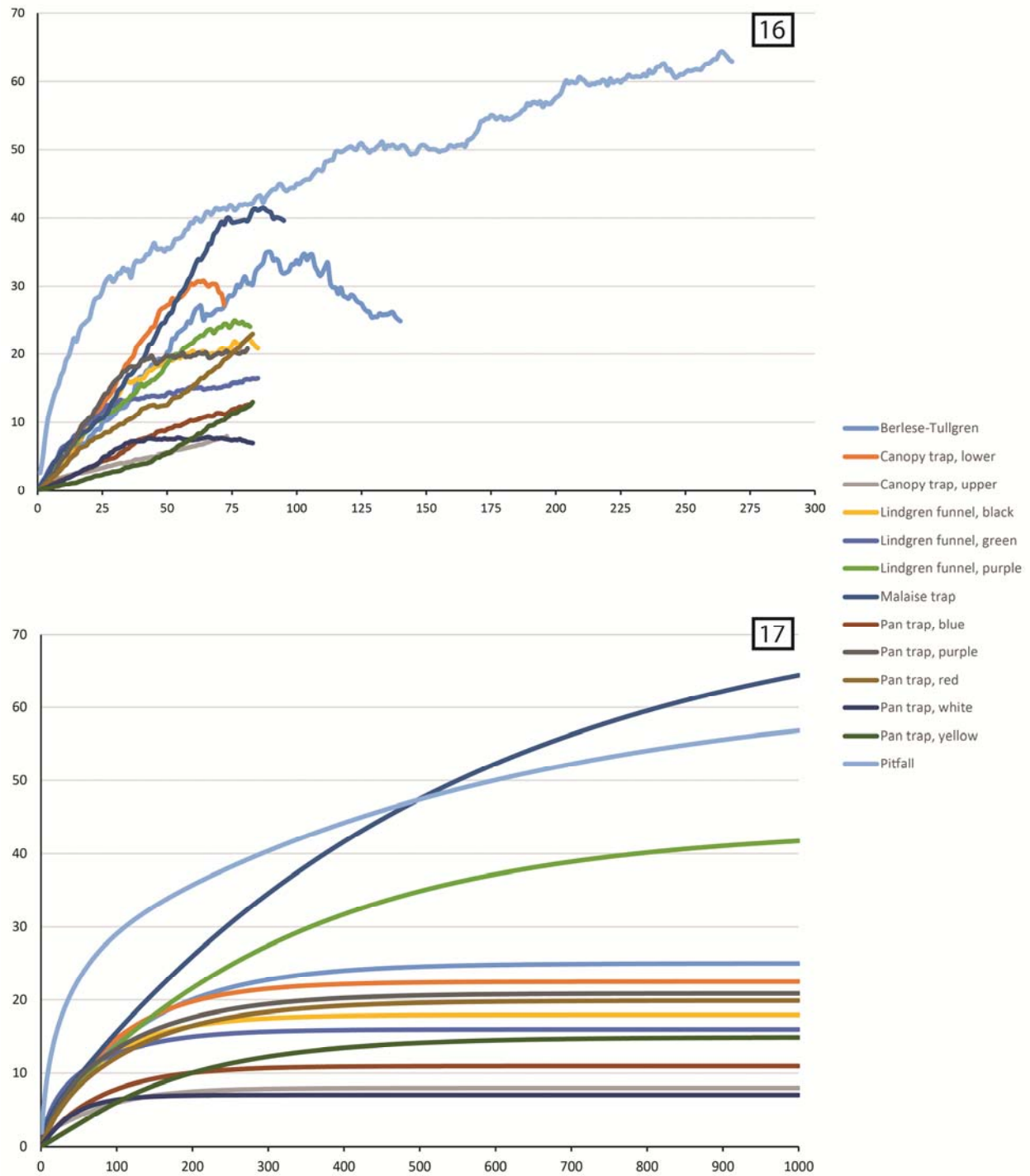
The effects of the color of Lindgren funnel traps was tested for three species. Color did not have a significant effect on the number of specimens collected at the $p < 0.05$ level (Table 2).

Species	ANOVA					Tukey-Kramer			
		df	ss	F	p-value	trap color	mean	sd	Separation of means
<i>Amara musculus</i>	Between groups	2	6.5	1.5	0.274	black	3.50	6.08	-
	Within groups	9	19.5			green	2.42	2.94	-
	Total	11	26			purple	3.83	6.53	-
<i>Cymindis limbata</i>	Between groups	2	13.17	0.22	0.801	black	0.75	1.50	-
	Within groups	33	971.58			green	2.00	2.00	-
	Total	35	984.75			purple	0.25	0.50	-
<i>Lebia viridis</i>	Between groups	2	10.11	1.64	0.228	black	0.50	1.22	-
	Within groups	15	46.33			green	2.33	2.42	-
	Total	17	56.44			purple	1.50	1.38	-

Table 2. Results of ANOVA tests comparing the effect of color on the number of specimens of different species of Carabidae collected in Lindgren funnel traps. $P < 0.05$ is considered significant. Significant values are indicated by as asterisk (*).

Species accumulation estimator curves for nine of the thirteen trap types (Berlese-Tullgren, upper and lower canopy traps, black and green Lindgren funnel traps, blue, purple, red, and white pan traps) became asymptotic (Figs. 16, A2a–m). However, those trap types collected the fewest carabids and in only the white pan traps, which collected the fewest species, did the estimators and actual number of specimens collected coalesce. Pitfall, Malaise, and purple Lindgren funnel traps were estimated to collect the most species after 1000 samples (Fig. 17).

Pitfall traps exhibited medium to very high similarity (Sørensen = 0.47, Chao's Sørensen = 0.82) with Berlese-Tullgren sampling (Fig. 18). Lindgren funnel, Malaise, and canopy traps exhibited medium to very high similarity (Sørensen = 0.26–0.70, Chao's Sørensen = 0.49–0.94) with each other, but, with a single exception, low to medium similarity (Sørensen = 0.13–0.32, Chao's Sørensen = 0.1–0.15) with pitfall traps. Blue, purple, red, and yellow pan traps exhibited high to very high similarity with each other (Sørensen = 0.50–0.73, Chao's Sørensen = 0.60–0.90), but low to medium similarity with white pan traps (Sørensen = 0–0.35, Chao's Sørensen = 0–0.22). Purple and white pan traps generally exhibited medium to very high similarity with non-pan traps (Sørensen = 0.26–0.59, Chao's Sørensen = 0.25–0.86), while yellow pan traps exhibited the lowest similarity with non-pan traps (Sørensen = 0–0.18, Chao's Sørensen = 0–0.19).



Figures 16, 17. Species rarefaction curves. **Fig. 16.** Chao 1 rarefaction curves based on the data.
Fig. 17. Estimated rarefaction curves (S(est)) extrapolated to 1000 samples.

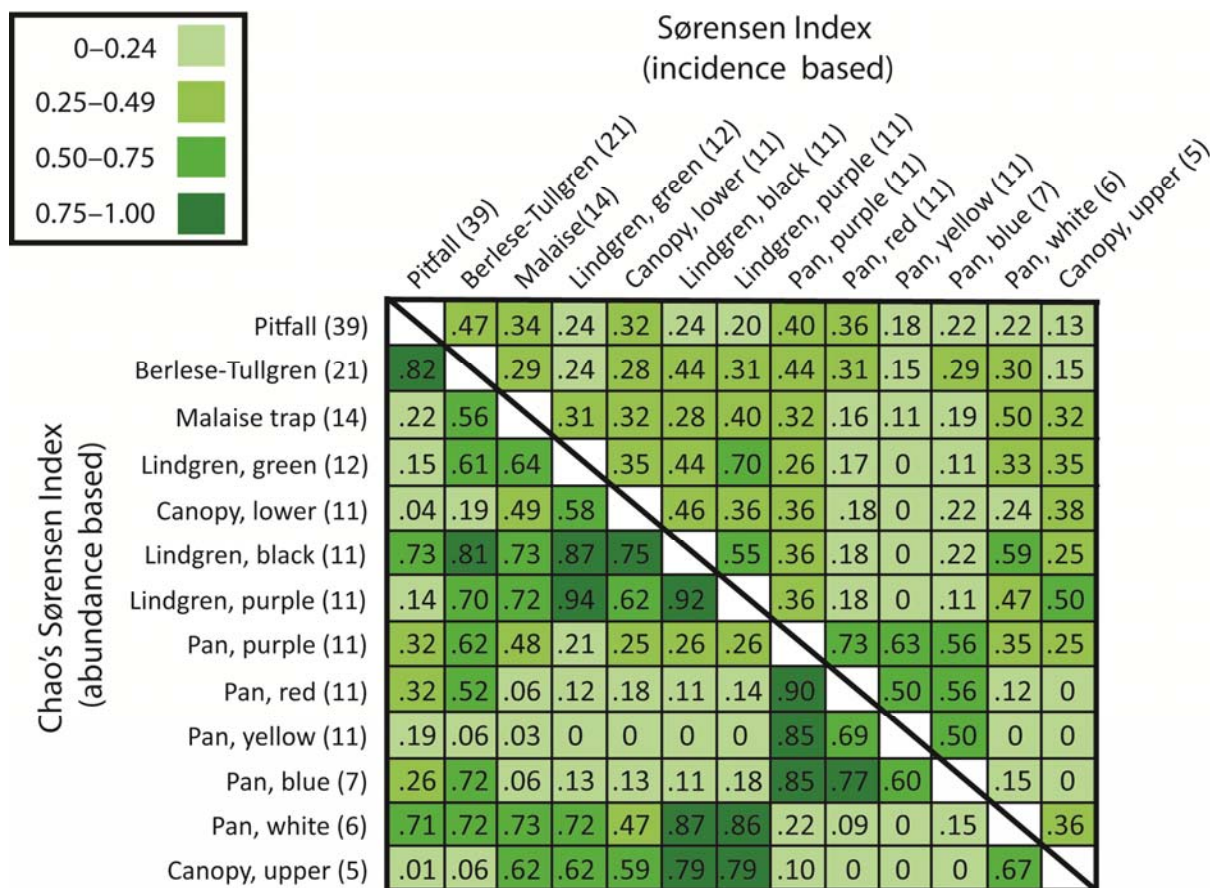
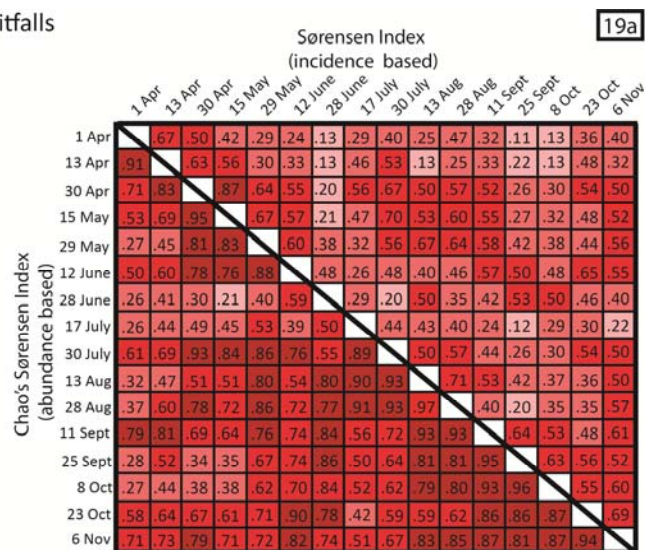


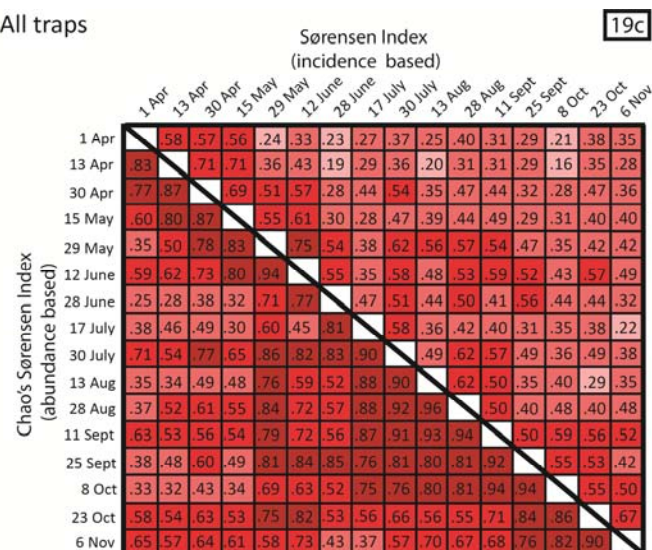
Figure 18. Similarity of trap catch as determined by Sørensen and Chao's Sørensen Indices. Number of species collected per trap type is indicated parenthetically after each trap type.

Carabidae exhibited distinct seasonal trends with the most specimens collected in late spring (late May – early June) and late summer/early fall (mid-August – mid-September) (Fig. 14b), although the number of species collected remained relatively constant throughout the study with a small increase in early summer (June) (Fig. 13b). When comparing trap collection dates using similarity indices, pitfall traps generally exhibited at least medium similarity regardless of the date considered and high to very high similarity between dates within two to four weeks of the date considered (Fig. 19a). Malaise traps exhibited high to very high similarity among spring and fall dates, but no similarity between them (Fig. 19b). When all traps were combined, the similarity between dates was similar to that exhibited by pitfall traps (Fig. 19c).

Pitfalls



All traps



Malaise trap

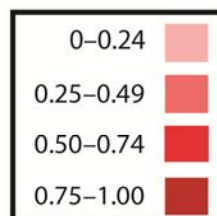
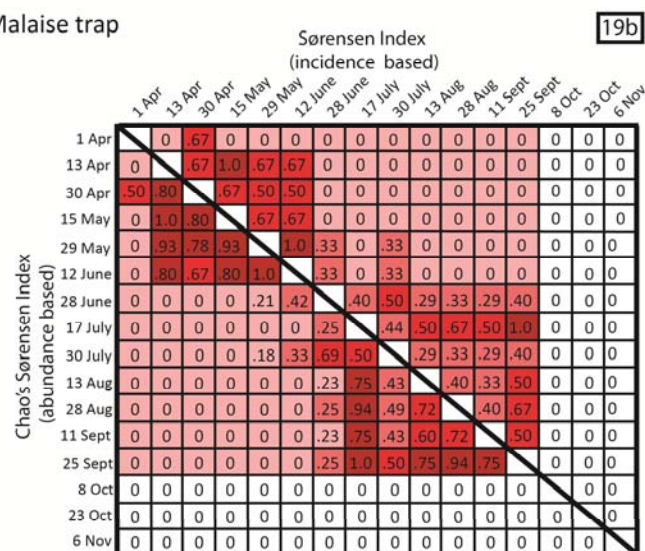


Figure 19. Sørensen and Chao's Sørensen Indices comparing similarity of trap catch by date in Malaise and green Lindgren funnel traps and all trap catch combined.

Approximately a third of the total species were collected in sufficient numbers to examine species-level phenology (Fig. 20a), while approximately a quarter were collected throughout the study but in low numbers that did not allow any interpretation of phenology (Fig. 20b). Forty percent of the species collected were found in low numbers during only a few trapping periods (Fig. 20c).

Fig. 20a

Carabidae

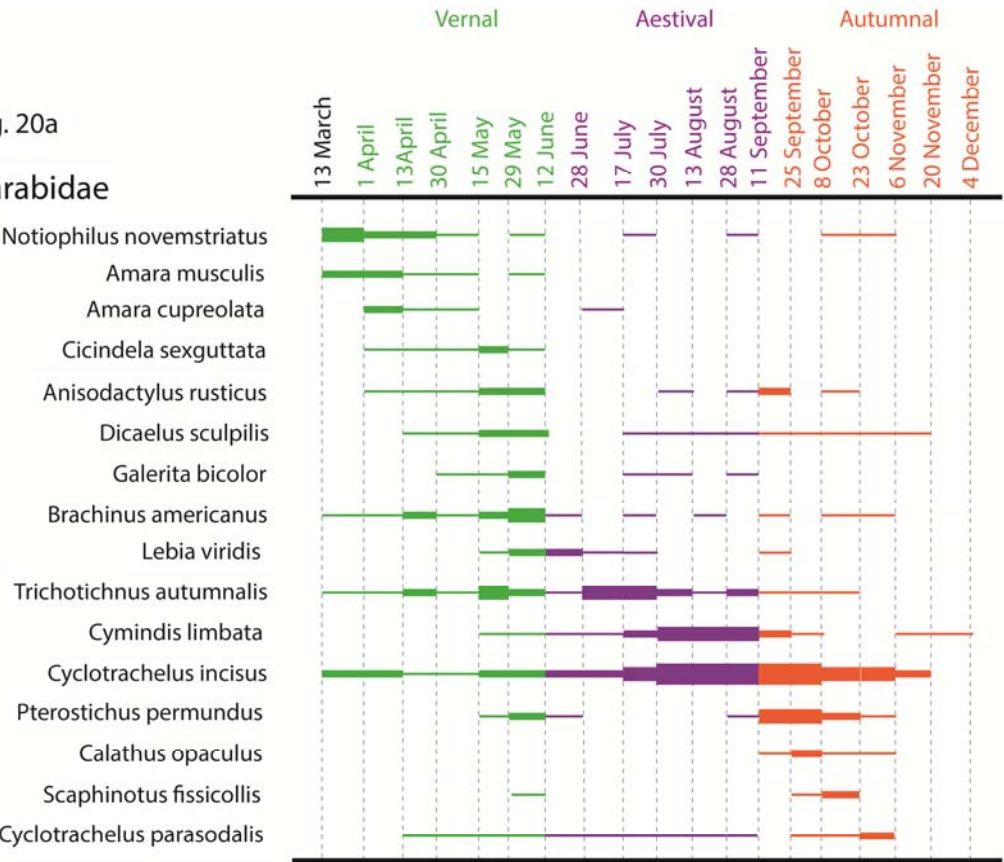


Fig. 20b

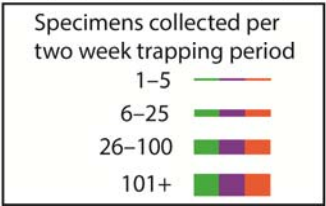
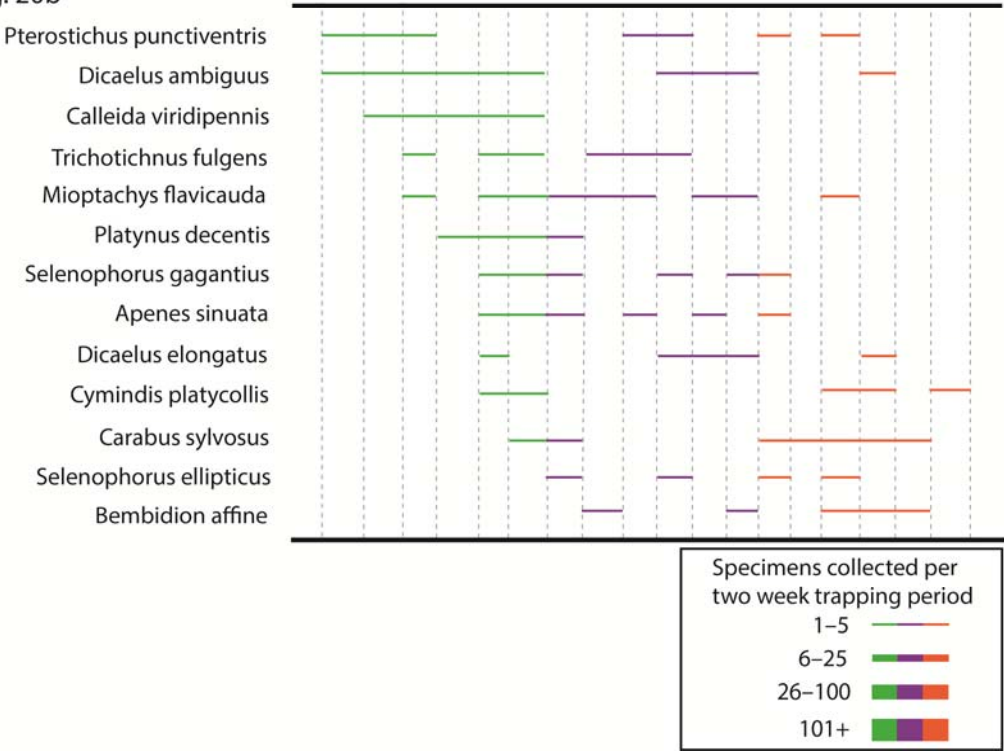


Fig. 20c

Carabidae

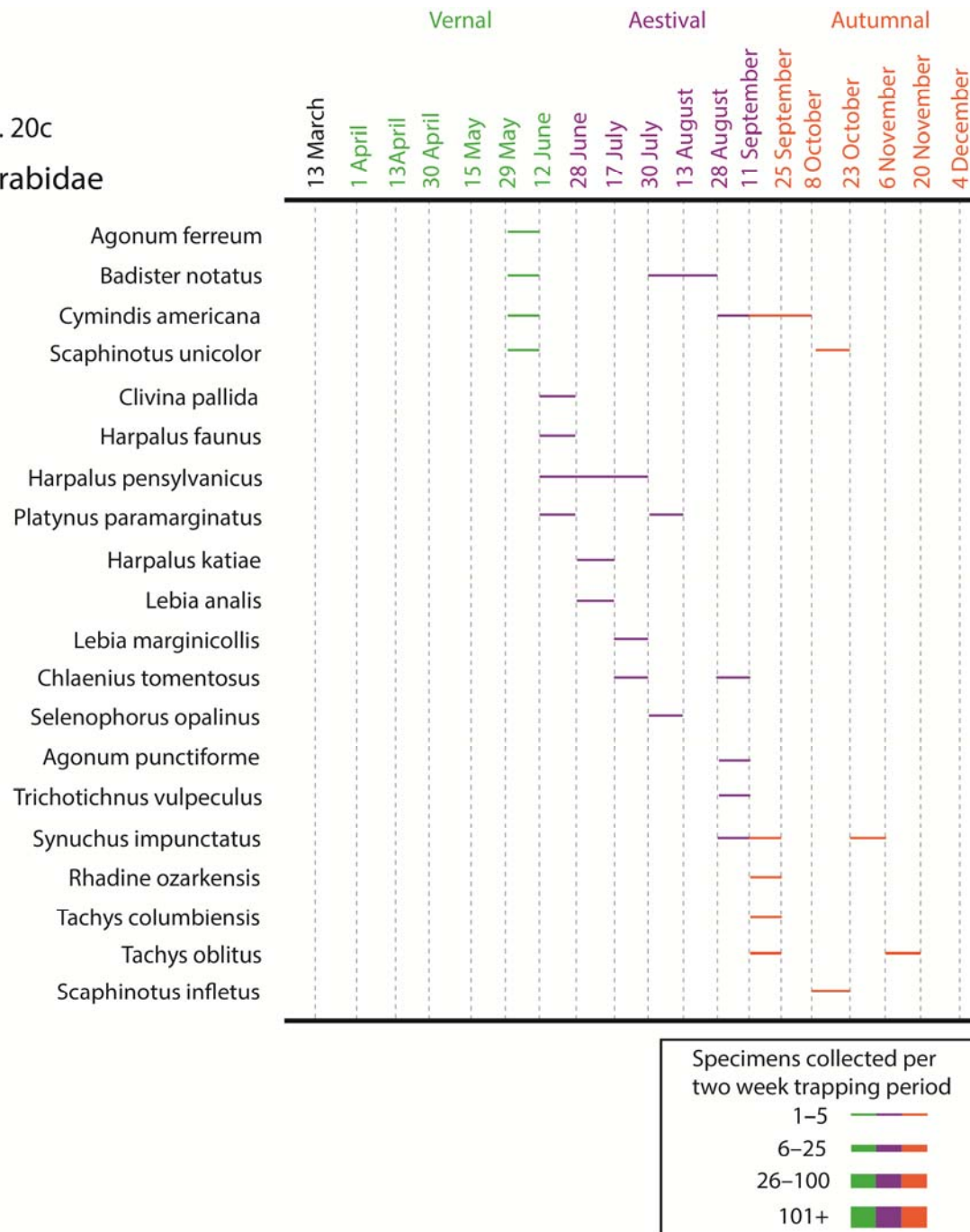


Figure 20. Phenology of carabids collected during this study summed across all trap types. **Fig. 20a.** Species with more than five specimens collected in at least one collecting period. **Fig. 20b.** Species with five or fewer specimens collected in any collection period but found in at least four collection periods. **Fig. 20c.** Species with five or fewer specimens collected in any collection period and found in three or fewer collection periods.

Cerambycidae

Collection efforts resulted in 1885 specimens representing 82 species. Malaise and upper canopy traps caught the most species (Figs. 21a,b) and specimens (Figs. 22a,b). Berlese-Tullgren extraction of leaf litter produced no cerambycids and is not considered in the analyses. Half of all species were represented by six or more specimens, while 16 species (19.5%) were represented by a single specimen (Fig. 23).

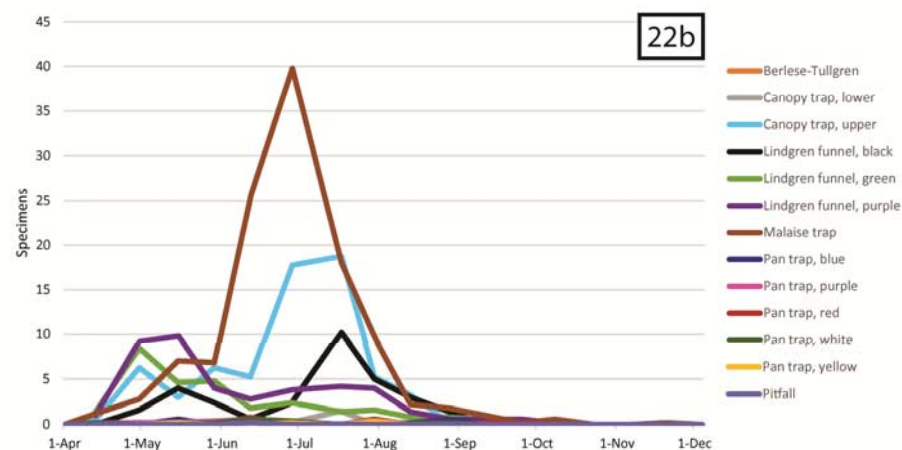
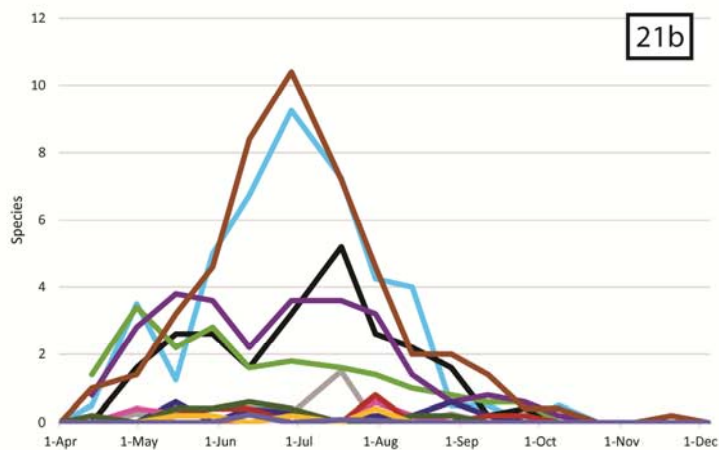
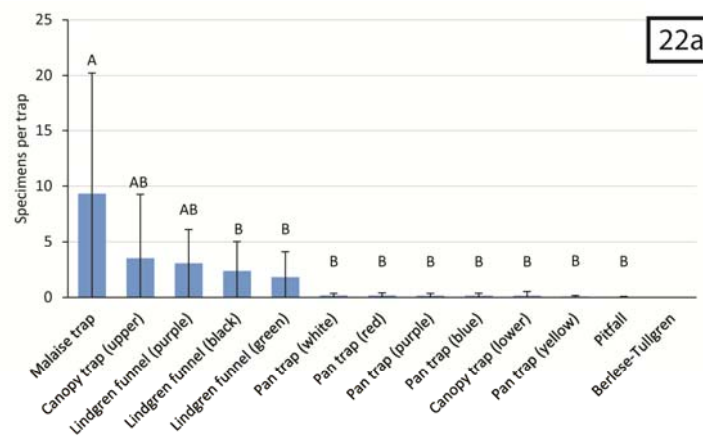
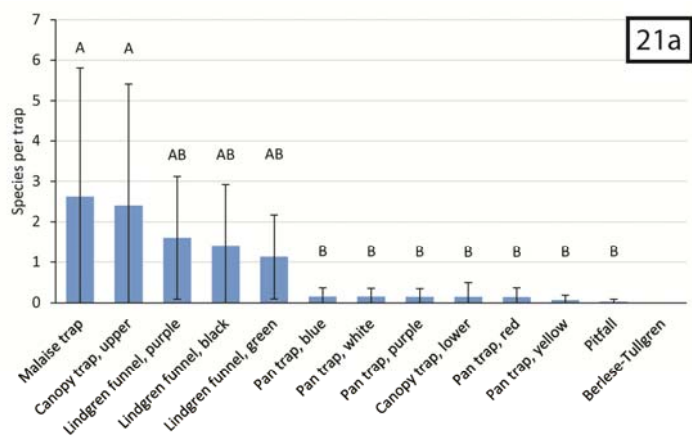
There was a significant ($p < 0.05$) effect of trap type on the number of species collected for the twelve trap types ($F(11,189) = 7.22$, $p = 3.10 \times 10^{-10}$). The mean number of species collected by Malaise traps ($M = 2.62$, $SD = 3.19$) and upper canopy traps ($M = 2.40$, $SD = 3.01$) were not significantly different from black Lindgren funnel traps ($M = 1.40$, $SD = 1.51$), green Lindgren funnel traps ($M = 1.12$, $SD = 1.04$) and purple Lindgren funnel traps ($M = 1.60$, $SD = 1.52$) but were significantly different than all other trap types ($p < 0.05$, Tukey-Kramer). Lindgren funnel traps were not significantly different from pan traps, lower canopy traps, or pitfall traps. Pan traps, lower canopy traps, and pitfall traps were not significantly different from each other: lower canopy trap ($M = 0.14$, $SD = 0.36$), blue pan trap ($M = 0.14$, $SD = 0.23$), purple pan trap ($M = 0.14$, $SD = 0.21$), red pan trap ($M = 0.14$, $SD = 0.20$), white pan trap ($M = 0.15$, $SD = 0.12$), yellow pan trap ($M = 0.06$, $SD = 0.20$), pitfall trap ($M = 0.02$, $SD = 0.06$) (Fig. 21a) ($p > 0.05$).

There was a significant ($p < 0.05$) effect of trap type on the number of specimens for the twelve trap types ($F(11,189) = 4.57$, $p = 3.80 \times 10^{-6}$). The mean number of specimens collected by Malaise traps ($M = 6.49$, $SD = 10.88$) was not significantly different from purple Lindgren funnel traps ($M = 2.48$, $SD = 3.08$) and upper canopy traps ($M = 3.76$, $SD = 5.76$) ($p > 0.05$, Tukey-Kramer) but was significantly different than all other trap types ($p < 0.05$). Purple Lindgren

funnel traps and upper canopy traps were not significantly different from all other trap types and no significant difference was detected between all other trap types: lower canopy trap ($M = 0.17$, $SD = 0.38$), black Lindgren funnel trap ($M = 1.82$, $SD = 2.67$), green Lindgren funnel trap ($M = 1.71$, $SD = 2.27$), blue pan trap ($M = 0.14$, $SD = 0.20$), purple pan trap ($M = 0.11$, $SD = 0.18$), red pan trap ($M = 0.10$, $SD = 0.23$), white pan trap ($M = 0.11$, $SD = 0.17$), yellow pan trap ($M = 0.04$, $SD = 0.11$), pitfall trap ($M = 0.02$, $SD = 0.06$) ($p > 0.05$) (Fig. 22a).

The effects of the color of Lindgren funnel traps was tested for twelve species. Color had a significant ($p < 0.05$) effect on the number of specimens collected for *Xylotrechus colonus* (Fab.) but not other species; the mean number of *X. colonus* specimens collected by black Lindgren funnel traps was significantly higher than green traps but not purple traps and that purple and green traps were not significantly different ($p < 0.05$, Tukey-Kramer) (Table 3).

Species accumulation estimator curves for six of the twelve trap types (lower canopy and blue, purple, red, white and yellow pan traps) became asymptotic (Figs. 24, A3a–m). However, those trap types collected the fewest cerambycids and in only the yellow pan traps, which collected the fewest species, did the estimators and actual number of specimens collected coalesce. Malaise and upper canopy traps were estimated to collect the most species and become asymptotic after approximately 400 samples (Fig. 25).



Figures 21,22. Average number of cerambycid species and specimens collected per trap. **Fig 21a.** Average number of species/trap. **Fig. 21b.** Average number of species/trap/date. **Fig. 22a.** Average number of specimens/trap. **Fig. 22b.** Average number of specimens/trap/date. **Figs. 21a,22a.** Bars indicate one standard deviation, letters indicate mean separation as determined by Tukey-Kramer test. **Figs. 21b, 22b.** Trap type indicated by the same color.

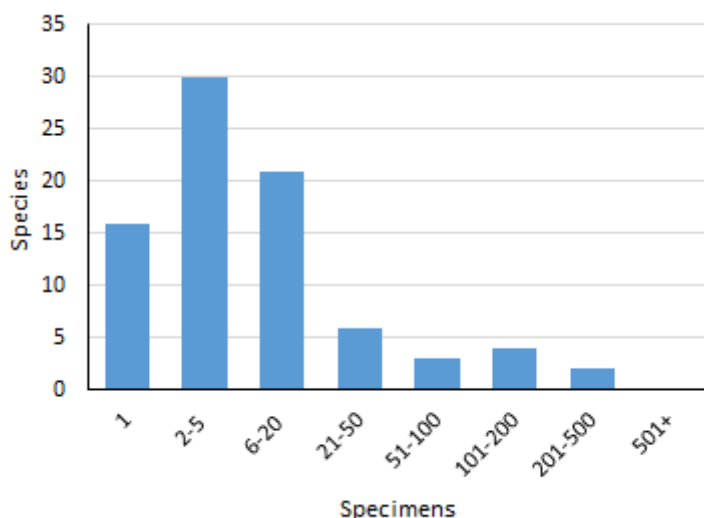


Figure 23. Total number of cerambycid specimens/species collected across all traps.

Malaise, upper canopy, and Lindgren funnel traps had high to very high similarity (Sørensen = 0.57–0.68, Chao's Sørensen = 0.62–0.94) (Fig. 26 – trap comparison). The remaining traps collected significantly fewer species and specimens and will not be considered further.

Cerambycidae exhibited distinct seasonality, with most species and specimens collected during the early summer (Figs. 21b, 22b). Overall, samples collected in the summer and fall were highly similar and distinct from samples collected in the spring when examining individual trap types (Fig. 27a,b) and all traps together (Fig. 27c).

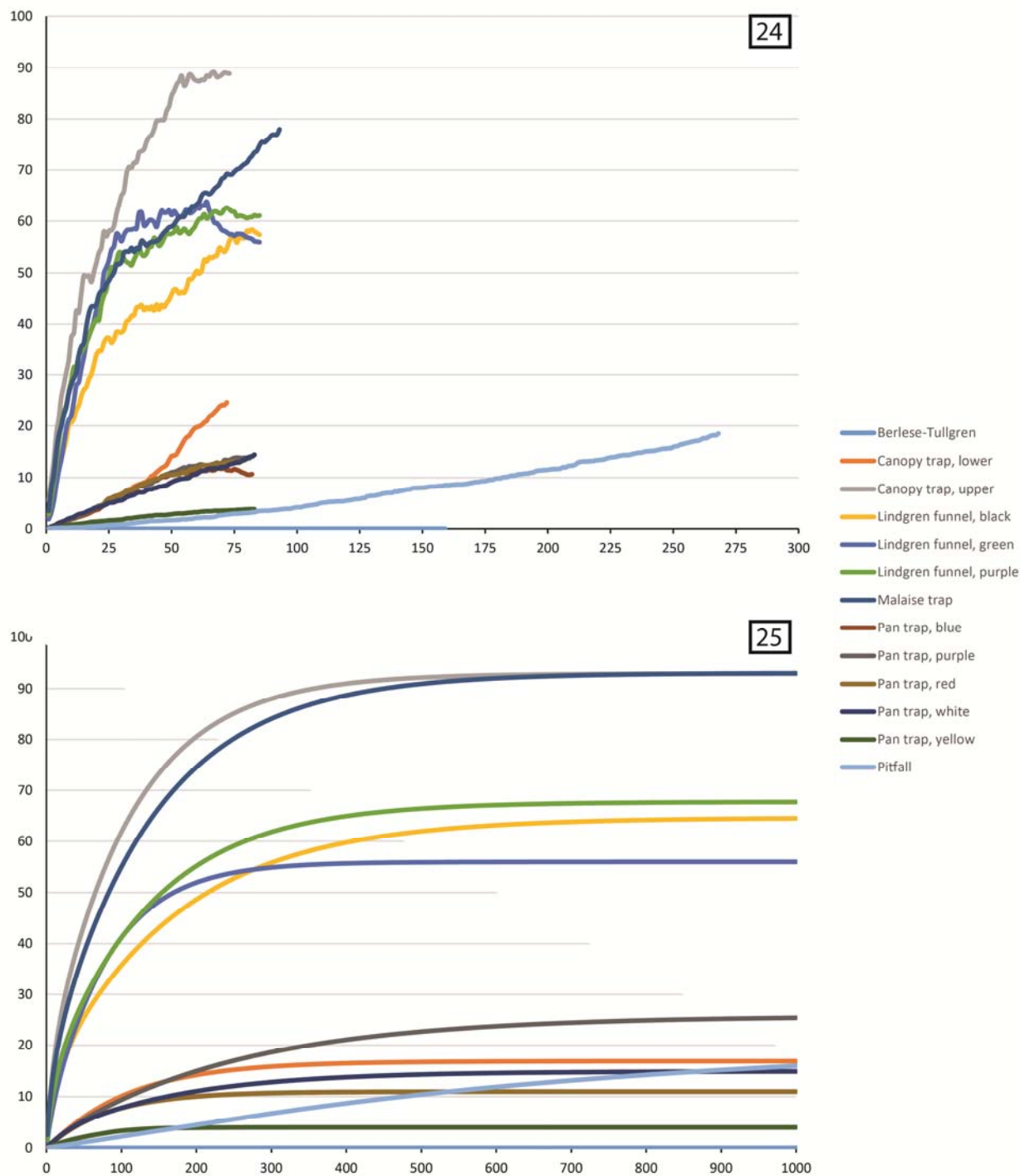
Approximately 24% of the total species were collected in sufficient numbers to examine species-level phenology (Fig. 28a), while 15% were collected throughout the study but in low numbers that do not allow any interpretation of phenology (Fig. 28b) and 62% were found in low numbers during only a few trapping periods (Fig. 28c).

Species	ANOVA					Tukey-Kramer			
		df	ss	F	p-value	trap color	mean	sd	Separation of means
<i>Anelaphus parallelus</i>	Between groups	2	66.89	0.96	0.433	black	4.67	4.73	-
	Within groups	6	208			green	8.33	2.31	-
	Total	8	274.89			purple	11.3	8.74	-
<i>Elaphidion mucronatum</i>	Between groups	2	26.47	0.77	0.472	black	4.20	6.29	-
	Within groups	27	462.5			green	1.90	1.52	-
	Total	29	488.97			purple	3.00	3.09	-
<i>Elytrimitatrix undata</i>	Between groups	2	0.13	0.06	0.94	black	1.00	1.22	-
	Within groups	12	12.8			green	0.80	0.84	-
	Total	14	12.93			purple	1.00	1.00	-
<i>Heterachthes quadrimaculatus</i>	Between groups	2	2.17	1.15	0.36	black	1.25	1.50	-
	Within groups	9	8.5			green	0.25	0.50	-
	Total	11	10.67			purple	0.50	0.58	-
<i>Molorchus bimaculatus</i>	Between groups	2	250.89	5.02	0.052	black	3.67	1.53	-
	Within groups	6	150			green	14.3	8.37	-
	Total	8	400.89			purple	2.67	1.53	-
<i>Neoclytus acuminatus</i>	Between groups	2	1.58	1.6	0.225	black	0.50	0.76	-
	Within groups	21	10.38			green	0.25	0.46	-
	Total	23	11.96			purple	0.88	0.83	-

Table 3. Results of ANOVA tests comparing the effect of color on the number of specimens of different species of Cerambycidae collected in Lindgren funnel traps. $P < 0.05$ is considered significant. Significant values are indicated by as asterisk (*).

Species		ANOVA				Tukey-Kramer			
		df	ss	F	p-value	trap color	mean	sd	Separation of means
<i>Neoclytus mucronatus</i>	Between groups	2	51.71	1.93	0.174	black	3.71	3.40	-
	Within groups	18	240.86			green	0	0	-
	Total	20	292.57			purple	2.71	5.35	-
<i>Neoclytus scutellaris</i>	Between groups	2	0.13	2.72	0.106	black	1.40	2.61	-
	Within groups	12	41.6			green	1.40	1.67	-
	Total	14	41.73			purple	1.60	0.89	-
<i>Parelaphidion aspersum</i>	Between groups	2	3.56	2.72	0.106	black	1.67	2.08	-
	Within groups	6	10.00			green	0.33	0.58	-
	Total	8	13.56			purple	0.33	0.58	-
<i>Saperda imitans</i>	Between groups	2	8.22	3.7	0.09	black	1.00	1.00	-
	Within groups	6	6.67			green	0	0	-
	Total	8	14.89			purple	2.33	1.53	-
<i>Stenosphenus notatus</i>	Between groups	2	370	2.72	0.106	black	0.40	0.55	-
	Within groups	12	817.6			green	1.40	1.34	-
	Total	14	1187.6			purple	11.4	14.2	-
<i>Xylotrechus colonus</i>	Between groups	2	78.79	4.83	0.015*	black	3.72	2.8	a,b
	Within groups	30	244.73			green	0.09	0.3	b
	Total	32	323.52			purple	2.82	4.07	a,b

Table 3 (cont.). Results of ANOVA tests comparing the effect of color on the number of specimens of different species of Cerambycidae collected in Lindgren funnel traps. $P < 0.05$ is considered significant. Significant values are indicated by as asterisk (*).



Figures 16, 17. Species rarefaction curves. **Fig. 16.** Chao 1 rarefaction curves based on the data. **Fig. 17.** Estimated rarefaction curves (S(est)) extrapolated to 1000 samples.

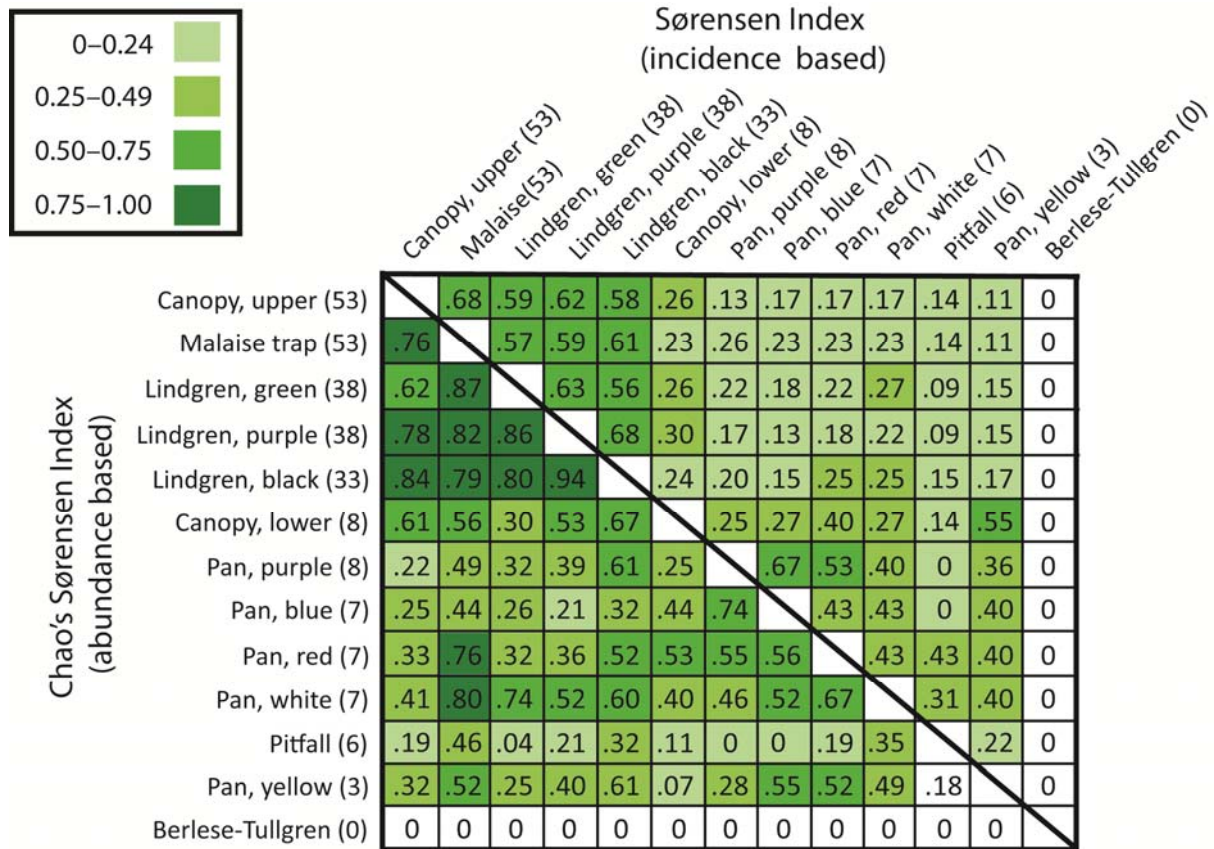


Figure 26. Similarity of trap catch as determined by Sørensen and Chao's Sørensen Indices.
Number of species collected per trap type is indicated parenthetically after each trap type.

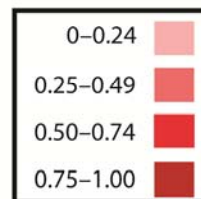
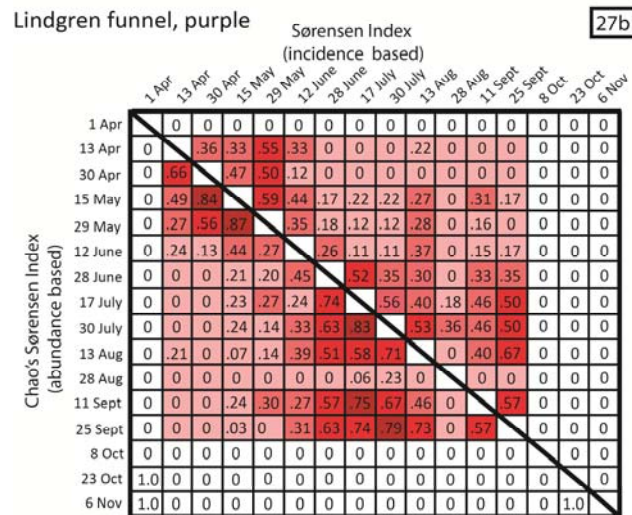
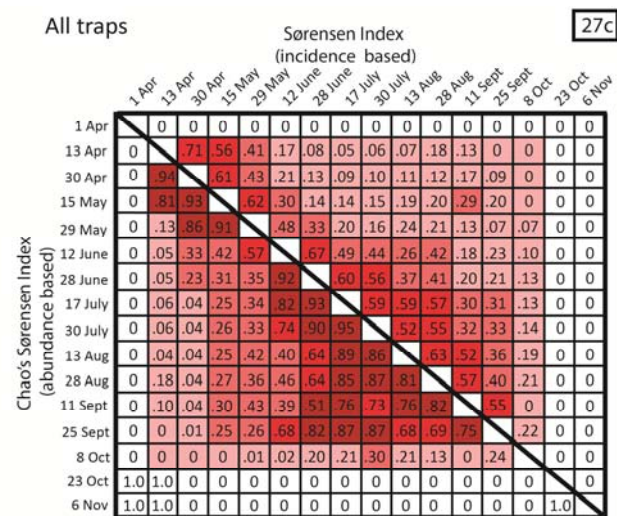
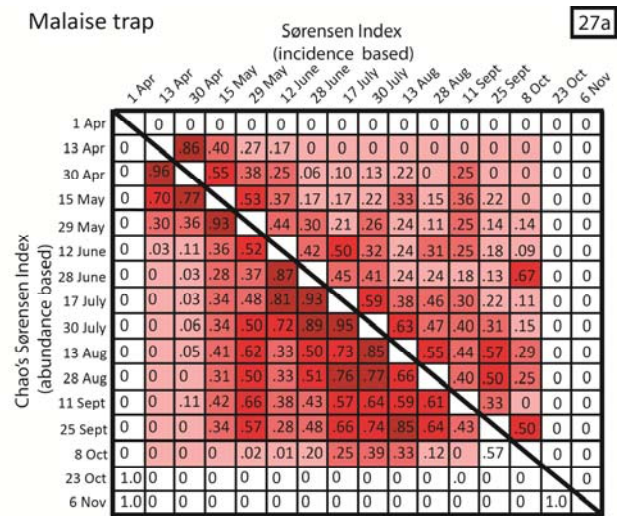


Figure 27. Sørensen and Chao's Sørensen Indices comparing similarity of trap catch by date in Malaise and purple Lindgren funnel traps and all trap catch combined.

Fig. 28a

Cerambycidae

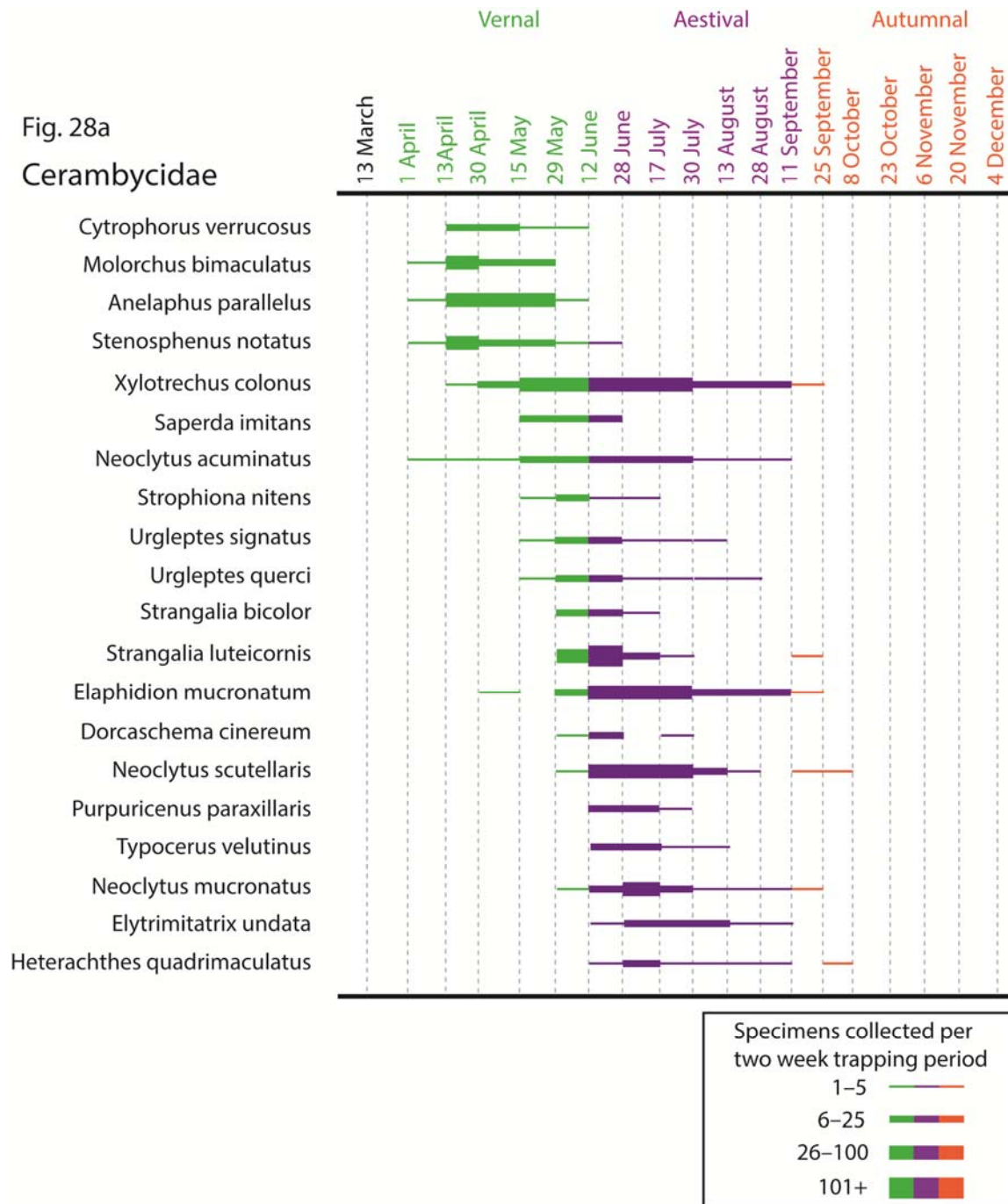


Fig. 28b
Cerambycidae

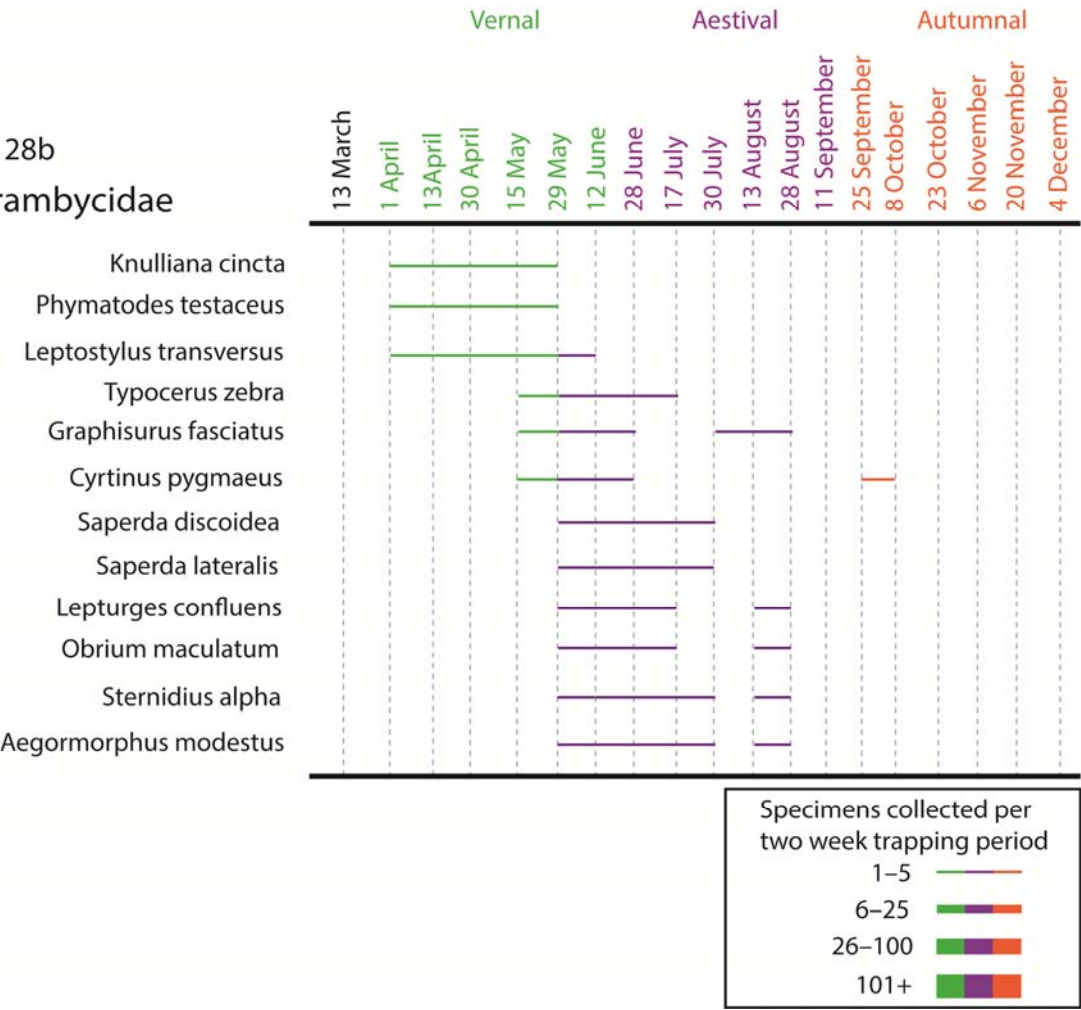


Fig. 28c

Cerambycidae

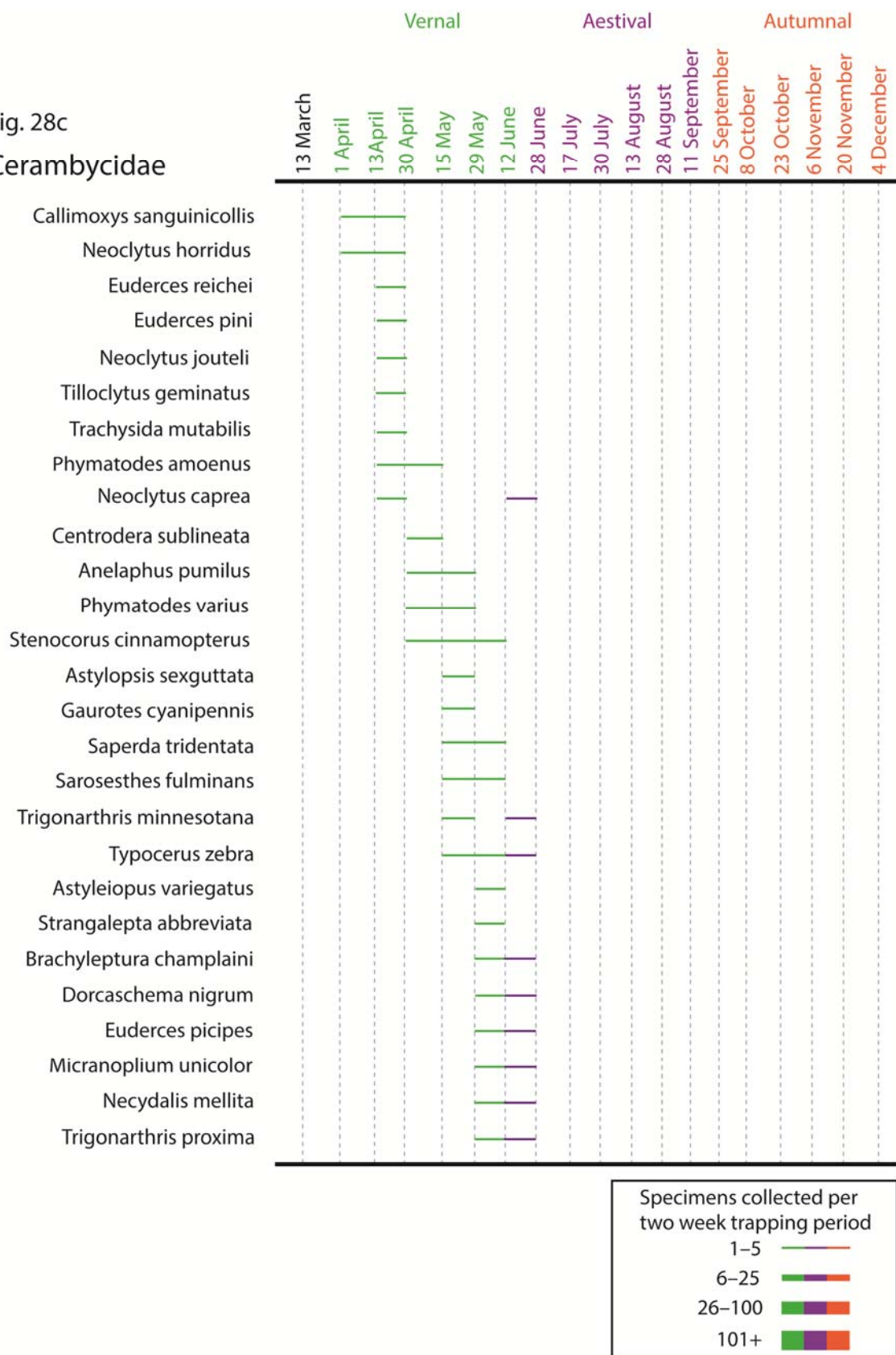


Fig. 28c (cont.)

Cerambycidae

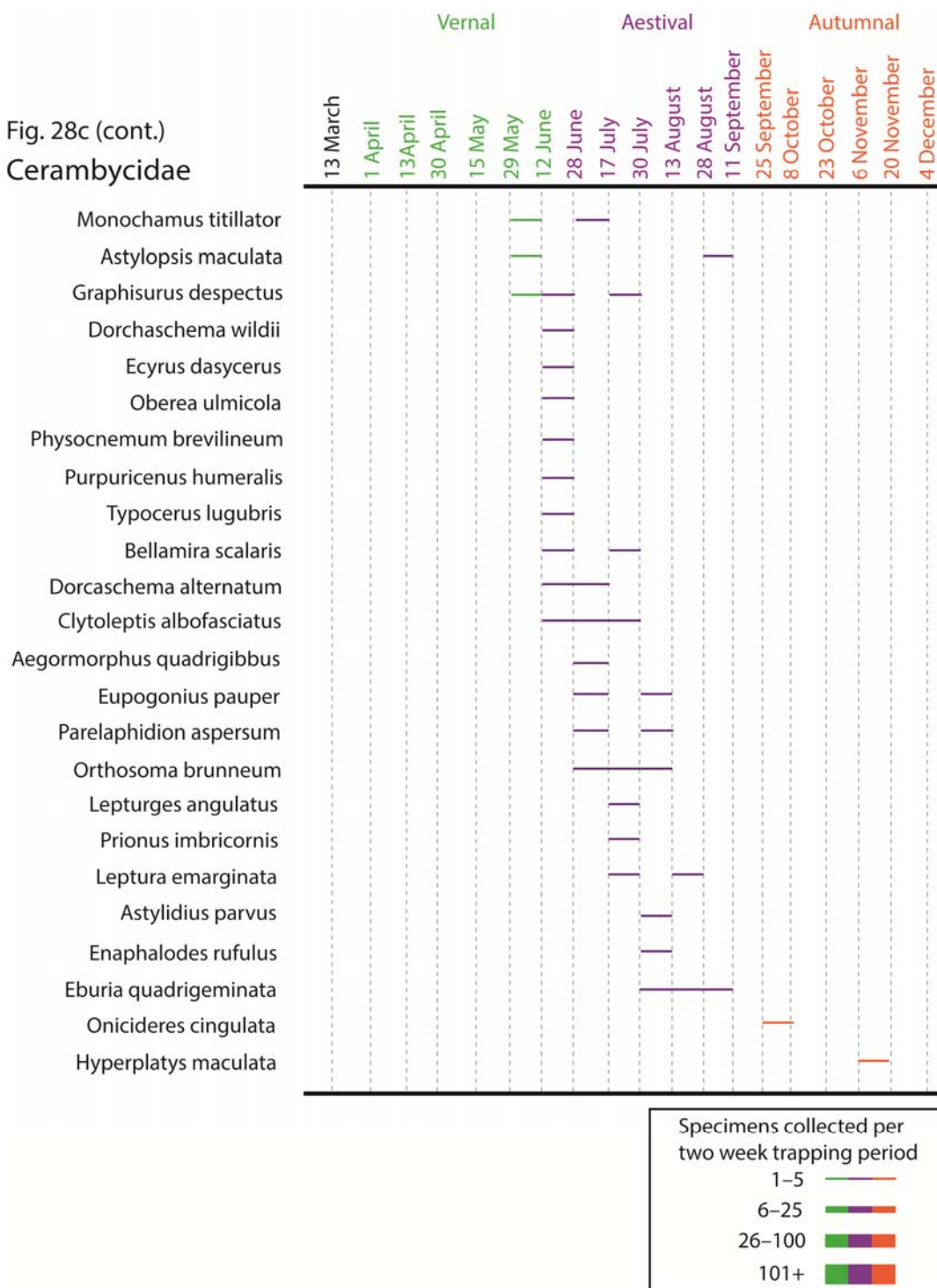


Figure 28. Phenology of cerambycids collected during this study summed across all trap types.

Fig. 28a. Species with more than five specimens collected in at least one collecting period. **Fig. 28b.** Species with five or fewer specimens collected in any collection period but found in at least four collection periods. **Fig. 28c.** Species with five or fewer specimens collected in any collection period and found in three or fewer collection periods.

Curculionoidea

Collecting efforts resulted in 3777 specimens representing 71 species. Malaise and pitfall traps caught the most species (Figs. 29a,b), while Malaise and upper canopy traps caught the most specimens (Fig. 30a,b). Half of the species collected were represented by five or fewer specimens and 28% of the species were represented by singletons (Fig. 31).

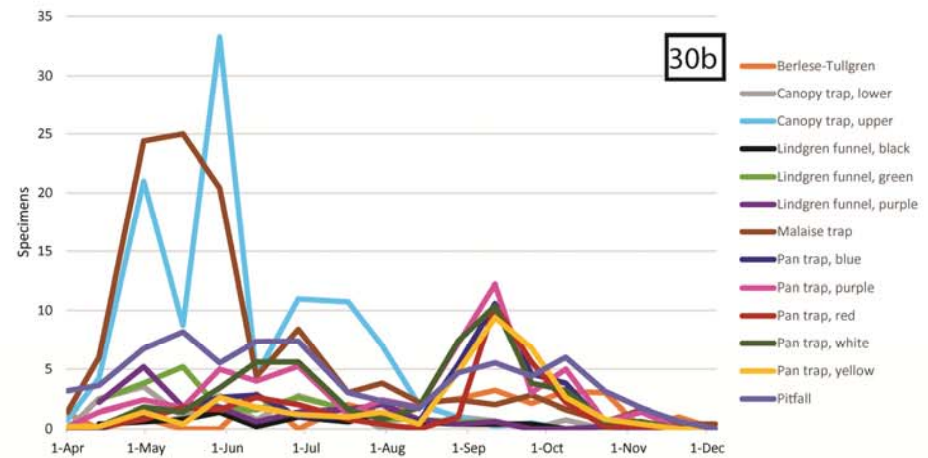
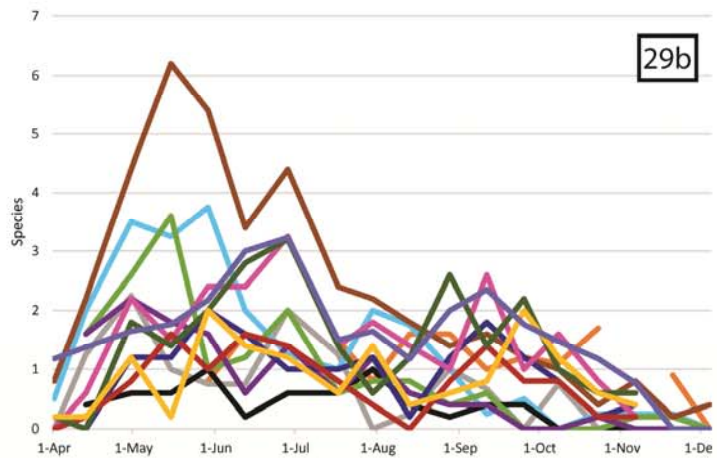
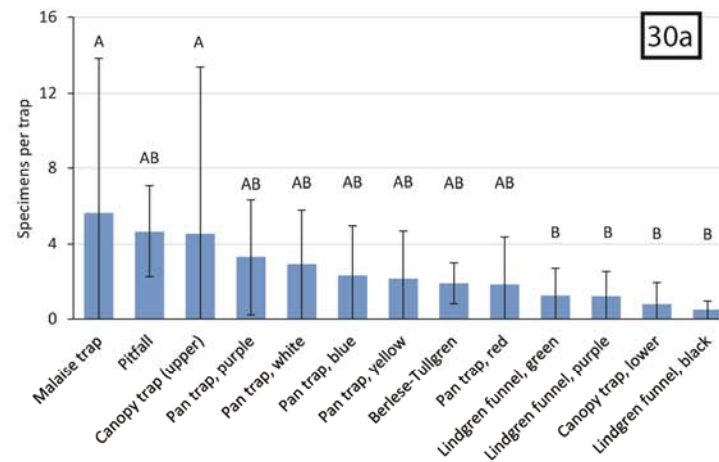
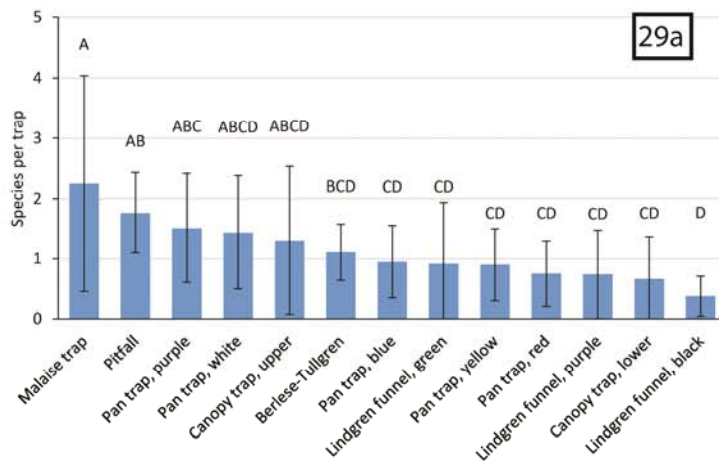
There was a significant ($p < 0.05$) effect of trap type on the number of species collected for the thirteen trap types ($F(12,203) = 5.45$, $p = 5.30 \times 10^{-8}$). The mean number of species collected by Malaise traps ($M = 2.24$, $SD = 1.79$) were not significantly different ($p > 0.05$, Tukey-Kramer) from pitfall ($M = 1.78$, $SD = 0.66$), purple pan ($M = 1.51$, $SD = 0.90$), white pan ($M = 1.43$, $SD = 0.94$), and upper canopy traps ($M = 1.31$, $SD = 1.23$) but were significantly different than all other trap types ($p > 0.05$). Pitfall traps were not significantly different from purple and white pan and upper canopy traps and Berlese-Tullgren sampling ($M = 1.11$, $SD = 0.47$), but were significantly different from blue, yellow, and red pan, lower canopy, and Lindgren funnel traps. Purple pan traps were significantly different from black Lindgren funnel traps ($M = 0.37$, $SD = 0.33$), but not significantly different from all other trap types. The remaining trap types were not significantly different from each other: Blue pan ($M = 0.95$, $SD = 0.60$), yellow pan ($M = 0.90$, $SD = 0.60$), red pan ($M = 0.75$, $SD = 0.54$), green Lindgren funnel ($M = 0.91$, $SD = 1.12$), purple Lindgren funnel ($M = 0.74$, $SD = 0.73$), and black Lindgren funnel ($M = 0.37$, $SD = 0.33$). (Fig. 29a).

There was a significant ($p < 0.05$) effect of trap type on the number of specimens collected for the thirteen trap types ($F(12,203) = 4.57$, $p = 3.80 \times 10^{-6}$). The mean number of specimens collected by Malaise traps ($M = 6.16$, $SD = 8.17$) and upper canopy traps ($M = 5.88$, $SD = 8.85$) were not significantly ($p > 0.05$, Tukey-Kramer) different from pitfall ($M = 4.41$, $SD = 2.25$),

purple Lindgren funnel traps ($M = 1.09$, $SD = 1.32$), white pan ($M = 3.03$, $SD = 2.92$), blue pan ($M = 2.35$, $SD = 2.74$), yellow pan ($M = 2.21$, $SD = 2.62$), red pan ($M = 1.86$, $SD = 2.68$), or Berlese-Tullgren extraction ($M = 1.80$, $SD = 1.09$) but were significantly ($p < 0.05$) different from black Lindgren funnel ($M = 0.48$, $SD = 0.46$), green Lindgren funnel ($M = 1.30$, $SD = 1.48$), purple Lindgren funnel ($M = 1.09$, $SD = 1.34$), and lower canopy ($M = 0.96$, $SD = 1.15$) traps. The number of specimens collected in pitfall, pan, Lindgren funnel and lower canopy traps were not significantly different from each other ($p > 0.05$) (Fig. 30a).

