# TABLE OF CONTENTS

<table>
<thead>
<tr>
<th>Title</th>
<th>Author(s)</th>
<th>Page</th>
</tr>
</thead>
<tbody>
<tr>
<td>Dispersal of <em>Fenusa dohrnii</em> (Hymenoptera: Tenthredinidae) from an <em>Alnus</em> short-rotation forest plantation</td>
<td>Elwood R. Hart, Richard B. Hall and Roger D. Hanna</td>
<td>63</td>
</tr>
<tr>
<td>Predation by amphibians and small mammals on the spruce budworm (Lepidoptera: Tortricidae)</td>
<td>Daniel T. Jennings, Hewlette S. Crawford, Jr. and Malcolm L. Hunter, Jr.</td>
<td>69</td>
</tr>
<tr>
<td>Alarm pheromone in a gregarious poduromorph collembolan (Collembola: Hypogastruridae)</td>
<td>Foster Forbes Purrington, Patricia A. Kendall, John E. Bater and Benjamin R. Stinner</td>
<td>75</td>
</tr>
<tr>
<td>A review of the genus <em>Gryllus</em> (Orthoptera: Gryllidae), with a new species from Korea</td>
<td>Richard D. Alexander</td>
<td>79</td>
</tr>
<tr>
<td>An annotated checklist of the Lepidoptera of the Beaver Island archipelago, Lake Michigan</td>
<td>Dennis Profant</td>
<td>85</td>
</tr>
<tr>
<td>Noteworthy range extensions of three emesine species (Heteroptera: Reduviidae)</td>
<td>J. E. McPherson</td>
<td>99</td>
</tr>
<tr>
<td>An array of spatulate sensillae on antennae of male <em>Brachymeria lasus</em> (Hymenoptera: Chalcididae)</td>
<td>D. H. Simser and H.C. Coppel</td>
<td>103</td>
</tr>
<tr>
<td>Foodplant processing adaptations in four <em>Hyalophora</em> species (Lepidoptera: Satur- niidae): Regional and taxonomic specialization</td>
<td>J. Mark Scriber and Eric Grabstein</td>
<td>109</td>
</tr>
<tr>
<td>A trap-nest design for small trap-nesting Hymenoptera</td>
<td>John M. Fricke</td>
<td>121</td>
</tr>
<tr>
<td>Trap-nest bore diameter preferences among sympatric <em>Passaloecus</em> spp. (Hymenoptera: Sphecidae)</td>
<td>John M. Fricke</td>
<td>123</td>
</tr>
</tbody>
</table>

**COVER ILLUSTRATION**

Buckeye butterfly, *Junonia coenia* Hübner (Nymphalidae) nectaring on *Apocynum*.

Photo by Brian Scholtens.
THE MICHIGAN ENTOMOLOGICAL SOCIETY

1990-1991 OFFICERS

President Eugene Kenaga
President-Elect Fred Stehr
Executive Secretary M.C. Nielsen
Journal Editor Mark F. O'Brien
Newsletter Editor Robert Haack

The Michigan Entomological Society traces its origins to the old Detroit Entomological Society and was organized on 4 November 1954 to "...promote the science of entomology in all its branches and by all feasible means, and to advance cooperation and good fellowship among persons interested in entomology." The Society attempts to facilitate the exchange of ideas and information in both amateur and professional circles, and encourages the study of insects by youth. Membership in the Society, which serves the North Central States and adjacent Canada, is open to all persons interested in entomology. There are four paying classes of membership:

Student (including those currently enrolled as college sophomores) — annual dues $4.00
Active — annual dues $8.00
Institutional — annual dues $20.00
Sustaining — annual contribution $25.00 or more
Life — $160.00

Dues are paid on a calendar year basis (Jan. 1 – Dec. 31).

Memberships accepted before July 1 shall begin on the preceding January 1; memberships accepted at a later date shall begin the following January 1 unless the earlier date is requested and the required dues are paid. All members in good standing receive the Newsletter of the Society, published quarterly. All active and sustaining members may vote in Society affairs.

All dues and contributions to the Society are deductible for Federal income tax purposes.

SUBSCRIPTION INFORMATION

Institutions and organizations, as well as individuals not desiring the benefits of membership, may subscribe to The Great Lakes Entomologist at the rate of $15.00 per volume. The journal is published quarterly; subscriptions are accepted only on a volume (4 issues) basis. Single copies of The Great Lakes Entomologist are available at $4.25 each, with a 20 percent discount for 25 or more copies sent to a single address.

MICROFILM EDITION: Positive microfilm copies of the current volume of The Great Lakes Entomologist will be available at nominal cost, to members and bona fide subscribers of the paper edition only, at the end of each volume year. Please address all orders and inquiries to University Microfilms, Inc., 300 Zeeb Road, Ann Arbor, Michigan 48106, USA.

Inquiries about back numbers, subscriptions and Society business should be directed to the Executive Secretary, Michigan Entomological Society, Department of Entomology, Michigan State University, East Lansing, Michigan 48824–1115, USA. Manuscripts and related correspondence should be directed to the Editor (see inside back cover).

Copyright © 1991. The Michigan Entomological Society
DISPERAL OF *FENUSA DOHRNII* (HYMENOPTERA: TENTHREDINIDAE) FROM AN *ALNUS* SHORT-ROTATION FOREST PLANTATION

Elwood R. Hart¹, ², Richard B. Hall², and Roger D. Hanna²

ABSTRACT

The European alder leafminer, *Fenusa dohrnii*, is a defoliating insect pest of *Alnus* in short-rotation forest plantations. A 2-year study was performed to quantify movement from infested stands to uninfested areas. Sticky traps and potted monitor trees were installed at different locations within and at various distances from (0, 5, 10, and 20 m) an infested stand to measure adult flight and oviposition activity, respectively. Trap catch and oviposition activity fell off sharply with distance, few insects being trapped or eggs laid at distances of 5 m or greater from the infestation.

The genus *Alnus*, the alders, has been considered a promising group for biomass plantations because of its rapid growth and symbiotic nitrogen-fixing capabilities. Genetic improvement work with *Alnus* began at Iowa State University in 1976 with a range-wide collection of germplasm for European black alder, *A. glutinosa* Gaertn. (Hall et al. 1983). Selected populations of other *Alnus* species have been grown here as well.

In 1982, the European alder leafminer, *Fenusa dohrnii* (Tischbein) (Hymenoptera: Tenthredinidae), began developing as a pest in our local plantations (Hart et al. 1991). By 1985, much defoliation was obvious, prompting concern about the impact of the insect on the plantations and possible management measures. Because little was known about the insect in the north-central United States, a series of studies was initiated in 1986 to define the field biology and impact of the insect in the plantation environment.

Sustained yield from biomass plantations involves having all growth and coppice-regrowth stages of trees present at various proximities within the growing area. Tree growth and survivorship are especially sensitive to growing conditions, including defoliation effects, in the first 2 years after planting (Meridian Corporation, 1986). The ability of *F. dohrnii* to move from infested to uninfested, newly-planted stands can thus determine, in part, the impact that it may have on the plantations and how they should be designed.

When a plantation is cut, the first adult flight of European alder leafminer in the spring precedes most of the resprouting of stems and leaves on the cut alder stumps (Hart et al. 1991). This provides an opportunity to reduce the leafminer population significantly in the area for a time. Reinfestation, however, may occur from late-emerging insects and from adjacent uncut stands. Adult dispersal is therefore important in the reinfestation of harvested areas.

¹Department of Entomology, Iowa State University, Ames, IA 50011.
²Department of Forestry, Iowa State University, Ames, IA 50011.
Because only the adult stage of the leafminer is capable of significant interplant movement, a study was designed to quantify adult movement from known infested stands to uninfested areas. The major objectives of this study were: (1) to determine the distances that the insect can migrate in significant numbers from infested stands to uninfested areas and (2) to use this information to project practical planting distances from such infested areas.

**MATERIALS AND METHODS**

The research was performed in central Iowa at the Iowa State University Rhodes Experimental Farm. The plantation site lies in a narrow stream valley, surrounded by low, forested hills. The site is well-buffered from the prevailing winds, which in the growing season are mostly from the southeast. The first alders were planted at this location in 1979, with additional plantings added each year as needed by the breeding and selection program.

In 1987, two infested, unharvested plantings were selected as European alder leafminer source areas (Fig. 1). Four replications were established, two at Unit 4 of the 1979 provenance test plantings and two at the 1981 clonal trial plantings. Each replication was organized along a transect line away from a planting into an open, grassy field. For each area, a single monitor station was established within the planting. Each replication also consisted of monitor stations at the edge of the planting and at 5, 10, and 20 m from the edge of the planting. Adult presence and activity were measured at different heights at each station by two different methods: (1) yellow sticky traps to attract and capture insects that flew into the area; (2) potted trees of susceptible *A. glutinosa* selections to measure oviposition activity in the area.
For the first week of the study, the sticky traps consisted of 15 cm by 3 m PVC pipe, painted yellow and coated with Tangle Trap® (The Tanglefoot Co., Grand Rapids, MI); trap catch was measured for each 0.5-m section to a height of 3.0 m. Because the large surface area created logistical problems in counting and cleaning, the traps were replaced after 1 week with Pherocon® AM (Trece, Inc., Salinas, CA) cards. The flattened cards were centered on and stapled to wooden standards at six 0.5-m heights above ground level, from 0.5 to 3.0 m. Leafminers were counted and removed weekly from the central 8 by 20 cm section of each trap. Adult activity was measured as the number of trapped adults/cm²/day.

To monitor oviposition activity at each station, greenhouse-grown potted trees, 45 to 60 cm tall, from two leafminer-susceptible *A. glutinosa* selections, SN-5 and SN-53 (Hall and Nyong'o 1987), were set out at two heights, ground level and 2.5 m. For each height, a potted tree from each selection was set into a beige plastic tub, watered from below, and replaced weekly. Four newly expanded leaves, those highly preferred for oviposition (Hart et al. 1991), were taken from each tree as it was removed from the field. Leaves were taken to the laboratory where egg counts and leaf area were measured. Female activity was defined as eggs/cm²/day.

Under the climatic conditions of the central Iowa *Alnus* plantations, three generations of the leafminer occur each year (Hart et al. 1991). In 1987, monitor stations were established from 30 June through 28 July and from 11 August through 22 September to include the second and third adult activity periods.

In 1988, the study was modified and expanded to include five replications. Three unharvested plantings were selected as leafminer-infested sources (Fig. 1). One replication was established at Unit 4 of the 1979 provenance test plantings, two at Unit 2 of the 1979 provenance test plantings, and two at the 1981 clonal trial planting. Each replication consisted of a transect line established into an open area away from a planting. Separate within-planting monitor stations were established for replications 1, 2, and 3; replications 4 and 5 shared a common within-planting monitor station. Transect stations were established at the edge and at distances from the edge as in 1987. Adult presence and activity again were measured at each station with sticky traps and potted trees.

The sticky traps were stapled to wooden standards and centered at three heights above ground level, 0.5, 1.5, and 3.0 m. Weekly, during each adult activity period, trap catches were counted from the central 8 by 20 cm section and the traps cleaned.

For each transect, trees were placed on platforms at the height of the prevailing foliage within and at the edge of the planting; at each of the outlying stations the trees were placed on the ground to simulate the location of newly planted trees. Two trees, 45 to 60 cm tall, one each from SN-5 and SN-53, were used in 1988. Trees again were placed in beige plastic tubs, watered from below, and replaced weekly. Two newly expanded leaves were taken from each tree as it was removed from the field. Leaves were taken to the laboratory and examined for oviposition density.

In 1988, monitor stations were established from 13 May to 3 June, from 14 June to 5 July, and from 26 July to 13 September to include the first, second, and third adult activity periods, respectively.

The data were analyzed by PROC GLM from the Statistical Analysis System (SAS Institute 1985a, b). When analysis indicated a significant difference (P < 0.05) for a given variable, the Duncan's option was used as the mean separation procedure.

RESULTS

Leafminer populations were quite high during the first adult activity period in 1988 and during the second adult activity period in both 1987 and 1988 (Hart et al. 1991), producing sufficiently measurable numbers for comparisons among locations
Table 1. European alder leafminer trap-catch density by sampling date.

<table>
<thead>
<tr>
<th>Date</th>
<th>N</th>
<th>EAL/cm²/day²</th>
<th>Date</th>
<th>N</th>
<th>EAL/cm²/day²</th>
</tr>
</thead>
<tbody>
<tr>
<td>July 14</td>
<td>108</td>
<td>0.0084 A</td>
<td>June 21</td>
<td>72</td>
<td>0.0193 A</td>
</tr>
<tr>
<td>July 9</td>
<td>108</td>
<td>0.0069 B</td>
<td>June 28</td>
<td>72</td>
<td>0.0187 A</td>
</tr>
<tr>
<td>Aug 18</td>
<td>108</td>
<td>0.0026 C</td>
<td>July 5</td>
<td>72</td>
<td>0.0074 B</td>
</tr>
<tr>
<td>July 28</td>
<td>108</td>
<td>0.0011 D</td>
<td>May 20</td>
<td>72</td>
<td>0.0048 C</td>
</tr>
<tr>
<td>July 21</td>
<td>108</td>
<td>0.0011 D</td>
<td>May 27</td>
<td>72</td>
<td>0.0031 C</td>
</tr>
<tr>
<td>Sept 1</td>
<td>108</td>
<td>0.0005 D</td>
<td>June 3</td>
<td>72</td>
<td>0.0005 D</td>
</tr>
<tr>
<td>Sept 22</td>
<td>107</td>
<td>0.0004 DE</td>
<td>Aug 2</td>
<td>72</td>
<td>0.0002 D</td>
</tr>
<tr>
<td>Sept 15</td>
<td>102</td>
<td>0.0001 E</td>
<td>Aug 9</td>
<td>72</td>
<td>0.0002 D</td>
</tr>
<tr>
<td>Aug 27</td>
<td>108</td>
<td>0.0001 E</td>
<td>Aug 30</td>
<td>59</td>
<td>0.0001 E</td>
</tr>
<tr>
<td>Sept 8</td>
<td>102</td>
<td>0.0001 E</td>
<td>Aug 23</td>
<td>64</td>
<td>0.0001 E</td>
</tr>
<tr>
<td>Aug 16</td>
<td>72</td>
<td>0.0000 E</td>
<td>Aug 16</td>
<td>72</td>
<td>0.0000 E</td>
</tr>
<tr>
<td>Sept 6</td>
<td>72</td>
<td>0.0000 E</td>
<td>Sept 13</td>
<td>69</td>
<td>0.0000 E</td>
</tr>
</tbody>
</table>

*Means followed by same letter are not significantly different. DNMRT*

Table 2. European alder leafminer trap-catch density by trap location.

<table>
<thead>
<tr>
<th>Trap Location</th>
<th>N</th>
<th>EAL/cm²/day²</th>
<th>Trap Location</th>
<th>N</th>
<th>EAL/cm²/day²</th>
</tr>
</thead>
<tbody>
<tr>
<td>Edge</td>
<td>48</td>
<td>0.0233 A</td>
<td>Within</td>
<td>60</td>
<td>0.3054 A</td>
</tr>
<tr>
<td>Within</td>
<td>24</td>
<td>0.0171 B</td>
<td>Edge</td>
<td>75</td>
<td>0.1027 B</td>
</tr>
<tr>
<td>5 m out</td>
<td>48</td>
<td>0.0013 C</td>
<td>5 m out</td>
<td>75</td>
<td>0.0072 C</td>
</tr>
<tr>
<td>10 m out</td>
<td>48</td>
<td>0.0010 C</td>
<td>10 m out</td>
<td>75</td>
<td>0.0024 C</td>
</tr>
<tr>
<td>20 m out</td>
<td>48</td>
<td>0.0003 C</td>
<td>20 m out</td>
<td>75</td>
<td>0.0017 C</td>
</tr>
</tbody>
</table>

*Means followed by same letter are not significantly different. DNMRT*

Table 3. European alder leafminer density by trap height.

<table>
<thead>
<tr>
<th>Trap Height (m)</th>
<th>N</th>
<th>EAL/cm²/day²</th>
<th>Trap Height (m)</th>
<th>N</th>
<th>EAL/cm²/day²</th>
</tr>
</thead>
<tbody>
<tr>
<td>3.0</td>
<td>36</td>
<td>0.0121 A</td>
<td>3.0</td>
<td>120</td>
<td>0.0836 A</td>
</tr>
<tr>
<td>2.5</td>
<td>36</td>
<td>0.0090 B</td>
<td>2.0</td>
<td>36</td>
<td>0.0076 BC</td>
</tr>
<tr>
<td>2.0</td>
<td>36</td>
<td>0.0069 C</td>
<td>1.5</td>
<td>36</td>
<td>0.0067 C</td>
</tr>
<tr>
<td>1.0</td>
<td>36</td>
<td>0.0036 D</td>
<td>0.5</td>
<td>36</td>
<td>0.0036 D</td>
</tr>
</tbody>
</table>

*Means followed by same letter are not significantly different. DNMRT*
Figure 2. Effects of sticky trap height and position on trap catch density relative to infested areas, Rhodes, Iowa, 1987, 1988.

(Hart et al. 1991), data from the two selections were pooled for further analyses. Oviposition density followed the same trend as sticky trap catches, but did not fall off quite as sharply with distance (1987: $F = 27.86$, df = 4, 31, $P < 0.0001$; 1988: $F = 10.92$, df = 4, 229, $P < 0.0001$) (Table 4).

The small amount of adult activity outside the infested area implies that new plantings at distances of 10 m or greater from infested areas should have 1 to 2 years of relatively low infestations, an important factor in the establishment of new trees. The less drastic decrease in oviposition density with distance from the population
Table 4. European alder leafminer oviposition density by tree location.

<table>
<thead>
<tr>
<th>Tree Location</th>
<th>1987</th>
<th>1988</th>
</tr>
</thead>
<tbody>
<tr>
<td></td>
<td>Eggs/cm²/day</td>
<td>Eggs/cm²/day</td>
</tr>
<tr>
<td>Within</td>
<td>0.2022 A</td>
<td>0.0979 A</td>
</tr>
<tr>
<td>Edge</td>
<td>0.1864 A</td>
<td>0.0500 B</td>
</tr>
<tr>
<td>5 m out</td>
<td>0.0329 B</td>
<td>0.0072 C</td>
</tr>
<tr>
<td>20 m out</td>
<td>0.0190 B</td>
<td>0.0062 C</td>
</tr>
<tr>
<td>10 m out</td>
<td>0.0029 B</td>
<td>0.0031 C</td>
</tr>
</tbody>
</table>

*Means followed by same letter are not significantly different. DNMRT*

source, compared with that of trap-catch density, however, suggests that a host-finding mechanism may be involved in dispersal.

