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## COVER ILLUSTRATION

A mourning cloak butterfly, *Nymphalis antiopa* (L.) at Ludington State Park, MI. Photograph by M. F. O'Brien
THE MICHIGAN ENTOMOLOGICAL SOCIETY

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ABSTRACT

Phyllophaga adults (June beetles) were surveyed from 1984 through 1987 at five locations in Wisconsin using blacklight traps. Data were collected at each location for three consecutive years. A total of 9,259 specimens representing 13 species were collected during the survey. Species captured, sex ratios, and flight periods for abundant species were recorded for each location. Species abundance differed from previous surveys of Phyllophaga in Wisconsin. Possible reasons for observed shifts in species abundance are discussed.

Phyllophaga larvae (white grubs) feed upon the roots of turfgrasses and other desirable plants, whereas adults (June beetles) feed on the foliage of trees and shrubs. In Wisconsin, most Phyllophaga are thought to have a three year life cycle, spending most of it as larvae in the soil (Luginbill and Painter 1953). Much is known about adult host plant preferences (Forbes 1916, Travis 1933, Ritcher 1940, Chamberlin et al. 1943), but although Ritcher (1940) made some general observations on white grub hosts in Kentucky, the host relationships of Phyllophaga larvae are poorly understood.

Since the last study of Wisconsin Phyllophaga (Chamberlin et al. 1943), agricultural practices and land use patterns have changed considerably. Major differences include increased use of persistent and non-persistent soil insecticides, modified weed control practices, increased irrigation in some areas, adoption of crop rotation, shifts in pasture forage species, adjustments in fertilization practices, and escalating suburbanization. These and other alterations in land use could seriously impact the species composition of the soil insect fauna, including white grubs.

Changes in the species complex of Phyllophaga within a region may be a reflection of modifications in larval and/or adult habitats. Proximity to adult host plants and soil moisture are two major factors influencing Phyllophaga populations (Forbes 1916, Sweetman 1931).

Light traps are commonly used to survey or monitor Phyllophaga adults (Sanders and Fracker 1916, Forbes 1916, Travis 1933, Henry and Heit 1940, Neiswander 1963, Lim et al. 1979). This paper reports a light trap study in Wisconsin which surveyed the species of Phyllophaga and their relative abundance at five different locations in the state. Goals of the study were to: (1) examine for species shifts since earlier studies and (2) analyze for possible periodicity in adult emergence. Additionally, we review the brood theory concept as it relates to Phyllophaga yearly abundance.

1Department of Entomology, University of Wisconsin, 1630 Linden Drive, Madison, WI 53706.
MATERIALS AND METHODS

Four-baffled blacklight traps (Ellisco Inc. cat. no. 110103-2) equipped with ultraviolet fluorescent bulbs (18" Norelco 15 watt bulb F15 T8/BL) were used to monitor *Phyllophaga* adults at five locations in Wisconsin from 1984 to 1987. Three consecutive years of trapping were conducted at each location. From 1984 through 1986, single traps were operated continuously from mid-April to mid-July on each of three suburban golf courses in Madison, Brookfield, and Menasha (Figure 1). From 1985 through 1987, two traps were operated in Mazomanie and Lancaster, agricultural locations in the southern part of the state. A dichlorvos-impregnated strip in the collecting pan of each trap killed the trapped insects.

Trap catches were collected weekly, placed in labeled paper bags and stored in a refrigerator until processed. *Phyllophaga* were sorted according to species, sexed and counted. Identification of species was according to the keys of Luginbill and Painter (1953). Voucher specimens from this study are deposited in the Insect Research Collection, Department of Entomology, University of Wisconsin, Madison.
RESULTS AND DISCUSSION

Species Collected

Thirteen species and 9,259 specimens of Phyllophaga were collected during the four years of the study (Table 1). All 13 species had been previously recorded for Wisconsin although previous studies recorded greater diversity of Phyllophaga than our study. For example, Sanders and Fracker (1916) collected 17 species at five Wisconsin collection sites with light traps whereas Chamberlin et al. (1943) collected 19 species at 39 different locations in Wisconsin by handpicking beetles from host plants at night. Data from Iowa (Travis 1933) and northern Illinois (Forbes 1916) also showed greater diversity of Phyllophaga than our study. The lower species diversity observed in our study may have been due in part to the restricted number of habitats associated with our survey locations.

The most commonly collected species in our study was P. jutius LeConte, which accounted for 71% of the total. The Madison sites surveyed previously (Sanders and Fracker 1916) and during our study show differences in species abundance. P. rugosa Melsheimer and P. fusca Froelich comprised 99% of the June beetles collected by Sanders and Fracker (1916) at Madison but in our study, P. jutius and P. anxia LeConte collectively accounted for 87% of the June beetles captured in Madison. Conversely, P. rugosa and P. fusca represented only 5.9% of our total and P. jutius and P. anxia constituted <1% of the total caught by Sanders and Fracker (1916). Several reasons may explain the difference in species abundance between our study and that of Sanders and Fracker (1916). First, it is highly probable that our light trap locations within Madison differed from those of the 1916 study. Second, the type of adult host trees and their proximity to light traps may have influenced the array of Phyllophaga species caught (e.g. Forbes 1916). Most importantly, significant changes in land use patterns have occurred in Madison over the 70 years between studies and it is likely that habitats of larvae and/or adults were altered during this time. Our Madison location was on a golf course adjacent to the University of Wisconsin Arboretum, a vegetationally diverse area where many native Wisconsin plant communities are recreated on one tract of land. However, data from a light trap on another golf course in the Madison area showed the same two most abundant Phyllophaga species (P. jutius and P. anxia) for a two year period (1985 and 1986) (unpublished). The most abundant species at each location in our study, except Mazomanie, was P. jutius (Table 1). The most abundant species at the Mazomanie location was P. implicita Horn. Each location had a different species complex and only three species were common to all five trap locations. Shenefelt and Simkover (1951) found that the Phyllophaga species composition differed greatly between localities and concluded that these differences were a manifestation of the environmental conditions under which the insects exist (e.g. edaphic factors).