The effects of the color of Lindgren funnel traps was tested for fourteen species. Color had a significant ($p < 0.05$) effect on the number of specimens collected at the $p < 0.05$ level for ten species; the mean number of specimens was significantly ($p < 0.05$, Tukey-Kramer) higher in green Lindgren funnel traps for two species, higher in purple traps for four species, could not be separated for two species, higher in green compared to black but not purple for one species, and higher in black compared to purple but not green for one species. (Table 4).

Species accumulation estimator curves for three of the thirteen trap types (black and purple Lindgren funnel and yellow pan traps) became asymptotic (Figs. 32, A4a–m), and in two trap types (black and purple Lindgren funnel traps) the estimators and actual number of specimens collected coalesced. However, those trap types collected the fewest curculionoids. Green Lindgren funnel traps were estimated to not become asymptotic and collect the most species after 1000 samples; however, Malaise traps were estimated to collect more species than green Lindgren funnel traps for the first 250 samples (Fig. 33).



Figures 29,30. Average number of cerambycid species and specimens collected per trap. **Fig 29a.** Average number of species/trap. **Fig. 29b.** Average number of species/trap/date. **Fig. 30a.** Average number of specimens/trap. **Fig. 30b.** Average number of specimens/trap/date. **Figs. 29a,30a.** Bars indicate one standard deviation, letters indicate mean separation as determined by Tukey-Kramer test. **Figs. 29b, 30b.** Trap type indicated by the same color.

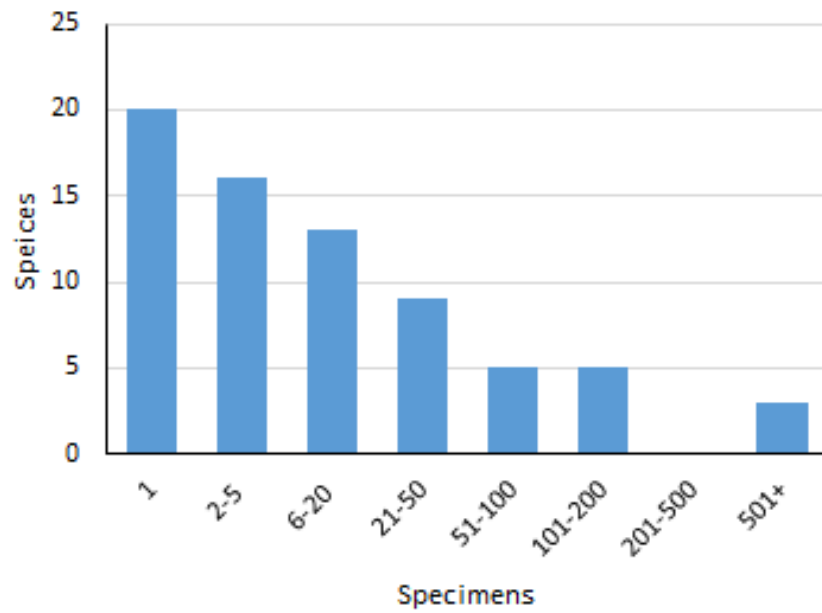


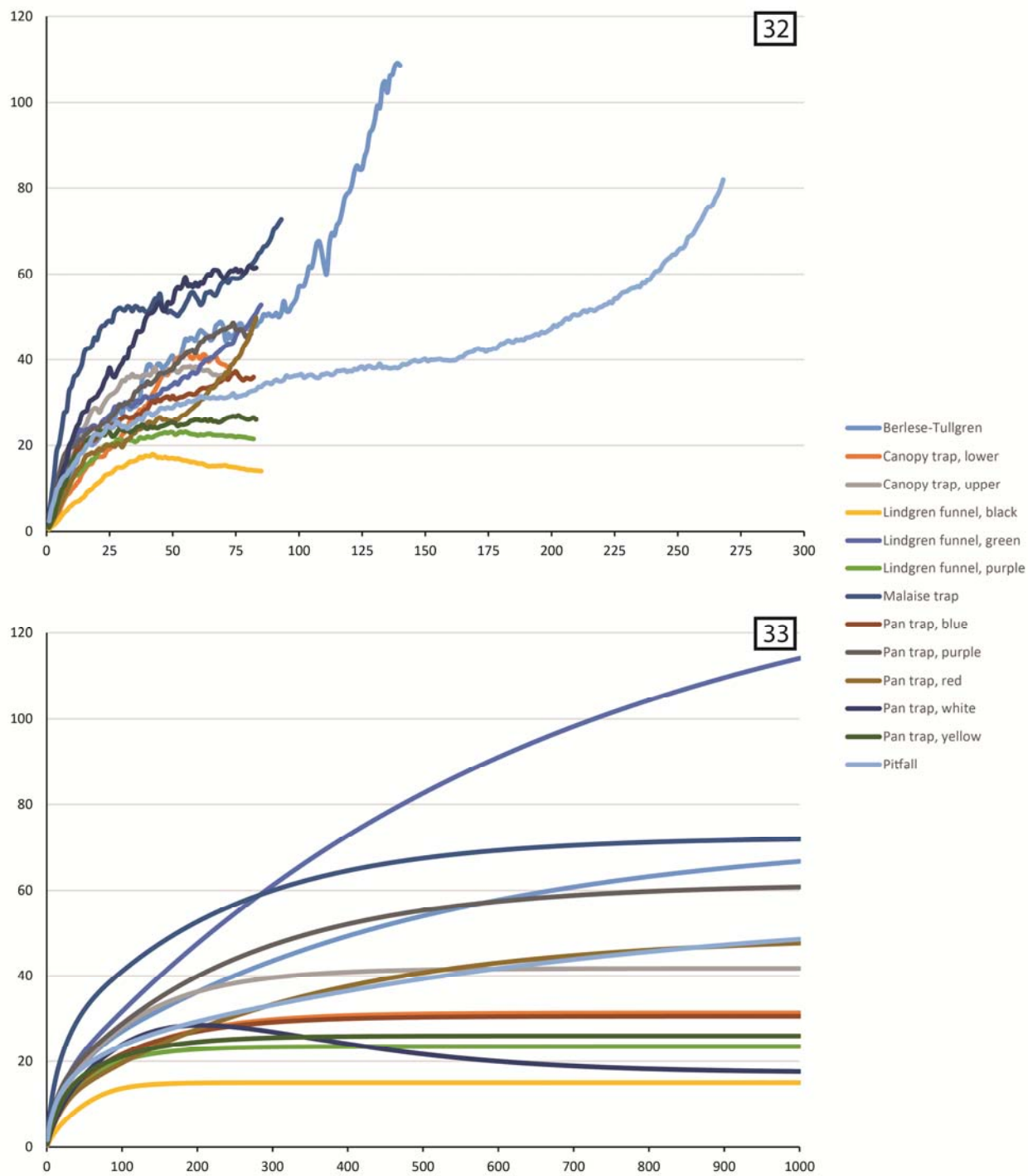
Figure 31. Total number of curculionoid specimens/species collected across all traps.

Species	ANOVA					Tukey-Kramer			
		df	ss	F	p-value	trap color	mean	sd	Separation of means
<i>Anthonomus rufipennis</i>	Between groups	2	3.56	16	0.004*	black	0	0	b
	Within groups	6	0.67			green	1.3	0.58	a
	Total	8	4.22			purple	0	0	b
<i>Anthonomus sutralis</i>	Between groups	2	22.93	4.05	0.045*	black	0.4	0.89	a
	Within groups	12	34			green	0	0	a
	Total	14	56.93			purple	0	0	a
<i>Apteromechus ferratus</i>	Between groups	2	0.08	0.02	0.983	black	1.63	1.77	-
	Within groups	21	51.25			green	1.75	1.67	-
	Total	23	51.33			purple	1.63	1.19	-
<i>Conotrachelus anaglypticus</i>	Between groups	2	4.95	4.46	0.027*	black	1.29	0.95	a
	Within groups	18	10			green	0.43	0.79	a,b
	Total	20	14.95			purple	0.14	0.38	b
<i>Conotrachelus aratus</i>	Between groups	2	28.58	1.59	0.228	black	0.25	0.71	-
	Within groups	21	189.25			green	1.13	0.99	-
	Total	23	217.83			purple	2.88	5.06	-
<i>Conotrachelus elegans</i>	Between groups	2	16.33	49	0.005*	black	0	0	b
	Within groups	3	0.5			green	0	0	b
	Total	5	16.83			purple	3.5	0.71	a
<i>Conotrachelus naso</i>	Between groups	2	1.78	1.54	0.247	black	0.33	0.52	-
	Within groups	15	8.67			green	1.00	1.10	-
	Total	17	10.44			purple	0.33	0.52	-

Table 4. Results of ANOVA tests comparing the effect of color on the number of specimens of different species of Curculionoidea collected in Lindgren funnel traps. $P < 0.05$ is considered significant. Significant values are indicated by as asterisk (*).

Species	ANOVA					Tukey-Kramer			
		df	ss	F	p-value	trap color	mean	sd	Separation of means
<i>Cossonus impressifrons</i>	Between groups	2	6	6.35	0.019*	black	0	0	b
	Within groups	9	4.25			green	0	0	b
	Total	11	10.25			purple	1.75	0.96	a
<i>Cyrtepidomus castaneus</i>	Between groups	2	16.44	4.4	0.031*	black	0.33	0.82	b
	Within groups	15	28			green	1.33	1.03	a,b
	Total	17	44.44			purple	2.67	1.97	a
<i>Dryophthorus americanus</i>	Between groups	2	25	25	<0.001*	black	0	0	b
	Within groups	9				green	0	0	b
	Total	11				purple	1.25	0	a
<i>Eugnamptus angustatus</i>	Between groups	2	5.56	25	0.001*	black	0	0	b
	Within groups	6	0.67			green	0	0	b
	Total	8	6.22			purple	1.67	0.58	a
<i>Hypera meles</i>	Between groups	2	8	12	0.008*	black	0	0	b
	Within groups	6	2			green	2	0	a
	Total	8	10			purple	0	0	b
<i>Lechriops oculatus</i>	Between groups	2	14.78	4.1	0.038*	black	0.17	0.41	a
	Within groups	15	27			green	2.17	2.14	a
	Total	17	41.78			purple	0.33	0.82	a
<i>Madarellus undulatus</i>	Between groups	2	4.33	2.17	0.262	black	0.50	0.71	-
	Within groups	3	3			green	1.00	1.41	-
	Total	5	7.33			purple	2.50	0.71	-

Table 4 (cont.). Results of ANOVA tests comparing the effect of color on the number of specimens of different species of Curculionoidea collected in Lindgren funnel traps. $P < 0.05$ is considered significant. Significant values are indicated by as asterisk (*).



Figures 32, 33. Species rarefaction curves. Fig. 32. Chao 1 rarefaction curves based on the data. Fig. 33. Estimated rarefaction curves (S(est)) extrapolated to 1000 samples.

Green Lindgren funnel, Malaise, and purple pan traps exhibited high to very high similarity with respect to the species collected with each other (Sørensen = 0.55–0.61, Chao's Sørensen = 0.71–0.90) (Fig. 34). With one exception, Berlese-Tullgren and pitfall sampling exhibited medium similarity with Green Lindgren funnel and Malaise traps (Sørensen = 0.33–0.47, Chao's Sørensen = 0.39–0.47), but high to very high similarity with purple pan traps (Sørensen = 0.56, 0.59, Chao's Sørensen = 0.70, 0.93). Pan traps exhibited high to very high similarity with each other (Sørensen = 0.55–0.78, Chao's Sørensen = 0.88–0.98) and Malaise and upper and lower canopy traps exhibited medium to very high similarity (Sørensen = 0.41–0.63, Chao's Sørensen = 0.79–0.95).

Curculionoidea exhibited seasonality, with the most species and specimens collected in late spring and a secondary peak in the number of specimens collected in the fall (Figs. 29b, 30b). Overall, samples collected within five collection periods (approximately 10 weeks) have high to very high similarity with each other and medium to high similarity with samples further removed in time (Fig. 35c). However, individual trap types show less similarity: for example, Malaise traps collected distinct spring and fall species assemblages that both had medium similarity with the assemblage collected in the summer (Fig. 35a), while purple pan traps collected a distinct spring assemblage that was different from that collected in summer and fall (Fig. 35b).

Thirty five percent of the curculionoid species were collected in sufficient numbers to examine species-level phenology (Fig. 36a), while fifteen percent were collected throughout the study but in low numbers that do not allow any interpretation of phenology (Fig. 36b). Sixty four percent of the species, including all of the non-curculionid curculionoids, were found in low numbers during only a few trapping periods (Fig. 36c).

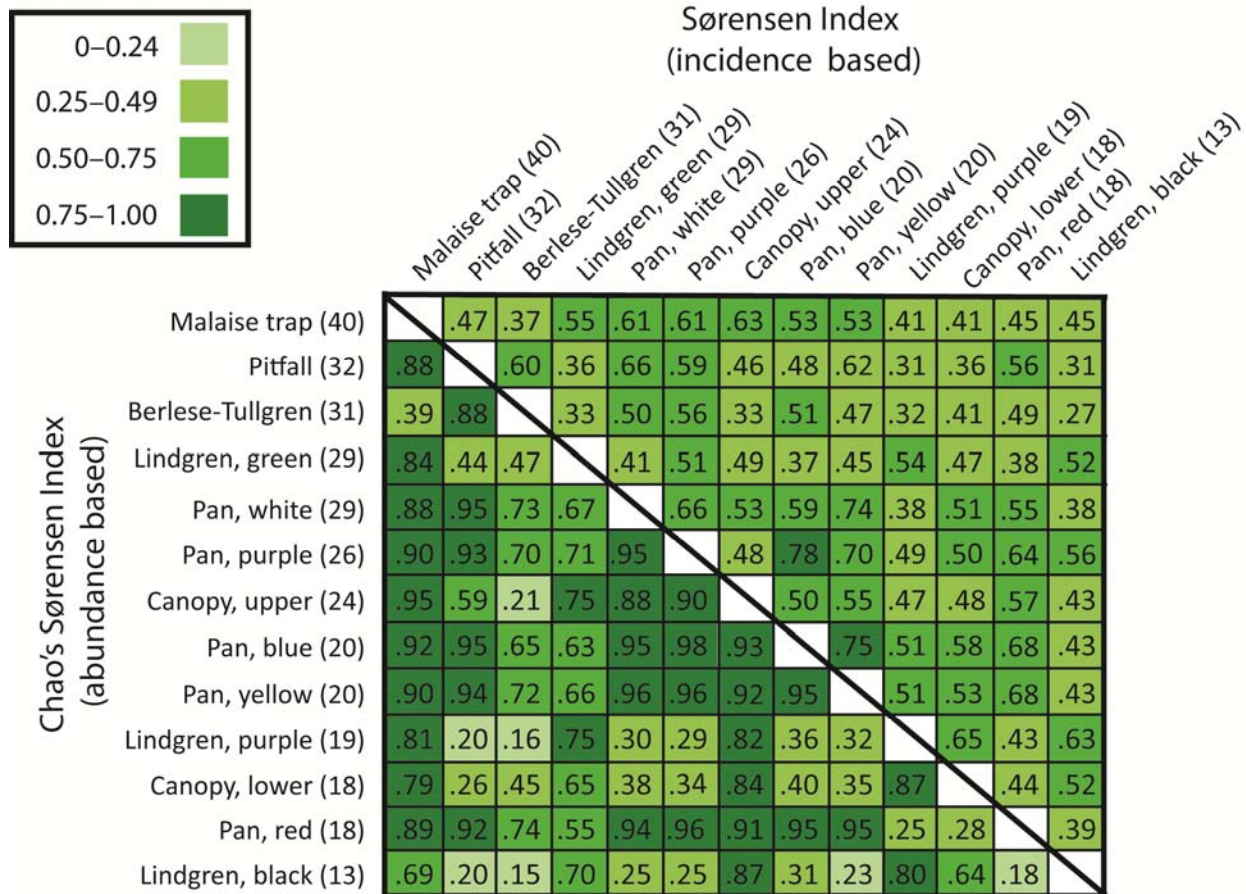


Figure 34. Similarity of trap catch as determined by Sørensen and Chao's Sørensen Indices. Number of species collected per trap type is indicated parenthetically after each trap type.

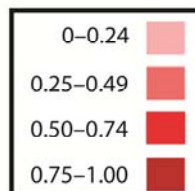
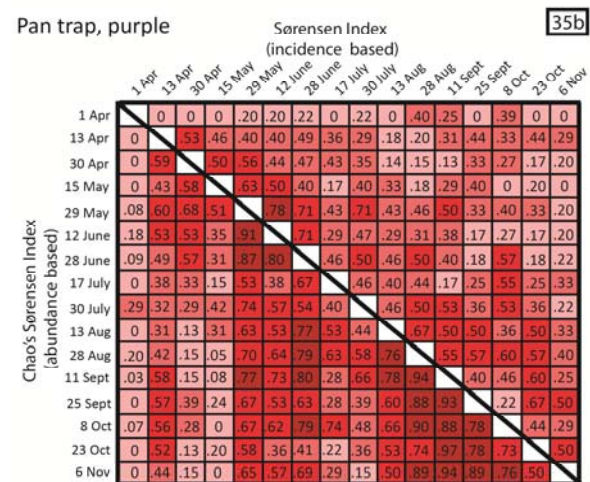
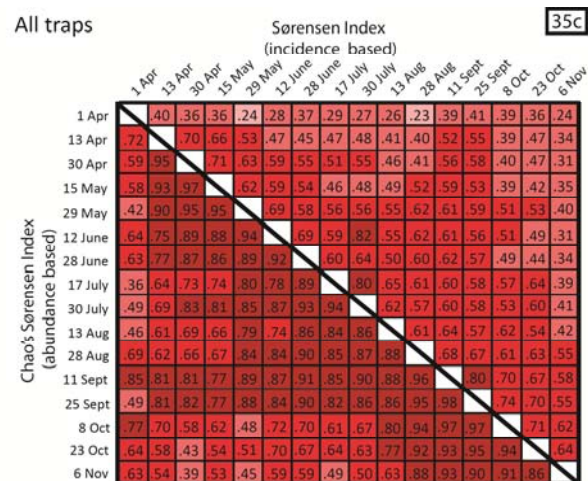
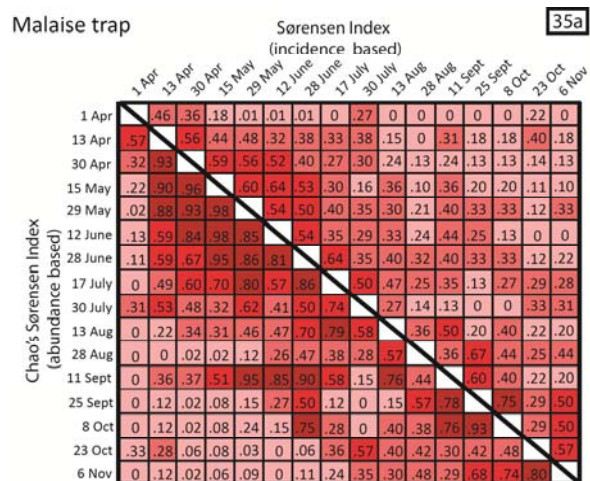


Figure 35. Sørensen and Chao's Sørensen Indices comparing similarity of trap catch by date in Malaise and purple pan traps and all trap catch combined.

Fig. 36a

Curculionidae

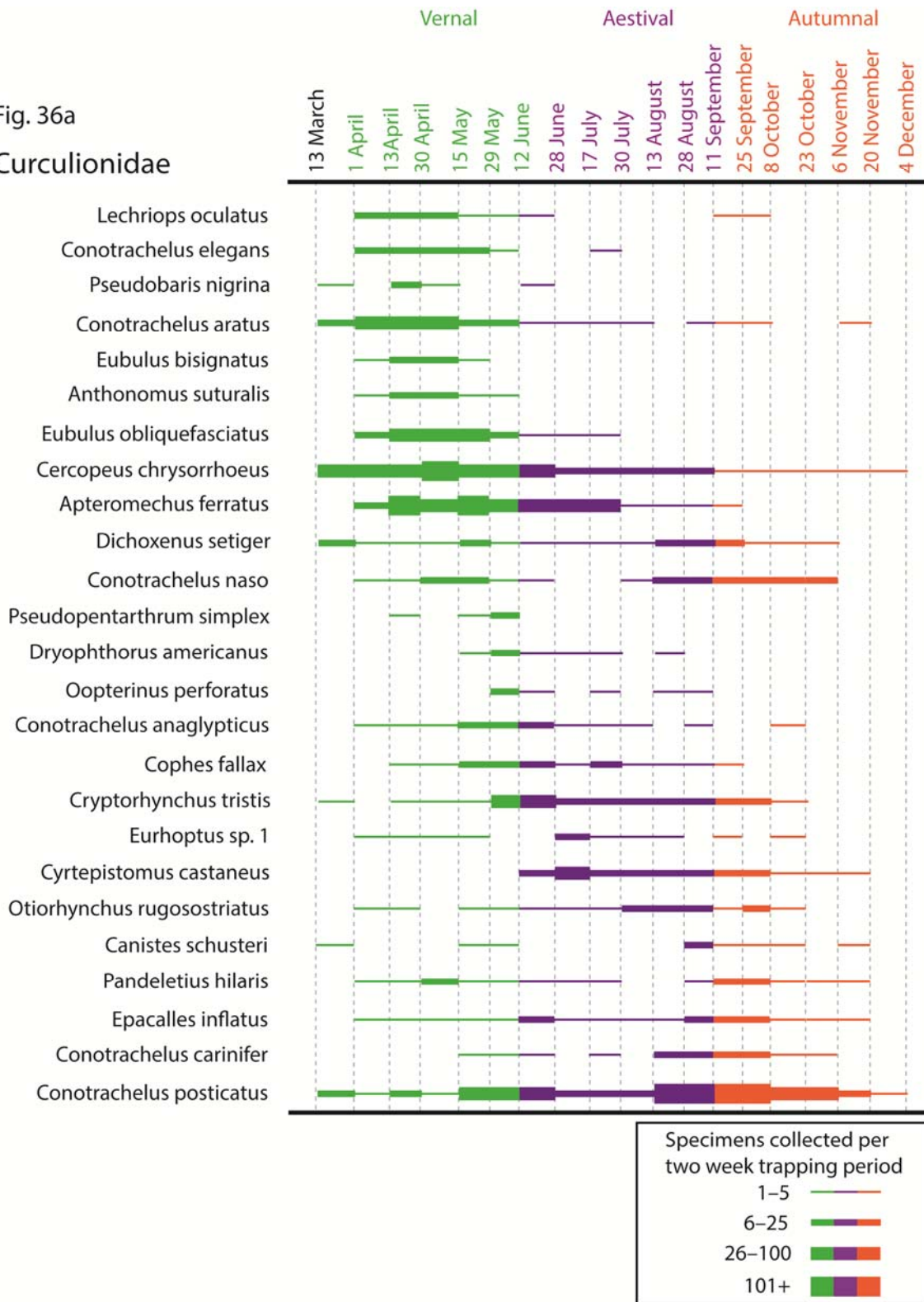


Fig. 36b
Curculionidae

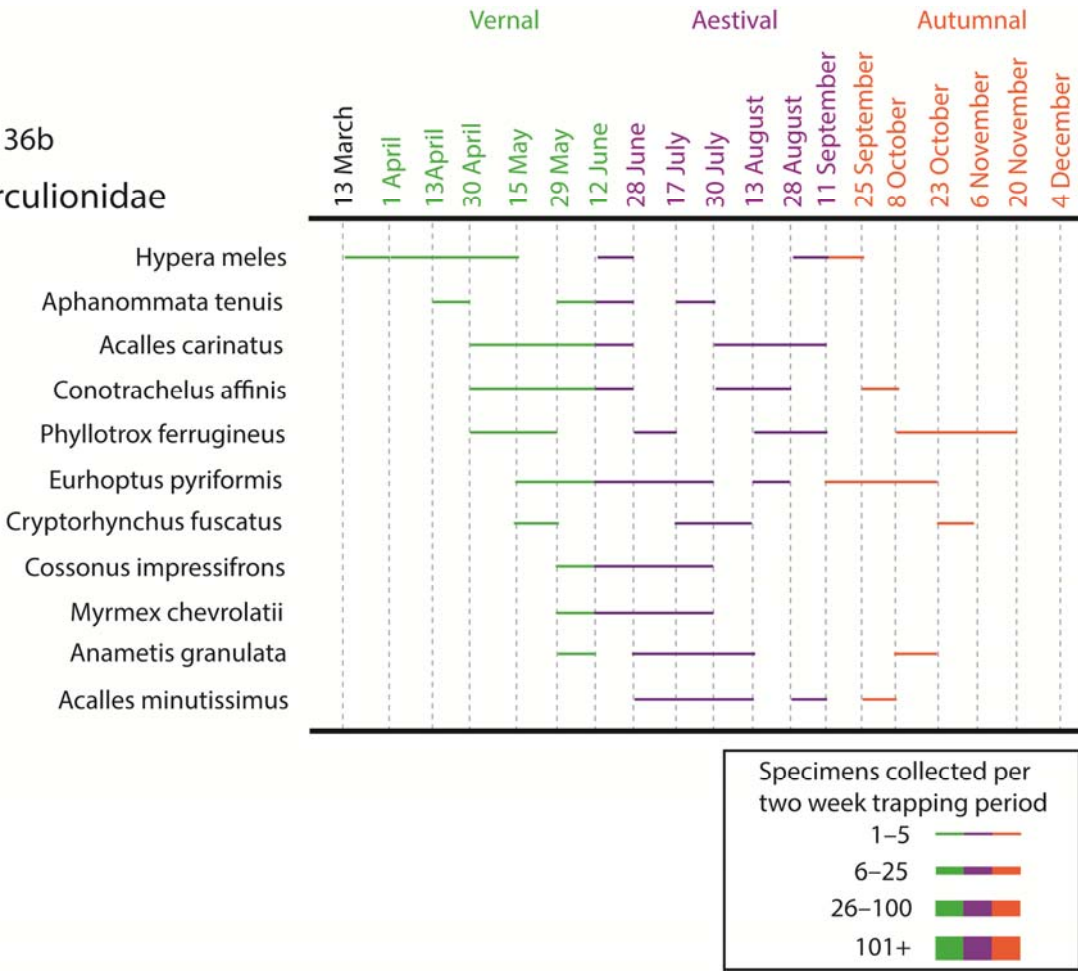


Fig. 36c

Curculionidae

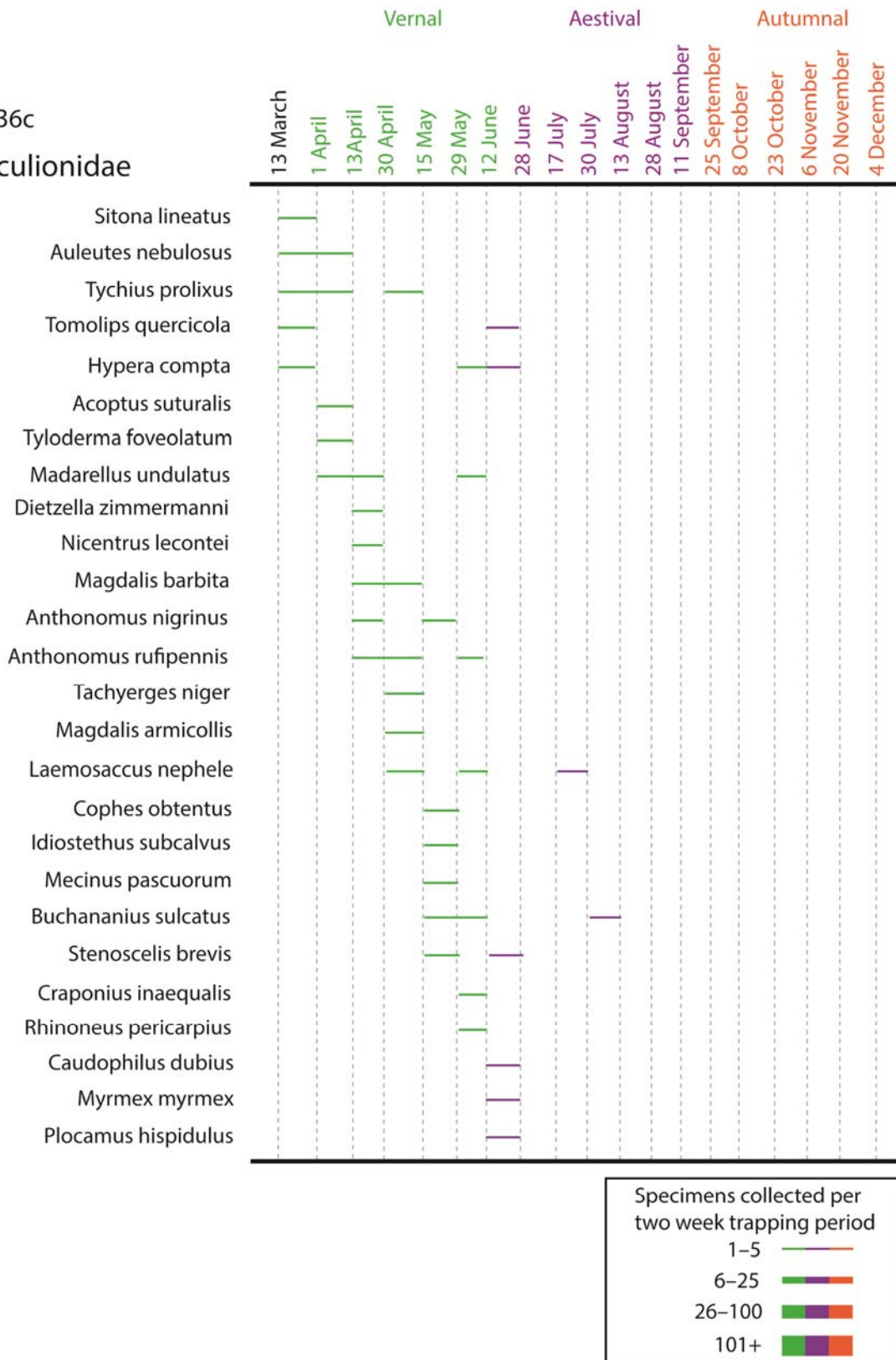


Fig. 36c (cont.)
Curculionidae

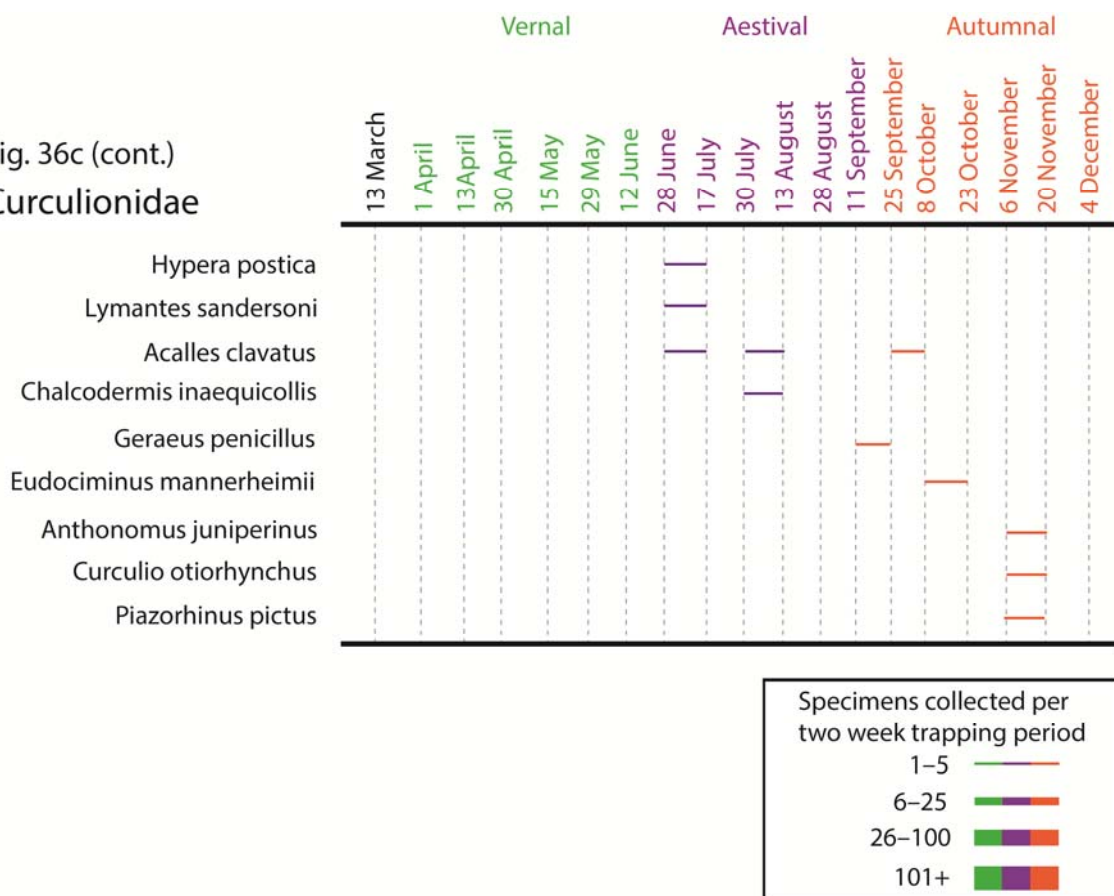


Fig. 36d

Anthribidae

Eurymycter fasciatus
Toxonotus cornutus
Euparius marmoreus
Ormiscus

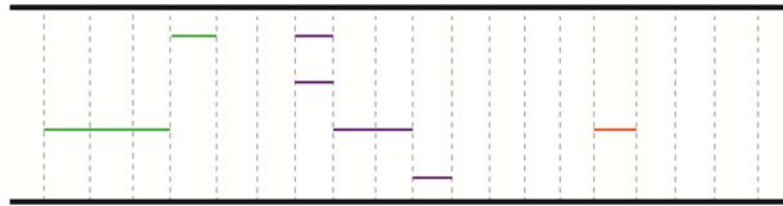


Fig. 36e

Attelabidae

Synolabus bipustulatus
Temnocerus aeratus
Eugnamptus angustatus



Fig. 36f

Brachyceridae

Notiodes limatulus



Fig. 36g

Brentidae

Arrhenodes minutus

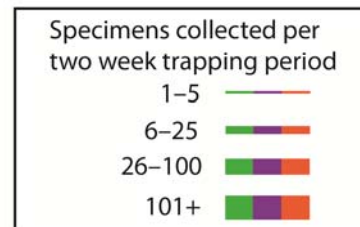
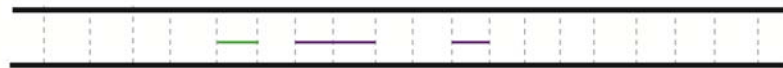


Figure 36. Phenology of curculionoids collected during this study summed across all trap types. **Fig. 36a.** Curculionidae with more than five specimens collected in at least one collecting period. **Fig. 36b.** Curculionidae with five or fewer specimens collected in any collection period but found in at least four collection periods. **Fig. 36c.** Curculionidae with five or fewer specimens collected in any collection period and found in three or fewer collection periods. **Fig. 36d.** Anthribidae. **Fig. 36 e.** Attelabidae. **Fig. 36f.** Brachyceridae. **Fig. 36g.** Brentidae.

Discussion.

Buprestidae

Malaise, upper canopy, green and purple Lindgren funnel traps collected the most buprestid species and specimens. Malaise and upper canopy traps exhibited high to very high similarity in the species collected with each other but, with two exceptions comparing Malaise traps to green and black Lindgren funnel traps using Chao's Sørensen index, only medium similarity with Lindgren funnel traps. Additionally, Malaise and upper canopy traps collected the largest buprestids at the site – *Chrysobothris* Eschscholtz and *Dicerca* Eschscholtz – in higher abundance than other methods. This indicated that Malaise and upper canopy traps, which were constructed from similar material and collect taxa in a similar fashion, targeted a species assemblage (i.e., large species) that other methods poorly sampled and also suggested that the large species are active both near the ground and in the canopy.

Trap color appeared to be an important component of Lindgren funnel traps when targeting buprestids. Green and purple Lindgren funnel traps exhibited only medium similarity in the species collected and differentially peaked in the number of species and specimens collected. Six of seven species analyzed were caught in significantly higher numbers by specific colored traps: four were caught in higher numbers by green traps, one by purple traps, and one by black traps. Other studies have examined the role of color in attraction and trapping of Buprestidae but most have either focusing at the family level or on economically important species (e.g., emerald ash borer, *Agrilus planipennis* Fairmaire) (Table 5). However, two studies (i.e., Petrice et al. 2013, Peatrice & Haack 2015) found that, while there was no difference in the attraction of emerald ash borer to green or purple traps, other *Agrilus* Curtis species demonstrate significant preference for green or green and purple traps. It is therefore probable that green and

purple Lindgren traps attract different species and that the bulk of studies that have examined color preference in emerald ash borer may not be applicable to other *Agrilus* or buprestids in general.

Highest-level taxon considered	Lowest taxonomic level identified	Reference
Insecta	family	Skvarla & Holland 2011
Coleoptera	family	Oliver <i>et al.</i> 2002
Coleoptera	species	Sakalian <i>et al.</i> 1993
Buprestidae	species	Sakalian 1993; Oliver <i>et al.</i> 2003; Sakalian & Langourov 2004; Peatrice <i>et al.</i> 2013; Peatrice & Haack 2015
<i>Agrilus</i>	species	Domingue <i>et al.</i> 2013
<i>Agrilus planipennis</i> Fairmaire	species	Francese <i>et al.</i> 2005; Otis <i>et al.</i> 2005; Francese <i>et al.</i> 2008; Lelito <i>et al.</i> 2008; Crook <i>et al.</i> 2009; Francese <i>et al.</i> 2010a; Francese <i>et al.</i> 2010b; Francese <i>et al.</i> 2011; Francese <i>et al.</i> 2013a; Francese <i>et al.</i> 2013b; Poland & McCullough 2014
<i>Agrilus sulcicollis</i> Lacordaire	species	Petrice & Haack 2014
<i>Agrilus bilineatus</i> (Weber)	species	Petrice & Haack 2014

Table 5. Select references pertaining to color attraction in Buprestidae.

Malaise and upper canopy traps were estimated to collect approximately the same number of buprestid species for the first fifty samples or so; however, species accumulation curves for upper canopy traps became asymptotic by 70 samples while the extrapolated rarefaction curve for Malaise traps was not estimated to approach an asymptote until nearly 1000 samples. This resulted in Malaise traps being expected to collect more than triple the number of species when large numbers of samples are taken.

Species accumulation curves for green and purple Lindgren funnel traps did not become asymptotic after 85 and 82 collections, respectively. Extrapolated rarefaction curves for both traps became asymptotic after approximately 350 samples

Pan traps generally collected the fewest buprestid species and specimens. Whether this was due to their placement under Malaise traps in this study, which may have obscured them to beetles in the canopy, or because buprestids were less likely to fly into traps placed on the ground is unclear. The only other study that compared the efficiency of pan traps to other trap types found that pan traps caught the fewest buprestid species and specimens (McIntosh et al. 2001), which suggests the results presented here were to be expected. However, blue pan traps may be an exception as they collected nearly as many species and specimens as black Lindgren funnel and upper canopy traps and were estimated to collect the third most species after approximately 220 samples.

Seasonality in buprestids is attracting interest as emerald ash borer and other invasive buprestids threaten native and managed landscapes. In temperate climates similar to the site studied herein, Dodds and Ross (2002) found buprestids active throughout the summer with a peak in late summer, while Sakalian and Langourov (2004), found them to be most active in the early summer. However, Klingeman et al. (2015), after accumulating collection data from 15,217 specimens of 135 species from North Carolina and Tennessee, found seasonality varied by species, with many species active in early summer while others are found only in the spring or are active throughout the warm months. Thus, while there is some seasonality to buprestids in general, it is likely that much of the apparent seasonality in this and other studies was due to a relatively few number of specimens from a limited number of species.

Carabidae

Pitfall traps are generally thought to be the most effective trap to collect carabids and are often used to collect them (e.g., Greenslade 1964; Baars 1979; Waage 1985; Desender & Maelfait 1986; Halsall & Wratten 1988; Morrill et al. 1990; Niemelä et al. 1990; Wiedenmann et al. 1992; Work et al. 2002; Raworth & Choi 2003; Buchholz et al. 2010). Unsurprisingly, pitfall traps collected the most carabid species and specimens. However, leaf litter samples processed with Berlese-Tullgren extractors exhibited high species similarity with pitfall traps, which suggests both methods target the same assemblage of ground-dwelling carabids when samples are taken from forest floor leaf litter habitat and Berlese-Tullgren samples are better suited for qualitative sampling (Sabu & Shiju 2010; Sabu et al. 2011) as the fauna collected by pitfall traps are affected by a number of factors, such as trap diameter, trap material, and activity level of target species (for a detailed discussion of issues with pitfall traps see Skvarla et al. 2014 [Chapter II]). Additionally, Spence and Niemelä 1994 found large-bodied carabids dominate pitfall catch and small-bodied species dominate litter samples, so while both methods primarily target terrestrial species and may adequately sample that community after many samples, they may preferentially sample certain species when a limited number of samples are taken.

Aerial traps (i.e., Malaise, canopy, and Lindgren funnel traps) generally exhibited only low to medium similarity with pitfall traps and collected fifteen species in four tribes not caught in pitfall traps (number of species noted parenthetically): Lebiini (7), Bembidiini (4), Harpalini (2), and Platynini (2). Lebiini and *Tachyta* Kirby (Bembidiini) are arboreal and an expected component of aerial traps (Ball & Bousquet 2001). Two species, *Agonum crenulatum* (LeConte) and *Tachys oblitus* Casey (Platynini and Bembidiini, respectively) are attracted to UV lights (Ciegler 2000), so may fly frequently and encounter aerial traps. The remaining five species are

hygro- or mesophilous (Ciegler 2000); we therefore suggest these species were collected in aerial traps as they moved between preferred habitat patches and that pitfall traps placed near such habitat may have collected them. Considering this, aerial traps appeared to target a different, complimentary assemblage of carabids to pitfalls. This has been previously suggested by Ulyshen et al. (2005), who reported that canopy traps (top + bottom collector) collect smaller, more aerial carabid species more effectively than pitfall traps and should be used in combination with pitfall traps when surveying carabid diversity.

Different colored Lindgren funnel traps did not collect significantly different numbers of specimens in the two species tested. Color was likely not an important consideration when targeting aerial carabids.

Pan traps (except white pans) exhibited low to medium similarity with pitfall and aerial traps. However, pan traps collectively only caught three species – *Clivina pallida* (Say), *Cyclotrachelus torvus* (LeConte), *Galerita janus* (Fab.) – that were unique to pan traps and one species – *Galerita bicolor* (Drury) – in higher numbers in pan traps than other trap types. Of the three unique species, two were represented by singletons and one by two specimens, suggesting they were either uncommon in the habitat or none of the methods employed were suitable for collecting them. We therefore suggest that, while pan traps exhibited low similarity with other trap types, they are generally unsuitable for collecting carabids, especially when other more effective methods are employed.

Species accumulation curves for pitfall, Malaise, and purple Lindgren funnel traps did not become asymptotic after 268, 95, and 82 2-week samples, respectively, and extrapolated rarefaction curves for all three trap types did not become asymptotic after 1000 samples. This indicated that significantly more trapping effort is needed in order to inventory all species at the

site. Additionally, the extrapolated rarefaction curves suggest Malaise traps may be more effective than pitfall traps after approximately 500 samples.

Most species collected in large numbers were active during at least two seasons and only four species – *Amara musculus* (Say), *Calathus opaculus* LeConte, *Calleida viridipennis* (Say), *Cicindela sexguttata* Fab. – were found during a single season. Of these, *Cicindela sexguttata* and *Calleida viridipennis* are most active in spring and early summer (Zhou et al. 1993; Pearson et al. 2006), while *Amara musculus* and *Calathus opaculus* are active outside the period they were collected (Ciegler 2000). It is unclear why *A. musculus* and *C. opaculus* exhibited marked seasonality, though it may be due in part to the fact the study was only conducted for a single year.

No single species appeared to account for the high number of specimens collected during the late spring as a handful of species reached their peak densities at that time (e.g., *Brachinus americanus* (LeConte), *Trichotichnus autumnalis* (Say), *Anisodactylus rusticus* (Say), *Dicaelus sculptilis* Say). In contrast, the large number of *Cyclotrachelus incisus* LeConte combined with smaller, but significant, numbers of *Cymindis limbata* Dejean and *Pterostichus permundus* Say collected between 1 August and 8 October drove the high number of total specimens collected during that time.

Most species collected in low numbers were taken during the summer, with one species collected only in the spring and four species collected only in the fall. *Rhadine ozarkensis* Sanderson and Miller is likely the only species that is truly rare, as it is known only from the type series, which was collected from the twilight and dark zone of Fincher's Cave in adjacent Washington County (Sanderson & Miller 1941; P. Messer *pers. comm.*). Other species that were

collected in low numbers were likely either uncommon transients in the surveyed habitat or were present in the habitat but not readily collected by the methods employed.

While the most abundant species were generally present throughout the warm months, we suggest traps be continuously employed rather than during a single season because species compositions varied somewhat between seasons and species abundances varied markedly. If traps can't be used continuously, then representative samples should be taken during each season.

Cerambycidae

Cerambycidae have been collected using a variety of methods, including active methods such as beat-sheeting and sweeping of vegetation (Yanega 1996) and passive methods such as light trapping (Yanega 1996), rearing traps (Yanega 1996; Ferro et al. 2009; Ferro & Carlton 2011), pan traps (Groot & Nott 2001), Malaise and canopy traps (Vance et al. 2003; Noguera et al. 2007; Dodds et al. 2010), clear window traps (Ulyshen & Hanula 2007; Bouget et al. 2009; Sama et al. 2011) and silhouette intercept traps such as Lindgren funnel and panel traps (Dodds et al. 2010; Dodds et al. 2010; Miller & Crowe 2011). Of the trap types included in this study, Malaise and canopy traps collected the highest number of species and had significant similarity. This is useful for vertical stratification studies (e.g., Vance et al. 2003) as they do not collect different assemblages so are comparable. However, when conducting faunal surveys it would be more efficient to choose a complimentary trap rather than include both Malaise and canopy traps.

Lindgren funnel traps were estimated to collect approximately the same number of species after 600 samples and exhibited high to very high similarity in the species collected with Malaise and upper canopy traps and between differently colored Lindgren funnel traps. Trap

color did not generally affect the response of species to the traps as only one of the nine species analyzed, *Xylotrechus colonus*, was attracted in significantly higher numbers to one color (black) over another (green). Only a few studies have examined the role of color in attraction and trapping of cerambycids: Shipman (2011) and Skvarla and Holland (2011) found that when analyzed at the family level, longhorns are preferentially attracted to red and purple, respectively, though neither study included a large diversity of color choices and Sakalian et al. (1993) and Imrei et al. (2014) found that individual species are attracted to yellow. Other studies (e.g., Macias-Samano n.d.) found no effect of color when trapping cerambycids. It is likely that color attraction is species-specific and tied to biological traits, such as flower feeding and host-finding. Our data suggest that many cerambycids were attracted to the vertical silhouette of the trap regardless of the color used. Additionally, all but two species – *Molorchus bimaculatus* Say and *Stenosphenus notatus* (Oliver), both of which were collected in the spring – were collected in similar or higher numbers in Malaise and/or upper canopy traps, so we suggest that Lindgren funnels should generally not be considered if Malaise or canopy traps are also used.

Species accumulation curves for Malaise, upper canopy, and black, green, and purple Lindgren funnel traps did not become asymptotic after 95, 72, 85, 85, and 82 samples, respectively, and extrapolated rarefaction curves for the five trap types became asymptotic after approximately 400, 500, 500, 350, and 200 samples, respectively. This indicated that significantly more trapping effort is needed in order to inventory all species at the site.

Of the twenty species collected in high enough abundance to examine phenology, four reached peak densities in the spring and 16 reached peak densities during the late spring to mid-summer. Species that were found in more than three collection periods but not in high numbers exhibited a similar pattern, with three of twelve species being present only in the spring and nine

of twelve species being present from late spring through summer. Of the rarely collected species found in low numbers during three or fewer collection periods, approximately half were found in the spring and half during the summer; only two species – *Hyperplatys maculata* Blatchley and *Oncideres cingulata* (Say) – were found only in the fall. While there were a few cerambycids that can be collected during the fall and a few that may be collected in the early spring, the most efficient collection effort was from the late spring through mid- to late summer when most species reach their peak populations.

Curculionoidea

Weevils are a diverse group of beetles and no one method is commonly used to collect their diversity (Table 6). The most effective combination of traps should target both aerial and terrestrial species. Of the traps included in this study, Malaise and upper canopy collected the most aerial species on average; however, when extrapolating to 1000 samples, Malaise traps were estimated to collect the most species for the first 250 samples and green Lindgren funnels were estimated to collect the most species after 250 samples. Depending on the number of samples to be collected, either trap would be an acceptable choice for collecting flying weevils.

Trap type	Select References
Malaise trap	Dutcher <i>et al.</i> 1986; Anderson 2008a; Ohsawa 2008; Hespenheide 2009
Pan trap	Setyo Leksono 2005
Pitfall trap	Raffa & Hunt 1988; Levesque & Levesque 1994; Hanula 1999; Lowe <i>et al.</i> 2010
Berlese extraction	Boland & Room 1983; Sakchoowong <i>et al.</i> 2007
Lindgren funnel trap	Anderson 2008b; Lowe <i>et al.</i> 2010; Hanula <i>et al.</i> 2011; Brar <i>et al.</i> 2012; Nam <i>et al.</i> 2013; Rassati <i>et al.</i> 2014
Window trap	Levesque & Levesque 1994; Anderson 2009a; Anderson 2008b

Table 6. Select references pertaining to trapping Curculionidae.

Pitfall traps and Berlese-Tullgren extraction collected the most terrestrial species on average and did not differ significantly with respect to the numbers collected. However, Berlese-Tullgren extraction is estimated to collect twenty additional species after 1000 samples. Depending on the facilities available, either method would be acceptable when targeting terrestrial weevils.

Purple and white pan traps also collected high numbers of species, but exhibited high similarity with Malaise, canopy, and pitfall traps and Berlese sampling in the species collected, which suggests pan traps were collecting both aerial and terrestrial species. Because pan traps were set under Malaise traps in this study, it is unknown whether pan traps set alone would be as effective as was suggested by these results. However, the addition of pan traps should be considered if Malaise traps are also being employed.

The attractiveness of various colors to different weevils has been previously investigated, almost exclusively in relation to pestiferous species in agricultural settings (e.g., Roach et al. 1972; Leggett & Cross 1978; Riley & Schuster 1994; Smart et al. 1997; Reddy & Raman 2001; Leskey 2006; Abuaglala & Al-Deeb 2012). In this study, ten of the fourteen weevil species analyzed were collected in significantly higher numbers by at least one color of Lindgren funnel trap: one species was most attracted to black traps, three were most attracted to green traps, four were most attracted to purple traps, and one was attracted to both green and purple traps. Three of the four species in which no difference was detected were collected in higher abundance in Malaise traps; these species were likely flying around in abundance and happened to be collected in funnel traps.

The weevils collected exhibited a diversity of activity periods. Some species were most abundant during one or two seasons (e.g., *Apteromechus ferratus* (Say), *Conotrachelus Aratus*

(Germar), *Cercopeus chrysorrhoeus* (Say)) but were collected in low numbers throughout the year; others exhibited a bimodal distribution in abundance (e.g., *Conotrachelus naso* LeConte, *C. posticatus* Boheman) or were present during only one season (e.g., *Eubulus bisignatus* (Say), *Anthonomus suturalis* LeConte). More than half (51%) of species represented by one or a few specimens were collected in the spring, while only 17% of such species were collected in the summer or more than one season and 14% were collected only in the fall,; additionally, only 16 of the 71 species collected (22%) were not collected at all during the spring.

The number of specimens collected exhibited a bimodal distribution, with the most collected in spring and fall, while the number of species peaked in the spring and declined thereafter. The spring peaks of species and specimens were likely driven by the fact that most species were collected in the spring but some were not present later in the year and because a handful of species reach peak densities at that time while the fall spike in the number of specimens appear driven solely by the high abundance of *Conotrachelus postacatus*.

If collection time is limited, spring is the most effective time to sample as the most species are present. A small percentage of species were present only in the summer or fall, and those were collected in low numbers that are not indicative of phenology. Additionally, only a few species were most abundant in the summer and fall and a majority of these were also present during the spring.

Conclusions.

The combination of pitfall and Malaise traps can be used to sample Carabidae, Cerambycidae, and Curculionoidea as pitfall traps effectively collected terrestrial carabids and curculionoids and Malaise traps effectively collected cerambycids and the aerial assemblage of

carabids and curculionoids. Large buprestids were collected by Malaise traps, but the smaller species (e.g., *Agrilus*) were most effectively green Lindgren funnel traps.

Pan traps were set under Malaise traps in this study with the intent that they double as intercept traps. However, they were generally ineffective at collecting aerial, wood-boring groups (Buprestidae, Cerambycidae). Whether the pan traps would have collected more wood-boring beetles if they had been placed in exposed areas rather than under Malaise traps is unknown, though the paucity of studies using pan traps to collect these taxa may be indicative of their effectiveness. When targeting terrestrial species, pan traps act as pitfall traps (Skvarla et al. 2014 [Chapter II]). The pan traps in this study were not sunk into the ground and flush with the surface as the pitfall traps were, so their effectiveness at collecting cursorial species may have been diminished.

The color of Lindgren funnel traps was an important factor for some species of Buprestidae Carabidae, and Curculionidae, but not Cerambycidae. The effect of color in trapping different taxa is understudied when the aim is to sample biodiversity and studies that examine the attraction of color to pest species may not apply to the genus or family more generally (e.g., EAB to *Agrilus*).

Most taxa exhibited seasonality, with the highest number of species in all families present in the spring or early summer, although a minority of species were present only during the summer or fall. When targeting these families, the most effort should be made during the spring and early summer with supplemental collections made during mid- to late-summer and fall.

Finally, none of the accumulation curves for the most effective collection methods per family became asymptotic after 85 (green Lindgren funnel), 95 (Malaise trap), or 268 (pitfall trap) samples. Extrapolated rarefaction curves were not estimated to become asymptotic until

350 to more than 1000 samples, depending on the trap and target taxon. This suggested that much more effort is needed when collecting beetles as the rarest species are often those that tell the most about biodiversity.

Acknowledgements.

We thank Danielle Fisher for her assistance processing samples and curating specimens; Kyle Schnepp and Peter Messer for their assistance identifying Buprestidae and confirming and correcting identifications of Carabidae, respectively; and Ray Fisher for his endless enthusiasm and willingness to discuss this project.

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Appendix I. Species rarefaction curves.

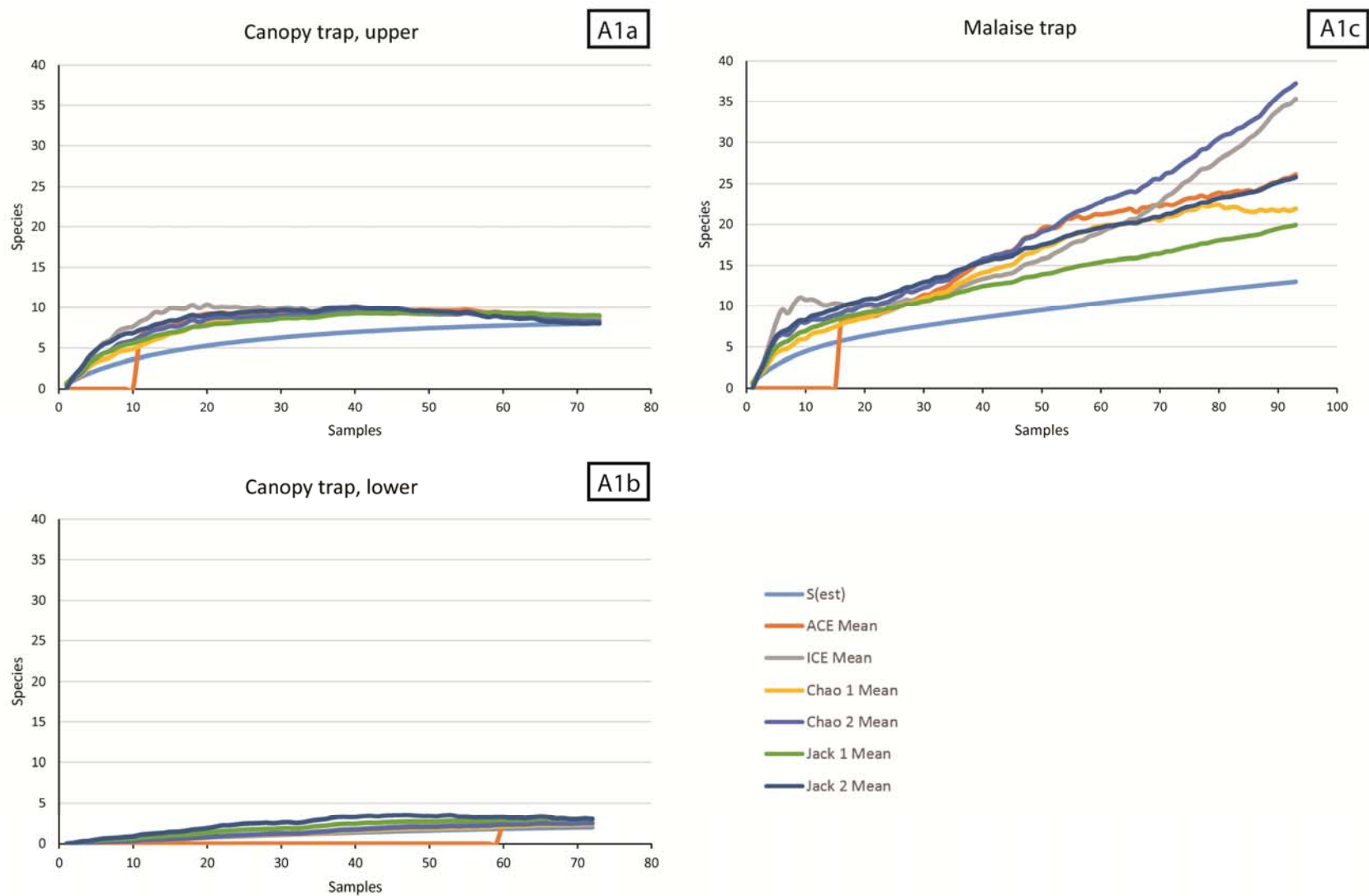
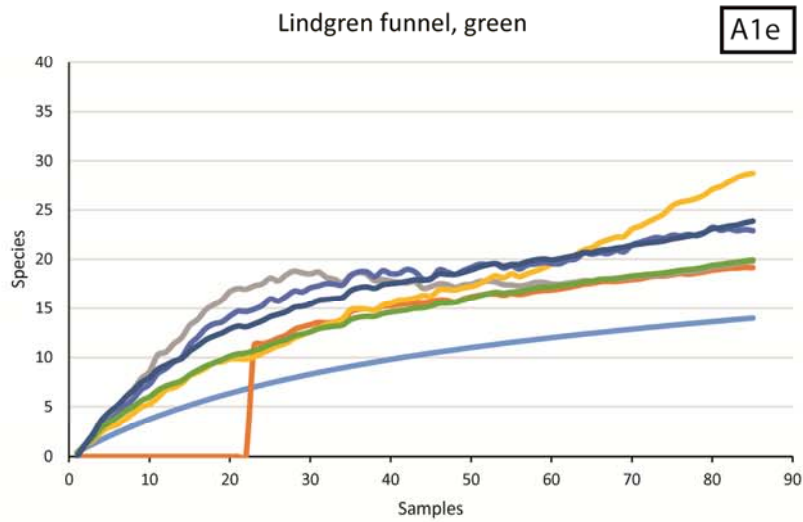
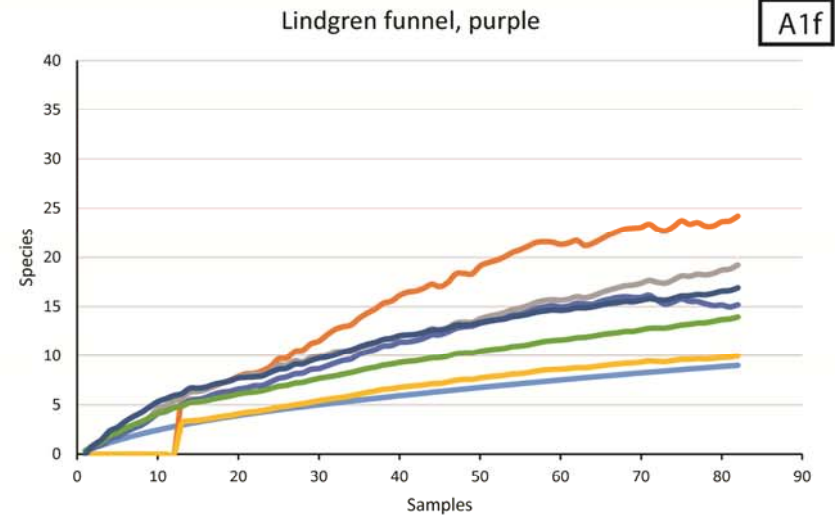
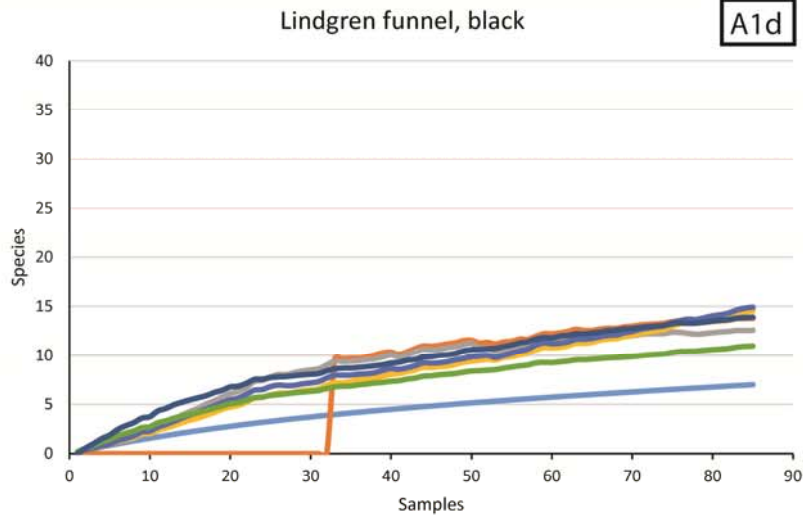
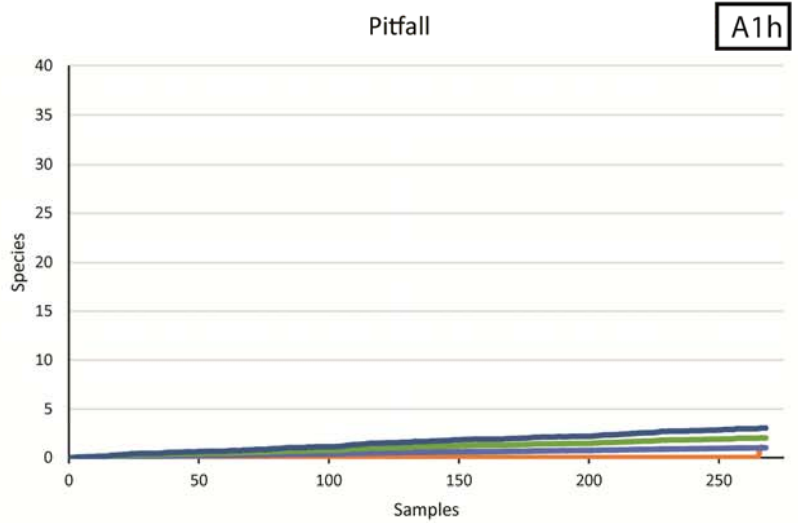
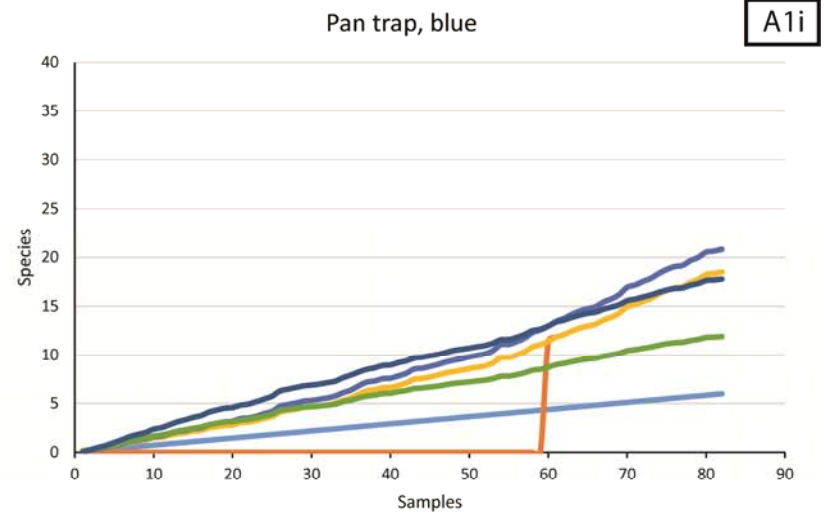
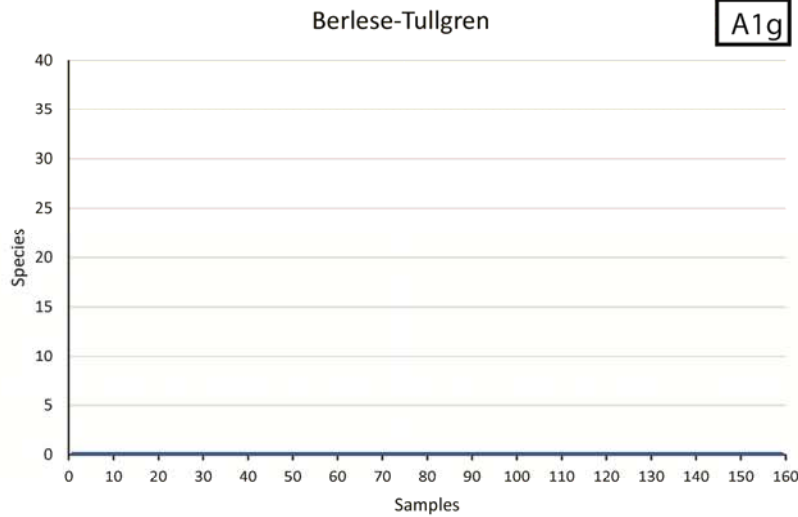


Figure A1. Buprestidae. See caption at the end of the figures for further explanation.



- S(est)
- ACE Mean
- ICE Mean
- Chao 1 Mean
- Chao 2 Mean
- Jack 1 Mean
- Jack 2 Mean

Figure A1 (cont.). Buprestidae. See caption at the end of the figures for further explanation.



- S(est)
- ACE Mean
- ICE Mean
- Chao 1 Mean
- Chao 2 Mean
- Jack 1 Mean
- Jack 2 Mean

Figure A1 (cont.). Buprestidae. See caption at the end of the figures for further explanation.