ACKNOWLEDGMENTS

We thank John Kean, Janelle Hall, and Carla Duncan for their assistance in gathering data. Research partly supported under Subcontract No. 19X-43391C with Oak Ridge National Laboratory under Martin Marietta Energy Systems, Inc. contract DE-840R21400 with the U. S. Department of Energy, and through Iowa Science Foundation grant ISF-87-47, administered by the Iowa Academy of Science. Journal Paper No. J-13815 of the Iowa Agriculture and Home Economics Experiment Station, Ames, Iowa, Projects No. 2210 and 2731.

LITERATURE CITED


Meridian Corporation. 1986. Short-rotation intensive culture of woody crops for energy: principles and practices for the Great Lakes region. Meridian Corporation, Falls Church, VA.


PREDATION BY AMPHIBIANS AND SMALL MAMMALS ON THE SPRUCE BUDWORM (LEPIDOPTERA: TORTRICIDAE)

Daniel T. Jennings¹ Hewlette S. Crawford, Jr.² and Malcolm L. Hunter, Jr.³

ABSTRACT

Stomach-content analyses of pitfall-trapped amphibians and small mammals showed that the eastern American toad, Bufo americanus americanus, and the wood frog, Rana sylvatica, preyed on late instars and moths of the spruce budworm, Choristoneura fumiferana. The spotted salamander, Ambystoma maculatum, and the masked shrew, Sorex cinereus, also preyed on late instars of the spruce budworm.

Known predators of the spruce budworm, Choristoneura fumiferana (Clem.), include both invertebrates and vertebrates (Jennings and Crawford 1985). Of the vertebrate predators, birds are the best known and most extensively studied (Dowden et al. 1953, Morris 1963, Crawford et al. 1983, Crawford and Jennings 1989). Although small mammals have long been implicated as potential predators of the spruce budworm (Morris et al. 1958, Morris 1963, Otvos 1981, Kelly and Régnière 1985), definitive evidence of such predation generally has been lacking, except for the red squirrel, Tamiasciurus hudsonicus (Bangs), (Dowden et al. 1953, Jennings and Crawford 1989). Because of their dependence on fresh water, amphibians generally are considered unimportant predators of forest insects (Buckner 1966). Nevertheless, budworm larvae and pupae are susceptible to predation by terrestrial vertebrates, such as amphibians and small, insectivorous mammals, when the large larvae drop from host-tree crowns to the forest floor (Morris and Mott 1963, Kelly and Régnière 1985). Amphibian populations were monitored during budworm suppression projects in Maine (Banasiak 1974, Peterson 1976, Sassaman 1978), but predation on the spruce budworm was not determined.

Here we describe for the first time predation by amphibians (both anurans and caudates) on late instars and moths of the spruce budworm. We also provide definitive evidence of predation by small mammals (insectivores) on late instars of C. fumiferana.

MATERIALS AND METHODS

Study Sites. We pitfall-trapped amphibians and small mammals for two years (1977, 1978) in a spruce-fir (Picea-Abies) forest that was heavily infested with the spruce budworm. Individual study sites were 48–61 km northwest of Millinocket,

¹Northeastern Forest Experiment Station, 180 Canfield St., Morgantown, WV 26505.
²Northeastern Forest Experiment Station, USDA Building, University of Maine, Orono, ME 04469.
³Department of Wildlife, College of Forest Resources, University of Maine, Orono, ME 04469.
Piscataquis County, Maine (45°45' - 46°10' N, 68°55' - 69°20' W). Portions of the forest had been strip clearcut, resulting in alternating clearcut and uncut residual strips. We investigated 5 strip-clearcut stands and 5 nearby dense (uncut) stands in 1977; 7 strip-clearcut stands and 3 dense stands were investigated in 1978. For details of study-site vegetation, sampling design, and associated invertebrates, see Jennings et al. (1984, 1986a, 1986b, 1988).

**Pitfall Traps.** For both study years, we used 40 large-capacity pitfall traps (Houseweart et al. 1979) to capture terrestrial amphibians and small mammals. Each trap bottle (1 liter) contained ca. 300 ml of a 1:1 mixture of ethylene glycol (antifreeze) and 70% ethanol as a killing-preservative agent. Four traps were placed in each strip-clearcut stand (5 replications, 1977; 7 replications, 1978); one trap each in two clearcut strips and in two adjacent uncut residual strips (see Fig. 1, Jennings et al. 1984). Correspondingly, four traps were placed in each nearby dense (uncut) stand investigated (5 replications, 1977, 3 replications, 1978). Traps were open continuously and their contents collected weekly for 10 weeks (26 May - 4 August) in 1977, and for 11 weeks (18 May - 3 August) in 1978. Total sampling effort was 5580 trap nights.

**Identifications.** Pitfall-trap contents were transferred to small jars and transported to the laboratory where collections were sorted and identified. Amphibian identifications follow Conant (1975); small mammal identifications follow Burt and Grossenheider (1976) and Godin (1977). All vertebrate identifications were verified by comparison with museum specimens retained in the Department of Wildlife, and Department of Zoology, University of Maine, Orono. Voucher specimens of collected amphibians and small mammals have been deposited in the collections of the Department of Wildlife, College of Forest Resources, University of Maine, Orono.

**Stomach-Content Analyses.** Digestive tracts of captured amphibians (n = 134; 5 desiccated specimens not dissected) and of small mammals (Insectivora only; n = 41) were removed and their contents examined with a stereomicroscope. Larval mandibles, pupal cremasters, and moth genitalia were compared with a reference collection of the spruce budworm and associated insects. The chitinized mandibles, cremasters, and genitalic parts are the most reliable diagnostic structures for identifying remains of *C. funiferana* in predator stomachs (Crawford and Jennings 1982). For amphibian stomach contents, larval instars of the spruce budworm were determined by head-capsule size (McGugan 1954). Because larval head capsules are seldom found intact in small mammal stomachs, we estimated instars based on relative mandible size, i.e., ocular estimates of mandible size compared with reference material.

**RESULTS**

**Amphibian Numbers and Species.** One hundred thirty-nine amphibians representing two orders, five families, five genera, and six species were pitfall-trapped in spruce-fir forests of northern Maine. The species and total numbers trapped were: eastern American toad, *Bufo americanus americanus* Holbrook, (n = 64); wood frog, *Rana sylvatica* LeConte, (n = 62); redback salamander, *Plethodon cinereus* (Green), (n = 9); red-spotted newt, *Notophthalmus viridescens viridescens* (Rafinesque), (n = 1); spotted salamander, *Ambystoma maculatum* (Shaw), (n = 1); and blue-spotted salamander, *Ambystoma laterale* Hallowell, (n = 2). Anura (toads, frogs) were found in all three forest conditions studied; Caudata (salamanders, newts) were found mostly in uncut residual strips and in dense (uncut) stands.

**Amphibian Predation on Spruce Budworm.** For both study years, amphibians fed on larvae and moths of the spruce budworm (Table 1). However, the percentage of pitfall-trapped amphibians that had eaten budworm prey was low (< 7.5%, n = 134). Numbers of spruce budworm prey per stomach ranged from 1 to 6; most
were late instars (L₅, L₆), the key age interval that influences generation survival of the spruce budworm (Morris 1963).

Small Mammal Numbers and Species. Forty-six small mammals representing two families, four genera, and five species were pitfall-trapped in strip-clearcut and dense (uncut) spruce-fir stands of northern Maine. The species and total numbers trapped were: masked shrew, *Sorex cinereus* Kerr, (n = 33); smoky shrew, *Sorex fumeus* Miller, (n = 1); pygmy shrew, *Microsorex hoyi* (Baird), (n = 7); red-backed vole, *Clethrionomys gapperi* (Vigors), (n = 2); and southern bog lemming, *Synaptomys cooperi* Baird, (n = 3). Most (90.2%) of the Insectivora were trapped in strip-clearcut stands, both in residual (uncut) strips and in strip clearcuts. Although few Rodentia were caught, they were trapped in all three forest conditions studied.

Small Mammal Predation on Spruce Budworm. Digestive tracts of 16 (48%, n = 33) masked shrews and 3 (44%, n = 7) pygmy shrews contained insect larval mandibles (range 1 - 8). One tract of a masked shrew had an insect pupal cremaster. However, only two of these insect parts could be positively identified as remains of *C. fumiferana*. One distinctive spruce budworm mandible was found in each digestive tract of 2 masked shrews, both captured on 30 June 1977 in residual strips. A possible mandible of the spruce budworm also was found in the digestive tract of 1 pygmy shrew, but positive identification was impossible because distinctive features (mandibular teeth and condyle) were missing.

DISCUSSION

Our stomach-content analyses of amphibian foods provides the first evidence that *Ambystoma maculatum* preys on larvae of the spruce budworm. The results of this study also indicate that both spruce budworm larvae and moths are included in the diets of *Bufo a. americanus* and *Rana sylvatica*. All three species of amphibians should be added to the list of known predators of the spruce budworm (Jennings and Crawford 1985).

We suspect that amphibian predation on the spruce budworm is opportunistic, and is influenced by prey availability and prey activity. Only the more active life stages (i.e., larvae and moths) of the spruce budworm were eaten by amphibians. Although pre-pupae and pupae of the spruce budworm also may drop from host-tree crowns to the forest floor (Morris and Mott 1963, Kelly and Régnière 1985), none were eaten by the pitfall-trapped amphibians (Table 1). Hence, these less active, generally immobile life stages may escape detection by amphibians.

Because some amphibians frequently eat ants (Hamilton 1954, LeClair and Vallières 1981), and most budworm life stages are susceptible to predation by ants (Finnegan 1978, McNeil et al. 1978), secondary predation may occur when ants transport budworm prey back to the nest. At least four of the eastern American toads examined during this study contained both budworm and ant prey in their stomachs.

Our stomach-content analyses of small mammals confirms previously suspected but unsubstantiated predation by *S. cinereus* on large larvae of the spruce budworm. Otvos (1981) included the masked shrew among three small mammals suspected to feed on the spruce budworm in Newfoundland; however, stomach-content analyses were not made. Our study indicates that the masked shrew, and possibly the pygmy shrew, should be added to the list of vertebrate predators of the spruce budworm (Jennings and Crawford 1985).

The extent and relative importance of predation by amphibians and small mammals on the spruce budworm generally are unknown. Kelly and Régnière (1985) concluded that predation on spruce budworm pupae on the forest floor was high (72.5% per day), and was largely attributable to vertebrate predators. No doubt the susceptibilities to predation by terrestrial vertebrates varies among the different
Table 1. Predation by amphibians on the spruce budworm based on stomach-content analyses (N = 134) of pitfall-trap collections, Piscataquis County, Maine.

<table>
<thead>
<tr>
<th>Date</th>
<th>Predator species</th>
<th>Habitat*</th>
<th>Spruce budworm number and life stage**</th>
</tr>
</thead>
<tbody>
<tr>
<td>1977</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>30 June</td>
<td><em>Bufo a. americanus</em></td>
<td>D</td>
<td>3 - L6</td>
</tr>
<tr>
<td>30 June</td>
<td><em>Rana sylvatica</em></td>
<td>R</td>
<td>1 - L5</td>
</tr>
<tr>
<td>30 June</td>
<td><em>Rana sylvatica</em></td>
<td>R</td>
<td>1 - L6</td>
</tr>
<tr>
<td>21 July</td>
<td><em>Bufo a. americanus</em></td>
<td>C</td>
<td>1 - L7</td>
</tr>
<tr>
<td>4 Aug.</td>
<td><em>Rana sylvatica</em></td>
<td>D</td>
<td>1 - ♀ moth</td>
</tr>
<tr>
<td>1978</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>1 June</td>
<td><em>Bufo a. americanus</em></td>
<td>D</td>
<td>1 - L5</td>
</tr>
<tr>
<td>15 June</td>
<td><em>Bufo a. americanus</em></td>
<td>D</td>
<td>1 - L3, 3 - L4, 1 - L5, 1 - L6</td>
</tr>
<tr>
<td>15 June</td>
<td><em>Ambystoma maculatum</em></td>
<td>D</td>
<td>1 - L4, 1 - L5, 1 - L6</td>
</tr>
<tr>
<td>22 June</td>
<td><em>Bufo a. americanus</em></td>
<td>D</td>
<td>1 - L5, 1 - L6</td>
</tr>
<tr>
<td>20 July</td>
<td><em>Bufo a. americanus</em></td>
<td>R</td>
<td>1 - ♀ moth</td>
</tr>
</tbody>
</table>

*D = dense (uncut) stand; R = uncut residual strip; C = clearcut strip.

**L3 . . . L6 instar.

budworm life stages (eggs, larvae, pre-pupae, pupae, moths). Such predation also may be influenced by budworm behavior (e.g., larval-pupal droppage from host-tree crowns; increased mobility of starved larvae; moth-flight activity) and population density (i.e., potential-prey abundance). Predator abundance and feeding behavior (generalist vs. specialist) also are factors.

The large-capacity pitfall trap used in this study was primarily designed to capture terrestrial invertebrates such as ants, carabid beetles, phalangids, and spiders (Houseweart et al. 1979). Because this trap has a plastic funnel (15 cm) that tapers to a 2.54-cm diameter spout, it no doubt was selectively biased toward capture of small-sized amphibians and insectivores. For future studies, larger-diameter traps with drift fences may yield substantially greater catches.

LITERATURE CITED


Peterson, J. W. 1976. Effects of Dylox, Matacil, and Sumithion on birds, small mammals and amphibians, pp. 2-10. In: 1975 Cooperative Pilot Control Project of Dylox, Matacil, and

ALARM PHEROMONE IN A GREGARIOUS PODUROMORPH COLLEMBOLAN (COLLEMBOLA: HYPOGASTRURIDAE)1

Foster Forbes Purrington2, Patricia A. Kendall3, John E. Bater2, and Benjamin R. Stinner4

ABSTRACT

We report an alarm pheromone in the gregarious poduromorph collembolan, Hypogastrura pannosa. Cuticular rupture results in emission of a rapidly vaporizing hexane-soluble material with an active space diameter of ca. 1 cm. Conspecifics encountering the vapor front respond with stereotypic aversion and dispersal behaviors. This is the first report on the presence of an alarm pheromone in the order Collembola.

Alarm pheromones are well known in eusocial insects (Isoptera, Hymenoptera) where responses involve complex defensive, recruitment and dispersal behaviors (reviewed by Blum 1985, Hölldobler and Wilson 1990). In his studies of the quasi-social treehopper, Umbonia crassicornis Amyut & Serville (Homoptera: Membracidae), Wood (1976) showed that abdominal wounding of nymphs prompted chemically mediated alarm and active defense of offspring by brooding female parents.

Socially less organized taxa such as several species of gregarious bugs in three families of the Hemiptera (reviewed by Blum 1985) also use alarm pheromones, and they are widespread if not universal among the aphids (Homoptera: Aphidae) (e.g. Nault and Bowers 1974). An alarm pheromone is present in the mold mite, Tyrophagus putrescentiae (Schrank) (Acarina: Acaridae) and in several related species (Kuwahara et al. 1980).

We now report an alarm pheromone for the first time in a fifth insect order, the Collembola.

MATERIALS AND METHODS

We obtained eggs and adults of Hypogastrura pannosa Macnamara (Collembola: Hypogastruridae) from rotting fruit of native persimmon (Diospyros virginiana L.) on 10 Oct. 1989 at Wooster, Ohio. Isotomurus bimus Christiansen & Bellinger (Isotomidae) and Lepidocyrtus pallidus Reuter (Entomobryidae) were collected from a greenhouse mist-bed on 15 Mar. 1990 at Wooster. Neanura muscorum (Templeton) (Neanuridae), Megalothorax minimus (Willem) (Neelidae), and Smin-
thurinus elegans (Fitch) (Sminthuridae) were also taken at Wooster, from extractions of leaf litter in a dense landscape planting of mature American holly (Ilex opaca). Folsomia candida Willem (Isotomidae) was obtained from laboratory cultures at the O.A.R.D.C., Wooster. We collected Onychiurus encarpatus Denis (Onychiuridae) from an agricultural field soil (winter wheat) at the USDA North Appalachian Experimental Watershed site, Coshocton, Ohio on 19 Apr. 1990. To obtain collembolans from soil and litter we used a modified Tullgren funnel extractor at the Soil Ecology Laboratory, O.A.R.D.C., Wooster, Ohio.

Collembola were maintained on active dry yeast in containers with moist plaster of Paris floors at 28°C.

To determine solubility characteristics and alarm inducing properties of the active material, we placed ca. 5000 H. pannosa adults into 1 ml acetone, and an equal number into 1 ml hexane (2 μl = ca. 10 adult-equivalents). We challenged a variety of collembolan species with these extracts dried onto filter paper points held with forceps or mounted on insect pins.

RESULTS AND DISCUSSION

Crushed Hypogastrura pannosa with broken cuticles evoked immediate alarm responses when held a few mm above conspecifics in a culture jar. The behavioral response sequence of a freely traversing individual to perception of the released volatiles typically included a stop with antennal waving, grading immediately into a general recoil. Extracts of H. pannosa eluted in hexane and completely evaporated onto filter paper points, at concentrations ranging from two to 10 adult-equivalents, gave results approximately similar to those using single crushed individuals, with no clear elevation of response intensity at higher concentrations. Neither extracts in acetone, pure acetone, nor pure hexane evaporated onto filter paper points evoked responses.

After flinching and general bodily contraction, behavior in the presence of the alarm releasing material included either ‘freezing’ or turn-and-run, although ‘freezing’ was not frequent. In this species the ultimate collembolan option of springing was seldom seen used as an escape strategy in the context of alarm behavior; by far the most frequently observed alarm sequence was stop-flinch-turn-run.

Emission of alarm releasing volatiles from a single freshly crushed H. pannosa generated an active space of ca. 1 cm diam. This zone, which was virtually cleared of conspecifics within 60 s, typically remained cleared for more than one h. All instars were alarmed similarly by the vaporizing materials; hatchling H. pannosa are well endowed with alarm releasing compounds and when crushed they too triggered strong aversion in conspecifics. Crushed eggs, however, did not cause noticeable alarm.

Recently, from studies he made of H. socialis (Uzel), Leinaas (1988) speculates on the function of eversible anal glands, suggesting their possible role in the release of alarm inducing volatiles, analogous to the terminal eversible sex pheromone gland of the female moth abdomen. He asserts that in H. socialis jumps are invariably accompanied by eversion of anal sacs. It is difficult for us to accept his implication that the putative release of alarm inducer chemicals is a concomitant of saltation in H. socialis or any gregarious collembolan, that such “crying wolf” has any adaptive value for sociality.

In our studies, short of rupturing the integument, no amount of jostling, probing, or other mechanical harassment of adult H. pannosa by us resulted in stereotypic aversion behavior by nearby conspecifics. For that reason we do not, on the basis of our preliminary findings, support a view that these insects propagate alarm pheromone by everting anal sacs or with any controlled glandular emission.