Sex Ratios of Phyllophaga Taken at Light Traps

Sex ratios for the most abundant species in the present study, given as the fraction of males, are as follows: P. anxia 0.88 (n = 75); P. crenulata Froelich 0.97 (n = 62); P. fusca 0.76 (n = 71); P. jutius 0.94 (n = 6214); P. gracilis Burmeister 0.75 (n = 16); P. hirticula 0.89 (n = 28); P. implicita 0.52 (n = 1423); P. rugosa 0.90 (n = 919). Generally, light traps capture more male Phyllophaga than females (e.g. Travis 1933, Lim et al. 1979) although some studies report differing results concerning sex ratios for certain species. For example, in three light trap studies involving P. anxia, Morofsky (1933) collected more females than males while Neiswander (1963) and Sanders and Fracker (1916) collected more males than females. In our study P. anxia captures were male biased. Only P. implicita had a nearly equal sex ratio and reasons for this were not apparent. Several factors could contribute to differences in sex ratios using light traps. For example, differences in flight habits and differential phototaxis (wavelength of light used in trap) among the sexes are two factors which could affect sex ratios.
Table 1. *Phyllophaga* spp. beetles collected at light traps in Wisconsin, 1984-1987.

<table>
<thead>
<tr>
<th>Phyllophaga species</th>
<th>BROOKFIELD</th>
<th>MADISON</th>
<th>MENASHA</th>
<th>MAZOMANIE</th>
<th>LANCASTER</th>
</tr>
</thead>
<tbody>
<tr>
<td>fuutilis</td>
<td>196 129 119</td>
<td>98 306 30</td>
<td>487 1046 345</td>
<td>358 3 0</td>
<td>1291 220 1944</td>
</tr>
<tr>
<td>rugosa</td>
<td>4 9 8</td>
<td>0 27 1</td>
<td>3 30 40</td>
<td>79 1 9</td>
<td>640 2 145</td>
</tr>
<tr>
<td>anxia</td>
<td>0 1 1</td>
<td>9 20 22</td>
<td>0 7 1</td>
<td>11 2 1</td>
<td>0 0 0</td>
</tr>
<tr>
<td>fusca</td>
<td>0 1 0</td>
<td>5 0 0</td>
<td>0 0 0</td>
<td>8 0 3</td>
<td>26 5 25</td>
</tr>
<tr>
<td>implicita</td>
<td>0 0 1</td>
<td>1 5 0</td>
<td>0 0 1</td>
<td>103 271 977</td>
<td>15 5 44</td>
</tr>
<tr>
<td>crenulata</td>
<td>0 0 0</td>
<td>14 4 6</td>
<td>0 0 0</td>
<td>8 11 15</td>
<td>0 0 4</td>
</tr>
<tr>
<td>tristis</td>
<td>0 0 1</td>
<td>1 2 1</td>
<td>0 0 0</td>
<td>0 0 0</td>
<td>0 0 0</td>
</tr>
<tr>
<td>hirticula</td>
<td>0 0 0</td>
<td>0 0 0</td>
<td>0 0 0</td>
<td>0 0 0</td>
<td>0 0 0</td>
</tr>
<tr>
<td>baltia</td>
<td>0 0 0</td>
<td>2 0 0</td>
<td>0 0 0</td>
<td>1 0 0</td>
<td>10 0 18</td>
</tr>
<tr>
<td>gracilis</td>
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<td>0 0 0</td>
<td>0 0 0</td>
<td>2 0 14</td>
<td>0 0 0</td>
</tr>
<tr>
<td>inversa</td>
<td>0 0 0</td>
<td>0 0 0</td>
<td>0 0 0</td>
<td>0 0 1</td>
<td>1 0 0</td>
</tr>
<tr>
<td>forsteri</td>
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<td>0 0 0</td>
<td>0 0 0</td>
<td>1 0 0</td>
<td>0 0 0</td>
</tr>
<tr>
<td>prunina</td>
<td>0 0 0</td>
<td>0 1 0</td>
<td>0 0 0</td>
<td>0 0 0</td>
<td>0 0 0</td>
</tr>
<tr>
<td>total</td>
<td>200 140 130</td>
<td>130 365 60</td>
<td>490 1083 387</td>
<td>571 288 1020</td>
<td>1983 232 2180</td>
</tr>
</tbody>
</table>
Seasonal Flight Periods and Yearly Trap Results

The three most abundant Phyllophaga species in our study were *P. jutilis*, *P. implicita*, and *P. rugosa*. Other Phyllophaga species (Table 1) were not well represented and are not discussed. The three most abundant species have similar adult seasonal activity periods. The fourth week of April through the end of June brackets the period of highest activity of adult *Phyllophaga* species. Many environmental factors (such as soil moisture and temperature, air temperature, and local rain fall patterns) potentially influence developmental rate or flight activity. Such factors could result in different flight periods for a species in areas of close proximity. For example, in 1985 Mazomanie and Lancaster had similar flight activity periods for *P. jutilis* (Fig. 2) while the activity periods for *P. implicita* in 1987 at the same two locations were quite different (Fig. 3).