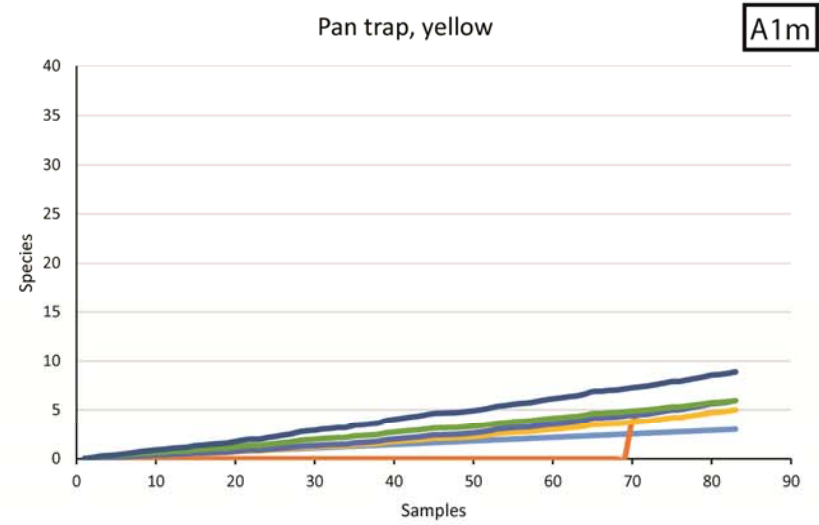
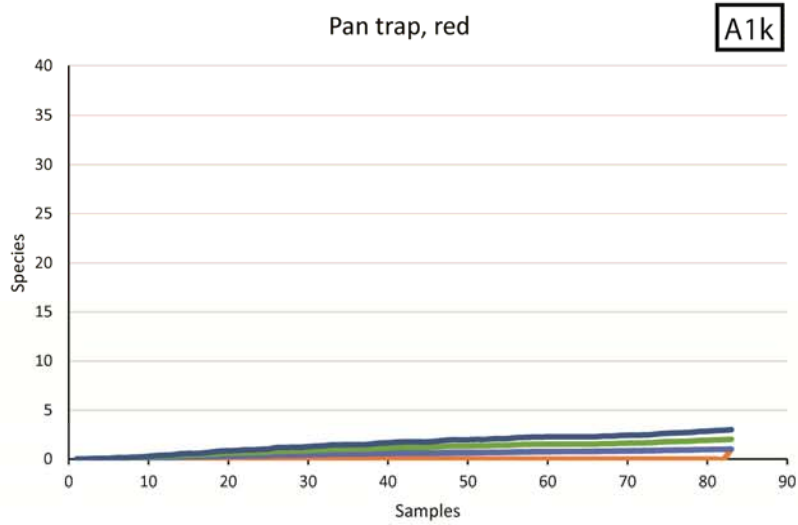
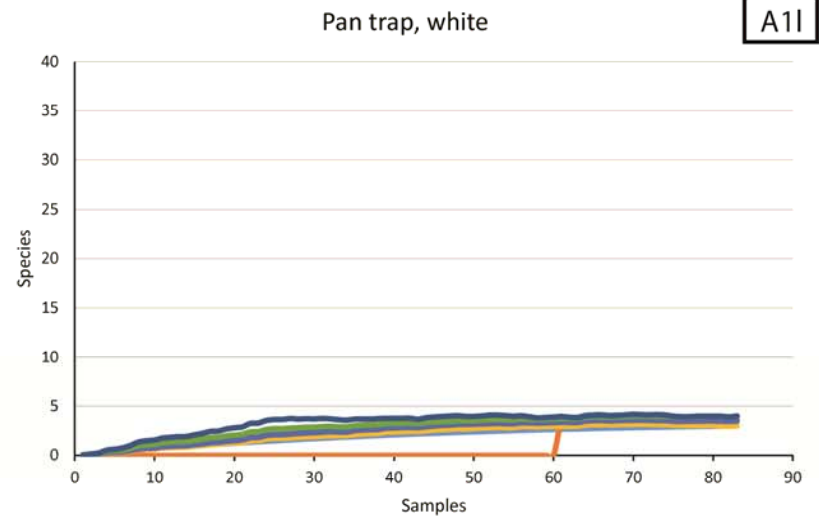
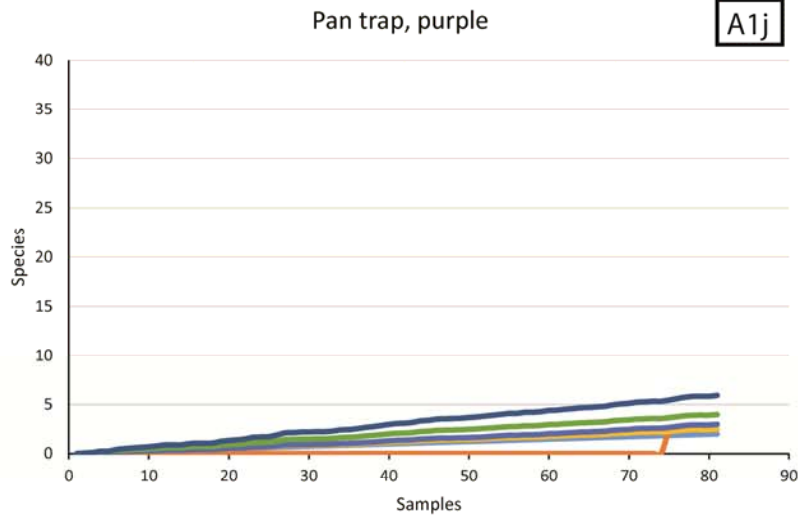
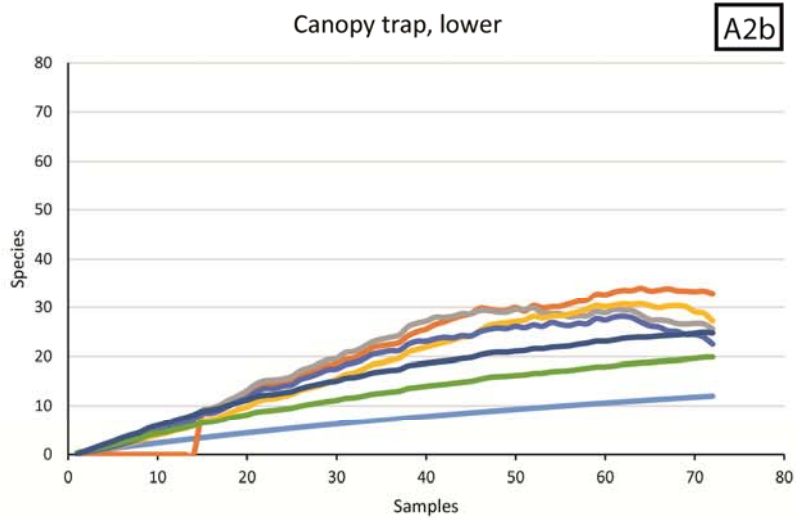
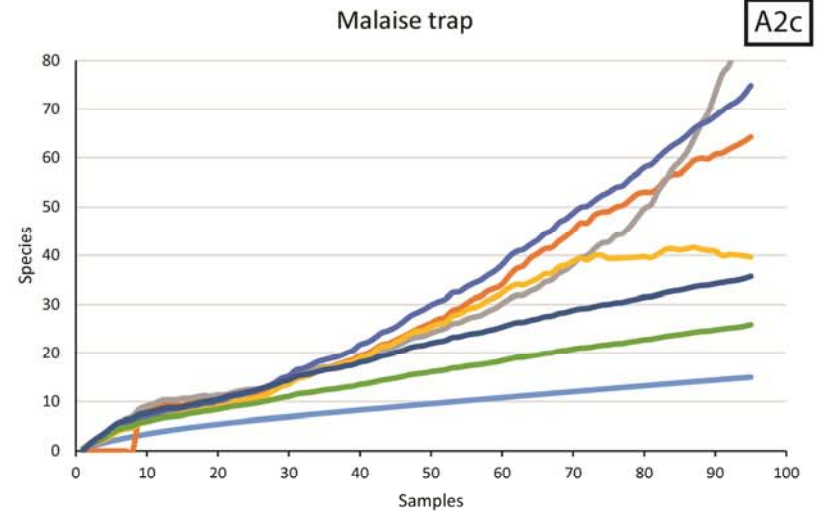
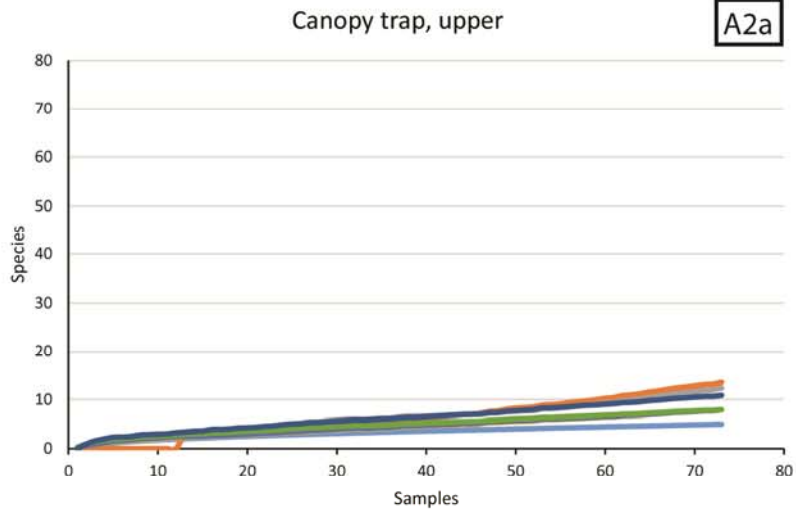
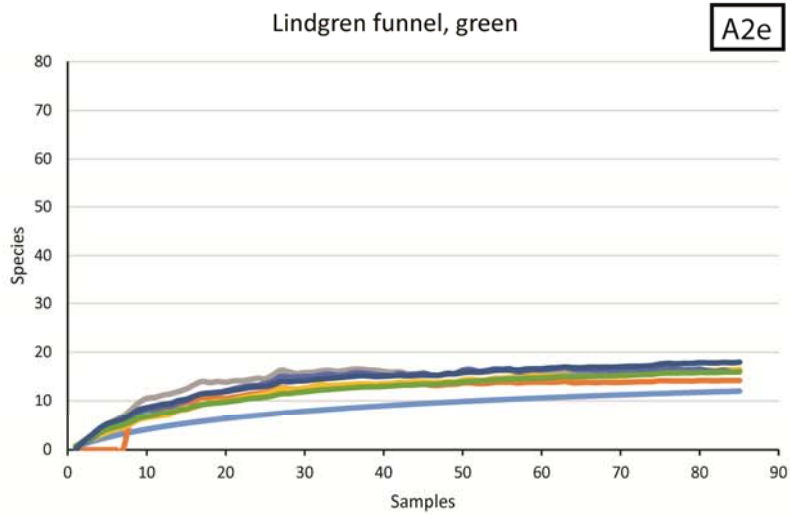
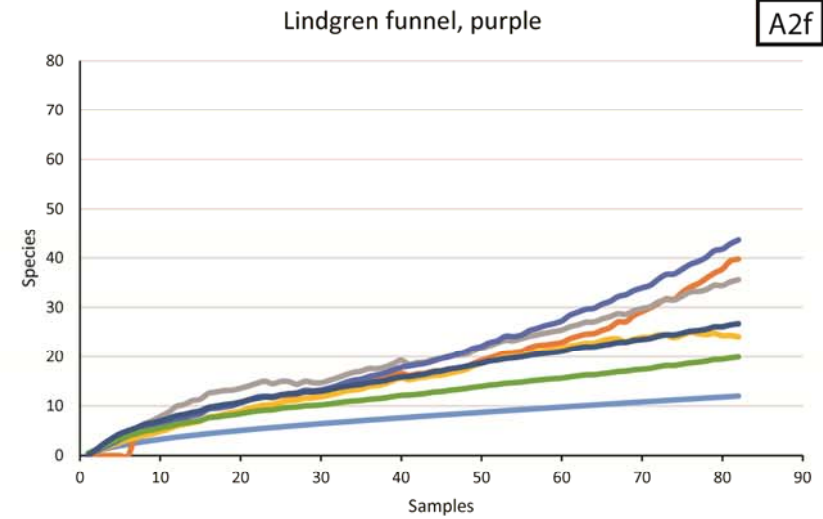
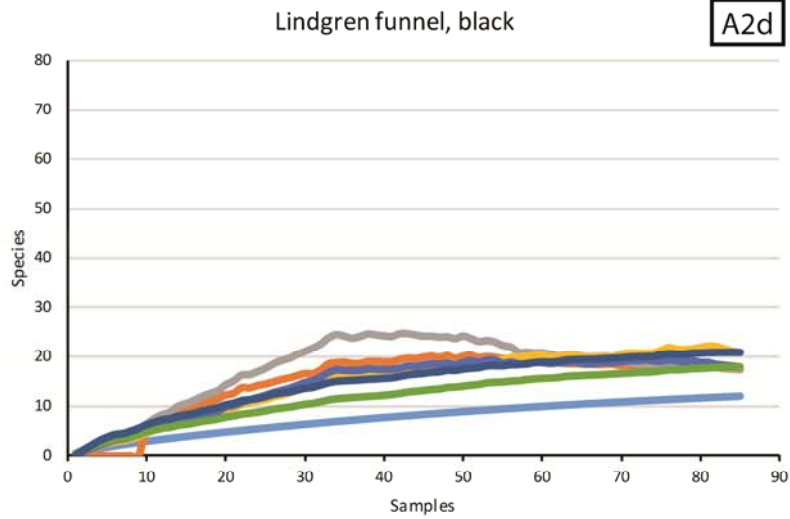


Figure A1 (cont.). Buprestidae. See caption at the end of the figures for further explanation.



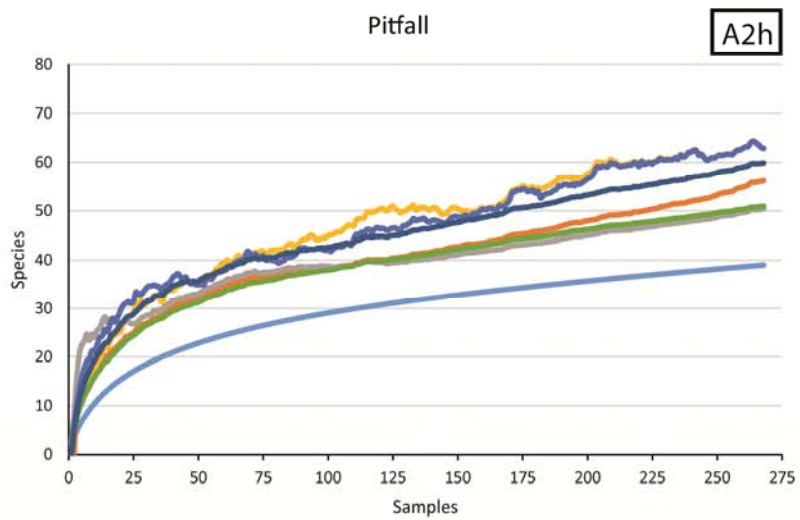
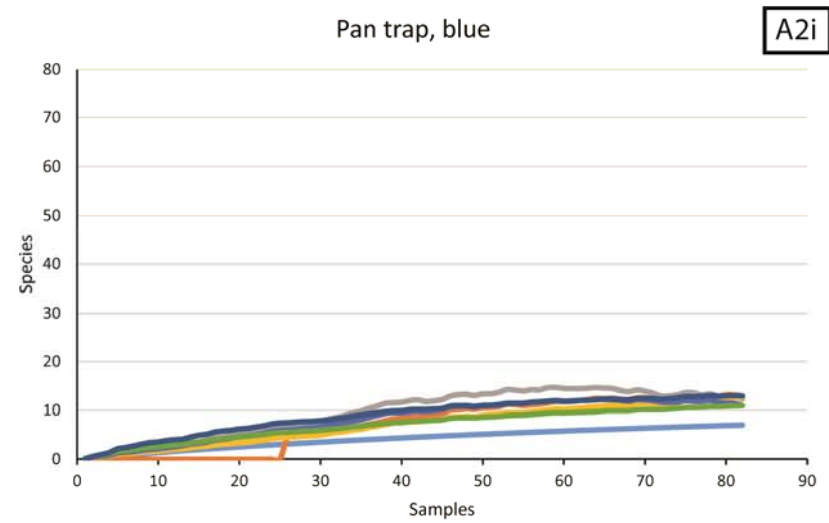
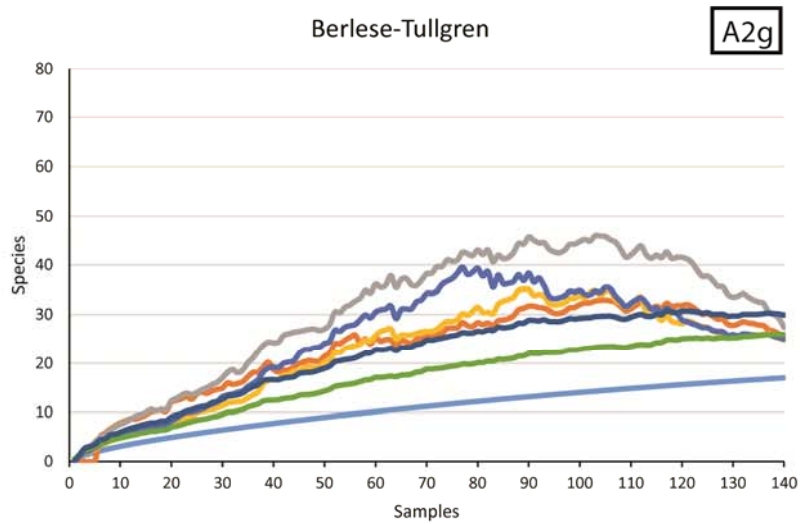
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Figure A2. Carabidae. See caption at the end of the figures for further explanation.



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 — Chao 2 Mean
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Figure A2 (cont.). Carabidae. See caption at the end of the figures for further explanation.



- S(est)
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- Chao 1 Mean
- Chao 2 Mean
- Jack 1 Mean
- Jack 2 Mean

Figure A2 (cont.). Carabidae. See caption at the end of the figures for further explanation.

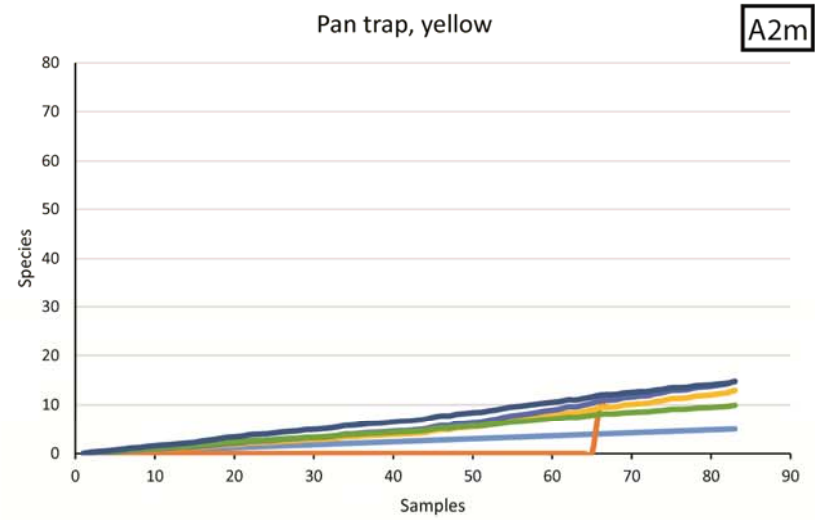
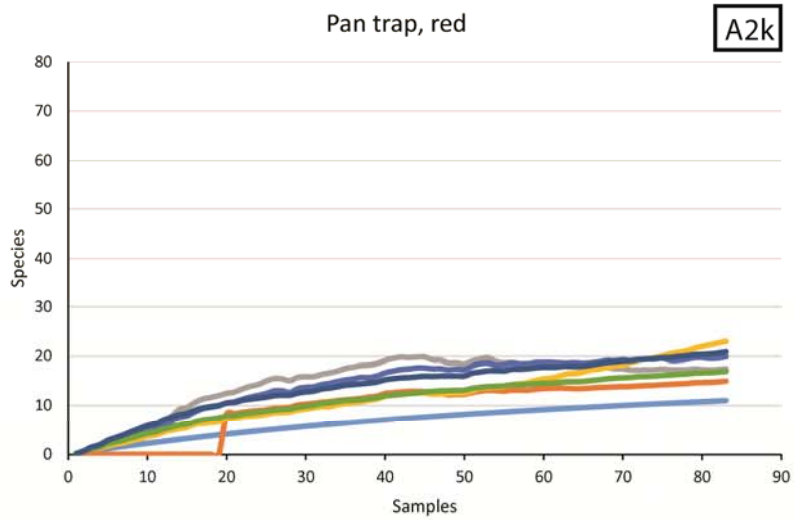
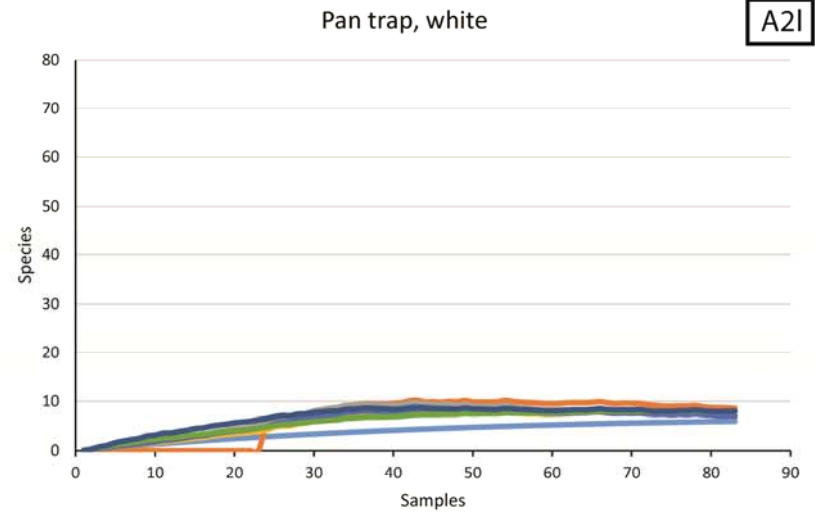
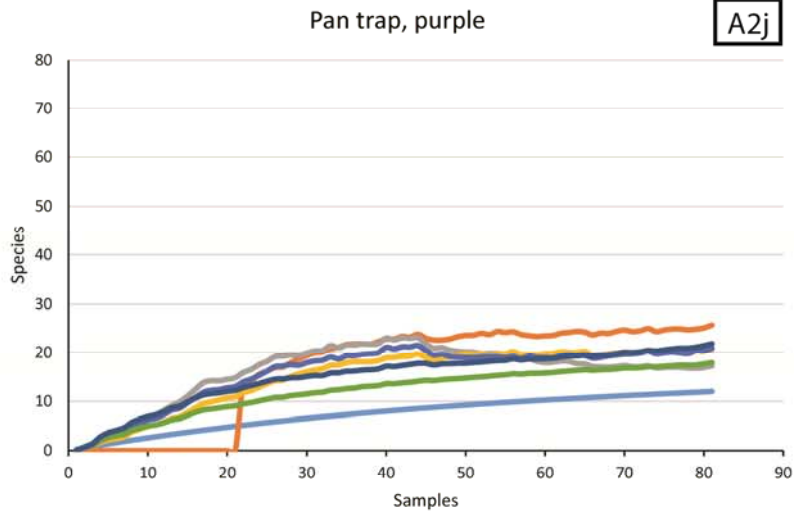
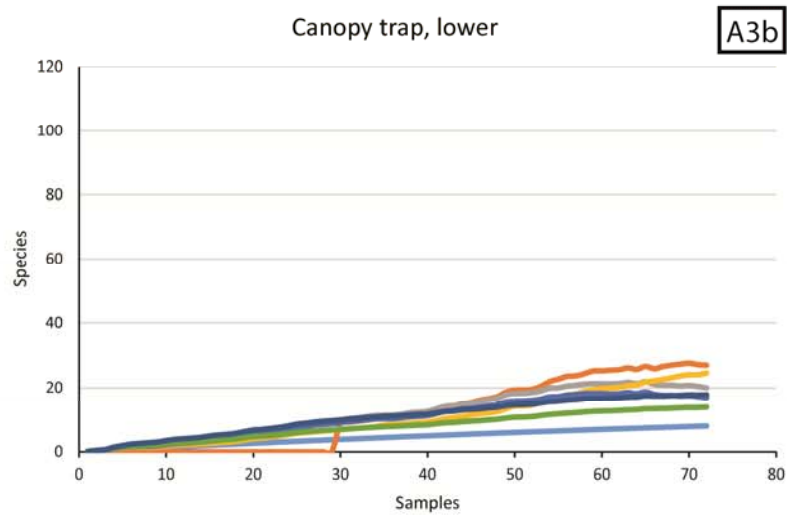
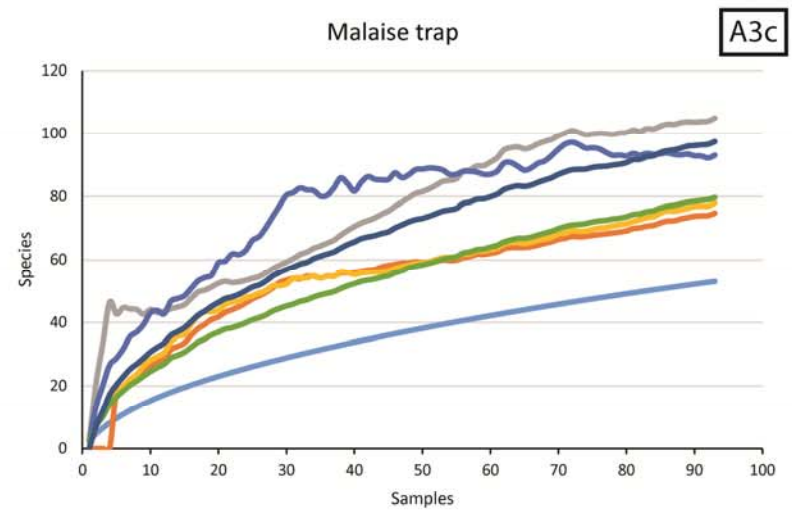
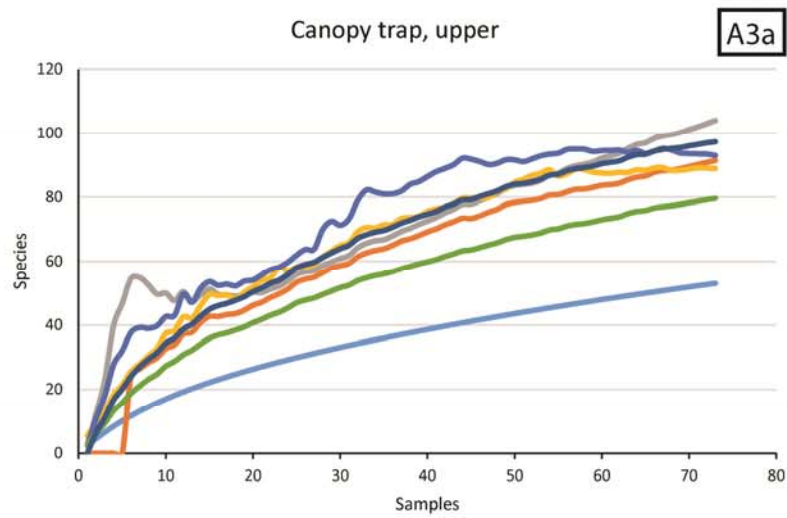
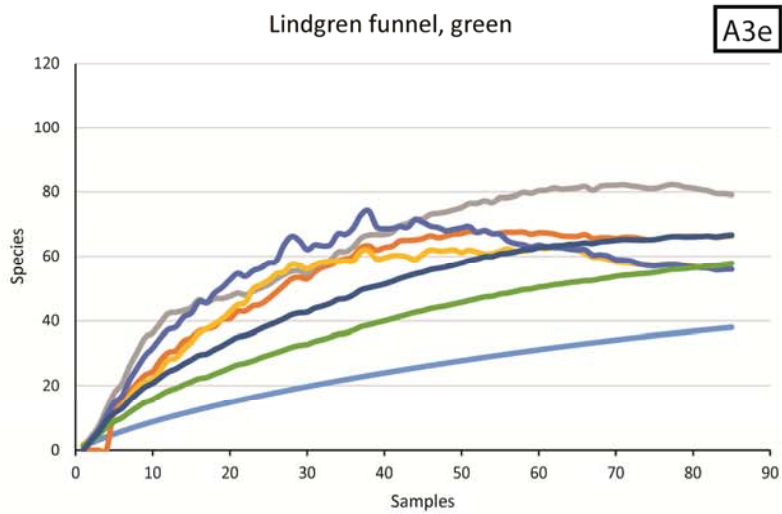
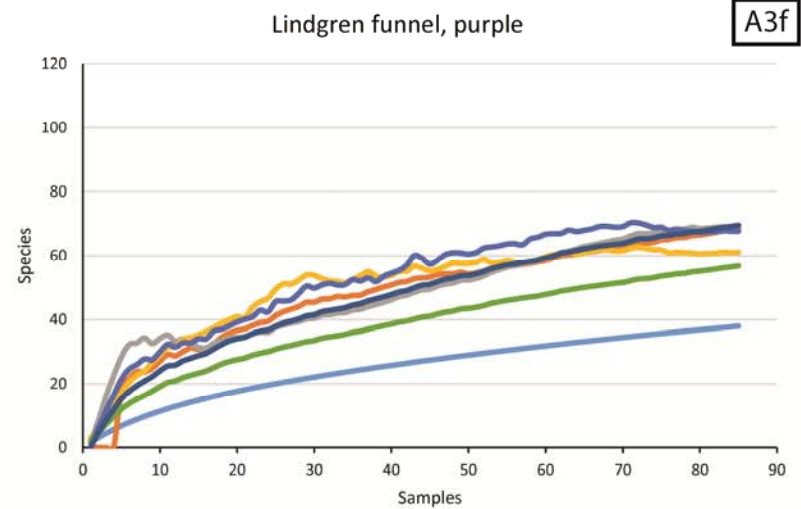
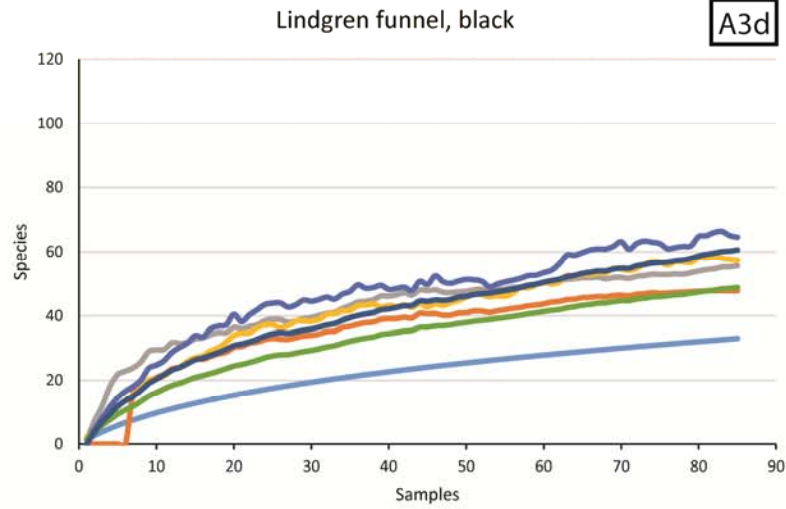


Figure A2 (cont.). Carabidae. See caption at the end of the figures for further explanation.



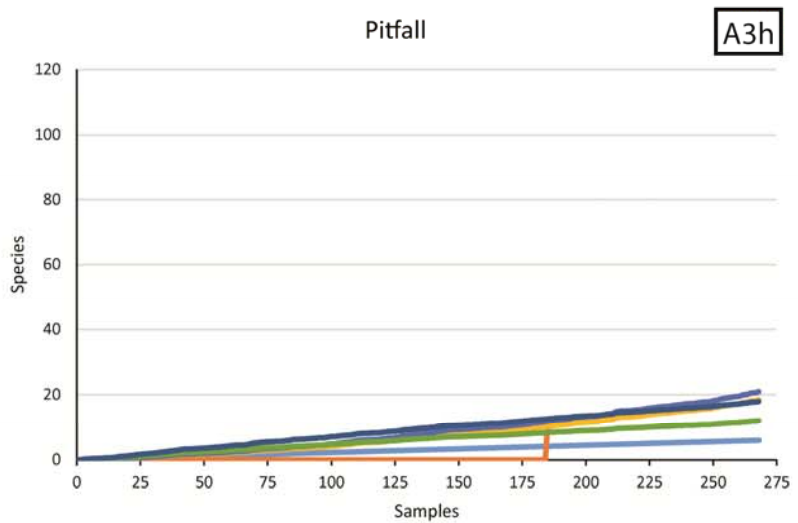
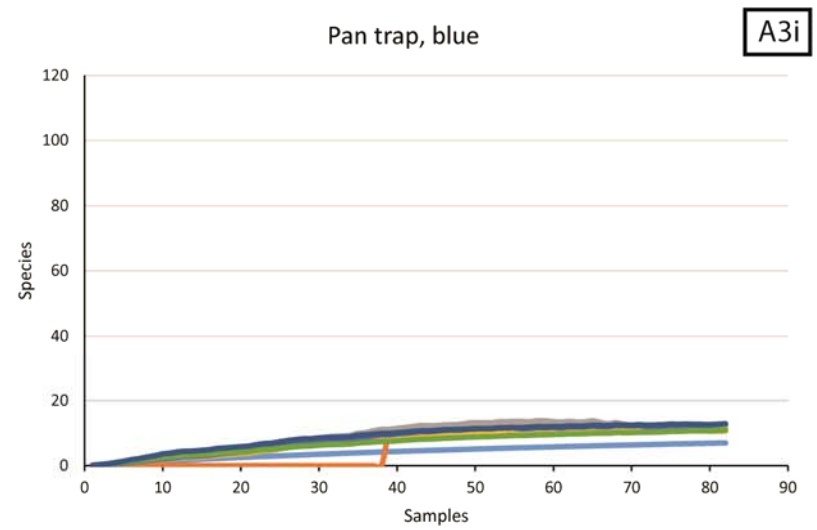
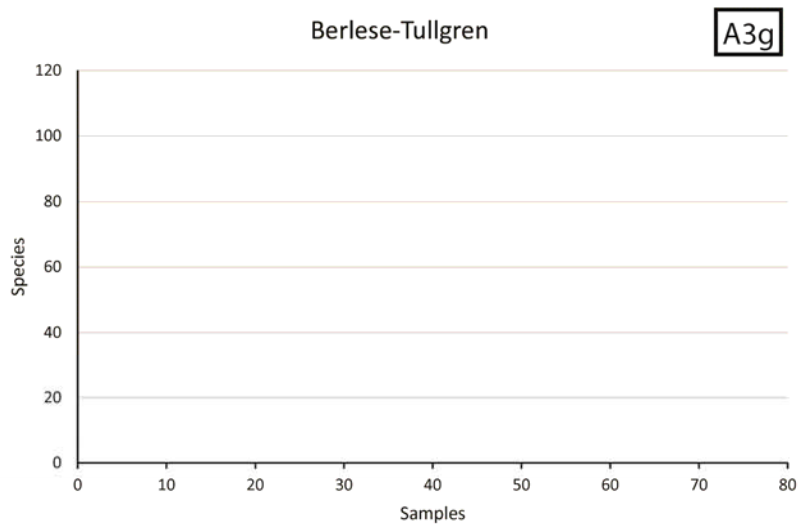
— S(est)
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 — Jack 1 Mean
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Figure A3. Cerambycidae. See caption at the end of the figures for further explanation.



— S(est)
 — ACE Mean
 — ICE Mean
 — Chao 1 Mean
 — Chao 2 Mean
 — Jack 1 Mean
 — Jack 2 Mean

Figure A3 (cont.). Cerambycidae. See caption at the end of the figures for further explanation.



- S(est)
- ACE Mean
- ICE Mean
- Chao 1 Mean
- Chao 2 Mean
- Jack 1 Mean
- Jack 2 Mean

Figure A3 (cont.). Cerambycidae. See caption at the end of the figures for further explanation.

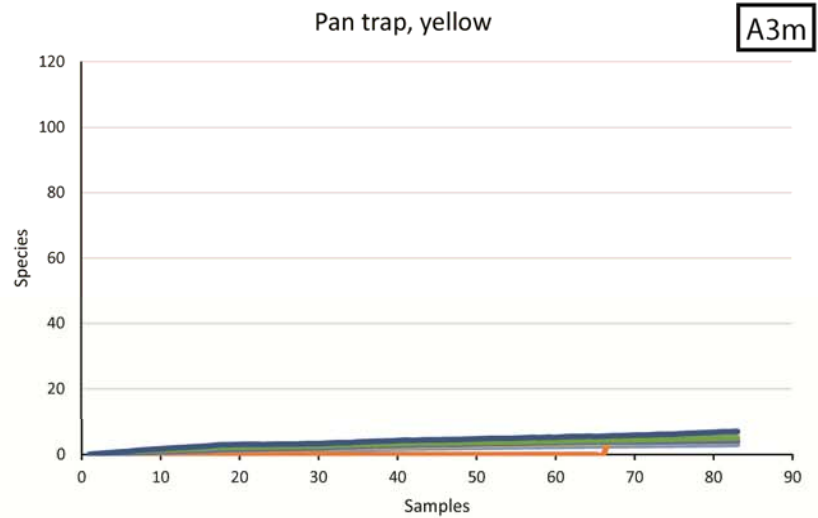
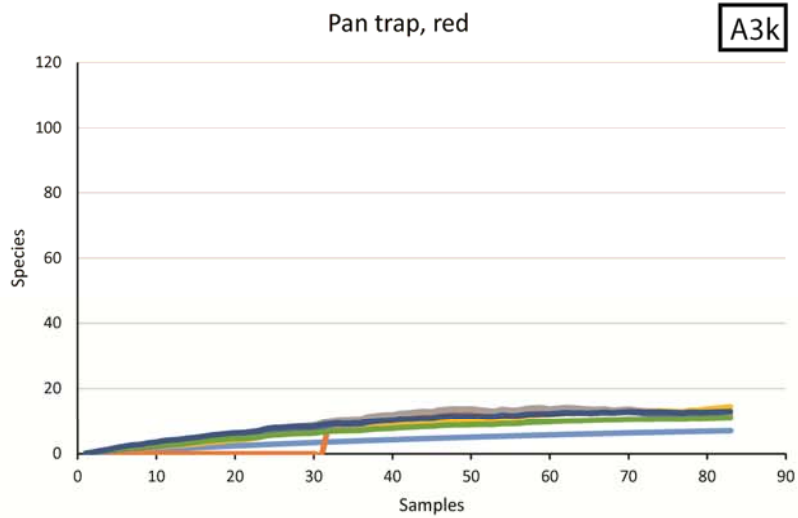
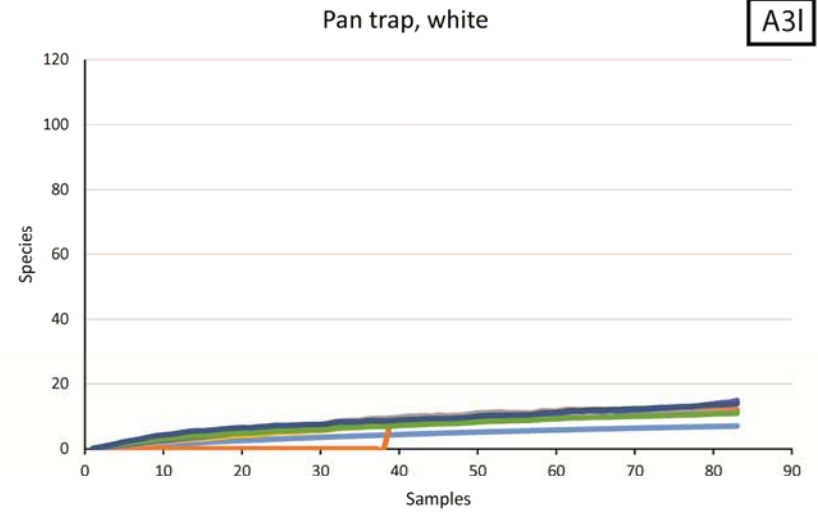
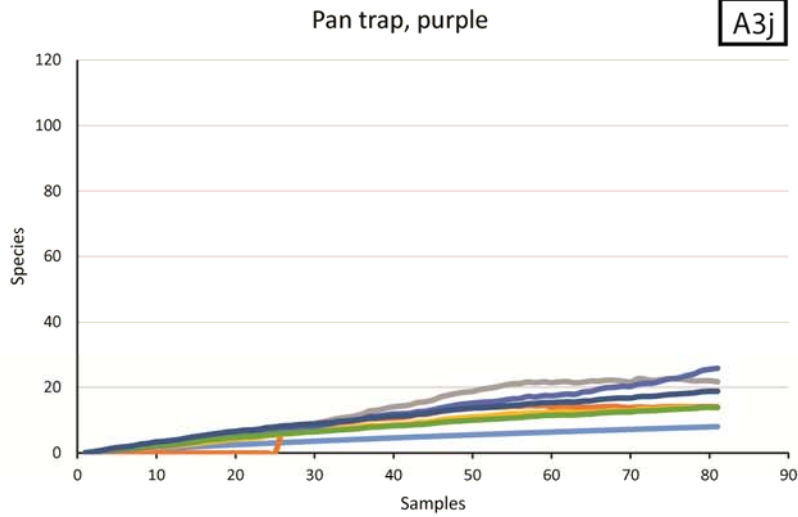
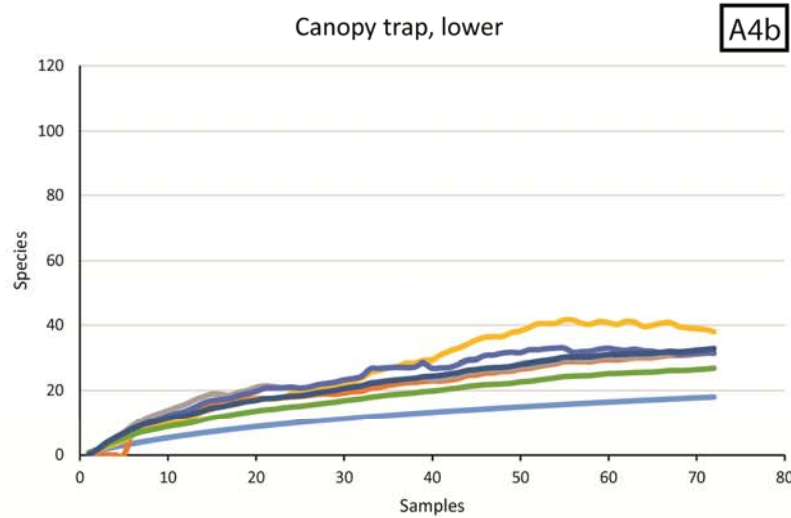
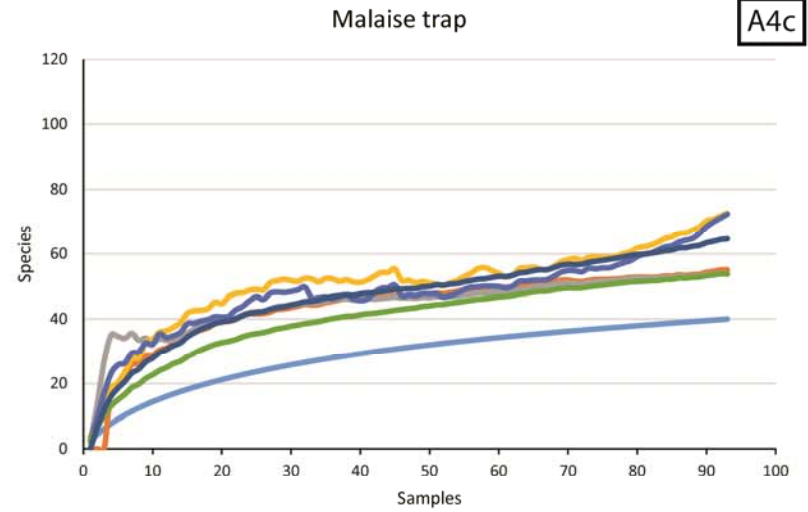
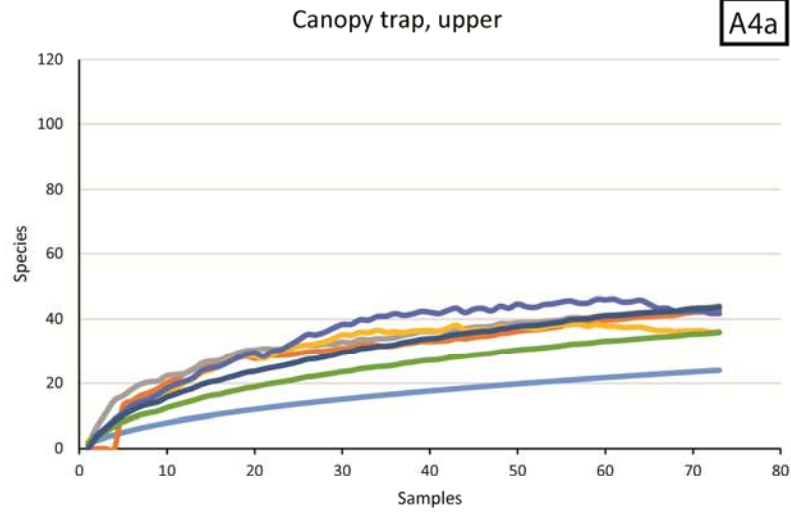
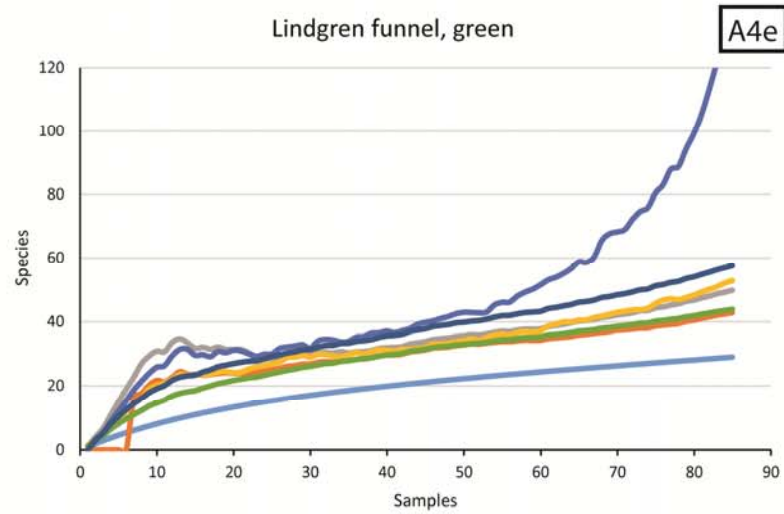
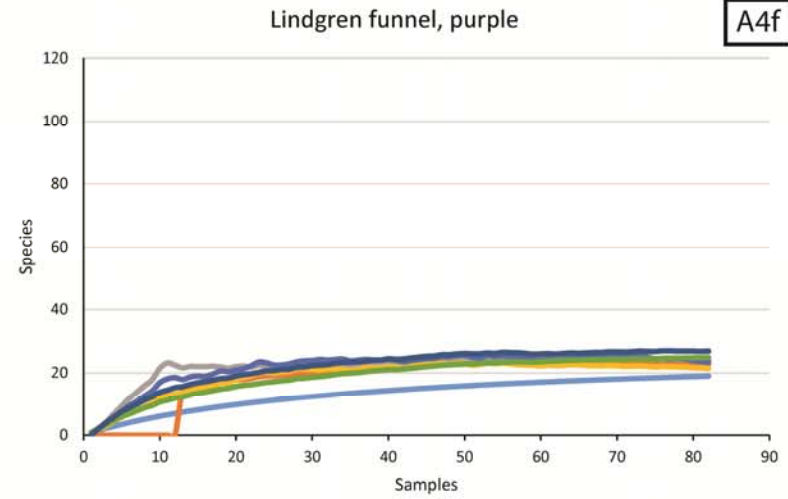
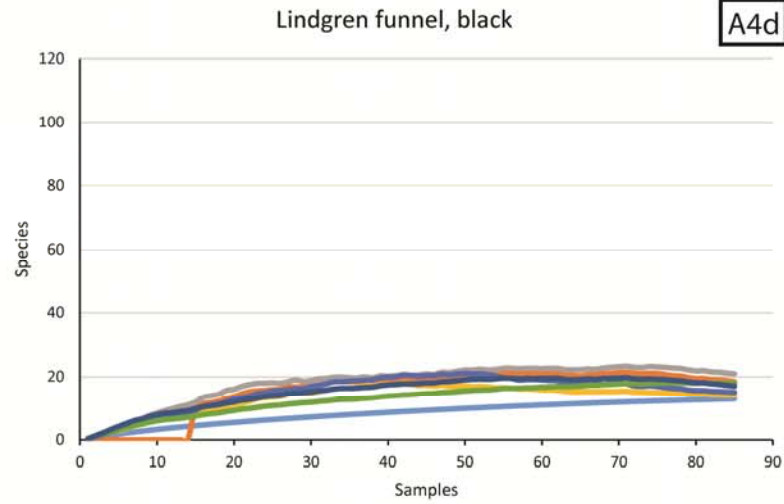


Figure A3 (cont.). Cerambycidae. See caption at the end of the figures for further explanation.



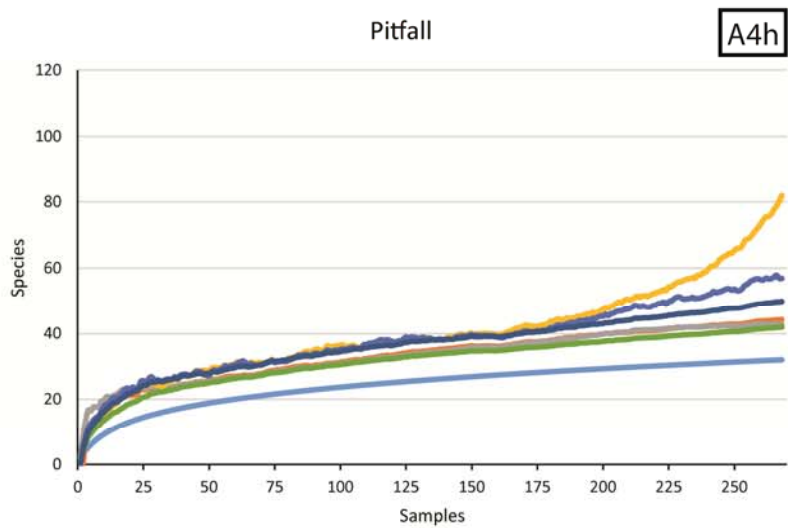
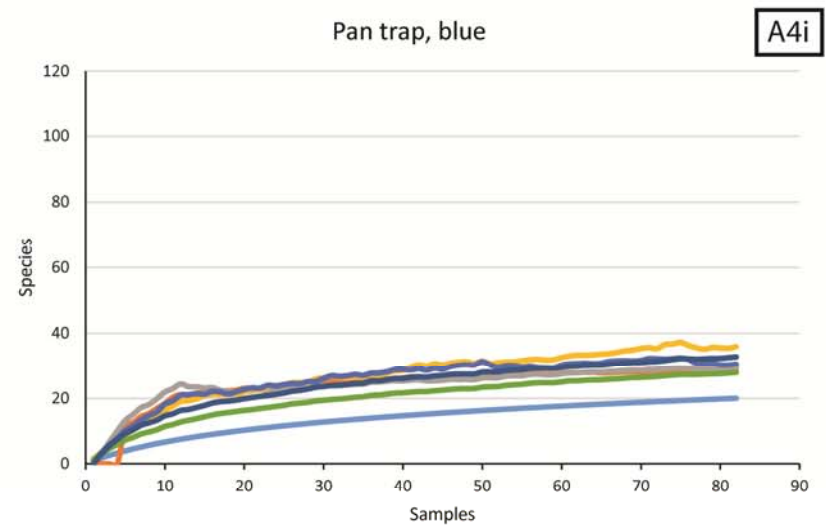
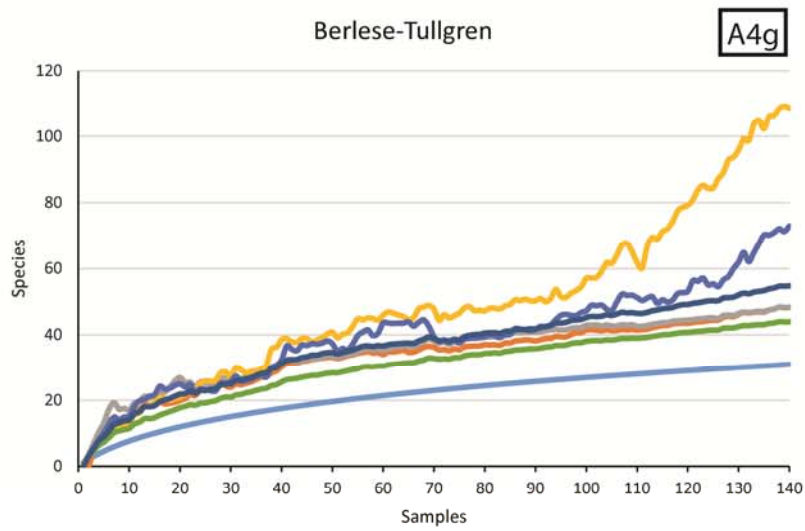
— S(est)
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 — Chao 1 Mean
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Figure A4. Curculionoidea. See caption at the end of the figures for further explanation.



— S(est)
 — ACE Mean
 — ICE Mean
 — Chao 1 Mean
 — Chao 2 Mean
 — Jack 1 Mean
 — Jack 2 Mean

Figure A4 (cont.). Curculionoidea. See caption at the end of the figures for further explanation.



— S(est)
 — ACE Mean
 — ICE Mean
 — Chao 1 Mean
 — Chao 2 Mean
 — Jack 1 Mean
 — Jack 2 Mean

Figure A4 (cont.). Curculionoidea. See caption at the end of the figures for further explanation.

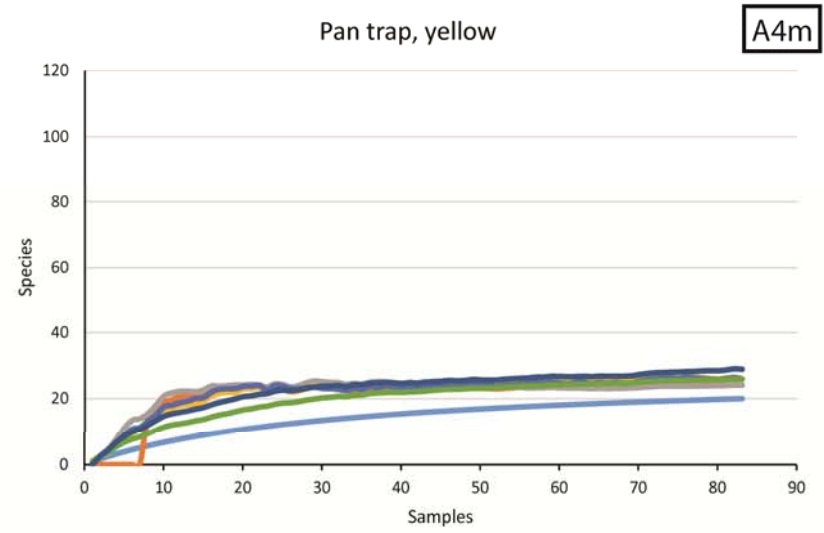
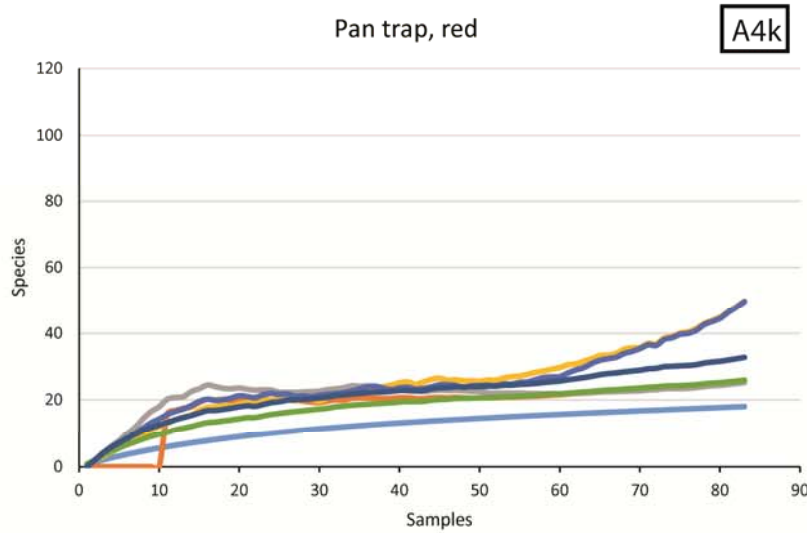
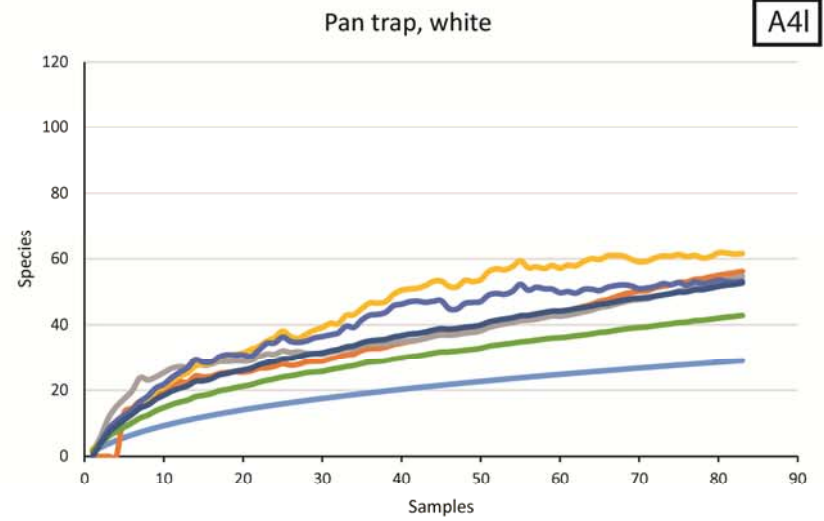
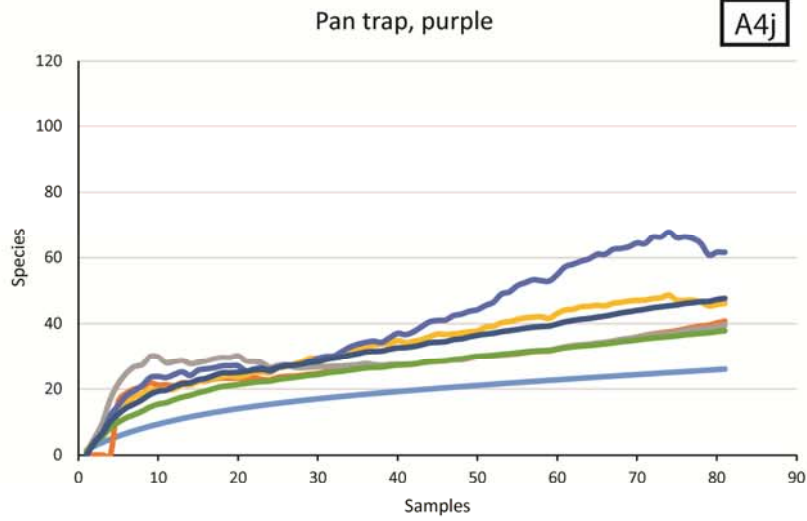


Figure A4 (cont.). Curculionoidea. See caption at the end of the figures for further explanation.

Figures A1–A4. Fig. A1. Buprestidae. **Fig. A2.** Carabidae. **Fig. A3.** Cerambycidae. **Fig. A4.** Curculionoidea. **a.** Canopy trap, upper collector. **b.** Canopy trap, lower collector. **c.** Malaise trap. **d.** Black Lindgren funnel trap. **e.** Green Lindgren funnel trap. **f.** Purple Lindgren funnel trap. **g.** Berlese-Tullgren extraction. **h.** Pitfall trap. **i.** Blue pan trap. **j.** Purple pan trap. **k.** Red pan trap. **l.** White pan trap. **m.** Yellow pan trap. Colors represent the same trap type throughout figures. The y-axis is standardized within a family but the x-axis is determined by the number of samples, which varies by trap type.

VI. Sampling terrestrial arthropod diversity: A case study

Abstract.

There is an increasing need to survey and document terrestrial arthropod assemblages as natural environments continue to be altered due to climate change, the introduction of invasive species, and habitat fragmentation and destruction. While the most effective survey methods have been studied for a few specific groups of arthropods, such as ants, few studies have attempted to determine the most effective methods for surveying the entire terrestrial arthropod assemblage at a site. In order to begin to answer this question, we surveyed a plot in the Boston Mountains of Arkansas using 70 traps of 12 trap types and Berlese-Tullgren extraction of leaf litter and identified 46,146 specimens representing 533 species from an array of higher taxa. We determined that Malaise and pitfall traps collected the most species and specimens and had the lowest similarity of the collection methods tested so were the best traps to deploy in tandem. We also estimated that 600 and 1000 samples were needed before the species accumulation curves for Malaise and pitfall traps, respectively, become asymptotic.

Introduction.

The Interior Highlands, which encompasses the Ouachita Mountains in west central Arkansas and eastern Oklahoma and the Ozarks in Missouri, northern Arkansas, and extreme southeast Kansas (Fig. 1), is an area of high biodiversity and endemism, with more than 200 species known only from the region (Allen 1990; Robison and Allen 1995; The Nature Conservancy, Ozarks Ecoregional Assessment Team 2003; Pringle and Witsell 2005; Zollner et al. 2005; Robison et al. 2008; McAllister *et al.* 2009;). However, with a few exceptions (e.g.,

Araneae, Carabidae, Pselaphinae, Formicidae: Table 1) many terrestrial arthropods have been historically understudied. This is underscored by recent work that has reported many species as new to Arkansas (e.g., Chordas *et al.* 2005; Chordas & Kovarik 2008a,b; Disney *et al.* 2010; Henry *et al.* 2010; MacGown *et al.* 2011; Bowles & Sites 2013; Tumblison 2013; Skvarla *et al.* 2014a [Chapter X], 2015 [Chapter VIII], *in press* [Chapter IX], *submitted* [Chapter IV]) and descriptions of new species from the state (Shelley *et al.* 2003; Cook & Lauder milk 2004; Tennessen 2004; Clark & Burke 2010; Hildebrandt & Maddison 2011). Additionally, establishing the composition of the terrestrial arthropod fauna of Arkansas and the Interior Highlands more generally is especially imperative in light of the many factors that continue to alter natural landscapes, such as global climate change (Thomas *et al.* 2004); the introduction of invasive species such as chestnut blight (*Cryphonectria parasitica* (Murrill) Barr), hemlock woolly adelgid (*Adelges tsugae* (Annand, 1928)), and emerald ash borer (*Agrilus planipennis* Fairmaire, 1888) that threaten keystone tree species, thereby altering forest composition (Ellison *et al.* 2005); and habitat fragmentation and destruction (Tilman *et al.* 1994; Brooks *et al.* 2002).

Recent efforts by the state of Arkansas, as laid out in the Arkansas Wildlife Action Plan (Anderson 2006) and implemented through the state wildlife grant system (Designing A Future For Arkansas Wildlife 2015), include an effort to survey, confirm the continued existence and known range, and locate additional populations of imperiled arthropods in the state. The list of at-risk arthropods contains a diverse array of terrestrial taxa such as butterflies, beetles, and true bugs for which multiple collection methods are needed. However, while other studies have employed a variety of techniques to collect arthropod biodiversity (e.g., Hammond 1990; Hammond *et al.* 1997), compared or discussed different collecting techniques for a target taxon (e.g., Formicidae: Agosti & Alonso 2000; Araneae: Duffey 1972), or compared trap catch at

higher taxonomic levels (i.e., order or family) (e.g., Hosking 1979; Juliet 1963), the authors are aware of no study that examined general arthropod biodiversity at the species level collected by a numerous techniques in order to determine the most efficient combination of methods that collect the widest array of species.

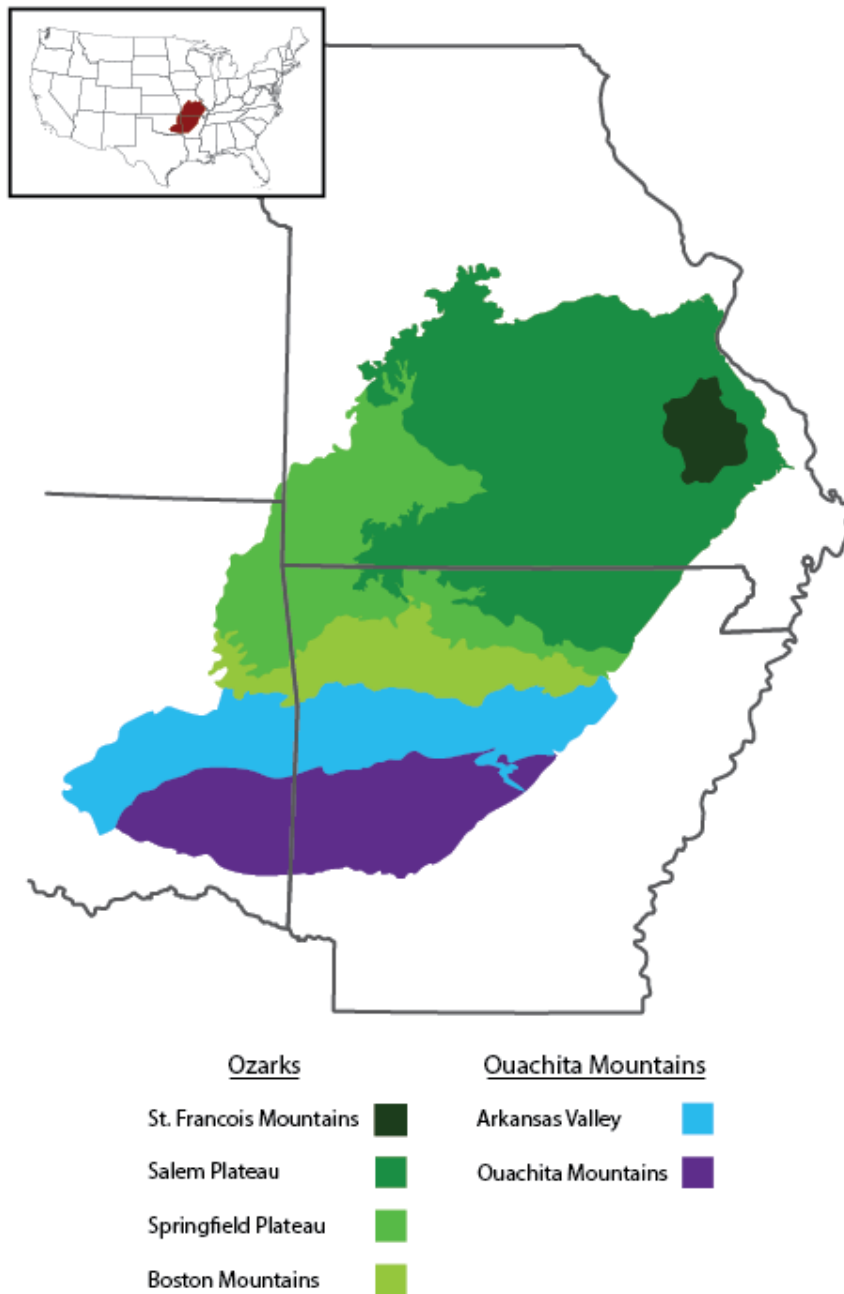


Figure 1. Geographic regions of the Interior Highlands. Modified from Skvarla *et al.* (submitted).

Taxon	Select references
Araneae	Dorris 1968, 1969, 1985, 1989, 1991; Dorris <i>et al.</i> 1995
Carabidae	Hemenway & Whitcomb 1967; Allen 1973; Allen & Carlton 1988
Staphylinidae, Pselaphinae	Carlton & Allen 1989; Carlton & Cox 1990; Carlton 1995 Warren & Rouse 1969, 1980; General & Thompson 2007, 2008,
Formicidae	2009; MacGown <i>et al.</i> 2011

Table 1. Examples of terrestrial arthropods that are well-sampled within the Interior Highlands.

While it would be nearly impossible to identify the hundreds of thousands to tens of millions of specimens collected in an extensive survey of even a small field site, we present here the results of a single year study that compared the catch of seventy traps comprising twelve trap types and Berlese-Tullgren extraction in an attempt to begin to answer the question of what is the most efficient combination of collection techniques for sampling the most arthropod biodiversity.

Materials and Methods.

The field site where this study was conducted and statistical methods used to analyze data were covered in detail by Skvarla et al. (submitted) (=Chapter IV) and in Chapter V, but are restated and summarized here for convenience.

Field site and collecting regime

A 4 ha plot was established at in the Boston Mountains of Arkansas at Steel Creek along the Buffalo National River in Newton County (centered at approximately N 36°02.269', W 93°20.434'). The site was dominated by mature second-growth oak (*Quercus*) and hickory (*Carya*), with other species such as American beech (*Fagus grandifolia*) and eastern red cedar (*Juniperus virginiana*) being abundant.

Five Malaise traps (MegaView Science Co., Ltd., Taichung, Taiwan) and twenty-five pan traps (five of each color: blue, purple, red, yellow, white) which were randomly arranged under

the Malaise traps (one of each color) so as to also act as intercept traps; four SLAM (Sea, Land, and Air Malaise) traps (MegaView Science Co., Ltd., Taichung, Taiwan) with top and bottom collectors; fifteen Lindgren multi-funnel traps (ChemTich International, S.A., Heredia, Costa Rica) (five of each color: black, green, purple); and seventeen pitfall trap sets. Sixteen of the seventeen pitfall sets were placed in two transects of sets spaced every five meters centered on two Malaise traps, while the final set was placed away from other traps. Additionally, ten leaf litter samples were collected for Berlese extraction when traps were serviced.

Pitfall traps were made using plastic soup containers based on a modified design proposed by Nordlander (1987). Each pitfall trap set was made by burying a single cup on either side of a 30.5 cm x 15.5 cm aluminum fence; trap catch from both cups was combined and treated as a single sample.

Traps were placed non-randomly within the plot in order to maximize the efficiency of each trap, although an attempt was made to evenly space similar traps in order to decrease the chance of interference between traps. Malaise traps were placed in perceived flight paths. SLAM and Lindgren funnel traps were suspended from the branches of large trees 4–10 meters above the ground in the lower canopy. Berlese-Tullgren samples were collected from a variety of habitats, including leaf litter, moss, tree holes, and bark from fallen, partially decayed trees. Litter was processed with a litter reducer until approximately one gallon of processed litter was collected and processed for four to seven days until the litter was dry throughout using modified Berlese-Tullgren funnels.

All traps were set by 13 March 2013, except Lindgren funnels, which were set on 1 April 2013. Traps were serviced approximately every two weeks (14 days \pm 3 days). The final collection of pitfall traps and pan traps occurred on 6 November 2013 and the final collection of

Malaise, SLAM, and Lindgren funnel traps occurred on 4 December 2013. Berlese-Tullgren samples from 13 April, 15 May, 28 June and 6 November were not taken or were lost. Pitfall sets were lost on 13 April (one set), 15 May (one set), 28 June (four sets), and 17 July (five sets). In total, 1311 samples were collected.

Propylene glycol (Peak RV & Marine Antifreeze) (Old World Industries, LLC, Northbrook, IL) was used as the preservative in all traps as it is non-toxic and generally preserves specimens well (Skvarla *et al.* 2014b [Chapter II]). Trap catch was sieved in the field and stored in Whirl-Pak bags (Nasco, Fort Atkinson, WI) in 90% ethanol until sorting.

Sample processing and identification

Samples were coarse-sorted using a Leica MZ16 stereomicroscope illuminated with a Leica KL1500 LCD light source and a Wild M38 stereomicroscope illuminated with an Applied Scientific Devices Corp. Eco-light 20 fiber optic light source. After sorting, specimens were stored individually or by family in 70% ethanol in 2 mL microtubes. Hard-bodied specimens such as beetles were pinned or pointed as appropriate.

Specimens were identified with the use of published keys (Table 2) (see Chapter IV for references pertaining to Buprestidae, Carabidae, Cerambycidae, and Curculionoidea). Pompilidae were identified by Clint Trammel (University of Arkansas). In some cases, difficult to key specimens were photographed through the eye piece of the stereomicroscope using the camera on an HTC Droid Incredible 4G LTE cell phone or Samsung Galaxy S5 cell phone; the photographs were uploaded to Bugguide (Iowa State University 2015) and identifications were proposed by Bugguide members. Proposed identifications were then double checked using published sources and either confirmed or corrected on the website.

Class	Order	Family	Genus	Reference
General arthropods identification				Robison & Allen 1995; Tripplehorn & Johnson 2005; SCAN 2014
Arachnida	Araneae			Dorris 1985; Dorris 1989; Dorris et al. 1995; Ubick 2005; Platnick 2014
Arachnida	Araneae	Agelenidae		Bennett & Ubick 2005
Arachnida	Araneae	Agelenidae	<i>Wadotes</i>	Muma 1947
Arachnida	Araneae	Agelenidae	<i>Agelenopsis</i>	Whitman <i>et al.</i> 2015
Arachnida	Araneae	Amaurobiidae		Leech 1972; Ubick 2005b
Arachnida	Araneae	Anyphaenidae		Richman & Ubick 2005a
Arachnida	Araneae	Anyphaenidae	<i>Anyphaena</i>	Platnick 1974
Arachnida	Araneae	Araneidae		Levi 2005a
Arachnida	Araneae	Araneidae	<i>Hypsosinga</i>	Levi 1971
Arachnida	Araneae	Araneidae	<i>Ocrepeira</i>	Levi 1976
Arachnida	Araneae	Atypidae		Gertsch & Platnick 1980
Arachnida	Araneae	Clubionidae		Edwards 1958; Dondale & Redner 1982; Richman & Ubick 2005b
Arachnida	Araneae	Corinnidae		Ubick & Richman 2005
Arachnida	Araneae	Corinnidae	<i>Castianeria</i>	Reiskind 1969
Arachnida	Araneae	Cybaeidae		Bennett 2005a
Arachnida	Araneae	Cyrtacheniidae	<i>Myrmekiaphila</i>	Bond & Platnick 2007
Arachnida	Araneae	Cyrtacheniidae		Bond <i>et al.</i> 2012
Arachnida	Araneae	Dictynidae		Bennett 2005b
Arachnida	Araneae	Dictynidae	<i>Cicurina</i>	Chamberlin & Ivie 1940; Paquin & Dupérré 2009
Arachnida	Araneae	Hahniidae		Opbell & Beatty 1976; Bennett 2005c
Arachnida	Araneae	Gnaphosidae		Ubick 2005c
Arachnida	Araneae	Gnaphosidae	<i>Callilepis</i>	Platnick 1975
Arachnida	Araneae	Gnaphosidae	<i>Drassodes</i>	Platnick & Shadab 1976a
Arachnida	Araneae	Gnaphosidae	<i>Gnaphosa</i>	Platnick & Shadab 1975a
Arachnida	Araneae	Gnaphosidae	<i>Haplodrassus</i>	Platnick & Shadab 1975b
Arachnida	Araneae	Gnaphosidae	<i>Micaria</i>	Platnick & Shadab 1988
Arachnida	Araneae	Gnaphosidae	<i>Rachodrassus</i>	Platnick & Shadab 1976b
Arachnida	Araneae	Gnaphosidae	<i>Tivodrassus</i>	Platnick & Shadab 1976a
Arachnida	Araneae	Gnaphosidae	<i>Scopodes</i>	Platnick & Shadab 1976b
Arachnida	Araneae	Gnaphosidae	<i>Sosticus</i>	Platnick & Shadab 1976b

Table 2. Keys used to identify specimens.