In Table 1 we show results of test responses of eight species in seven collembolan
Table 1. Cross-taxa alarm/aversion responses between and within 8 species in 7 collembolan families to freshly crushed individuals of these species. 1/, 2/.


<table>
<thead>
<tr>
<th>CHALLENGER SPECIES (crushed)</th>
<th>A</th>
<th>B</th>
<th>C</th>
<th>D</th>
<th>E</th>
<th>F</th>
<th>G</th>
<th>H</th>
</tr>
</thead>
<tbody>
<tr>
<td>A</td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td>+</td>
</tr>
<tr>
<td>B</td>
<td>+</td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>C</td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td>+</td>
<td>+</td>
</tr>
<tr>
<td>D</td>
<td></td>
<td>+</td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>E</td>
<td>+</td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>F</td>
<td>+</td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>G</td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td>+</td>
<td></td>
<td></td>
</tr>
<tr>
<td>H</td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td>+</td>
<td></td>
</tr>
</tbody>
</table>

1/ Each score ≥ 5 replications.
2/ Species codes:
   A. Onychiurus encarpatus
   B. Hypogastrura pannosa
   C. Neanura muscorum
   D. Folsomia candida
   E. Isotomurus bisus
   F. Lepidocyrtus pallidus
   G. Megalothorax minimus
   H. Sminthurinus elegans

families to freshly crushed individuals of the eight species. *Megalothorax minimus* and *Folsomia candida* both responded slightly to *H. pannosa* volatiles. Crushed *Onychiurus encarpatus* elicited mild aversion in both *F. candida* and *H. pannosa*. Except for *H. pannosa*, however, none of the eight tested species was alarmed by crushed conspecifics, although slight aversion was evident in *M. minimus*.

We interpret these observations and test results as fitting Blum's (1985) correlation of alarm pheromones in arthropod taxa with a gregarious *modus vivendi*. Of the collembolans we observed, *H. pannosa* was by far the most gregarious. Even in sparse cultures they typically formed compact aggregations with frequent and prolonged physical contact between individuals. Their eggs are almost invariably deposited in dumps that can contain into the thousands. Butcher et al. (1971) noted that *H. nivicola* (Fitch) also lays eggs in batches or clumps. Pheromones mediating aggregation behavior have been noted in *H. viatica* Tullberg (Mertens and Bourgoignie 1975, 1977). Synchronized moulting in colonies of *H. lapponica* (Axelson) and *H. socialis* has been demonstrated to result from chemical communication among members, rather than control by external factors (Leinaas 1983). We anticipate that further studies of gregarious hypogastrurid species will reveal additional details of chemically mediated interactions between aggregation members. Particularly, in regard to alarm behavior, it would be valuable to study the responses to known collembolan predators such as mesostigmatid mites and carabid beetles, and to elucidate the chemistry of the alarm pheromones involved.

ACKNOWLEDGMENTS

We thank Richard J. Snider, Department of Zoology, Michigan State University, East Lansing for sharing his insights and kindly identifying all collembolans. Murray Blum, University of Georgia, Athens, Larry Phelan, and John M. Blair gave us helpful advice and encouragement. We greatly appreciate the bibliographic support of Connie J. Britton and the Library staff at the O.A.R.D.C., Wooster.
LITERATURE CITED


A REVIEW OF THE GENUS GRYLLUS (ORTHOPTERA: GRYLLIDAE), WITH A NEW SPECIES FROM KOREA

Richard D. Alexander

ABSTRACT

Gryllus is the most widely distributed genus of the Tribe Gryllini, and may be the largest; it includes 69 described species occupying most of the New World, Africa, and Europe, and much of Asia. A new species from Korea significantly extends the known range of the genus.

Recent Work on Gryllus

Chopard’s (1961, 1967) restrictions of the genus Gryllus Linnaeus (1758), based largely on male genitalia, reduced it from over 200 species to 42 nominal species, 32 in the New World, one Bermudan, six African and Mediterranean, one Madagascar, one European, and one, Gryllus bimaculatus, a flying species, widely distributed in southern Europe, Africa, Asia, and the Pacific, in some cases by human transport. Of these 42, nigra Harris and scudderianus Saussure are synonyms of pennsylvanicus Burmeister (Alexander 1957), comptus Walker (Brazil) is a Brachytrupinae (Alexander, unpubl. obs.), and afer Saussure is a Teleogryllus species (Otte and Cade, 1983). Chopard (1970) later described G. abnormis from St. Helena Island. Walker (1974) described G. ovisopus from Florida, and Weissman et al. (1980) added two western U.S. species, brevicaudus Weissman, Rentz, and Alexander, and cohni Weissman. Weissman et al. also synonymized determinatus Walker (Jamaica), contingens Walker (Brazil), and mundus Walker (Brazil). It is likely, however, that for these three type specimens—at least those from Brazil—nothing more can be said than that they belong to the assimilis group (my notes from the types in the British Museum, examined in 1963, say “probably = assimilis”). Undescribed species in this group, with distinctive songs, are known from Mexico (Alexander, unpubl.), and probably occur throughout Central and South America.


In terms of species numbers, Gryllus appears to thin out gradually eastward across the Asian continent. A. V. Popov (in corresp.) describes G. bimaculatus and G. campestris as sympatric in Azerbaijan and states that G. bimaculatus “occupies the whole Middle Asia from the Caspian to Tadjikstan”; G. bimaculatus is also

---

1Museum of Zoology and Department of Biology, University of Michigan, Ann Arbor, MI 48109.
known from China and tropical Asia (Chopard 1967). Chopard (1969) placed an Indian species *facialis* F. Walker (1871) in the genus *Gryllus*. In his (1967) world catalogue, however, written later but published prior to the Indian work, Chopard transferred this species to the genus *Modicogryllus*. Nevertheless, Chopard's (1969) illustration of the genitalic of *facialis* appears to represent a *Gryllus*. The specimen used for the genitalic drawing is not identified, nor is it indicated whether or not Chopard saw the types of either *facialis* or *Scapsipeded hastatus* Saussure, which he synonymized with *facialis* Walker. A second possible endemic *Gryllus* species from the same region is the Assam (India) species, *Lenigryllus quadrimaculatus* Saussure (Chopard 1967).

As known to the present, then, *Gryllus* includes approximately 68 species and is the most widely distributed and probably the largest genus in the Tribe Gryllini (Otte and Chopard unpubl.). It occurs throughout North, Central, and South America, on islands in the Atlantic (Bahamas, Bermuda, St. Helena), more or less throughout Africa and Europe, and extends eastward to the middle of the Asian continent. Except for *Gryllus bimaculatus*, however, the genus has been unknown in eastern Asia. Nor are endemic *Gryllus* species known on the Australian continent or in the Pacific, except along the west coast of the Americas on the Galapagos Islands (*galapageius* Scudder) and the Revilla Gigedo Islands (Clarion Island, a new species: Otte, pers. comm.). In eastern Asia and the Pacific what appears as the niche occupied by these large, usually shallowly burrowing field crickets is commonly filled by members of the African, Asian, and Australian genus *Teleogryllus*, which are quite similar in overall appearance and behavior but differ from *Gryllus* in song pattern, genitalic, karyotype, and mating behavior (Otte and Alexander 1967—pers. obs., Alexander and Otte 1967, Alexander 1962a,b; Lim, Vickery, and Kevan 1973).

On the basis of numbers of species, *Gryllus* might be thought to have originated in the New World; at least 15 undescribed species are known from North America alone (Alexander ms., unpubl.; Weisman, unpubl.; Alexander and Cade, ms.). Africa, however, contains many more genera similar to *Gryllus* (Chopard 1967, Otte and Cade 1984, Otte, pers. comm., Otte and Chopard, unpubl.).

*Gryllus nigrohirsutus*, new species

**Holotype male.** Body length, 17.55 mm.; tegmen, 7.488; hind femur, 9.5; cercus, broken; hind tibia, 6.76; head width, 3.9; pronotal width (widest), 4.75; pronotal length, 3.25.

**Male tegmina.** Three chords; three harp veins; diagonal vein with two cross-veins connecting it to Cu, anterior to mirror; mirror with a Y-shaped dividing vein; apical field much reduced; stridulatory vein length, 2.3 mm; with 94 teeth (fig. 2).

**Male genitalia.** As in figure 2 (compare to figs. 1–7 Alexander 1962a, fig. 2 Alexander and Otte 1967, Lam. II Chopard, 1961); epiphallus apparently a little thicker and more blunt than those of American and European *Gryllus* species.

**Allotype female.** Body length, 16.25 mm.; ovipositor, 11.7; tegmen, 4.5; hind femur, 10.4; cercus (right), 7.15; hind tibia, 6.84; head width, 4.49; pronotal width, 5.2; pronotal length, 3.5.

**Color.** Solid black in both sexes, with spurs on hind tibiae brownish; tegmina and ovipositor with some brownish areas.

**Armature of hind tibiae.** Eight spurs on each margin of each tibia of both sexes, including three apical, second apical (center) spur longest of apical spurs.

**Tympana.** Inner and outer on each front tibia in both sexes, both oval, inner one much smaller and, in the holotype male, with a smaller projection extending partway across its diameter from central part of rear margin; male: outer, 0.653 x 0.223 mm.; inner, 0.373 x 0.155; female: outer, 0.808 x 0.290; inner, 0.180 x 0.077.

**Type Data.** Holotype male and allotype female: Korea, Central National Forest,
Figure 1a-b. Gryllus nigrohirtus, n. sp.: a—holotype male; b—allotype female.
Figure 2a-d. Gryllus nigrohirsutus, n. sp.: a-c, genitalia of holotype male; a - dorsal view, b - lateral view, c - ventral view; d - stridulum of holotype male.


This small, black, pubescent, micropterous Gryllus was taken in the Central National Forest 16 mi northeast of Seoul, Korea (two mi west of Pupyongi), by Dr. George Byers, now of the University of Kansas. The male genitalia (fig. 2) place it in the genus Gryllus, thus very far outside the presently known range of micropterous forms in this evidently ancient genus.

The resemblance of G. nigrohirsutus to the American wood cricket, G. vernalis Blatchley (Blatchley 1920, Alexander 1957), is superficially close, although G. nigrohirsutus is considerably more pubescent and distinctive in appearance, owing partly to a slightly flattened shape and partly to the venation of the male tegmen. The general appearance of G. nigrohirsutus, and its micropterousness, are consistent with a life in leaf litter, like that of G. vernalis. Byers' field notes (pers. comm.) indicate a flat, fairly open hardwood forest with considerable leaf litter and occasional patches of grass. No Gryllus species can be identified as a close relative.
DISCUSSION

This species extends the known range of *Gryllus* significantly on the Asian continent, suggesting that historically the genus has occurred essentially everywhere within the range of the Gryllidae except in southeast Asia, on Pacific islands, and in Australia.

ACKNOWLEDGMENTS

I thank Dan Otte for providing access to Otte and Chopard (unpublished), and for assisting with every aspect of the manuscript.

LITERATURE CITED


——. (in prep.). A review of recent systematic and genetic work on the *Gryllus firmus* complex (Orthoptera: Gryllidae).


AN ANNOTATED CHECKLIST OF THE LEPIDOPTERA OF THE BEAVER ISLAND ARCHIPELAGO, LAKE MICHIGAN.

Dennis Profant

ABSTRACT

A survey of Lepidoptera was conducted in 1987 and 1988 on Beaver Island, Lake Michigan. When combined with a 1930 survey of the Beaver Island Archipelago, 757 species from 41 families have now been recorded from these islands.

Only one study has been published on the Lepidoptera of Beaver Island and the surrounding islands of Garden, High, Hog, Whiskey, Squaw, Trout, Gull, and Hat (Moore 1930). The present study has produced a more complete inventory of lepidopteran species on Beaver Island. Collecting was done in a variety of habitats using several different light sources. Emphasis was placed on the Microlepidoptera which were largely omitted in the original work by Moore (1930).

Beaver Island is the largest of a group of nine named islands located in the extreme northeastern portion of Lake Michigan, (Fig. 1). Situated 51 km northwest of Charlevoix, Michigan, the island is 20 km long by 11 km wide and consists of several hundred permanent residents who reside primarily in the town of St. James. The island has a maritime climate and frosts can occur well into May and again in September. While the summer air is usually hot during the day, 26°C, it often is very cool at night. 10°C is not unusual in June, July, and August, (Jacque LaFreniere, pers. comm.).

The northern part of the island is underlain with limestone formations which form outcrops in various places. The interior is gently rolling, corresponding to the original glacial drift. Behind the shoreline are a series of sand dunes and terraces, the largest of which occurs on the west side of the island. The highest point in this dune complex is approximately 61 m high (Darlington 1940). It is probable that the Beaver archipelago has not been connected to the mainland since the last glacial epoch. The fauna must have crossed the water after the retreat of the Wisconsin ice sheet some 15,000 years ago. The water would be only a slight barrier for winged insects, especially Lepidoptera. The species found on the island have a marked northern affinity and may have been carried from the Upper Peninsula by the frequent strong northwest winds (Moore 1930).

Beaver Island occurs in the hemlock-white pine-northern hardwoods forest region (Braun 1950). The beaches and dunes consist of vegetation characteristic of this latitude. Much of the island perimeter consists of coniferous forests (mixed fir, spruce, cedar, hemlock, and pine). A number of inland lakes contain sphagnum bogs or bog forests. Years ago the island residents put much of the land into cultivation and many of these farmlands have reverted to old fields and second growth forests of paper birch (*Betula papyrifera*) and a bracken fern (*Pteridium aquilinum*) understory.
MATERIALS AND METHODS

Lepidoptera were collected between July and September 1987, and periodically between May and August 1988. A few records were obtained from a small teaching collection at the Central Michigan University (CMU) Biological Station. Most species in table 1 without collecting dates came from this collection. White sheets illuminated by a 200 watt incandescent light bulb and/or several filtered black lights (BLB) were used for night collecting. Buildings with mercury vapor lights were also utilized. Sugaring for moths with a beer-molasses bait proved unproductive. Moths were collected 4-5 times a week and at all hours of the night.

Habitats sampled on the island included hemlock woods, beech-maple forest, sphagnum bog, cedar woods, northern hardwoods, coniferous forest, lake edges, marshes, and open fields. The primary collection sites responsible for the largest percentage of species collected were the CMU Biological Station and campground.
The beach and dunes at the station were typified by *Prunus pennsylvanica*, *Populus balsamifera*, *Salix cordata*, *Potentilla fruticosa*, *Juniper spp.*, *Arctostaphylos uva-ursi*, *Campanula rotundifolia*, *Lithospermum caroliniense*, and *Zigadenus glaucus*. Behind the dunes was an area dominated by *Pinus resinosa*, and *P. strobus*, with an understory of bracken fern, *Smilacina stellata*, *Linnaea borealis*, and *Silene vulgaris*. The campground was a mixed hardwood-coniferous community. Common plants included paper birch, *Tsuga canadensis*, *Thuja occidentalis*, *Populus tremuloides*, *Abies balsamea*, *Picea glauca*, *Ostrya virginiana*, *Betula alleghaniensis*, *Cornus canadensis*, *Clintonia borealis*, *Maianthemum canadense*, and *Polygala paucifolia*.


**RESULTS**

A total of 757 lepidopteran species from 41 families has now been recorded from the Beaver Island archipelago. 701 of these species were from Beaver Island; 517 species were collected in this study and 184 additional species were reported by Moore (1930). Approximately 35 species of microlepidoptera remain unidentified. Table 1 lists all identified species collected from the Beaver Island archipelago. Species not collected on Beaver Island are listed with the island on which they were originally recorded by Moore (1930).

Table 1. An annotated checklist of the Lepidoptera of the Beaver Island archipelago, Lake Michigan. M, recorded by Moore (1930); P, recorded by Profant.