Peak captures of *P. jutilis* in 1984 occurred during the same time period for the three golf course locations (Fig. 2). In 1985 all trap locations had earlier flight activity of *P. jutilis* than other years; the two agricultural locations had earlier peak flights than the three golf course sites (Fig. 2). Flight activity of *P. jutilis* in 1986 was
more variable temporally than the previous two years. Both agricultural locations had low numbers of *P. futilis* captured in 1986. Total *P. futilis* collected at all sites was lowest in 1986, except at Mazomanie (Table 1). In 1987, Lancaster had the largest capture of *P. futilis* for any year or location, whereas Mazomanie had no *P. futilis* captured in 1987, the lowest for any year at any location during the study.

Abundance of *P. rugosa* was greatest in 1985 at all sites except Menasha (Table 1). Captures were very small in 1984 and 1986 at all locations, Menasha excluded. Captures differed substantially in 1987 between Mazomanie and Lancaster. The total capture of *P. rugosa* at Lancaster in 1987 was substantially greater than at Mazomanie (Fig. 4).

Seasonal flight periods were similar for *P. rugosa* at Brookfield and Menasha in 1984 (Fig. 4). In 1985, seasonal flight periods were earlier than 1984 (for Brookfield and Menasha) and similar for all locations except Mazomanie which peaked later (Fig. 4). Peak flight periods in 1986 were similar among the golf course locations and 1987 peak flight periods were similar between the two agricultural locations (Fig. 4).

*P. implicita* was collected at all five sites but was well represented only at the two agricultural locations (Table 1). More *P. implicita* were captured yearly at Mazomanie than at Lancaster, and Mazomanie trap catches increased with each year of trapping (Table 1). Peak captures of *P. implicita* were generally earlier at Mazomanie for a given year (Fig. 3). Although *P. implicita* was captured mainly at the agricultural locations, the total number captured represented >15% of the 9,259 beetles collected.

**The Brood Theory in Phyllophaga**

Several authors have alluded to a three year synchronous life-cycle ("broods") for *Phyllophaga* (Neiswander 1963, Hammond 1954, Ritcher 1940). The idea of synchronous and periodic emergence of June beetles may have originated from European work. For example, Forbes (1916) briefly mentioned a European origin for forecasting white grub outbreaks but lists no references. Unfortunately the notion of synchronicity with respect to *Phyllophaga* populations has been perpetuated without sound data. Neiswander (1963) claimed that entomologists have "essentially
accepted the early alignment of broods without subsequent study of populations."
The life cycles of *Phyllophaga* species generally exhibit a periodicity of two to four years but individuals of the same species may differ greatly (e.g. Davis 1916). Determination of life cycle length in *Phyllophaga* is complex; the interplay of temperature, food quality and quantity, soil conditions and larval population density may all be involved. In particular, cumulative seasonal soil temperatures can have a strong influence on developmental time. For instance, Davis (1916) found *P. drakii* Kirby to have a three year life cycle at Lafayette, Indiana and a four year life cycle at Trout Lake, in northern Wisconsin. Furthermore, he pointed out that the combined seasonal temperatures in Lafayette for three years were approximately the same as for four years at Trout Lake. Additionally, Shenefelt and Simkover (1951) found *P. tristis* Fabricius to have a two year life cycle in central Wisconsin but a one year life cycle at the same latitude in Michigan. Michigan's more moderate climate may account for the shorter life cycle. The roles of food quality and larval density are more difficult to assess. These factors could, separately or together, account for differences in life cycle length for individuals of the same species in the same location. Lloyd and White (1976) proposed that *Magicicada* species might emerge four years early with crowding of early instar nymphs. These factors make prediction of destructive broods difficult except on a very local basis.

Mahr (1984) predicted that 1986 would be a major flight year for *P. rugosa* in Wisconsin. No trap location in this study supported that prediction. Although the Menasha location had the largest capture of *P. rugosa* in 1986, the total was fairly insignificant and *P. rugosa* was not the most abundant species in any year. The literature shows discrepancies in major flight years and relative abundance for a given
species in different areas (Forbes 1916, Sanders and Fracker 1916). It is therefore difficult, if not impossible, to compare results of different studies in predicting *Phyllophaga* outbreaks, even in the same locale. In some areas a shift in the most abundant brood can occur over several years (Neiswander 1963). The supposition that most June beetles have synchronous life cycles appears to be a dubious generality on a regional basis. In restricted geographical areas populations of *Phyllophaga* may be temporarily synchronous due to localized environmental conditions. However, it is likely that this synchronicity breaks down through time and the concept should not be extrapolated on a larger geographical or temporal scale.

**ACKNOWLEDGMENTS**

Funding for this study was provided, in part, by the Wisconsin Turfgrass Association and the University of Wisconsin College of Agricultural and Life Sciences, and by Hatch Grant nos. 2011 and 2566. We thank Ed Arnold of the Wisconsin Dept. of Agriculture, Trade and Consumer Protection and the following golf course superintendents: Randall Smith, Nakoma Country Club, Madison; Roger Bell, Northshore Golf Club, Menasha; Jerry Kushasky, Westmoor Country Club, Brookfield. We also thank Phil Pellitteri for assistance in identification of many June beetles.