Class	Order	Family	Genus	Reference
Arachnida	Araneae	Lycosidae		Gertsch & Wallace 1935; Gertsch & Wallace 1936; Dondale 2005
Arachnida	Araneae	Lycosidae	<i>Acantholycosa</i>	Vogel 2004
Arachnida	Araneae	Lycosidae	<i>Allocosa</i>	Dondale & Redner 1983a
Arachnida	Araneae	Lycosidae	<i>Arctosa</i>	Dondale & Redner 1983b
Arachnida	Araneae	Lycosidae	<i>Camptocosa</i>	Dondale <i>et al.</i> 2005
Arachnida	Araneae	Lycosidae	<i>Gladicosa</i>	Brady 1986
Arachnida	Araneae	Lycosidae	<i>Lycosa</i>	Wallace 1942
Arachnida	Araneae	Lycosidae	<i>Pardosa</i>	Vogel 2004
Arachnida	Araneae	Lycosidae	<i>Pirata</i>	Wallace & Exline 1978
Arachnida	Araneae	Lycosidae	<i>Rabidosa</i>	Brady & McKinley 1994
Arachnida	Araneae	Lycosidae	<i>Schizocosa</i>	Dondale & Redner 1978
Arachnida	Araneae	Lycosidae	<i>Tigrosa</i>	Brady 2012
Arachnida	Araneae	Lycosidae	<i>Trochosa</i>	Brady 1980
Arachnida	Araneae	Mimetidae		Lew & Mott 2005; Harms & Dunlop 2009
Arachnida	Araneae	Mimetidae	<i>Mimetus</i>	Mott 1989
Arachnida	Araneae	Mysmenidae		Lopardo & Coddington 2005
Arachnida	Araneae	Oxyopidae		Brady 1963
Arachnida	Araneae	Philodromidae		Dondale 2005b
Arachnida	Araneae	Philodromidae	<i>Ebo</i>	Sauer & Platnick 1970
Arachnida	Araneae	Philodromidae	<i>Philodromus</i>	Dondale & Redner 1976
Arachnida	Araneae	Phrurolithidae		Chamberlin & Gertsch 1930; Chamberlin & Ivie 1935; Ivie & Barrows 1935; Chamberline & Ivie 1944; Ubick & Richman 2005
Arachnida	Araneae	Phrurolithidae	<i>Phruronellus</i>	Chamberlin 1921
Arachnida	Araneae	Phrurolithidae	<i>Scotinella</i>	Penniman 1985
Arachnida	Araneae	Salticidae		Richman et al. 2005; Richman et al. 2012
Arachnida	Araneae	Salticidae	<i>Habronattus</i>	Griswold 1987
Arachnida	Araneae	Salticidae	<i>Maevia</i>	Barnes 1955
Arachnida	Araneae	Salticidae	<i>Naphrys</i>	Richman 1981; Edwards 2002
Arachnida	Araneae	Salticidae	<i>Peckhamia</i>	Peckham & Peckham 1909
Arachnida	Araneae	Salticidae	<i>Pelegrina</i>	Maddison 1996
Arachnida	Araneae	Salticidae	<i>Phidippus</i>	Edwards 2004
Arachnida	Araneae	Salticidae	<i>Synageles</i>	Cutler 1987

Table 2 (cont.). Keys used to identify specimens.

Class	Order	Family	Genus	Reference
Arachnida	Araneae	Salticidae	<i>Thiodina</i>	Richman & Vetter 2004
Arachnida	Araneae	Theridiidae		Levi 2005b
Arachnida	Araneae	Theridiidae	<i>Argyrodes/</i> <i>Neospintharus</i>	Exline & Levi 1962
Arachnida	Araneae	Theridiidae	<i>Crustulina</i>	Levi 1957
Arachnida	Araneae	Theridiidae	<i>Steatoda</i>	Levi 1957
Arachnida	Araneae	Thomisidae		Gertsch 1939; Dondale & Redner 1978; Cokendolpher et al. 1979; Dondale 2005d
Arachnida	Araneae	Thomisidae	<i>Coriachne</i>	Bowling & Sauer 1975
Arachnida	Araneae	Thomisidae	<i>Coriachne</i>	Gertsch 1953
Arachnida	Araneae	Thomisidae	<i>Oxyptila</i>	Gertsch 1953
Arachnida	Araneae	Thomisidae	<i>Xysticus</i>	Gertsch 1953
Arachnida	Araneae	Titanoecidae		Cutler 2005
Arachnida	Araneae	Titanoecidae	<i>Titanoeca</i>	Leech 1972
Arachnida	Araneae	Trachelidae		Ubick & Richman 2005
Arachnida	Araneae	Trachelidae	<i>Meriola</i>	Platnick & Shadab 1974
Arachnida	Mesostigmata	Ixodidae		Clifford et al. 1961; Lancaster 1973
Arachnida	Opiliones	Phalangodidae		Clarence & Goodnight 1942
Insecta	Blattodea	Rhinotermitidae	<i>Reticulitermes</i>	Lim & Forscler 2012
Insecta	Dermaptera			Hoffman 1987
Insecta	Diptera			McAlpine <i>et al.</i> 1981
Insecta	Diptera	Anisopodidae	<i>Sylvicola</i>	Pratt & Pratt 1980
Insecta	Diptera	Oestridae	<i>Cephenemyia</i>	Bennett & Sabrosky 1972; Taber & Fleenor 2004; Fleenor & Tabor 2007
Insecta	Diptera	Oestridae	<i>Cuterebra</i>	Sabrosky 1986
Insecta	Diptera	Scathophagidae		James 1950
Insecta	Diptera	Stratiomyiidae		McFadden 1972; Williston 1885
Insecta	Diptera	Stratiomyiidae	<i>Ptecticus</i>	McFadden 1971
Insecta	Diptera	Tabanidae		Carlton & Lancaster 1995
Insecta	Diptera	Xylophagidae	<i>Rachicerus</i>	Webb 1984
Insecta	Hemiptera	Caliscelidae		Doering 1939
Insecta	Hemiptera	Lygaeidae		Slater & Baranowski 1990
Insecta	Hymenoptera	Aulacidae		Smith 1996

Table 2 (cont.). Keys used to identify specimens.

Class	Order	Family	Genus	Reference
Insecta	Hymenoptera	Formicidae		Ross et al. 1971; MacGown 2003; Coover 2005; Fisher & Cover 2007
Insecta	Hymenoptera	Formicidae	<i>Solenopsis</i>	Pacheco 2007
Insecta	Hymenoptera	Orussidae		Middlekauff 1983
Insecta	Hymenoptera	Siricidae		Schiff <i>et al.</i> 2006
Insecta	Hymenoptera	Vespidae		Akre <i>et al.</i> 1980
Insecta	Mecoptera			Thornhill & Johnson 1974; Cheung et al. 1996; Robison et al. 1997
Insecta	Mecoptera			Webb <i>et al.</i> 1975
Insecta	Mecoptera	Panorpidae	<i>Panorpa</i>	Byers 1993; Capinera et al. 2004
Insecta	Orthoptera	Myrmecophilidae		Capinera <i>et al.</i> 2005; MacGown & Hill 2006
Malacostraca	Isopoda			Muchmore 1990
Myriapoda	Diplopoda	Polyxenidae	<i>Polyxenus</i>	Kincaid 1898; Pierce 1940; Kane 1981; Chamberlin 1922

Table 2 (cont.). Keys used to identify specimens.

One to five voucher specimens of each species were retained in the Dowling Lab Collection at the University of Arkansas while the remaining species were deposited in the University of Arkansas Arthropod Museum (UAAM) and several private collections. with the

Taxa selection

Taxa were chosen for identification and inclusion in the statistical analysis for the following of reasons: Formicidae was selected because they are often used in biodiversity assessment (Alonso 2000; Underwood & Fisher 2006; Maleque *et al.* 2009) studies and the lead author is familiar with ant identification. Carabidae, Araneae, and Isopoda were chosen because they are bioindicators (Paoletti & Hassall 1999; Buddle *et al.* 2000; Ranio & Niemelä 2003; Oxbrough *et al.* 2005; Pearce & Venier 2006; Maleque *et al.* 2009; Avgin & Luff 2009) and have a wealth of material such as keys and checklists to aid in the identification of North

American species. Cerambycidae were included because they are diverse and relatively easily identified, and are used as bioindicators in forests (Maeto & Makihara 1999; Maeto *et al.* 2002; Makino *et al.* 2007). Curculionoidea were included because they are abundant and diverse. Buprestidae and Parasitengona were selected because an expert was willing to identify them or teach the lead author how to identify them. Finally, the remaining species (e.g., *Merope tuber*, *Polistes* spp., *Orussus minutus*, &c.) were included because they are distinctive and could be readily identified to species-level as samples were coarse-sorted. While this may introduce some bias towards large, showy species, an effort was made to include smaller distinctive species (e.g., Ixodidae spp., *Lygistorrhina sanctaecatharinae*, *Polyxenus largurus*) to counteract this.

Statistical analysis

Specimen abundance per trap per date was recorded in Microsoft Excel (Microsoft 2013). For each family analyzed, the following procedures were performed.

The effect of trap type on the number of species and specimens was analyzed by performing a one-way analysis of variance (ANOVA) test ($\alpha = 0.05$) in Excel. Due to uneven trapping effort and because some traps were randomly lost due during the study, we compared the average number of species and specimens collected per trap type per date.

If a significant difference was detected, the means were separated using a Tukey-Kramer test ($\alpha = 0.05$) performed in Excel using the Real Statistics Resource Pack add-in (Zaiontz 2015). We chose to use ANOVA and Tukey-Kramer rather than their non-parametric equivalents as both tests are relatively robust with respect to violations of the normality assumption (Kirk 1995; Samuels & Witmer 2003) and easily performed within Excel.

EstimateS (Colwell 2013) was used to calculate the following species accumulation estimators, with abbreviations used in graphs noted parenthetically, for each trap type using all samples collected per trap type: abundance coverage-based estimator of species richness (ACE); incidence coverage-based estimator of species richness (ICE); Chao 1 richness estimator (Chao1); Chao 2 richness estimator (Chao2); first-order Jackknife richness estimator (Jack1); second-order Jackknife richness estimator (Jack2) (see Gotelli & Colwell [2010] for a synopsis of each estimator). Additionally, the sample-based rarefaction curve ($S(\text{est})$), which is the expected number of species in t pooled samples given the reference sample, was also calculated. EstimateS was run on default settings except that classic Chao1 and Chao2 estimators were used instead of the default bias-corrected Chao1 and Chao2 as suggested by the program. One hundred randomizations of sample order were performed. As the various estimators generally calculated similar trends, we reported only Chao1 estimators in a single graph and included graphs of all of the estimators in Appendix I. Because uneven sampling effort between trap types did not allow the number of species collected by each trap type to be directly compared, EstimateS was used to extrapolate the number of samples per trap type to 1000 samples, at which point the number of estimated species collected per trap type were compared. Samples were randomized across traps and dates within a trap type. Error bars were excluded from accumulation and rarefaction graphs in order to enhance clarity.

Species similarity between trap types and collecting dates were investigated by calculating shared species indices using EstimateS. EstimateS output was organized in Excel and final graphs were constructed in Adobe Illustrator (Adobe 2012). EstimateS calculates a number of different shared species estimators; herein we report the Sørensen similarity index, an incidence-based (i.e., presence/absence) index, and Chao's Sørensen similarity index, an

abundance-based index (Chao et al. 2005). These indices indicate the similarity of the compared samples, which ranges between 0 and 1 and indicate no to complete similarity. The statistical significance of similarity cannot be determined from these indices; therefore, when discussing the estimated similarity, we used the terms low (0–0.24), medium (0.25–0.49), high (0.50–0.74) and very high (0.75–1.0). Graphs are color-coded to reflect these categories; dates for which no samples of a given trap type were collected are indicated by a dash (-).

Shared species indices for trap types were calculated based on the total number of specimens per species collected per trap type. Shared species indices for collection dates were calculated based on the total specimens collected per species per date.

Results.

We identified 46,146 specimens representing 533 species; 15 species, 10 of which were parasitengone mites, are putatively undescribed, at least 36 are new state records for Arkansas (the status of some species could not be confirmed so this is likely an underestimate), and 13 are non-native introduced species (Appendix II). Formicidae represented 60.7% of the specimens identified (28,032 specimens) but only represented 13.9% of the species collected (74 species); because Formicidae were numerically dominant in the number of specimens collected but only represented a small proportion of the species collected, all statistics were performed including and excluding Formicidae in the event their inclusion skewed results. Reported results assume the inclusion of Formicidae unless otherwise specified.

Pompilidae were deposited in the C. Trammel collection; *Cicurina* (Dictynidae) were sent to Pierre Paquin; the following specimens were sent to Peter Messer for identification confirmation and are deposited in the P. W. Messer collection: *Agonum crenulatum* (MS 13-

0529-072, #136215), *Agonum ferreum* (MS 13-0612-022, #139663), *Cicindela rufiventris* (MS 13-0717-001, #134492), *Cyclotrachelus incisus* (MS 13-0413-023, #139591; MS 13-0413-019, #139592; MS 13-0413-006, #139594; MS 13-1008-075, #139596), *Cyclotrachelus parasodalis* (MS 13-0430-019, #131983; MS 13-0529-037, #135057; MS 13-1106-002, #138280), *Cyclotrachelus torvus* (MS 13-0529-066, #135053), *Pterostichus punctiventris* (MS 13-0401-018, #135065; MS 13-1023-021, # 136216), *Rhadine ozarkensis* (MS 13-0925-027, #134547), *Scaphinotus fissicollis* (MS 13-1106-037, #137830), *Selenophorus ellipticus* (MS 13-0925-005, #136223), *Selenophorus opalinus* (MS 13-0813-034, # 136217), *Trichotichus autumnalis* (MS 13-0730-005, #136226), *Trichotichnus vulpeculus* (MS 13-0911-027, #136218).

The following issues with identification should be noted: *Abacion* (Diplopoda: Abacionidae) specimens can only be identified to species based on the shape of the male gonopods. Two species, *A. texense* and *A. tessellatum*, were identified at the field site (a third species, *A. wilhelminae*, is known from Arkansas but was not found at the site and is apparently restricted to Rich Mountain in Polk County [Shelley *et al.* 2003,]). The majority of males (147/150 specimens examined) were identified as *A. texense* so immature and female *Abacion* were assigned that species. While it is probable that a small percentage of immature and female *Abacion* represent *A. tessellatum* and not *A. texense*, it is unlikely that their inclusion with *A. texense* will alter statistical analysis and excluding all immatures and females would certainly reduce statistical power.

Multiple species of *Polyxenus* (Diplopoda: Polyxenidae) have been identified from North America but the only revision of the genus synonymized them under *P. lagurus* (Kane 1981). Unfortunately, the revision is an unpublished Ph.D. dissertation and not recognized by ICZN.

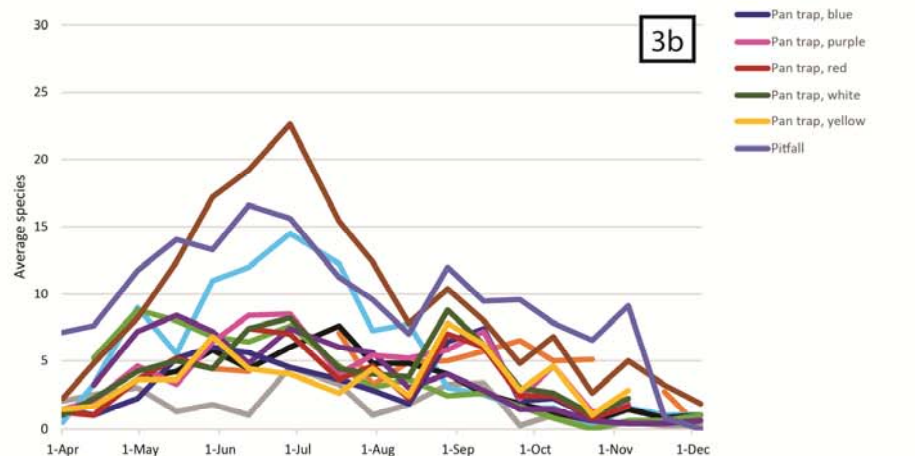
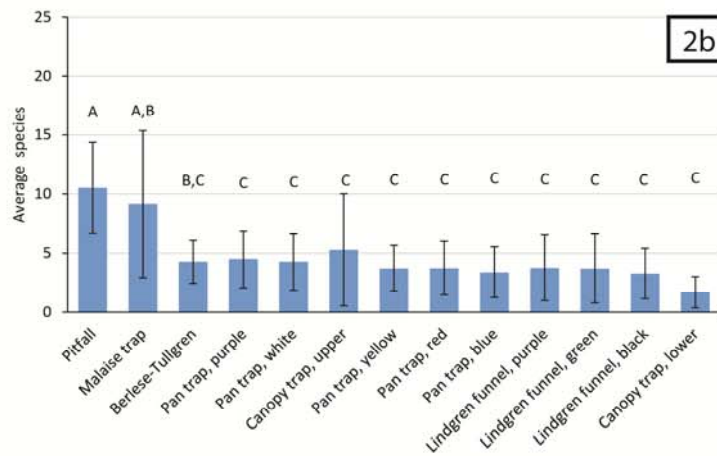
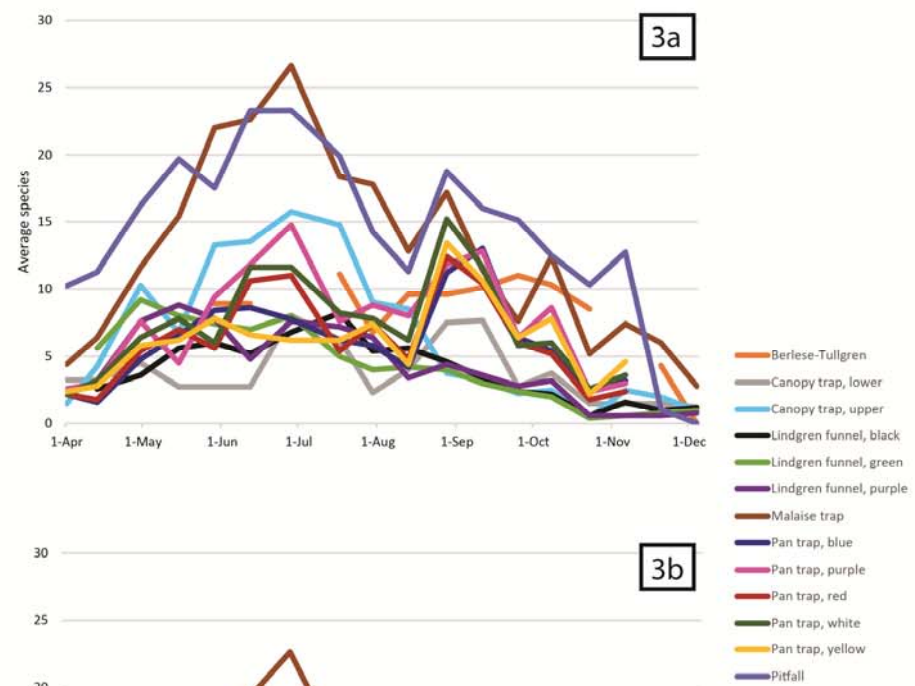
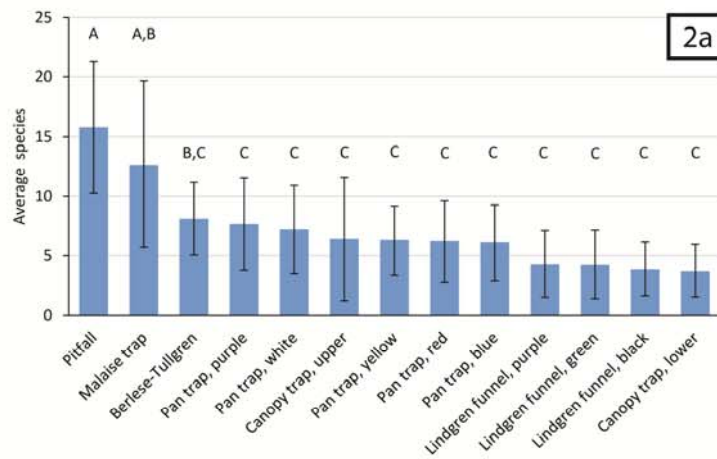
Even so, the characters used to separate the various “species” are dubious and identification of the “species” impossible. I therefore follow Kane (1981).

Tmarus (Araneae: Thomisidae) is represented by six species in North America, three of which may occur in Arkansas: *T. floridensis*, *T. rubromaculatus*, and *T. angulatus* (Gertsch 1939, Dondale & Redner 1978). As with many other spiders, only adults are identifiable beyond genus as species identification relies on genital morphology. *Tmarus floridensis* is known from Florida, Georgia, Mississippi, Louisiana, and Texas (Gertsch 1939); while it may be found in southern Arkansas, it is unlikely that it is present in northwestern Arkansas. *Tmarus rubromaculatus* occurs in the southeastern United States and Ohio, but is uncommonly encountered (Gertsch 1939); it has not been recorded from Arkansas but may eventually be found. All adults (n=2) collected in this study were identified as *T. angulatus*. Given the low number of species possible, all immature *Tmarus* (n=3) were assigned to *T. angulatus* for statistical analysis.

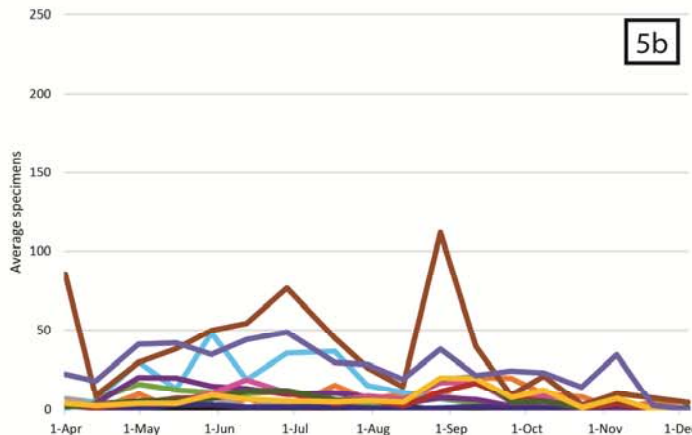
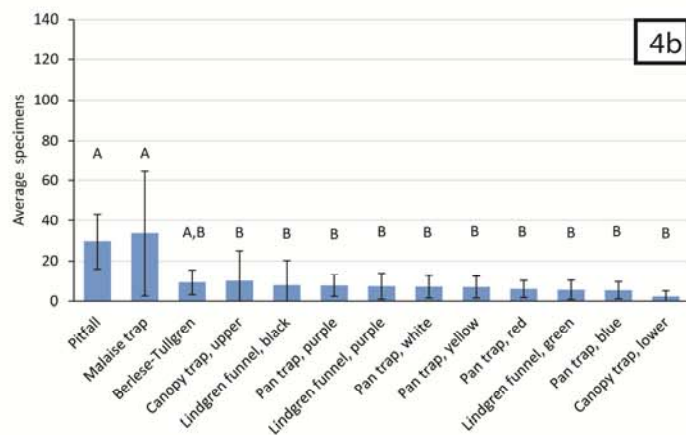
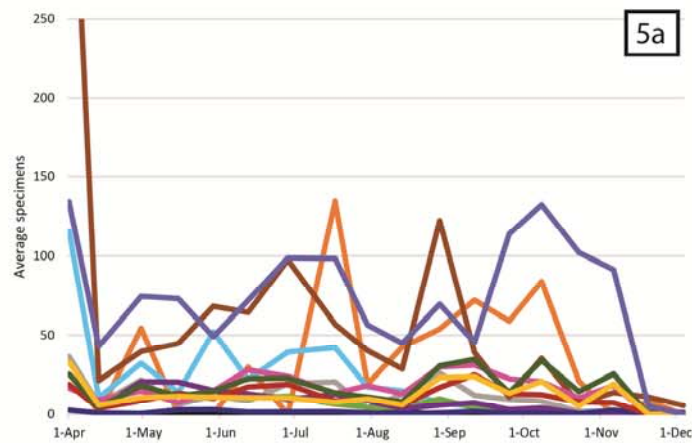
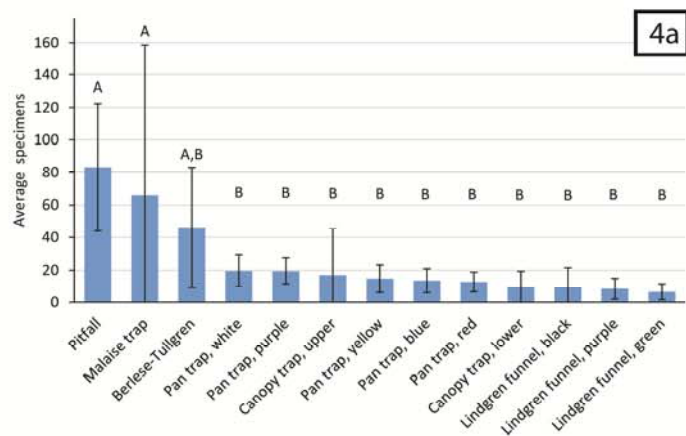
Phrurotimpus (Araneae: Phrurolithidae) consists of 16 described species and at least as many undescribed species. Two described species and two undescribed morphotypes, one represented by females and one by males, were collected. The two undescribed morphotypes were treated as a single species in the statistical analysis – *Phrurotimpus* sp. 3 – as it is unlikely that two undescribed species occur at the site, one of which was represented only by females and the other only by males.

Pitfall and Malaise traps collected the most species (Figs. 2, 3) and specimens (Figs. 4, 5) of the targeted taxa. Twenty five percent of the species (135) were represented by a single specimen and 51% percent of the species (274) were represented by five or fewer specimens, while 3.3% of the species (18) were represented by more than 500 specimens (Fig. 6).

There was a significant ($p < 0.05$) effect of trap type on the number of species collected for the twelve trap types ($F(12,203) = 4.61$, $p = 1.60 \times 10^{-18}$). The mean number of species collected by pitfall ($M = 14.91$, $SD = 5.51$) were significantly ($p < 0.05$, Tukey-Kramer) different from all other traps except Malaise traps, Malaise traps ($M = 12.67$, $SD = 6.98$) were significantly different from all other traps except Berlese-Tullgren extraction ($M = 8.12$, $SD = 3.07$) and that Berlese-Tullgren was not significantly different from all other traps: lower canopy trap ($M = 3.74$, $SD = 2.20$), upper canopy trap ($M = 6.40$, $SD = 5.12$), black Lindgren funnel trap ($M = 3.89$, $SD = 2.24$), green Lindgren funnel trap ($M = 4.27$, $SD = 2.89$), purple Lindgren funnel trap ($M = 4.31$, $SD = 2.81$), blue pan trap ($M = 6.10$, $SD = 3.18$), purple pan trap ($M = 7.69$, $SD = 3.87$), red pan trap ($M = 6.12$, $SD = 3.41$), white pan trap ($M = 7.24$, $SD = 3.70$), yellow pan trap ($M = 6.3$, $SD = 2.90$) ($p > 0.05$, Tukey-Kramer) (Figs. 2a,b). Exclusion of Formicidae did not significantly alter the results of the ANOVA or Tukey-Kramer tests, although it did reduce the standard deviation slightly across all traps.



Figures 2, 3. Average number of species collected per trap. **Fig. 2.** Average number of species/trap. Error bars represent standard deviation; letters represent mean separation as determined by Tukey-Kramer test. **Fig. 3.** Average number of species/trap/date. Standard deviations are omitted for clarity. **a.** All taxa including Formicidae. **b.** All taxa excluding Formicidae.



Figures 4,5. Average number of specimens collected per trap. **Fig. 4.** Average number of specimens/trap. Error bars represent standard deviation; letters represent mean separation as determined by Tukey-Kramer test. **Fig. 5.** Average number of specimens /trap/date. Standard deviations are omitted for clarity. **a.** Formicidae included. **b.** Formicidae excluded.

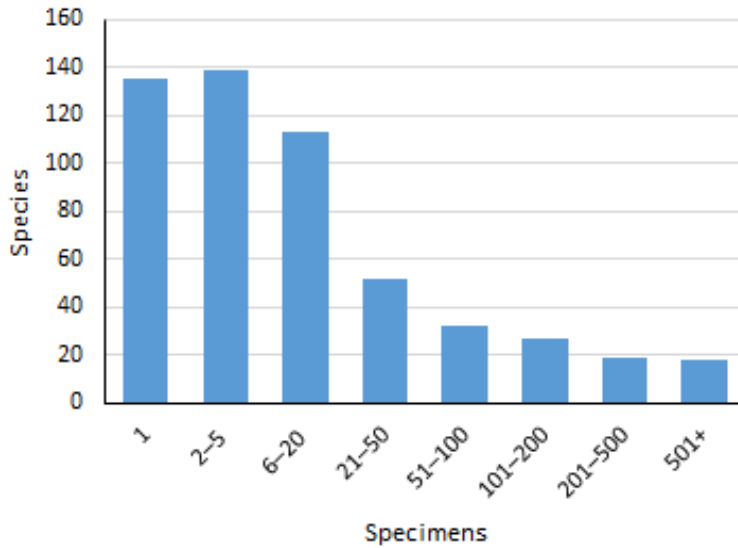


Figure 6. Total number of specimens/species collected across all traps.

There was a significant ($p < 0.05$) effect of trap type on the number of specimens collected for the thirteen trap types ($F(12,203) = 3.79$, $p = 1.3 \times 10^{-13}$). The mean number of specimens collected by pitfall ($M = 79.93$, $SD = 35.40$) and Malaise traps ($M = 64.46$, $SD = 92.86$) was significantly ($p < 0.05$, Tukey-Kramer) different from all other traps except Berlese-Tullgren extraction ($M = 44.05$, $SD = 36.92$) and that Berlese-Tullgren was not significantly different from all other trap types: lower canopy trap ($M = 12.01$, $SD = 9.90$), upper canopy trap ($M = 21.54$, $SD = 28.31$), black Lindgren funnel trap ($M = 9.07$, $SD = 12.45$), green Lindgren funnel trap ($M = 6.47$, $SD = 4.93$), purple Lindgren funnel trap ($M = 8.13$, $SD = 6.47$), blue pan trap ($M = 13.84$, $SD = 7.46$), purple pan trap ($M = 19.15$, $SD = 7.95$), red pan trap ($M = 13.13$, $SD = 6.02$), white pan trap ($M = 19.98$, $SD = 9.72$), yellow pan trap ($M = 15.17$, $SD = 8.61$) ($p > 0.05$, Tukey-Kramer) (Figs. 4a,b). Exclusion of Formicidae did not significantly alter the results of the ANOVA or Tukey-Kramer tests, although it did reduce the standard deviation across all traps, most especially in pitfall, Malaise, and Berlese-Tullgren extraction.

Species accumulation curves for four of the thirteen trap types (upper and lower canopy traps, red pan trap, green Lindgren funnel traps) became asymptotic within the number of samples collected during this study (72, 72, 83, and 85) (Figs. 7, A1a–m). The accumulation curves for the majority of the remaining traps were estimated to become asymptotic by 300 samples, except Malaise, purple pan, and pitfall traps, which were estimated to become asymptotic by 600, 600, and 1000 samples, respectively (Fig. 8). Excluding Formicidae did not significantly alter the number of samples required before each trap became asymptotic, though did lower the number of species expected.

When Formicidae were included, pitfall and pan traps all exhibited very high similarity with each other. Aerial traps (upper canopy, Malaise, and Lindgren funnel traps) exhibited high to very high similarity with each other but medium to high similarity with pitfall and pan traps, except for Malaise traps which exhibited high to very high similarity with pitfall and pan traps. Berlese-Tullgren extraction exhibited the lowest similarity with most trap types, except for pitfall traps (Fig. 9a). When Formicidae were excluded, the same general patterns appeared to be evident though with less similarity between trap types (Fig. 9b).

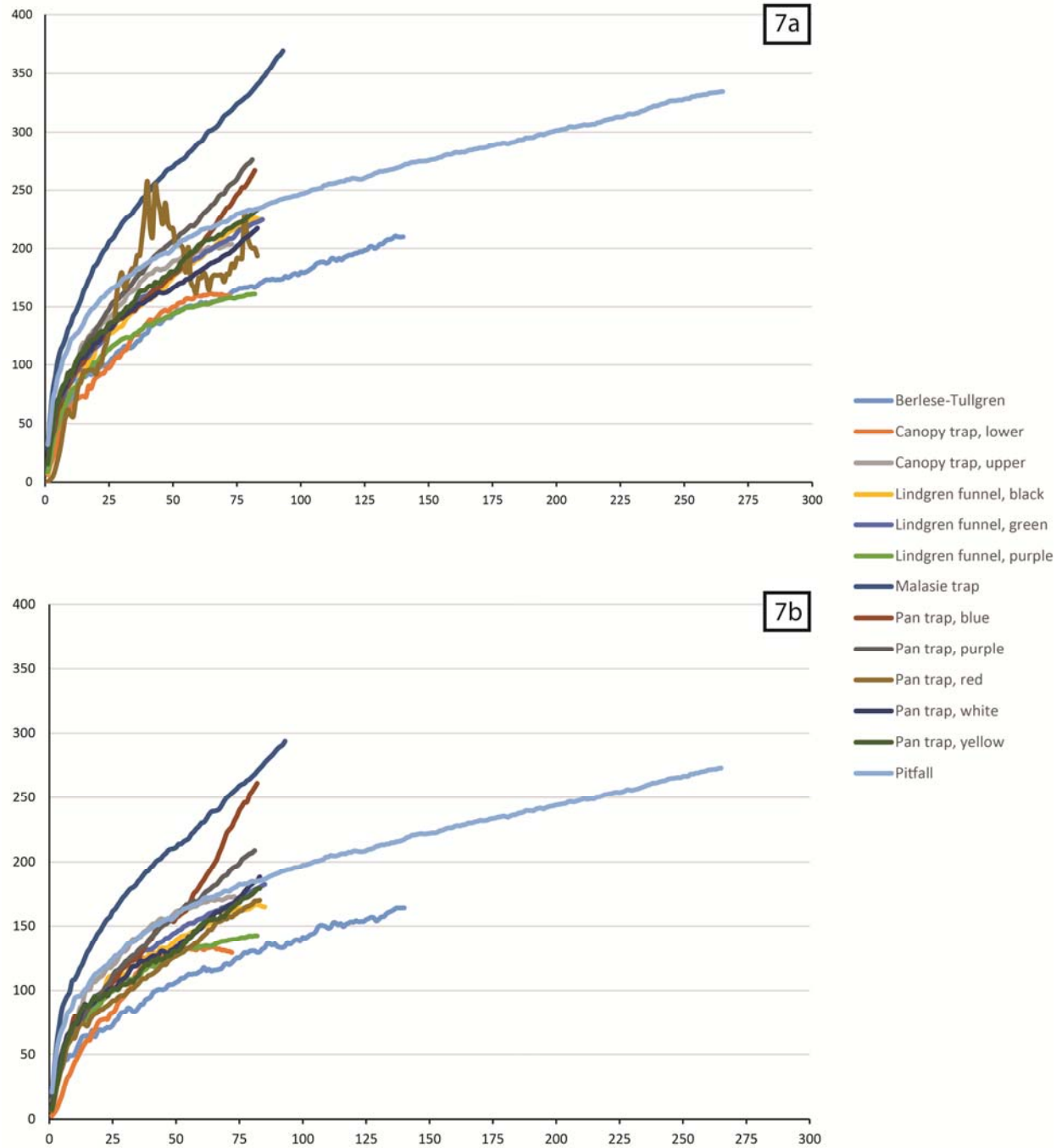


Figure 7. Chao 1 rarefaction curves based on the data. **a.** Formicidae included. **b.** Formicidae excluded.

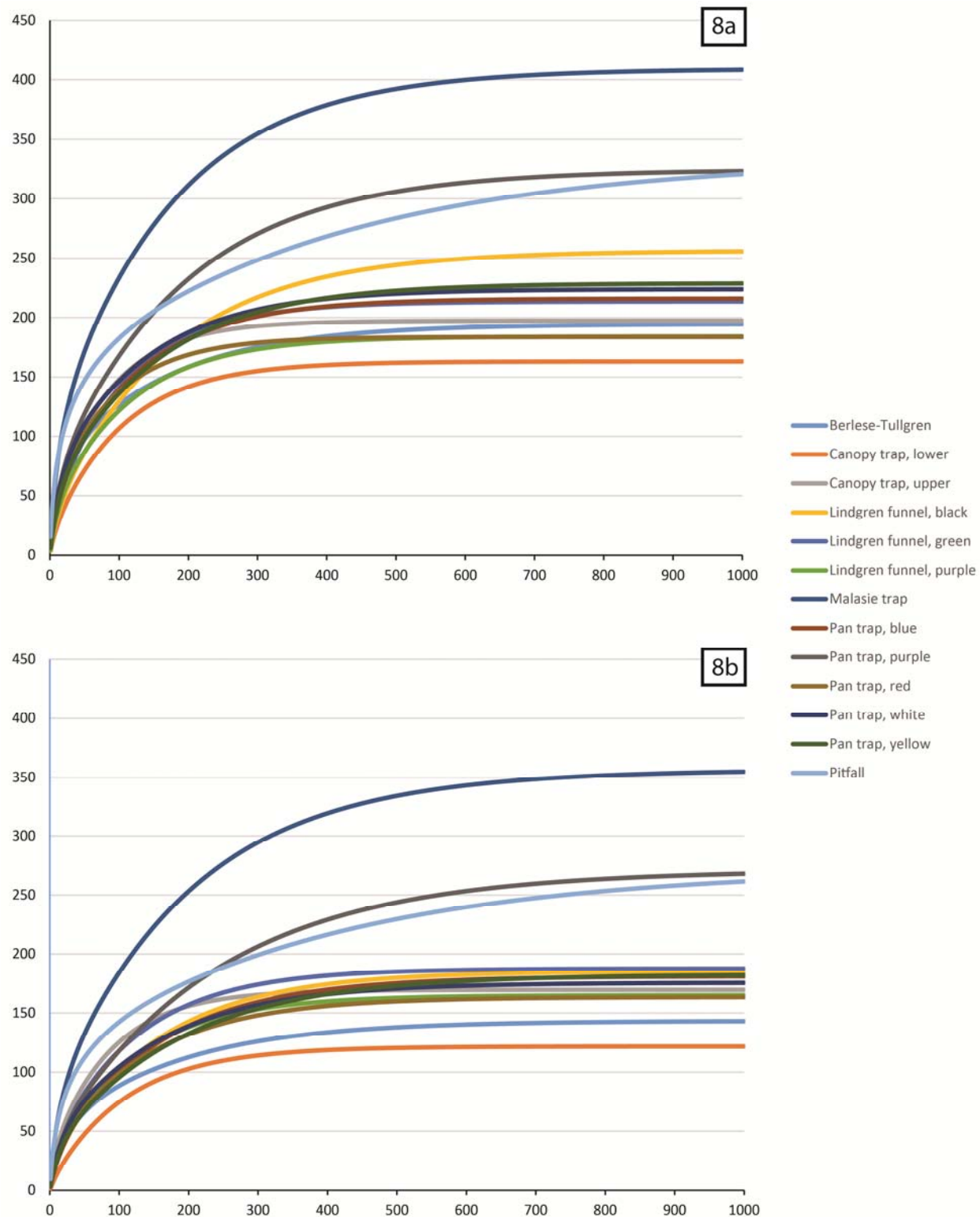


Figure 8. Estimated rarefaction curves (S(est)) extrapolated to 1000 samples. **a.** Formicidae included. **b.** Formicidae excluded.

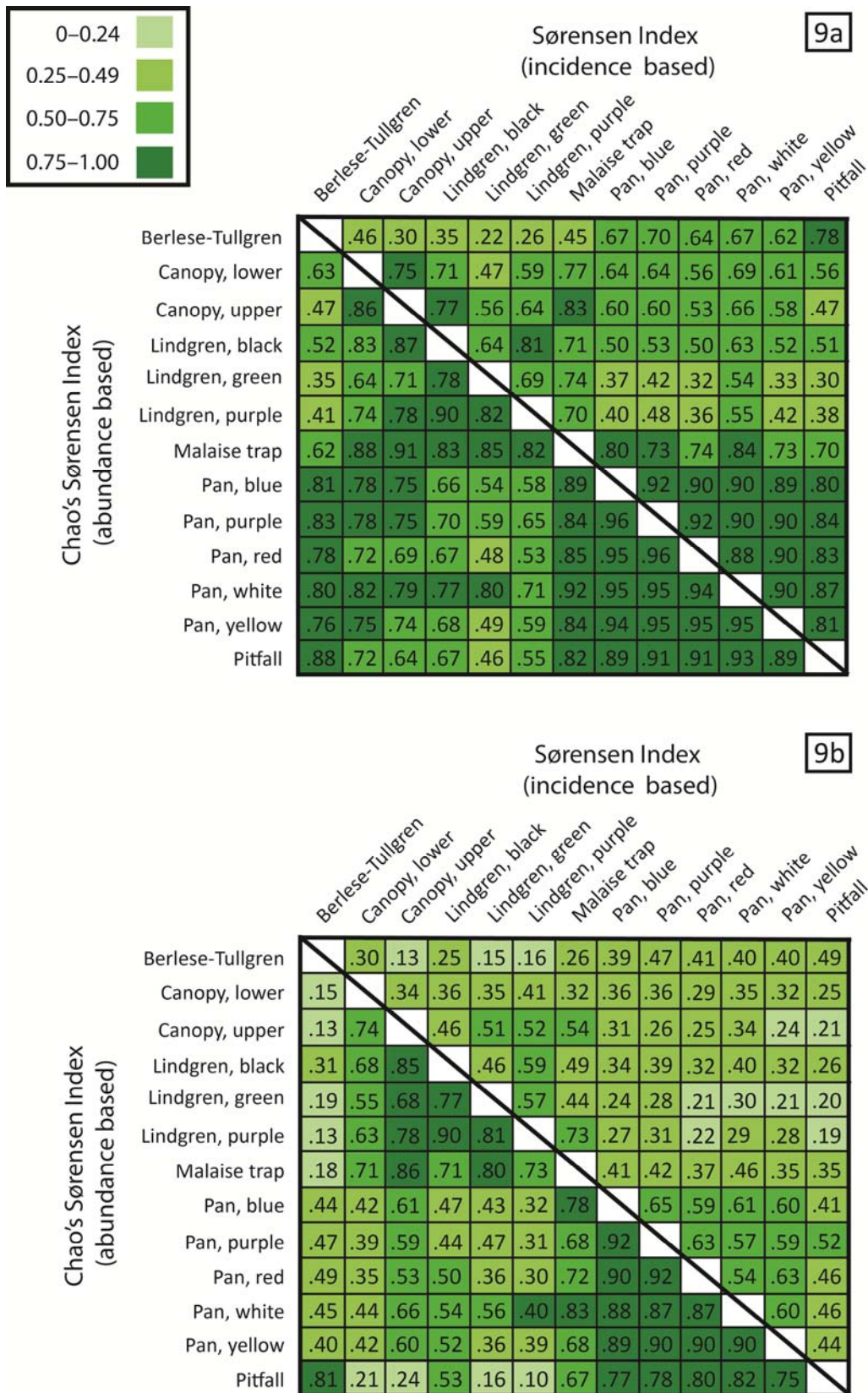
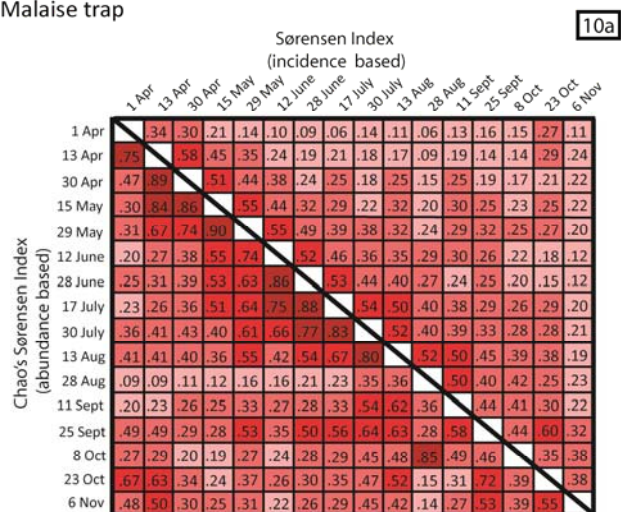


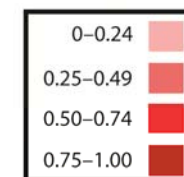
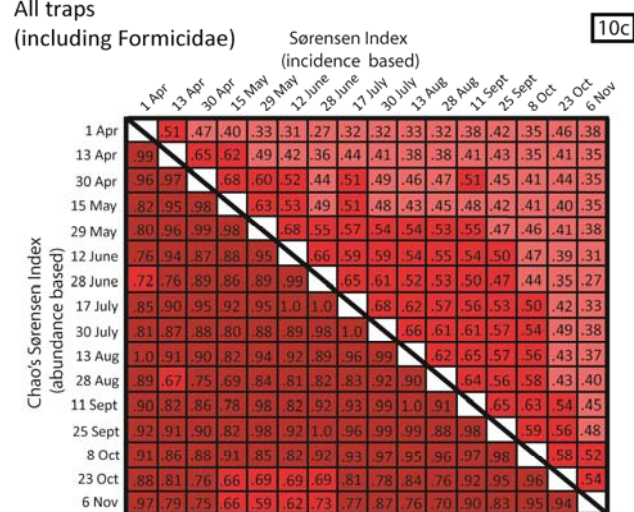
Figure 9. Similarity of trap catch as determined by Sørensen and Chao's Sørensen Indices. **Fig. 9a.** Formicidae included. **Fig. 9b.** Formicidae excluded.

Many species that were collected in large enough numbers to examine phenology exhibited season trends in diversity (Figs. A2a–r). However, when considered in aggregate, the average number of species and specimens collected per trap showed less distinct trends. The number of species collected increased in the early spring and decreased in early winter, with local peaks in early summer and fall (Fig. 3). The similarity and turnover between collection dates varied between traps: for example, collection dates for Malaise traps, depending on the method of analysis, generally exhibited very high or high similarity within one or two collection periods (approximately 2–4 weeks) (Fig. 10a) while collection dates for pitfall traps exhibited very high or high similarity throughout nearly the entire collecting season (Fig. 10b). Collection dates for other traps exhibited a range of similarity between collection dates (Figs. A3a–m). When all traps are combined and Formicidae included, dates from late spring through early fall exhibit high similarity (Sørensen) or exhibit very high similarity throughout the collection period (Chao's Sørensen) (Fig. 10c). However, when ants are excluded, samples taken two to three collection periods (4–6 weeks) around a given collection date exhibit high to very high similarity, but collections beyond that only exhibit medium to high similarity, depending on the analysis (Fig. 10d).

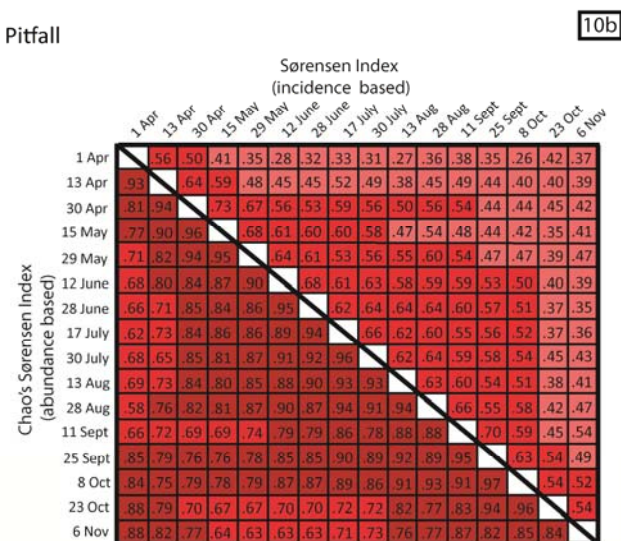
Malaise trap



All traps



Pitfall



All taxa

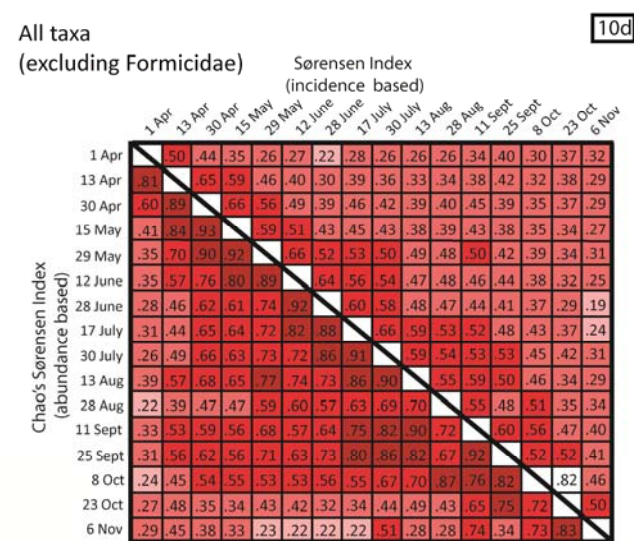


Figure 10. Sørensen and Chao's Sørensen Indices comparing similarity of trap catch by date. **Fig. 10a.** Malaise traps. **Fig. 10b.** Pitfall traps. **Fig. 10c.** All traps combined, including Formicidae. **Fig. 10d.** All traps combined, excluding Formicidae.

Discussion.

As may be expected, the species sampled by terrestrial collection methods (i.e., pitfall traps and Berlese-Tullgren extraction) and aerial traps (i.e., Malaise, canopy, and Lindgren funnel traps) generally exhibited high to very high similarity within each group (terrestrial vs aerial) but lower similarity between the groups. They are likely targeting different arthropod assemblages and a combination of techniques is required if maximum diversity is to be sampled.

Pitfall and Malaise traps collected the most species on average; pitfall, Malaise, and purple pan traps were estimated to collect the most species after species accumulation curves become asymptotic; and pitfall and Malaise traps and Berlese-Tullgren extraction of leaf litter collected the most specimens on average. While this was certainly influenced by the taxa included in the analysis, and slightly different results might be obtained if different taxa were included, it likely reflected the true performance of the different trap types for two reasons: 1) all individuals from a diversity of higher taxa were included, limiting the influence any one taxon would have on the results and 2) the inclusion of a number of easily-identified species from an even wider range of orders and families introduced additional variation in life-history and minimized the impact of expert-bias when picking which taxa were included.

The species collected by pan traps generally exhibited high to very high similarity with terrestrial and aerial traps (except green Lindgren funnel traps), probably because they collected both flying and crawling insects, although generally underperformed in species and specimen collection when compared to Malaise or pitfall traps. The pan traps in this study, however, were not buried flush with the substrate and likely missed many arthropods that would fall into a pitfall trap but could not scale the sides of the pan. One potential solution is to combine pitfall and pan traps by using open, colored pitfalls flush with the substrate (Skvarla *et al.* 2014

[Chapter II]; Ernst *et al.* 2015). The only study that examined the effect of color in pitfall traps found that flying pollinators and carabids were collected in higher numbers in white and yellow (except carabids) pitfall traps compared to green and brown pitfalls and that terrestrial taxa, such as Isopoda, were not affected by trap color (Buchholz *et al.* 2010). Such pitfalls cannot be run with rain covers and will be more affected by rainfall than covered traps when run for extended periods of time. However, it may be possible to employ a clear rain cover without affecting the attractiveness of the trap to flying insects.

Different species exhibited markedly different phenologies, as should be expected from a diverse assemblage of taxa. Taken collectively, two activity peaks were apparent in the spring and fall, with the larger peak occurring in the spring. The number of specimens collected, however, showed less variation overall, although individual traps may collect more or less when abundant species are present.

The species turnover, as reflected in the similarity between collection dates, varied by trap; species collected in Malaise and other aerial traps exhibited high or very high similarity between collections two to four weeks apart and decreased in similarity thereafter. Species collected by pitfall traps and Berlese-Tullgren extraction exhibited high to very high similarity throughout the trapping period. This suggested that aerial species were present for shorter periods of time, possibly because of changing abiotic factors such as precipitation, moisture, and temperature in the relatively exposed canopy, and collections targeting this group should either be continuous throughout the warm seasons or be made at least during every season.

Conversely, terrestrial species were present for longer periods and many of the most abundant species, especially ants, were present throughout the warm months; this may be because the leaf litter on the forest floor experiences less dramatic abiotic fluctuations and protected areas, such

as leaf litter next to logs or under rocks, retain moisture. It may be possible to collect most terrestrial diversity in only a few collection periods as long as a relative increase in effort is made. However, it is unlikely that collecting during only one period during the year is sufficient to sample most diversity, as many terrestrial species fluctuate in abundance or, depending on the group, the percent of the population that are adults and therefore identifiable to species.

Including data for Formicidae generally did not have a significant impact on the statistical analyses, except that including the data increased the standard deviations of the average number of species and specimens collected per trap type (Figs. 2, 4). Additionally, including Formicidae caused all collection dates to exhibit high to very high similarity with respect to species collected while excluding Formicidae resulted in collection dates within two or three collections exhibiting high to very high similarity and dates beyond that exhibiting low to medium similarity with respect to the species collected (Figs. 10c, d). This suggested Formicidae did not exhibit much seasonality once they became active and the numerically dominant (in terms of specimens) ants overwhelmed other species when they were included in the analysis. It also suggested that caution should be employed when including species that are dominant in specimens but not species, as they can affect some analyses.

Fifty one percent of the species collected were represented by five or fewer specimens and 25% were represented by singletons. The species accumulation curves for most trap types did not become asymptotic and extrapolated rarefaction curves predicted 300–600 samples per trap type (1000 for pitfall traps), far more than were collected during this study. This suggested that even though the site was relatively small, a great deal more effort would be required to sample the majority of species present.

Finally, even though fewer than half of the species predicted by the species accumulation curves were collected, the survey still produced 15 new species and 36 new state records within the taxa identified. This highlights not only how much work remains to be done in Arkansas but also how much is left to discover even in a relatively well-studied area such as North America north of Mexico.

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Appendix I. Species rarefaction curves.

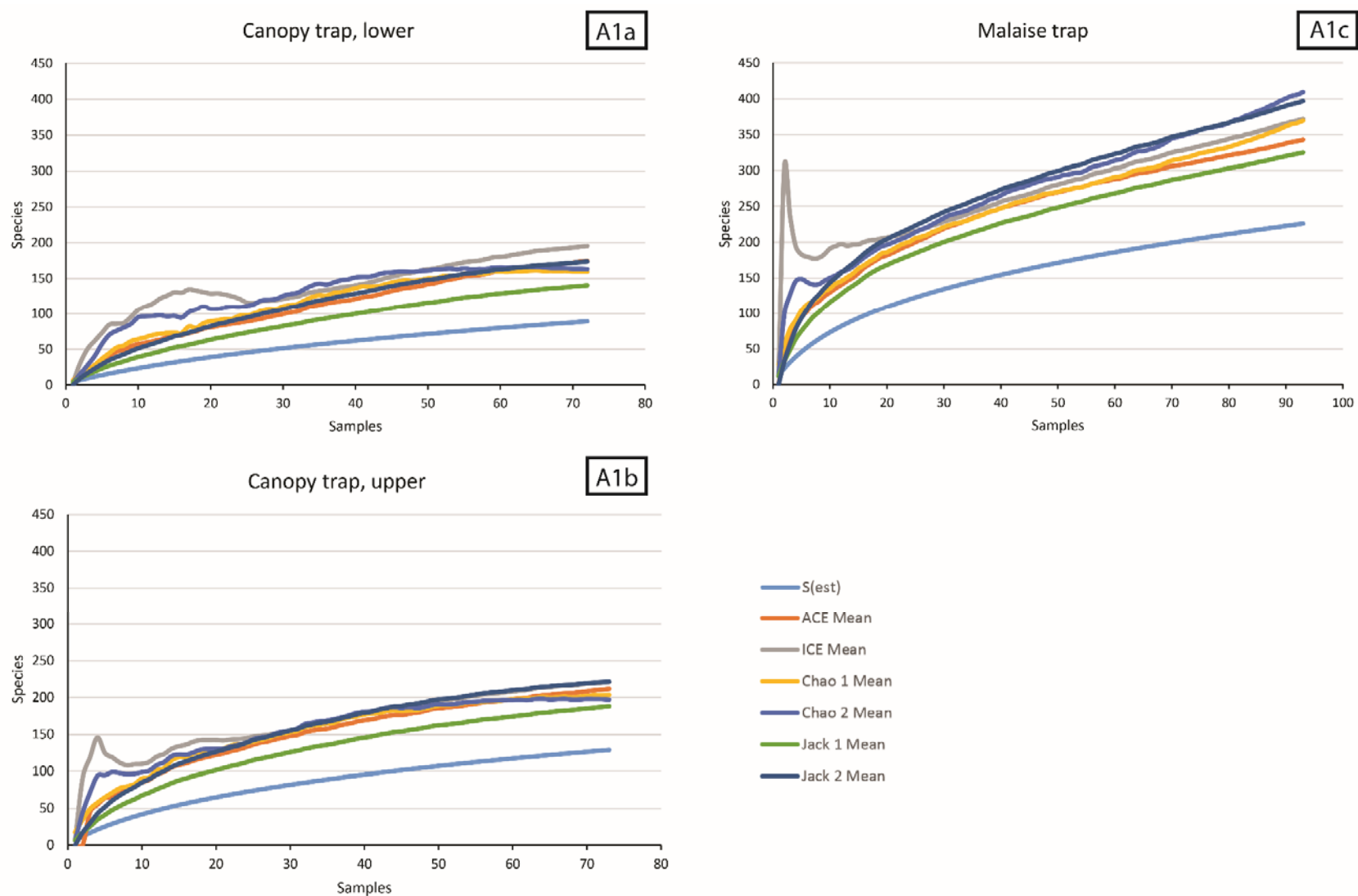
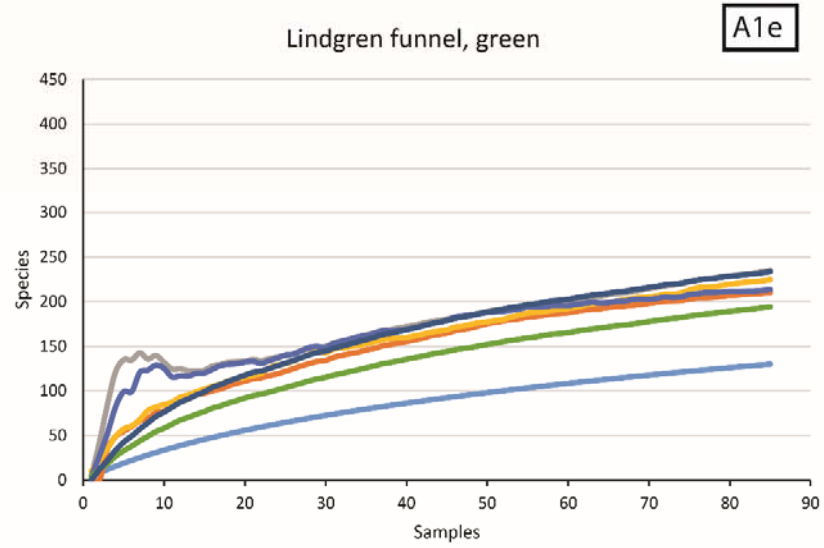
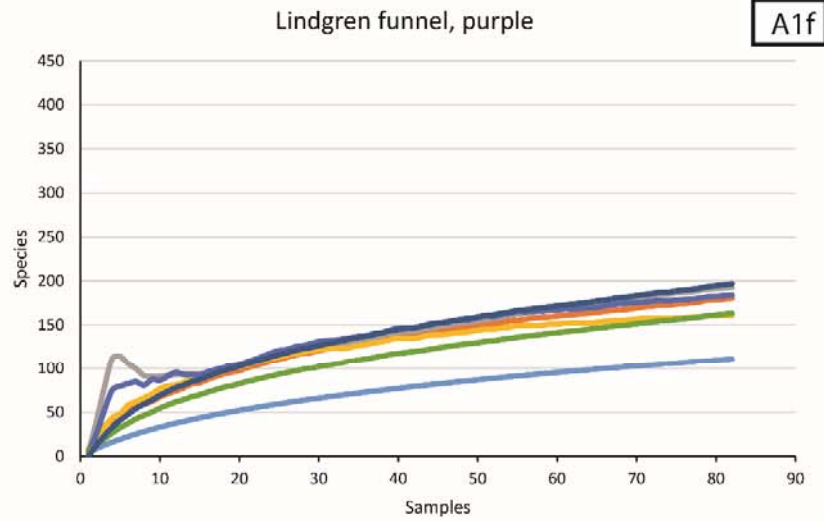
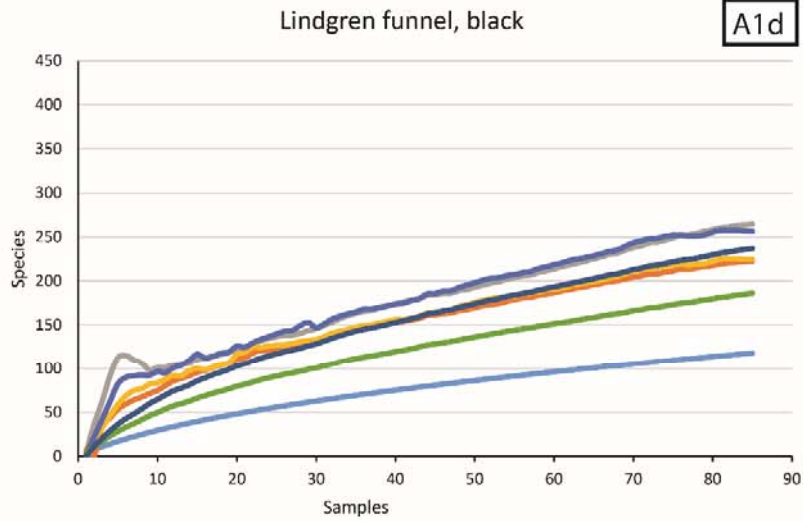
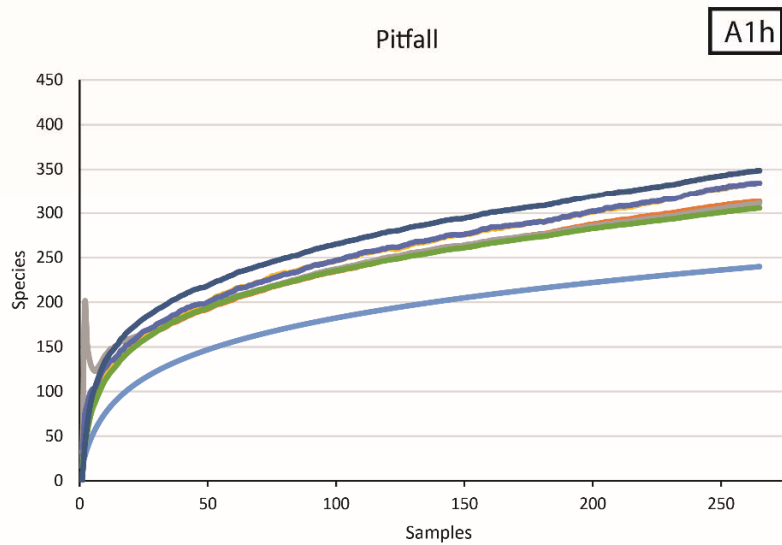
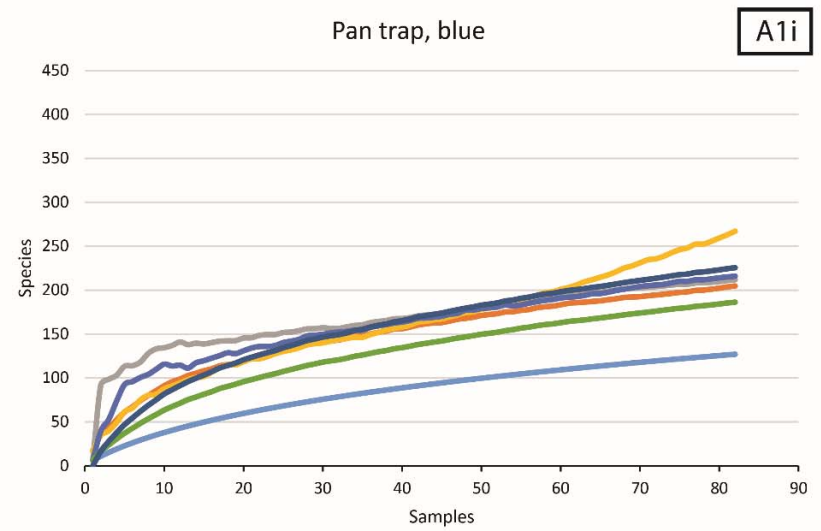
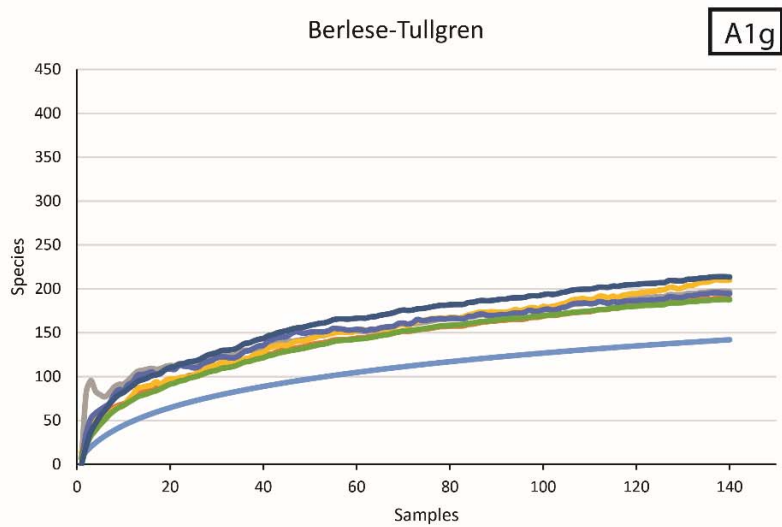


Figure A1. Species rarefaction curves. See caption at the end of the figures for further explanation.



- $S(\text{est})$
- ACE Mean
- ICE Mean
- Chao 1 Mean
- Chao 2 Mean
- Jack 1 Mean
- Jack 2 Mean

Figure A1. Species rarefaction curves. See caption at the end of the figures for further explanation.



- S(est)
- ACE Mean
- ICE Mean
- Chao 1 Mean
- Chao 2 Mean
- Jack 1 Mean
- Jack 2 Mean

Figure A1. Species rarefaction curves. See caption at the end of the figures for further explanation.

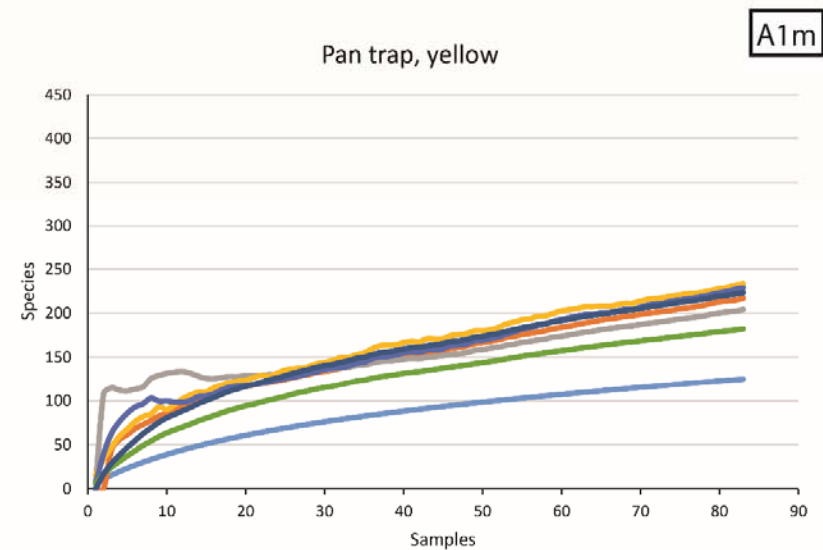
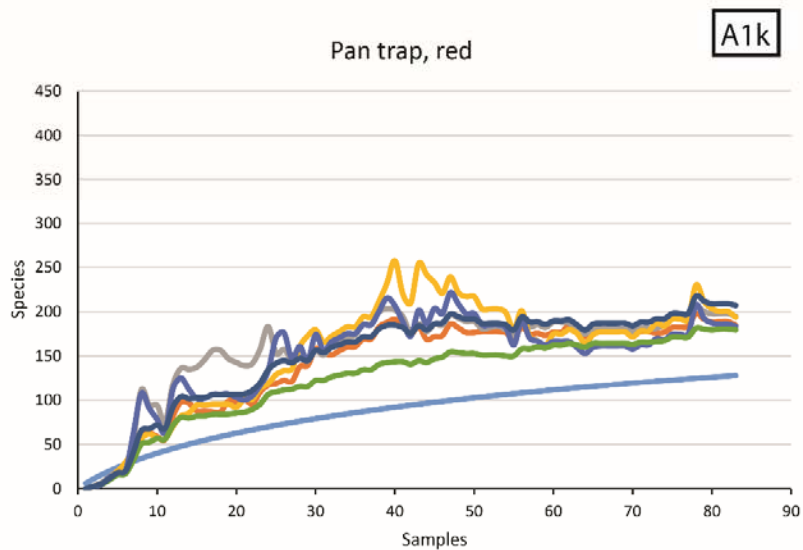
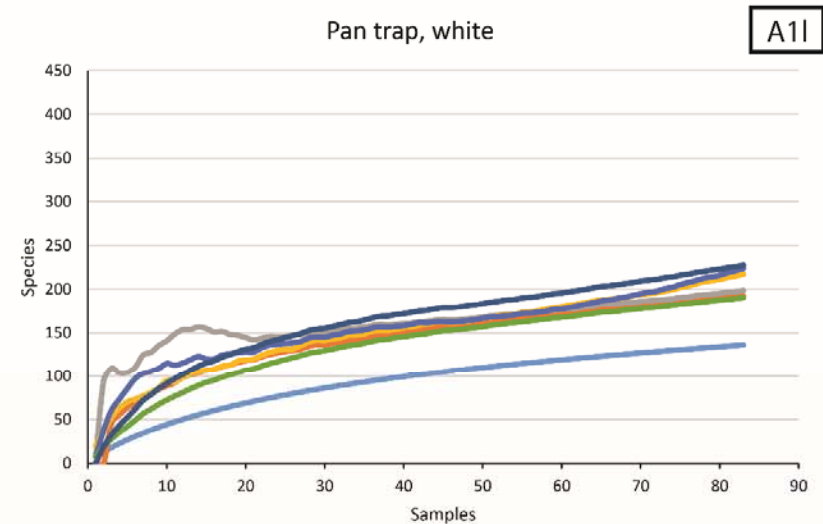
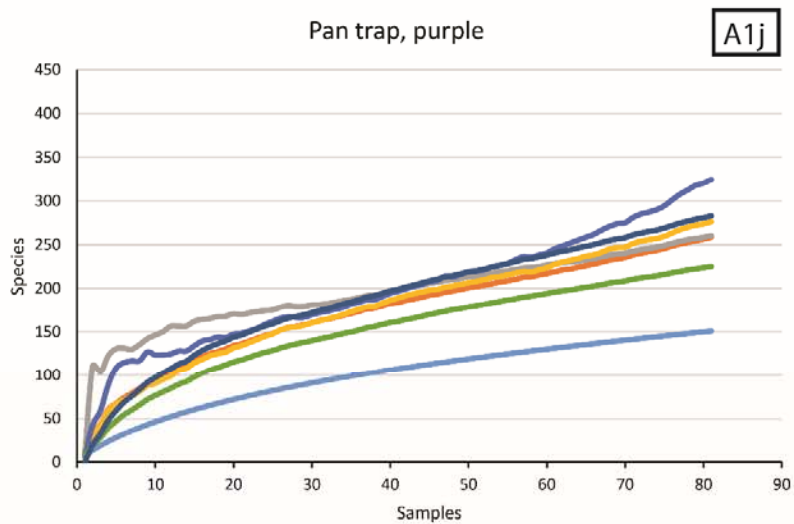


Figure A1. Species rarefaction curves. See caption at the end of the figures for further explanation.