<table>
<thead>
<tr>
<th>FAMILY</th>
<th>SPECIES</th>
<th>STATUS</th>
<th>COLLECTION</th>
</tr>
</thead>
<tbody>
<tr>
<td>HEPIALIDAE</td>
<td><em>Monopis dorsistrigella</em> (Clem.)</td>
<td>P 29 July</td>
<td></td>
</tr>
<tr>
<td>Sthenopis quadriguttatus (Grt.)</td>
<td>P July</td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Hepialus gracilis</em> Grt.</td>
<td>P 12 July</td>
<td></td>
<td></td>
</tr>
<tr>
<td>OPOSTEGIDAE</td>
<td><em>Lyonetiidae</em></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Opostega quadristrigella Cham.</td>
<td>P 22-29 June</td>
<td></td>
<td></td>
</tr>
<tr>
<td>INCURVARIIDAE</td>
<td><em>Gracillariidae</em></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Chalceopla cyanella (Bk.)</td>
<td>P 27 May</td>
<td></td>
<td></td>
</tr>
<tr>
<td>Adela ridingella Clem.</td>
<td>M July</td>
<td></td>
<td></td>
</tr>
<tr>
<td>TINEIDAE</td>
<td><em>Heliozelidae</em></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Antispila sp.</td>
<td>P 4 July</td>
<td></td>
<td></td>
</tr>
<tr>
<td>Nemapogon sp.</td>
<td>P 22 July</td>
<td></td>
<td></td>
</tr>
<tr>
<td>Amydria effrentella Clem.</td>
<td>P 21 July</td>
<td></td>
<td></td>
</tr>
<tr>
<td>TINEIDAE</td>
<td><em>Oecophoridae</em></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Agonopterix atrodorsella (Clem.)</td>
<td>P 27 May</td>
<td></td>
<td></td>
</tr>
<tr>
<td>Agonopterix pulvipersella (Clem.)</td>
<td>P 26 May</td>
<td></td>
<td></td>
</tr>
<tr>
<td>Agonopterix flavicomella (Engel)</td>
<td>P 9 July</td>
<td></td>
<td></td>
</tr>
<tr>
<td>Semioscopis packardella (Clem.)</td>
<td>P May</td>
<td></td>
<td></td>
</tr>
<tr>
<td>Depressaria pastinacella (Dup.)</td>
<td>M June-Aug. Garden, High</td>
<td></td>
<td></td>
</tr>
</tbody>
</table>
Ethmia monticola fuscopedella (Wlsm.) P 22 June
Callima argenticinctella Clem. P July
BLASTOBASIDAE
Dryoperia mursfeldiella (Cham.) P 3 Aug.
COLEOPHORIDAE
Coleophora fagicorticella ? Cham. P 4 July
Coleophora spissicomis (Haw.) P 4 July
GELECHIIDAE
Dichomeris ligulella Hbn. P 29 July
Dichomeris serrafivittellalisa complex P 7 July
Anacampsis niveopulvella (Cham.) P 6 Aug.
Trichotaphe flavocostella (Clem.) P 21 July
ALUCITIDAE
Alucita hexadactyla L. P 7 Aug.
PLUTELLIDAE
Plutella porrectella ? (L.) P 26 May
Plutella xylostella (L.) P 3 Aug.
Ypsolopha dentiferella (Wism.) P 2 Aug.
YPONOMEUTIDAE
Atteva punctella (Cram.) P 21 July
Swammerdamia sp. P 9 July
ARGYRESTHIIDAE
Argyresthia annettella Bsk. P July
Argyresthia prob. austerella Zell. P 1 July
Argyresthia oreasella Clem. P 29 July
Argyresthia sp. P 1 July
SESIIIDAE
Pennisetia marginata (Harr.) M Aug.
Albuna pyramidalis (Wlk.) M June-July High
Synanthedon pictipes (Grote & Robinson) M 5 July High
Synanthedon acerni (Clem.) MP July-Aug.
Synanthedon proxima (Hy. Edw.) P July
CHOREUTIDAE
Prochoreutis inflatella (Clem.) P July-Aug.
Choreutis pariana (Cl.) P 29 July
COSSIDAE
Acossus centerensis (Lint.) M July-Aug.
Prionoxystus robiniae (Peck) P 1 July
Prionoxystus macmurtrei (Guer.) P 29 June
TORTRICIDAE
Aterpla approximana (Heinr.) P July-Aug.
Apotomis capreaana (Hbn.) M Sept.
Apotomis funerea (Meyr.) P June-Aug.
Apotomis apateticana (McD.) P 18 July
Apotomis deceptana (Kft.) P July-Aug.
Pseudosciaphila duplex (Wlsm.) P 27 June
Olethreutes punctana (Wlsm.) P 9 July
Olethreutes quadridifus (Zell.) P July
Olethreutes clavana (Walker) P 9 July
Olethreutes nigrana (Heinr.) P 29 July
Olethreutes merrickana ? (Kft.) P 18 July
Olethreutes cespitana (Hbn.) MP July-Aug.
Olethreutes bipartitana (Clem.) P 4 July
Hedyia ochroleucana (Frolich) P 7 July
Rhyacionia buoliana (Denis & Schiffermuller) P July
Spilonota ocelana (Denis & Schiffermuller) P 29 July
Phaneta formosana (Clem.) P 7 July
Phaneta ochrocepha (Wlsm.) P July-Aug.
Phaneta ochrosternina (Kft.) P Aug.
Phaneta parmatana (Clem.) P 2 Aug.
Phaneta tarandana ? (Mosch.) P July-Aug.
Phaneta prob. olivacea (Riley) P 4 July
Eucosma tocullionana Heinr. P 7 July
Eucosma palabunda Heinr. P June-July
Eucosma dorsissignata (Clem.) MP Aug.-Sept.
Eucosma derelecta Heinr. P 21 July
Eucosma consobrina Heinr. P 29 July
Epiblema oitosana (Clem.) P 7 July
Notocelia culminana (Wlsm.) P 3 Aug.
Gypsonoma haimbachiana (Kft.) P 29 July
Gypsonoma substitutionis Heinr. P June-July
Gypsonoma adjecta Heinr. P 1-4 July
Proteoiras moffittiana Fern. P 29 July
Epinotia solandriana (L.) P July-Aug.
Epinotia huroniens Brown P 22 July
Epinotia criddleana ? (Kft.) P 18 July
Epinotia transmissana (Walker) P 29 July
Ancyis metalmelana (Wlk.) P 7 July
Ancyis semiolana (Zell.) P 1 July
Ancyis burgessiana (Zell.) MP May-June
Dichrorampha bittana (Bsk.) P 7 July
Pammene feliciana Heinr. P 7 July
Grapholita pruniwora (Walsh) P 29 July
Cydia flexifoqua (Heinr.) P July
Cydia pomonella (L.) P 29 July
<table>
<thead>
<tr>
<th>Taxonomy</th>
<th>Host Plant</th>
<th>Month</th>
</tr>
</thead>
<tbody>
<tr>
<td>Croesia albicomana (Clem.)</td>
<td>MP</td>
<td>Aug.</td>
</tr>
<tr>
<td>Acleris nivisellana (Wlsm.)</td>
<td>P</td>
<td>22 July</td>
</tr>
<tr>
<td>Pandemis lamprosana (Rob.)</td>
<td>P</td>
<td>4 July</td>
</tr>
<tr>
<td>Pandemis limitata (Rob.)</td>
<td>P</td>
<td>July</td>
</tr>
<tr>
<td>Pandemis canadana Kft.</td>
<td>P</td>
<td>18-29 July</td>
</tr>
<tr>
<td>Argyrotaenia velutinana (Wlk.)</td>
<td>P</td>
<td>July</td>
</tr>
<tr>
<td>Pandemis rosacea (Harr.)</td>
<td>P</td>
<td>28 July</td>
</tr>
<tr>
<td>Pandemis limpitata</td>
<td>P</td>
<td>July</td>
</tr>
<tr>
<td>Pandemis canadana Kft.</td>
<td>P</td>
<td>July</td>
</tr>
<tr>
<td>Argyrotaenia quadridasciana</td>
<td>P</td>
<td>July</td>
</tr>
<tr>
<td>P. packardiana (Fern.)</td>
<td>P</td>
<td>July-Aug.</td>
</tr>
<tr>
<td>Clepsis pemcana (Fitch)</td>
<td>P</td>
<td>July</td>
</tr>
<tr>
<td>Clepsis meialeueana (Wlk.)</td>
<td>MP</td>
<td>July-Aug.</td>
</tr>
<tr>
<td>Ptycholoma virescana (Clem.)</td>
<td>P</td>
<td>4 July</td>
</tr>
<tr>
<td>Sparganothis suljureana</td>
<td>P</td>
<td>21 July</td>
</tr>
<tr>
<td>Sparganothis reticulatana</td>
<td>MP</td>
<td>Aug.</td>
</tr>
<tr>
<td>Sparganothis directana</td>
<td>P</td>
<td>July</td>
</tr>
<tr>
<td>Sparganothis pettitana</td>
<td>MP</td>
<td>July</td>
</tr>
<tr>
<td>COCHYLIDAE</td>
<td></td>
<td></td>
</tr>
<tr>
<td>Hysterosia hospe Wlsm.</td>
<td>P</td>
<td>18 July</td>
</tr>
<tr>
<td>Hysterosia romonana Kft.</td>
<td>P</td>
<td>July</td>
</tr>
<tr>
<td>HESPERIIDAE</td>
<td></td>
<td></td>
</tr>
<tr>
<td>Erynnis icelus (Scudder &amp; Burgess)</td>
<td>M</td>
<td>May-Aug.</td>
</tr>
<tr>
<td>Carterocephalus palaemon (Pallas)</td>
<td>M</td>
<td>18 June</td>
</tr>
<tr>
<td>Hesperia comta laurentina (Lyman)</td>
<td>MP</td>
<td>July-Aug.</td>
</tr>
<tr>
<td>Hesperia leonardus Harr.</td>
<td>P</td>
<td></td>
</tr>
<tr>
<td>Hesperia sussacca Harr.</td>
<td>M</td>
<td>18 June</td>
</tr>
<tr>
<td>Polites coras (Cram.)</td>
<td>P</td>
<td></td>
</tr>
<tr>
<td>Polites isimostolo (Latr.)</td>
<td>M</td>
<td>June-July,</td>
</tr>
<tr>
<td>Amblyscirtes white (Edw.)</td>
<td>M</td>
<td>July</td>
</tr>
<tr>
<td>PAPILIONIDAE</td>
<td></td>
<td></td>
</tr>
<tr>
<td>Papilio polyxenes F. P</td>
<td></td>
<td></td>
</tr>
<tr>
<td>Papilio glaucus L. MP</td>
<td></td>
<td></td>
</tr>
<tr>
<td>PIERIDAE</td>
<td></td>
<td></td>
</tr>
<tr>
<td>Argea napi (L.) M</td>
<td>P</td>
<td>May-June Garden, Hog</td>
</tr>
<tr>
<td>Argea rapae (L.) MP</td>
<td></td>
<td>May-Sept.</td>
</tr>
<tr>
<td>Colias philodice Godt. MP</td>
<td></td>
<td>May-Oct.</td>
</tr>
<tr>
<td>Colias interior Scudder P</td>
<td></td>
<td>2 July</td>
</tr>
<tr>
<td>Eurema leda Bdv. &amp; Leconte MP</td>
<td>P</td>
<td>July-Aug.</td>
</tr>
<tr>
<td>LYCAENIDAE</td>
<td></td>
<td></td>
</tr>
<tr>
<td>Lycaena phlaeas (L.) MP</td>
<td></td>
<td>May-Aug.</td>
</tr>
<tr>
<td>Hylophila phlanaus (Cram.)</td>
<td>MP</td>
<td>28 June High</td>
</tr>
<tr>
<td>Epidemia epixanthe (Bdv. &amp; Leconte)</td>
<td>P</td>
<td>June-July</td>
</tr>
<tr>
<td>Epidemia dorcas (Kby.) P</td>
<td></td>
<td>July-Aug.</td>
</tr>
<tr>
<td>Epidemia helioidea (Bdv.)</td>
<td>M</td>
<td>June-Aug.</td>
</tr>
<tr>
<td>Satyrium cancer (Edw.) P</td>
<td></td>
<td>20 July</td>
</tr>
<tr>
<td>Satyrium calanus fauler (Godart)</td>
<td>M</td>
<td>June-July</td>
</tr>
<tr>
<td>Satyrium liprups strigosum (Harr.)</td>
<td>MP</td>
<td>27 July</td>
</tr>
<tr>
<td>Incisalia augustus (Kby.) MP</td>
<td>M</td>
<td>July-Aug.</td>
</tr>
<tr>
<td>Strymon melinus Hbn. M</td>
<td>29 July Squaw</td>
<td></td>
</tr>
<tr>
<td>Everes comytas (Godt.) P</td>
<td></td>
<td>4 Aug.</td>
</tr>
<tr>
<td>Celastrina ladon (Cram.) MP</td>
<td></td>
<td>May-Aug.</td>
</tr>
<tr>
<td>Plebejus saepiolus (Bdv.) M</td>
<td></td>
<td>June-July Garden, Hog, High</td>
</tr>
<tr>
<td>NYMPHALIDAE</td>
<td></td>
<td></td>
</tr>
<tr>
<td>Polygonia interrogationis (F.)</td>
<td>P</td>
<td>June</td>
</tr>
<tr>
<td>Polygonia comta Harr. MP</td>
<td></td>
<td>July</td>
</tr>
<tr>
<td>Polygonia faunus (Edw.) M</td>
<td>20 July Squaw</td>
<td></td>
</tr>
<tr>
<td>Polygonia progne (Cram.) MP</td>
<td></td>
<td>July-Sept.</td>
</tr>
<tr>
<td>Nymphalis va-album (Denis &amp; Schiffermuller)</td>
<td>MP</td>
<td>Aug.</td>
</tr>
<tr>
<td>Nymphalis antiocha (L.) MP</td>
<td></td>
<td>June-Oct.</td>
</tr>
<tr>
<td>Aglais milberti (Godt.) MP</td>
<td></td>
<td>June-July</td>
</tr>
<tr>
<td>Vanessa virginienis (Drury)</td>
<td>MP</td>
<td>June-July</td>
</tr>
<tr>
<td>Vanessa cardui (L.) MP</td>
<td></td>
<td>May-Sept.</td>
</tr>
<tr>
<td>Vanessa atalanta (L.) MP</td>
<td></td>
<td>June-Aug.</td>
</tr>
<tr>
<td>Junonia coenia (Hbn.) MP</td>
<td></td>
<td>Aug.-Sept.</td>
</tr>
<tr>
<td>Speyeria cybelle (F.) MP</td>
<td></td>
<td>July</td>
</tr>
<tr>
<td>Speyeria aphrodite alccestis (Edw.) MP</td>
<td>July</td>
<td></td>
</tr>
<tr>
<td>Speyeria alicia (Edw.) MP</td>
<td></td>
<td>July-Aug.</td>
</tr>
<tr>
<td>Clossiana sebina myrina (Cram.)</td>
<td>MP</td>
<td>June-Aug.</td>
</tr>
<tr>
<td>Clossiana bellina (F.) P</td>
<td></td>
<td>6 Aug.</td>
</tr>
<tr>
<td>Phyciodes morpheus (F.) MP</td>
<td></td>
<td>June-Aug.</td>
</tr>
<tr>
<td>Charidryas nyctea (Doubleday) M</td>
<td>June-High</td>
<td></td>
</tr>
<tr>
<td>Basilarchia arthemis (Drury) &amp; astynax (F.) MP</td>
<td>June-Aug.</td>
<td></td>
</tr>
</tbody>
</table>
Basilarchia archippus (Cram.) MP Aug.

SATYRIDAE

Enodia anthedon A.H. Clark P 7 July
Satyrodes eurydice (Johansson) MP July-Aug.
Megisto cymela (Cram.) P 5 July
Coenonympha inornata Edw. MP July
Cercyonis pega nephele (Kby.) MP July-Aug.

DANAIDAE

Danaus plexippus (L.) MP Aug.

LIMACODIDAE

Tortricidia testacea Pack. MP June-July
Tortricidia prob. jlexuosa (Grt.) P 4 July
Lithacodes jasciola (HAl.) MP July-Aug.

PYRALIDAE

Scoparia basalis Wlk. P 29 July
Eudonia strigalis (Dyar) P 22 July
Nymphula ekthipitis (Grt.) M 18 July High
Munroessa icciualis (Wlk.) MP July-Aug.
Parapoyx maculalis (Clem.) MP Aug.
Parapoyx badiusalis (Wlk.) P Aug.-Sept.
Parapoyx allionesalis Wlk. P 29 July
Dicytomolonia julianalis (Wlk.) P 29 July
Evergestis pallidata (Hufn.) M June-Sept.
Perispa caucalitis Zell. P July
Phylaenia coronata (Hufn.) M July-Aug.
Nealgedonia extricalis (Gn.) M 10 Sept.
Anania fanebris (Strom) M June-July
Sitochroa chortalis (Gn.) M June
Loxostege sticticalis (L.) MP May-June
Pyrausta nicalis (Gn.) M 25 June High
Pyrausta signalinis (Wlk.) M 2 Aug. High
Pyrausta bicoloralis (Gn.) P July
Pyrausta acrionalis (Wlk.) P 21 July
Pyrausta fodiaulis (Led.) M 30 July Garden
Udea rubigalis (Gn.) MP June-Oct.
Diathrausta reconditalis (Wlk.) M 3 Aug.
Desmia funerals (Hbn.) P
Anageshna primordialis (Dyar) P June-July
Palpita magniferalis (Wlk.) P May-June
Herpetogramma pertextalis (Led.) M Aug.-Sept.
High

PTEROPHORIDAE

Geina tenuidactyla (Fitch) MP July
Cnaemidophorus rhododactylus (Denis & Schiffermuller) P 22 July
Platyptilia pallidactyla (Haw.) M June
Platyptilia cardiacactyla (Riley) P June-Aug.
Oidaematophorus eliotii (Fern.) P 2 Aug.
Oidaematophorus kellicottii (Fish) M June High
Oidaematophorus inconditus ? (Wlsm.) M June High
Emmelina monodactyla (L.) M June-July High

THYATIRIDAE

Habrosyne scripta (Gosse) MP June-July
Habrosyne gloriosa (Gn.) M Hog from pupae
Pseudothylitura cymatophoroides (Gn.) M June-July
Euthyatra pudens (Gn.) M 7 June High
DREPANIDAE

*Drepanta arcuata* Wik. MP July-Aug.

*Drepanta bilineata* (Pack.) MP July

*Oreta rosea* (Wik.) P 4 July

GEOMETRIDAE

*Protolyme virginalis* (Hulst) P 2 Aug.

*Itame pustularia* (Gn.) MP June-Sept.

*Itame ribearia* (Fitch) M 6 Aug.

*Itame brunnnea* (Thund.) M July

*Itame subcessaria* (Wik.) P 21 July

*Itame bitactata* (Wik.) M July-Aug.

*Semiothisa aemulataria* (Wik.) M June-July

*Semiothisa minorata* (Pack.) MP June-July

*Semiothisa angustioria* (Gn.) MP June-Aug.

*Semiothisa eremiasia* (Gn.) M 18 July High

*Semiothisa neptaria trijasciata* (Pack.) M June-July Squaw, Hog

*Ematurga amitaria* (Pack.) P 2 July

*Anacamptodes velivaria* (Hulst) P 27 May

*Anacemethia larvaria* (Gn.) MP July-Aug.

*Anavitrelia pampinaria* (Gn.) M June-July High

*Ectropis crepuscularia* (Denis & Schiffermuller) M May-July

*Protoboaemlia porcellaria indicataria* (Wik.) M July-Sept.

*Melanolophia signataria* (Wik.) MP May-June

*Eufidonia discospilata* (Wik.) P May-July

*Biston betularia cognataria* (Gn.) MP July-Aug.

*Hypagyrtis unipunctata* (Haw.) M July-Aug.

*Hypagyrtis piniaria* (Pack.) P June-July

*Erannis illaria* (Harr.) M 6 Oct.

*Lomographa semiclararia* (Wik.) M May-June

*Lomographa vestalitaria* (Gn.) M June-July High

*Cabera erythemaria* Gn. MP June-Aug.

*Cabera varioloria* Gn. M June-July

*Euchlaena serrata* (Drury) MP July

*Euchlaena obtusaria* (Hbn.) P 4 July

*Euchlaena effecta* (Wik.) M July-Aug.

*Euchlaena johnsonaria* (Fitch) MP July

*Euchlaena amoenaestria* astylaria (Wik.) MP July

*Euchlaena marginaria* (Minot) M 4 June

*Euchlaena irriaria* (Barnes & McDunnough) MP July

*Euchlaena effecta* (Wik.) P 27 June

*Xanthotype sospeta* (Drury) M July

*Pero honestaria* (Wik.) MP June-July

*Pero morrisonaria* (Hy. Edw.) P 4 July

*Campaea perlata* (Gn.) P June-July

*Ennomos magnumaria* Gn. MP Aug.-Sept.

*Ennomos subsignaria* (Hbn.) M July-Aug.

*Petrophora subaequaria* (Wik.) MP May-July

*Homoeclides fritillaria* (Gn.) MP June-July

*Seleina kentaria* (Grote & Robinson) P May 27

*Melanema inatomaria* Gn. MP June-Aug.

*Melanema determinata* Wik. M Aug. 6

*Metarranthis duaria* (Gn.) MP June

*Metarranthis hypochoria* (Herrich-Schaffer) M June Garden, High

*Cephis armatoria* (H. & S.) MP June-July

*Anagoga occiduaria* (Wik.) M June

*Probole amicaria* (Herrich-Schaffer) MP May-July

*Plagodis serinaria* Herrich-Schaffer P 28 May

*Plagodis phlogosaria approximaria* Dyar MP May-July

*Caripeta divisata* Wik. MP June-Aug.