**LITERATURE CITED**


ACOUSTIC SIGNALS OF *GRAMINELLA NIGRIFRONS*  
(HOMOPTERA: CICADELLIDAE)  

S. E. Heady and L. R. Nault

**ABSTRACT**

The deltocephaline leafhopper, *Graminella nigrifrons*, produces low intensity substrate transmitted vibrations (signals) to facilitate location of virgin females by males during courtship. In the laboratory, signals produced on maize leaves were received by a phonographic cartridge, amplified, and analyzed on an oscillograph and sonograph. Male calls, that are produced spontaneously, are complex, consisting of three consecutive sections. Section 1 consists of ca. 3 sec of irregular clicks. Section 2 has ca. 4 sec of repeated phrases consisting of a continuous series of 0.4 sec chirps and a roll. Section 3 consists of ca. 5 sec of an intermittent series of 0.2 sec chirps and a roll. Female calls are produced in response to male calls. Female calls are simple compared to male calls and consist of ca. 4–5 sec of low frequency clicking. Signal patterns of *G. nigrifrons* are compared to those of other leafhoppers and evolutionary scenarios are presented to account for the observed gender differences in signals.

The black faced leafhopper, *Graminella nigrifrons* (Forbes), is one of the most common leafhoppers occurring in grasslands of the Eastern United States (Kramer 1967). *G. nigrifrons* is the primary vector of the virus causing one of the most important stunting diseases of maize (*Zea mays* mays) in the United States. The pathogen, initially described as the 'Ohio corn stunt agent' by Rosenkranz (1969), was later identified as maize chlorotic dwarf virus (MCDV) (Gingery 1988 and references therein). In the corn belt states, MCDV has been reported in Ohio, Illinois, and Indiana, but is generally a problem only in areas where its alternate host, johnsongrass (*Sorghum halepense* L.), occurs (Gordon et al. 1981). Use of tolerant maize genotypes can reduce losses in some years and locations. MCDV remains the most destructive and widespread maize virus in the Southeastern and adjacent maize growing regions of the United States (Gordon and Nault 1977).

The biology of *G. nigrifrons* and its role as the vector of MCDV are well documented. Studies have been conducted on field biology (Boyd and Pitre 1968, Stoner and Gustin 1967), host range (Boyd and Pitre 1969), host utilization (Hunt and Nault 1990, Larsen et al. 1990), distribution (Douglas et al. 1965, Durant 1968, Durant and Hepner 1968), and vector relationships (Nault et al. 1973, Choudhury and Rosenkranz 1983, Knoke et al. 1983). The reproductive biology of *G. nigrifrons* has been studied in terms of laboratory life tables (Sedlecek et al. 1986); however, studies on the mating behavior were lacking. Leafhoppers were discovered to produce low intensity substrate signals by Ossiannilsson (1949). These signals are integral in courtship and facilitate males locating virgin females for copulation. To aid
our understanding of the mating behavior of *G. nigrifrons*, male and female signals are described herein. Other aspects of the mating behavior, particularly those that may influence vector movement and disease spread, were conducted concurrently (Hunt 1988).

MATERIALS AND METHODS

Acoustic recordings were made using adults taken from laboratory colonies kept in rearing cages (D'Arcy and Nault 1982) at 26–28°C with a photoperiod of 12:12 (L:D). Late-instar nymphs were individually separated to obtain virgin adults for recording (Heady et al. 1986). To record signals, a leafhopper was placed on a maize leaf piece (ca. 15 cm length) and quickly covered with a plastic dome (1 cm diam) (Heady 1987). The leaf piece was laid over a phonographic cartridge (ElectroVoice 5146) so that it lightly touched the needle. Signal output from the cartridge was sent to a preamplifier (Omega EQ-25), a DC amplifier at 100x (Dana 3640), displayed on an oscilloscope (Dumont/Fairchild 766), monitored with earphones, and recorded on a tape recorder (Nagra E) using magnetic tape (3M 250) set at 19 cm/s. Recordings were made in a laboratory where temperatures ranged from 25 ± 2°C. Calls on acoustic tape were printed using a pen and ink polygraph (Grass Model 7, D.C. driver amplifier) (Heady et al. 1986). For male *G. nigrifrons*, the average duration of a spontaneous call was measured from 30 randomly chosen individuals. Characterizations of portions of the male signals were analyzed from oscillograms of five calls per individual and 10 individuals using a repeated measures analysis of covariance, with temperature as the covariable. Calls of female *G. nigrifrons* were analyzed from eleven individuals. Frequency spectra were printed using a DSP Sona-Graph (Model 5500, Kay Elemetrics Corp.).

Call terminology is described based on onomatopoeic interpretation and by pulse structure (Alexander 1967, Booij 1982, Heady et al. 1986, Huber et al. 1989). Leafhopper call repertoire includes male calling signals and male and female courtship signals. These calls are made up of sections composed of phrases, which are themselves made up of chirps, clicks, rolls, and intervals of silence. Chirps are distinct sounds to the human ear and are composed of simple or complex pulses of sound. Clicks have shorter durations than chirps. A roll is composed of a chirp and 3–6 pulses.