Figures A1a–m. **Fig. A1a.** Canopy trap, upper collector. **Fig. A1b.** Canopy trap, lower collector. **Fig. A1c.** Malaise trap. **Fig. A1d.** Black Lindgren funnel trap. **Fig. A1e.** Green Lindgren funnel trap. **Fig. A1f.** Purple Lindgren funnel trap. **Fig. A1g.** Berlese-Tullgren extraction. **Fig. A1h.** Pitfall trap. **Fig. A1i.** Blue pan trap. **Fig. A1j.** Purple pan trap. **Fig. A1k.** Red pan trap. **Fig. A1l.** White pan trap. **Fig. A1m.** Yellow pan trap. Colors represent the same trap type throughout figures. The y-axis is standardized across graphs but the x-axis is determined by the number of samples, which varies by trap type.

Fig. A2a

Parasitengona

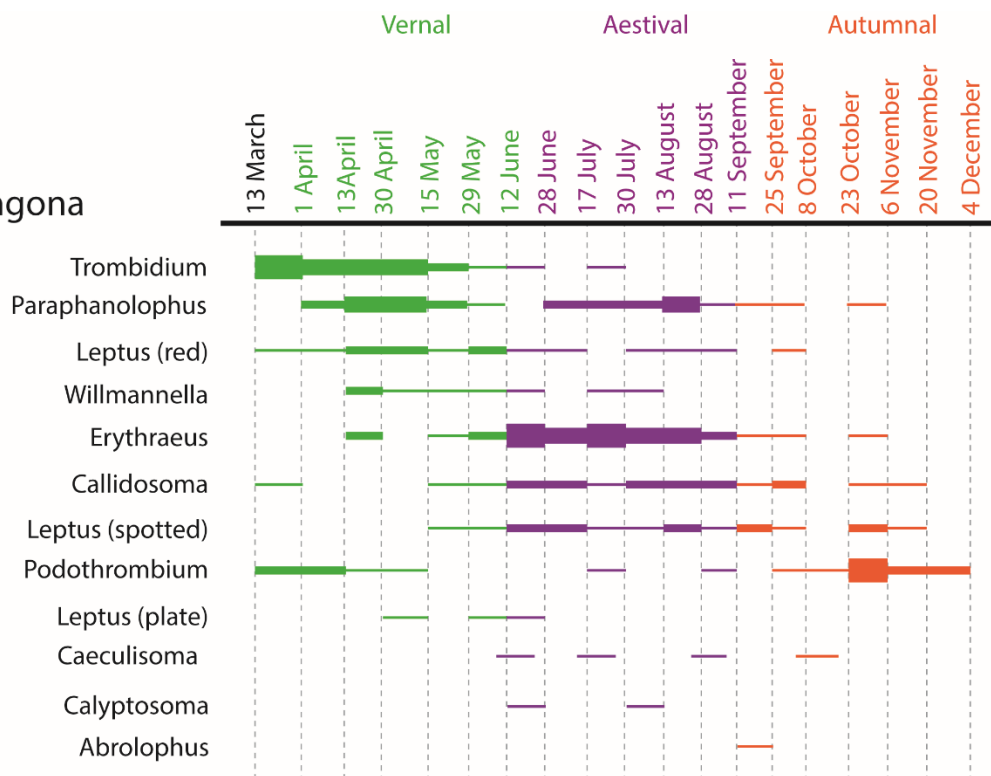


Fig. A2b

Ixodidae

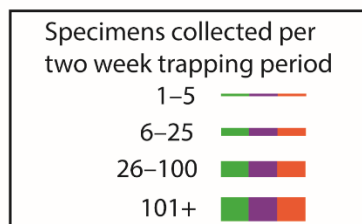
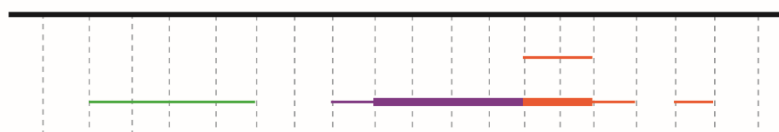
Amblyomma americanum
 Dermacentor variabilis
 Ixodes scapularis



Fig. A2c

Phalangodidae

Crosbyella
 Wespus arkansasensis



Figures A2a–c. Phenology of collected Chelicerata excluding Araneae. **Fig. A2a.** Parasitengona (Acari). **Fig. A2b.** Ixodidae (Parasitiformes). **Fig. A2c.** Phalangodidae (Opiliones).

Fig. A2d

Gnaphosidae

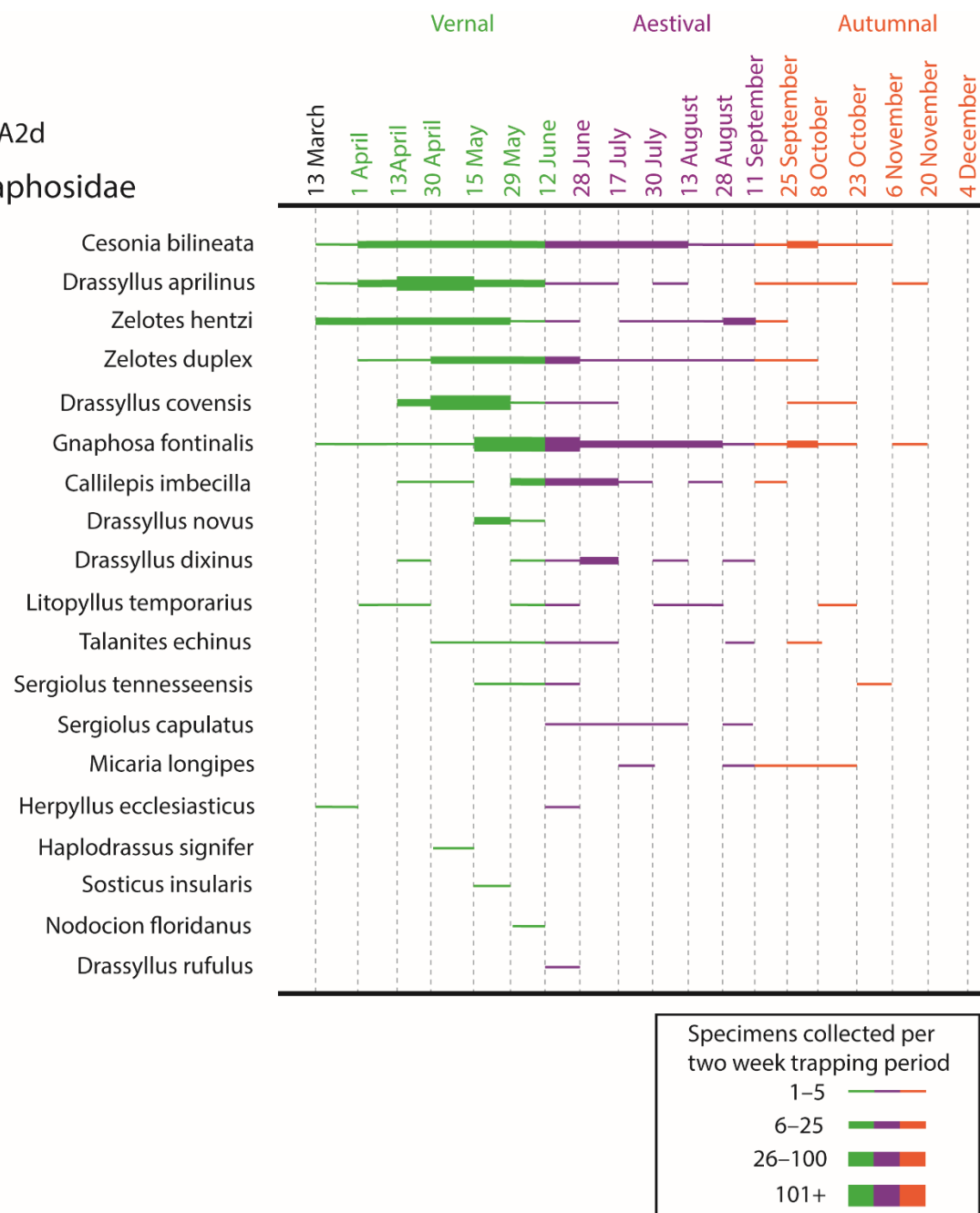


Figure A2d. Phenology of collected Gnaphosidae (Araneae).

Fig. A2e
Lycosidae

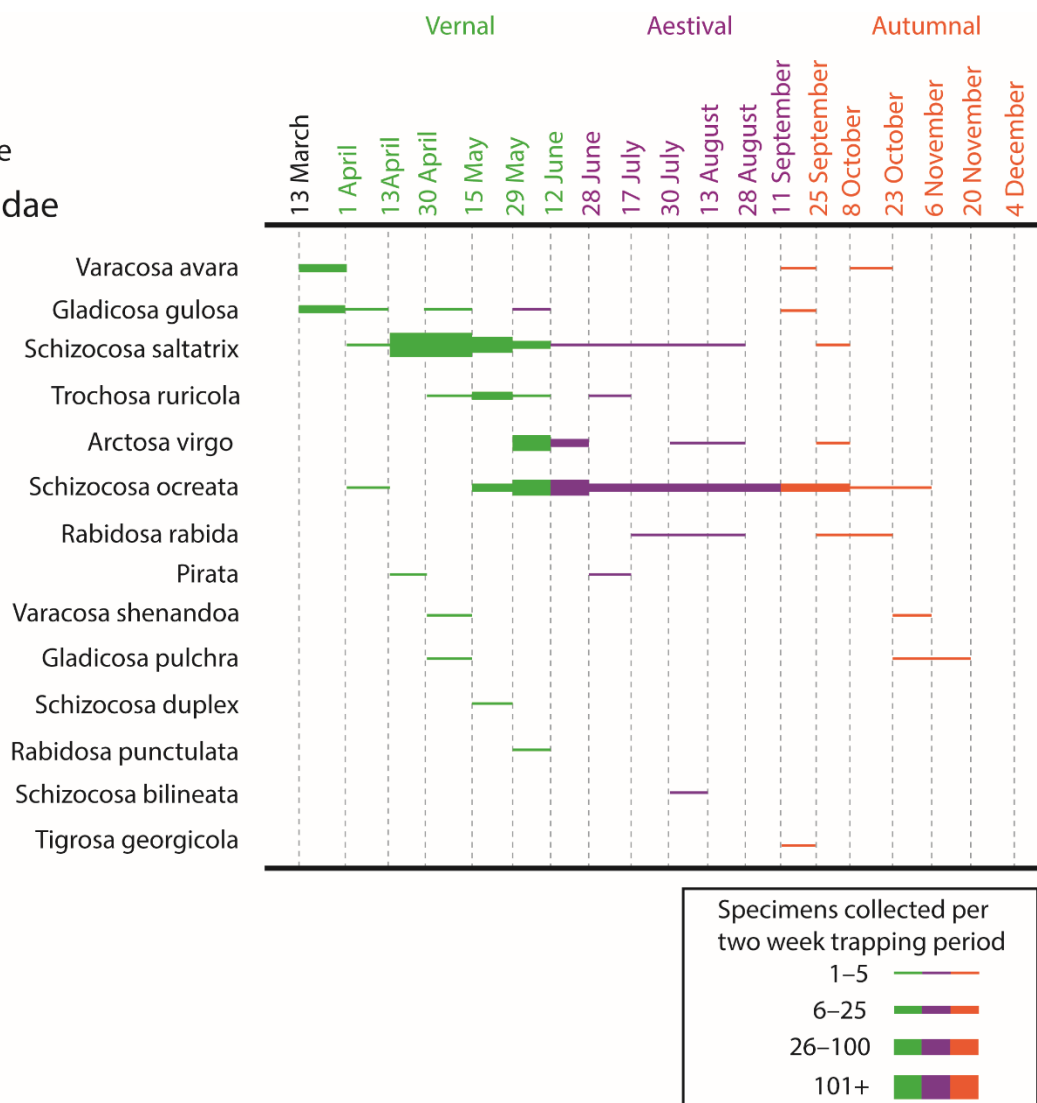


Figure A2e. Phenology of collected Lycosidae (Araneae).

Fig. A2f

Salticidae

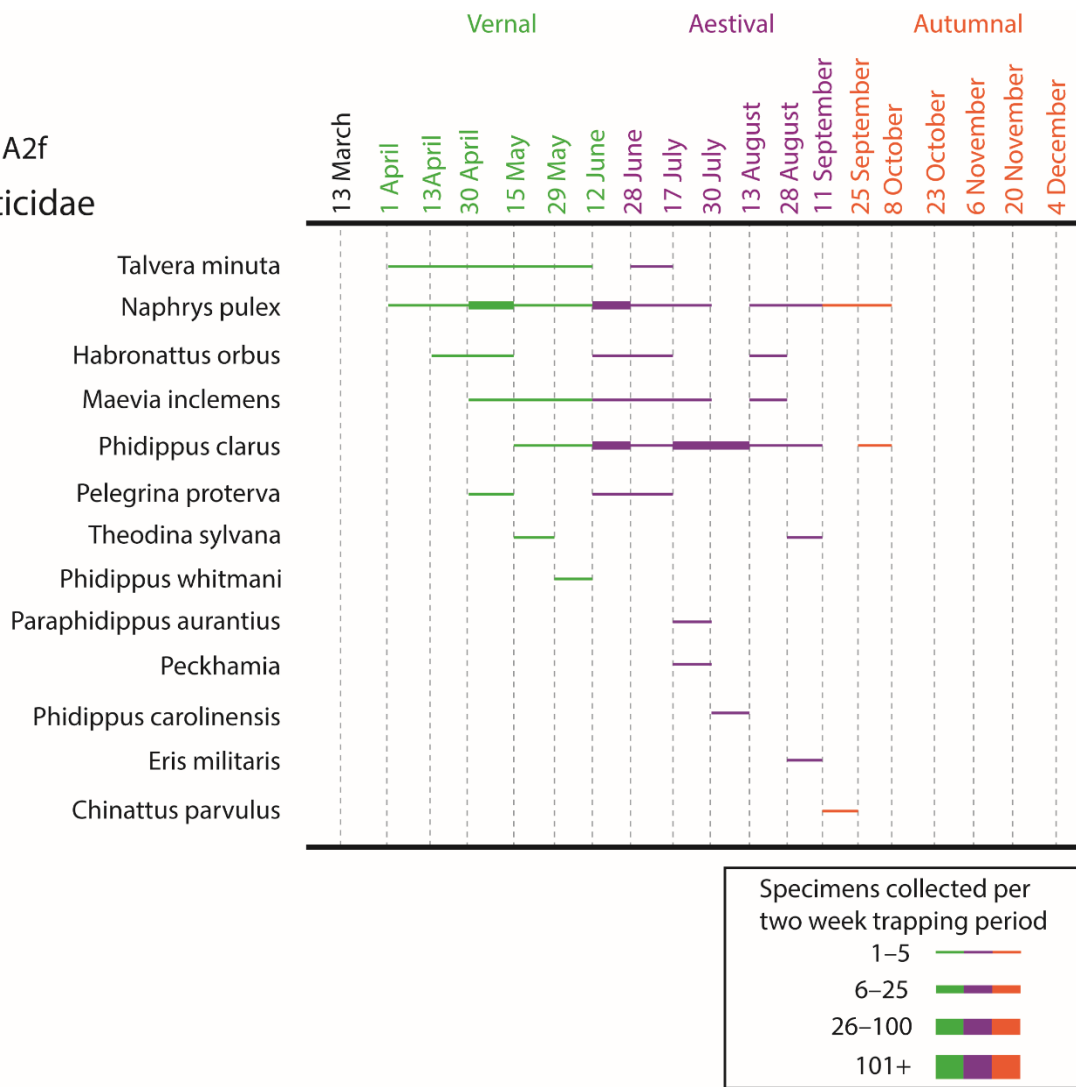


Figure A2f. Phenology of collected Salticidae (Araneae).

Fig. A2g

Agelenidae

Agelenopsis kastoni

Agelenopsis naevia

Agelenopsis pennsylvanica

Wadotes

Anyphaenidae

Anyphaena celer

Areneidae

Araniella displicata

Hypsosinga rubens

Neoscona crucifera

Ocrepeira

Araneus partitus

Atypidae

Sphodros niger

Clubionidae

Elaver excepta

Corrinidae

Castianeira cingulata

Castianeira longipalpa

Castianeira amoena

Castianeira crocata

Castianeira descripta

Casitaneira trilineata

Ctenidae

Cteneus exlineae

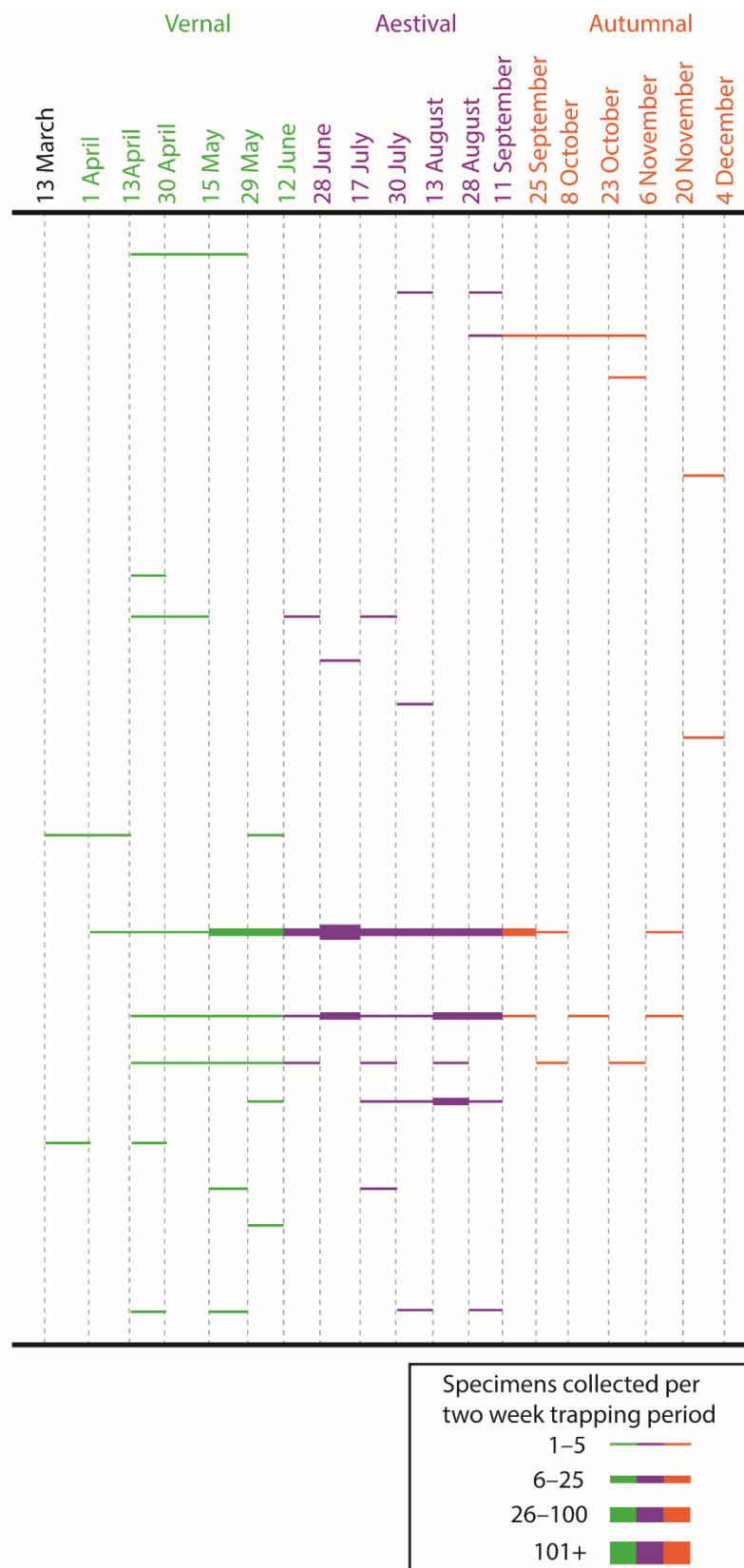


Figure A2g. Phenology of collected Araneae excluding Gnaphosidae, Lycosidae, and Salticidae.

Fig. A2g (cont.)

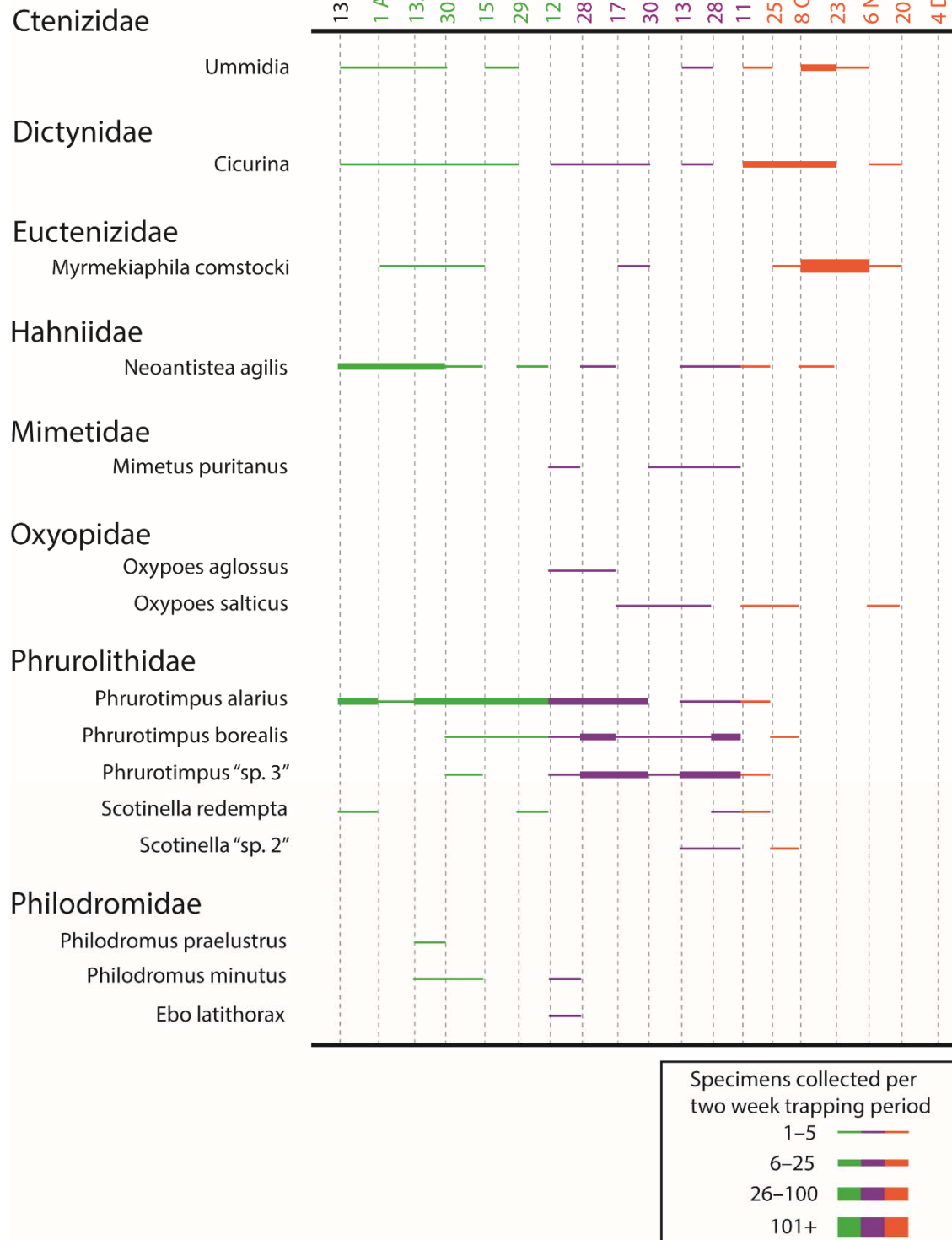


Figure A2g (cont.). Phenology of collected Araneae excluding Gnaphosidae, Lycosidae, and Salticidae.

Fig. A2g (cont.)

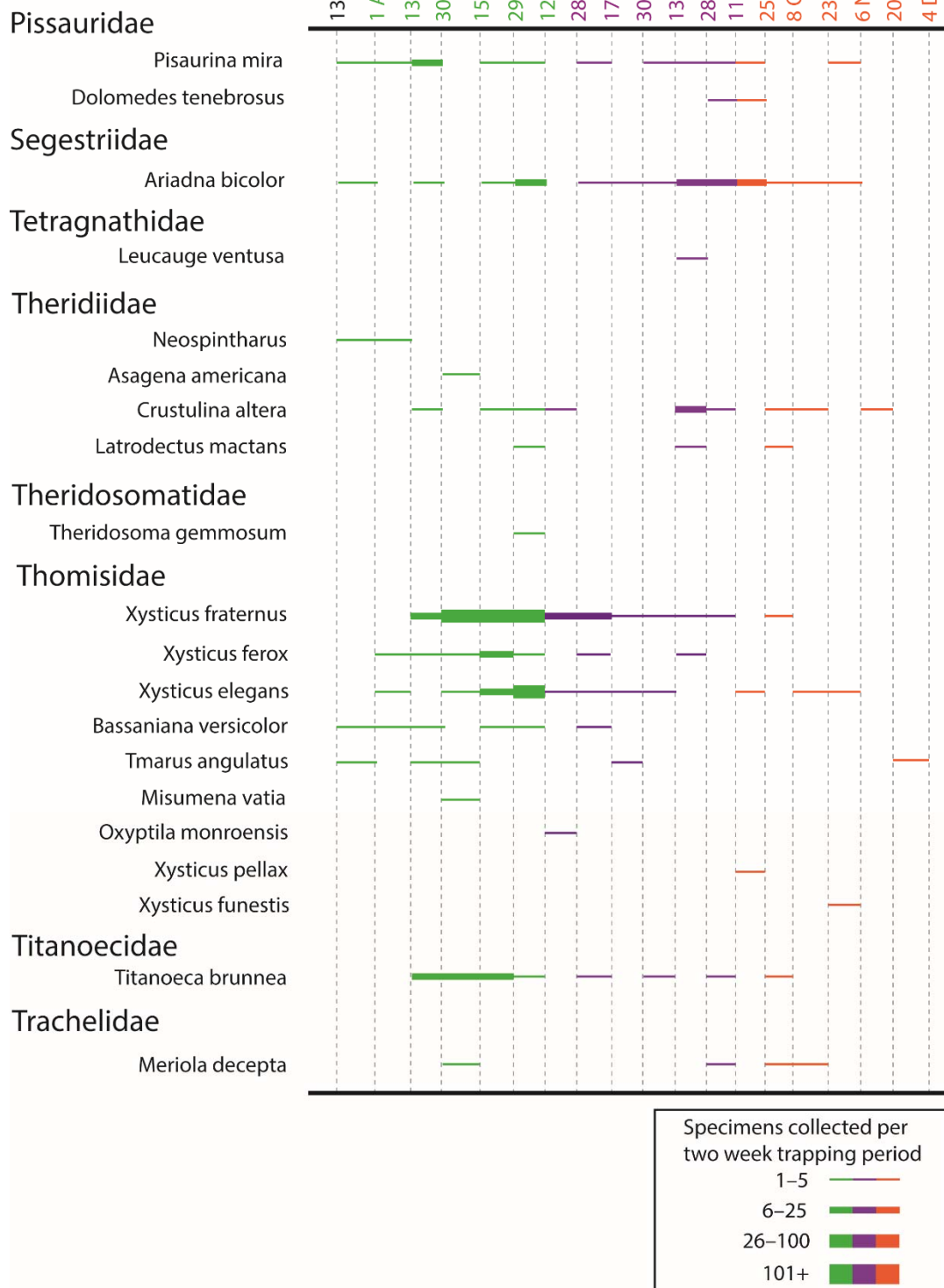


Figure A2g (cont.). Phenology of collected Araneae excluding Gnaphosidae, Lycosidae, and Salticidae.

Fig. A2h

Formicidae

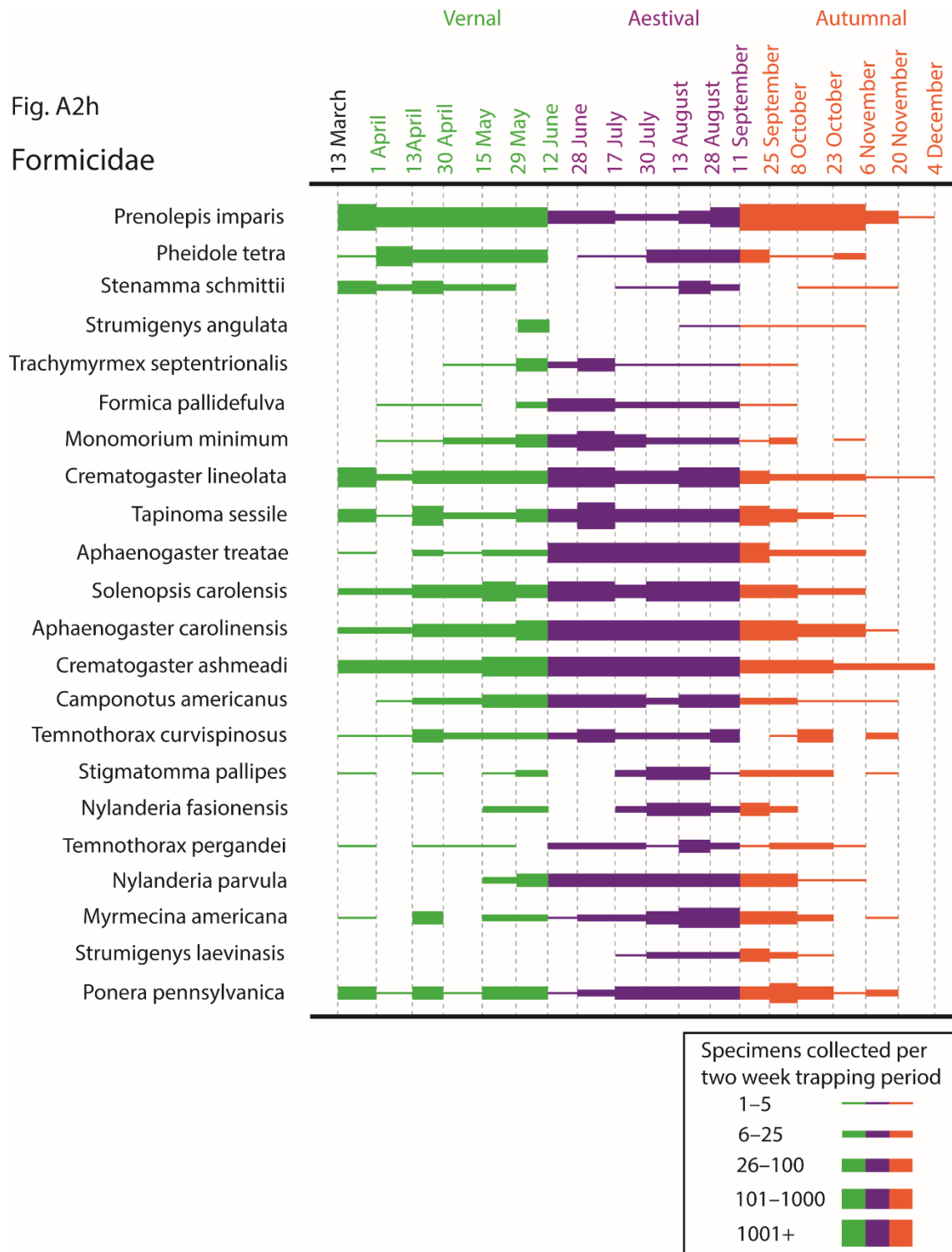


Figure A2h. Phenology of collected Formicidae, abundant species.

Fig. A2h (cont.)

Formicidae

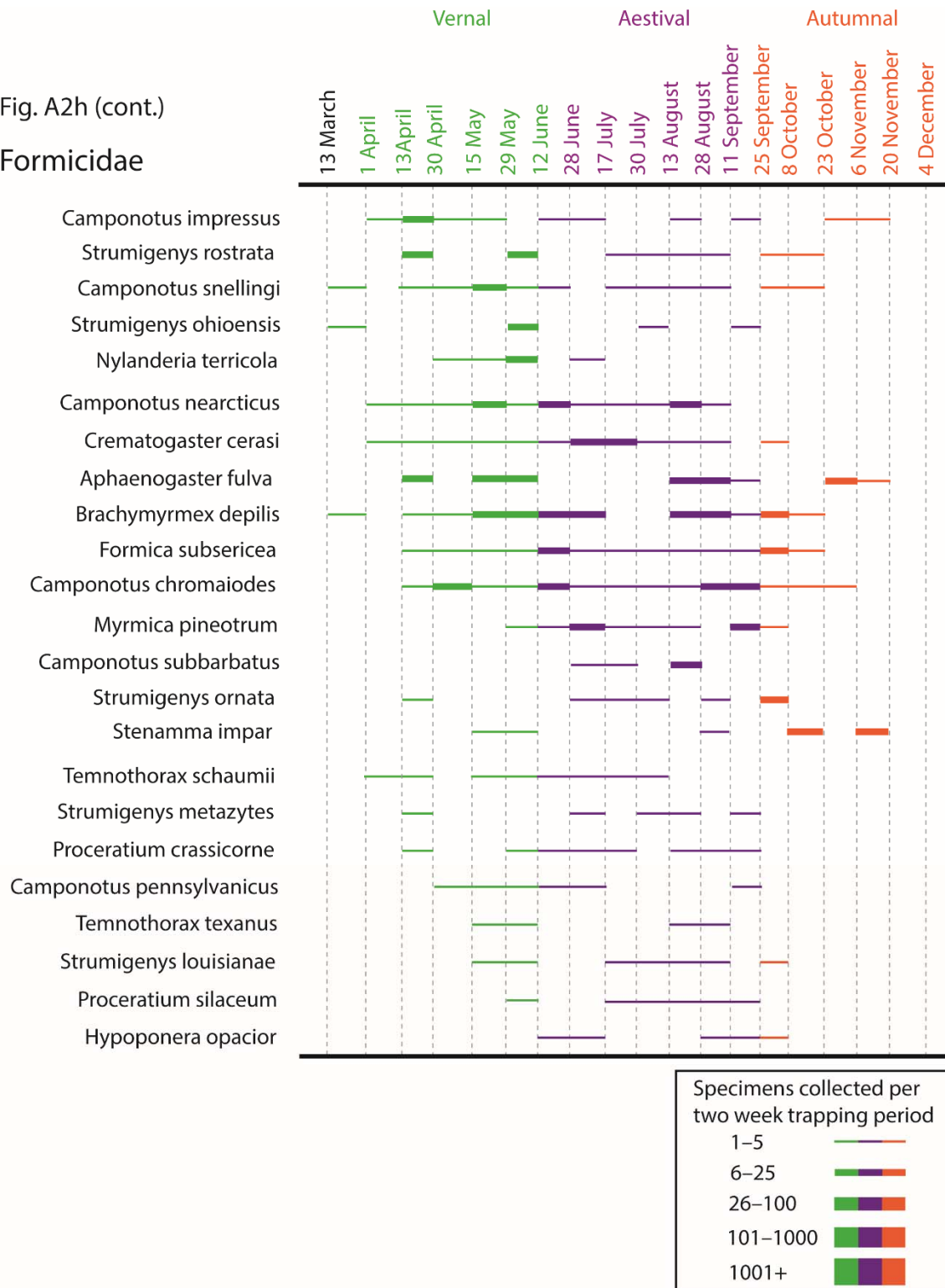


Figure A2h (cont.). Phenology of collected Formicidae, less abundant species.

Fig. A2h (cont.)

Formicidae

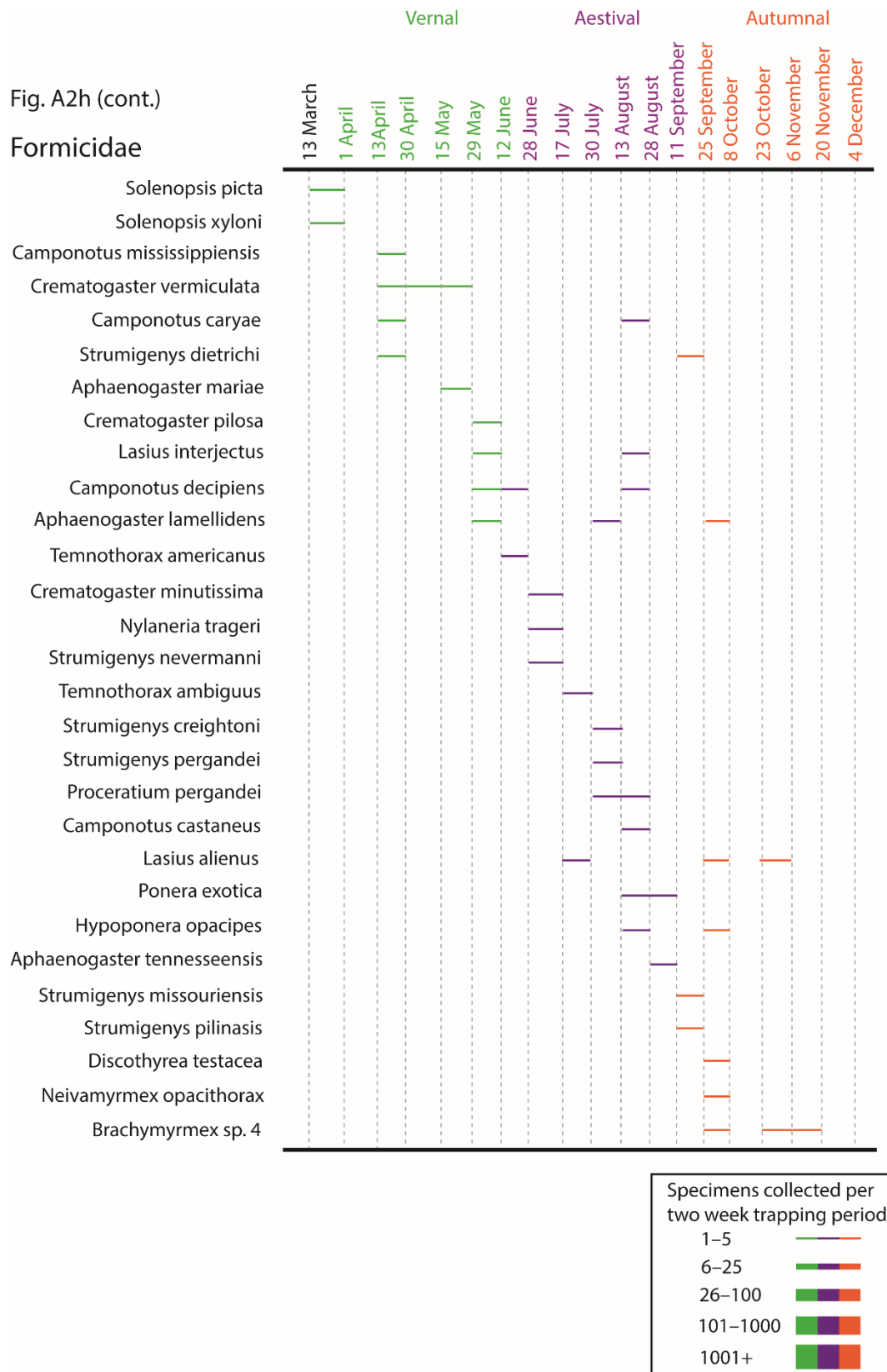


Figure A2h. Phenology of collected Formicidae, rare species collected on three or fewer collection dates and represented by five or fewer specimens per date.

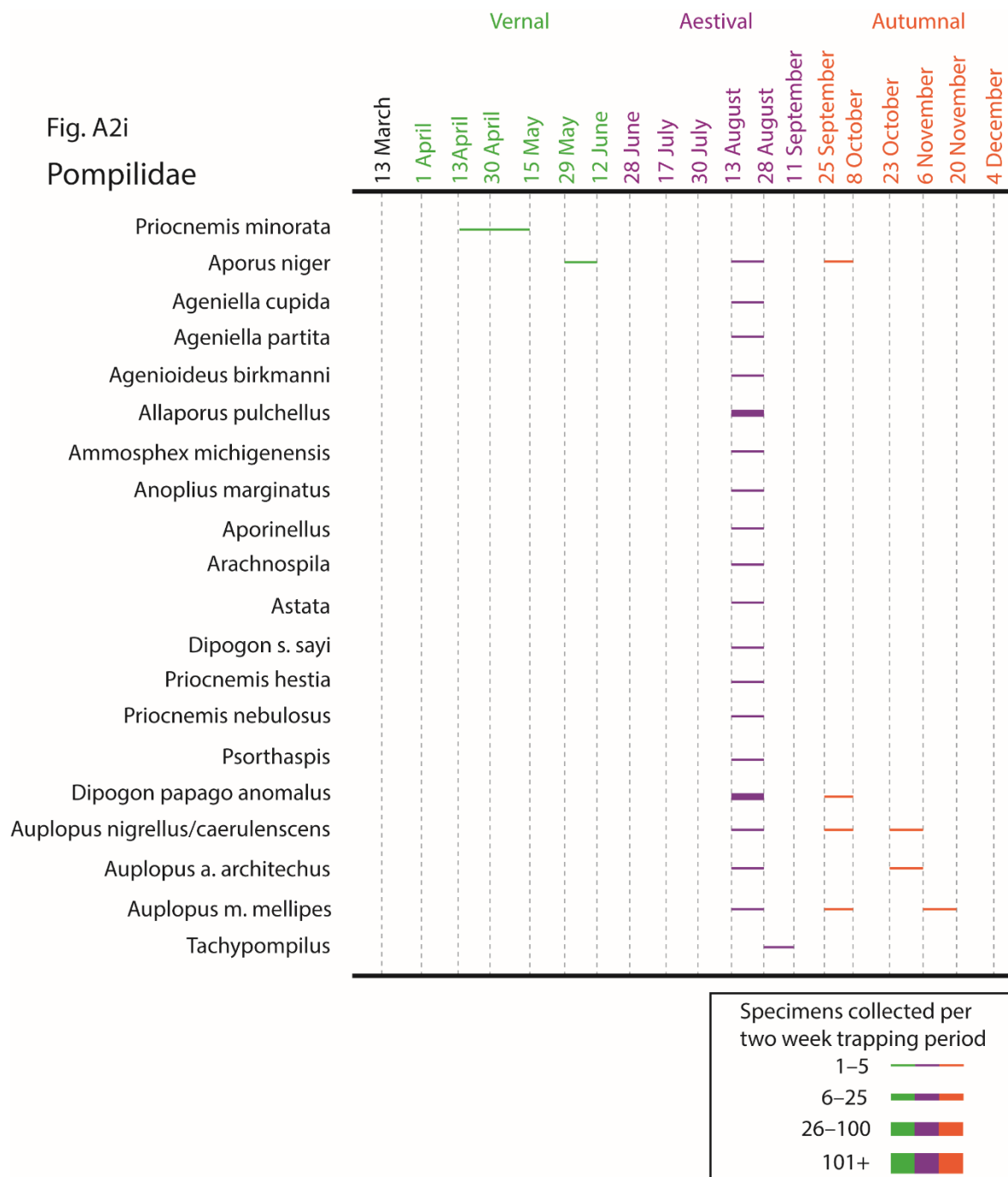


Figure A2i. Phenology of collected Pompilidae.

Fig. A2j

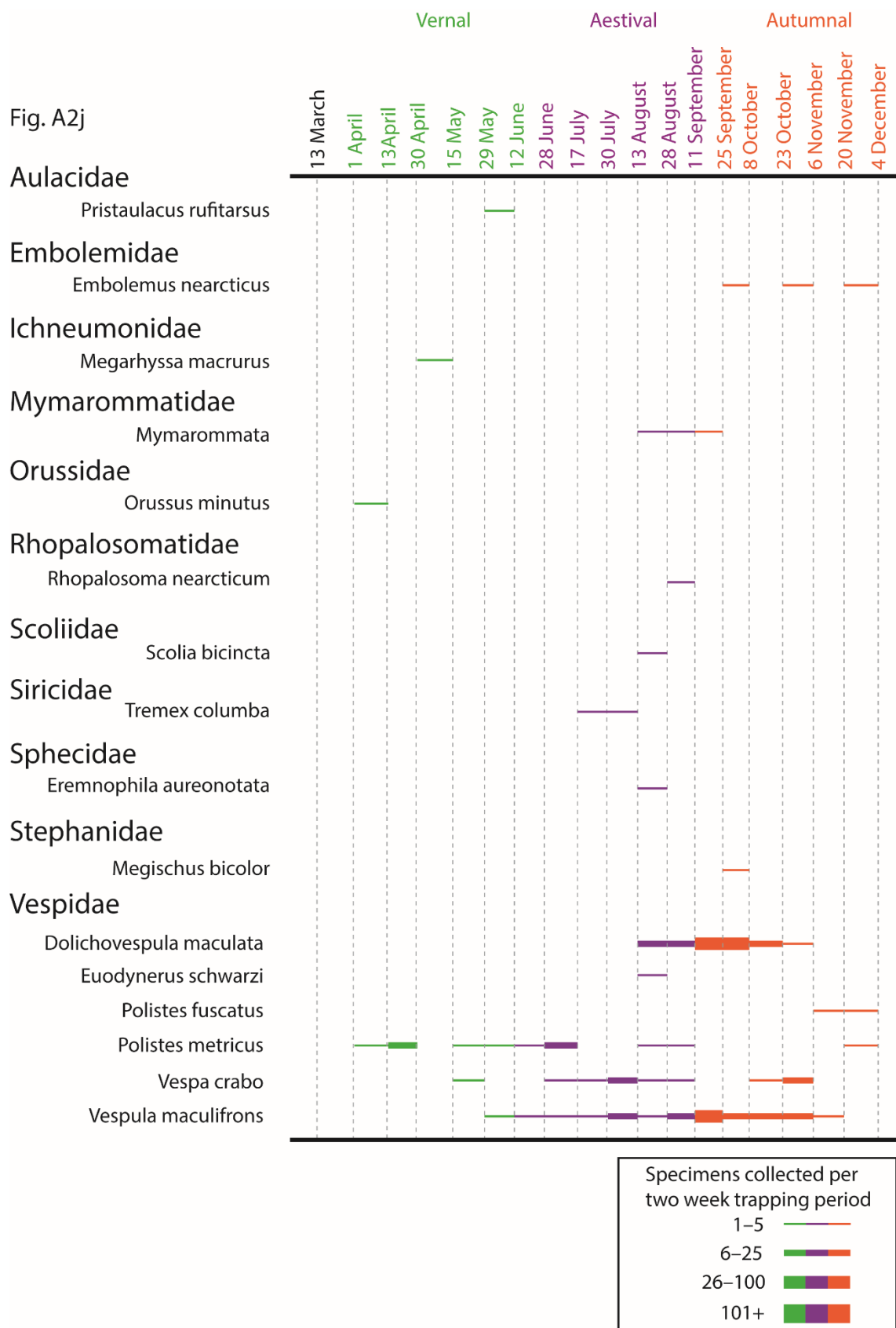


Figure A2j. Phenology of select collected Hymenoptera, excluding Formicidae and Pompilidae.

Fig. A2k

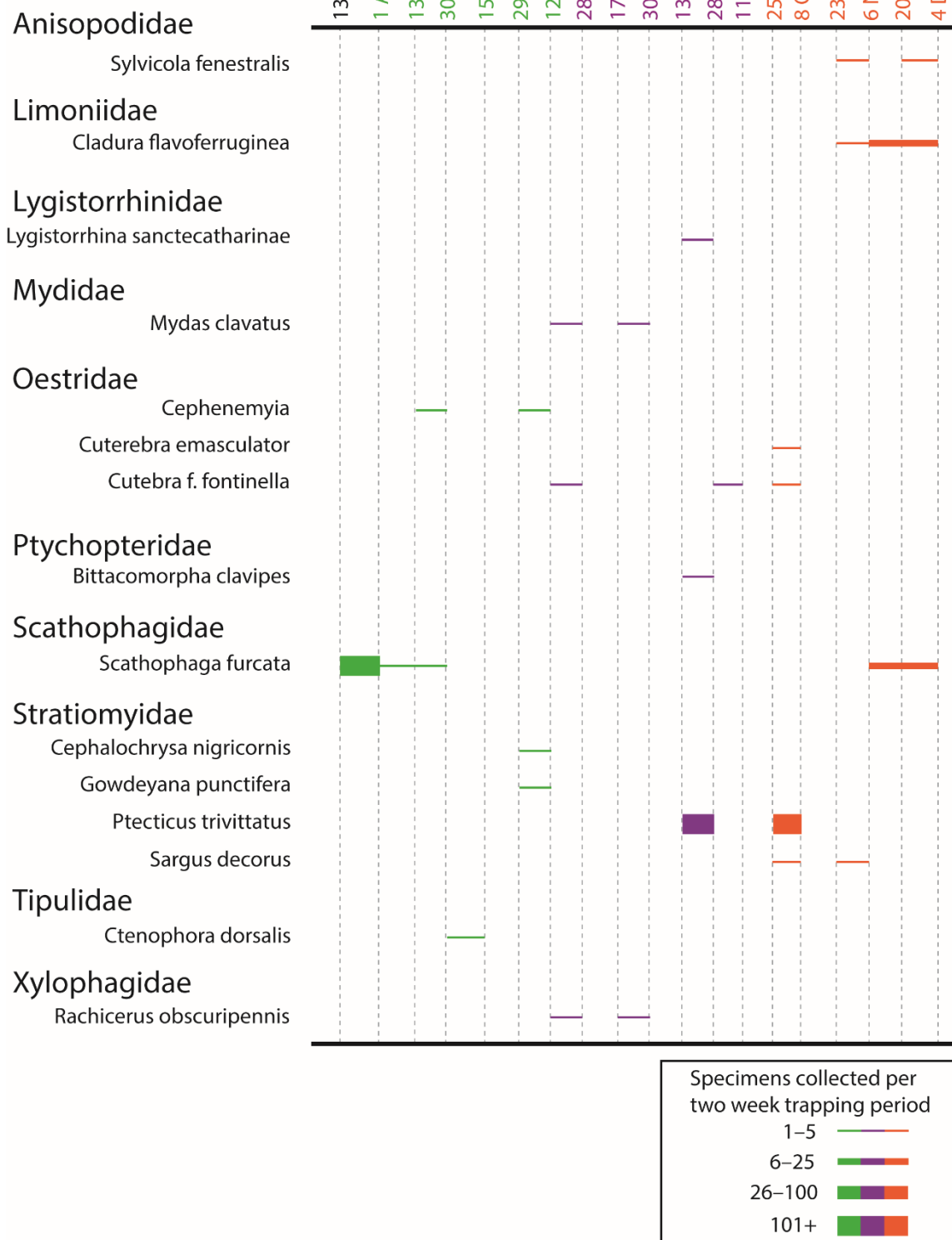


Figure A2k. Phenology of select collected Diptera

Fig. A2I

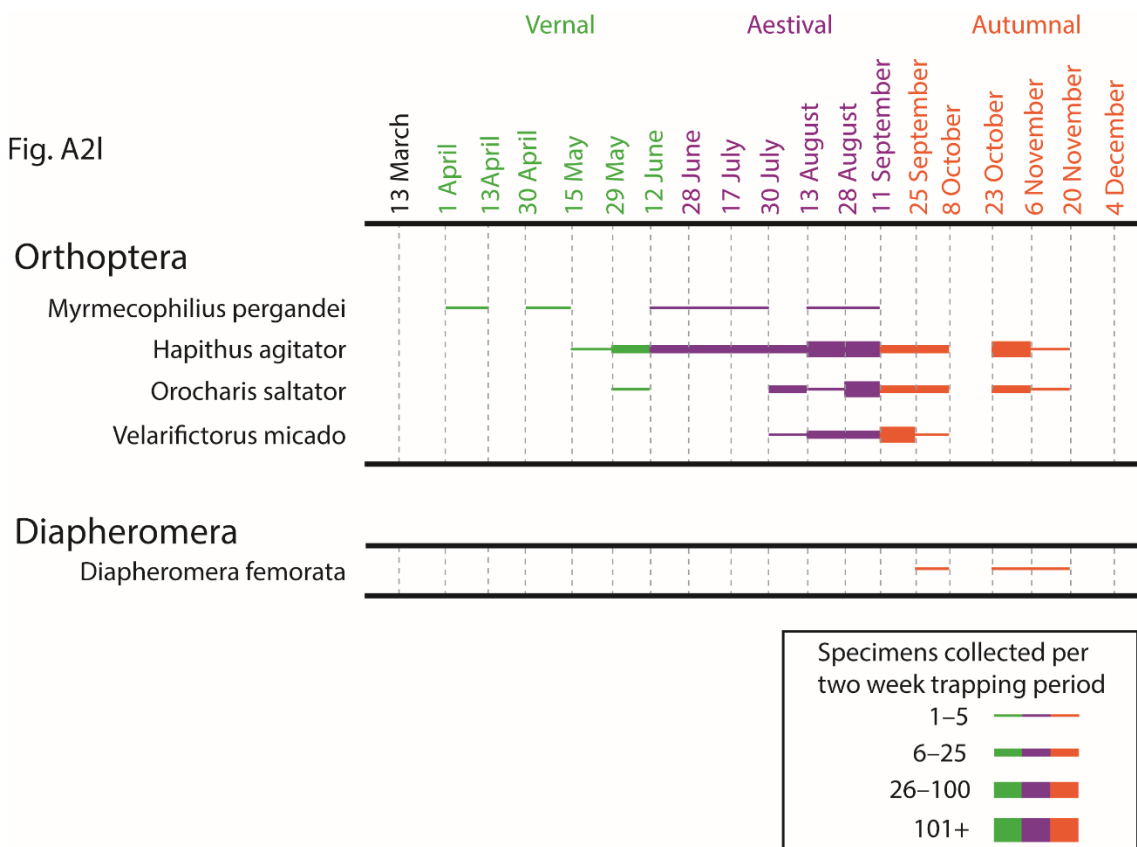


Figure A2I. Phenology of select collected orthopteroids.

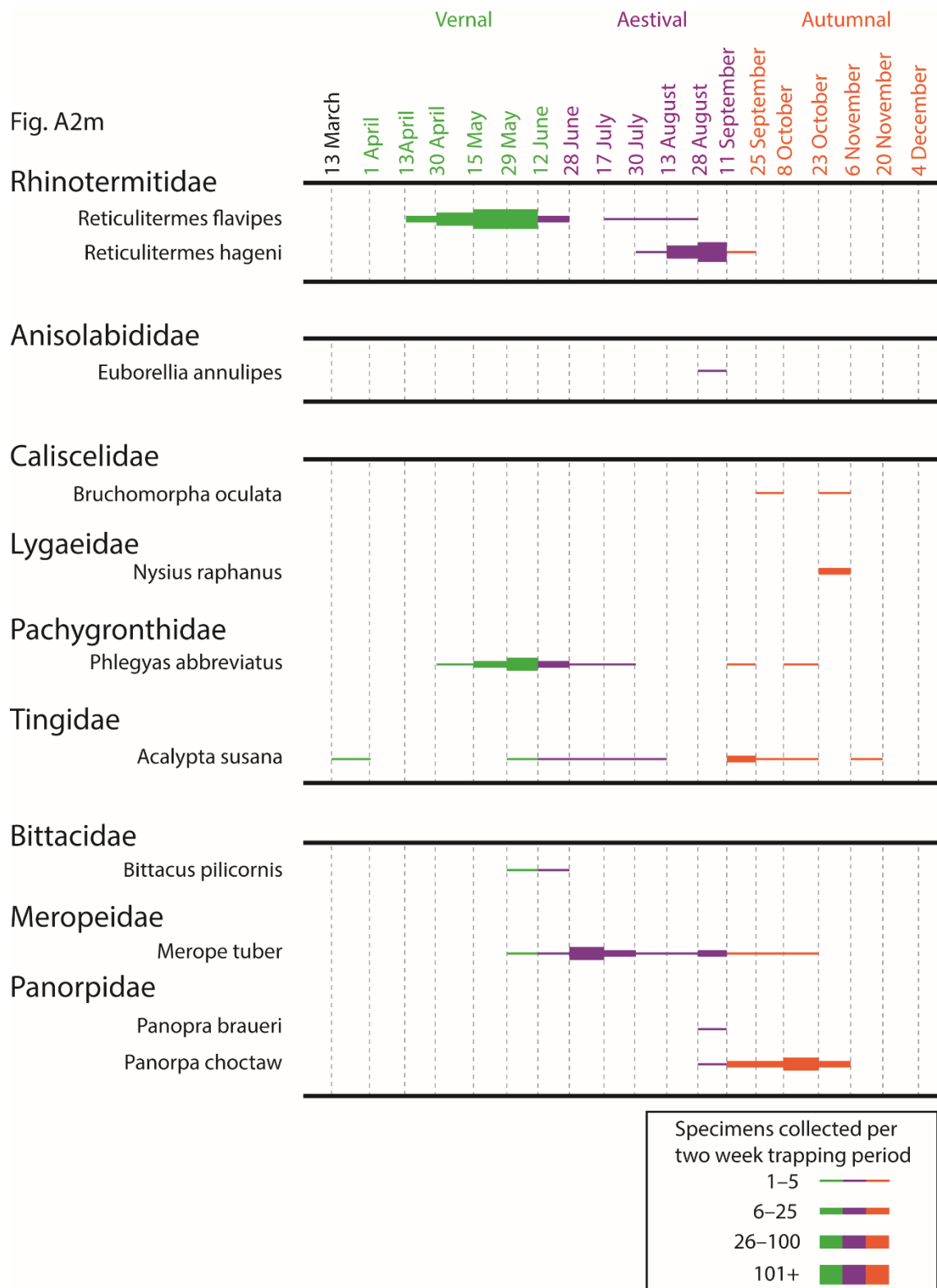


Figure A2m. Phenology of select collected Blattodea, Dermaptera, Hemiptera, and Mecoptera.

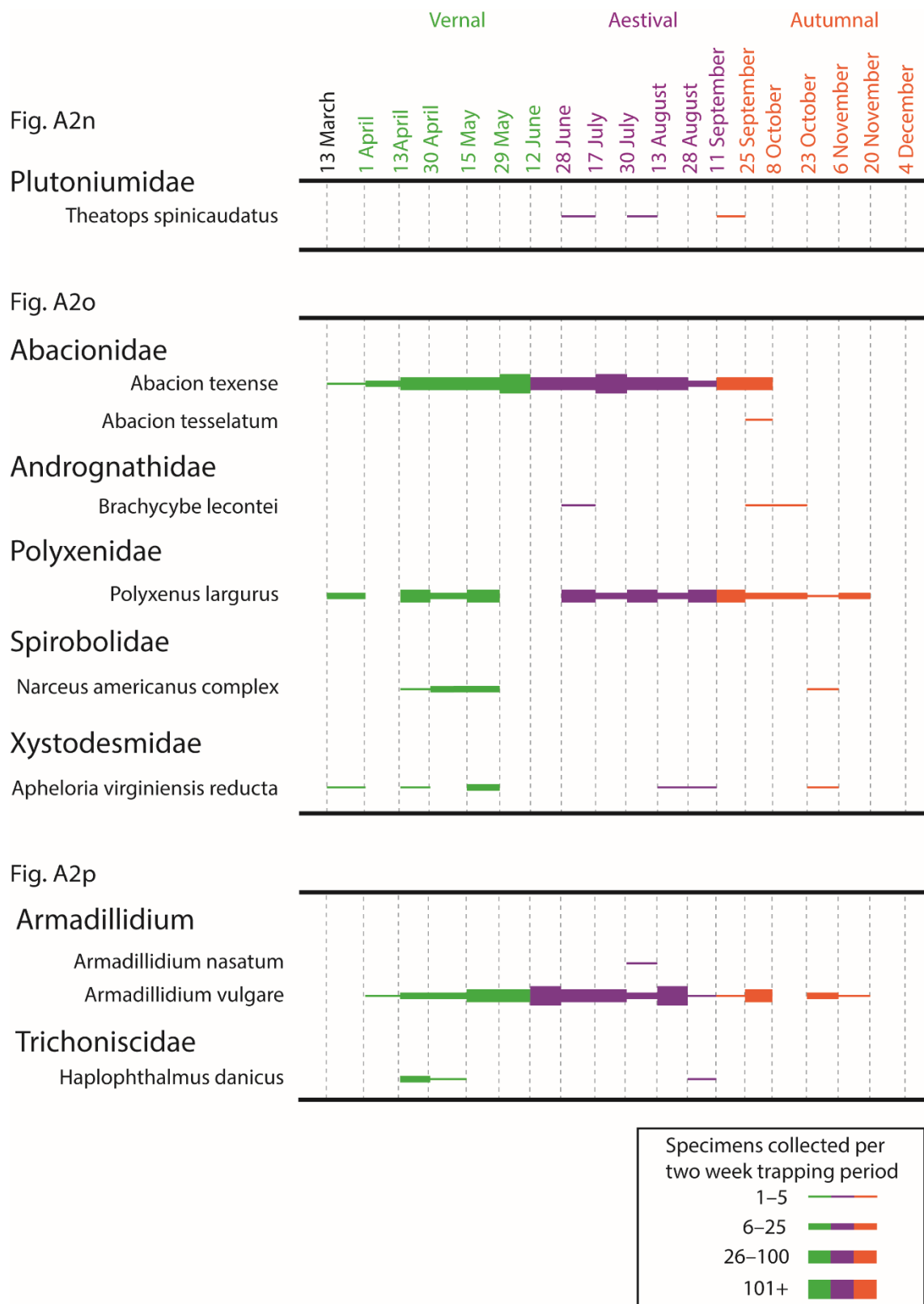


Figure A2n–p. Phenology of select collected Myriapoda and Isopoda. **Fig. A2n.** Chilopoda. **Fig. A2o.** Diplopoda. **Fig. A2p.** Isopoda.

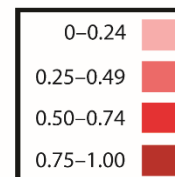
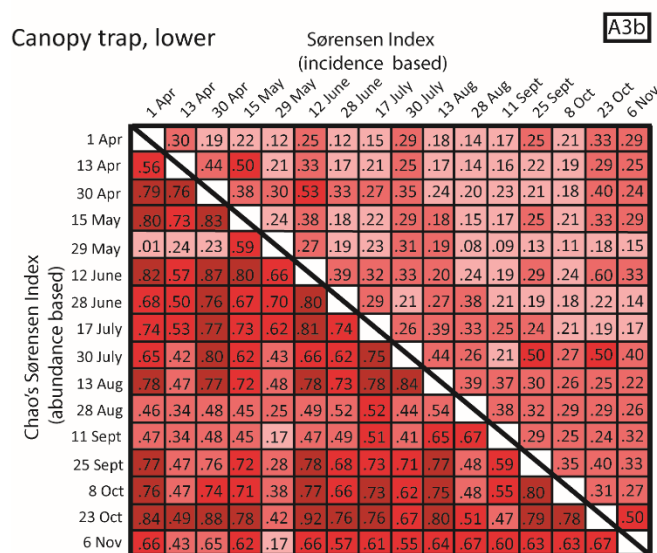
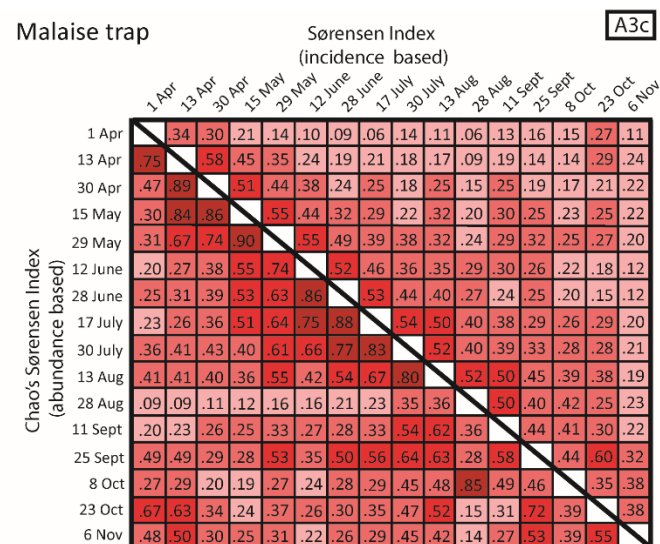
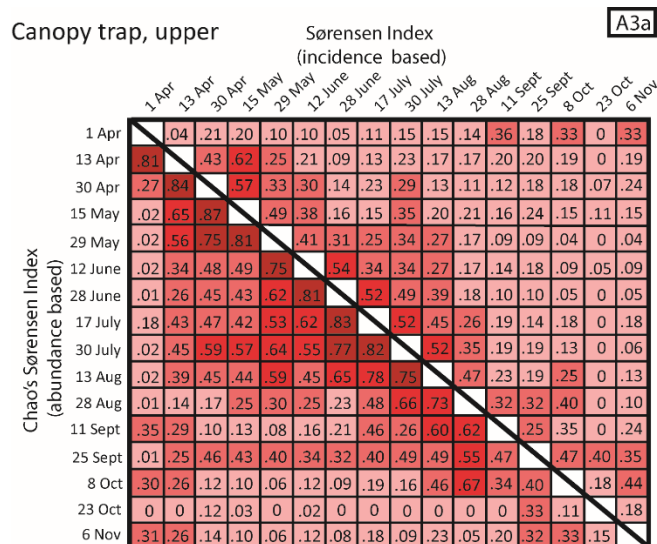


Figure A3. Sørensen and Chao's Sørensen Indices comparing similarity of trap catch by date. **Fig. A3a.** Upper canopy trap. **Fig. A3b.** Lower canopy trap. **Fig. A3c.** Malaise trap.

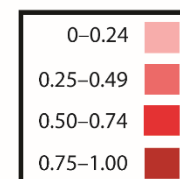
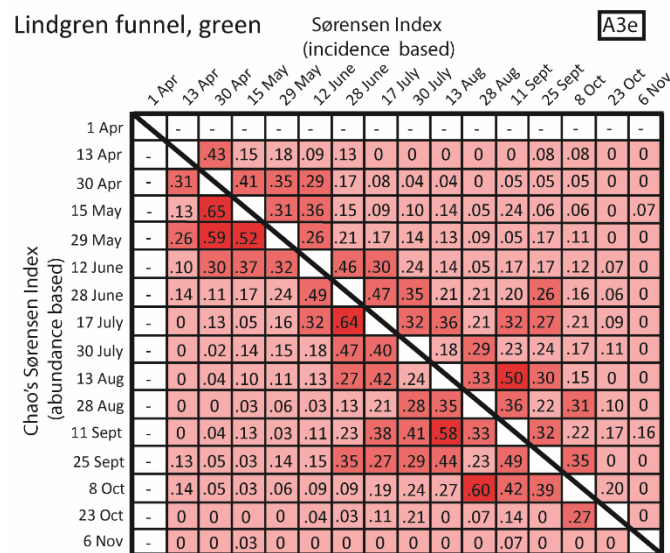
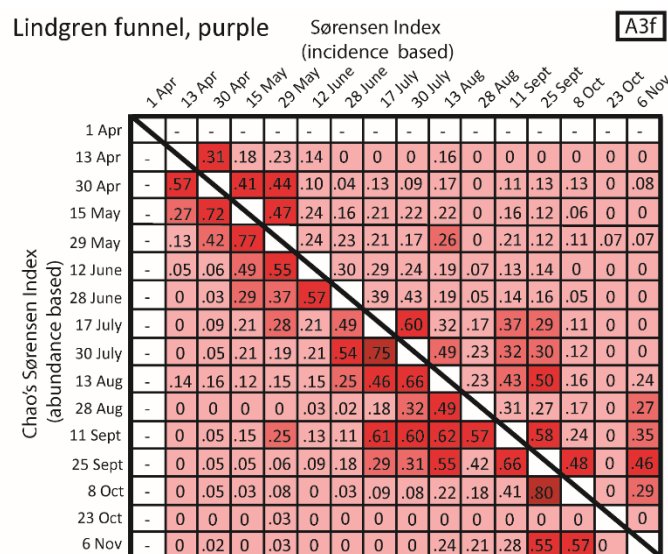
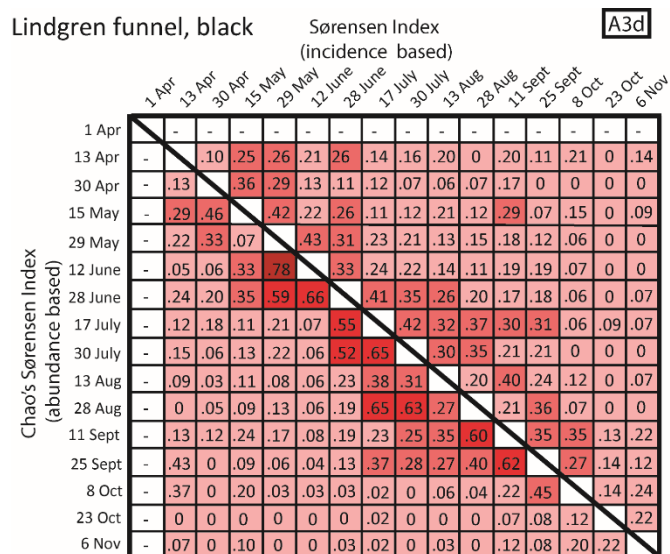
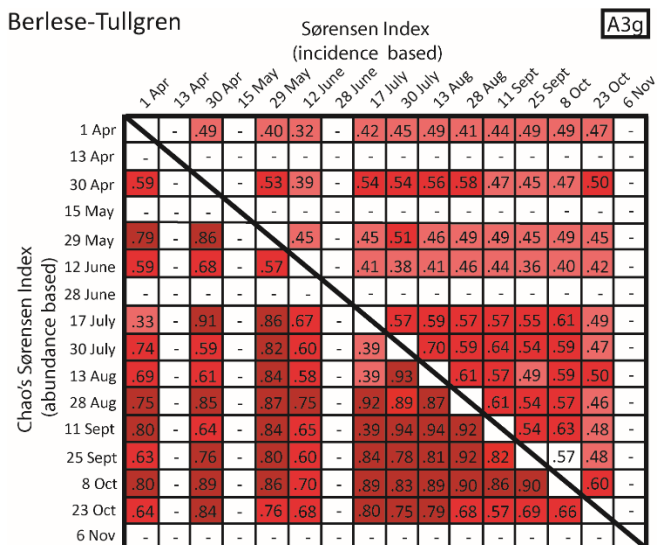
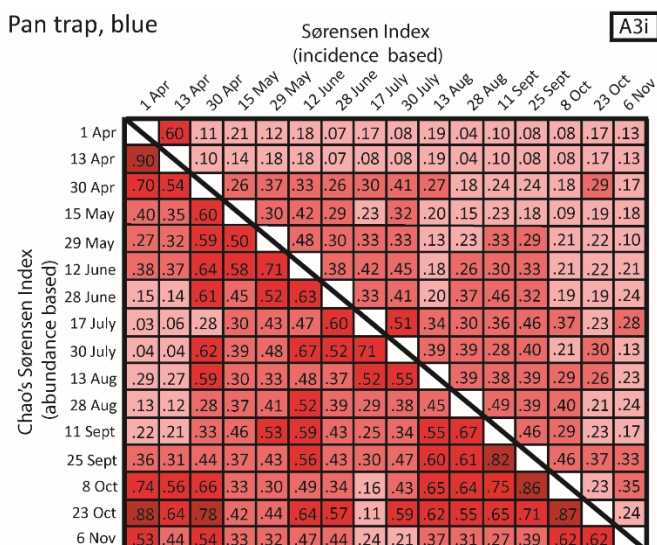


Figure A3 (cont.). Sørensen and Chao's Sørensen Indices comparing similarity of trap catch by date. **Fig. A3d.** Black Lindgren funnel trap. **Fig. A3e.** Green Lindgren funnel trap. **Fig. A3f.** Purple Lindgren funnel trap.