*Caripeta piniaria* (Pack.) P 27 June

*Caripeta angustioria* Wik. MP June-Aug.

*Besma quercivoraria* (Gn.) MP July

*Lambdana fiscellaria* (Gn.) MP Aug.-Oct.

*Cingilia catenaria* (Drury) M Sept.

*Nepytia canosaria* (Wik.) P Aug.

*Nepytia semicrusaria* (Wik.) M Aug.-Sept.

*Sicya macularia* (Harr.) MP Aug.

*Eusarca confusaria* Hbn. MP July

*Tetracis crocallata* Gn. MP July

*Tetracis cachexiata* Gn. M June-July

*Eutrapela clemataria* (J.E. Smith) MP May-June


*Nematocampa limbata* (Haw.) M July-Aug.

*Nemoria mimosaria* (Gn.) MP May-July

*Synchlora aerata* (F.) M July

*Chlorochlamys chloroctoria* (Gn.) M June-Aug.

*Hethemia pistasciaria* (Gn.) MP May-June

*Mesothea incertaria* (Wik.) M 25 May Hog

*Idea demissaria* (Hbn.) P 9 July

*Pleuropana insularia* (Gn.) M 4 Sept.

*Cyclophora pendulinaria* (Gn.) M June-Aug.

*Scopula limbounata* (Haw.) MP July-Aug.

*Scopula junctaria* (Wik.) M July 12 High

* Dysstroma truncata* (Hufn.) MP Aug.-Sept.

*Dysstroma hersiliata* (Gn.) M July

*Eulithis diversilineata* (Hbn.) M July-Sept.

*Eulithis testata* (L.) MP Aug.-Sept.

*Eulithis molliculata* (Wik.) P 6 Aug.

*Eulithis explanata* (Wik.) MP Aug.
Ecliptopera silacea albolineata (Pack.) M June-Sept.
Thera contractata (Pack.) MP July-Sept.
Hydriomena californiata Pack. M June-July
Hydriomena coerulea Fehr. M June-July Hog
Tripbosa haesitata affinartia (Wlk.) MP May-Aug.
Coryphista meadii (Pack.) P 6 Aug.
Hydria undulata (L.) MP June-July
Rheumaptera hastata (L.) MP June-July Hog, Garden, High
Mesoleuca ruicjillata (Gn.) MP June-July
Spargania magnoliata Gn. MP June-Sept.
Perizoma basaliata (Wlk.) M Aug.
Anticlea vassiliata Gn. P 24 May
Anticlea multijerata (Wlk.) M 10 July
Xanthorhoe labradorescens (Pack.) M June-Aug.
Xanthorhoe iduata (Gn.) M July
Xanthorhoe muniata convallaria (Gn.) M 16 July
Xanthorhoe lacustra (Gn.) M 7 July
Xanthorhoe inornata (Hulst) P 4-7 July
Trichodesia albovittata (Gn.) MP May-July
Eubaphe mendica (Wlk.) MP June-Aug.
Horisme intestinata (Gn.) MP June-Aug.
Eupithecia palpata Pack. M 1 July
Eupithecia subfuscata (Haw.) M June-July
Eupithecia satryata fumata Tayl. M 18 July High
Eupithecia coagulata Gn. MP May & Aug.
Eupithecia antecaria Wlk. M 7 July
Eupithecia ravoscatalisata Pack. P 28 May
Cladara limitalia (Wlk.) P May
Lobophora nivigerata Wlk. P July
Lobophora montanata Pack. M June-July
EPIPLEMIDAE
Callizia amorata Pack. MP June-July
LASIOCAMPIDAE
Tolype laricis (Fitch) MP Aug.-Sept.
Phylloidesma americana (Harr.) MP June
Malacosoma disstria Hbn. MP July
Malacosoma americanum (F.) MP July-Aug.
SATURNIIDAE
Dryocampa rubicunda (F.) MP June-July
Automeris io (F.) P
Antheraea polyphemus (Cram.) MP June-July
Actias luna (L.) MP June-July
Hyalaphora cecropia (L.) P
SPHINGIDAE
Manula quinquemaculata (Haz.) P
Ceratomia amyntor (Geyer) MP July
Ceratomia undulosa (Okl.) MP June-Aug.
Sphinx chersis (Hbn.) MP June-Aug.
Sphinx kalmiae J.E. Smith MP Aug.
Sphinx gordius Cram. P 27 June
Sphinx drupiferarum J.E. Smith P 6 July
Lapara bombycoides Wlk MP June-July
Smerinthus jalamecensis (Drury) MP June
Smerinthus cerisyi Kby. MP June-July
Paonia exacetus (J.E. Smith) MP June-July
Paonia myops (J.E. Smith) P 1 July
Laothoe juglandis (J.E. Smith) MP June-July
Pachysphinx modesta (Harr.) MP July-Aug.
Hemaris thysbe (F.) M 24 July High
Hemaris diffinis (Bed.) M 2 June High
Hyles gallii (Rottemburg) P
NOTODONTIDAE
Clostera albosigma Fitch MP June-Aug.
Clostera strigosa (Grt.) M July
Clostera apicalis (Wlk.) M June-July
Datana ministra (Drury) MP July
Nadata gibbosa (J.E. Smith) MP July
Peridea basiriens (Wlk.) M July
Peridea angulosa (J.E. Smith) MP July-Aug.
Peridea farraginea (Pack.) MP June-Aug.
Pheosia rimoso Pack. MP July-Aug.
Odontasia elegans (Skr.) M July
Notodonta scitipennis Wlk. M June-July
Notodonta simplaria Graef P 1-6 Aug.
Nerice bidentata Wlk. P 27 June
Gluphisia septentrionis Wlk. MP June-Aug.
Gluphisia avimacula Hudson P May
Furcula cinerea (Wlk.) MP July-Aug.
Furcula scolopendrina (Bdv.) M 12 Aug.
Furcula modesta (Hudson) P May-Aug.
Furcula occidentalis (Lint.) P 4 July
Symmerista albifrons (J.E. Smith) MP June-July
Dasylophia thyatiroidea (Wlk.) MP June-Aug.
Heterocampa umbra Wlk. MP June-July
Heterocampa guttivata (Wlk.) P 29 June
Heterocampa biundata M June-July
Lochmaeus manteo Doubleday M 21 July Hog
Schizura ipomoeae Doubleday M 5 Aug.
Schizura unicornis (J.E.Smith) MP June-Aug.
Schizura apicalis (Grote & Robinson) M 16 July High
Schizura concinna (J.E.Smith) M reared
Oligocentria semirujescens (Wlk.) P 18 July
Oligocentria lignicolor (Wlk.) MP July
Hyparpax aurora (J.E.Smith) P June

ARCTIIDAE

Eilema bicolor (Grt.) MP Aug.
Crambida casta (Pack.) MP Aug.-Sept.
Lycomorpha pholus (Drury) P July
Hyponypria miniata (Kby.) MP July-Aug.
Clemensia alibata Pack. MP Aug.
Uetheisa bella (L.) M July-Aug.
Haploa confusa (Lyman) MP July-Aug.
Holometina laeta (Guer.-Menemille) MP July
Holometina aurantiaca (Hbn.) MP July-Aug.
Pyrharctia aurora (J.E.Smith) M 10 July
Spilosoma virginica (F.) MP June-Aug.
Arctica caja americana Harr. MP Aug.
Apanesthis phyllira (Drury) MP Aug.
Apanesthis parthenice (W.Kirby) MP Aug.-Sept.
Halysidota tessellaris (J.E.Smith) MP July-Aug.

LYMANTRIIDAE

Dasychira plagata (Wlk.) P 2-6 Aug.
Orgyia antiqua (L.) M 5 Sept.
Orgyia leucostigma (J.E.Smith) MP Aug.-Sept.

NOCTUIDAE

Idia americalis (Gn.) MP July-Sept.
Idia rotundalis (Wlk.) M July-Sept.
Idia scrobicollis (Grt.) M 28 July
Idia liberata (Gey.) M July-Aug.
Phalaenophana pyramusalis (Wlk.) M June-July
Zanclognatha theralis (Wlk.) M July-Aug.
Zanclognatha laevigata (Grt.) M 3 Aug. Garden
Zanclognatha cruralis (Gn.) M 20 June High

Chrysolina ochreipennis (Grt.) MP July-Aug.
Chrysolina moridallis (Gn.) M July High
Chrysolina petrelis (Grt.) M June-July
Hormista occiferalis Wlk. P July
Hormista absorptalis Wlk. P July
Phalaenostola larenioides Gt. P 29 July
Biepina caradrinalis Gt. M July
Renia iavigipunctalis (Gey.) MP July-Sept.
Palthis angulalis (Hbn.) M June-July Hog, High
Rivula propinqualis Gt. MP July-Aug.
Hypenodes caducus (Dyar) P 2 Aug.
Hypenodes fractilinea (Sm.) P July-Aug.
Dyspyralis illoca Warr. P 18 July
Bomolochi batmorialis (Gn.) MP July-Aug.
Bomolochi palparia (Wlk.) MP July
Bomolochi deceptalis (Wlk.) M 22 June High
Plathypena scabra (F.) MP June-Aug.
Spargaloma sexpunctata Gt. M 14 July
Pangrapta decoralis Hbn. M July
Metalacta quadrisignata (Wlk.) M June-Sept.
Alabama argillacea (Hbn.) M Sept.
Scoliopyrgus libatrix (L.) MP Aug.
Snyedoida adumbrata (Behr) M June-July
Zale luna (Drury) MP Sept.
Zale minerea (Gn.) P 27 May
Zale duplicata largera (Sm.) P May
Parallela bistriaria Hbn. M June
Euclidia cupida (Hbn.) M 27 May Hog
Caenurgina crassiuscula (Haw.) M Aug.
Caenurgina erechtes (Cram.) MP July-Aug.
Catocala antinympha (Hbn.) M 21 July Garden
Catocala ita (Cram.) MP Aug.
Catocala cerogama Gn. P July
Catocala relicta Wlk. MP Aug.-Sept.
Catocala unijuga Wlk. MP July-Sept.
Catocala briseis Edw. MP July-Aug.
Catocala concumbens Wlk. M 27 Sept. High
Catocala ultronia (Hbn.) MP Aug.-Sept.
Catocala grynea (Cram.) P 6 Aug.
Diachrysia aeroides (Grt.) M July-Aug.
Diachrysia baullina Gey. MP July
Autographa precationis (Gn.) MP May-Sept.
Autographa bipunctula (Steph.) MP July-Aug.
Autographa mappa (Grote & Robinson) P 4 July

1991 THE GREAT LAKES ENTOMOLOGIST 93
<table>
<thead>
<tr>
<th>Species</th>
<th>Appearance</th>
<th>Larvae</th>
<th>Host(s)</th>
<th>Notes</th>
</tr>
</thead>
<tbody>
<tr>
<td><em>Lacinipolia olivacea</em> (Morr.)</td>
<td>MP Aug.-Sept.</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Faromia diffusa</em> (Wlk.)</td>
<td>M June-Aug.</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Aletia oxygala</em> luteopallens (Sm.)</td>
<td>M July-Aug.</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Pseudaletia unipuncta</em> (Haw.)</td>
<td>MP May-Aug.</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Leucania commodoides</em> Gn.</td>
<td>MP July</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Leucania insueta</em> Gn.</td>
<td>M June</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Leucania pseudargyria</em> Gu.</td>
<td>MP 7 July</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Orthosia revieta</em> (Morr.)</td>
<td>P May</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Crocigrapha normani</em> (Grt.)</td>
<td>27 May</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Egira d%sa</em> (Grt.)</td>
<td>P 27 May</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Nephelodes minians</em> Gn.</td>
<td>MP Aug.-Sept.</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Homorthodes surgurata</em> (Grt.)</td>
<td>M 28 July</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Protorthodes incincta</em> (Morr.)</td>
<td>P Aug.</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Protorthodes ovilaca</em> (Gn.)</td>
<td>MP June-Sept.</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Ulolonche modesta</em> (Morr.)</td>
<td>MP June</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Orthodes crenulata</em> (Butler)</td>
<td>M 3 Aug.</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Orthodes cynica</em> Gn.</td>
<td>M June-July</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Tricholita signata</em> (Wlk.)</td>
<td>MP July-Aug.</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Agrotis vetusta</em> Wlk.</td>
<td>MP Aug.-Sept.</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Agrotis mollis</em> Wlk.</td>
<td>P 22 July</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Agrotis venerabilis</em> Wlk.</td>
<td>M 15 June</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Agrotis ipsilon</em> (Hufn.)</td>
<td>MP Aug.-Sept.</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Feltia jaculifera</em> (Gn.)</td>
<td>MP Aug.-Sept.</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Feltia subgothica</em> (Haw.)</td>
<td>MP Aug.</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Feltia herlits</em> (Grt.)</td>
<td>MP July-Aug.</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Feltia geniculata</em> Grote &amp; Robinson MP</td>
<td>Aug.-Sept.</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Euxoa diversens</em> (Wlk.)</td>
<td>M June-Aug.</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Euxoa sinelinea</em> Hdwk.</td>
<td>P 22 July</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Euxoa messoria</em> (Harr.)</td>
<td>M Aug.</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Euxoa scandens</em> (Riley)</td>
<td>M July-Aug.</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Euxoa pleuritica</em> (Grt.)</td>
<td>P 22 July</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Euxoa mimallonis</em> (Grt.)</td>
<td>MP Aug.</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Euxoa ochrogaster</em> (Gn.)</td>
<td>M 6 Aug.</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Euxoa tessellata</em> (Harr.)</td>
<td>M 9 July-3 Sept.</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Euxoa catenula</em> (Grt.)</td>
<td>M Aug.-Sept.</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Euxoa persevelens</em> (Grt.)</td>
<td>M Aug.</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Euxoa detersa</em> (Wlk.)</td>
<td>MP Aug.-Sept.</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Euxoa redimicula</em> (Grt.)</td>
<td>M July</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Ochropiura plecta</em> (L.)</td>
<td>M July-Aug.</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Peridroma saucia</em> (Hbn.)</td>
<td>M June-Sept.</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Diarsia rubifera</em> (Grt.)</td>
<td>MP July-Sept.</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Diarsia jucunda</em> (Wlk.)</td>
<td>M July-Aug.</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Spatelotis clandestina</em> (Harr.)</td>
<td>M June-Aug.</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Graphiphora haruspica</em> (Grt.)</td>
<td>MP July-Aug.</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Eurois occultula</em> (L.)</td>
<td>MP July-Aug.</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Xestia dolosa</em> Franc.</td>
<td>MP June-Aug.</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Xestia normaniana</em> (Grt.)</td>
<td>MP Aug.</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Xestia smithii</em> (Sned.)</td>
<td>M July-Aug.</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Xestia bicarnea</em> (Gn.)</td>
<td>MP July-Aug.</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Xestia tenuicula</em> (?) (Morr.)</td>
<td>M 23 July</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Anomogyra elimate</em> (Gn.)</td>
<td>M July-Aug.</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Anomogyra badicollis</em> (Grt.)</td>
<td>P Aug.</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Anomogyra dilucida</em> (Morr.)</td>
<td>M 8 Sept.</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Paradiarsia littoralis</em> (Pack.)</td>
<td>MP June-July</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Anaplectoides prasina</em> (Denis &amp; Schiffermuller)</td>
<td>MP Aug.-Sept.</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Anaplectoides pressus</em> (Grt.)</td>
<td>M July-Aug.</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Protolampra brunnecollis</em> (Grt.)</td>
<td>P</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Eueretagrotis peratetina</em> (Grt.)</td>
<td>M June-Aug.</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Heptagrotis phyllophora</em> (Grt.)</td>
<td>M July-Aug.</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Cryptocala acadiensis</em> (Bethune)</td>
<td>MP July-Aug.</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Abagrotis alternata</em> (Grt.)</td>
<td>M 6 Sept.</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Rhynchagrotis cupida</em> (Grt.)</td>
<td>P 3 Aug.</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Rhynchagrotis anchocellioides</em> (Gn.)</td>
<td>M Aug.-Sept.</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Pyrhia umbra</em> (Hufn.)</td>
<td>M June-Aug.</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Heliothis zeu</em> (Boddie)</td>
<td>M Sept.</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Schinia rivicola</em> (Gn.)</td>
<td>M 24 Aug.</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Schinia florida</em> (Gn.)</td>
<td>M 10 Aug.</td>
<td></td>
<td></td>
<td></td>
</tr>
</tbody>
</table>

**DISCUSSION**

Moore (1930) listed *Gluphisia lintneri* (Notodontidae) as occurring on Beaver Island. Originally *G. lintneri* and *G. avimacula* were synonymous and therefore mixed in Moore's data. The two are now valid species and *G. avimacula* is extremely abundant on Beaver Island. Although *G. lintneri* may occur, none were collected, and upon examination of Moore's specimens at the University of Michigan collection, all were *G. avimacula*. *Enargia decolor* (Noctuidae) and *E. infumata*, were also originally thought to be one species. Although Moore (1930) listed only *E. decolor*, his specimens include data on both species. Another noctuid, listed by Moore (1930) as *Euplexia lucipara* is now referred to as *E. benesimilis* (Hodges et al. 1983). *E. lucipara* is a palearctic species and does not occur in North America. No endangered or threatened species were found based on the list of endangered lepidopteran species of Michigan (DNR 1988). One "species of special concern" (DNR 1988), *Papaipema aweme* (one specimen), (Noctuidae), was recorded by Moore (1930). This species is known only from a few specimens collected in various parts of North America (M. C. Nielsen, pers. comm.). Little is known about its life history, but the
species may be associated with wetlands or shoreline environments. Repeated efforts to recollect it along lake edges were unsuccessful.

Further investigations should turn up additional species new to Beaver Island, especially moths at ultraviolet/mercury vapor lights. Future collecting should be concentrated in the southern portions of the island and in a variety of habitats. With the exception of Iron Ore Bay, collecting has been virtually nonexistent in areas surrounding Green’s Bay, French Bay, Miller’s Marsh, Lake Geneserath, and on the satellite islands.

ACKNOWLEDGMENTS

I thank J. Gillingham, Central Michigan University (CMU) Biological Station, for his cooperation in collecting at the station, and R. G. Bland, CMU, for reviewing the manuscript and being advisor on this project. R. L. Fischer, Michigan State University and M. F. O’Brien, University of Michigan Museum of Zoology, allowed access to the university insect collections. M. C. Nielsen, Lansing, provided valuable assistance in identification.