RESULTS

*G. nigrifrons* males spontaneously produced calling signals with mean durations of 15.2 (S.E. = 1.4, N = 30) sec. In the presence of a responding female, the male would call almost continuously as he walked to the female and joined genitalia. The male calling signal of *G. nigrifrons* was composed of three sections (Fig. 1A). Section 1 consisted of irregular clicks which were produced with increasing frequency over ca. 3 sec. Section 2 consisted of ca. 4 sec of repeated phrases consisting of a continuous series of 5–9 chirps and a roll (Fig. 1B). Section 3 has ca. 5 sec of repeated phrases consisting of an intermittent series of 1–4 chirps and a roll (Fig. 1C). The repeating phrase in section 2 was longer than the repeating phrase in section 3, 0.4 and 0.2 sec, respectively (Table 1). The rolls found in section 2 and 3 were not significantly different in duration and pulse rate (Table 1). Additionally, the rate of roll production (rolls/msec) in section 2 and 3 was not significantly different (P > 0.20). Thus the main difference between section 2 and 3 was the chirping before rolls. When the chirps, rolls, and silent interval between phrases were totaled (Table 1, phrase duration + phrase interval) and compared for section 2 and 3, section 3 phrases (x = 597.8, S.E. = 9.9) were only slightly longer than section 2 phrases (x = 537.5, S.E. = 17.2). Temperature did not significantly (P >
Table 1. — Male calling signal variables of *Graminella nigrijrons*.

<table>
<thead>
<tr>
<th>Variable</th>
<th>Section 2a</th>
<th>x (S.E.)</th>
<th>Section 3a</th>
<th>x (S.E.)</th>
</tr>
</thead>
<tbody>
<tr>
<td>Phrase Duration (msec)</td>
<td>449.4 (15.7)</td>
<td></td>
<td>215.8 (4.5)</td>
<td></td>
</tr>
<tr>
<td>Phrase Interval (msec)</td>
<td>88.1 (10.2)</td>
<td></td>
<td>382.0 (10.6)</td>
<td></td>
</tr>
<tr>
<td>Chirp Rate (chirps/sec)b</td>
<td>0.02 (0.0003)</td>
<td></td>
<td></td>
<td>0.15</td>
</tr>
<tr>
<td>Roll Duration (msec)</td>
<td>64.6 (1.1)</td>
<td></td>
<td>64.6 (0.9)</td>
<td></td>
</tr>
<tr>
<td>Roll Pulse Rate (pulses/sec)b</td>
<td>0.15 (0.0044)</td>
<td></td>
<td>0.15 (0.0066)</td>
<td></td>
</tr>
<tr>
<td>Roll Rate (rolls/sec)b</td>
<td>2.00 (0.043)</td>
<td></td>
<td>1.87 (0.064)</td>
<td></td>
</tr>
</tbody>
</table>

a Corresponds to Fig. 1.

b Values are temperature corrected for 24°C.

0.20) affect duration of phrases or intervals between phrases, but did impact on the pulse rate of chirping in section 2, pulse rate in the rolls, and rate of roll production (P<0.05). The variation in calls among individuals for all variables analyzed was 2-7 times higher than the variation in calls within individuals. The frequency spectrum of calls of male *G. nigrijrons* consisted of frequency bands of energy concentrated between DC to 2000 Hz, with dominant frequencies at ca. 250 and 390 Hz.

In courtship, male calling began with section 1 type calls followed by section 2 and section 3 type calls which were repeated until copulation. Females called only when males produced section 2 and 3 calls. Calls of female *G. nigrijrons* consisted of low frequency (dominant frequencies at 170 and 250 Hz) clicking sounds. Average durations of female pulses were 15.4 msec (S.E. = 0.8, N = 11) and average intervals of 778.1 msec (S.E. = 106.3, N = 11)(Fig. 2).

**DISCUSSION**

Leafhoppers and planthoppers produce vibrational signals by means of a sound-producing organ similar to the cicada timbal organs in the first and second abdominal segments (Ossiannilsson 1949, Mitomi et al. 1984). The signals are transmitted through the plant substrate on which the insect is sitting (Ichikawa and Ishii 1974, Michelsen et al 1982). Males and females alternate calls and the male walks to the female whereby copulation may occur. The mechanism for male mate location has been described as phonoklinotaxis for the leafhopper, *Amrasca devastans* (Dist.).(Saxena and Kumar 1984). In contrast, Michelsen et al. (1982) theorized that signal frequency-time domain information might be used to determine the direction, distance, or both to a female. Claridge (1985) proposed that no directional information is received by the male, rather female calling elicits spontaneous random searching movements by males. Hunt (1988) proposed that male *Graminella nigrijrons* receives information on distance but not direction to females. Additionally, Hunt (1988) has demonstrated that phototactic responses in addition to acoustic signaling facilitate male location of females.

When a male *G. nigrijrons* flies or hops to a plant it will produce a call spontaneously. This signal, designated as a calling signal, attraction call, or common call (Ossiannilsson 1949, Heady et al. 1986), is complex and consists of three distinct sections (Fig. 1). The *G. nigrijrons* call becomes more patterned over time. The first portion of the call contains irregularly spaced clicks that increase in amplitude as section 2 calls are produced. In section 3 the signal pattern is well defined. A similar pattern of irregular clicking that changes to a patterned signal is found in the leafhoppers *Aphrodes trijasciatus* (Fourer.) (Ossiannilsson 1949), *Nephoptetix nigropicntus* (Dist.) (Claridge 1988), and short-winged forms of *Macrosteles fascifrons* (Stål) (Purcell and Loher 1976). In the latter species, the male call was described as a trill with gradual transition to chirping. However, other leafhoppers
Figure 1. *Graminella nigrifrons* male calling signal. (A) Entire calling signal composed of three labeled sections. (B) Expanded Section 2 type calls with three repeated phrases. A phrase (a) consists of chirps (b) and a roll (c). (C) Expanded Section 3 type calls with three repeated phrases (a) consisting of chirps (b) and a roll (c). There are 6 pulses in the labeled roll.