Berlese-Tullgren



Pan trap, blue



Pitfall

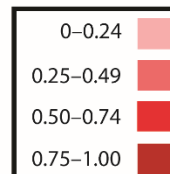
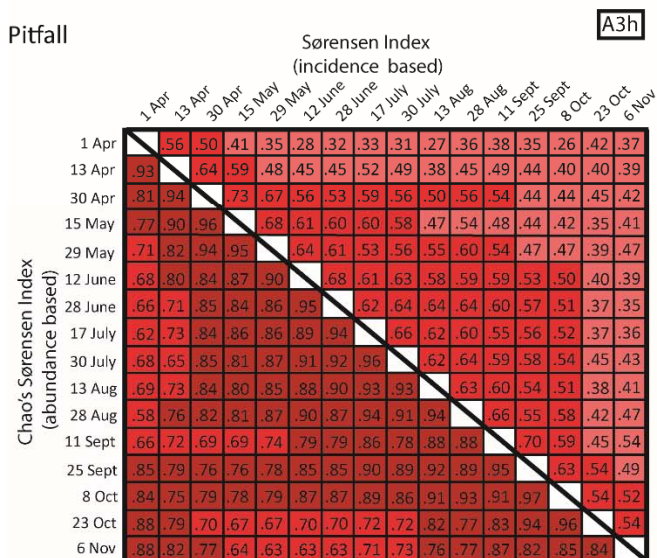
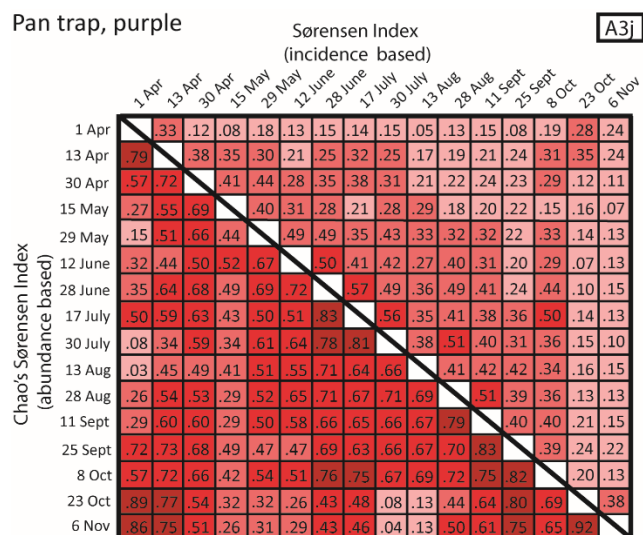
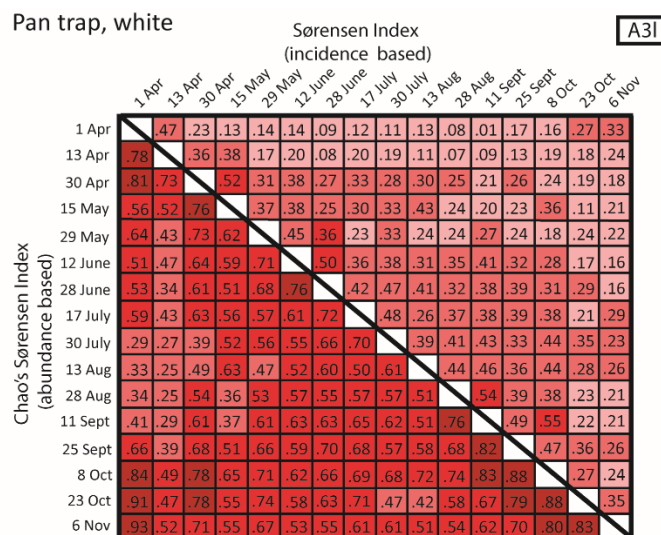


Figure A3 (cont.). Sørensen and Chao's Sørensen Indices comparing similarity of trap catch by date. **Fig. A3g.** Berlese-Tullgren extraction. Dashes indicate dates when no samples were collected **Fig. A3h.** Pitfall trap. **Fig. A3i.** Blue pan trap.

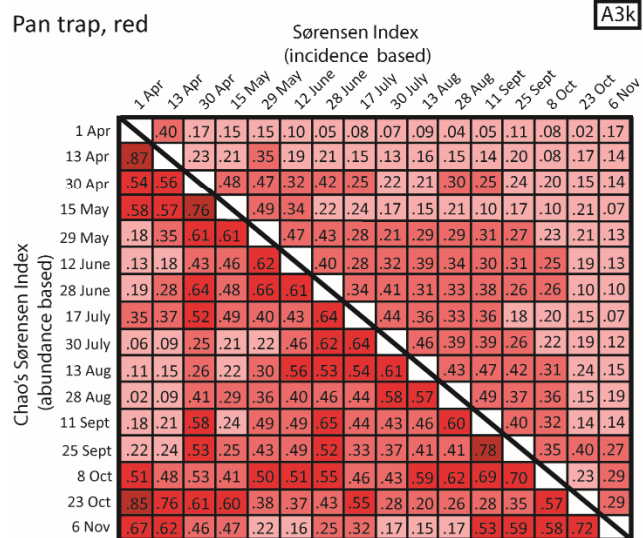
Pan trap, purple



Pan trap, white



Pan trap, red



Pan trap, yellow

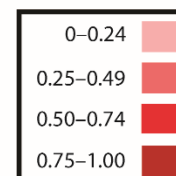
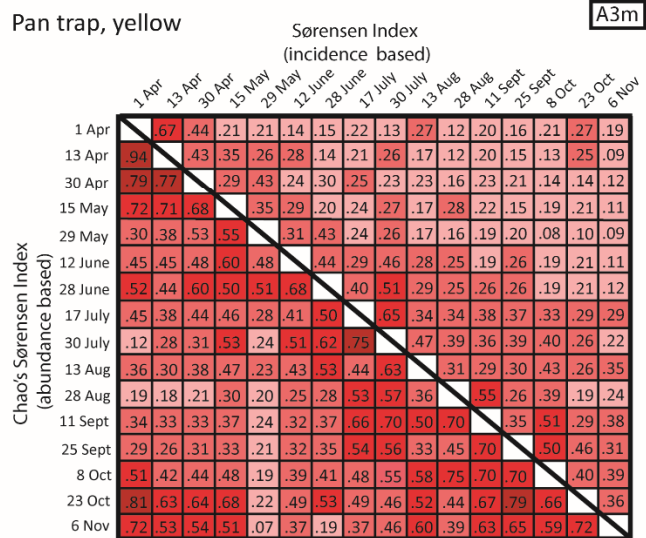


Figure A3 (cont.). Sørensen and Chao's Sørensen Indices comparing similarity of trap catch by date. **Fig. A3j.** Purple pan trap. **Fig. A3k.** Red pan trap. **Fig. A3l.** White pan trap. **Fig. A3m.** Yellow pan trap.

Chapter VII. First report of gynandromorphism in *Temnothorax curvispinosus* (Mayr, 1866) (Hymenoptera: Formicidae).

Abstract.

We report for the first time a *Temnothorax curvispinosus* (Mayr, 1866) ergatandromorph.

Body.

Gynandromorphism is when an organism possesses tissue that is genotypically and phenotypically male and female (Laugé 1985). A gynandromorph can have bilateral symmetry, in which one side is male and one is female, or be a mosaic, in which case male and female tissues are spread in patches across the body and may not be clearly defined (Campos et al 2011). While this phenomenon has been reported in vertebrates (Brodkorb 1935; Patten 1993), it is most commonly reported from invertebrates, especially insects (Turrisi & Foucart 2008).

Gynandromorphism has been described from 69 families of insects across 13 orders (Cui and Cai 2003). Within Hymenoptera the condition has been reported from Agaonidae (Pereira et al. 2003), Andrenidae (Xu & Cui 2007), Apidae (Wcislo et al. 2004), Braconidae (Whiting & Whiting 1927), Chalcididae (Haltead 1988), Colletidae (Wcislo et al. 2004), Diprionidae (Martini et al. 1999), Encyrtidae (Zhang & Zhu 2007), Halictidae (Wcislo et al. 2004), Ichneumonidae (Tarasco 1996) Megachilidae (Gerber and Akre 1969), Melittidae (Wcislo et al. 2004), Mutillidae (Turrisi & Foucart 2008), Scelionidae (Huggert 1977), Sphecidae (Schneider & Feitz 2003) Tenthredinidae (Peacock 1925), Trichogrammatidae (Beserra et al. 2003), and Vespidae (Turrisi & Borsato 2008).

However, the condition has most often been reported in Formicidae, with gynandromorphs described in *Acromyrmex octospinosus* (Reich) (Wheeler 1937), *Anergates atratulus* (Schenck) (Wheeler 1914), *Aphaenogaster picea* Wheeler (Wheeler 1903), *Camponotus (Colobopsis) albocinctus* (Ashmead) (Wheeler 1919), *Camponotus ligniperdus*

(Latreille) (Wheeler 1903) *Cardiocondyla batesi* Forel (Kugler 1983), *Cardiocondyla kagutsuchi* Terayama (Yoshizawa et al. 2009), *Cardiocondyla nigra* Forel (Wheeler 1914), *Diacamma* Mayr (Dobata et al. 2012), *Formica microgyna* Wheeler (Wheeler 1903), *Formica rufa* Linnaeus (Forel 1874; Forbes 1954), *Formica sanguinea* Latreille (Wheeler 1914), *Lasius (Acanthomyops) latipes* (Walsh) (Wheeler 1919), *Monomorium floricola* (Jerdon) (Donisthorpe 1929; Campos et al. 2011), *Monomorium pharaonis* (Linnaeus) (Berndt & Kremer 1983), *Myrmecia gulosa* (Fabricius) (Crosland et al. 1988) *Myrmica sabuleti* Meinert (Scupola 1994), *Myrmica scabrinodis* Nylander (Wheeler 1914), *Pheidole dentata* Mayr (Jones & Phillips Jr. 1985), *Pheidole inquilina* (Wheeler) (Wheeler 1903), *Pheidole morrisi* Forel (Yang & Abouheif 2011), *Pogonomyrmex occidentalis* (Cresson) (Taber & Francke 1986), *Polyergus rufescens* (Forel 1874; Forbes 1954); *Solenopsis aurea* Wheeler (Cokendolpher and Francke 1983), *Solenopsis fugax* (Latreille) (Wheeler 1914), *Solenopsis invicta* Buren (Hung et al. 1975), *Solenopsis quinquecuspis* (Forel) (Pitts 2002), *Stenamma* Westwood (Munsee 1994), *Temnothorax obturator* (Wheeler) (Wheeler 1903), *Tetramorium guineense* (Bernard) (Wheeler 1926), *Tetramorium simillimum* (Smith) (Wheeler 1903) and *Vollenhovia emeryi* Wheeler (Kubota 1984; Kinomura and Yamauchi 1994).

Because a cast system exists in ants, different combinations of male and female tissue can occur, for which Campos et al. (2011) proposed the names gynandromorph (queen-male), ergatandromorph (worker-male), and dynergatandromorph (soldier-male). “Inter caste” individuals, in which different female castes are combined (e.g., queen-worker [gynergatandromorph], queen-soldier [ergatogynandromorph], and worker-soldier [androergatogynandromorph]), also occur, but are not true gynandromorphs because both castes are female (Yang & Abouheif 2011).

Although found in numerous species as described above, the probability of encountering a gynandromorph is very low. Out of the 14,442 ant specimens examined and identified, only a single specimen displaying signs of gynandromorphy was collected.

We report for the first time a *Temnothorax curvispinosus* ergatandromorph. The specimen was collected in a purple pan trap between 15–29 May, 2013 in the Steel Creek Wilderness Area of the Buffalo National River in Newton County, Arkansas (36°02.231' N, 93°20.461' W) and is deposited in the University of Arkansas Arthropod Museum.

The specimen exhibits male characteristics on the right side of the head – darker brown pigmentation, enlarged eye, ocelli present, reduced mandible, and 12-segmented antennae – and pronotum – lighter sclerotization – (Fig. 1a) and female worker characteristics on the left side of the head – lighter yellow pigmentation, smaller eye, ocelli absent, larger mandible, and 11-segmented antennae – and pronotum – heavier, darker sclerotization (Fig. 1b). The remaining thoracic segments, including the prothoracic leg, and abdominal segments are characteristic of a female worker (Fig. 1c). The internal anatomy of the head and prothorax were not examined.



Figure 1. **A**, Head. Male tissue to the left, female worker tissue to the right; **B**, Head and prothorax, dorsum. Male tissue to the left, female worker tissue to the right; **C**, Profile, dextral. Male tissue can be seen on the head and pronotum; the rest of the body is composed of female worker tissue.

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**VIII. New records of *Orussus minutus* Middlekauff, 1983 (Hymenoptera: Orussidae)
represent a significant western range expansion**

Abstract.

Background

Orussus minutus is an uncommonly collected parasitoid sawfly known from the eastern United States.

New information

We report specimens *Orussus minutus* Middlekauff, 1983, from Arkansas, Iowa, Minnesota, and Manitoba, which represent new state and province records and significantly expand the known range of the species west from previous records; provide collection information for unpublished specimens housed in the United States National Museum collection, which includes new state records for West Virginia and Michigan; and report two specimens housed in the Biological Museum at Lund University that represent new state records for Connecticut.

Introduction.

Orussidae have long interested entomologists because of their parasitoid larvae, which are unique among non-apocritan Hymenoptera, phylogenetically important position between basal Hymenoptera ("Symphyta") and Apocrita, and because they are rarely collected (Middlekauff 1983, Pesarini and Turrisi 2003, Vilhelmsen 2003). Middlekauff (1983) provided an excellent review of the literature concerning the feeding biology and hosts of orussid larvae. Briefly summarized, a number of authors reported orussid larvae develop in wood (Harrington 1887a, Konow 1902, Gaulle 1906) and associate with beetle and sawfly larvae (Wachtl 1882,

Rudow 1909). Harrington (1887b) first hypothesized that orussid larvae may be parasitoids, though he considered it more likely they fed on wood. Rohwer (1912) and Burke (1918) provided convincing evidence that orussids are parasitoids as they reported *Orussus* larvae pupating in old cerambycid larval galleries and attacking buprestid larvae. Subsequent authors investigated oviposition behavior and larval feeding; they found that adult female orussids deposit eggs into frassfilled galleries of and directly onto larvae of wood-boring Coleoptera and Hymenoptera and that larval orussids feed upon those larvae (Cooper 1953, Rawlings 1957, Powell and Turner 1975). Currently, Orussidae are known or suspected to parasitize Buprestidae, Cerambycidae, Siricidae, and Xiphydriidae (Table 1).

Host family	Reference
Buprestidae	Wachtl 1882, Harrington 1887, Burke 1918, Ahnlund & Ronquist 2001, Vilhelmsen & Smith 2002
Cerambycidae	Rowher 1925, Hellrigl 1984 Ahnlund & Ronquist 2001
Siricidae	Gourlay 1951, Rawlings 1957, Vilhelmsen & Smith 2002
Xiphydriidae	Rudow 1909

Table 1. Known and suspected hosts of Orussidae.

Ashmead (1896) published the first phylogenetic hypothesis of Hymenoptera and placed Oryssidae (=Orussidae) transitionally between sawflies and other Hymenoptera. Recent phylogenetic analyses of morphological characters (Rasnitsyn 1988, Vilhelmsen 1997, Vilhelmsen 2000, Vilhelmsen 2001, Ronquist et al. 1999, Schulmeister 2003b), large molecular datasets and combined molecular and morphological datasets (Schulmeister 2003b, Heraty et al. 2011, Sharkey et al. 2011) have corroborated the placement of Orussidae (and Paroryssidae when fossil taxa are included) as sister to Apocrita. For relationships within Orussidae, the most robust phylogenetic analysis was produced by Vilhelmsen (2003). His analysis recovered most

genera as monophyletic, though Vilhelmsen abandoned the use of subfamilies and tribes, as “[e]nforcing a strictly cladistics classification at these levels would require recognition of many redundant taxa without enhancing the information content”.

Orussidae are uncommonly collected and rare in collections. For example, despite a cumulative 25,000 trapping hours (314 separate 1–2 week collection events) using Malaise traps over the last five years by the authors around Arkansas, no additional specimens beyond the three reported herein were captured with this trapping method and David Smith (USDA, SEL), who has had success collecting orussids in Malaise traps (e.g., Smith 2006, Smith 2008, Barrows and Smith 2014), has only collected 33 specimens of *O. minutus* in 35 years of collecting with an average of 15 Malaise traps set per year (David R. Smith, pers. comm. 18 August 2015).

Additionally, new species continue to be described, even in heavily collected areas such as California (e.g., Vilhelmsen 2005, Blank et al. 2010, Vilhelmsen et al. 2014). Several species are known only from one or a few localities and specimens and the known ranges of many species continue to expand as new specimens are collected (Ahnlund and Ronquist 2001, Vilhelmsen and Smith 2002, Pesarini and Turrisi 2003, Pesarini and Turrisi 2006, Choi and Suh 2011).

Orussus is represented five species in North America north of Mexico: *O. occidentalis* (Cresson, 1879) has been reported from Southern British Columbia east to Ontario, south in the western United States to southern California, Nevada, and New Mexico; *O. thoracicus* (Ashmead, 1898) has been reported from Colorado, Washington, Oregon, and California; *O. sayii* (Westwood, 1835) has been reported from Ontario south to Louisiana, west to Indiana; *O. terminalis* (Newman, 1838) has been reported from New England and Ontario west to Iowa and Illinois, south to Maryland; and *O. minutus* (Middlekauff, 1983) has been reported from New

York to Georgia west to Illinois (Middlekauff 1983, Vilhelmsen 2003, Blank et al. 2010, Vilhelmsen et al. 2013).

Materials and methods.

Two orussids (1 male, 1 female) were collected along the Buffalo National River in the lower collector of an aerial SLAM (sea-land-air-Malaise) trap (MegaView Science Co., Ltd., Taichung, Taiwan) and a black multifunnel trap (ChemTica International, S.A., Heredia, Costa Rica); a third specimen (1 female) was collected via aerial netting in the Kessler Mountain Reserve. Both localities are mixed secondary deciduous forest dominated by oak and hickory that were logged approximately 80–100 years ago. Specimens were identified to species using published keys (Middlekauff 1983, Vilhelmsen et al. 2014) and have been deposited in the University of Arkansas Arthropod Museum.

Stereomicrographs of the Arkansas specimens were taken with a Cannon EOS 40D camera (Tokyo, Japan) attached using a Diagnostic Instruments DD20NLT 2.0X camera mount (Sterling Heights, Michigan, USA) to a Nikon SMZ1500 stereomicroscope (Tokyo, Japan). The micrographs were processed and final plates arranged in Adobe Illustrator (San Jose, California, USA).

DNA of one Arkansas specimen (MS 13-0413-047, #138295) was sequenced for comparison with previously characterized *Orussus*. Genomic DNA was extracted from a single mid-leg using the Qiagen DNeasy Tissue kit (Qiagen, Inc., Valencia, California), following manufacturer's instructions. PCR was conducted using the primers LR-J-13017 (5'-TTACGCTGTTATCCTAA-3') and LR-N-13398 (5'-CACCTGTTTAACAAAAACAT-3') (Kambhampati and Smith 1995), which amplify an approximately 415 bp portion of the 16S

rRNA region of the mitochondrial genome. Reaction conditions were 94°C for 2 min, followed by 40 cycles of 94°C for 45 s, 48°C for 1 min, and 72°C for 1 min, with a final 5 min extension step at 72°C. Amplified DNA was purified, concentrated with PES 30k centrifugal filter devices (VWR, Radnor, PA) and sent for direct sequencing in both directions (Eurofins MWG Operon, Huntsville, Alabama).

David R. Smith kindly provided label information for specimens housed in the United States National Museum; previously unpublished specimens are reported herein. Additional unpublished specimens were found by searching the databased collection of Lund University Biological Museum (Lund University 2015), BugGuide (Hatfield 2008, Alexander 2011, Liberta 2014, Zhang 2014), and Flickr (King 2014).

Published locality data for Figure 3 was compiled from Cooper (1953), Middlekauff (1983), Smith (2006), Barrows and Smith (2014).

Institution abbreviations follow Evenhuis (2015) and are as follows: United States National Museum (USNM), University of Arkansas Arthropod Museum (UAAM), Lund University, Sweden (MZLU).

Taxon treatment.

Orussus minutus Middlekauff, 1983

Materials

- a. scientificName: *Orussus minutus* Middlekauff, 1983; kingdom: Animalia; phylum: Arthropoda; class: Insecta; order: Hymenoptera; family: Orussidae; genus: *Orussus*; specificEpithet: *minutus*; scientificNameAuthorship: Middlekauff, 1983; country: United States; countryCode: US; stateProvince: Arkansas; county: Newton; locality: Buffalo National River, Steel Creek; locationRemarks: 80-100 year old mature second-growth Eastern mixed deciduous forest dominated by oak (*Quercus*) and hickory (*Carya*); verbatimCoordinates: 36°02.218' N, 93°20.439 W; decimalLatitude: 36.036967; decimalLongitude: -93.34065; georeferenceProtocol: GPS; samplingProtocol: black Lindgren multifunnel trap; eventDate: 201313-4-13; individualCount: 1; lifeStage: adult; catalogNumber: 138295; recordedBy: Michael J Skvarla; identifiedBy: Michael J. Skvarla; dateIdentified: 2014; language: en; collectionID: MS 13-0413-047; institutionCode: UAAM; basisOfRecord: PreservedSpecimen
- b. scientificName: *Orussus minutus* Middlekauff, 1983; kingdom: Animalia; phylum: Arthropoda; class: Insecta; order: Hymenoptera; family: Orussidae; genus: *Orussus*; specificEpithet: *minutus*; scientificNameAuthorship: Middlekauff, 1983; country: United States; countryCode: US; stateProvince: Arkansas; county: Newton; locality: Buffalo National River, Steel Creek; locationRemarks: 80-100 year old mature second-growth Eastern mixed deciduous forest dominated by oak (*Quercus*) and hickory (*Carya*); verbatimCoordinates: 36°02.314' N, 93°20.425 W; decimalLatitude: 36.038567; decimalLongitude: -93.34041; georeferenceProtocol: GPS; samplingProtocol: SLAM canopy trap, lower collector; eventDate: 201313-4-13; individualCount: 1; lifeStage: adult; catalogNumber: 138296; recordedBy: Michael J Skvarla; identifiedBy: Michael J. Skvarla; dateIdentified: 2014; language: en; collectionID: MS 13-0413-060; institutionCode: UAAM; basisOfRecord: PreservedSpecimen

- c. scientificName: *Orussus minutus* Middlekauff, 1983; kingdom: Animalia; phylum: Arthropoda; class: Insecta; order: Hymenoptera; family: Orussidae; genus: *Orussus*; specificEpithet: *minutus*; scientificNameAuthorship: Middlekauff, 1983; country: United States; countryCode: US; stateProvince: Arkansas; county: Washington; locality: Fayetteville, Kessler Mountain Reserve, Wino Trail; locationRemarks: 80-100 year old mature second-growth Eastern mixed deciduous forest dominated by oak (*Quercus*) and hickory (*Carya*); verbatimCoordinates: 36°02'19.45" N, 94°13'01.98" W; decimalLatitude: 36.038611; decimalLongitude: -94.216944; georeferenceProtocol: GoogleEarth; samplingProtocol: hand collected with net; eventDate: 41755.00; individualCount: 1; lifeStage: adult; recordedBy: Amber Tripodi; identifiedBy: Michael J. Skvarla; dateIdentified: 2014; language: en; institutionCode: UAAM; basisOfRecord: PreservedSpecimen
- d. scientificName: *Orussus minutus* Middlekauff, 1983; kingdom: Animalia; phylum: Arthropoda; class: Insecta; order: Hymenoptera; family: Orussidae; genus: *Orussus*; specificEpithet: *minutus*; scientificNameAuthorship: Middlekauff, 1983; country: United States; countryCode: US; stateProvince: Virginia; county: Warren; locality: Skyland Estates; locationRemarks: 4 km NNW of Linden; georeferenceProtocol: label; samplingProtocol: Malaise trap; eventDate: 1985-4-20/1985-4-27; individualCount: 1; lifeStage: adult; recordedBy: T. P. Nuhn; identifiedBy: David R. Smith; language: en; institutionCode: USNM; basisOfRecord: PreservedSpecimen
- e. scientificName: *Orussus minutus* Middlekauff, 1983; kingdom: Animalia; phylum: Arthropoda; class: Insecta; order: Hymenoptera; family: Orussidae; genus: *Orussus*; specificEpithet: *minutus*; scientificNameAuthorship: Middlekauff, 1983; country: United States; countryCode: US; stateProvince: Virginia; county: Warren; locality: Skyland Estates; locationRemarks: 4 km NNW of Linden; georeferenceProtocol: label; samplingProtocol: Malaise trap; eventDate: 1996-4-27/1996-5-12; individualCount: 1; lifeStage: adult; recordedBy: T. P. Nuhn; identifiedBy: David R. Smith; language: en; institutionCode: USNM; basisOfRecord: PreservedSpecimen
- f. scientificName: *Orussus minutus* Middlekauff, 1983; kingdom: Animalia; phylum: Arthropoda; class: Insecta; order: Hymenoptera; family: Orussidae; genus: *Orussus*; specificEpithet: *minutus*; scientificNameAuthorship: Middlekauff, 1983; country: United States; countryCode: US; stateProvince: Virginia; locality: Great Dismal Swamp National Wildlife Refuge; georeferenceProtocol: label; samplingProtocol: Malaise trap; eventDate: 1965-4-16/1965-4-17; individualCount: 1; lifeStage: adult; recordedBy: P. J. Spangler; identifiedBy: David R. Smith; language: en; institutionCode: USNM; basisOfRecord: PreservedSpecimen
- g. scientificName: *Orussus minutus* Middlekauff, 1983; kingdom: Animalia; phylum: Arthropoda; class: Insecta; order: Hymenoptera; family: Orussidae; genus: *Orussus*; specificEpithet: *minutus*; scientificNameAuthorship: Middlekauff, 1983; country: United States; countryCode: US; stateProvince: Virginia; county: Louisa; locationRemarks: 4 mi south of Cuckoo; georeferenceProtocol: label; samplingProtocol: Malaise trap; eventDate: 1989-4-26/1989-5-12; individualCount: 1; lifeStage: adult; recordedBy: J. Kloke & D. R. Smith; identifiedBy: David R. Smith; language: en; institutionCode: USNM; basisOfRecord: PreservedSpecimen
- h. scientificName: *Orussus minutus* Middlekauff, 1983; kingdom: Animalia; phylum: Arthropoda; class: Insecta; order: Hymenoptera; family: Orussidae; genus: *Orussus*; specificEpithet: *minutus*; scientificNameAuthorship: Middlekauff, 1983; country: United States; countryCode: US; stateProvince: Virginia; county: Louisa; locationRemarks: 4 mi south of Cuckoo; georeferenceProtocol: label; samplingProtocol: Malaise trap; eventDate: 1989-5-27/1989-6-7; individualCount: 1; lifeStage: adult; recordedBy: J. Kloke & D. R. Smith; identifiedBy: David R. Smith; language: en; institutionCode: USNM; basisOfRecord: PreservedSpecimen
- i. scientificName: *Orussus minutus* Middlekauff, 1983; kingdom: Animalia; phylum: Arthropoda; class: Insecta; order: Hymenoptera; family: Orussidae; genus: *Orussus*; specificEpithet: *minutus*; scientificNameAuthorship: Middlekauff, 1983; country: United States; countryCode: US; stateProvince: Virginia; county: Louisa; locationRemarks: 4 mi south of Cuckoo; georeferenceProtocol: label; samplingProtocol: Malaise trap; eventDate: 1988-3-19/1988-4-11; individualCount: 1; lifeStage: adult; recordedBy: J. Kloke & D. R. Smith; identifiedBy: David R. Smith; language: en; institutionCode: USNM; basisOfRecord: PreservedSpecimen
- j. scientificName: *Orussus minutus* Middlekauff, 1983; kingdom: Animalia; phylum: Arthropoda; class: Insecta; order: Hymenoptera; family: Orussidae; genus: *Orussus*; specificEpithet: *minutus*; scientificNameAuthorship: Middlekauff, 1983; country: United States; countryCode: US; stateProvince: Virginia; county: Fairfax; locality: Holmes Run; locationRemarks: ~1/4 mi NW jct. Gallows Rd & I-495; verbatimCoordinates: 38°50'N, 77°12'W; georeferenceProtocol: label; samplingProtocol: Malaise trap; eventDate: 1990-4-22/1990-4-28; individualCount: 1; lifeStage: adult; recordedBy: D. R. Smith; identifiedBy: David R. Smith; language: en; institutionCode: USNM; basisOfRecord: PreservedSpecimen
- k. scientificName: *Orussus minutus* Middlekauff, 1983; kingdom: Animalia; phylum: Arthropoda; class: Insecta; order: Hymenoptera; family: Orussidae; genus: *Orussus*; specificEpithet: *minutus*; scientificNameAuthorship: Middlekauff, 1983; country: United States; countryCode: US; stateProvince: Virginia; county: Fairfax; locality: Holmes Run; locationRemarks: ~1/4 mi NW jct. Gallows Rd & I-496; verbatimCoordinates: 38°50'N, 77°12'W; georeferenceProtocol: label; samplingProtocol: Malaise trap; eventDate: 1990-3-11/1990-30-17; individualCount: 1; lifeStage: adult; recordedBy: D. R. Smith; identifiedBy: David R. Smith; language: en; institutionCode: USNM; basisOfRecord: PreservedSpecimen
- l. scientificName: *Orussus minutus* Middlekauff, 1983; kingdom: Animalia; phylum: Arthropoda; class: Insecta; order: Hymenoptera; family: Orussidae; genus: *Orussus*; specificEpithet: *minutus*; scientificNameAuthorship: Middlekauff, 1983; country: United States; countryCode: US; stateProvince: Virginia; county: Fairfax; locality: Holmes Run; locationRemarks: ~1/4 mi NW jct. Gallows Rd & I-497; verbatimCoordinates: 38°50'N, 77°12'W; georeferenceProtocol: label; samplingProtocol: Malaise trap; eventDate: 2008-4-13/2008-4-19; individualCount: 1; lifeStage: adult; recordedBy: D. R. Smith; identifiedBy: David R. Smith; language: en; institutionCode: USNM; basisOfRecord: PreservedSpecimen
- m. scientificName: *Orussus minutus* Middlekauff, 1983; kingdom: Animalia; phylum: Arthropoda; class: Insecta; order: Hymenoptera; family: Orussidae; genus: *Orussus*; specificEpithet: *minutus*; scientificNameAuthorship: Middlekauff, 1983; country: United States; countryCode: US; stateProvince: Virginia; county: Clarke; locality: University of Virginia Blandy Experiment Farm; locationRemarks: 2 mi south of Boyce; verbatimCoordinates: 39°05'N, 78°10'W; georeferenceProtocol: label; samplingProtocol: Malaise trap; eventDate: 1992-5-2/1992-5-16; individualCount: 1; lifeStage: adult; recordedBy: D. R. Smith; identifiedBy: David R. Smith; language: en; institutionCode: USNM; basisOfRecord: PreservedSpecimen
- n. scientificName: *Orussus minutus* Middlekauff, 1983; kingdom: Animalia; phylum: Arthropoda; class: Insecta; order: Hymenoptera; family: Orussidae; genus: *Orussus*; specificEpithet: *minutus*; scientificNameAuthorship: Middlekauff, 1983;

[illegible]

- z. scientificName: *Orussus minutus* Middlekauff, 1983; kingdom: Animalia; phylum: Arthropoda; class: Insecta; order: Hymenoptera; family: Orussidae; genus: *Orussus*; specificEpithet: *minutus*; scientificNameAuthorship: Middlekauff, 1983; country: United States; countryCode: US; stateProvince: Virginia; county: Essex; locationRemarks: 1 mi southeast of Dunnsville; verbatimCoordinates: 37°52'N, 76°48'W; georeferenceProtocol: label; samplingProtocol: Malaise trap; eventDate: 1999-5-6/1999-5-20; individualCount: 1; lifeStage: adult; recordedBy: D. R. Smith; identifiedBy: David R. Smith; language: en; institutionCode: USNM; basisOfRecord: PreservedSpecimen
- aa. scientificName: *Orussus minutus* Middlekauff, 1983; kingdom: Animalia; phylum: Arthropoda; class: Insecta; order: Hymenoptera; family: Orussidae; genus: *Orussus*; specificEpithet: *minutus*; scientificNameAuthorship: Middlekauff, 1983; country: United States; countryCode: US; stateProvince: Virginia; county: Fairfax; locality: Great Falls Park; verbatimCoordinates: 38°59.4'N, 77°15.26'W; georeferenceProtocol: label; samplingProtocol: Malaise trap; eventDate: 2007-4-19/2007-5-2; individualCount: 1; lifeStage: adult; recordedBy: D. R. Smith; identifiedBy: David R. Smith; language: en; institutionCode: USNM; basisOfRecord: PreservedSpecimen
- ab. scientificName: *Orussus minutus* Middlekauff, 1983; kingdom: Animalia; phylum: Arthropoda; class: Insecta; order: Hymenoptera; family: Orussidae; genus: *Orussus*; specificEpithet: *minutus*; scientificNameAuthorship: Middlekauff, 1983; country: United States; countryCode: US; stateProvince: West Virginia; county: Hardy; locationRemarks: 3 mi northeast of Mathias; verbatimCoordinates: 38°55'N, 78°49'W; georeferenceProtocol: label; samplingProtocol: Malaise trap; eventDate: 2000-5-1/2000-5-15; individualCount: 1; lifeStage: adult; recordedBy: D. R. Smith; identifiedBy: David R. Smith; language: en; institutionCode: USNM; basisOfRecord: PreservedSpecimen
- ac. scientificName: *Orussus minutus* Middlekauff, 1983; kingdom: Animalia; phylum: Arthropoda; class: Insecta; order: Hymenoptera; family: Orussidae; genus: *Orussus*; specificEpithet: *minutus*; scientificNameAuthorship: Middlekauff, 1983; country: United States; countryCode: US; stateProvince: West Virginia; county: Hardy; locationRemarks: 3 mi northeast of Mathias; verbatimCoordinates: 38°55'N, 78°49'W; georeferenceProtocol: label; samplingProtocol: Malaise trap; eventDate: 2001-4-1/2001-5-14; individualCount: 1; lifeStage: adult; recordedBy: D. R. Smith; identifiedBy: David R. Smith; language: en; institutionCode: USNM; basisOfRecord: PreservedSpecimen
- ad. scientificName: *Orussus minutus* Middlekauff, 1983; kingdom: Animalia; phylum: Arthropoda; class: Insecta; order: Hymenoptera; family: Orussidae; genus: *Orussus*; specificEpithet: *minutus*; scientificNameAuthorship: Middlekauff, 1983; country: United States; countryCode: US; stateProvince: West Virginia; county: Hardy; locationRemarks: 3 mi northeast of Mathias; verbatimCoordinates: 38°55'N, 78°49'W; georeferenceProtocol: label; samplingProtocol: Malaise trap; eventDate: 2007-5-4/2007-5-21; individualCount: 1; lifeStage: adult; recordedBy: D. R. Smith; identifiedBy: David R. Smith; language: en; institutionCode: USNM; basisOfRecord: PreservedSpecimen
- ae. scientificName: *Orussus minutus* Middlekauff, 1983; kingdom: Animalia; phylum: Arthropoda; class: Insecta; order: Hymenoptera; family: Orussidae; genus: *Orussus*; specificEpithet: *minutus*; scientificNameAuthorship: Middlekauff, 1983; country: United States; countryCode: US; stateProvince: West Virginia; county: Hardy; locationRemarks: 3 mi northeast of Mathias; verbatimCoordinates: 38°55'N, 78°49'W; georeferenceProtocol: label; samplingProtocol: Malaise trap; eventDate: 2007-5-22/2007-6-7; individualCount: 1; lifeStage: adult; recordedBy: D. R. Smith; identifiedBy: David R. Smith; language: en; institutionCode: USNM; basisOfRecord: PreservedSpecimen
- af. scientificName: *Orussus minutus* Middlekauff, 1983; kingdom: Animalia; phylum: Arthropoda; class: Insecta; order: Hymenoptera; family: Orussidae; genus: *Orussus*; specificEpithet: *minutus*; scientificNameAuthorship: Middlekauff, 1983; country: United States; countryCode: US; stateProvince: West Virginia; county: Hardy; locationRemarks: 3 mi northeast of Mathias; verbatimCoordinates: 38°55'N, 78°49'W; georeferenceProtocol: label; samplingProtocol: Malaise trap; eventDate: 2008-5-30/2008-6-17; individualCount: 1; lifeStage: adult; recordedBy: D. R. Smith; identifiedBy: David R. Smith; language: en; institutionCode: USNM; basisOfRecord: PreservedSpecimen
- ag. scientificName: *Orussus minutus* Middlekauff, 1983; kingdom: Animalia; phylum: Arthropoda; class: Insecta; order: Hymenoptera; family: Orussidae; genus: *Orussus*; specificEpithet: *minutus*; scientificNameAuthorship: Middlekauff, 1983; country: United States; countryCode: US; stateProvince: West Virginia; county: Tucker; locality: Fernow Experimental Forest; verbatimCoordinates: 39°03'N, 79°40'W; georeferenceProtocol: label; samplingProtocol: Malaise trap; eventDate: 1993-4-30/1993-5-10; individualCount: 1; lifeStage: adult; recordedBy: E. M. Barrows; language: en; institutionCode: USNM; basisOfRecord: PreservedSpecimen
- ah. scientificName: *Orussus minutus* Middlekauff, 1983; kingdom: Animalia; phylum: Arthropoda; class: Insecta; order: Hymenoptera; family: Orussidae; genus: *Orussus*; specificEpithet: *minutus*; scientificNameAuthorship: Middlekauff, 1983; country: United States; countryCode: US; stateProvince: Maryland; county: Montgomery; locality: Plimmers Island; georeferenceProtocol: label; samplingProtocol: hand collected with net; eventDate: 1971-4-11; individualCount: 5; lifeStage: adult; behavior: specimens taken on trunk of dead, standing, barked samplings, trunk diam. 2"; recordedBy: K. V. Krombein; associatedReferences: Smith, D.R. 2008. Hymenoptera (Insecta) of Plimmers Island, Maryland: Symphyta and selected families of Apocrita. Bulletin of the Biological Society of Washington, 15(1): 160–167; identifiedBy: David R. Smith; language: en; institutionCode: USNM; basisOfRecord: PreservedSpecimen
- ai. scientificName: *Orussus minutus* Middlekauff, 1983; kingdom: Animalia; phylum: Arthropoda; class: Insecta; order: Hymenoptera; family: Orussidae; genus: *Orussus*; specificEpithet: *minutus*; scientificNameAuthorship: Middlekauff, 1983; country: United States; countryCode: US; stateProvince: Michigan; county: Wayne; locality: Grosse Ile; georeferenceProtocol: label; eventDate: 1957-5-25; individualCount: 1; lifeStage: adult; recordedBy: Geo. Steyskal; language: en; institutionCode: USNM; basisOfRecord: PreservedSpecimen
- aj. scientificName: *Orussus minutus* Middlekauff, 1983; kingdom: Animalia; phylum: Arthropoda; class: Insecta; order: Hymenoptera; family: Orussidae; genus: *Orussus*; specificEpithet: *minutus*; scientificNameAuthorship: Middlekauff, 1983; country: United States; countryCode: US; stateProvince: Michigan; county: Washtenaw; georeferenceProtocol: label; eventDate: 1967-6-10; individualCount: 1; lifeStage: adult; recordedBy: R. W. Carlson; language: en; institutionCode: USNM; basisOfRecord: PreservedSpecimen
- ak. scientificName: *Orussus minutus* Middlekauff, 1983; kingdom: Animalia; phylum: Arthropoda; class: Insecta; order: Hymenoptera; family: Orussidae; genus: *Orussus*; specificEpithet: *minutus*; scientificNameAuthorship: Middlekauff, 1983; country: United States; countryCode: US; stateProvince: Indiana; county: Tippecanoe; locality: West Lafayette; georeferenceProtocol: label; samplingProtocol: hand collected; eventDate: 1970-5-5; individualCount: 1; lifeStage: adult;

behavior: collected in flight; recordedBy: M. & N. Deyrup; identifiedBy: David R. Smith; language: en; institutionCode: USNM; basisOfRecord: PreservedSpecimen

- al. scientificName: *Orussus minutus* Middlekauff, 1983; kingdom: Animalia; phylum: Arthropoda; class: Insecta; order: Hymenoptera; family: Orussidae; genus: *Orussus*; specificEpithet: *minutus*; scientificNameAuthorship: Middlekauff, 1983; country: United States; countryCode: US; stateProvince: Indiana; county: Tippecanoe; locality: West Lafayette; georeferenceProtocol: label; eventDate: 1981-4-16; individualCount: 1; lifeStage: adult; behavior: collected from branches of *Acer saccharum*; recordedBy: M. & N. Deyrup; associatedReferences: Deyrup, M.A. 1984. A maple wood wasp, *Xiphydria maculate*, and its insect enemies (Hymenoptera: Xiphydriidae). Great Lakes Entomologist, 17: 17–28. [referred to as "*Orussus* sp."]; identifiedBy: David R. Smith; language: en; institutionCode: USNM; basisOfRecord: PreservedSpecimen
- am. scientificName: *Orussus minutus* Middlekauff, 1983; kingdom: Animalia; phylum: Arthropoda; class: Insecta; order: Hymenoptera; family: Orussidae; genus: *Orussus*; specificEpithet: *minutus*; scientificNameAuthorship: Middlekauff, 1983; country: United States; countryCode: US; stateProvince: Indiana; county: Tippecanoe; locality: West Lafayette; georeferenceProtocol: label; eventDate: 1981-4-26; individualCount: 1; lifeStage: adult; behavior: collected from branches of *Acer saccharum*; recordedBy: M. & N. Deyrup; associatedReferences: Deyrup, M.A. 1984. A maple wood wasp, *Xiphydria maculate*, and its insect enemies (Hymenoptera: Xiphydriidae). Great Lakes Entomologist, 17: 17–28. [referred to as "*Orussus* sp."]; identifiedBy: David R. Smith; language: en; institutionCode: USNM; basisOfRecord: PreservedSpecimen
- an. scientificName: *Orussus minutus* Middlekauff, 1983; kingdom: Animalia; phylum: Arthropoda; class: Insecta; order: Hymenoptera; family: Orussidae; genus: *Orussus*; specificEpithet: *minutus*; scientificNameAuthorship: Middlekauff, 1983; country: United States; countryCode: US; stateProvince: Pennsylvania; county: Cumberland; verbatimCoordinates: 40.22479, -76.96278; decimalLatitude: 40.22479; decimalLongitude: -76.96278; georeferenceProtocol: label; samplingProtocol: Lindgren multifunnel trap; eventDate: 2011-5-4; individualCount: 2; lifeStage: adult; recordedBy: Shu Ambree; language: en; institutionCode: USNM; basisOfRecord: PreservedSpecimen
- ao. scientificName: *Orussus minutus* Middlekauff, 1983; kingdom: Animalia; phylum: Arthropoda; class: Insecta; order: Hymenoptera; family: Orussidae; genus: *Orussus*; specificEpithet: *minutus*; scientificNameAuthorship: Middlekauff, 1983; country: United States; countryCode: US; stateProvince: Pennsylvania; county: Cumberland; verbatimCoordinates: 40.22519, -76.96252; decimalLatitude: 40.22519; decimalLongitude: -76.96252; georeferenceProtocol: label; samplingProtocol: Lindgren multifunnel trap; eventDate: 2011-5-4; individualCount: 1; lifeStage: adult; recordedBy: Shu Ambree; language: en; institutionCode: USNM; basisOfRecord: PreservedSpecimen
- ap. scientificName: *Orussus minutus* Middlekauff, 1983; kingdom: Animalia; phylum: Arthropoda; class: Insecta; order: Hymenoptera; family: Orussidae; genus: *Orussus*; specificEpithet: *minutus*; scientificNameAuthorship: Middlekauff, 1983; country: United States; countryCode: US; stateProvince: Pennsylvania; county: Northumberland; verbatimCoordinates: 40.87671, -76.50962; decimalLatitude: 40.87671; decimalLongitude: -76.50962; georeferenceProtocol: label; samplingProtocol: Lindgren multifunnel trap; eventDate: 2011-6-1; individualCount: 1; lifeStage: adult; recordedBy: Jay Bagley; language: en; institutionCode: USNM; basisOfRecord: PreservedSpecimen
- aq. scientificName: *Orussus minutus* Middlekauff, 1983; kingdom: Animalia; phylum: Arthropoda; class: Insecta; order: Hymenoptera; family: Orussidae; genus: *Orussus*; specificEpithet: *minutus*; scientificNameAuthorship: Middlekauff, 1983; country: United States; countryCode: US; stateProvince: Pennsylvania; county: Lehigh; verbatimCoordinates: 40.45855, -75.473198; decimalLatitude: 40.45855; decimalLongitude: -75.473198; georeferenceProtocol: label; samplingProtocol: Lindgren multifunnel trap; eventDate: 2012-5-31; individualCount: 1; lifeStage: adult; recordedBy: Sam Louenwirth; language: en; institutionCode: USNM; basisOfRecord: PreservedSpecimen
- ar. scientificName: *Orussus minutus* Middlekauff, 1983; kingdom: Animalia; phylum: Arthropoda; class: Insecta; order: Hymenoptera; family: Orussidae; genus: *Orussus*; specificEpithet: *minutus*; scientificNameAuthorship: Middlekauff, 1983; country: United States; countryCode: US; stateProvince: Pennsylvania; county: Bedford; verbatimCoordinates: 40.04287, -78.36906; decimalLatitude: 40.04287; decimalLongitude: -78.36906; georeferenceProtocol: label; samplingProtocol: Lindgren multifunnel trap; eventDate: 2012-5-15; individualCount: 1; lifeStage: adult; recordedBy: Nathan Delp; language: en; institutionCode: USNM; basisOfRecord: PreservedSpecimen
- as. scientificName: *Orussus minutus* Middlekauff, 1983; kingdom: Animalia; phylum: Arthropoda; class: Insecta; order: Hymenoptera; family: Orussidae; genus: *Orussus*; specificEpithet: *minutus*; scientificNameAuthorship: Middlekauff, 1983; country: United States; countryCode: US; stateProvince: Pennsylvania; county: Fulton; verbatimCoordinates: 40.02970, -77.637133; decimalLatitude: 40.0297; decimalLongitude: -77.637133; georeferenceProtocol: label; samplingProtocol: Lindgren multifunnel trap; eventDate: 2014-7-8; individualCount: 2; lifeStage: adult; language: en; institutionCode: USNM; basisOfRecord: PreservedSpecimen
- at. scientificName: *Orussus minutus* Middlekauff, 1983; kingdom: Animalia; phylum: Arthropoda; class: Insecta; order: Hymenoptera; family: Orussidae; genus: *Orussus*; specificEpithet: *minutus*; scientificNameAuthorship: Middlekauff, 1983; country: United States; countryCode: US; stateProvince: Pennsylvania; county: Chester; verbatimCoordinates: 40.6765, -75.71953; decimalLatitude: 40.6765; decimalLongitude: -75.71953; georeferenceProtocol: label; samplingProtocol: Lindgren multifunnel trap; eventDate: 2012-5-15; individualCount: 2; lifeStage: adult; recordedBy: Thea Stimmler; language: en; institutionCode: USNM; basisOfRecord: PreservedSpecimen
- au. scientificName: *Orussus minutus* Middlekauff, 1983; kingdom: Animalia; phylum: Arthropoda; class: Insecta; order: Hymenoptera; family: Orussidae; genus: *Orussus*; specificEpithet: *minutus*; scientificNameAuthorship: Middlekauff, 1983; country: United States; countryCode: US; stateProvince: Pennsylvania; county: Centre; verbatimCoordinates: 41.030522, -77.98226; decimalLatitude: 41.030522; decimalLongitude: -77.98226; georeferenceProtocol: label; samplingProtocol: Lindgren multifunnel trap; eventDate: 2012-5-18; individualCount: 1; lifeStage: adult; recordedBy: Ryan Weston; language: en; institutionCode: USNM; basisOfRecord: PreservedSpecimen
- av. scientificName: *Orussus minutus* Middlekauff, 1983; kingdom: Animalia; phylum: Arthropoda; class: Insecta; order: Hymenoptera; family: Orussidae; genus: *Orussus*; specificEpithet: *minutus*; scientificNameAuthorship: Middlekauff, 1983; country: United States; countryCode: US; stateProvince: Pennsylvania; county: Bradford; verbatimCoordinates: 41.81719, -76.79818; decimalLatitude: 41.81719; decimalLongitude: -76.79818; georeferenceProtocol: label; samplingProtocol: Lindgren multifunnel trap; eventDate: 2012-5-31; individualCount: 1; lifeStage: adult; recordedBy: Rick Malak; language: en; institutionCode: USNM; basisOfRecord: PreservedSpecimen

- [illegible]

- Lindgren multifunnel trap; eventDate: 2011-5-2; individualCount: 1; lifeStage: adult; recordedBy: L. Donovan; language: en; institutionCode: USNM; basisOfRecord: PreservedSpecimen
- bi. scientificName: *Orussus minutus* Middlekauff, 1983; kingdom: Animalia; phylum: Arthropoda; class: Insecta; order: Hymenoptera; family: Orussidae; genus: *Orussus*; specificEpithet: *minutus*; scientificNameAuthorship: Middlekauff, 1983; country: United States; countryCode: US; stateProvince: Pennsylvania; county: Franklin; verbatimCoordinates: 39.930884, -77.637928; decimalLatitude: 39.930884; decimalLongitude: -77.637928; georeferenceProtocol: label; samplingProtocol: Lindgren multifunnel trap; eventDate: 2011-4-28; individualCount: 1; lifeStage: adult; recordedBy: L. Donovan; language: en; institutionCode: USNM; basisOfRecord: PreservedSpecimen
- bj. scientificName: *Orussus minutus* Middlekauff, 1983; kingdom: Animalia; phylum: Arthropoda; class: Insecta; order: Hymenoptera; family: Orussidae; genus: *Orussus*; specificEpithet: *minutus*; scientificNameAuthorship: Middlekauff, 1983; country: United States; countryCode: US; stateProvince: Pennsylvania; county: Franklin; verbatimCoordinates: 39.930884, -77.637928; decimalLatitude: 39.930884; decimalLongitude: -77.637928; georeferenceProtocol: label; samplingProtocol: Lindgren multifunnel trap; eventDate: 2011-4-21; individualCount: 1; lifeStage: adult; recordedBy: L. Donovan; language: en; institutionCode: USNM; basisOfRecord: PreservedSpecimen
- bk. scientificName: *Orussus minutus* Middlekauff, 1983; kingdom: Animalia; phylum: Arthropoda; class: Insecta; order: Hymenoptera; family: Orussidae; genus: *Orussus*; specificEpithet: *minutus*; scientificNameAuthorship: Middlekauff, 1983; country: United States; countryCode: US; stateProvince: Pennsylvania; county: Franklin; verbatimCoordinates: 39.930884, -77.637928; decimalLatitude: 39.930884; decimalLongitude: -77.637928; georeferenceProtocol: label; samplingProtocol: Lindgren multifunnel trap; eventDate: 2011-5-19; individualCount: 1; lifeStage: adult; recordedBy: L. Donovan; language: en; institutionCode: USNM; basisOfRecord: PreservedSpecimen
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- bm. scientificName: *Orussus minutus* Middlekauff, 1983; kingdom: Animalia; phylum: Arthropoda; class: Insecta; order: Hymenoptera; family: Orussidae; genus: *Orussus*; specificEpithet: *minutus*; scientificNameAuthorship: Middlekauff, 1983; country: United States; countryCode: US; stateProvince: Pennsylvania; county: Franklin; verbatimCoordinates: 39.93094, -77.637133; decimalLatitude: 39.93094; decimalLongitude: -77.637133; georeferenceProtocol: label; samplingProtocol: Lindgren multifunnel trap; eventDate: 2011-4-21; individualCount: 1; lifeStage: adult; recordedBy: L. Donovan; language: en; institutionCode: USNM; basisOfRecord: PreservedSpecimen
- bn. scientificName: *Orussus minutus* Middlekauff, 1983; kingdom: Animalia; phylum: Arthropoda; class: Insecta; order: Hymenoptera; family: Orussidae; genus: *Orussus*; specificEpithet: *minutus*; scientificNameAuthorship: Middlekauff, 1983; country: United States; countryCode: US; stateProvince: Pennsylvania; county: Franklin; verbatimCoordinates: 39.93021, -77.638025; decimalLatitude: 39.93021; decimalLongitude: -77.638025; georeferenceProtocol: label; samplingProtocol: Lindgren multifunnel trap; eventDate: 2011-6-1; individualCount: 1; lifeStage: adult; recordedBy: L. Donovan; language: en; institutionCode: USNM; basisOfRecord: PreservedSpecimen
- bo. scientificName: *Orussus minutus* Middlekauff, 1983; kingdom: Animalia; phylum: Arthropoda; class: Insecta; order: Hymenoptera; family: Orussidae; genus: *Orussus*; specificEpithet: *minutus*; scientificNameAuthorship: Middlekauff, 1983; country: United States; countryCode: US; stateProvince: Pennsylvania; county: Franklin; verbatimCoordinates: 39.93021, -77.638025; decimalLatitude: 39.93021; decimalLongitude: -77.638025; georeferenceProtocol: label; samplingProtocol: Lindgren multifunnel trap; eventDate: 2011-5-2; individualCount: 1; lifeStage: adult; recordedBy: L. Donovan; language: en; institutionCode: USNM; basisOfRecord: PreservedSpecimen
- bp. scientificName: *Orussus minutus* Middlekauff, 1983; kingdom: Animalia; phylum: Arthropoda; class: Insecta; order: Hymenoptera; family: Orussidae; genus: *Orussus*; specificEpithet: *minutus*; scientificNameAuthorship: Middlekauff, 1983; country: United States; countryCode: US; stateProvince: Pennsylvania; county: Franklin; verbatimCoordinates: 39.93097, -77.637695; decimalLatitude: 39.93097; decimalLongitude: -77.637695; georeferenceProtocol: label; samplingProtocol: Lindgren multifunnel trap; eventDate: 2011-4-21; individualCount: 1; lifeStage: adult; recordedBy: L. Donovan; language: en; institutionCode: USNM; basisOfRecord: PreservedSpecimen
- bq. scientificName: *Orussus minutus* Middlekauff, 1983; kingdom: Animalia; phylum: Arthropoda; class: Insecta; order: Hymenoptera; family: Orussidae; genus: *Orussus*; specificEpithet: *minutus*; scientificNameAuthorship: Middlekauff, 1983; country: United States; countryCode: US; stateProvince: Connecticut; county: New London; municipality: Groton; georeferenceProtocol: label; eventDate: 17695.00; individualCount: 2; lifeStage: adult; recordedBy: Anton Jansson; institutionCode: MZLU; basisOfRecord: PreservedSpecimen

Distribution.

New York south to Georgia west to Manitoba, Iowa, and Arkansas.

Analysis.

The Arkansas specimens were identified morphologically as *Orussus minutus* Middlekauff, 1983 (Figs 1, 2). The 16S rRNA sequence (GenBank #KM379143) was a 99.5% match with an existing *O. minutus* sequence (EF032174), differing by two base pairs.

Discussion.

The Arkansas specimens and those shared as photographs on Bugguide and Flickr significantly expand the known range of *O. minutus* westward (Fig. 3). Morphological determination of the Arkansas specimens was confirmed by genetic data and the species is easily identified due to its small size and distinct markings, so it is highly unlikely the photographed specimens are not *O. minutus*.

Many of the USNM specimens were collected by David R. Smith during 35 years of Malaise trapping specifically for sawflies. However, most recently collected specimens, especially those from Pennsylvania, were found as non-target species during various exotic species monitoring programs that utilized Lindgren multifunnel traps (David Smith, pers. comm., 28 Aug. 2015). The abundance of these specimens emphasize the utility of examining, or at least collecting and sending to the appropriate specialist, non-target species in mass trapping surveys, such as was suggested by Skvarla and Holland (2011). Precise figures for the number of traps and amount of effort that was involved in the Pennsylvania surveys is unavailable, so we are unable to compare the efficiency of Malaise trapping compared to Lindgren funnel trapping; however, the number of *O. minutus* that were collected in Lindgren funnel traps suggests that it may be a useful tool for collecting *Orussus*.

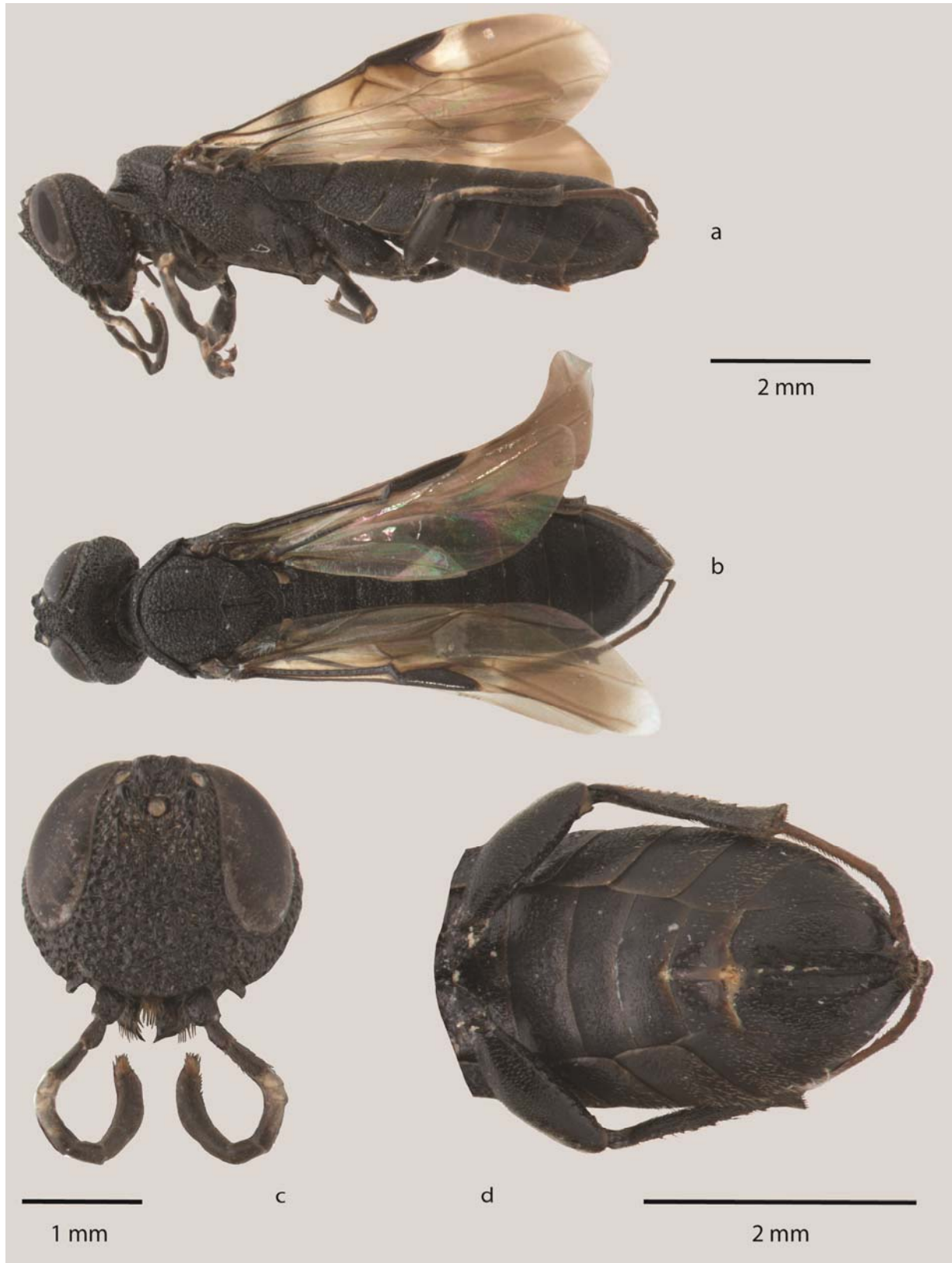


Figure 1. *Orussus minutus*, female. **a:** Lateral habitus. **b:** Dorsal habitus. **c:** Head. **d:** Ventral abdomen.

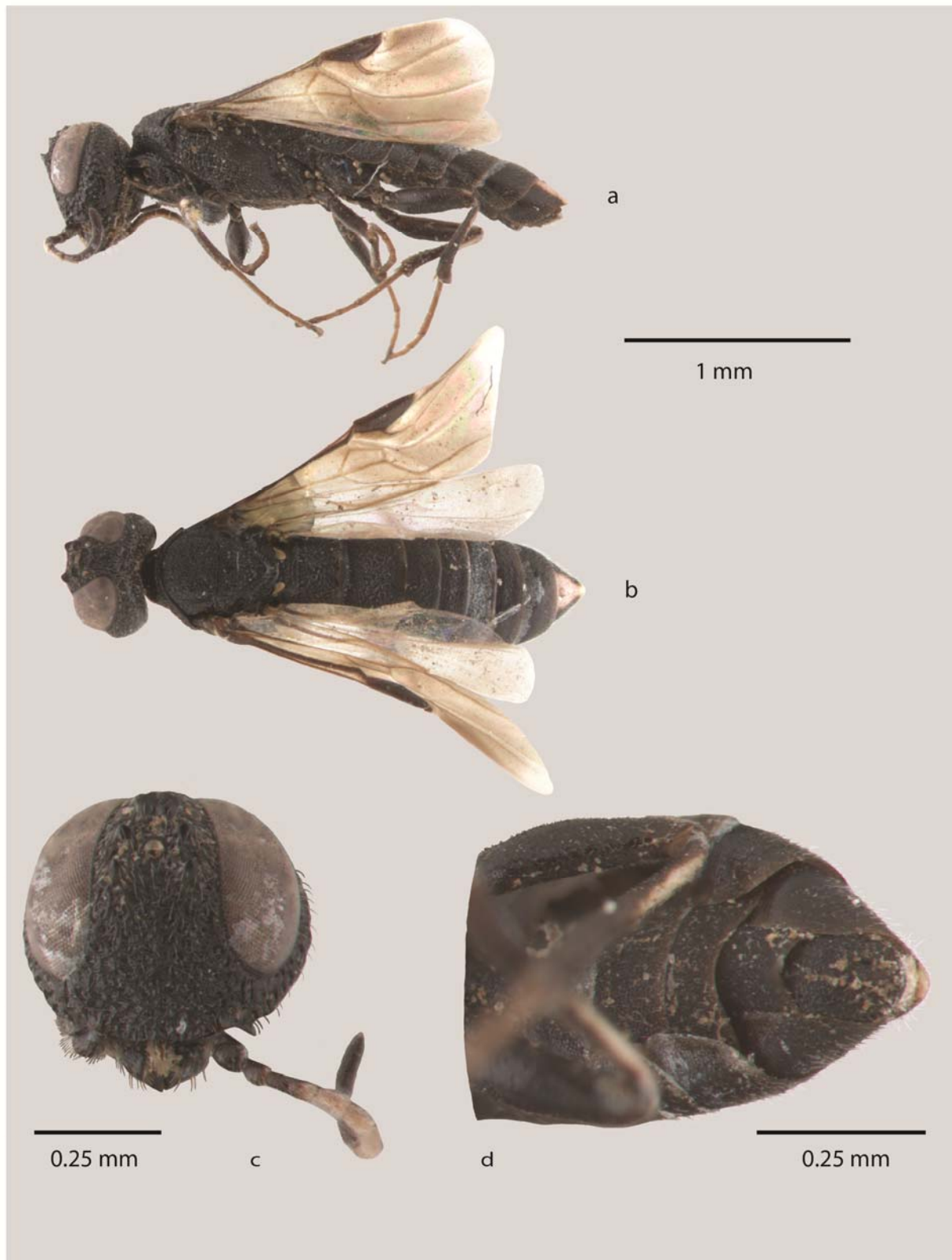


Figure 2. *Orussus minutus*, male. **a:** Lateral habitus. **b:** Dorsal habitus. **c:** Head. **d:** Ventral abdomen.

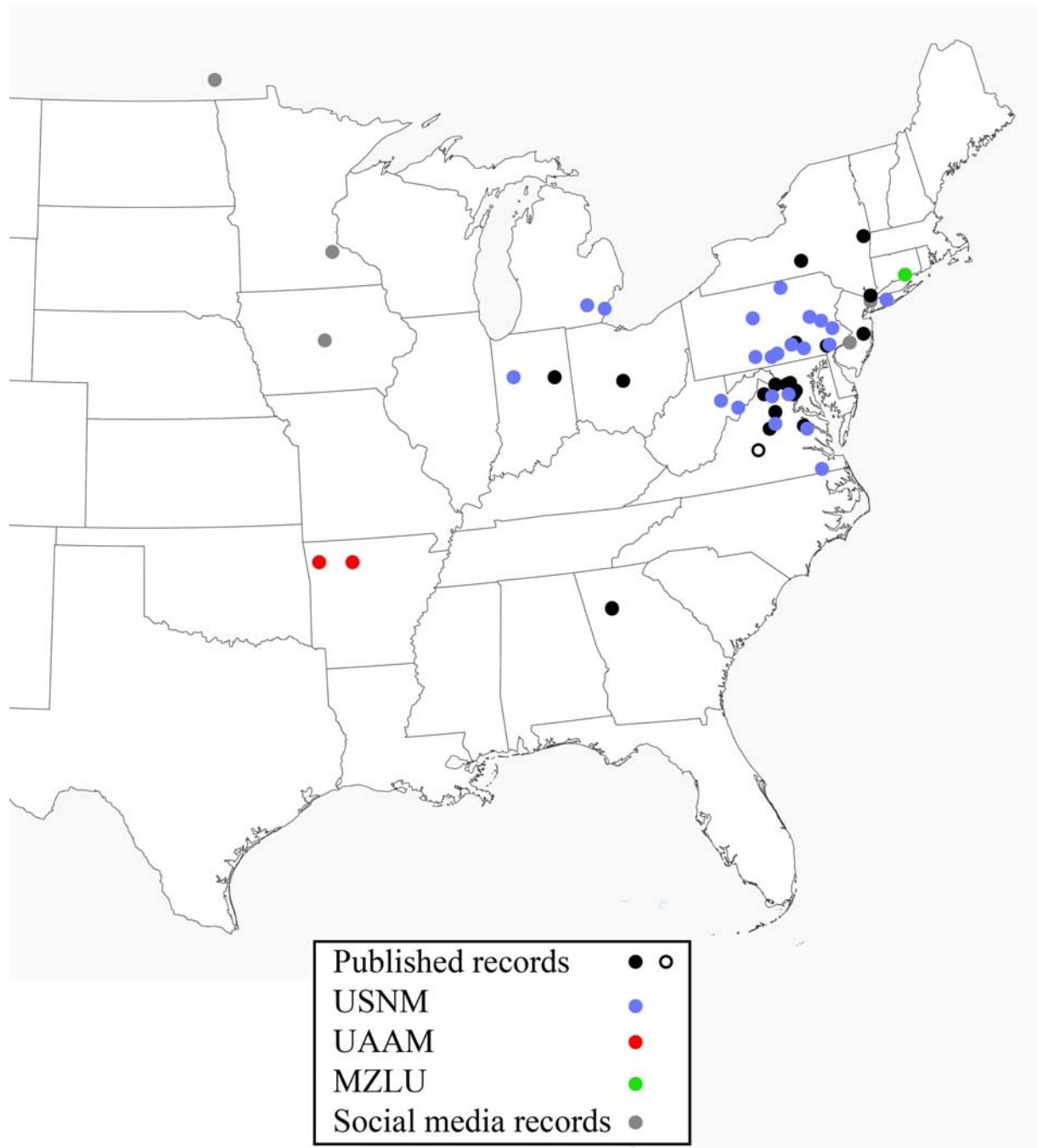


Figure 3. Known range of *Orussus minutus*. Solid circles represent collection localities, open circles represent state records lacking additional locality data.

Given the current records, *O. minutus* is likely present throughout most of Eastern North America. The concentration of specimens from northern Virginia and Pennsylvania reflect collecting effort and specimen recognition rather than true abundance and further collecting in the southeastern United States and Canada should produce additional specimens from those areas.

Finally, records found through Bugguide and Flickr join a growing list of discoveries made via citizen science and social media websites (e.g., Otto and Hill 2011, Winterton et al. 2012, Gonella et al. 2015) and help underscore the importance of such resources in descriptive biology and natural history.

Acknowledgements.

We thank David Smith for providing the collection data for the USNM specimens and his willingness and enthusiasm to discuss sawflies; Christer Hansson for providing the collection data for the MZLU specimens and Lund University for digitizing their collection; Danielle Fisher for her assistance in sorting trap material and finding the first specimen;

Clinton Trammel for laboratory assistance; and Lars Vilhelmsen, Stephen Blank, and Andy Deans for their helpful comments and corrections. This project and the preparation of this publication was funded in part by the State Wildlife Grants Program (Grant # T39-05) of the U.S. Fish and Wildlife Service through an agreement with the Arkansas Game and Fish Commission.

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IX. New information about the cypress weevil, *Eudocimus mannerheimii* (Boheman, 1836) (Coleoptera: Curculionidae: Molytinae): redescription, range expansion, new host records, and a report as a possible causative agent of tree mortality

Abstract.

The cypress weevil, *Eudocimus mannerheimii* (Boheman, 1836), is reported from northwest Arkansas (**new state** record). The suspected host in this area is eastern red cedar (*Juniperus virginiana* L.), which represents a new host record. Additional new host records from arborvitae (*Thuja* L.) in North Carolina are reported. A brief redescription of the adults that expands upon the original description and photographs are included. Although cypress weevils are not generally considered pestiferous, a case of landscape trees likely killed by this species is included.

Introduction.

Eudocimus mannerheimii (Boheman, 1836) (Figs. 1–8, 12), commonly called cypress weevils, are large native hylobiine weevils (Curculionidae: Molytinae) that breed in stressed bald cypress and related trees (Cupressaceae). Although not generally considered a pest, damage to small diameter nursery stock and girdling of sprouts and seedlings has been occasionally reported (Mayfield 2004; Randall *et al.* 2005). Aside from checklist and catalogue entries (e.g., Hopkins 1904; Blatchley and Leng 1916; Alonso-Zarazaga and Lyal 1999), information about *E. mannerheimii* is limited (Mayfield 2004).

Cypress weevils range from New York south to Florida and west to Louisiana (O'Brien and Wibmer 1982; Peck and Thomas 1998). Recently, it has also been reported from Querétaro and Jalisco, Mexico (Jones *et al.* 2003; Sánchez-Martínez *et al.* 2010).

Recorded hosts include bald cypress (*Taxodium distichum* (L.) Rich.) (Hopkins 1904; Anderson 2002; Bambara 2004), pond cypress (*T. ascendens* Brongn.), Montezuma cypress (*T. mucronatum* Ten.) (Jones *et al.* 2003; Sánchez-Martínez *et al.* 2010), Japanese cedar (*Cryptomeria* (L.f.) D. Don), and Leyland cypress (\times *Cupressocyparis leylandii* A. B. Jacks. and Dallim.) (Bambara 2004). Additionally, Baker and Bambara (1999) suggested *E. mannerheimii* may feed on Atlantic white cedar (*Chamaecyparis thyoides* (L.) Britton, Sterns and Poggenb.) in New York, as bald cypress is not native so far north.

Herein, we discuss the collection of *E. mannerheimii* outside its known range and present a situation in which the beetle was involved in the death of landscape trees. Furthermore, as the original description of this species is in Latin, and therefore inaccessible to most modern readers, and subsequent redescrptions (e.g., Blatchley and Leng 1916) do not encompass the variation, especially in color, seen in the species, we provide a brief updated description of the adults.

Materials and Methods.

In Arkansas, adult weevils (Figs. 1–2) were collected at Steel Creek along the Buffalo National River (Newton County) by Malaise traps in an eastern red cedar (*Juniperus virginiana* L.) glade and in a purple Lindgren funnel trap in a mixed forest containing eastern red cedar.