LITERATURE CITED

Department of Natural Resources 1988. Michigan’s Special Animals; Endangered, Threatened, Special Concern, and Probably Extirpated. Nongame & Endangered Species Program, Lansing, MI.
NOTEWORTHY RANGE EXTENSIONS OF THREE EMESINE SPECIES (HETEROPTERA: REDUVIIDAE)

J. E. McPherson

ABSTRACT

The first records of *Empicoris culiciformis* and *E. winnemana* from Michigan and of *Pseudometapterus umbrosus* from Illinois are reported. All represent considerable extensions of their recorded ranges.

The emesine reduviids or thread-legged bugs (known in earlier literature as the Ploiariinae or Ploiariidae) are cosmopolitan in distribution (Wygodzinsky 1966). These secretive predaceous insects occur over much of North America but, generally, their biology here is poorly known. The primary exception to this is *Emesaya b. brevipennis* (Say) for which scattered notes on life history have been published (e.g., Uhler 1884, Weed 1889, Wickham 1909, 1910; McAtee 1911; Howes 1919; Readio 1926, 1927; Brown and Lollis 1963; Whitcomb and Bell 1964). Interestingly, both it and *E. brevicoxa* (Banks) are often associated with spider webs (Usinger 1941). For most other North American emesines, biological information consists primarily of data associated with their collection sites (e.g., Wygodzinsky 1966).

Most species in eastern North America are rarely collected, particularly those occurring in the northern states. This may be, in part, because individuals often occur at “considerable heights on bushes and trees” (Wygodzinsky 1966), are stick-like in appearance, and often are less than 12 mm long (the most notable exception being species of *Emesaya*, which often are more than 30 mm long (McAtee and Malloch 1925, Wygodzinsky 1966).

*Empicoris* and *Pseudometapterus* both occur in eastern North America; *Empicoris* is represented by 12 species and *Pseudometapterus* by one (Froeschner 1988). All of the *Empicoris* species average less than 8 mm long (Wygodzinsky 1966); *P. umbrosus* (Blatchley) averages about 15 mm long (Blatchley 1926).

Herein, *E. culiciformis* (De Geer) and *E. winnemana* McAtee and Malloch are reported from Michigan, and *P. umbrosus* is reported from Illinois. These new state records represent considerable extensions of their known ranges.

*Empicoris culiciformis*

This species has been reported from Connecticut, Maryland, Virginia, and Oregon, and from Africa, Europe, and South America (Froeschner 1988); it probably has been dispersed by man (Wygodzinsky 1966). It has been collected at light (McAtee and Malloch 1925, label information) and from the bark of a dead willow (Wygodzinsky 1966, label information); information on its biology in England is provided by Butler (1923). Prior to this paper, the possibility of man's influence
could have explained its odd North American distribution. However, its discovery in Michigan strongly suggests it is more widely distributed in North America than previously thought.

Deposited in the Michigan State University Entomology Museum (MSU), East Lansing, is an adult specimen of *E. culiciformis* with the following label information: MI: Ingham Co., 7 Aug. 1949 (1♀) (No collector label).

*Empicoris winnemana*

This species, originally described in 1925 from specimens collected in Maryland and Virginia, is now also known from Connecticut (Froeschner 1988). It has been collected in October at light (McAtee and Malloch 1925, original label information). Its discovery in Michigan is a considerable westward extension of its known range. I would suspect that rather than representing expansion, this species simply has been overlooked in most of its range because of its small size (4-5 mm long).

Deposited in the MSU and University of Michigan Museum of Zoology (UMMZ), Ann Arbor, are one and two adult specimens, respectively, of *E. winnemana* with the following label information: MI: Livingston Co., E. S. George Reserve, 18 March 1950 (1♀, UMMZ), 15 Oct. 1950 (1♀, UMMZ), K. Bohnsack (Coll., both specimens; previously identified by R. F. Hussey); Midland Co., 5 Sept. 1959 (1♂, MSU), R. R. Dreisbach (Coll.).

*Pseudometapterus umbrosus*

This species is known only from Florida (Froeschner 1988). It has been collected from fallen dead leaves of royal palm in a dense hammock on Paradise Key (Blatchley 1926) and from Spanish moss (Wygodzinsky 1966).

Deposited in the Southern Illinois University Entomology Collection (SIUEC) are two adult specimens of *P. umbrosus*, both collected in the La Rue-Pine Hills Ecological Area, southern Illinois. Its presence there is more difficult to explain. Certainly it is possible that it occurs between Florida and southern Illinois but is rare and, as with *E. culiciformis* and *E. winnemana*, has been overlooked. However, discovery of species in Pine Hills that occur elsewhere only in the south and southeast is not unprecedented. Recently (1986), I coauthored a publication with T. E. Vogt on the Odonata occurring at Pine Hills. Included was a record of a population of *Telebasis byersi* Westfall, a damselfly that previously had been known only from North Carolina, South Carolina, Florida, and Alabama. Pine Hills is well known for its high biological diversity (McPherson and Mohlenbrock 1976). It may, in fact, represent a refugium for several species, based on their presently known distributions.

The label information for the two specimens is as follows: IL: Union Co., (La Rue) Pine Hills, 27 July 1972 (2♂♂, SIUEC), J. F. Walt, Coll. (both specimens).

ACKNOWLEDGMENTS

I thank M. F. O'Brien, Insect Division, UMMZ; and F. W. Stehr and R. L Fischer, Department of Entomology, MSU; for permission to examine the collections at their respective institutions. I am also grateful to A. V. Provonsha, Department of Entomology, Purdue University, West Lafayette, IN; and R. T. Schuh, Department of Entomology, American Museum of Natural History, NY; for allowing me to examine the lectotype and paratypes of *Pseudometapterus umbrosus* and *P. wygodzinskyi* (Elkins), respectively, to determine which of these closely related species was represented by the Pine Hills specimens.
LITERATURE CITED


AN ARRAY OF SPATULATE SENSILLA ON ANTENNAE OF MALE BRACHYMERIA LASUS (HYMENOPTERA: CHALCIDIDAE)

D. H. Simser¹ and H. C. Coppel²

ABSTRACT

An array of spatulate sensilla on the ventral flagellar surface of each antenna of male Brachymeria lasus occurs only on segments IV-VII and is absent on female antennae. Most such sensilla are on segment VI. Each spatulate sensillum was 15 μ by 16.7 μ with a stalk extending 17 μ from the antennal base. Pores were not apparent, but the sensillum surface was imbricated. The sensilla are speculated to have a role in the courtship sequence of this chalcid by functioning both as chemoreceptors of the female-produced sex pheromone and as mechanoreceptors to indicate female receptivity, as female B. lasus typically raise the abdomen to expose the genital pocket.

The chalcid wasp, Brachymeria lasus (Walker), a native of Japan, India and Indonesia (Joseph et al. 1973, Habu 1960) was imported and released as a potential biological control agent against the gypsy moth, Lymantria dispar (L.), in the United States between 1908-1914 (Burgess and Crossman 1929). This solitary pupal parasitoid failed to establish, possibly due to climatic differences, and subsequent releases were discontinued. Recently, B. lasus has been reintroduced and released in gypsy moth infested areas in North America and renewed interest has prompted ongoing biological investigations (Weseloh and Anderson 1982).

Observations of the courtship behavior of B. lasus confirmed that the antennae were preemptory to successful mating. We describe the gross morphology and antennal location of these apparent sensory receptors and propose their possible function in mate location and courtship.

MATERIALS AND METHODS

A laboratory culture of B. lasus was maintained (Simser and Coppel 1980a) and wasps were provided with pupae of the greater wax moth, Galleria mellonella L. Parasitized host pupae were placed singly in gelatin capsules (#00) until parasitoid eclosion. Newly eclosed males and females were placed within a freezer for one hour, then examined with a dissecting microscope at 20-40x. Ten male and ten female wasps were examined. The location and number of spatulate sensilla per segment was recorded. Antennae were also prepared for scanning electron observation after freezing. Male and female heads were ablated, fixed to a metal stub with silver conducting paint, and vacuum coated with a 100 Å layer of gold-palladium in

¹New Alchemy Institute, 237 Hatchville Rd., Falmouth, MA 02536.
²Department of Entomology, University of Wisconsin, Madison, WI 53706.
RESULTS AND DISCUSSION

The geniculate, 11-segmented antenna of *B. lasus* is composed of a scape, pedicel and ring segment (I-III) and flagellum (IV-XI) terminating with a concave distal tip. Although sensilla trichoidea and chaetica (Zacharuk 1980) are distributed on all antennal segments, discrete spatulate sensilla are restricted to the ventral surfaces of segments IV-VIII (Fig. 1) in a uniform arrangement. No other sensillar types were noted within this zone. The morphological nature of these discrete sensilla permit their description as spatulate, or 'spoon-shaped and attached at the narrow end.' Thus, spatulate sensilla are restricted to a localized section of male *B. lasus* antennae and are not present on the female. Male *B. lasus* had 86.8 (ave.) spatulate sensilla, the majority of which are on segments V and VI (Table 1). Numbers varied with different males.

The sensilla were oriented to be flattened in the ventral plane, thus affording greatest surface area, although the sensillar surfaces were not contiguous, nor were individual sensillum in contact (Fig. 2 and 3). Each sensillum originates from the ventral antennal surface by a stalk 17 μ long from base to point of proximal attachment. This stalk may serve as a point of articulation. The flattened spatula was 16.7 μ long and 15 μ at its widest diameter (Fig. 2) and gradually tapered to both proximal and distal ends. The dorsal surface is apparently imbricated with a series of discrete and localized zones (Fig. 4), but pores were not observed. Numerous sensilla trichoidea, placodea and basiconica were observed on all flagellar segments, but none in the spatulate sensillar zone.

Joseph et al. (1973) described spatulate sensilla on male *B. lasus* as flattened sensilla trichoidea. However, morphological comparison reveals differences, as sensilla trichoidea are represented by filamentous or hair-like structures (Zacharuk 1980), whereas the spatulate sensilla are typically shaped with a broad surface extending from a basal stalk. Spatulate shaped receptors have been described in other Insecta, including a tortricid moth, *Cydia nigricana* F. (Wall 1978) and a tenebrionid beetle, *Tenebrio molitor* L. (Harbach and Larsen 1977). In the tortricid, the spatulate structures were identified as sensilla auricolica. These sensilla were morphologically distinct from the spatulate sensilla of *B. lasus*, however, as they were longitudinally grooved and elongate. The male tenebrionid has an antennal spatulate bristle that is 50.6 μ long and is common on antennal segments V-IX. Each bristle has a corrugated surface and an apical pore. In either case, the spatulate receptors were not found in an array as noted in *B. lasus*. Scanning or transmission electron microscope examinations of hymenopterous parasites have not revealed zones or arrays of distinct sensilla as in *B. lasus*, although specialized receptors are noted (Borden et al. 1973, 1978, Norton and Vinson 1974, Richerson et al. 1972, Voegele et al. 1975, Weseloh 1972).

The few studies of courtship behavior by *Brachymeria* species have reflected the importance of antennae in the behavioral sequence between male and female conspecifics. Leonard and Ringo (1978) documented that 69% of discrete courtship behaviors involved use of the male antennae for female location and subsequent courtship. Similarly, Simser and Coppel (1980b) demonstrated that *B. lasus* males utilized a similar proportion of antennal behaviors in their courtship sequence. Males respond to a female pheromone by directing their antennae forward and ceasing random movements. This behavior orients the spatulate sensilla toward the female pheromone source. The male then advances with a side to side oscillatory movement and presses the antennae on to the wings and dorsum of the female.
Figures 1–2. Spatulate sensilla on male *Brachymeria lasus*: 1 ventral surface of antennae (× 65); 2 surface of spatulate sensilla showing array (× 1,500).
Figures 3–4. Spatulate sensilla on male *Brachymeria lasus*: 3 lateral view of sensilla showing orientation (× 500); 4 surface of sensilla showing imbricated surface (× 3,000).
Table I. Segmental location of spatulate sensilla on male *B. lasus* antennae

<table>
<thead>
<tr>
<th>Antennal segment</th>
<th>Average number of spatulate sensilla</th>
<th>SEM</th>
</tr>
</thead>
<tbody>
<tr>
<td>I</td>
<td>0</td>
<td>-</td>
</tr>
<tr>
<td>II</td>
<td>0</td>
<td>-</td>
</tr>
<tr>
<td>III</td>
<td>0</td>
<td>-</td>
</tr>
<tr>
<td>IV</td>
<td>15.3</td>
<td>0.88</td>
</tr>
<tr>
<td>V</td>
<td>29.8</td>
<td>1.02</td>
</tr>
<tr>
<td>VI</td>
<td>23.4</td>
<td>0.75</td>
</tr>
<tr>
<td>VII</td>
<td>13.1</td>
<td>0.64</td>
</tr>
<tr>
<td>VIII</td>
<td>5.2</td>
<td>0.28</td>
</tr>
<tr>
<td>IX</td>
<td>0</td>
<td>-</td>
</tr>
<tr>
<td>X</td>
<td>0</td>
<td>-</td>
</tr>
<tr>
<td>XI</td>
<td>0</td>
<td>-</td>
</tr>
</tbody>
</table>

*Average number of sensilla from ten male *B. lasus*; number of sensilla from right and left antenna combined.

Following a series of antennal presses and wing buzzes, receptive females respond by raising the abdomen about 45°. This action appresses the spatulate sensilla to her abdomen. At this point, the male ceases courtship activity and attempts intromission. The presence and positioning of spatulate sensilla indicates their possible role in reception of mechanical and/or chemical cues to afford the continuing success of *B. lasus*.

ACKNOWLEDGMENTS

Gratitude is extended to Mr. Louis Kerr, Marine Biological Laboratory, Woods Hole, MA for preparation and scanning electron micrography of the specimens. This research was supported by the College of Agricultural and Life Sciences, University of Wisconsin-Madison, by Federal Hatch support (Project N. 2116) to H. C. Coppel and by USDA, SEA AR Agreement No. 58-519B-1-99 to H.C. Coppel and W.E. Burkholder.

LITERATURE CITED


FOODPLANT PROCESSING ADAPTATIONS IN FOUR HYALOPHORA SPECIES (LEPIDOPTERA: SATURNIIDAE): REGIONAL AND TAXONOMIC SPECIALIZATION

J. Mark Scriber¹ and Eric Grabstein²

ABSTRACT

To determine whether local populations of four Hyalophora species (Lepidoptera: Saturniidae) had improved survival or were physiologically adapted for rapid and/or efficient growth on their local hosts, a series of larval feeding studies were conducted using gravimetric techniques on several host plant species. Significantly better survival and growth performances were observed for H. columbia (a tamarack specialist) on its host, Larix laricina. Similarly, H. gloveri had the best growth performance on Elaeagnus angustifolia (its favorite) as did certain sympatric populations of H. cecropia on black cherry, Prunus serotina. Hyalophora gloveri and H. columbia are largely sympatric with Betula papyrifera and perform better than their allopatric congeners H. cecropia in the east and H. euryalus in the west. While survival of the tamarack specialist was poor, all three of the other North American Hyalophora species survived and grew very well on choke cherry, Prunus virginiana, which is sympatric with all four Hyalophora species. The extent to which these are genetically based adaptations is not known, nor are the specific mechanisms of biochemical adaptation involved in these differential performances of larvae.

Dethier (1954) predicted that monophagous species should utilize their host more efficiently than polyphagous species. Hypotheses regarding the evolution of feeding specialization usually assume there is a cost to adaptation, with an implied trade-off principal that "a jack of all trades is master of none" (see Futuyma and Moreno 1988 for a review). Scriber and Feeny (1979) empirically tested this hypothesis using butterfly (Papilionidae) and silkmoth (Saturniidae) species. Variability in plant nutritional and allelochemical content with various plant species and regional variation in insect feeding preferences were found to be major problems in assessing these physiological and toxicological "costs". Comparisons of congeners with different degrees of specialization, but with a shared host plant proved to be a better test of the hypothesis, since nutritional/allelochemical variability are minimized (Scriber 1984). One of the best tests of the feeding specialization hypothesis (that larval performance will be better on local host plant favorites that populations have adapted to, than on hosts used elsewhere) appears to be possible with a comparison of larval performance of the giant silkmoth family (Saturniidae). For this study we chose the widespread, but endemic North American silkmoth genus Hyalophora with its four largely parapatrically distributed species (cecropia, euryalus, gloveri, and columbia; Fig. 1).

¹Current address and correspondence to: Dr. J. Mark Scriber, Department of Entomology, Michigan State University, East Lansing, MI 48824.
²Dept. of Entomology, University of Wisconsin, Madison, WI 53706.
The widespread eastern species *Hyalophora cecropia* (L.) is polyphagous, capable of feeding on at least 88 different species of plants from 20 families of angiosperms including introduced ornamentals (Waldbauer and Sternburg 1967, Scarbrough et al. 1974). The most common natural hosts are geologically old families, the Rosaceae, Salicaceae, and Aceraceae (Ferguson 1972). The two western species *H. gloveri* (Strecker) and *H. euryalus* (Boisduval) prefer the Rhamnaceae (*Ceanothus* and *Rhamnus* spp.) and Ericaceae (*Arctostaphylos* and *Arbutus*), members of the geologically more recent Madro-Tertiary flora (Alexrod 1977, Collins 1984). These plants are important members of sclerophyll chaparral and scrub communities in the California Coast Ranges and Sierra Nevada and are adapted to a climate of winter rains and summer drought; the range of *H. euryalus* is nearly congruent with the broad extent of these plant communities. While *H. gloveri* includes manzanita and other *Arctostaphylos* spp. (Ericaceae) as hosts in Arizona and *Ceanothus velutinus* (Rhamnaceae) in the Great Basin and Rocky Mts., its principal hosts are *Salix* spp. (Salicaceae) and *Prunus* spp. and *Purshia tridentata* (Rosaceae) (Collins 1984). Northern populations of *H. gloveri* occupying recently deglaciated terrain are more specialized feeders. In the northern plains *H. gloveri nokomis* prefers buffalo berry
1991 THE GREAT LAKES ENTOMOLOGIST

(Shepherdia argentea Pursh), silverberry (Elaeagnus commutata), and the introduced Russian olive (Elaeagnus angustifolia), all in the Elaeagnaceae (Collins 1973).

The primarily Canadian form H. columbia (Smith) inhabits acid bogs and is a specialist on the conifer tamarack (Larix laricina). Although H. columbia is commonly treated as a separate species, Kohalmi and Moens (1975) and Collins (1973) describe intergradient populations in nature, and female hybrids between columbia and nominate gloveri are fully fertile. In northern Wisconsin, H. columbia and H. cecropia are sympatric and fly at the same time of the season, however due to various behavioral prezygotic isolating mechanisms there appears to be a low incidence of hybridization (Ferge 1983, Tuttle 1985).