studied do not produce a long building call, but rather, discrete chirps. *Amrasca devastans* (Distant) was described as producing "croaking" sounds (Saxena and Kumar 1984). Males of 10 *Dalbulus* species spontaneously produced calls composed of chirps (Heady et al. 1986). For example, *D. maidis* (DeLong and Wolcott) emitted 1-11 repeated chirps spontaneously. Male common calls of 10 *Dalbulus* species were analyzed using cluster analyses and the resulting phenogram was similar to groupings of species based on cladistic analysis of morphological characteristics (Heady et al. 1986). Although calls of members within a genus appeared similar in *Dalbulus*, calls of members among related tribes do not seem similar using the characteristics examined here. *Dalbulus* and *Macrosteles*, members of the tribe Macrosteolini, have very different calls. The call of *G. nigrifrons* is similar to *N. nigropictus*, yet they are members of different Deltocephalinae tribes, Deltocephalini and Euscelini, respectively. Thus calls may be useful in resolving phylogenetic relationships within genera but not at higher taxonomic levels.

Female calls of *G. nigrifrons* were very simple in structure compared to male calls, and were produced only when males were producing section 2 and 3 signals. Section 1 male clicks do not elicit female calling nor any other obvious behaviors such as walking, perhaps because they are not patterned and are not species specific, or because their low amplitude may not be received by the female. Female calls of other leafhoppers are also less complex (lacking different patterned sections) compared to male calls. Examples are *Dalbulus* species (Heady et al. 1986; Heady, unpublished data); *Nephotettix* species (Claridge 1988); *M. fascifrons*, whose female call was described as a buzz (Purcell and Loher 1976); and *A. devastans*, whose call was described as cooing (Saxena and Kumar 1984).
The differences in call complexity between males and females may have evolved as a result of (1) the energetic cost of calling, (2) the risks associated with searching for mates, and/or (3) runaway sexual selection. Integral to these three hypotheses is the inherent asymmetry of reproductive roles between the sexes (West-Eberhard 1984). Male leafhoppers, like many insects, produce large numbers of small gametes (sperm) and multiply mate, whereas females invest more in gametes (large eggs), produce relatively few of them, and mate once or twice. In most crickets, katydids, and cicadas, males alone bear this energetically expensive activity of calling while a mute female finds him (Alexander 1967). In leafhoppers, males bear most of calling expense by producing more or longer calls than females.

Acoustic communication is the most energetically expensive of all forms of communication, i.e., chemical, visual, and tactile (Alcock 1989) and is known to be exploited by predators and parasites due to the ease of locating the signaler. A tachinid fly, *Euphasiopteryx ochracea* (Bigot), was attracted to taped songs of the field cricket *Gryllus integer* Scudder (Cade 1975) and a vertebrate predator (domesticated cat) was observed locating male crickets using their acoustic signals (Walker 1964). Additionally, the sarcophagid fly, *Colcondamyia auditrix* Shewell, acoustically intercepts calling male cicadas (Soper et al. 1976). Specific predators of the leafhopper, *G. nigrifrons* have not been identified. Parasites identified from *G. nigrifrons* include three pipunculid flies, one strepsipteran, two drynid wasps and one encyrtid wasp (Freytag 1987); however, their host-finding strategies have not been elucidated. Generalist insect and spider predators may be attracted to calling male leafhoppers by detecting the vibrations on the plant and by cueing on the
movement of males as they walk to females. Predators and parasites probably exert selective pressures on general male reproductive behaviors including song characteristics (Cade 1975, Zuk 1987a, b). Several studies of insect mating systems have shown that males suffer greater predation and parasitism as a result of their conspicuous displays and movements through habitats in search for females (Gwynne 1987, Thornhill 1978, Burk 1982, Walker and Masaki 1989). Hunt (1988) found that male *G. nigrifrons* flew plant-to-plant, calling on each, until landing on a plant that harbored sedentary but calling virgin females. This "call-and-fly" tactic was previously found in tick-tock cicadas, *Cicadetta quadricincta* and explained the higher incidence of males than females caught in spider webs (Gwynne 1987).

Lastly, the complex calls of males could be the result of rapid or "runaway" evolutionary change (Fisher 1958). If superior signalling is at a premium then there is selection on females to favor superior signallers as mates, because they will produce sons who are superior signallers. This leads to increasing selection on signalling ability and a genetic correlation between female preference and male signalling ability which will accelerate the evolution of both. Runaway change would end when natural selection against too costly or too risky displays balances sexual selection in favor of traits that are appealing to females (Alcock 1989). We expect that male calls of *G. nigrifrons* contain different section types and are elaborate compared to female calls due to female preference and yet are likely constrained by the energetics of calling and the risks associated with mate-finding.

**ACKNOWLEDGMENTS**

We thank Peggy Rhee and Andrea Sweazy (College of Wooster), Rex Alvey (ARS-USDA, OARDC-OSU), L.V. Madden (Dept. of Plant Pathology, OARDC-OSU), and The Borror Bioacoustic Laboratory, OSU for assistance. Salaries and research support provided by State and Federal funds appropriated to the Ohio Agric. Res. and Dev. Center, The Ohio State Univ. This is Journal Article No. 100-90.