Recent, 2013, specimens from North Carolina (locality data below) were collected as larvae (Fig. 7) and pupae (Fig. 8), or reared to adulthood (Figs. 3–5), from a ca. 30 cm x 12 cm trunk section of ‘Green Giant’ arborvitae (*Thuja plicata* x *T. standishii*). The section was received at the Plant Disease and Insect Clinic at North Carolina State University on 30 January 2013 and isolated in a covered 5-gallon bucket at room temperature. Late instar larvae were observed under the bark and the sample was maintained until adults emerged around April 18th, 2013.

Specimens collected in Arkansas have been deposited in the University of Arkansas Arthropod Museum (**UAAM**). Specimens collected in North Carolina have been deposited in the North Carolina State University Insect Museum (**NCSU**). Institutional abbreviations follow Evenhuis (2014).

Taxonomy.

***Eudociminus* Leng 1918**

Eudocinus Dejean 1835: 276 [nomen nudum]

Eudocimus Boheman 1836: 240 [preoccupied by Wagler, 1832 (Aves)]

Eudocinus Laporte 1840: 335 [lapsus]

Eudociminus Leng 1918: 210

LSID: urn:lsid:zoobank.org:act:8652B3EE-8CC9-49F0-8930-6D3EF060A0F3

Type species: *Eudociminus mannerheimii*

***Eudociminus mannerheimii* (Boheman 1836)**

Eudocinus mannerheimii Schönherr Dejean 1835: 276 [nomen nudum]

Eudocimus mannerheimii Boheman 1836: 241

Eudociminus mannerheimii Leng 1918

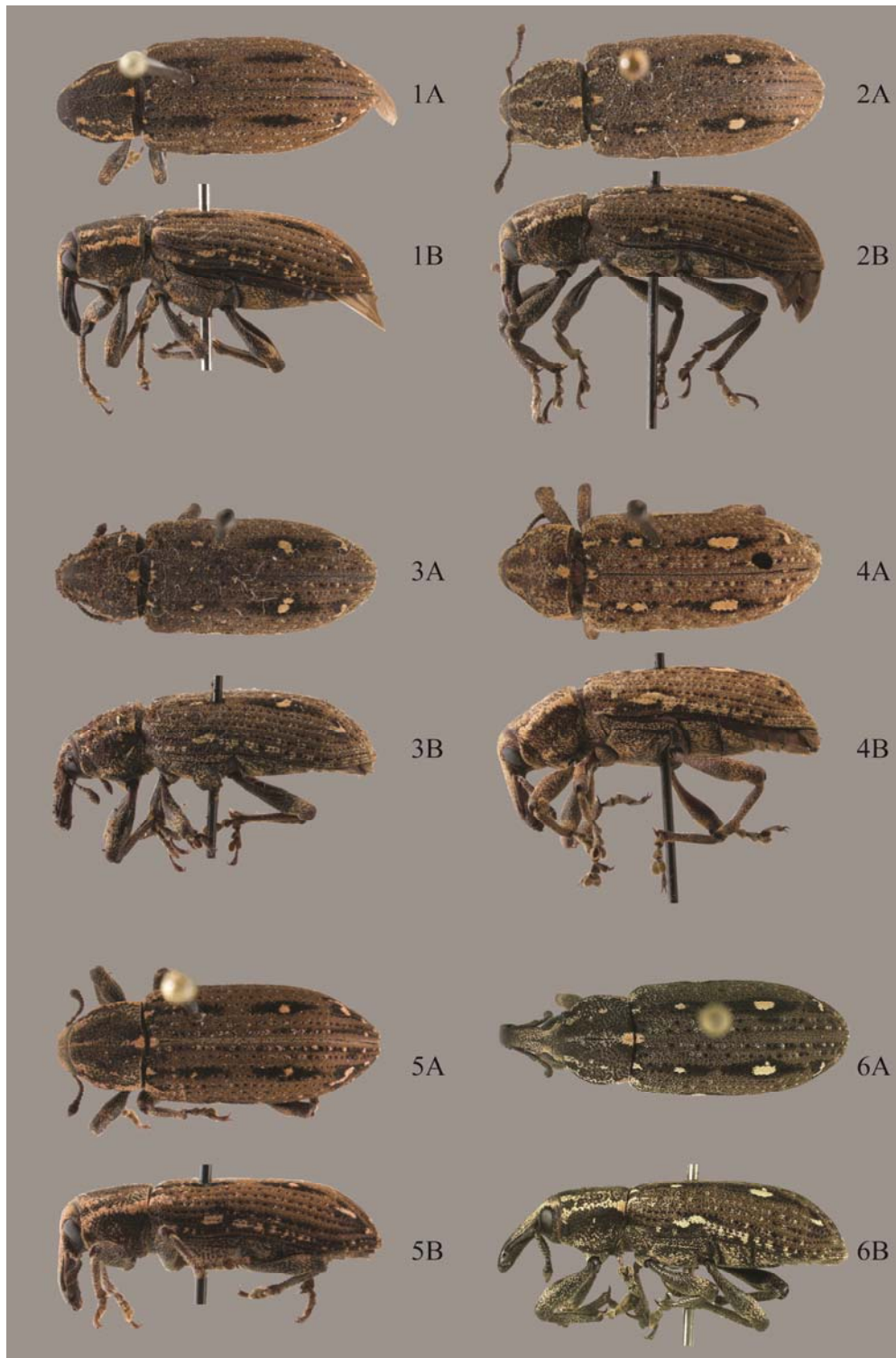
LSID: urn:lsid:zoobank.org:act:51E0D421-BFBC-4BDE-8AE4-C987AD13038F

Description (n=14). Body 10–17 mm long and 3.5–5.5 mm wide. Cuticle dark red to black, generally clothed in colored scale-like setae. **Dorsum:** dark gray to brown, with scale-like

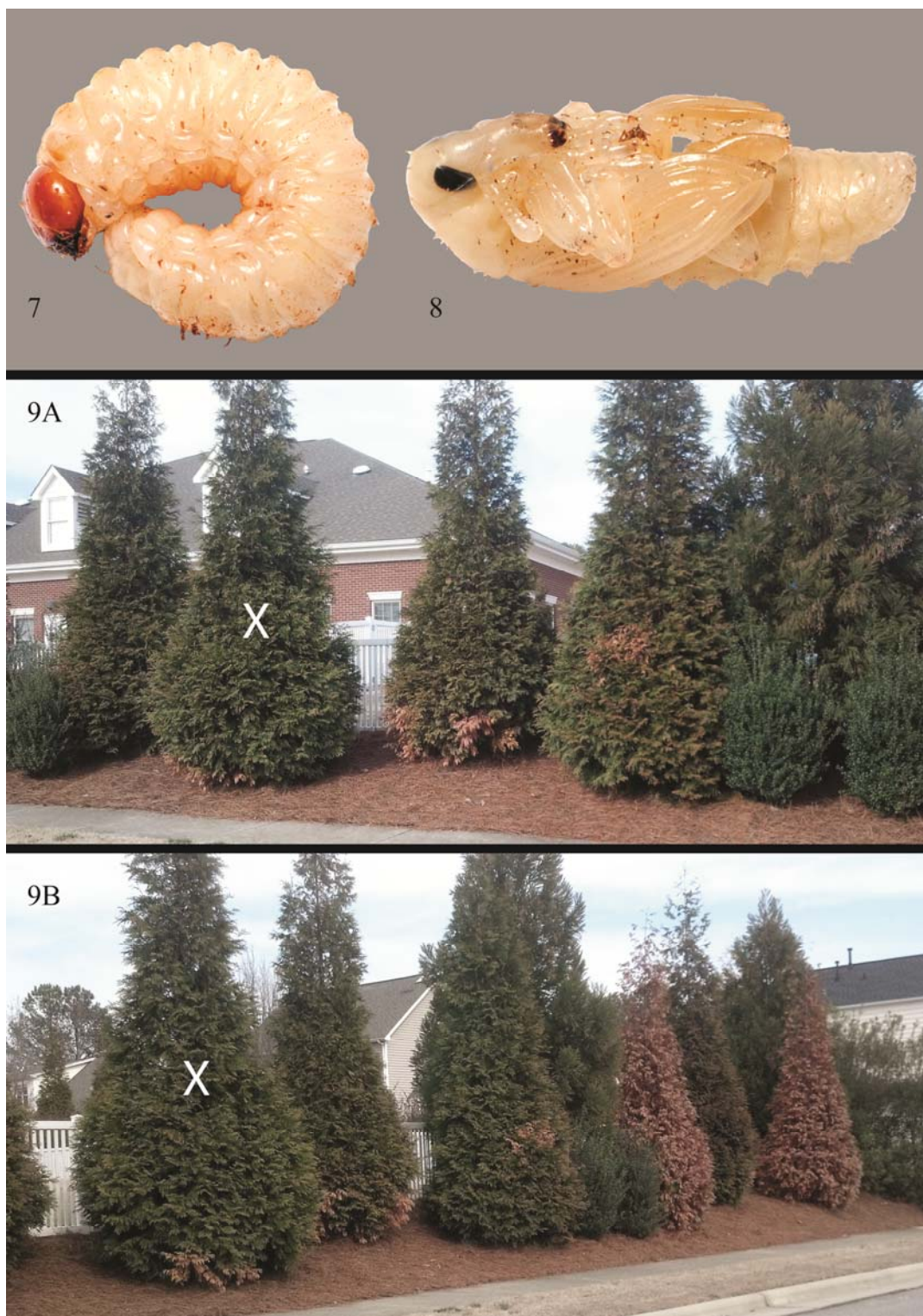
setae densely packed. **Head:** punctate with smooth median line; light tan, orange, or white scales dorsolaterally and immediately posterior and ventral to eye, otherwise without setae; rostrum 2/3 length of pronotum; eyes elongate, reniform. **Pronotum:** slightly longer than wide and sides moderately rounded; disc coarsely punctate with smooth median line; with five lines of variable color (light tan, orange, pink, or white): two complete lines dorsolaterally that connect anteriorly midway between eyes and posteriorly to spots on sixth elytral intervals; two incomplete lines that connect anteriorly to the dorsal apex of the eye and terminate in the anterior third of the pronotum; and a median line, which may be indistinct in the middle. **Scutellum:** triangular and light tan to white. **Elytra** slightly wider than pronotum and parallel-sided, with humeral angle distinct; preapical elytral hump present in some specimens (e.g., Fig. 4b); striae deeply impressed, intervals flat. Elytra with four dark brown to black spots on fourth intervals, sometimes coalescent into stripes, with or without four light tan to white spots; elytral bases usually with two to four light tan to white spots on intervals four and six; intervals nine and ten with tan to white spots, sometimes coalescent into longer lines. **Venter** (including legs) generally appearing dark, with sparse scale-like setae light tan, orange, pink, or white; legs additionally have simple setae. Tibiae with strong hook-like unci. Tarsal claws simple, without teeth.

Specimens examined: (14 pinned specimens) 2 males, USA, North Carolina, Phelps Lake, ex. cypress bark, 25 October 1928, coll. B. B. Fulton (NCSU) • 1 female, USA, North Carolina, Bladen Co, White Lake, cypress, 14 March 1953, coll. D. M. Weisman (NCSU) • 4 females, 1 male, USA, North Carolina, Wake Co, Holly Springs, reared from *Thuja* sp., 30 January 2013, coll. M. A. Bertone (NCSU) • 1 female, USA, Arkansas, Newton Co, Steel Creek, ex Malaise trap set in eastern red cedar glade, 10 July 2010, coll. J. R. Fisher and D. Keeler (UAAM) • 1 female (APGD 10-0618-003, #135701), USA, Arkansas, Newton Co, Steel Creek

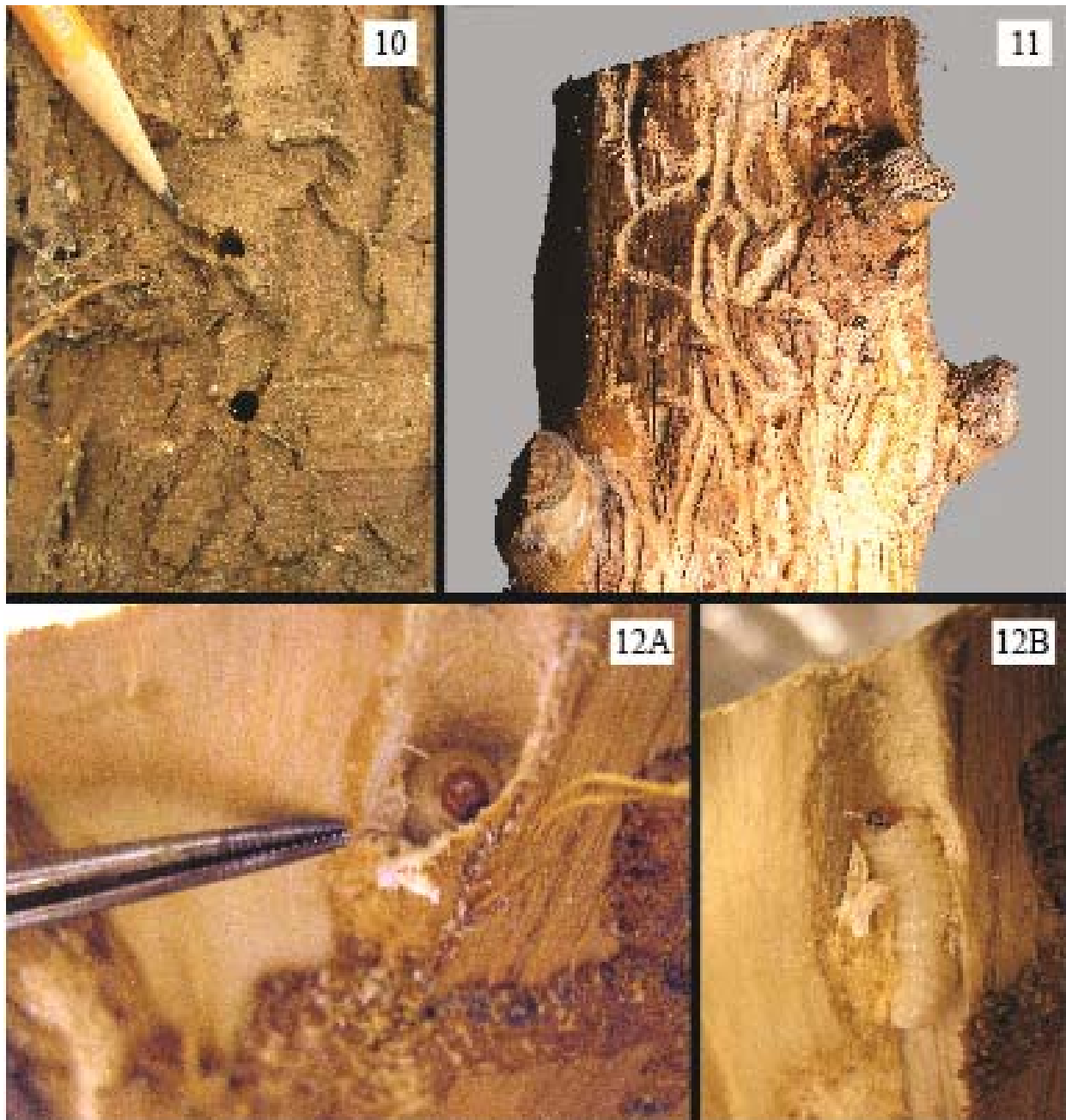
(36°01'56" N, 93°20'02" W), ex Malaise trap set in eastern red cedar glade, 18 June 2010, col. J. R. Fisher and M. J. Skvarla (UAAM) • 2 males (APGD 10-0618-003, #135702), USA, Arkansas, Newton Co, Steel Creek (36°01'56" N, 93°20'02" W), ex Malaise trap set in eastern red cedar glade, 18 June 2010, col. J. R. Fisher and M. J. Skvarla (UAAM) • 1 female (MS 13-1023-017, #133546), USA, Arkansas, Newton Co, Steel Creek (36°02'19" N, 93°20'27" W), ex. purple Lindgren funnel trap, 23 October 2013, col. M. J. Skvarla (UAAM) • 1 male, "Univ. of Ark. Student Coll.", no other data (UAAM).



Figs 1–6. *Eudociminus mannerheimii*, adults. **A–B**) dorsal and lateral habitus; **1–6**) Locality: **1**) Steel Creek, Newton Co., Arkansas; **2**) “Univ. of Ark. Student Coll.”; **3–4**) Phelps Lake, North Carolina; **5**) Holly Springs, Wake Co., North Carolina; **6**) Gainesville, Alachua Co., Florida. Photograph by Michael C. Thomas. Used with permission. Not to scale.



Figs. 7–9. *Eudocimus mannerheimii*, immature stages and landscape damage. **7)** Larva; **8)** Pupa; **9)** Landscape damage. “X” indicates the same tree in both photographs. **9A)** View left showing one undamaged tree and three minimally damaged trees; **9B)** View right showing three minimally damaged trees and two dead trees.



Figs 10–12. Tree damage. **10)** exit holes; **11)** larval galleries after bark was removed; **12A–B)** larva *in situ*.

Discussion.

Specimens collected from Steel Creek along the Buffalo River in the Boston Mountains represent the first report of the species from the Interior Highlands of Arkansas and significantly expand the range of the species north from previous records in Louisiana. While we did not observe oviposition, larval feeding, or adult emergence, we suggest the beetles were breeding in eastern red cedar as they were collected in a cedar glade and no other Cupressaceae were present at or near the site. All specimens collected were brown with orange scale-like setae, with dark brown spots on the fourth elytral interval, and lacking light elytral spots (Figs. 1–2).

The sole specimen located in the Arthropod Museum at the University of Arkansas lacks collection data beyond “Univ. of Ark. Student Coll.” (Fig. 2). While it was most likely collected in Arkansas, it cannot be assigned to the state conclusively and therefore does not represent an earlier record for the species in the state. It is similar in coloration to the Steel Creek specimens except that light elytral spots are present.

North Carolina specimens (Figs. 3–5) reared from arborvitae were similar in coloration to the Arkansas specimens. They also exhibited variation in the extent of light-colored elytral spots and presence/absence of a preapical elytral hump, which suggests these characters do not represent geographic variation. No dark grey specimens with black elytral stripes and white spots (e.g., Fig. 6) were examined. Further investigation is needed to determine if this variation in color has any correlation with geography or phylogenetic history.

The cypress weevil appears to be an occasional primary pest and, more frequently, a secondary invader of trees (Baker and Bambara 1999; Bambara 2004). Adult feeding damage to young shoots and green twigs (Baker and Bambara 1999; Bambara 2004; Randall *et al.* 2005) may cause aesthetic damage to trees. Tunneling by the larvae in small saplings is known

(Mayfield 2004) and likely causes mortality in some plants. Most infestations of this beetle, however, occur in stressed, dying or dead trees. In the case of the first record of this beetle in arborvitae (*Thuja* L.), a row of mature trees (Fig. 9) planted outside a school began to decline rapidly due to unknown factors. Landscape contractors stated that only some of the plants were affected, and adjacent Japanese cedars (*Cryptomeria japonica* (L.f.) D. Don) were unaffected. A trunk section from one of the dead trees revealed approximately 12 large larvae residing in tunnels (Figs. 10–12). The large number of specimens found in such a small portion of the plant, and located largely in the vascular tissue just below the bark, suggests the weevil likely overwhelmed some of the plants, resulting in rapid death. At this time we do not know why some plants were so heavily infested while others were not.

Specimen records indicate two to three generations of this weevil per year in North Carolina. Final instars were abundant in the arborvitae collected in January, signifying initial colonization during the previous fall. Adults emerged under laboratory conditions in March, similar to the suggested early spring timing of adults as mentioned in the literature (Bambara 2004; Mayfield 2004). Mid- to late instar larvae were also found in a small arborvitae branch in North Carolina during May and probably represented the second generation. Based on the specimens described here, adult beetles can be found in the summer (July specimens) and fall (October specimens).

Acknowledgments.

We thank Robert Anderson for confirming the identification of the first specimens collected in Arkansas and providing the full citation for Boheman (1836); Michael C. Thomas and Mark Schaffer for the use of their photographs; Sarah Skvarla for her help editing photos for publication; David Stephan for advice on rearing the North Carolina specimens; Mark Schaffer

for information about the site containing infested arborvitae; and the anonymous reviewers for their constructive comments

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X. Report on a large collection of *Merope tuber* Newman, 1838 (Mecoptera: Meropeidae) from Arkansas with notes on collection technique, sex ratio, and male clasper size

Abstract.

A large collection of earwigflies, *Merope tuber*, is reported from Arkansas and flight period and sex ratio are discussed. In contrast to previous studies, earwigflies were caught more frequently in pan traps than in Malaise traps and male clasper size was found not to be bimodal.

Introduction.

Merope tuber Newman, 1838, known as earwigflies or forcepflies, are uncommonly collected and have fascinated entomologists since their discovery in 1837 (Fig. 1). This fascination was initially due to their presumed rarity – only 16 specimens were collected between their discovery and 1904 [1]. Since then, they have continued to receive attention due to their previously assumed basal phylogenetic position within Mecoptera; relatively unknown life history; undescribed larvae; and odd appearance relative to other Mecoptera (e.g., a flattened body, opisthognathus head, and broad wings folded over the abdomen) [2, 3].

Only two other extant meropeids exist: *Austromerope poultoni* Killington, 1933 [4] from Western Australia and *Austromerope brasiliensis* Machado et al., 2013 [3] from Brazil. One extinct species, *Boreomerope antiqua* Novokschonov, 1995 [5], is known from Middle Jurassic lacustrine claystone near Kubekovo village in Siberia. Four extinct species of *Thaumatomerope* (i.e. *T. madygenica* Rasnitsyn 1974, *T. minuta* Rasnitsyn 1974, *T. oligoneura* Rasnitsyn 1974, and *T. sogdiana* Rasnitsyn 1974) were originally assigned to Meropeidae but were later reassigned to Thaumatomeropidae [6].



Figure 1. *Merope tuber*, male.

Collections of *M. tuber* continue to be infrequent. Prior to 1954 it was reported only from areas in or east of the Appalachian Mountains. Since then, the known range has been extended north to southern Ontario [7, 8, 9], west to Minnesota [10, 11], Iowa [12], Missouri [13, 14, 15], Arkansas [12, 15, 16], and Kansas [12], and south to Alabama [17], Georgia [16], and Florida [18, 19]. Rather than true emigration, this range expansion is best explained by the increased use of various passive trapping techniques [13]. *Merope tuber* have been collected using Malaise traps, picric acid traps, European chafer traps, carbon dioxide traps, molasses traps, and glue traps [2, 11, 20], with the most effective being Malaise traps [21].

Little is known about the life history of *M. tuber*. Adults are nocturnal, attracted to light at night, and spend daylight hours under logs and stones [1, 20]. They seem to be associated with moist deciduous woodlands near water [20, 22], although are occasionally caught in dry grasslands far from any stream or creek [9]. Feeding preferences are unknown, although they

may be attracted to carrion [2] similar to another mecopteran, *Notiothauma reedi* McLachlan, 1877, which has been reported from vertebrate carrion [23]. Adults stridulate by rubbing the jugum of the forewing against the metanotum [24]. The larvae of all meropeids, including *M. tuber*, remain undescribed [25] and their discovery “is certainly the most exciting thing left to be done in the study of North American Mecoptera” [13].

The flight period of *M. tuber* lasts throughout the summer with some variation depending on latitude. They have been reported to occur from June through October in Connecticut [26], June through September in Maryland [27], July through September in Ohio [25], May through September in Alabama [17], and April through December in Florida [18, 19].

Few studies have reported *M. tuber* in significant numbers, but in those that do, the sex ratio appears to be female biased. Scarbrough [28] collected 8 males and 18 females (1 male: 2.25 females) in two Malaise traps over a period of three years. Maier [26] collected 26 males and 43 females (1 male: 1.65 females) in a single Malaise trap over three years. Barrows and Flint [27], in six Malaise traps over the course of seven months, caught no males and 35 females. Johnson [25], in a single Malaise trap over two years, caught 61 males and 102 females (1 male: 1.67 females), the largest number of earwigflies yet reported from a single site. It is not known whether the sex ratio is truly skewed or if sampling bias is the cause.

Unlike life history, much is known about the morphology of *M. tuber*, with both internal and external anatomy of both sexes being well documented [29, 30, 31, 32]. Males have elongated genital styli (= claspers) that are thought to be used in mating as in other Mecoptera, either holding the female during copulation, fighting rival males, or both [25]. A bimodal distribution in clasper size has been demonstrated for at least one population with differential mating strategies being suggested as a possible cause [25].

Materials and Methods.

As part of a more extensive arthropod sampling project, five blocks were established at a four ha plot located at Steel Creek along the Buffalo National River in Arkansas (Fig. 2). In each block, five pan traps (one each of blue, red, green, yellow, and white) were randomly arranged under a terrestrial Malaise trap (MegaView Science Co. Ltd., Taichung, Taiwan), which was placed in perceived flight paths. In addition, three Lindgren funnel traps (ChemTica Internacional, S.A., Heredia, Costa Rica) (one in each color of green, purple, and black) were suspended non-randomly from large trees 4-10 meters from the ground in the lower canopy.

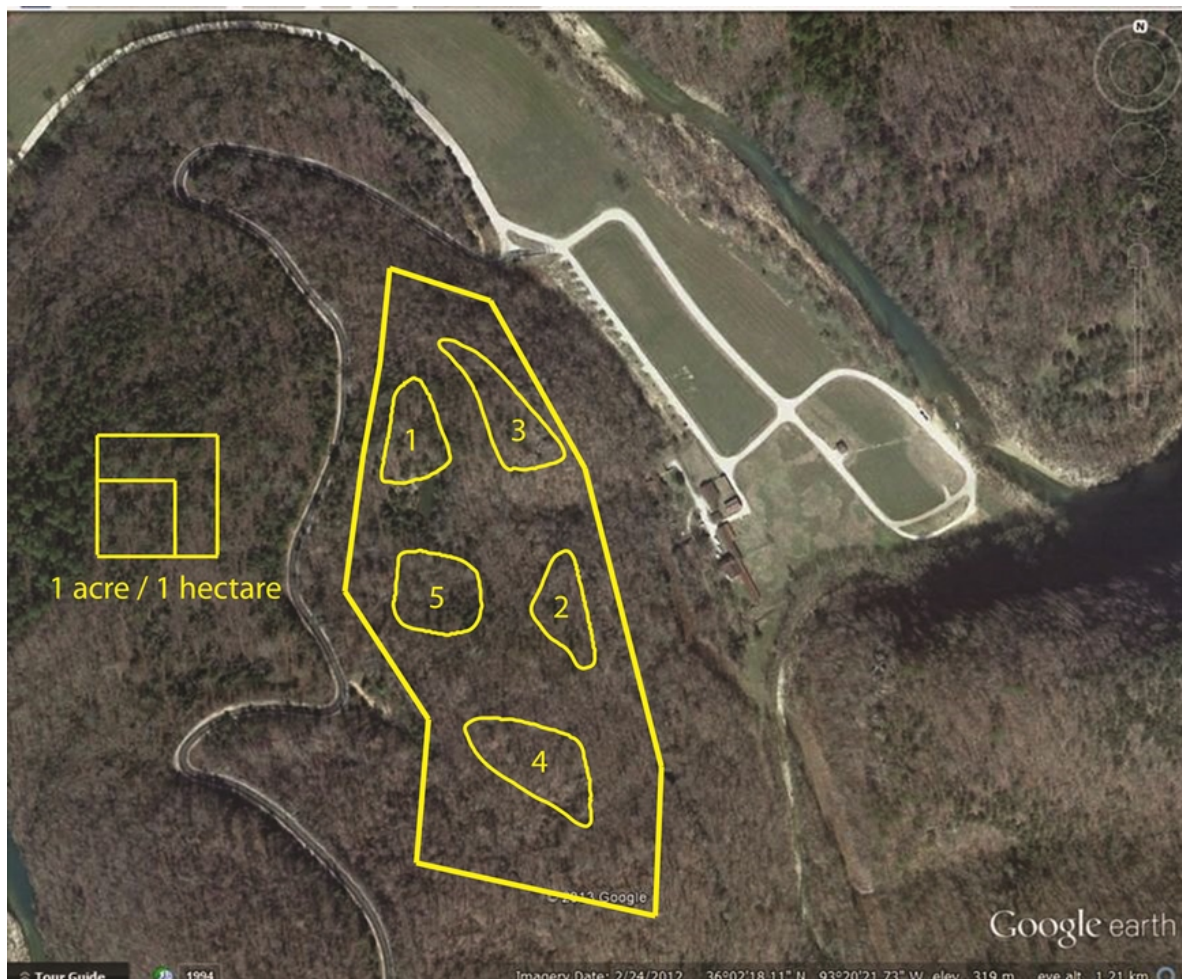


Figure 2. Overhead view of the field site at Steel Creek, with approximate limits of the site and blocks and acre/hectare scales in yellow. Base image taken from with Google Earth [36].

Four blocks contained a SLAM (Sea, Land, and Air Malaise, MegaView Science Co., Ltd., Taichung, Taiwan) trap (with top and bottom collectors counted as separate traps). Three blocks contained pitfall trap sets placed every five meters along a transect centered on a Malaise trap. Two of these blocks contained eight pitfall trap sets and one block contained a single set.

Pitfall traps were modified from a design proposed by Nordlander [33], which Lemieux and Lindgren [34] demonstrated catches carabids in similar numbers but is more efficient at excluding small vertebrate bycatch. Rather than cutting circular entrances in the sides of pitfall traps, we cut three slots, 2 cm tall x 9.3 cm wide, 2 cm under the rim in the sides of plastic soup containers leaving three 1.5 cm posts, equidistant apart, resulting in a 28 cm collecting surface. Diameter at the base of slots is approximately 10.5 cm and the cups are 10.5 cm deep below these slots, resulting in a collecting volume of 2,988 cm³. This allowed the matching lid to be secured to the cup instead of using a separate cover. A single cup was placed on either side of a 30.5 cm x 15.5 cm aluminum fence to make a pitfall trap set and the catch from both cups was combined and treated as a single sample.

Propylene glycol (Peak RV & marine antifreeze) (Old World Industries, LLC, Northbrook, IL) was used as a preservative in all trap types. Traps were placed on 13 March 2013, taken down on 4 December 2013, and collected approximately every two weeks. Trap catch was sieved in the field and stored in whirl-pak bags (Nasco, Fort Atkinson, WI) in 90% ethanol until sorting. After sorting, specimens were stored individually in 2 mL microtubes (VWR International, LLC, Randor, PA) in 70% ethanol. Voucher specimens have been submitted to the University of Arkansas Arthropod Museum.

Head width, pronotum width, wing length, and abdomen length were measured for both sexes. The length of the basistylus and dististylus (Fig. 3) were measured on the right side of males and combined to measure total clasper length.

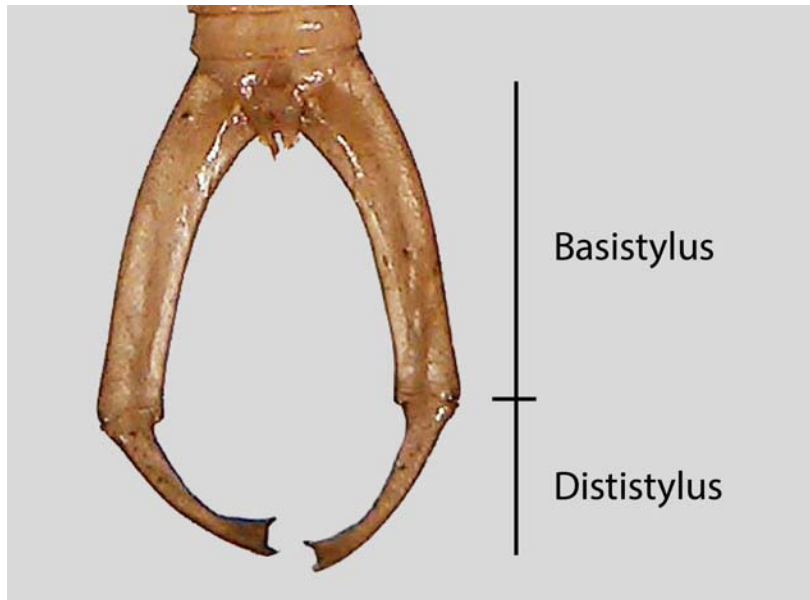


Figure 3. Clasper of male *Merope tuber* with basistylus and dististylus labeled.

Measurements were made in the following manner: photographs of a millimeter ruler and dorsal and ventral aspect of each specimen were taken through the eye piece of a Leica MZ 16 stereomicroscope with the camera on an HTC Droid Incredible 4G LTE; zoom was not adjusted between photographs to ensure they were to the same scale. All photographs were exported onto a desktop computer, opened in Image J [35], and measurements were taken by tracing the structures. Measurements were recorded in Microsoft Excel (Redmond, WA).

Shapiro-Wilk goodness-of-fit tests ($\alpha = 0.05$) were performed in JMP (SAS Institute, Cary, NC) to test normality of previously described measurements. An F-test for significance was performed by creating a generalized linear model (GLM) with a Gaussian distribution ($\alpha = 0.05$). Count data were not normally distributed and required transformation. Because the data contained many zeroes, one was added to each count and before a natural log transformation.

Because five pan traps were placed with a single Malaise trap, trap type could not be compared due to extremely skewed sample sizes. Instead, Malaise traps were considered a ‘color’ in analyses and tested against each pan trap color. This simultaneously allowed for comparisons among variables of equal sample sizes for both trap type and pan color.

Results and Conclusions.

All totaled eighty two earwigflies – 24 males and 58 females (1 male: 2.42 females) – were collected (Table 1). This female-biased collection is in line with previous studies [25, 26, 27, 28]. Earwigflies were first collected in late June, with the largest collection occurring in July, followed by low, but consistent, numbers caught until late October (Fig. 4). The beginning and end of the flight period were consistent with other areas at similar latitudes [18, 25, 26, 27].

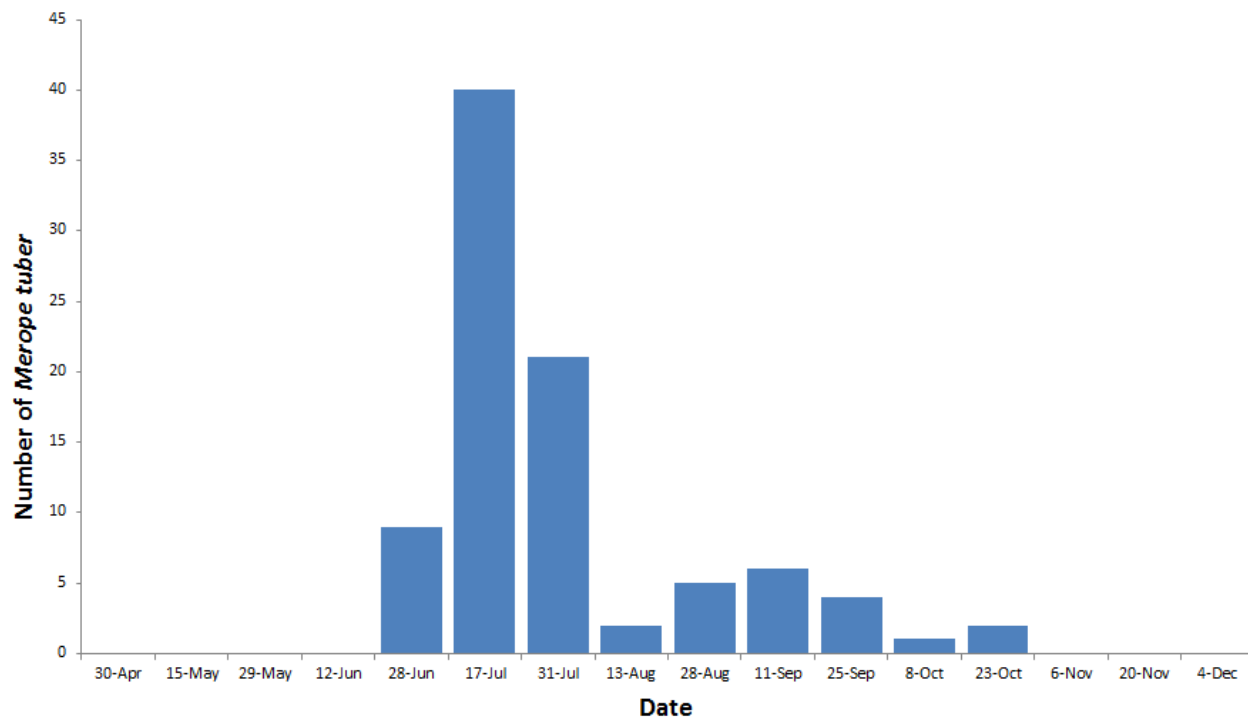


Figure 4. Number of *Merope tuber* collected across all traps per date.

Only a single body measurement, the dististylus, differed significantly from a normal distribution, but not in a bimodal manner (Table 2). These results are in contrast to previous studies (e.g., 25), which found a bimodal distribution in the size of male basistyli, dististyli, and total clasper length. As the use of the claspers is unknown the significance of this is also unknown.

Earwigflies were not caught in SLAM traps, Lindgren funnel traps, or pitfall trap sets, therefore, these traps were excluded from analyses. Significantly fewer *M. tuber* were caught in Malaise traps compared to pan traps [$t = -2.455$, d.f. = 1, $p = 0.0145$], although pan trap colors were not significantly different from each other. This is the first report of earwigflies being collected in pan traps, however, previous studies which reported large collections of *M. tuber* traditionally used Malaise traps alone. It should be noted that because pan traps were directly under Malaise traps, it is unknown whether those pan trap-collected individuals would have been captured in the Malaise trap collecting head, had pan traps had not been present.

Significantly more earwigflies were caught in block 4 [$t = 4.307$, d.f. = 1, $p = 0.00002$] and 5 [$t = 2.479$, d.f. = 1, $p = 0.0136$] than blocks 1, 2, and 3. This suggests that trap placement and microhabitat, even within a relatively small area of a few hectares, are important factors when collecting earwigflies. If earwigflies are specifically targeted, we suggest placing multiple traps in an area of known occurrence in order to maximize the microhabitats sampled and increase the chance of collecting these enigmatic insects.

Trap type	Block	Number of females caught	Number of males caught	Total caught
Malaise trap	1	0	0	0
Pan trap (purple)	1	1	1	2
Pan trap (yellow)	1	1	0	1
Pan trap (blue)	1	0	0	0
Pan trap (white)	1	1	0	1
Pan trap (red)	1	0	0	0
Malaise trap	2	0	1	1
Pan trap (purple)	2	2	0	2
Pan trap (yellow)	2	1	0	1
Pan trap (blue)	2	2	1	3
Pan trap (white)	2	2	1	3
Pan trap (red)	2	4	1	5
Malaise trap	3	0	0	0
Pan trap (purple)	3	2	0	2
Pan trap (yellow)	3	0	0	0
Pan trap (blue)	3	0	1	1
Pan trap (white)	3	1	0	1
Pan trap (red)	3	1	1	2
Malaise trap	4	0	0	0
Pan trap (purple)	4	5	3	8
Pan trap (yellow)	4	8	2	10
Pan trap (blue)	4	7	3	10
Pan trap (white)	4	2	2	4
Pan trap (red)	4	2	1	3
Malaise trap	5	1	0	1
Pan trap (purple)	5	2	3	5
Pan trap (yellow)	5	5	1	6
Pan trap (blue)	5	2	1	3
Pan trap (white)	5	4	0	4
Pan trap (red)	5	2	1	3

Table 1. Total number of *Merope tuber* collected per trap type per block, with subtotals of trap type and block.

Trap type	Block	Number of females caught	Number of males caught	Total caught
Trab subtotal				
Malaise trap	-	1	1	2
Pan trap (purple)	-	12	7	19
Pan trap (yellow)	-	15	3	18
Pan trap (blue)	-	11	6	17
Pan trap (white)	-	10	3	13
Pan trap (red)	-	9	4	13
Block subtotal				
-	1	3	1	4
-	2	11	4	15
-	3	4	2	6
-	4	24	11	35
-	5	16	6	22
Total	-	58	24	82

Table 1 (Cont.). Total number of *Merope tuber* collected per trap type per block, with subtotals of trap type and block.

Measurement	Sex	Minimum (mm)	Maximum (mm)	Mean (mm)	SD (mm)	W	Prob < W
Head width	Female	0.8	1.32	1.1	0.12	0.97	0.247
Pronotum width	Female	1.06	1.69	1.41	0.16	0.97	0.196
Forewing length	Female	8.86	13.28	11.66	0.9	0.98	0.337
Abdomen length	Female	4.1	8.96	6.44	1.3	0.97	0.153
Head width	Male	0.77	1.39	1.11	0.15	0.96	0.534
Pronotum width	Male	0.95	1.63	1.31	0.17	0.97	0.756
Forewing length	Male	9.52	13.39	11.82	1.04	0.971	0.695
Abdomen length	Male	4.07	7.61	5.8	0.78	0.95	0.206
Basistylus length	Male	2.21	5.09	4.05	0.77	0.95	0.265
Dististylus length	Male	1.47	2.91	2.34	0.43	0.91	0.036*
Clasper total length	Male	3.68	7.97	6.38	1.17	0.94	0.138

Table 2. Minimum, maximum, and mean measurements of various body parts, and results of Shapiro-Wilk goodness-of-fit tests on the same. $P < 0.05$ is considered significant.

Acknowledgments.

We thank Danielle Fisher for her assistance in sorting samples. This project and the preparation of this publication was funded in part by the State Wildlife Grants Program (Grant # T39-05) of the U.S. Fish and Wildlife Service through an agreement with the Arkansas Game and Fish Commission.

Disclosure.

The authors declare that there is no conflict of interests regarding the publication of this paper.

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XIII. Conclusions

The Interior Highlands is a biodiversity hotspot, with at least 200 known endemic species, more than half of which are arthropods, but the area is under studied compared to other regions of high biodiversity and endemism such as the Southern Appalachians (Chapters I, IV). Three goals were established for this dissertation in order to begin to rectify the lack of study in the Interior Highlands: 1) intensively survey a single site and identify as many terrestrial arthropods to species as possible in order to establish a baseline list of taxa against which future change can be compared; 2) compare collecting methods used for terrestrial arthropods in order to determine the most efficient combination of traps and the minimum number of samples needed to collect most species so future surveys in similar environments can maximize the return of effort; and 3) report rare and endemic terrestrial arthropods, as well as species that are new to Arkansas, in order to better understand the arthropods native to the state.

An intensive nine month survey was conducted at a 4 hectare plot established at Steel Creek, Buffalo National River, in Newton County, Arkansas. Thirteen collecting methods – twelve trap types (three colors of Lindgren funnel trap, five colors of pan trap, Malaise traps, canopy traps with upper and lower collectors, and pitfall traps), which were run continuously between collections, and Berlese-Tullgren extraction of leaf litter, which was collected when traps were serviced – were employed, with a total of 80 samples being collected approximately every two weeks. A total of 1311 samples were taken during the course of 17 sample dates; 49 samples were lost to rain, animal disturbance, &c. and account for the disparity in the total number of samples collected.

Target bioindicator groups – including Formicidae, Carabidae, Cerambycidae, Curculionoidea, and Araneae – and easily identified taxa – including Isopoda, Mecoptera,

Phasmida, Vespidae, Ixodidae, Phalangodidae, and select Diplopoda and Orthoptera – as well as taxa that were identified by willing experts (Parasitengona, ‘Symphyta’, Pompilidae) were coarse-sorted and identified to species. This resulted in 47,481 specimens representing 706 species that were curated and identified, including 18 putatively undescribed species, 56 species that represented new state records, 15 non-native species, and three species endemic to the Interior Highlands, two of which were previously known only from the original type series (Appendix II).

Four beetle taxa – Buprestidae (375 specimens, 27 species), Carabidae (1970 specimens, 62 species), Cerambycidae (1885 specimens, 82 species), and Curculionoidea (Anthribidae: 15 specimens, 4 species; Attelabidae: 19 specimens, 3 species, 3 genera; Brachyceridae: 1 specimen, 1 species; Brentidae: 6 specimens, 1 species; Curculionidae: 3777 specimens, 71 species) were analyzed individually. The phenology of the sampled populations at higher (superfamily/family) and species level was examined. The number of species and specimens collected per trap type were compared using ANOVA and, if statistical differences were found, further compared using Tukey’s HSD statistics and by estimating the total number of species a trap type is expected to collect at the site using extrapolated rarefaction curves. Extrapolated rarefaction curves were also used to determine the minimum number of samples that should be collected per trap type before species saturation – that is, the number of samples after which no new species are collected – is reached. Overlap between trap types was compared by determining the similarity of species collected between trap types using Sørensen and Chao’s Sørensen indices. The most effective collecting method or methods for each superfamily/family was the method or methods that collected the highest number of species and, in the case of two methods, exhibited reasonably low similarity.

Pitfall and Malaise traps were determined to be the most effective combination of collection methods for Carabidae, Curculionoidea, and the combined taxa. Pitfall traps are generally thought to be the best method to collect carabids and are often employed as many species are epigeal (e.g., Greenslade 1964; Baars 1979; Waage 1985; Desender & Maelfait 1986; Halsall & Wratten 1988; Morrill et al. 1990; Niemelä et al. 1990; Wiedenmann et al. 1992; Work et al. 2002; Raworth & Choi 2003; Buchholz et al. 2010). However, Ulyshen et al. (2005) reported that canopy traps (top + bottom collector) collect smaller, more aerial carabid species more effectively than pitfall traps and should be used in combination with pitfall traps when surveying carabid diversity, so it is unsurprising that Malaise traps, which operate similarly to canopy traps, were an excellent complement to pitfall traps.

Weevils are a diverse group of beetles and no one method can be employed that adequately samples their diversity. The most effective combination of traps should target both aerial and terrestrial species and both pitfall (Raffa & Hunt 1988; Levesque & Levesque 1994; Hanula 1990) and Malaise traps (Dutcher et al. 1986; Anderson 2008; Ohsawa 2008; Hespenheide 2009) have been used to survey weevils. That pitfall and Malaise traps are the most effective combination of terrestrial and aerial traps is unsurprising given they were the most effective combination of traps when collecting all taxa. It may also be that weevils, with their diverse habits, are good indicators of terrestrial arthropod biodiversity, though a more definitive statement cannot be made based on the data presented herein.

Malaise traps were the most effective method for collecting Cerambycidae as all other aerial trap types (canopy, all colors of Lindgren funnel) exhibited high similarity with Malaise traps. If Lindgren funnel traps are to be used, it is useful to note that one species, *Xylotrechus colonus*, of the nine analyzed was collected in significantly higher numbers in one trap color

(black). Only a handful of studies examining color attraction in Cerambycidae have been published; some found increased attraction to red (Shipman 2011), purple (Skvarla and Holland 2011), and yellow (Sakalian et al. 1993, Imrei et al. 2014) at the family level or within individual species, though others found no effect of color (Macias-Samano n.d.). It is likely that color attraction is species-specific and tied to biological traits, such as nectivory. The response to unbaited Lindgren funnel traps regardless of color suggests that many cerambycids may be attracted to the vertical silhouette of the trap. A comprehensive study with multiple colors and multiple trap types is needed before this issue is settled.

The combination of Malaise or canopy trap and green Lindgren funnel traps was most effective for Buprestidae. Malaise and canopy traps exhibited very high similarity with each other but much lower similarity with Lindgren funnel traps. Malaise and canopy traps collected large species (e.g., *Chrysobothris*, *Dicerca*) in much higher abundance than other trap types while Lindgren funnels collected smaller species (e.g., *Agrilus*, *Taphrocerus*) in higher abundance than other trap types.

Trap color is an important component of Lindgren funnel traps when targeting buprestids. Green and purple Lindgren funnel traps exhibited only medium similarity in the species collected and differentially peaked in the number of species and specimens collected. Six of seven species analyzed were collected in significantly higher numbers by specific colored traps: four were caught in higher numbers by green traps, one by purple traps, and one by black traps. Other studies have examined the role of color in attraction and trapping of Buprestidae but most have either focused at the family level or on economically important species (e.g., emerald ash borer, *Agrilus planipennis*). However, two studies (i.e., Petrice et al. 2013, Peatrice & Haack 2015) found that, while there was no difference in the attraction of emerald ash borer to green or purple

traps, other *Agrilus* species demonstrated significant preference for green or green and purple traps. It is probable then that green and purple Lindgren traps differentially attract *Agrilus* species and that the bulk of studies that have examined color preference in emerald ash borer, which have focused primarily on purple traps, may not be applicable to other *Agrilus* or buprestids in general.

A combined “all taxa” analysis was performed using the same statistics that were used to analyze beetle data. It should be noted that a number of species were excluded from the all taxa analyses for a variety of reasons: 1) only one or a few specimens of a species were examined and identified, so the data did not accurately reflect the total abundance of the species. For example, the first specimen of *Lygistorrhina sancthecatharinae* (Diptera) was found and identified after more than half of the samples were sorted. It was examined because it is a rare and interesting fly that is new to Arkansas, but the species presence or absence in previously sorted samples could not be determined; 2) the collecting methods employed preferentially damaged certain specimens, resulting in biased collections. Lepidoptera is an excellent example of this as the wet collection jars frequently resulted in poor specimens that lacked wing scales, especially among smaller species; 3) specimens were identified after the analyses were completed. These are included in the final list of arthropods collected at Steel Creek for completeness but were not available when the analyses were conducted. After excluding species based on these criteria, 46,146 specimens representing 533 species were included in the all taxa analyses.

Terrestrial collection methods (i.e., pitfall traps and Berlese-Tullgren extraction) and aerial traps (i.e., Malaise, canopy, and Lindgren funnel traps) generally exhibited high to very high similarity within each group but lower similarity between the groups. They are likely

targeting different arthropod assemblages and a combination of techniques is required if maximum diversity is to be sampled.

Pitfall and Malaise traps collected the most species on average, with pitfall, Malaise, and purple pan traps estimated to collect the most species after species accumulation curves become asymptotic. While this is certainly influenced by the taxa included in the analysis, and slightly different results would be obtained if different taxa were included, it likely reflects the true performance of the different trap types because: 1) all individuals from a diversity of higher taxa were included, limiting the influence any one taxon would have on the results and 2) the inclusion of a number of easily-identified species from an even wider range of orders and families introduces additional variation in life-history and minimizes the impact of expert-bias when picking which taxa are included.

Pan traps generally exhibited high similarity with terrestrial and aerial traps (except green Lindgren funnel traps), probably because they collect both flying and crawling insects, though generally underperformed in the number of species collected when compared to Malaise or pitfall traps. The pan traps in this study, however, were placed under Malaise traps and were not buried flush with the substrate. It is possible that many flying insects did not see the pan traps because of their placement under the Malaise traps and they likely missed many arthropods that would fall into a pitfall trap but could not scale the sides of the pan. One potential solution is to combine pitfall and pan traps by using open, colored pitfalls flush with the substrate (Skvarla *et al.* 2014; Ernst *et al.* 2015). The only study that compared the effect of color in pitfall traps found that flying pollinators and carabids were collected in higher numbers in white and yellow (except carabids) pitfall traps compared to green and brown pitfalls and that terrestrial taxa, such as Isopoda, were not affected by trap color (Buchholz *et al.* 2010). However, such pitfalls cannot

be run with rain covers and will be more affected by rainfall than covered traps when run for extended periods of time, though it may be possible to employ a clear rain cover without affecting the attractiveness of the trap to flying insects.

51% of the species analyzed were represented by five or fewer specimens and 25% were represented by singletons. The species accumulation curves for most trap types did not become asymptotic and extrapolated rarefaction curves predicted 300–600 samples are required per trap type (1000+ for pitfall traps), far more than were collected during this study, before species accumulation is saturated. This suggests that even though the site was relatively small, a great deal more effort is required before the majority of species are sampled.

However, even though fewer than half of the species predicted by the species accumulation curves were collected, the survey still produced 18 new species and 56 new state records within the identified taxa.

Finally, one specimen and three species collected during the survey that represent rarely collected mutations or taxa were examined in detail.

A single specimen of *Temnothorax curvispinosus* (Formicidae) exhibiting gynandromorphism was collected and represented the first time this anomaly was seen in the species; additionally, it was the only gynandromorph collected out of more than 28,000 ants examined during the study and demonstrates the potential rarity of the condition among able-bodied, foraging workers.

One and two specimens of *Eudociminus mannerheimii* (Curculionidae) and *Orussus minutus* (Orussidae) were collected and represent major range extensions for the species. *Eudociminus mannerheimii* has been previously recorded only from coastal states from New York south to Florida, west to Louisiana and Mexico, so the specimens (four additional

specimens were collected a few hundred yards from the Steel Creek survey site) represent the northwestern-most, inland records for the species. Additionally, we hypothesized the larval host plant in Arkansas to be eastern red cedar (*Juniperus virginiana*) as it is the only representative of Cupressaceae, the only family the beetles are known to colonize, at the collection site. *Orussus minutus* was previously known from as far west as Indiana and Georgia, so the Arkansas specimens represent a significant western range extension. Prior to publication, only approximately 30 specimens of *O. minutus* were known; by incorporating locality and collection data from specimens housed in the United States National Collection, we nearly tripled the number of published specimens and showed that both Malaise and black funnel traps can be used to collect the species.

Ninety one specimens of *Merope tuber* (Meropeidae) were collected at Steel Creek, the second largest collection of the species recorded. The species has generally been considered to be rare and is often found incidentally as bycatch in Malaise traps. Eighty seven specimens were collected in pan traps during the survey – the first time the species had been collected with that method. The high proportion of specimens collected in pan traps suggests that pan traps or pan traps combined with intercept traps may be a more effective alternative than Malaise and other trap types which have been used to collect it in the past. Additionally, we discussed the phenology of the specimens collected and tested a previously proposed hypothesis that male exhibit a bimodal distribution of large and small claspers; we found that the claspers in the population sampled at Steel Creek did not exhibit a bimodal distribution and instead were normally distributed.

The number of new species, new state records, and highlighted specimen and species illustrate the fact that not only does much work remain to be done in Arkansas, which is under

studied compared to similar areas, but also how much is left to discover even in a well-worked region such as North America north of Mexico.

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XIV. Appendix I. Statistical Analyses Workflow.

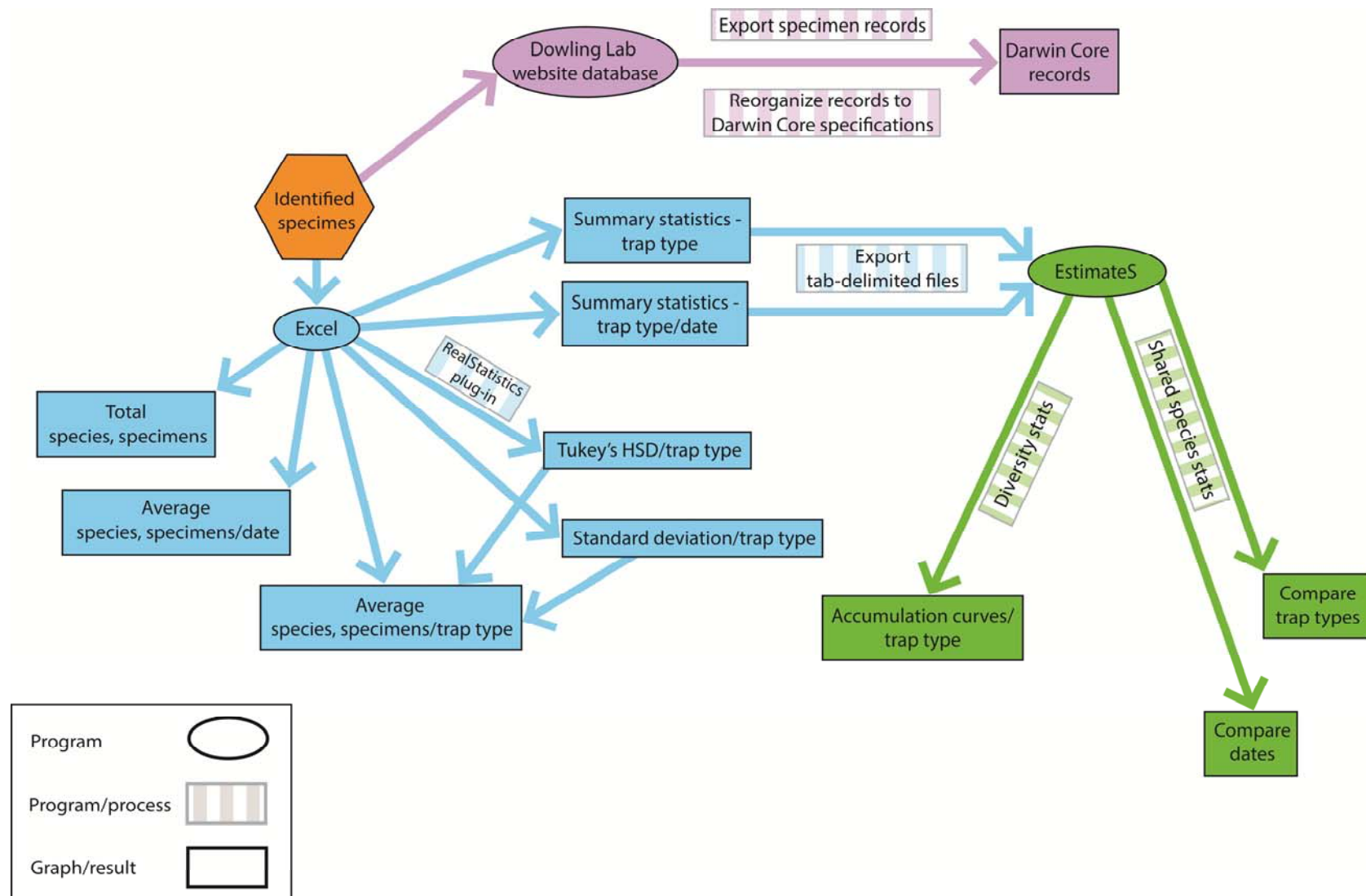


Figure A2a. Workflow for statistical analyses conducted in Chapters V and VI. Different colors represent different programs or websites.

XV. Appendix II. Arthropod species collected at Steel Creek

The following list includes all arthropod species identified from Steel Creek, including those species included in Chapter IV–XI and species that were excluded from analyses because 1) every specimen of the species was not identified (e.g., a few Encyrtidae were sorted and sent to John Noyes for identification at the Natural History Museum in London, but many specimens were left unsorted in trap residue); 2) the collection methods were obviously biased or were poor at collecting/preserving the specimens (e.g., Lepidoptera); or 3) the identifications were made after the final analyses were completed but before this dissertation was submitted (e.g., “Symphyta”, Heteroptera). Species indicated as new state records here may have been reported previously in publications that have resulted from this work (e.g., *Orussus minutus* in Chapter VIII, *Eudocimus mannerheimii* in Chapter IX). Additionally, the number of new state records reported here likely underrepresents the true number of new records as no attempt was made to establish previous occurrences of some species (e.g., Cerambycidae, Encyrtidae, “Symphyta”).

Class	Order	Family	Genus	Species
Arachnida	Acariformes	Calyptostomatidae	<i>Calyptostoma</i>	<i>Calyptosoma</i>
Arachnida	Acariformes	Cunaxidae	<i>Parabonzia</i>	<i>Parabonzia</i> <i>bdelliformis</i>
Arachnida	Acariformes	Erythraeidae	<i>Abrolophus</i>	sp. 1 [†]
Arachnida	Acariformes	Erythraeidae	<i>Caeculisoma</i>	sp. 1 [†]
Arachnida	Acariformes	Erythraeidae	<i>Callidosoma</i>	sp. 1 [†]
Arachnida	Acariformes	Erythraeidae	<i>Erythraeus</i>	sp. 1 [†]
Arachnida	Acariformes	Erythraeidae	<i>Leptus</i>	plate Leptus [†]
Arachnida	Acariformes	Erythraeidae	<i>Leptus</i>	red Leptus [†]
Arachnida	Acariformes	Erythraeidae	<i>Leptus</i>	spotted Leptus [†]
Arachnida	Acariformes	Erythraeidae	<i>Paraphanolophus</i>	sp. 1 [†]
Arachnida	Acariformes	Microtrombidiidae	<i>Willmannella</i>	sp. 1 [†]
Arachnida	Acariformes	Podothrombidiidae	<i>Podothrombidium</i>	sp. 1 [†]
Arachnida	Acariformes	Trombidiidae	<i>Trombidium</i>	Trombidium, yellow-shouldered [†]
Arachnida	Araneae	Agelenidae	<i>Agelenopsis</i>	<i>Agelenopsis</i> <i>kastoni</i>
Arachnida	Araneae	Agelenidae	<i>Agelenopsis</i>	<i>Agelenopsis naevia</i>
Arachnida	Araneae	Agelenidae	<i>Agelenopsis</i>	<i>Agelenopsis</i> <i>pennsylvanica</i>
Arachnida	Araneae	Agelenidae	<i>Wadotes</i>	sp. 1
Arachnida	Araneae	Anyphaenidae	<i>Anyphaena</i>	<i>Anyphaena celer</i>
Arachnida	Araneae	Araneidae	<i>Araneus</i>	<i>Araneus partitus</i>
Arachnida	Araneae	Araneidae	<i>Araniella</i>	<i>Araniella</i> <i>displicata</i>
Arachnida	Araneae	Araneidae	<i>Eustala</i>	<i>Eustala anastera</i>
Arachnida	Araneae	Araneidae	<i>Hypsosinga</i>	<i>Hypsosinga rubens</i>
Arachnida	Araneae	Araneidae	<i>Mangora</i>	<i>Mangora placida</i>
Arachnida	Araneae	Araneidae	<i>Neoscona</i>	<i>Neoscona</i> <i>crucifera</i>
Arachnida	Araneae	Araneidae	<i>Ocrepeira</i>	
Arachnida	Araneae	Atypidae	<i>Sphodros</i>	<i>Sphodros niger</i>
Arachnida	Araneae	Clubionidae	<i>Elaver</i>	<i>Elaver excepta</i>
Arachnida	Araneae	Corinnidae	<i>Castianeira</i>	<i>Castianeira</i> <i>amoena</i>
Arachnida	Araneae	Corinnidae	<i>Castianeira</i>	<i>Castianeira</i> <i>cingulata</i>

Table A3. Species identified from Steel Creek during this dissertation.

[†] - putative new species, [‡] - introduced, non-native species, [§] - new state record

Class	Order	Family	Genus	Species
Arachnida	Araneae	Corinnidae	<i>Castianeira</i>	<i>Castianeira crocata</i>
Arachnida	Araneae	Corinnidae	<i>Castianeira</i>	<i>Castianeira descripta</i>
Arachnida	Araneae	Corinnidae	<i>Castianeira</i>	<i>Castianeira longipalpa</i>
Arachnida	Araneae	Corinnidae	<i>Castianeira</i>	<i>Castianeira trilineata</i>
Arachnida	Araneae	Ctenidae	<i>Ctenus</i>	<i>Ctenus exlineae</i>
Arachnida	Araneae	Ctenizidae	<i>Ummidia</i>	sp. 1 (small) [†]
Arachnida	Araneae	Dictynidae	<i>Cicurina</i>	
Arachnida	Araneae	Euctenizidae	<i>Myrmekiaphila</i>	<i>Myrmekiaphila comstocki</i>
Arachnida	Araneae	Gnaphosidae	<i>Callilepis</i>	<i>Callilepis imbecilla</i>
Arachnida	Araneae	Gnaphosidae	<i>Cesonia</i>	<i>Cesonia bilineata</i>
Arachnida	Araneae	Gnaphosidae	<i>Drassyllus</i>	<i>Drassyllus aprilinus</i>
Arachnida	Araneae	Gnaphosidae	<i>Drassyllus</i>	<i>Drassyllus covensis</i>
Arachnida	Araneae	Gnaphosidae	<i>Drassyllus</i>	<i>Drassyllus dixinus</i>
Arachnida	Araneae	Gnaphosidae	<i>Drassyllus</i>	<i>Drassyllus novus</i>
Arachnida	Araneae	Gnaphosidae	<i>Drassyllus</i>	<i>Drassyllus rufulus</i>
Arachnida	Araneae	Gnaphosidae	<i>Gnaphosa</i>	<i>Gnaphosa fontinalis</i>
Arachnida	Araneae	Gnaphosidae	<i>Haplodrassus</i>	<i>Haplodrassus signifer</i>
Arachnida	Araneae	Gnaphosidae	<i>Herpyllus</i>	<i>Herpyllus ecclesiasticus</i>
Arachnida	Araneae	Gnaphosidae	<i>Litopyllus</i>	<i>Litopyllus temporarius</i>
Arachnida	Araneae	Gnaphosidae	<i>Micaria</i>	<i>Micaria longipes</i>
Arachnida	Araneae	Gnaphosidae	<i>Nodocion</i>	<i>Nodocion floridanus</i>
Arachnida	Araneae	Gnaphosidae	<i>Sergiolus</i>	<i>Sergiolus capulatus</i>
Arachnida	Araneae	Gnaphosidae	<i>Sergiolus</i>	<i>Sergiolus tennesseensis</i>
Arachnida	Araneae	Gnaphosidae	<i>Sosticus</i>	<i>Sosticus insularis</i>
Arachnida	Araneae	Gnaphosidae	<i>Talanites</i>	<i>Talanites echinus</i>
Arachnida	Araneae	Gnaphosidae	<i>Zelotes</i>	<i>Zelotes duplex</i>

Table A3 (cont.). Species identified from Steel Creek during this dissertation.

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Class	Order	Family	Genus	Species
Arachnida	Araneae	Hahniidae	<i>Neoantistea</i>	<i>Neoantistea agilis</i>
Arachnida	Araneae	Lycosidae	<i>Arctosa</i>	<i>Arctosa virgo</i>
Arachnida	Araneae	Lycosidae	<i>Gladicosa</i>	<i>Gladicosa gulosa</i>
Arachnida	Araneae	Lycosidae	<i>Gladicosa</i>	<i>Gladicosa pulchra</i>
Arachnida	Araneae	Lycosidae	<i>Pirata</i>	sp. 1
Arachnida	Araneae	Lycosidae	<i>Rabidosa</i>	<i>Rabidosa punctulata</i>
Arachnida	Araneae	Lycosidae	<i>Rabidosa</i>	<i>Rabidosa rabida</i>
Arachnida	Araneae	Lycosidae	<i>Schizocosa</i>	<i>Schizocosa bilineata</i>
Arachnida	Araneae	Lycosidae	<i>Schizocosa</i>	<i>Schizocosa duplex</i>
Arachnida	Araneae	Lycosidae	<i>Schizocosa</i>	<i>Schizocosa ocreata</i>
Arachnida	Araneae	Lycosidae	<i>Schizocosa</i>	<i>Schizocosa saltatrix</i>
Arachnida	Araneae	Lycosidae	<i>Tigrosa</i>	<i>Tigrosa georgicola</i>
Arachnida	Araneae	Lycosidae	<i>Trochosa</i>	<i>Trochosa ruricola</i>
Arachnida	Araneae	Lycosidae	<i>Varacosa</i>	<i>Varacosa avara</i>
Arachnida	Araneae	Lycosidae	<i>Varacosa</i>	<i>Varacosa shenandoa</i>
Arachnida	Araneae	Mimetidae	<i>Mimetes</i>	<i>Mimetes puritanus</i>
Arachnida	Araneae	Oxyopidae	<i>Oxyopes</i>	<i>Oxyopes aglossus</i>
Arachnida	Araneae	Oxyopidae	<i>Oxyopes</i>	<i>Oxyopes salticus</i>
Arachnida	Araneae	Philodromidae	<i>Ebo</i>	<i>Ebo latithorax</i>
Arachnida	Araneae	Philodromidae	<i>Philodromus</i>	<i>Philodromus minutus</i>
Arachnida	Araneae	Philodromidae	<i>Philodromu</i>	<i>Philodromus praelustrus</i>
Arachnida	Araneae	Phrurolithidae	<i>Phrurotimpus</i>	<i>Phrurotimpus alarius</i>
Arachnida	Araneae	Phrurolithidae	<i>Phrurotimpus</i>	<i>Phrurotimpus borealis</i>
Arachnida	Araneae	Phrurolithidae	<i>Phrurotimpus</i>	<i>Phrurotimpus</i> sp. 3 [†]
Arachnida	Araneae	Phrurolithidae	<i>Scotinella</i>	<i>Scotinella redempta</i>
Arachnida	Araneae	Phrurolithidae	<i>Scotinella</i>	<i>Scotinella</i> sp 2
Arachnida	Araneae	Pisauridae	<i>Dolomedes</i>	<i>Dolomedes tenebrosus</i>
Arachnida	Araneae	Pisauridae	<i>Pisaurina</i>	<i>Pisaurina mira</i>

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Class	Order	Family	Genus	Species
Arachnida	Araneae	Salticidae	sp. 1 male	
Arachnida	Araneae	Salticidae	<i>Chinattus</i>	<i>Chinattus parvulus</i>
Arachnida	Araneae	Salticidae	<i>Eris</i>	<i>Eris militaris</i>
Arachnida	Araneae	Salticidae	<i>Habronattus</i>	<i>Habronattus orbus</i>
Arachnida	Araneae	Salticidae	<i>Maevia</i>	<i>Maevia inclemens</i>
Arachnida	Araneae	Salticidae	<i>Naphrys</i>	<i>Naphrys pulex</i>
Arachnida	Araneae	Salticidae	<i>Paraphidippus</i>	<i>Paraphidippus aurantius</i>
Arachnida	Araneae	Salticidae	<i>Peckhamia</i>	
Arachnida	Araneae	Salticidae	<i>Pelegrina</i>	<i>Pelegrina galathea</i>
Arachnida	Araneae	Salticidae	<i>Pelegrina</i>	<i>Pelegrina proterva</i>
Arachnida	Araneae	Salticidae	<i>Phidippus</i>	<i>Phidippus carolinensis</i>
Arachnida	Araneae	Salticidae	<i>Phidippus</i>	<i>Phidippus clarus</i>
Arachnida	Araneae	Salticidae	<i>Phidippus</i>	<i>Phidippus whitmani</i>
Arachnida	Araneae	Salticidae	<i>Talvera</i>	<i>Talvera minuta</i>
Arachnida	Araneae	Salticidae	<i>Thiodina sylvana</i>	<i>Thiodina sylvana</i>
Arachnida	Araneae	Salticidae	<i>Zygoballus</i>	<i>Zygoballus rufipes</i>
Arachnida	Araneae	Segestriidae	<i>Ariadna</i>	<i>Ariadna bicolor</i>
Arachnida	Araneae	Tetragnathidae	<i>Leucauge</i>	<i>Leucauge ventusa</i>
Arachnida	Araneae	Theridiidae	<i>Asagena</i>	<i>Asagena americana</i>
Arachnida	Araneae	Theridiidae	<i>Crustulina</i>	<i>Crustulina altera</i>
Arachnida	Araneae	Theridiidae	<i>Latrodectus</i>	<i>Latrodectus mactans</i>
Arachnida	Araneae	Theridiidae	<i>Neospintharus</i>	
Arachnida	Araneae	Theridosomatidae	<i>Theridiosoma</i>	<i>Theridiosoma gemmosum</i>
Arachnida	Araneae	Thomisidae	<i>Bassaniana</i>	<i>Bassaniana versicolor</i>
Arachnida	Araneae	Thomisidae	<i>Misumena</i>	<i>Misumena vatia</i>
Arachnida	Araneae	Thomisidae	<i>Ozyptila</i>	<i>Ozyptila monroensis</i>
Arachnida	Araneae	Thomisidae	<i>Tmarus</i>	<i>Tmarus angulatus</i>
Arachnida	Araneae	Thomisidae	<i>Xysticus</i>	<i>Xysticus elegans</i>
Arachnida	Araneae	Thomisidae	<i>Xysticus</i>	<i>Xysticus ferox</i>
Arachnida	Araneae	Thomisidae	<i>Xysticus</i>	<i>Xysticus fraternus</i>

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Class	Order	Family	Genus	Species
Arachnida	Araneae	Thomisidae	<i>Xysticus</i>	<i>Xysticus funestis</i>
Arachnida	Araneae	Thomisidae	<i>Xysticus</i>	<i>Xysticus pella</i>
Arachnida	Araneae	Titanoecidae	<i>Titanoeca</i>	<i>Titanoeca brunnea</i>
Arachnida	Araneae	Trachelidae	<i>Meriola</i>	<i>Meriola decepta</i>
Arachnida	Opiliones	Phalangodidae	<i>Crosbyella</i>	
Arachnida	Opiliones	Phalangodidae	<i>Wespus</i>	<i>Wespus</i> <i>arkansasensis</i>
Arachnida	Parasitiformes	Ixodidae	<i>Amblyomma</i>	<i>Amblyomma</i> <i>americanum</i>
Arachnida	Parasitiformes	Ixodidae	<i>Dermacentor</i>	<i>Dermacentor</i> <i>variabilis</i>
Arachnida	Parasitiformes	Ixodidae	<i>Ixodes</i>	<i>Ixodes scapularis</i>
Chilopoda	Scolopendromorpha	Plutoniumidae	<i>Theatops</i>	<i>Theatops</i> <i>spinicaudatus</i>
Diplopoda	Callipodida	Abacionidae	<i>Abacion</i>	<i>Abacion texense</i>
Diplopoda	Callipodida	Abacionidae	<i>Abacion</i>	<i>Abacion tessellatum</i>
Diplopoda	Platydesmida	Andrognathidae	<i>Brachycybe</i>	<i>Brachycybe lecontei</i>
Diplopoda	Polydesmida	Euryuridae	<i>Auturus</i>	<i>Auturus evides</i>
Diplopoda	Polydesmida	Sphaeriodesmidae	<i>Desmonus</i>	<i>Desmonus pudicus</i>
Diplopoda	Polydesmida	Xystodesmidae	<i>Apheloria</i>	<i>Apheloria</i> <i>virginiensis reducta</i>
Diplopoda	Polydesmida	Xystodesmidae	<i>Nannaria</i>	<i>Nannaria</i> <i>davidcauseyi</i>
Diplopoda	Polyxenida	Polyxenidae	<i>Polyxenus</i>	<i>Polyxenus</i> <i>largurus</i> ^{†?}
Diplopoda	Spirobolida	Spirobolidae	<i>Narceus</i>	<i>Narceus americanus</i> <i>complex</i>

Table A3 (cont.). Species identified from Steel Creek during this dissertation.