Our objective in this study was to determine whether there is evidence that local foodplant specialization has an associated component of improved larval survival, growth rate, and/or efficiency. To conduct the study, we obtained specimens of all four Hyalophora silkmoth species and proceeded with controlled environment feeding experiments on selected foodplant species, including: black cherry, Prunus serotina, and choke cherry, P. virginiana (Rosaceae), paper birch, Betula papyrifera (Betulaceae), Russian olive, Elaeagnus angustifolia (Elaeagnaceae), and tamarack, Larix laricina (Pinaceae).

METHODS AND MATERIALS

Breeding stock was collected in the wild as cocoons or obtained from breeders who could verify its regional source (Fig. 1). Interpopulational and interspecific hybrids were obtained by laboratory cage mating and with funnel traps in the field which were baited with virgin Hyalophora females. Mated females were placed in paper bags to oviposit. Clusters of ova were cut out and placed in 10 cm diameter plastic petri dishes. Small twigs of host plant were maintained in “aquapics” placed in the petri dishes and changed at 1 to 3 day intervals, at which time larvae were censused for survival and stage of growth.

For penultimate instar feeding studies, Hyalophora larvae of each species were reared from neonate stages through to the penultimate instar en masse on the particular foodplant to be used in feeding experiments. These groups of individuals were kept in an environmental chamber set at 16 hr photo-: 8 hr scotophase, and at 23.5°C and 19.5°C during the photo- and scotophase, respectively. The relative humidity in the chamber was roughly 60% and 90% during the photo- and scotophase, respectively.

At the beginning of the penultimate stadium, larvae were placed individually in 150 x 25 mm plastic petri dishes with moist filter paper to maintain humidity. Petri dishes were kept in an environmental chamber under the same conditions described for rearing. Sample larvae were weighed at the beginning of the penultimate stadium, frozen, lyophilized and then weighed again (dry). The percentage of dry biomass was determined for each larva.

Foodplant leaves were excised from branches (collected the previous day and stored in water at ca. 5–10°C) immediately before use, weighed and placed by their petioles in Aquapics® to maintain their turgidity (Scriber 1977). Sample aliquots of leaves were taken from each branch for later determinations of percentage dry weights. Fresh leaves were presented at approximately 48 hr intervals; uneaten food was collected, dried and weighed. The food consumption of each larva for the whole penultimate stadium was estimated by standard gravimetric techniques (Waldbauer 1968).

Nutritional indices were calculated based upon dry weight (biomass) of leaves, feces, and larvae. The mean larval weight during the stadium (B) was estimated
by measuring the the initial plus final weight and dividing by 2. Indices of larval performance are reported as in Sibler and Slansky (1981).

RESULTS

While all four Hyalophora species survived well in the early instar stages on the two cherry species (black cherry and choke cherry), penultimate instar consumption rates and efficiencies of conversion of plant tissue into insect biomass were significantly different among species and populations. On black cherry, larvae of the tamarack feeding specialist (H. columbia) did not survive to the feeding study and H. gloveri larvae from a Utah population grew at a significantly lower rate than eastern H. cecropia larvae (Table 1). Larvae of H. euryalis from California grew significantly slower than H. cecropia from Wisconsin, but were only slightly slower than H. cecropia from Colorado (Table 1). Of all populations, the fastest growing and most efficient (E.C.I.) were those of Wisconsin H. cecropia which are also the only populations of cecropia tested that are sympatric with the black cherry host plant. Slower and less efficient growth of Colorado H. cecropia compared to Wisconsin H. cecropia suggest that interpopulation as well as interspecific physiological adaptations exist.

Nearly all H. columbia larvae died on choke cherry, P. virginiana, as with black cherry, before reaching the penultimate instar feeding experiment (Table 2). Unlike the case with black cherry, on choke cherry all of the other Hyalophora species grew at the same rate, with some trade-offs in consumption rates and efficiencies (Table 2). Unlike black cherry, the geographic range of choke cherry extends across almost all of Canada and the northern half of the United States and is sympatric in certain locations with all four species of Hyalophora in this study. It should be noted that choke cherry of California has been considered taxonomically distinct and is sometimes called Prunus demissa. Whether or not the identical growth rates of the various Hyalophora species reflect similar physiological adaptations evolved due to local use of choke cherry by their populations in areas of sympatry is unclear (since local preferences are poorly documented). However, it would appear that Rosaceae is a generally suitable host plant family for the group, with H. columbia as a possible exception.

Another host plant that has considerable geographic overlap with the Hyalophora group is paper birch, Betula papyrifera, with a range across Canada at least as extensive as is that of Prunus virginiana. Paper birch extends from New England south into the Smoky Mts. and across the Great Lakes states and essentially all of Canada into Alaska. It does not extend south throughout the Rockies nor does it occur in California. Nonetheless, larvae of H. euryalis from California and H. gloveri from Utah grew as fast and as efficiently on this plant as did the Wisconsin H. cecropia (Table 3). We were unable to test H. columbia larvae on this plant, but hybrid columbia x gloveri larvae grow at an extremely rapid rate (195 mg g⁻¹ day⁻¹). It would be especially valuable to bioassay H. gloveri from Canada, where the extensive use of paper birch is perhaps most likely. In fact, Hyalophora eggs were collected on paper birch in the putative hybrid zone between gloveri and columbia in the Riding Mts. of Manitoba (J. M. Sibler personal observation), however too few were found for experimental feeding studies.

In the heart of the range of H. gloveri, Elaeagnaceae are the primary host plants (Ferguson 1972, Collins 1973). One of these host plant species, Russian olive (Elaeagnus angustifolia), was bioassayed with all four Hyalophora species to see if differential adaptations may be evident in growth of their larvae. All of the H. columbia died before or during the penultimate instar while feeding on E. angustifolia (Sibler and Collins, unpublished data). All H. euryalis also died in the neonate (first instar) stage and no feeding experiments were possible. Both H. gloveri and H. cecropia
Table 1. Utilization of black cherry, *Prunus serotina* by penultimate instar larvae of four *Hyalophora* species.

<table>
<thead>
<tr>
<th>Hyalophora species</th>
<th>Consumption Rate (RCR) mg/day/g</th>
<th>(AD) %</th>
<th>Efficiency (ECD) %</th>
<th>(ECI) %</th>
<th>Growth Rate (RGR) mg/day/g</th>
</tr>
</thead>
<tbody>
<tr>
<td><em>H. eurialis</em>¹</td>
<td>(4)</td>
<td>859 ± 36 b</td>
<td>34.3 ± 1.0 ab</td>
<td>45.1 ± 1.2 a</td>
<td>15.5 ± 0.2 abc</td>
</tr>
<tr>
<td><em>H. gloveri</em>²</td>
<td>(UT) (5)</td>
<td>645 ± 33 b</td>
<td>33.4 ± 0.9 ab</td>
<td>43.1 ± 3.0 a</td>
<td>14.3 ± 0.9 bc</td>
</tr>
<tr>
<td><em>H. cecropia</em>³</td>
<td>(CO) (8)</td>
<td>988 ± 37 ab</td>
<td>36.8 ± 1.8 ab</td>
<td>39.0 ± 1.6 a</td>
<td>14.1 ± 0.1 bc</td>
</tr>
<tr>
<td><em>H. cecropia</em>³</td>
<td>(CO) (6)</td>
<td>993 ± 64 ab</td>
<td>42.0 ± 2.2 a</td>
<td>38.9 ± 1.8 a</td>
<td>16.2 ± 0.4 abc</td>
</tr>
<tr>
<td><em>H. cecropia</em>³</td>
<td>(CO) (5)</td>
<td>943 ± 38 ab</td>
<td>33.8 ± 1.4 ab</td>
<td>47.0 ± 1.6 a</td>
<td>15.8 ± 0.5 abc</td>
</tr>
<tr>
<td>(WI) (5)</td>
<td>1088 ± 33 a</td>
<td>43.0 ± 0.6 a</td>
<td>41.7 ± 2.0 a</td>
<td>17.9 ± 0.9 ab</td>
<td>194 ± 5 ab</td>
</tr>
<tr>
<td>(WI) (14)</td>
<td>1043 ± 39 a</td>
<td>42.4 ± 1.0 a</td>
<td>46.7 ± 2.2 a</td>
<td>19.5 ± 0.5 a</td>
<td>202 ± 6 a</td>
</tr>
<tr>
<td><em>H. columbia</em>⁵</td>
<td>(all died)</td>
<td>--</td>
<td>--</td>
<td>--</td>
<td>--</td>
</tr>
<tr>
<td>(LSD)⁶</td>
<td>(390)</td>
<td>(9.9)</td>
<td>(21.1) n.s.</td>
<td>(4.3)</td>
<td>(38)</td>
</tr>
</tbody>
</table>

¹ *H. eurialis* from California (Nevada Co.)
² *H. gloveri* two source female from K. Thorne (Utah).
³ *H. cecropia* three source females from Colorado (outside the natural range of black cherry); two source females from Madison, WI which use black cherry as one of the natural host plants.
⁴ *H. cecropia* from S. Stone (Colorado); *H. columbia* from D. Robacker (Price Co., WI).
⁵ *H. columbia* from L. Ferge (Oneida Co., WI).
⁶ n.s. = no significant differences between the means.

Significant differences between the means are indicated (P = 0.05 via Tukey's test for unequal sample sizes).
Table 2. Utilization of choke cherry (*Prunus virginiana*) may penultimate instar larvae of *Hyalophora* species. Data are presented as a mean ± SE (see methods for procedures).

<table>
<thead>
<tr>
<th>Insect taxa</th>
<th>(n)</th>
<th>Consumption Rate (RCR) mg/day/g</th>
<th>(AD) %</th>
<th>Efficiency (ECD) %</th>
<th>(ECI) %</th>
<th>Growth Rate (RGR) mg/day/g</th>
</tr>
</thead>
<tbody>
<tr>
<td><em>H. euryalus</em></td>
<td>3</td>
<td>734 ± 31 b</td>
<td>37.2 ± 1.4 a</td>
<td>44.7 ± 2.8 b</td>
<td>16.6 ± 0.7 a</td>
<td>121 ± 7 a</td>
</tr>
<tr>
<td><em>H. gloveri</em></td>
<td>3</td>
<td>896 ± 84 a</td>
<td>26.8 ± 0.9 b</td>
<td>45.1 ± 0.8 b</td>
<td>12.8 ± 0.3 c</td>
<td>109 ± 12 a</td>
</tr>
<tr>
<td><em>H. cecropia</em></td>
<td>8</td>
<td>680 ± 22 b</td>
<td>27.6 ± 0.6 b</td>
<td>54.0 ± 1.3 a</td>
<td>14.9 ± 0.2 b</td>
<td>101 ± 2 a</td>
</tr>
<tr>
<td><em>H. columbia</em></td>
<td></td>
<td>(all but one died earlier)</td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
</tbody>
</table>

(L.S.D.) (162) (3.5) (6.9) (1.5) (23) n.s.

1From California (Nevada Co.). The choke cherry used by *gloveri* and *euryalus* in nature is *P. demissa*, now considered a separate species from *virginiana*.  
2From K. Thorne (Utah).  
3From Wisconsin (Dane Co.)  
4From Wisconsin (Lincoln Co.)

Precise counts of the initial number of neonate larvae were not made in this study; however replication is less than the desired ten 4th instar larvae because too few survived to reach this penultimate instar feeding experiment stage.

Significant differences between the means are indicated (P = 0.05) via Tukey's test for unequal sample sizes (Winer 1962, Snedecor and Cochran 1967).

Table 3. Utilization of paper birch, *Betula papyrifera*, by various *Hyalophora* species. Data are presented as a mean ± SE.

<table>
<thead>
<tr>
<th>Insect species</th>
<th>(n)</th>
<th>Consumption Rate (RCR) mg/day/g</th>
<th>(AD) %</th>
<th>Efficiency (ECD) %</th>
<th>(ECI) %</th>
<th>Growth Rate (RGR) mg/day/g</th>
</tr>
</thead>
<tbody>
<tr>
<td><em>H. euryalus</em></td>
<td>4</td>
<td>963 ± 67 b</td>
<td>36.1 ± 3.1 a</td>
<td>36.2 ± 4.3 b</td>
<td>13.0 ± 1.8 a</td>
<td>128 ± 24 ab</td>
</tr>
<tr>
<td><em>H. gloveri</em></td>
<td>2</td>
<td>1432 ± 62 a</td>
<td>34.3 ± 2.2 a</td>
<td>24.1 ± 2.4 b</td>
<td>8.2 ± 0.3 a</td>
<td>117 ± 1 b</td>
</tr>
<tr>
<td><em>H. cecropia</em></td>
<td>8</td>
<td>833 ± 51 b</td>
<td>27.2 ± 0.8 a</td>
<td>63.6 ± 2.2 a</td>
<td>17.3 ± 0.5 a</td>
<td>147 ± 9 ab</td>
</tr>
<tr>
<td><em>H. columbia</em></td>
<td></td>
<td>(all but one died earlier)</td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
</tbody>
</table>

(L.S.D.) (274) (12.5) n.s. (26.4) (9.6) n.s. (68)

1From California (Nevada Co.).  
2Utah *gloveri* × Panamint Mts. *gloveri* CA.  
3From Wisconsin (Dane Co.)  
4From Wisconsin (Lincoln Co.)

Significant differences between the means are indicated (P = 0.05, Tukey's test for unequal sample sizes).
survived and grew to pupation on this plant. *Hyalophora gloveri*, the species normally using Elaeagnaceae as host plants, grew at nearly twice the rate (146 mg g\(^{-1}\) day\(^{-1}\)) of the generalist (70 mg g\(^{-1}\) day\(^{-1}\); Table 4). Only occasionally (where used as ornamentals) might *H. cecropia* encounter and use Russian olive. *Elaeagnus commutata* or *Shepherdia canadensis* (which extends to south Michigan) could be used by *columbia*, but no reports exist. This difference in growth rate was due to both greater efficiencies and faster consumption by *H. cecropia* (Table 4), and it would appear that differential adaptations (behavioral, physiological, and toxicological) exist among the four North American *Hyalophora* species with regard to *Elaeagnus angustifolia*.

The larch feeding specialist, *Hyalophora columbia* survived and grew exceptionally well on eastern larch, *Larix laricina* (Fig. 2). In fact, consumption rates of *Larix* by *H. columbia* were faster than any other *Hyalophora* species on any foodplant (Tables 1–5). Also, their growth rate was as fast as *H. cecropia* on black cherry, and these represent the fastest growth rates reported for any *Hyalophora* on any host plant species (Table 5). While 1st instar survival on tamarack is excellent for all four species of *Hyalophora* (77%–100%, M. Collins and J.M. Scriber unpublished), none of the larvae in our lab except *H. columbia* survived to the penultimate instar feeding experiments here.

**DISCUSSION**

Differential survival and growth on a particular plant species by different species of Lepidoptera could be expected if different adaptations (behavioral, physiological and toxicological) have evolved. This study, using the four North American species of giant silkmoths (*Hyalophora cecropia*, *H. euryalus*, *H. gloveri*, and *H. columbia*), illustrates that differential survival and performance does occur in certain cases. However, it is important to note that survival and growth of larvae from widely scattered localities in laboratory feeding experiments may not perfectly reflect adaptations of the species as a whole, or even the adaptations of the source population under their natural (outdoor) conditions (see Scriber 1983, 1984).

While black cherry and choke cherry (*P. serotina* and *P. virginiana* of the Rosaceae) are generally suitable for survival and growth of *Hyalophora*, there appear to exist interspecific differences, and intraspecific (interpopulation) differences that might reflect local adaptations (Tables 1 and 2). Similarly, paper birch (*Betula papyrifera*) of the Betulaceae appears generally suitable to various *Hyalophora* species, although comparable growth is achieved differently by different species. For example, *H. gloveri* consumes paper birch rapidly but metabolically are inefficient in processing the leaves compared to *H. cecropia* larvae that consume slowly (Table 3). *Hyalophora euryalus* is intermediate in both consumption rate and efficiency (Table 3).

The differential adaptations of *H. gloveri* to *Elaeagnus angustifolia* appear quite prominent. Toxic to neonate larvae of *H. euryalus* and unsuitable for development of *H. columbia*, Russian olive also extracts a cost in *H. cecropia* that halves its growth rate compared to *H. gloveri* (Table 4). This appears to represent a significant divergence in host plant adaptations, and may reflect significant underlying phytochemical bases of the interspecific antibiosis observed with *Hyalophora*.

While the tamarack feeding specialist grows extremely well on tamarack, other *Hyalophora* can also survive and grow relatively well as early instars on this plant (M. Collins and J.M. Scriber, unpublished). The degree to which the tamarack feeding abilities are shared by other *Hyalophora* deserves additional study.

The determination of whether the differences observed in these feeding studies have a genetic basis awaits a series of hybridization and backcross studies as has been done for other Lepidoptera (Peigler 1977, Thompson 1988a, Scriber et al.
Table 4. Nutritional indices of penultimate instar larvae of *H. gloveri* and *H. cecropia* fed Russian olive, *Elaeagnus angustifolia*.

<table>
<thead>
<tr>
<th></th>
<th>Consumption Rate (RCR) mg/day/g</th>
<th>Efficiency (ECD) %</th>
<th>Efficiency (ECI) %</th>
<th>Growth Rate (RGR) mg/day/g</th>
</tr>
</thead>
<tbody>
<tr>
<td><em>H. gloveri</em> 1,2</td>
<td>892 ± 60 a</td>
<td>31.3 ± 0.8 a</td>
<td>52.8 ± 2.1 a</td>
<td>146 ± 07 a</td>
</tr>
<tr>
<td><em>H. cecropia</em> 2</td>
<td>585 ± 31 b</td>
<td>28.9 ± 0.8 a</td>
<td>47.0 ± 2.1 a</td>
<td>79 ± 05 b</td>
</tr>
<tr>
<td><em>H. columbica</em> 3</td>
<td>-</td>
<td>-</td>
<td>31.0 ± 3.6</td>
<td>All died</td>
</tr>
<tr>
<td><em>H. euryalis</em> 4</td>
<td>-</td>
<td>-</td>
<td>13.7 ± 0.9</td>
<td>All died</td>
</tr>
</tbody>
</table>

1From K. Thorne (Utah).
2From Wisconsin (Dane County).
3From Ontario. All larvae died before or during (n = 4) the penultimate (4th) instar.
4From California. All larvae died in the first instar (see also Table 1).

Significant differences indicates F-test (P = 0.05)

Table 5. Utilization of eastern larch (tamarack), *Larix laricina*, by *Hyalophora columbica*. Data are presented as a mean ± SE.