**LITERATURE CITED**


NO INTERSEXUAL DIFFERENCES IN HOST SIZE AND SPECIES USAGE IN *SPALANGIA ENDIUS* (HYMENOPTERA: PTEROMALIDAE)

B. H. King

ABSTRACT

*Spalangia endius* were collected from fly pupae, primarily house fly and stable fly, from a poultry house in Indiana. Male and female wasps did not differ within and across host species in host size usage. Also, despite stable fly pupae being significantly smaller than house fly pupae, the proportion of male wasps emerging from the two host species was similar.

Early in the 1900's, entomologists observed that in some species of parasitoid wasps, males tended to emerge from smaller hosts than did females, resulting in a negative relationship between host size and parasitoid sex ratio (proportion males) (reviewed in Flanders 1939, 1946). Since then, a group of sex ratio models, the host quality models have been developed to explain this pattern (Charnov 1979, Charnov et al. 1981, Werren 1984). These models were designed for solitary species (species in which one offspring completes development per host). In these models, the prediction that male parasitoids should emerge from smaller hosts than females is based on the assumption that developing on a small host will be more detrimental to a female than to a male in terms of future ability to reproduce. The rationale of the assumption was that wasps will be smaller when developing on smaller hosts; and even small males may be able to mate successfully, whereas small females probably lay fewer eggs than large females (Charnov et al. 1981). There is some support for this idea in a few species of parasitoid wasps (Charnov et al. 1981, Jones 1982, van den Assem et al. 1989), but not in all species (King 1988). In those species or populations in which the assumption is valid, natural selection is expected to favor females that oviposit a greater proportion of males in small than in large hosts (Charnov et al. 1981). Female wasps can potentially control the sex of their offspring by controlling fertilization because they have haplodiploid sex determination. Under haplodiploid sex determination, males develop from unfertilized eggs and females from fertilized eggs.

Here the host quality models' prediction of a greater proportion of sons from small than from large hosts is tested using field collections of the solitary parasitoid wasp *Spalangia endius* Walker (Hymenoptera: Pteromalidae). The wasps emerged from house fly pupae (*Musca domestica* Linnaeus) and stable fly pupae (*Stomoxys calcitrans* [Linnaeus]) (Diptera: Muscidae) collected from a poultry house in northern Indiana.

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Table 1. — X ± SD (n) size (mm³) of house fly pupae from which male and female Spalangia endius emerged for five collection dates.

<table>
<thead>
<tr>
<th>Date</th>
<th>Male wasps</th>
<th>Female wasps</th>
<th>Test Comparison</th>
</tr>
</thead>
<tbody>
<tr>
<td>Sep 03</td>
<td>26.24 ± 3.09 (38)</td>
<td>25.86 ± 3.02 (110)</td>
<td>t = 0.66, P = 0.51</td>
</tr>
<tr>
<td>Sep 10</td>
<td>28.15 ± 2.27 (3)</td>
<td>25.16 ± 2.94 (14)</td>
<td>t = 1.65, P = 0.12</td>
</tr>
<tr>
<td>Oct 01</td>
<td>16.57 ± 5.04 (14)</td>
<td>18.51 ± 5.16 (30)</td>
<td>t = 1.17, P = 0.12</td>
</tr>
<tr>
<td>Oct 08</td>
<td>19.31 ± 7.19 (5)</td>
<td>23.94 ± 6.27 (14)</td>
<td>t = 1.37, P = 0.09</td>
</tr>
<tr>
<td>Oct 15</td>
<td>27.02 ± 3.21 (2)</td>
<td>25.81 ± 4.72 (15)</td>
<td>t = 0.35, P = 0.73</td>
</tr>
</tbody>
</table>

* t-tests of whether females emerged from larger hosts than did males; two-tailed P values given when means were in direction opposite to that predicted.

MATERIALS AND METHODS

Weekly from 28 May to 12 November 1985, fly pupae were collected from an enclosed, shallow-pit, egg-layer poultry house in Delphi, Indiana (the Hilltop poultry house described in Merchant 1984, Merchant et al. 1987), using pupal traps (Hogsette and Butler 1981, Merchant et al. 1985). The pupae were held until flies emerged and died. Then pupae from which no flies emerged and which had not been depredated (Merchant 1984) were held individually in gelatin capsules for parasitoid emergence. Wasp species were identified following Boucek (1963) and Rueda and Axtell (1985) and house fly and stable fly pupae following Skidmore (1985). Voucher specimens have been deposited at the museum of the Department of Entomology at Purdue University, West Lafayette, Indiana.

Width of fly pupae was measured to the nearest 0.05 mm at a magnification of 360 X under a dissecting microscope. These widths were converted to volumes (mm³) using the following formulas: for house flies volume = (22.68)(width)³ - 37.63 and for stable flies volume = (11.31)(width)³ - 11.18. These regression equations had been determined by measuring both width and length on 17 stable flies and 36 house flies, then calculating volume with the equation for a prolate spheroid (Holdaway and Smith 1932), and finally regressing volume against width.

RESULTS

There were no significant differences among collection dates in S. endius sex ratio (G = 3.82, df = 6, P > 0.50). Pooling across all dates, S. endius sex ratio was 26.1% males (n = 330 male and female wasps).