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Class	Order	Family	Genus	Species
Insecta	Blattodea	Rhinotermitidae	<i>Reticulitermes</i>	<i>Reticulitermes flavipes</i>
Insecta	Blattodea	Rhinotermitidae	<i>Reticulitermes</i>	<i>Reticulitermes hageni</i>
Insecta	Coleoptera	Anthribidae	<i>Euparius</i>	<i>Euparius marmoreus</i>
Insecta	Coleoptera	Anthribidae	<i>Eurymycter</i>	<i>Eurymycter fasciatus</i>
Insecta	Coleoptera	Anthribidae	<i>Ormiscus</i>	sp 1
Insecta	Coleoptera	Anthribidae	<i>Toxonotus</i>	<i>Toxonotus cornutus</i>
Insecta	Coleoptera	Attelabidae	<i>Eugnamptus</i>	<i>Eugnamptus angustatus</i>
Insecta	Coleoptera	Attelabidae	<i>Synolabus</i>	<i>Synolabus bipustulatus</i>
Insecta	Coleoptera	Attelabidae	<i>Temnocerus</i>	<i>Temnocerus aeratus</i>
Insecta	Coleoptera	Brachyceridae	<i>Notiodes</i>	<i>Notiodes limatulus</i>
Insecta	Coleoptera	Brentidae	<i>Arrhenodes</i>	<i>Arrhenodes minutus</i>
Insecta	Coleoptera	Buprestidae	<i>Acmaeodera</i>	<i>Acmaeodera tubulus</i>
Insecta	Coleoptera	Buprestidae	<i>Acmaeodera</i>	<i>Acmaeodera pulchella</i>
Insecta	Coleoptera	Buprestidae	<i>Actenodes</i>	<i>Actenodes acornis</i> [§]
Insecta	Coleoptera	Buprestidae	<i>Agrilus</i>	<i>Agrilus arcuatus complex</i>
Insecta	Coleoptera	Buprestidae	<i>Agrilus</i>	<i>Agrilus bilineatus</i>
Insecta	Coleoptera	Buprestidae	<i>Agrilus</i>	<i>Agrilus cephalicus</i> [§]
Insecta	Coleoptera	Buprestidae	<i>Agrilus</i>	<i>Agrilus defectus</i>
Insecta	Coleoptera	Buprestidae	<i>Agrilus</i>	<i>Agrilus fallax</i>
Insecta	Coleoptera	Buprestidae	<i>Agrilus</i>	<i>Agrilus geminatus</i>
Insecta	Coleoptera	Buprestidae	<i>Agrilus</i>	<i>Agrilus lecontei</i>
Insecta	Coleoptera	Buprestidae	<i>Agrilus</i>	<i>Agrilus masculinus</i>
Insecta	Coleoptera	Buprestidae	<i>Agrilus</i>	<i>Agrilus ohioensis</i> [§]
Insecta	Coleoptera	Buprestidae	<i>Agrilus</i>	<i>Agrilus olentangyi</i>
Insecta	Coleoptera	Buprestidae	<i>Agrilus</i>	<i>Agrilus obsoletoguttatus</i>
Insecta	Coleoptera	Buprestidae	<i>Agrilus</i>	<i>Agrilus paracelti</i> [§]
Insecta	Coleoptera	Buprestidae	<i>Anthaxia</i>	<i>Anthaxia viridifrons</i>
Insecta	Coleoptera	Buprestidae	<i>Brachys</i>	<i>Brachys aerosus</i>
Insecta	Coleoptera	Buprestidae	<i>Chrysobothris</i>	<i>Chrysobothris adelpha</i>
Insecta	Coleoptera	Buprestidae	<i>Chrysobothris</i>	<i>Chrysobothris femorata complex</i>
Insecta	Coleoptera	Buprestidae	<i>Chrysobothris</i>	<i>Chrysobothris sexsignata</i>
Insecta	Coleoptera	Buprestidae	<i>Dicerca</i>	<i>Dicerca divaricata</i>
Insecta	Coleoptera	Buprestidae	<i>Dicerca</i>	<i>Dicerca lurida</i>
Insecta	Coleoptera	Buprestidae	<i>Dicerca</i>	<i>Dicerca obscura</i>
Insecta	Coleoptera	Buprestidae	<i>Dicerca</i>	<i>Dicerca spreta</i>
Insecta	Coleoptera	Buprestidae	<i>Ptosima</i>	<i>Ptosima gibbicollis</i>
Insecta	Coleoptera	Buprestidae	<i>Taphrocerus</i>	<i>Taphrocerus gracilis</i>
Insecta	Coleoptera	Buprestidae	<i>Taphrocerus</i>	<i>Taphrocerus nicolayi</i> [§]

Table A3 (cont.). Species identified from Steel Creek during this dissertation.

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Class	Order	Family	Genus	Species
Insecta	Coleoptera	Carabidae	<i>Agonoleptus</i>	<i>Agonoleptus conjunctus</i>
Insecta	Coleoptera	Carabidae	<i>Agonum</i>	<i>Agonum striatopunctatum</i>
Insecta	Coleoptera	Carabidae	<i>Agonum</i>	<i>Agonum punctiforme</i> [§]
Insecta	Coleoptera	Carabidae	<i>Amara</i>	<i>Amara aenea</i> [‡]
Insecta	Coleoptera	Carabidae	<i>Amara</i>	<i>Amara cupreolata</i>
Insecta	Coleoptera	Carabidae	<i>Amara</i>	<i>Amara musculus</i>
Insecta	Coleoptera	Carabidae	<i>Anisodactylus</i>	<i>Anisodactylus rusticus</i>
Insecta	Coleoptera	Carabidae	<i>Apenes</i>	<i>Apenes sinuata</i>
Insecta	Coleoptera	Carabidae	<i>Badister</i>	<i>Badister notatus</i>
Insecta	Coleoptera	Carabidae	<i>Bembidion</i>	<i>Bembidion affine</i>
Insecta	Coleoptera	Carabidae	<i>Bembidion</i>	<i>Bembidion rapidum</i>
Insecta	Coleoptera	Carabidae	<i>Brachinus</i>	<i>Brachinus americanus</i>
Insecta	Coleoptera	Carabidae	<i>Calathus</i>	<i>Calathus opaculus</i>
Insecta	Coleoptera	Carabidae	<i>Calleida</i>	<i>Calleida viridipennis</i>
Insecta	Coleoptera	Carabidae	<i>Carabus</i>	<i>Carabus sylvosus</i>
Insecta	Coleoptera	Carabidae	<i>Chlaenius</i>	<i>Chlaenius platyderus</i>
Insecta	Coleoptera	Carabidae	<i>Chlaenius</i>	<i>Chlaenius tomentosus</i>
Insecta	Coleoptera	Carabidae	<i>Cicindela</i>	<i>Cicindela rufiventris</i>
Insecta	Coleoptera	Carabidae	<i>Cicindela</i>	<i>Cicindela sexguttata</i>
Insecta	Coleoptera	Carabidae	<i>Clinidium</i>	<i>Clinidium sculptile</i>
Insecta	Coleoptera	Carabidae	<i>Clivina</i>	<i>Clivina pallida</i>
Insecta	Coleoptera	Carabidae	<i>Cyclotrachelus</i>	<i>Cyclotrachelus incisus</i>
Insecta	Coleoptera	Carabidae	<i>Cyclotrachelus</i>	<i>Cyclotrachelus parasodalis</i>
Insecta	Coleoptera	Carabidae	<i>Cymindis</i>	<i>Cymindis americana</i>
Insecta	Coleoptera	Carabidae	<i>Cymindis</i>	<i>Cymindis limbata</i>
Insecta	Coleoptera	Carabidae	<i>Cymindis</i>	<i>Cymindis platycollis</i>
Insecta	Coleoptera	Carabidae	<i>Dicaelus</i>	<i>Dicaelus ambiguus</i>
Insecta	Coleoptera	Carabidae	<i>Dicaelus</i>	<i>Dicaelus elongatus</i>
Insecta	Coleoptera	Carabidae	<i>Dicaelus</i>	<i>Dicaelus sculptilis</i>
Insecta	Coleoptera	Carabidae	<i>Dromius</i>	<i>Dromius piceus</i>
Insecta	Coleoptera	Carabidae	<i>Elaphropus</i>	<i>Elaphropus granarius</i>
Insecta	Coleoptera	Carabidae	<i>Galerita</i>	<i>Galerita bicolor</i>
Insecta	Coleoptera	Carabidae	<i>Galerita</i>	<i>Galerita janus</i>
Insecta	Coleoptera	Carabidae	<i>Harpalus</i>	<i>Harpalus faunus</i>
Insecta	Coleoptera	Carabidae	<i>Harpalus</i>	<i>Harpalus katiae</i>
Insecta	Coleoptera	Carabidae	<i>Harpalus</i>	<i>Harpalus pensylvanicus</i>
Insecta	Coleoptera	Carabidae	<i>Lebia</i>	<i>Lebia analis</i>

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Class	Order	Family	Genus	Species
Insecta	Coleoptera	Carabidae	<i>Lebia</i>	<i>Lebia marginicollis</i>
Insecta	Coleoptera	Carabidae	<i>Lebia</i>	<i>Lebia viridis</i>
Insecta	Coleoptera	Carabidae	<i>Lophoglossus</i>	<i>Lophoglossus haldemanni</i>
Insecta	Coleoptera	Carabidae	<i>Mioptachys</i>	<i>Mioptachys flavicauda</i>
Insecta	Coleoptera	Carabidae	<i>Notiophilus</i>	<i>Notiophilus novemstriatus</i>
Insecta	Coleoptera	Carabidae	<i>Platynus</i>	<i>Platynus decentis</i>
Insecta	Coleoptera	Carabidae	<i>Platynus</i>	<i>Platynus paramarginatus</i>
Insecta	Coleoptera	Carabidae	<i>Plochionus</i>	<i>Plochionus timidus</i>
Insecta	Coleoptera	Carabidae	<i>Pterostichus</i>	<i>Pterostichus permundus</i>
Insecta	Coleoptera	Carabidae	<i>Pterostichus</i>	<i>Pterostichus punctiventris</i>
Insecta	Coleoptera	Carabidae	<i>Rhadine</i>	<i>Rhadine ozarkensis</i>
Insecta	Coleoptera	Carabidae	<i>Scaphinotus</i>	<i>Scaphinotus unicolor</i>
Insecta	Coleoptera	Carabidae	<i>Scaphinotus</i>	<i>Scaphinotus fissicollis</i>
Insecta	Coleoptera	Carabidae	<i>Scaphinotus</i>	<i>Scaphinotus infletus</i>
Insecta	Coleoptera	Carabidae	<i>Selenophorus</i>	<i>Selenophorus ellipticus</i>
Insecta	Coleoptera	Carabidae	<i>Selenophorus</i>	<i>Selenophorus gagatinus</i>
Insecta	Coleoptera	Carabidae	<i>Selenophorus</i>	<i>Selenophorus opalinus</i>
Insecta	Coleoptera	Carabidae	<i>Stenolophus</i>	<i>Stenolophus ochropezus</i>
Insecta	Coleoptera	Carabidae	<i>Synuchus</i>	<i>Synuchus impunctatus</i> [§]
Insecta	Coleoptera	Carabidae	<i>Tachyta</i>	<i>Tachyta parvicornis</i>
Insecta	Coleoptera	Carabidae	<i>Tachys</i>	<i>Tachys columbiensis</i>
Insecta	Coleoptera	Carabidae	<i>Tachys</i>	<i>Tachys oblitus</i>
Insecta	Coleoptera	Carabidae	<i>Trichotichnus</i>	<i>Trichotichnus autumnalis</i>
Insecta	Coleoptera	Carabidae	<i>Trichotichnus</i>	<i>Trichotichnus fulgens</i>
Insecta	Coleoptera	Carabidae	<i>Trichotichnus</i>	<i>Trichotichnus vulpeculus</i>
Insecta	Coleoptera	Cerambycidae	<i>Aegormorphus</i>	<i>Aegormorphus modestus</i>
Insecta	Coleoptera	Cerambycidae	<i>Aegormorphus</i>	<i>Aegormorphus quadrigibbus</i>
Insecta	Coleoptera	Cerambycidae	<i>Anelaphus</i>	<i>Anelaphus parallelus</i>
Insecta	Coleoptera	Cerambycidae	<i>Anelaphus</i>	<i>Anelaphus pumilus</i>
Insecta	Coleoptera	Cerambycidae	<i>Astyleiopus</i>	<i>Astyleiopus variegatus</i>
Insecta	Coleoptera	Cerambycidae	<i>Astylidius</i>	<i>Astylidius parvus</i>
Insecta	Coleoptera	Cerambycidae	<i>Astylopsis</i>	<i>Astylopsis macula</i>
Insecta	Coleoptera	Cerambycidae	<i>Astylopsis</i>	<i>Astylopsis sexguttata</i>
Insecta	Coleoptera	Cerambycidae	<i>Bellamira</i>	<i>Bellamira scalaris</i>
Insecta	Coleoptera	Cerambycidae	<i>Brachyleptura</i>	<i>Brachyleptura champlaini</i>
Insecta	Coleoptera	Cerambycidae	<i>Callimoxys</i>	<i>Callimoxys sanguinicollis</i>
Insecta	Coleoptera	Cerambycidae	<i>Centrodera</i>	<i>Centrodera sublineata</i>

Table A3 (cont.). Species identified from Steel Creek during this dissertation.

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Class	Order	Family	Genus	Species
Insecta	Coleoptera	Cerambycidae	<i>Clytoleptus</i>	<i>Clytoleptus albofasciatus</i>
Insecta	Coleoptera	Cerambycidae	<i>Cyrtinus</i>	<i>Cyrtinus pygmaeus</i>
Insecta	Coleoptera	Cerambycidae	<i>Cyrtophorus</i>	<i>Cyrtophorus verrucosus</i>
Insecta	Coleoptera	Cerambycidae	<i>Dorcaschema</i>	<i>Dorcaschema alternatum</i>
Insecta	Coleoptera	Cerambycidae	<i>Dorcaschema</i>	<i>Dorcaschema cinereum</i>
Insecta	Coleoptera	Cerambycidae	<i>Dorcaschema</i>	<i>Dorcaschema nigrum</i>
Insecta	Coleoptera	Cerambycidae	<i>Dorcaschema</i>	<i>Dorcaschema wildii</i>
Insecta	Coleoptera	Cerambycidae	<i>Eburia</i>	<i>Eburia quadrigeminata</i>
Insecta	Coleoptera	Cerambycidae	<i>Ecyrus</i>	<i>Ecyrus dasycerus</i>
Insecta	Coleoptera	Cerambycidae	<i>Elytrimitatrix</i>	<i>Elytrimitatrix undata</i>
Insecta	Coleoptera	Cerambycidae	<i>Elaphidion</i>	<i>Elaphidion mucronatum</i>
Insecta	Coleoptera	Cerambycidae	<i>Enaphalodes</i>	<i>Enaphalodes rufulus</i>
Insecta	Coleoptera	Cerambycidae	<i>Euderces</i>	<i>Euderces reichei</i>
Insecta	Coleoptera	Cerambycidae	<i>Euderces</i>	<i>Euderces picipes</i>
Insecta	Coleoptera	Cerambycidae	<i>Euderces</i>	<i>Euderces pini</i>
Insecta	Coleoptera	Cerambycidae	<i>Eupogonius</i>	<i>Eupogonius pauper</i>
Insecta	Coleoptera	Cerambycidae	<i>Gaurotes</i>	<i>Gaurotes cyanipennis</i>
Insecta	Coleoptera	Cerambycidae	<i>Graphisurus</i>	<i>Graphisurus despectus</i>
Insecta	Coleoptera	Cerambycidae	<i>Graphisurus</i>	<i>Graphisurus fasciatus</i>
Insecta	Coleoptera	Cerambycidae	<i>Heterachthes</i>	<i>Heterachthes</i> <i>quadrimaculatus</i>
Insecta	Coleoptera	Cerambycidae	<i>Hyperplatys</i>	<i>Hyperplatys maculata</i>
Insecta	Coleoptera	Cerambycidae	<i>Knulliana</i>	<i>Knulliana cincta</i>
Insecta	Coleoptera	Cerambycidae	<i>Leptostylus</i>	<i>Leptostylus transversus</i>
Insecta	Coleoptera	Cerambycidae	<i>Leptura</i>	<i>Leptura emarginata</i>
Insecta	Coleoptera	Cerambycidae	<i>Lepturges</i>	<i>Lepturges angulatus</i>
Insecta	Coleoptera	Cerambycidae	<i>Lepturges</i>	<i>Lepturges confluens</i>
Insecta	Coleoptera	Cerambycidae	<i>Micranoplium</i>	<i>Micranoplium unicolor</i>
Insecta	Coleoptera	Cerambycidae	<i>Molorchus</i>	<i>Molorchus bimaculatus</i>
Insecta	Coleoptera	Cerambycidae	<i>Monochamus</i>	<i>Monochamus titillator</i>
Insecta	Coleoptera	Cerambycidae	<i>Neoclytus</i>	<i>Neoclytus acuminatus</i>
Insecta	Coleoptera	Cerambycidae	<i>Neoclytus</i>	<i>Neoclytus caprea</i>
Insecta	Coleoptera	Cerambycidae	<i>Neoclytus</i>	<i>Neoclytus horridus</i>
Insecta	Coleoptera	Cerambycidae	<i>Neoclytus</i>	<i>Neoclytus jouteli</i>
Insecta	Coleoptera	Cerambycidae	<i>Neoclytus</i>	<i>Neoclytus mucronatus</i>
Insecta	Coleoptera	Cerambycidae	<i>Neoclytus</i>	<i>Neoclytus scutellaris</i>
Insecta	Coleoptera	Cerambycidae	<i>Necydalis</i>	<i>Necydalis mellita</i>

Table A3 (cont.). Species identified from Steel Creek during this dissertation.

† - putative new species, ‡ - introduced, non-native species, § - new state record

Class	Order	Family	Genus	Species
Insecta	Coleoptera	Cerambycidae	<i>Oberea</i>	<i>Oberea ulmicola</i>
Insecta	Coleoptera	Cerambycidae	<i>Obrium</i>	<i>Obrium maculatum</i>
Insecta	Coleoptera	Cerambycidae	<i>Onicideres</i>	<i>Onicideres cingulata</i>
Insecta	Coleoptera	Cerambycidae	<i>Orthosoma</i>	<i>Orthosoma brunneum</i>
Insecta	Coleoptera	Cerambycidae	<i>Parelaphidion</i>	<i>Parelaphidion aspersum</i>
Insecta	Coleoptera	Cerambycidae	<i>Phymatodes</i>	<i>Phymatodes amoenus</i>
Insecta	Coleoptera	Cerambycidae	<i>Phymatodes</i>	<i>Phymatodes testaceus</i>
Insecta	Coleoptera	Cerambycidae	<i>Phymatodes</i>	<i>Phymatodes varius</i>
Insecta	Coleoptera	Cerambycidae	<i>Physocnemum</i>	<i>Physocnemum brevilineum</i>
Insecta	Coleoptera	Cerambycidae	<i>Prionus</i>	<i>Prionus imbricornis</i>
Insecta	Coleoptera	Cerambycidae	<i>Purpuricen</i>	<i>Purpuricen humeralis</i>
Insecta	Coleoptera	Cerambycidae	<i>Purpuricen</i>	<i>Purpuricen paraxillaris</i>
Insecta	Coleoptera	Cerambycidae	<i>Saperda</i>	<i>Saperda discoidea</i>
Insecta	Coleoptera	Cerambycidae	<i>Saperda</i>	<i>Saperda imitans</i>
Insecta	Coleoptera	Cerambycidae	<i>Saperda</i>	<i>Saperda lateralis</i>
Insecta	Coleoptera	Cerambycidae	<i>Saperda</i>	<i>Saperda tridentata</i>
Insecta	Coleoptera	Cerambycidae	<i>Sarosesthes</i>	<i>Sarosesthes fulminans</i>
Insecta	Coleoptera	Cerambycidae	<i>Stenocorus</i>	<i>Stenocorus cinnamopterus</i>
Insecta	Coleoptera	Cerambycidae	<i>Stenosphenus</i>	<i>Stenosphenus notatus</i>
Insecta	Coleoptera	Cerambycidae	<i>Sternidius</i>	<i>Sternidius alpha</i>
Insecta	Coleoptera	Cerambycidae	<i>Strangalepta</i>	<i>Strangalepta abbreviata</i>
Insecta	Coleoptera	Cerambycidae	<i>Strangalia</i>	<i>Strangalia bicolor</i>
Insecta	Coleoptera	Cerambycidae	<i>Strangalia</i>	<i>Strangalia luteicornis</i>
Insecta	Coleoptera	Cerambycidae	<i>Strophiona</i>	<i>Strophiona nitens</i>
Insecta	Coleoptera	Cerambycidae	<i>Tilloclytus</i>	<i>Tilloclytus geminatus</i>
Insecta	Coleoptera	Cerambycidae	<i>Trachysida</i>	<i>Trachysida mutabilis</i>
Insecta	Coleoptera	Cerambycidae	<i>Trigonarthris</i>	<i>Trigonarthris minnesotana</i>
Insecta	Coleoptera	Cerambycidae	<i>Trigonarthris</i>	<i>Trigonarthris proxima</i>
Insecta	Coleoptera	Cerambycidae	<i>Typocerus</i>	<i>Typocerus lugubris</i>
Insecta	Coleoptera	Cerambycidae	<i>Typocerus</i>	<i>Typocerus velutinus</i>
Insecta	Coleoptera	Cerambycidae	<i>Typocerus</i>	<i>Typocerus zebra</i>
Insecta	Coleoptera	Cerambycidae	<i>Urgleptes</i>	<i>Urgleptes querci</i>
Insecta	Coleoptera	Cerambycidae	<i>Urgleptes</i>	<i>Urgleptes signatus</i>
Insecta	Coleoptera	Cerambycidae	<i>Xylotrechus</i>	<i>Xylotrechus colonus</i>
Insecta	Coleoptera	Cleridae	<i>Enoclerus</i>	<i>Enoclerus ichneumoneus</i>
Insecta	Coleoptera	Cleridae	<i>Enoclerus</i>	<i>Enoclerus nigripes</i>
Insecta	Coleoptera	Coccinellidae	<i>Coccinella</i>	<i>Coccinella septempunctata</i>

Table A3 (cont.). Species identified from Steel Creek during this dissertation.

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Class	Order	Family	Genus	Species
Insecta	Coleoptera	Curculionidae	<i>Acalles</i>	<i>Acalles carinatus</i>
Insecta	Coleoptera	Curculionidae	<i>Acalles</i>	<i>Acalles clavatus</i> [§]
Insecta	Coleoptera	Curculionidae	<i>Acalles</i>	<i>Acalles minutissimus</i> [§]
Insecta	Coleoptera	Curculionidae	<i>Acoptus</i>	<i>Acoptus suturalis</i> [§]
Insecta	Coleoptera	Curculionidae	<i>Anthonomus</i>	<i>Anthonomus juniperinus</i> [§]
Insecta	Coleoptera	Curculionidae	<i>Anthonomus</i>	<i>Anthonomus nigrinus</i>
Insecta	Coleoptera	Curculionidae	<i>Anthonomus</i>	<i>Anthonomus rufipennis</i>
Insecta	Coleoptera	Curculionidae	<i>Anthonomus</i>	<i>Anthonomus suturalis</i>
Insecta	Coleoptera	Curculionidae	<i>Aphanommata</i>	<i>Aphanommata tenuis</i>
Insecta	Coleoptera	Curculionidae	<i>Apteromechus</i>	<i>Apteromechus ferratus</i>
Insecta	Coleoptera	Curculionidae	<i>Anametis</i>	<i>Anametis granulata</i> [§]
Insecta	Coleoptera	Curculionidae	<i>Auleutes</i>	<i>Auleutes nebulosus complex</i>
Insecta	Coleoptera	Curculionidae	<i>Buchananius</i>	<i>Buchananius sulcatus</i>
Insecta	Coleoptera	Curculionidae	<i>Canistes</i>	<i>Canistes schusteri</i>
Insecta	Coleoptera	Curculionidae	<i>Caulophilus</i>	<i>Caulophilus dubius</i>
Insecta	Coleoptera	Curculionidae	<i>Cercopeus</i>	<i>Cercopeus chrysorrhoeus</i>
Insecta	Coleoptera	Curculionidae	<i>Chalcodermus</i>	<i>Chalcodermus inaequicollis</i>
Insecta	Coleoptera	Curculionidae	<i>Conotrachelus</i>	<i>Conotrachelus affinis</i>
Insecta	Coleoptera	Curculionidae	<i>Conotrachelus</i>	<i>Conotrachelus anaglypticus</i>
Insecta	Coleoptera	Curculionidae	<i>Conotrachelus</i>	<i>Conotrachelus aratus</i>
Insecta	Coleoptera	Curculionidae	<i>Conotrachelus</i>	<i>Conotrachelus carinifer</i>
Insecta	Coleoptera	Curculionidae	<i>Conotrachelus</i>	<i>Conotrachelus elegans</i>
Insecta	Coleoptera	Curculionidae	<i>Conotrachelus</i>	<i>Conotrachelus naso</i>
Insecta	Coleoptera	Curculionidae	<i>Conotrachelus</i>	<i>Conotrachelus posticatus</i>
Insecta	Coleoptera	Curculionidae	<i>Cophes</i>	<i>Cophes fallax</i>
Insecta	Coleoptera	Curculionidae	<i>Cophes</i>	<i>Cophes obtentus</i>
Insecta	Coleoptera	Curculionidae	<i>Cossonus</i>	<i>Cossonus impressifrons</i>
Insecta	Coleoptera	Curculionidae	<i>Craponius</i>	<i>Craponius inaequalis</i>
Insecta	Coleoptera	Curculionidae	<i>Cryptorhynchus</i>	<i>Cryptorhynchus fuscatus</i>
Insecta	Coleoptera	Curculionidae	<i>Cryptorhynchus</i>	<i>Cryptorhynchus tristis</i>
Insecta	Coleoptera	Curculionidae	<i>Curculio</i>	<i>Curculio othorhynchus</i>
Insecta	Coleoptera	Curculionidae	<i>Cyrtepidomus</i>	<i>Cyrtepidomus castaneus</i> [‡]
Insecta	Coleoptera	Curculionidae	<i>Dichoxenus</i>	<i>Dichoxenus setiger</i>
Insecta	Coleoptera	Curculionidae	<i>Dietzella</i>	<i>Dietzella zimmermanni</i>
Insecta	Coleoptera	Curculionidae	<i>Dryophthorus</i>	<i>Dryophthorus americanus</i>
Insecta	Coleoptera	Curculionidae	<i>Epacalles</i>	<i>Epacalles inflatus</i>
Insecta	Coleoptera	Curculionidae	<i>Eubulus</i>	<i>Eubulus bisignatus</i>
Insecta	Coleoptera	Curculionidae	<i>Eubulus</i>	<i>Eubulus obliquefasciatus</i>

Table A3 (cont.). Species identified from Steel Creek during this dissertation.

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Class	Order	Family	Genus	Species
Insecta	Coleoptera	Curculionidae	<i>Eudociminus</i>	<i>Eudociminus mannerheimii</i> [§]
Insecta	Coleoptera	Curculionidae	<i>Eurhoptus</i>	<i>Eurhoptus</i> sp. 1 [†]
Insecta	Coleoptera	Curculionidae	<i>Eurhoptus</i>	<i>Eurhoptus pyriformis</i>
Insecta	Coleoptera	Curculionidae	<i>Geraeus</i>	<i>Geraeus penicillus</i>
Insecta	Coleoptera	Curculionidae	<i>Hypera</i>	<i>Hypera compta</i> [‡]
Insecta	Coleoptera	Curculionidae	<i>Hypera</i>	<i>Hypera meles</i> [‡]
Insecta	Coleoptera	Curculionidae	<i>Hypera</i>	<i>Hypera postica</i>
Insecta	Coleoptera	Curculionidae	<i>Idiostethus</i>	<i>Idiostethus subcalvus</i> [§]
Insecta	Coleoptera	Curculionidae	<i>Laemosaccus</i>	<i>Laemosaccus nephele</i> group
Insecta	Coleoptera	Curculionidae	<i>Leichrops</i>	<i>Lechriops oculatus</i>
Insecta	Coleoptera	Curculionidae	<i>Lymantes</i>	<i>Lymantes sandersoni</i>
Insecta	Coleoptera	Curculionidae	<i>Madarellus</i>	<i>Madarellus undulatus</i> [§]
Insecta	Coleoptera	Curculionidae	<i>Magdalis</i>	<i>Magdalis armicollis</i> [§]
Insecta	Coleoptera	Curculionidae	<i>Magdalis</i>	<i>Magdalis barbata</i> [§]
Insecta	Coleoptera	Curculionidae	<i>Mecinus</i>	<i>Mecinus pascuorum</i> [§]
Insecta	Coleoptera	Curculionidae	<i>Myrmex</i>	<i>Myrmex chevrolatii</i> [§]
Insecta	Coleoptera	Curculionidae	<i>Myrmex</i>	<i>Myrmex myrmex</i> [§]
Insecta	Coleoptera	Curculionidae	<i>Nicentrus</i>	<i>Nicentrus lecontei</i> [§]
Insecta	Coleoptera	Curculionidae	<i>Oopterinus</i>	<i>Oopterinus perforatus</i>
Insecta	Coleoptera	Curculionidae	<i>Otiorhynchus</i>	<i>Otiorhynchus rugosostriatus</i> ^{‡§}
Insecta	Coleoptera	Curculionidae	<i>Pandeleiteius</i>	<i>Pandeleiteius hilaris</i>
Insecta	Coleoptera	Curculionidae	<i>Piazorhinus</i>	<i>Piazorhinus pictus</i> [§]
Insecta	Coleoptera	Curculionidae	<i>Phyllotrox</i>	<i>Phyllotrox ferrugineus</i> [§]
Insecta	Coleoptera	Curculionidae	<i>Plocamus</i>	<i>Plocamus hispidulus</i> [§]
Insecta	Coleoptera	Curculionidae	<i>Pseudobaris</i>	<i>Pseudobaris nigrina</i> [§]
Insecta	Coleoptera	Curculionidae	<i>Pseudopentarthrum</i>	<i>Pseudopentarthrum simplex</i> [§]
Insecta	Coleoptera	Curculionidae	<i>Rhinoncus</i>	<i>Rhinoncus pericarpus</i> [§]
Insecta	Coleoptera	Curculionidae	<i>Sitona</i>	<i>Sitona lineatus</i> ^{‡§}
Insecta	Coleoptera	Curculionidae	<i>Stenoscelis</i>	<i>Stenoscelis brevis</i> [§]
Insecta	Coleoptera	Curculionidae	<i>Tachyerges</i>	<i>Tachyerges niger</i>
Insecta	Coleoptera	Curculionidae	<i>Tomolips</i>	<i>Tomolips quercicola</i> [§]
Insecta	Coleoptera	Curculionidae	<i>Tychius</i>	<i>Tychius picirostris</i>
Insecta	Coleoptera	Curculionidae	<i>Tyloderma</i>	<i>Tyloderma foveolatum</i>
Insecta	Coleoptera	Elateridae	<i>Alaus</i>	<i>Alaus oculatus</i>
Insecta	Coleoptera	Endomychidae	<i>Phymaphora</i>	<i>Phymaphora pulchella</i>

Table A3 (cont.). Species identified from Steel Creek during this dissertation.

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Class	Order	Family	Genus	Species
Insecta	Coleoptera	Meloidae	<i>Lytta</i>	<i>Lytta aenea</i>
Insecta	Coleoptera	Nitidulidae	<i>Glischrochilus</i>	<i>Glischrochilus fasciatus</i>
Insecta	Coleoptera	Salpingidae	<i>Salpingus</i>	<i>Salpingus viridiaeneus</i>
Insecta	Coleoptera	Scarabaeidae	<i>Copris</i>	<i>Copris fricator</i>
Insecta	Coleoptera	Scarabaeidae	<i>Cotinus</i>	<i>Cotinus nitida</i>
Insecta	Coleoptera	Scarabaeidae	<i>Onthophagus</i>	<i>Onthophagus orpheus</i>
Insecta	Coleoptera	Silphidae	<i>Necrophila</i>	<i>Necrophila americana</i>
Insecta	Coleoptera	Tenebrionidae	<i>Polyporus</i>	<i>Polyporus perforatus</i>
Insecta	Dermaptera	Forficulidae	<i>Forficula</i>	<i>Forficula auricularia</i>
Insecta	Diptera	Anisopodidae	<i>Sylvicola</i>	<i>Sylvicola fenestralis</i>
Insecta	Diptera	Bombyliidae	<i>Bombylius</i>	<i>Bombylius (bald-backed)[†]</i>
Insecta	Diptera	Drosophilidae	<i>Drosophila</i>	<i>Drosophila suzukii[‡]</i>
Insecta	Diptera	Drosophilidae	<i>Zaprionus</i>	<i>Zaprionus indianus[‡]</i>
Insecta	Diptera	Limoniidae	<i>Cladura</i>	<i>Cladura flavoferruginea</i>
Insecta	Diptera	Lygistorrhinidae	<i>Lygistorrhina</i>	<i>Lygistorrhina sancthecatharinae[§]</i>
Insecta	Diptera	Mydidae	<i>Mydas</i>	<i>Mydas clavatus</i>
Insecta	Diptera	Osetridae	<i>Cephenemyia</i>	<i>Cephenemyia sp. nov.[†]</i>
Insecta	Diptera	Osetridae	<i>Cuterebra</i>	<i>Cuterebra emasculator</i>
Insecta	Diptera	Osetridae	<i>Cuterebra</i>	<i>Cuterebra f. fontinella</i>
Insecta	Diptera	Ptychopteridae	<i>Bittacomorpha</i>	<i>Bittacomorpha clavipes</i>
Insecta	Diptera	Scathophagidae	<i>Scathophaga</i>	<i>Scathophaga furcata</i>
Insecta	Diptera	Scathophagidae	<i>Scathophaga</i>	<i>Scathophaga stercoraria</i>
Insecta	Diptera	Stratiomyidae	<i>Cephalochrysa</i>	<i>Cephalochrysa nigricornis[§]</i>
Insecta	Diptera	Stratiomyidae	<i>Gowdeyana</i>	<i>Gowdeyana punctifera[§]</i>
Insecta	Diptera	Stratiomyidae	<i>Ptecticus</i>	<i>Ptecticus trivattus</i>
Insecta	Diptera	Stratiomyidae	<i>Sargus</i>	<i>Sargus decorus[§]</i>
Insecta	Diptera	Tipulidae	<i>Ctenophora</i>	<i>Ctenophora dorsalis</i>
Insecta	Diptera	Ulidiidae	<i>Callopistromyia</i>	<i>Callopistromyia annulipes</i>
Insecta	Diptera	Ulidiidae	<i>Idana</i>	<i>Idana marginata</i>
Insecta	Diptera	Xylophagidae	<i>Rachicercus</i>	<i>Rachicercus obscuripennis[§]</i>
Insecta	Dermaptera	Anisolabididae	<i>Euborellia</i>	<i>Euborellia annulipes[‡]</i>

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Class	Order	Family	Genus	Species
Insecta	Hemiptera	Alydidae	<i>Alydus</i>	<i>Alydus eurinus</i>
Insecta	Hemiptera	Alydidae	<i>Megalotomus</i>	<i>Megalotomus quinquespinosus</i>
Insecta	Hemiptera	Aradidae	<i>Aradus</i>	<i>Aradus acutus</i>
Insecta	Hemiptera	Aradidae	<i>Aradus</i>	<i>Aradus approximatus</i> [§]
Insecta	Hemiptera	Aradidae	<i>Aradus</i>	<i>Aradus crenatus</i>
Insecta	Hemiptera	Aradidae	<i>Aradus</i>	<i>Aradus duzei</i> [§]
Insecta	Hemiptera	Aradidae	<i>Aradus</i>	<i>Aradus ornatus</i> [§]
Insecta	Hemiptera	Aradidae	<i>Aradus</i>	<i>Aradus similis</i>
Insecta	Hemiptera	Aradidae	<i>Mezira</i>	<i>Mezira sayi</i>
Insecta	Hemiptera	Aradidae	<i>Neuroctenus</i>	<i>Neuroctenus elongatus</i> [§]
Insecta	Hemiptera	Aradidae	<i>Neuroctenus</i>	<i>Neuroctenus pseudonymus</i> [§]
Insecta	Hemiptera	Aradidae	<i>Notapictinus</i>	<i>Notapictinus aurivilli</i> [§]
Insecta	Hemiptera	Caliscelidae	<i>Bruchomorpha</i>	<i>Bruchomorpha oculata</i>
Insecta	Hemiptera	Coreidae	<i>Acanthocephala</i>	<i>Acanthocephala terminalis</i>
Insecta	Hemiptera	Coreidae	<i>Leptoglossus</i>	<i>Leptoglossus oppositus</i>
Insecta	Hemiptera	Cydnidae	<i>Amnestus</i>	<i>Amnestus basidentatus</i>
Insecta	Hemiptera	Cydnidae	<i>Melanaethus</i>	<i>Melanaethus subpunctatus</i>
Insecta	Hemiptera	Cydnidae	<i>Pangaeus</i>	<i>Pangaeus bilineatus</i>
Insecta	Hemiptera	Cydnidae	<i>Sehirus</i>	<i>Sehirus cinctus</i> [§]
Insecta	Hemiptera	Gerridae	<i>Gerris</i>	<i>Gerris argenticollis</i>
Insecta	Hemiptera	Gerridae	<i>Gerris</i>	<i>Gerris marginatus</i>
Insecta	Hemiptera	Lygaeidae	<i>Nysius</i>	<i>Nysius raphanus</i> [§]
Insecta	Hemiptera	Miridae	<i>Prepops</i>	<i>Prepops insitivus</i> [§]
Insecta	Hemiptera	Pachygronthidae	<i>Oedancala</i>	<i>Oedancala dorsalis</i>
Insecta	Hemiptera	Pachygronthidae	<i>Phlegyas</i>	<i>Phlegyas abbreviatus</i>
Insecta	Hemiptera	Pentatomidae	<i>Banasa</i>	<i>Banasa euchlora</i>
Insecta	Hemiptera	Pentatomidae	<i>Brochymena</i>	<i>Brochymena arborea</i>
Insecta	Hemiptera	Pentatomidae	<i>Chinavia</i>	<i>Chinavia hilaris</i>
Insecta	Hemiptera	Pentatomidae	<i>Coenus</i>	<i>Coenus delius</i>
Insecta	Hemiptera	Pentatomidae	<i>Euschistus</i>	<i>Euschistus servus</i>
Insecta	Hemiptera	Pentatomidae	<i>Euschistus</i>	<i>Euschistus tristigmus</i>
Insecta	Hemiptera	Pentatomidae	<i>Meneclis</i>	<i>Meneclis insertus</i>
Insecta	Hemiptera	Pentatomidae	<i>Mormidea</i>	<i>Mormidea lugens</i>
Insecta	Hemiptera	Pentatomidae	<i>Podisus</i>	<i>Podisus maculiventris</i>

Table A3 (cont.). Species identified from Steel Creek during this dissertation.

† - putative new species, ‡ - introduced, non-native species, § - new state record

Class	Order	Family	Genus	Species
Insecta	Hemiptera	Reduviidae	<i>Arilus</i>	<i>Arilus cristatus</i>
Insecta	Hemiptera	Reduviidae	<i>Barce</i>	
Insecta	Hemiptera	Reduviidae	<i>Melanolestes</i>	<i>Melanolestes picipes</i>
Insecta	Hemiptera	Reduviidae	<i>Oncocephalus</i>	<i>Oncocephalus geniculatus</i>
Insecta	Hemiptera	Reduviidae	<i>Pselliopus</i>	<i>Pselliopus barberi</i>
Insecta	Hemiptera	Reduviidae	<i>Rhiginia</i>	<i>Rhiginia cruciata</i>
Insecta	Hemiptera	Reduviidae	<i>Rocconota</i>	<i>Rocconota annulicornis</i>
Insecta	Hemiptera	Reduviidae	<i>Sinea</i>	<i>Sinea diadema</i>
Insecta	Hemiptera	Reduviidae	<i>Sinea</i>	<i>Sinea spinipes</i>
Insecta	Hemiptera	Reduviidae	<i>Stenopoda</i>	<i>Stenopoda spinulosa</i>
Insecta	Hemiptera	Reduviidae	<i>Zelus</i>	<i>Zelus tetracanthus</i> [§]
Insecta	Hemiptera	Rhyparochromidae	<i>Antilocoris</i>	<i>Antilocoris pilosulus</i>
Insecta	Hemiptera	Rhyparochromidae	<i>Cryphula</i>	<i>Cryphula trimaculata</i>
Insecta	Hemiptera	Rhyparochromidae	<i>Kolenetrus</i>	<i>Kolenetrus plenus</i> [§]
Insecta	Hemiptera	Rhyparochromidae	<i>Myodocha</i>	<i>Myodocha serripes</i>
Insecta	Hemiptera	Rhyparochromidae	<i>Ozophora</i>	<i>Ozophora picturata</i>
Insecta	Hemiptera	Rhyparochromidae	<i>Xestocoris</i>	<i>Xestocoris nitens</i>
Insecta	Hemiptera	Scutelleridae	<i>Stethaulax</i>	<i>Stethaulax marmorata</i>
Insecta	Hemiptera	Thyreocoridae	<i>Corimelaena</i>	<i>Corimelaena pulicaria</i>
Insecta	Hemiptera	Thyreocoridae	<i>Galgupha</i>	<i>Galgupha loboprostethia</i>
Insecta	Hemiptera	Tingidae	<i>Acalypta</i>	<i>Acalypta susana</i>
Insecta	Hymenoptera	Apidae	<i>Apis</i>	<i>Apis mellifera</i>
Insecta	Hymenoptera	Argidae	<i>Arge</i>	<i>Arge humeralis</i>
Insecta	Hymenoptera	Argidae	<i>Arge</i>	<i>Arge macleayi</i>
Insecta	Hymenoptera	Argidae	<i>Sterictiphota</i>	<i>Sterictiphota serotina</i>
Insecta	Hymenoptera	Aulacidae	<i>Pristaulacus</i>	<i>Pristaulacus rufitarsis</i> [§]
Insecta	Hymenoptera	Chrysididae	<i>Amisega</i>	<i>Amisega bella</i> [§]
Insecta	Hymenoptera	Chrysididae	<i>Amisega</i>	<i>Amisega kahlii</i> [§]
Insecta	Hymenoptera	Chrysididae	<i>Trichrysis</i>	<i>Trichrysis areolata</i> [§]
Insecta	Hymenoptera	Cimbididae	<i>Abia</i>	<i>Abia americana</i>
Insecta	Hymenoptera	Diprionidae	<i>Monoctenus</i>	<i>Monoctenus fulvus</i>
Insecta	Hymenoptera	Embolemidae	<i>Embolemus</i>	<i>Embolemus nearcticus</i>

Table A3 (cont.). Species identified from Steel Creek during this dissertation.

† - putative new species, ‡ - introduced, non-native species, § - new state record

Class	Order	Family	Genus	Species
Insecta	Hymenoptera	Encyrtidae	<i>Chrysoplatycerus</i>	<i>Chrysoplatycerus ferrisi</i>
Insecta	Hymenoptera	Encyrtidae	<i>Chrysoplatycerus</i>	<i>Chrysoplatycerus spendens</i>
Insecta	Hymenoptera	Encyrtidae	<i>Forcipestricis</i>	<i>Forcipestricis gaseaui</i>
Insecta	Hymenoptera	Encyrtidae	<i>Metaphycus</i>	<i>Metaphycus</i> , nr <i>matteolus</i> [†]
Insecta	Hymenoptera	Encyrtidae	<i>Ooencyrtus</i>	<i>Ooencyrtus anasae</i>
Insecta	Hymenoptera	Encyrtidae	<i>Ooencyrtus</i>	<i>Ooencyrtus sp nov</i> [†]
Insecta	Hymenoptera	Encyrtidae	<i>Syrphophagus</i>	<i>Syrphophagus aphidivorus</i>
Insecta	Hymenoptera	Formicidae	<i>Aphaenogaster</i>	<i>Aphaenogaster carolinensis</i>
Insecta	Hymenoptera	Formicidae	<i>Aphaenogaster</i>	<i>Aphaenogaster fulva</i>
Insecta	Hymenoptera	Formicidae	<i>Aphaenogaster</i>	<i>Aphaenogaster lamellidens</i>
Insecta	Hymenoptera	Formicidae	<i>Aphaenogaster</i>	<i>Aphaenogaster mariae</i>
Insecta	Hymenoptera	Formicidae	<i>Aphaenogaster</i>	<i>Aphaenogaster tennesseensis</i>
Insecta	Hymenoptera	Formicidae	<i>Aphaenogaster</i>	<i>Aphaenogaster treatae</i>
Insecta	Hymenoptera	Formicidae	<i>Brachymyrmex</i>	<i>Brachymyrmex sp. 04</i> [†]
Insecta	Hymenoptera	Formicidae	<i>Brachymyrmex</i>	<i>Brachymyrmex depilis</i>
Insecta	Hymenoptera	Formicidae	<i>Camponotus</i>	<i>Camponotus americanus</i>
Insecta	Hymenoptera	Formicidae	<i>Camponotus</i>	<i>Camponotus caryae</i>
Insecta	Hymenoptera	Formicidae	<i>Camponotus</i>	<i>Camponotus castaneus</i>
Insecta	Hymenoptera	Formicidae	<i>Camponotus</i>	<i>Camponotus chromaiodes</i>
Insecta	Hymenoptera	Formicidae	<i>Camponotus</i>	<i>Camponotus decipiens</i>
Insecta	Hymenoptera	Formicidae	<i>Camponotus</i>	<i>Camponotus impressus</i>
Insecta	Hymenoptera	Formicidae	<i>Camponotus</i>	<i>Camponotus mississippiensis</i>
Insecta	Hymenoptera	Formicidae	<i>Camponotus</i>	<i>Camponotus nearcticus</i>
Insecta	Hymenoptera	Formicidae	<i>Camponotus</i>	<i>Camponotus pennsylvanicus</i>
Insecta	Hymenoptera	Formicidae	<i>Camponotus</i>	<i>Camponotus subbarbatus</i>
Insecta	Hymenoptera	Formicidae	<i>Camponotus</i>	<i>Camponotus snellingi</i>
Insecta	Hymenoptera	Formicidae	<i>Crematogaster</i>	<i>Crematogaster ashmeadi</i>
Insecta	Hymenoptera	Formicidae	<i>Crematogaster</i>	<i>Crematogaster cerasi</i>
Insecta	Hymenoptera	Formicidae	<i>Crematogaster</i>	<i>Crematogaster lineolata</i>
Insecta	Hymenoptera	Formicidae	<i>Crematogaster</i>	<i>Crematogaster minutissima</i>
Insecta	Hymenoptera	Formicidae	<i>Crematogaster</i>	<i>Crematogaster pilosa</i>
Insecta	Hymenoptera	Formicidae	<i>Crematogaster</i>	<i>Crematogaster vermiculata</i>
Insecta	Hymenoptera	Formicidae	<i>Discothyrea</i>	<i>Discothyrea testacea</i>
Insecta	Hymenoptera	Formicidae	<i>Formica</i>	<i>Formica pallidefulva</i>
Insecta	Hymenoptera	Formicidae	<i>Formica</i>	<i>Formica subsericea</i>

Table A3 (cont.). Species identified from Steel Creek during this dissertation.

[†] - putative new species, [‡] - introduced, non-native species, [§] - new state record

Class	Order	Family	Genus	Species
Insecta	Hymenoptera	Formicidae	<i>Hypoponera</i>	<i>Hypoponera opaciceps</i>
Insecta	Hymenoptera	Formicidae	<i>Hypoponera</i>	<i>Hypoponera opacior</i>
Insecta	Hymenoptera	Formicidae	<i>Lasius</i>	<i>Lasius alienus</i>
Insecta	Hymenoptera	Formicidae	<i>Lasius</i>	<i>Lasius interjectus</i>
Insecta	Hymenoptera	Formicidae	<i>Monomorium</i>	<i>Monomorium minimum</i>
Insecta	Hymenoptera	Formicidae	<i>Myrmica</i>	<i>Myrmica pinetorum</i>
Insecta	Hymenoptera	Formicidae	<i>Myrmecina</i>	<i>Myrmecina americana</i>
Insecta	Hymenoptera	Formicidae	<i>Neivamyrmex</i>	<i>Neivamyrmex opacithorax</i>
Insecta	Hymenoptera	Formicidae	<i>Nylanderia</i>	<i>Nylanderia fasionensis</i>
Insecta	Hymenoptera	Formicidae	<i>Nylanderia</i>	<i>Nylanderia parvula</i>
Insecta	Hymenoptera	Formicidae	<i>Nylanderia</i>	<i>Nylanderia terricola</i>
Insecta	Hymenoptera	Formicidae	<i>Nylanderia</i>	<i>Nylanderia trageri</i>
Insecta	Hymenoptera	Formicidae	<i>Pheidole</i>	<i>Pheidole tetra</i>
Insecta	Hymenoptera	Formicidae	<i>Ponera</i>	<i>Ponera exotica</i>
Insecta	Hymenoptera	Formicidae	<i>Ponera</i>	<i>Ponera pennsylvanica</i>
Insecta	Hymenoptera	Formicidae	<i>Proceratium</i>	<i>Proceratium crassicorne</i>
Insecta	Hymenoptera	Formicidae	<i>Prenolepis</i>	<i>Prenolepis imparis</i>
Insecta	Hymenoptera	Formicidae	<i>Proceratium</i>	<i>Proceratium pergandei</i>
Insecta	Hymenoptera	Formicidae	<i>Proceratium</i>	<i>Proceratium silaceum</i>
Insecta	Hymenoptera	Formicidae	<i>Solenopsis</i>	<i>Solenopsis carolinensis</i>
Insecta	Hymenoptera	Formicidae	<i>Solenopsis</i>	<i>Solenopsis picta</i>
Insecta	Hymenoptera	Formicidae	<i>Solenopsis</i>	<i>Solenopsis xyloni</i>
Insecta	Hymenoptera	Formicidae	<i>Stenamma</i>	<i>Stenamma impar</i>
Insecta	Hymenoptera	Formicidae	<i>Stenamma</i>	<i>Stenamma schmittii</i>
Insecta	Hymenoptera	Formicidae	<i>Stigmatomma</i>	<i>Stigmatomma pallipes</i>
Insecta	Hymenoptera	Formicidae	<i>Strumigenys</i>	<i>Strumigenys angulata</i>
Insecta	Hymenoptera	Formicidae	<i>Strumigenys</i>	<i>Strumigenys creightoni</i>
Insecta	Hymenoptera	Formicidae	<i>Strumigenys</i>	<i>Strumigenys dietrichi</i>
Insecta	Hymenoptera	Formicidae	<i>Strumigenys</i>	<i>Strumigenys laevinasis</i>
Insecta	Hymenoptera	Formicidae	<i>Strumigenys</i>	<i>Strumigenys louisianae</i>
Insecta	Hymenoptera	Formicidae	<i>Strumigenys</i>	<i>Strumigenys metazytes</i>
Insecta	Hymenoptera	Formicidae	<i>Strumigenys</i>	<i>Strumigenys missouriensis</i>
Insecta	Hymenoptera	Formicidae	<i>Strumigenys</i>	<i>Styrumigenys ohioensis</i>
Insecta	Hymenoptera	Formicidae	<i>Strumigenys</i>	<i>Strumigenys ornata</i>
Insecta	Hymenoptera	Formicidae	<i>Strumigenys</i>	<i>Strumigenys nevermanni</i>
Insecta	Hymenoptera	Formicidae	<i>Strumigenys</i>	<i>Strumigenys pergandei</i>
Insecta	Hymenoptera	Formicidae	<i>Strumigenys</i>	<i>Strumigenys pilinasis</i>
Insecta	Hymenoptera	Formicidae	<i>Strumigenys</i>	<i>Strumigenys rostrata</i>

Table A3 (cont.). Species identified from Steel Creek during this dissertation.

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Class	Order	Family	Genus	Species
Insecta	Hymenoptera	Formicidae	<i>Tapinoma</i>	<i>Tapinoma sessile</i>
Insecta	Hymenoptera	Formicidae	<i>Temnothorax</i>	<i>Temnothorax ambiguus</i>
Insecta	Hymenoptera	Formicidae	<i>Temnothorax</i>	<i>Temnothorax americanus</i>
Insecta	Hymenoptera	Formicidae	<i>Temnothorax</i>	<i>Temnothorax curvispinosus</i>
Insecta	Hymenoptera	Formicidae	<i>Temnothorax</i>	<i>Temnothorax pergandei</i>
Insecta	Hymenoptera	Formicidae	<i>Temnothorax</i>	<i>Temnothorax schaumii</i>
Insecta	Hymenoptera	Formicidae	<i>Temnothorax</i>	<i>Temnothorax texanus</i>
Insecta	Hymenoptera	Formicidae	<i>Trachymyrmex</i>	<i>Trachymyrmex septentrionalis</i>
Insecta	Hymenoptera	Ichneumonidae	<i>Charops</i>	<i>Charops annulipes</i>
Insecta	Hymenoptera	Ichneumonidae	<i>Enicospilus</i>	<i>Enicospilus americanus</i>
Insecta	Hymenoptera	Ichneumonidae	<i>Megarhyssa</i>	<i>Megarhyssa macrurus</i>
Insecta	Hymenoptera	Mymarommatidae		
Insecta	Hymenoptera	Orussidae	<i>Orussus</i>	<i>Orussus minutus</i> [§]
Insecta	Hymenoptera	Orussidae	<i>Orussus</i>	<i>Orussus terminalis</i> [§]
Insecta	Hymenoptera	Pamphiliidae	<i>Onycholyda</i>	<i>Onycholyda luteicornis</i>
Insecta	Hymenoptera	Pamphiliidae	<i>Pamphilius</i>	<i>Pamphilius ocreatus</i>
Insecta	Hymenoptera	Pamphiliidae	<i>Pamphilius</i>	<i>Pamphilius periscum</i>
Insecta	Hymenoptera	Pamphiliidae	<i>Pamphilius</i>	<i>Pamphilius rileyi</i>
Insecta	Hymenoptera	Pergidae	<i>Acordulecera</i>	<i>Acordulecera dorsalis</i>
Insecta	Hymenoptera	Pergidae	<i>Acordulecera</i>	<i>Acordulecera mellina</i>
Insecta	Hymenoptera	Pergidae	<i>Acordulecera</i>	<i>Acordulecera pellucida</i>
Insecta	Hymenoptera	Pompilidae	<i>Ageniella</i>	<i>Ageniella cupida</i>
Insecta	Hymenoptera	Pompilidae	<i>Ageniella</i>	<i>Ageniella partita</i>
Insecta	Hymenoptera	Pompilidae	<i>Agenioideus</i>	<i>Agenioideus birkmanni</i>
Insecta	Hymenoptera	Pompilidae	<i>Allaporus</i>	<i>Allaporus pulchellus</i>
Insecta	Hymenoptera	Pompilidae	<i>Ammosphex</i>	<i>Ammosphex michigenensis michigenensis</i>
Insecta	Hymenoptera	Pompilidae	<i>Anoplius</i>	<i>Anoplius marginatus</i>
Insecta	Hymenoptera	Pompilidae	<i>Aporus</i>	<i>Aporus niger</i>
Insecta	Hymenoptera	Pompilidae	<i>Aporinellus</i>	
Insecta	Hymenoptera	Pompilidae	<i>Arachnospila</i>	
Insecta	Hymenoptera	Pompilidae	<i>Astata</i>	
Insecta	Hymenoptera	Pompilidae	<i>Auplopus</i>	<i>Auplopus architectus architectus</i>
Insecta	Hymenoptera	Pompilidae	<i>Auplopus</i>	<i>Auplopus mellipes mellipes</i>

Table A3 (cont.). Species identified from Steel Creek during this dissertation.

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Class	Order	Family	Genus	Species
Insecta	Hymenoptera	Pompilidae	<i>Auplopus</i>	<i>Auplopus nigrellus</i> or <i>caerulescens</i>
Insecta	Hymenoptera	Pompilidae	<i>Cryptocheilus</i>	<i>Cryptocheilus attenuatum</i>
Insecta	Hymenoptera	Pompilidae	<i>Dipogon</i>	<i>Dipogon sayi sayi</i>
Insecta	Hymenoptera	Pompilidae	<i>Dipogon</i>	<i>Dipogon papago</i> <i>anomalus</i>
Insecta	Hymenoptera	Pompilidae	<i>Priocnemis</i>	<i>Priocnemis hestia</i>
Insecta	Hymenoptera	Pompilidae	<i>Priocnemis</i>	<i>Priocnemis minorata</i>
Insecta	Hymenoptera	Pompilidae	<i>Priocnesis</i>	<i>Priocnensis nebulosus</i>
Insecta	Hymenoptera	Pompilidae	<i>Psorthaspis</i>	
Insecta	Hymenoptera	Pompilidae	<i>Tachypompilus</i>	<i>Tachypompilus</i> <i>ferrugineus ferrugineus</i>
Insecta	Hymenoptera	Rhopalosomatidae	<i>Rhopalosoma</i>	<i>Rhopalosoma nearcticum</i>
Insecta	Hymenoptera	Scoliidae	<i>Scolia</i>	<i>Scolia bicincta</i>
Insecta	Hymenoptera	Siricidae	<i>Tremex</i>	<i>Tremex columba</i>
Insecta	Hymenoptera	Sphecidae	<i>Eremnophila</i>	<i>Eremnophila aureonotata</i>
Insecta	Hymenoptera	Stephanidae	<i>Megischus</i>	<i>Megischus bicolor</i>
Insecta	Hymenoptera	Tenthredinidae	<i>Caliroa</i>	<i>Caliroa quercuscoccineae</i>
Insecta	Hymenoptera	Tenthredinidae	<i>Craterocercus</i>	<i>Craterocercus obtusus</i>
Insecta	Hymenoptera	Tenthredinidae	<i>Dolerus</i>	<i>Dolerus neoagcistus</i>
Insecta	Hymenoptera	Tenthredinidae	<i>Empria</i>	<i>Empria coryli</i>
Insecta	Hymenoptera	Tenthredinidae	<i>Empria</i>	<i>Empria maculata</i>
Insecta	Hymenoptera	Tenthredinidae	<i>Eupareophora</i>	<i>Eupareophora parca</i>
Insecta	Hymenoptera	Tenthredinidae	<i>Hoplocampa</i>	<i>Hoplocampa marlatti</i>
Insecta	Hymenoptera	Tenthredinidae	<i>Macrophya</i>	<i>Macrophya cassandra</i>
Insecta	Hymenoptera	Tenthredinidae	<i>Macrophya</i>	<i>Macrophya formosa</i>
Insecta	Hymenoptera	Tenthredinidae	<i>Macrophya</i>	<i>Macrophya macgillivrayi</i>
Insecta	Hymenoptera	Tenthredinidae	<i>Macrophya</i>	<i>Macrophya pulchella</i>
Insecta	Hymenoptera	Tenthredinidae	<i>Monophadnoides</i>	<i>Monophadnoides</i> <i>conspiculatus</i>
Insecta	Hymenoptera	Tenthredinidae	<i>Monophadnoides</i>	<i>Monophadnoides pauper</i>
Insecta	Hymenoptera	Tenthredinidae	<i>Monophadnoides</i>	<i>Monophadnoides rubi</i>
Insecta	Hymenoptera	Tenthredinidae	<i>Monophadnus</i>	<i>Monophadnus bakeri</i>
Insecta	Hymenoptera	Tenthredinidae	<i>Nefusa</i>	<i>Nefusa ambigua</i>
Insecta	Hymenoptera	Tenthredinidae	<i>Nematus</i>	<i>Nematus abbotii</i>
Insecta	Hymenoptera	Tenthredinidae	<i>Nematus</i>	<i>Nematus tibialis</i>
Insecta	Hymenoptera	Tenthredinidae	<i>Neopareophora</i>	<i>Neopareophora litura</i>

Table A3 (cont.). Species identified from Steel Creek during this dissertation.

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Class	Order	Family	Genus	Species
Insecta	Hymenoptera	Tenthredinidae	<i>Pachynematus</i>	<i>Pachynematus corniger</i>
Insecta	Hymenoptera	Tenthredinidae	<i>Paracharactus</i>	<i>Paracharactus rudis</i>
Insecta	Hymenoptera	Tenthredinidae	<i>Periclista</i>	<i>Periclista marginicollis</i>
Insecta	Hymenoptera	Tenthredinidae	<i>Pristiphora</i>	<i>Pristiphora banski</i>
Insecta	Hymenoptera	Tenthredinidae	<i>Pristiphora</i>	<i>Pristiphora chlorea</i>
Insecta	Hymenoptera	Tenthredinidae	<i>Strongylogaster</i>	<i>Strongylogaster impressata</i>
Insecta	Hymenoptera	Tenthredinidae	<i>Strongylogaster</i>	<i>Strongylogaster remota</i>
Insecta	Hymenoptera	Tenthredinidae	<i>Taxonus</i>	<i>Taxonus eipcera</i>
Insecta	Hymenoptera	Tenthredinidae	<i>Thrinax</i>	<i>Thrinax albidopictus</i>
Insecta	Hymenoptera	Tenthredinidae	<i>Thrinax</i>	<i>Thrinax multicinctus</i>
Insecta	Hymenoptera	Tenthredinidae	<i>Zaschizonyx</i>	<i>Zaschizonyx montana</i>
Insecta	Hymenoptera	Vespidae	<i>Dolcihovespula</i>	<i>Dolcihovespula maculata</i>
Insecta	Hymenoptera	Vespidae	<i>Euodynerus</i>	<i>Euodynerus schwarzi</i>
Insecta	Hymenoptera	Vespidae	<i>Polistes</i>	<i>Polistes fuscatus</i>
Insecta	Hymenoptera	Vespidae	<i>Polistes</i>	<i>Polistes metricus</i>
Insecta	Hymenoptera	Vespidae	<i>Vespa</i>	<i>Vespa crabo</i> [‡]
Insecta	Hymenoptera	Vespidae	<i>Vespula</i>	<i>Vespula maculifrons</i>
Insecta	Hymenoptera	Xyelidae	<i>Xyela</i>	<i>Xyela pini</i>
Insecta	Hymenoptera	Xiphydriidae	<i>Xyphydria</i>	<i>Xyphydria tibialis</i>
Insecta	Lepidoptera	Drepanidae	<i>Euthyatira</i>	<i>Euthyatira pudens</i>
Insecta	Lepidoptera	Erebidae	<i>Apantesis</i>	<i>Apantesis nais</i>
Insecta	Lepidoptera	Erebidae	<i>Apantesis</i>	<i>Apantesis vittata</i>
Insecta	Lepidoptera	Erebidae	<i>Catocala</i>	<i>Catocala dejecta</i>
Insecta	Lepidoptera	Erebidae	<i>Catocala</i>	<i>Catocala epione</i>
Insecta	Lepidoptera	Erebidae	<i>Catocala</i>	<i>Catocala insolabilis</i>
Insecta	Lepidoptera	Erebidae	<i>Catocala</i>	<i>Catocala nebulosa</i>
Insecta	Lepidoptera	Erebidae	<i>Hypsoropha</i>	<i>Hypsoropha monilis</i>
Insecta	Lepidoptera	Erebidae	<i>Euparthenos</i>	<i>Euparthenos nubilis</i>
Insecta	Lepidoptera	Erebidae	<i>Grammia</i>	<i>Grammia anna</i>
Insecta	Lepidoptera	Erebidae	<i>Lycomorpha</i>	<i>Lycomorpha pholus</i>
Insecta	Lepidoptera	Erebidae	<i>Phoberia</i>	<i>Phoberia atomeris</i>
Insecta	Lepidoptera	Erebidae	<i>Zale</i>	<i>Zale lunata</i>
Insecta	Lepidoptera	Geometridae	<i>Epimecis</i>	<i>Epimecis hortaria</i>
Insecta	Lepidoptera	Geometridae	<i>Eutrapela</i>	<i>Eutrapela clemataria</i>
Insecta	Lepidoptera	Hesperiidae	<i>Atalopedes</i>	<i>Atalopedes campestris</i>
Insecta	Lepidoptera	Hesperiidae	<i>Poanes</i>	<i>Poanes hobomok</i>
Insecta	Lepidoptera	Hesperiidae	<i>Poanes</i>	<i>Poanes zabulon</i>

Table A3 (cont.). Species identified from Steel Creek during this dissertation.

† - putative new species, ‡ - introduced, non-native species, § - new state record

Class	Order	Family	Genus	Species
Insecta	Lepidoptera	Lasiocampidae	<i>Malacosoma</i>	<i>Malacosoma americana</i>
Insecta	Lepidoptera	Lasiocampidae	<i>Malacosoma</i>	<i>Malacosoma distria</i>
Insecta	Lepidoptera	Lycaenidae	<i>Calycopis</i>	<i>Calycopis cecrops</i>
Insecta	Lepidoptera	Lycaenidae	<i>Feniseca</i>	<i>Feniseca tarquinius</i>
Insecta	Lepidoptera	Lycaenidae	<i>Satyrrium</i>	<i>Satyrrium favonius</i>
Insecta	Lepidoptera	Noctuidae	<i>Amphipyra</i>	<i>Amphipyra pyramidoides</i>
Insecta	Lepidoptera	Noctuidae	<i>Eupsilia</i>	
Insecta	Lepidoptera	Noctuidae	<i>Eupsilia</i>	<i>Eupsilia vinulenta</i>
Insecta	Lepidoptera	Noctuidae	<i>Morrisonia</i>	<i>Morrisonia confusa</i>
Insecta	Lepidoptera	Noctuidae	<i>Psychomorpha</i>	<i>Psychomorpha epimenis</i>
Insecta	Lepidoptera	Noctuidae	<i>Sericaglaea</i>	<i>Sericaglaea signata</i>
Insecta	Lepidoptera	Nymphalidae	<i>Asterocampa</i>	<i>Asterocampa clyton</i>
Insecta	Lepidoptera	Nymphalidae	<i>Cercyonis</i>	<i>Cercyonis pegala</i>
Insecta	Lepidoptera	Nymphalidae	<i>Chlosyne</i>	<i>Chlosyne nycteis</i>
Insecta	Lepidoptera	Nymphalidae	<i>Lethe</i>	<i>Lethe anthedon</i>
Insecta	Lepidoptera	Nymphalidae	<i>Megisto</i>	<i>Megisto cymela</i>
Insecta	Lepidoptera	Nymphalidae	<i>Nymphalis</i>	<i>Nymphalis antiopa</i>
Insecta	Lepidoptera	Nymphalidae	<i>Speyeria</i>	<i>Speyeria cybele</i>
Insecta	Lepidoptera	Papilionidae	<i>Eurytides</i>	<i>Eurytides marcellus</i>
Insecta	Lepidoptera	Papilionidae	<i>Papilo</i>	<i>Papilo glaucus</i>
Insecta	Lepidoptera	Papilionidae	<i>Papilo</i>	<i>Papilo troilus</i>
Insecta	Lepidoptera	Pieridae	<i>Anthocharis</i>	<i>Anthocharis midea</i>
Insecta	Lepidoptera	Sphingidae	<i>Amphion</i>	<i>Amphion floridensis</i>
Insecta	Lepidoptera	Sphingidae	<i>Hemaris</i>	<i>Hemaris thysbe</i>
Insecta	Lepidoptera	Zygaenidae	<i>Pyromorpha</i>	<i>Pyromorpha dimidiata</i>
Insecta	Mecoptera	Bittacidae	<i>Bittacus</i>	<i>Bittacus pilicornis</i>
Insecta	Mecoptera	Meropeidae	<i>Merope</i>	<i>Merope tuber</i>
Insecta	Mecoptera	Panorpidae	<i>Panorpa</i>	<i>Panorpa braueri</i>
Insecta	Mecoptera	Panorpidae	<i>Panorpa</i>	<i>Panorpa choctaw</i>
Insecta	Orthoptera	Acrididae	<i>Arphia</i>	<i>Arphia sulphurea</i>
Insecta	Orthoptera	Acrididae	<i>Arphia</i>	<i>Arphia xanthoptera</i>
Insecta	Orthoptera	Acrididae	<i>Boopedon</i>	<i>Boopedon gracile</i>
Insecta	Orthoptera	Acrididae	<i>Chortophaga</i>	<i>Chortophaga viridifasciata</i>
Insecta	Orthoptera	Acrididae	<i>Syrbula</i>	<i>Syrbula admirabilis</i>
Insecta	Orthoptera	Gryllidae	<i>Hapithus</i>	<i>Hapithus agitator</i>
Insecta	Orthoptera	Gryllidae	<i>Orocharis</i>	<i>Orocharis saltator</i>
Insecta	Orthoptera	Gryllidae	<i>Velarifictorus</i>	<i>Velarifictorus micado</i> ^{‡§}
Insecta	Orthoptera	Myrmecophilidae	<i>Myrmecophilus</i>	<i>Myrmecophilus pergandei</i>
Insecta	Phasmida	Diapheromeridae	<i>Diapheromera</i>	<i>Diapheromera femorata</i>

Table A3 (cont.). Species identified from Steel Creek during this dissertation.

† - putative new species, ‡ - introduced, non-native species, § - new state record

Class	Order	Family	Genus	Species
Malacostraca	Isopoda	Armadillidiidae	<i>Armadillidium</i>	<i>Armadillidium nasatum</i> [†]
Malacostraca	Isopoda	Armadillidiidae	<i>Armadillidium</i>	<i>Armadillidium vulgare</i> [‡]
Malacostraca	Isopoda	Trichoniscidae	<i>Haplophthalmus</i>	<i>Haplophthalmus danicus</i> [§]

Table A3 (cont.). Species identified from Steel Creek during this dissertation.

† - putative new species, ‡ - introduced, non-native species, § - new state record