<table>
<thead>
<tr>
<th></th>
<th>Consumption Rate (RCR) mg/day/g</th>
<th>Efficiency (ECD) %</th>
<th>Efficiency (ECI) %</th>
<th>Growth Rate (RGR) mg/day/g</th>
</tr>
</thead>
<tbody>
<tr>
<td><em>H. columbica</em> 1</td>
<td>1471 ± 72</td>
<td>42.7 ± 2.9</td>
<td>31.0 ± 3.6</td>
<td>185 ± 09</td>
</tr>
</tbody>
</table>

1Original cocoons from Les Kohalmi (Ontario, CANADA).
Figure 2. The geographic distribution of tamarack, *Larix laricina*, and *Shepherdia* (Eleagnaceae) with *Hyalophora* collection sites (adapted from Harlow and Harrar 1949, Collins 1973, Kohalmi and Moens 1975). Question mark indicates Riding Mountains site of *Hyalophora* egg collection on paper birch, *Betula papyifera* (JMS).

It is also unknown for any *Hyalophora* species whether physiological adaptation to particular host plants is also paralleled by more specific choices in host plant selection by ovipositing females (i.e., a preference/
The importance of these adaptations in host choice and host use abilities for interpretation of the geographic distribution of *Hyalophora* is of major significance. It would be of considerable interest to know whether the inability to survive and grow rapidly and/or efficiently on specific plants is a consequence or a cause of feeding specialization. Such uncertainties about cause and effect of feeding specialization might be best resolved by additional and extensive comparisons of interpopulation and intrapopulation variation in growth performance than by interspecific comparisons. Generalist species such as *H. cecropia* provide a uniquely suited opportunity to pursue such questions.

The common assumption that specialists evolve from generalists may be true most of the time, but is likely not to be true in certain cases. In North America, for example, the polyphagous tiger swallowtail, *Papilio glaucus* (Papilionidae) is felt to have evolved from Lauraceae, Magnoliaceae, and/or Rutaceae specialists (Miller 1987, Scriber et al. 1991, Hagen and Scriber 1991). In a similar case, it is likely that the polyphagous North American moth *Antheraea polyphemus* (Cramer) (Saturniidae) is derived from an Asian oak-feeding (Fagaceae) specialized form similar to *A. pernyi* (Guerin) and is thought to have migrated from Eurasia via the Bering land bridge during the Miocene (Ferguson 1972, Michener 1952). The North American saturniids *Actias luna* Linn. and *Antheraea polyphemus* closely resemble their Asian congeners and are evidence of this migration, while the genus *Hyalophora* appears to have evolved originally as a polyphagous Nearctic endemic (Collins, pers. comm) and *H. columba* has become specialized.

This polyphagy of *H. cecropia* (and of the tiger swallowtail butterfly, *Papilio glaucus*) suggests a long association with the species-rich flora of the southeastern U.S., which includes many plant species with Asian affinities (Graham, 1964). Thus the present distribution of *H. cecropia* and its polyphagy in association with "ancient" hosts are evidence that *H. cecropia* may be more closely related to the progenitor of modern *Hyalophora* than are its western congeners (M. Collins, pers. comm.). A similar, and strikingly parallel host range pattern is evident with the *Papilio glaucus* group (Dethier 1954, Scriber 1988, Hagen and Scriber 1991). Furthermore, the southeastern forest flora, or at least important elements of it, appear to have survived the pleistocene glaciation more or less intact in refugia (Watts 1980, Davis 1981, Solomon and Webb 1985) which makes it plausible that *cecropia* as a taxon may have occurred prior to this event. Intraspecific adaptations to host plants, with the individual variation in oviposition choice and larval host use carefully monitored will help determine the genetic basis of these phenotypic responses. Such studies may help resolve these speculations about the relationships of insect phylogeny and host plant affiliations.

In summary, it does appear that improved physiological and toxicological performance may be the result of adaptations evolved on local hosts in the case of *H. columba* on its primary host (tamarack) and for *H. gloveri* on its primary host (*Elaeagnus*), and possibly for certain *H. cecropia* populations on their locally preferred host (*Prunus serotina*). We do not know the extent to which these improved performances are genetically based.

**ACKNOWLEDGMENTS**

This work was supported in part by the College of Agriculture and Life Sciences of the University of Wisconsin (Hatch 5134), the National Science Foundation (DEB 7921749 and BSR 8306060) and Michigan State University (MAES #8051 and 1644). We extend special thanks to Michael Collins, Mark Evans, Les Ferge, Dave Robacker, Steve Stone, and Ken Thorne for their help in collecting these *Hyalophora* species. We particularly recognize the inspirational enthusiasm of Dr. Michael Collins, whether in the field hunting cocoons or in the lab discussing...
evolutionary scenarios of *Hyaiophora*. His comments have also helped improve the text.

**LITERATURE CITED**


—. 1983. The evolution feeding specialization, physiological efficiency, and host races.


TRAP-NEST DESIGN FOR SMALL TRAP-NESTING HYMENOPTERA

John M. Fricke

Many solitary bees and wasps construct brood cells in pre-existing natural cavities such as beetle borings or in excavations of pithy stems and twigs like *Sambucus* and *Juglans*. Artificial nesting materials are also acceptable and provide a convenient approach to study nest architecture, nesting activity, provisions and parasites. Artificial nesting materials have included bamboo, glass tubes, plastic straws, cuttings of twigs and stems, and trap-nests. However, use of many of these materials have significant drawbacks. Condensation in glass tubes and plastic straws make these materials ineffective. Bamboo has a varying bore diameter and cuttings of twigs and stems are split with great difficulty. Trap-nests, small rectangular wooden blocks with holes drilled into their longitudinal axes, eliminate or greatly reduce all of these problems. Trap-nest bores can be varied in depth and diameter and are analogous to natural cavities used as nesting sites. Trap-nests of clear straight-grain pine can be split in half lengthwise to expose nest contents with relative ease, especially if bore diameters are greater than 3.2 mm.

Initial trap-nest construction techniques for my studies were derived from Fye (1965) and Krombein (1967). Pine boards were cut into trap-nests (19 x 19 x 140 mm) with bores drilled to depths of 60 and 120 mm. Bore diameters and bore depths varied seasonally, dependent upon prior experience and current study focus.

Several problems encountered in early studies were resolved with modifications of trap-nest construction techniques. Small bore trap-nests (1.6 mm-3.2 mm) are split with much difficulty. The splitting plane frequently did not intercept the bore, since the axis of the bore was seldom parallel to the long axis of the wood grain. These problems were solved by the use of pre-split trap-nests. Stages in the construction of pre-split trap-nests are illustrated in Figure 1. Several steps were required for their construction. A band saw was used to cut trap-nest blocks length-wise into two sections with dimensions nominally 6.4 x 19 x 140 mm and 12.6 x 19 x 140 mm. A drill-guide channel was routed in a longitudinal face of the larger section. Trap-nest sections were bound together with masking tape and drilled to selected depths and diameters with high speed twist-steel bits.

Pre-split trap-nest sections did not fit well together. Irregularities across split surfaces admitted light and excess moisture, both detrimental to trap-nest use. These difficulties were eliminated by modifying a technique from Krombein (1967). Pre-split trap-nests were coated with melted paraffin and then re-drilled to their appropriate bore diameter and depths. Paraffin provided water-proof seal across split surfaces and re-drilling removed paraffin that blocked the bore and produced exceptionally smooth bore surfaces. Completed trap-nests were bound together into trap-nest bundles of nine (3 x 3), twelve (3 x 4), or twenty (4 x 5) trap-nests. Cotton cord, rubber bands and plastic strapping were used to secure individual trap-nests in bundles. Fye's (1965) design for a bundle carrier was used to place bundles in the field and fence staples were used to attach bundles to the trunks of trees in the study area. A typical trap-nest bundle is illustrated in Figure 2.

---

1Natural Science and Mathematics Division, Concordia College, Ann Arbor, MI 48105.
Figure 1. Construction of pre-split trap-nests: A—pine trap-nest block; B—a 6.4 mm section removed from side of trap-nest; C—60° channel routed in trap-nest face; D—pre-split trap-nest with bore drilled out.

Figure 2. A 3 by 3 bundle of trap-nests ready for placement in the field.

LITERATURE CITED


TRAP-NEST BORE DIAMETER PREFERENCES AMONG SYMPATRIC PASSALOECUS SPP. (HYMENOPTERA: SPHECIDAE)

John M. Fricke

ABSTRACT

Five species of Passaloecus used trap-nests in a study area in southern Michigan. Significant differences in trap-nest bore diameter selection were noted among P. annulatus, P. areolatus, P. cuspidatus, and P. monilicornis. P. annulatus and P. areolatus selected bore diameters 1.6 to 2.4 mm, P. cuspidatus used bore diameters of 2.0 to 6.4 mm, and P. monilicornis selected diameters of 1.6 to 3.6 mm.

It is questionable whether previous reports of trap-nesting data indicate bore diameter preferences of Passaloecus spp. or that they are artifacts due to the bore diameter selection of the investigators. Fye (1965), Krombein (1967), and Vincent (1978) reported on Passaloecus spp. and gave data on bore diameters selected. Vincent (1978) noted P. annulatus (Say) reared from a 1.5 mm bore trap-nest and P. areolatus Vincent from two 1.5 mm bore trap-nests. All of these authors reported trap-nest bores used by P. cuspidatus Smith and their pooled data (bore diameters and numbers of nests) are as follows: 3.2 mm—20; 4.0 mm—83; and 6.4 mm—3. Fye (1965) reported that P. monilicornis Dahlbom preferred 6.4 mm bores and Krombein (1967) noted P. monilicornis from four 3.2 mm and two 4.8 mm borings.

Bore diameters most frequently used by these authors were 4.8 mm or greater, with increments of 1.6 mm. Fye used 6.4 and 8.0 mm drillings; Krombein also used these sizes and included a few 3.2 mm bores. Ratios or actual frequencies were not reported. Vincent was the only author to report use of bores as small as 1.5 mm. Passaloecus are small wasps (4–9 mm long) and bore diameters used in trap-nesting survey studies may be inappropriate for studies focused on this genus, because bore sizes usually presented may be too large to be used by the majority of these small wasps.

MATERIALS AND METHODS

Bore diameter preferences among Passaloecus spp. were investigated from 1984 through 1987 on the campus of Concordia College, Ann Arbor, Michigan. Trap-nest stations were established in a mixed hardwood forest edge between a small red pine plantation and an old field. The long axis of the edge ran from north-west to south-east. Bundles of trap-nests were positioned so that bore openings faced north-west,
Table 1. Bore diameters, bundle configurations, and bundle heights used in studies of *Passaloecus* trap-nest bore diameter preferences, 1984–1987.

<table>
<thead>
<tr>
<th>Year</th>
<th>Bore Diameter (mm)</th>
<th>Number of Bundles &amp; Bundle Configuration</th>
<th>Staitiaons</th>
<th>Bundle Heights (meters)</th>
</tr>
</thead>
<tbody>
<tr>
<td>1984a</td>
<td>3.2–4.8</td>
<td>48(3 x 3)</td>
<td>Juglans</td>
<td>0.5–1.5</td>
</tr>
<tr>
<td>1984b</td>
<td>3.2–8.0</td>
<td>17(3 x 3)</td>
<td>Juglans</td>
<td>0.5–1.5</td>
</tr>
<tr>
<td>1985</td>
<td>1.6–8.8</td>
<td>64(4 x 5)</td>
<td>Juglans</td>
<td>0.5–2.0</td>
</tr>
<tr>
<td>1986a</td>
<td>1.6–4.8</td>
<td>96(3 x 4)</td>
<td>Populus Fraxinus Prunus</td>
<td>0.5–2.0</td>
</tr>
<tr>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>1986b</td>
<td>1.6–4.8</td>
<td>36(3 x 4)</td>
<td>Juglans Fagus</td>
<td>1.0–9.0</td>
</tr>
<tr>
<td>1987</td>
<td>2.4–7.2</td>
<td>147(3 x 4)</td>
<td>Juglans Pinus</td>
<td>0.75–1.75</td>
</tr>
</tbody>
</table>

Table 2. Bore diameter selections for five *Passaloecus* spp., 1984–1986.

<table>
<thead>
<tr>
<th>Bore Diameters (mm)</th>
<th>1.6</th>
<th>2.0</th>
<th>2.4</th>
<th>2.8</th>
<th>3.2</th>
<th>3.6</th>
<th>4.0</th>
<th>4.4</th>
<th>4.8</th>
<th>5.6</th>
<th>6.4</th>
</tr>
</thead>
<tbody>
<tr>
<td>Bore Diameter Frequencies</td>
<td>256</td>
<td>128</td>
<td>256</td>
<td>128</td>
<td>430</td>
<td>128</td>
<td>430</td>
<td>128</td>
<td>430</td>
<td>128</td>
<td>158</td>
</tr>
</tbody>
</table>

Species

- *annulatus* (Say) 3 1 7 0 0 0 0 0 0 0
- *areolatus* Vincent 18 20 9 0 0 0 0 0 0 0
- *cuspidatus* Smith 0 1 4 6 32 7 19 3 21 0 1
- *monilicornis* Dahlbom 2 3 13 2 2 2 0 0 0 0 0
- *singularis* Dahlbom 0 0 1 0 0 0 0 0 0 0

north-east, south-east, and south-west. Trap-nests were arranged in bundles presenting a mixture of regular and randomized patterns of drilled and blank trap-nest faces. Trap-nest design has been previously described (Fricke, 1991). Bore diameters, bundle configurations, stations, and bundle heights are summarized in Table 1.

Table 2 summarizes data for 1984–1986 on frequency of bore diameter availability and selection as nesting sites by five *Passaloecus* spp. The Kruskal-Wallis test for differences in ranks of trap-nest bore selection by four of these species (*P. annulatus, P. areolatus, P. cuspidatus, and P. monilicornis*) is very significant ($H = 120.9749$, df $= 3$, $p < .0005$). The chi-square (I) test for differences of bore diameter selection by *P. cuspidatus* (based upon three bore diameter classes: 2.0–2.8, 3.2–4.0, and 4.4–6.4 mm) and the t(II) test for differences of bore diameter preferences between *P. cuspidatus* and *P. monilicornis* are both very significant ($\chi^2 = 15.2583$, df $= 2$, $p < .0005$ and $t = 7.4316$, df $= 116$, $p < .0005$).

*P. cuspidatus* trap-nest selection data from 1984–1987 were pooled for analysis and are given in Table 3. Expected and observed frequencies of bore selection are significantly different indicating that *P. cuspidatus* prefers trap-nest bore diameters ranging from 2.8 to 4.8 mm.

<table>
<thead>
<tr>
<th>Bore Diameter Class (mm)</th>
<th>Diameter Frequency</th>
<th>Expected Selection Frequency</th>
<th>Observed Selection Frequency*</th>
</tr>
</thead>
<tbody>
<tr>
<td>2.0–2.4</td>
<td>595</td>
<td>27.17</td>
<td>14</td>
</tr>
<tr>
<td>2.8–3.2</td>
<td>865</td>
<td>39.50</td>
<td>75</td>
</tr>
<tr>
<td>3.6–4.0</td>
<td>865</td>
<td>39.50</td>
<td>54</td>
</tr>
<tr>
<td>4.4–4.8</td>
<td>865</td>
<td>39.50</td>
<td>32</td>
</tr>
<tr>
<td>5.6–6.4</td>
<td>708</td>
<td>32.33</td>
<td>3</td>
</tr>
</tbody>
</table>

\[X^2 = 71.6456, \text{df} = 4, p < .0005\]

**DISCUSSION**

A possible factor influencing bore diameter selections among *Passaloecus* spp. is wasp size. A relatively simple index to wasp size is head width. Head width measurements, to the nearest 0.1 mm, were taken from samples of ten females of *P. cuspidatus*, *P. monilicornis*, and *P. areolatus*. The respective average head widths for these samples were 1.46, 1.19, and 1.0 mm. *Passaloecus* spp. appear to partition nesting sites on the basis of bore diameter and wasp size may limit the minimum acceptable bore diameter. An additional factor in this regard may be the size of aphids selected as prey. Aphids are carried in the mandibles with the prey's body lying below the wasp's head. Under these circumstances the dorsal-ventral dimension of the wasp's head plus an aphid will be greater than head width and may possibly influence acceptable bore diameters.

**LITERATURE CITED**


INSTRUCTIONS FOR AUTHORS

Papers dealing with any aspect of entomology will be considered for publication in *The Great Lakes Entomologist*. Appropriate subjects are those of interest to professional and amateur entomologists in the North Central States and Canada, as well as general papers and revisions directed to a larger audience while retaining an interest to readers in our geographic area.

All manuscripts are refereed by two reviewers, except for short notes, which are reviewed at the discretion of the Editor. Manuscripts must be typed, double-spaced, with 1" margins on 8 1/2 x 11" or equivalent size paper, and submitted in triplicate. Please underline only those words that are to be italicized. Use subheadings sparingly. Footnotes (except for author's addresses, which must be on the title page, and treated as a footnote), legends, and captions of illustrations should be typed on separate sheets of paper. Titles should be concise, identifying the order and family discussed. The author of each species must be given fully at least once in the text, but not in the title or abstract. If a common name is used for a species or group, it should be in accordance with the common names published by the Entomological Society of America. The format for references must follow that used in previous issues of *The Great Lakes Entomologist*. Literature cited is just that—no unpublished manuscripts or internal memos. Photographs should be glossy finish, and mounted on stiff white cardboard (transparencies are not acceptable). Drawings, charts, graphs, and maps must be scaled to permit proper reduction without loss of detail. Please reduce illustrations or plates to a size no greater than 9 x 12" to permit easier handling. Attach a figure number to the reverse side of each figure and include the authors' names. Unsuitably mounted photographs or poor figures will be returned to authors for revision.

Tables should be kept as uncluttered as possible, and should fit normally across a page when typeset by the printers. Contributors should follow the *Council of Biology Editors Style Manual*, 5th ed., and examine recent issues of *The Great Lakes Entomologist* for proper format of manuscripts.

Manuscripts may also be submitted on computer disk (MS-DOS, Apple II, or Macintosh format) along with a printed copy, after they have been accepted for publication. Authors are especially encouraged to do so if their manuscript is longer than 2 printed journal pages, or if they are without funds for page costs.

Papers published in *The Great Lakes Entomologist* are subject to a page charge of $30.00 per published page. Members of the Society, who are authors without funds from grants, institutions, or industry, and are unable to pay costs from personal funds, may apply to the Society for financial assistance. Application for subsidy must be made at the time a manuscript is initially submitted for publication.

Authors will receive page proof, together with an order blank for separates. Extensive changes to the proof by the author will be billed at a rate of $1.00 per line.

All manuscripts for *The Great Lakes Entomologist* should be sent to the Editor, Mark F. O'Brien, Insect Division, Museum of Zoology, The University of Michigan, Ann Arbor, MI, 48109-1079, USA. Other correspondence should be directed to the Executive Secretary (see inside front cover).