There was no significant difference in the size of hosts from which male and female S. endius emerged. This was true regardless of whether one looked (1) within host species, (2) at all host species combined, or (3) between different host species. Looking at house fly hosts, among the five dates with sample sizes of greater than ten, a two-way analysis of variance on host size showed no significant effect of wasp sex (F = 0.34, P = 0.56) and no significant interaction between wasp sex and date (F = 2.26, P = 0.06). Because the interaction approached significance, I also did individual t-tests for each date, but these also revealed no significant difference in the size of hosts from which male and female wasps emerged (Table 1). Looking at stable fly hosts, combining all dates, there was no significant difference in host size between male and female wasps, though sample sizes are small (males: X ± SD 14.50 ± 0.73, n = 4; females 14.50 ± 0.97, n = 7; t = 0.07, P = 0.94). Combining host species, among the five dates with sample sizes of greater than ten, a two-way analysis of variance on host size showed no significant effect of wasp sex (F = 0.51, P = 0.48) and no significant interaction between sex and date (F = 1.57, P = 0.18). Looking between host species, though stable fly pupae are on average smaller than
house fly pupae (Skidmore 1985, King 1991), the sex ratio of \textit{S. endius} on stable flies (36% males, \(n = 11\) wasps) was not significantly greater than on house flies (25% males, \(n = 264\) wasps) (\(G = 0.67, P > 0.30\)).

**DISCUSSION**

The absence of host size differences between the sexes for \textit{S. endius} in this study is consistent with results of laboratory experiments by Donaldson and Walter (1984). Their experiments indicated that \textit{S. endius} females do not manipulate the sex of their offspring in response to the size of house fly hosts. The lack of any significant relationship between host size and parasitoid sex ratio for \textit{S. endius} contrasts both with the pattern observed in most other species of parasitoid wasps that have been examined and with the prediction of the host quality models. In most, though not all, species of parasitoid wasps that have been examined, females emerge from larger hosts than do males (about 44 of 65 parasitoid wasp species (reviewed in King 1987, 1989, in press).

Results with other species of \textit{Spalangia} besides \textit{S. endius} have been mixed. These other species of \textit{Spalangia} are also parasitoids of fly pupae. Legner (1969) found the predicted negative relationship between host size and offspring sex ratio for \textit{S. nigra}, but not for \textit{S. cameroni} or \textit{S. drosophilae} (statistical analyses of his results in Table 7 of King 1987). King's (1988) laboratory results support the prediction for \textit{S. cameroni} parasitizing house flies. However, field results with \textit{S. cameroni} are more complicated: the prediction is supported on a within host species basis for two of three dates—two dates on which \textit{S. cameroni} emerged only from house fly pupae—but not on the date when stable fly pupae were also parasitized by \textit{S. cameroni} (King 1991). Looking just at stable fly pupae, \textit{S. cameroni} males actually emerged from significantly larger hosts than did females, opposite the prediction. As was the case with \textit{S. endius}, \textit{S. cameroni} sex ratio did not differ between small and large host species (i.e., stable fly and house fly pupae) (King 1991).

Because the members of the genus \textit{Spalangia} exhibit such a variety of relationships between sex ratio and host size, it is a promising group for a comparative study. A comparison of behavioral, ecological, and life history factors among the species may help to explain the interspecific differences in sex ratio patterns. One possibility is that the species that do not support the host quality models' prediction may not meet the models' assumption of a more positive effect of host size on female than male reproduction. For example, competition among males for mates may increase the importance of large male body size in some species. In general, among parasitoid wasps, the importance of size on male reproductive success has been little studied (Charnov et al. 1981, Jones 1982, King 1988, van den Assem et al. 1989), especially relative to the attention given females (e.g., Charnov et al. 1981, Jones 1982, King 1988, van den Assem et al. 1989; 14 references in Table 6 of King 1987).

**ACKNOWLEDGMENTS**

I thank R. Flanders for introducing me to parasitoid wasps, the Brubakers of Hilltop Egg Farm for the use of their poultry house, M. Merchant for helpful tips on identification, R. Howard for valuable discussions in the planning, analyzing, and writing of this research, and R. King for comments on the manuscript. This research was aided by a National Science Foundation Graduate Fellowship and by Grants-in-Aid of Research from Sigma Xi, The Scientific Research Society.
LITERATURE CITED


ACROBASIS SHOOT MOTH (LEPIDOPTERA: PYRALIDAE) INFESTATION-TREE HEIGHT LINK IN A YOUNG BLACK WALNUT PLANTATION

George Rink¹, Barbara C. Weber², D. Michael Baines³, and David T. Funk⁴

ABSTRACT

Acrobasis shoot moth infestations were evaluated in a young black walnut progeny test for 4 years, from ages 3 to 6. Infestation levels were greatest on the largest trees in the fourth and fifth year after plantation establishment, and were declining by the sixth year. Acrobasis infestation appears to be a problem primarily on young trees less than 2.5 m in height. There was no evidence for genetic resistance to Acrobasis infestation in black walnut.

Black walnut (Juglans nigra) is commonly grown for production of high-quality veneer logs. Only trees with stems free of form defects, such as forks or crooks in the first 3 to 5 m, qualify as veneer logs. Stem defects in black walnut often result from loss of apical dominance due to damage or mortality of the terminal bud, often associated with late spring frosts. More recently, case bearer moths (Acrobasis spp.) have been shown to damage terminal shoots and buds of young black walnut trees when larvae tunnel into them. Our study was designed as a survey to assess damage to trees in a young plantation with a relatively serious Acrobasis infestation. Insect identification techniques followed those of McKeague and Simmons (1979).

METHODS

Open-pollinated seeds from 54 walnut trees were used to produce 1–0 seedlings for a progeny test. Parent trees were selected from within a 250-mile semi-circular radius south of the Shawnee National Forest in southern Illinois. This included an area bounded by southern Illinois, western Kentucky, western Tennessee, and southeast Kansas. However, most of the seed collections were from western Kentucky and western Tennessee. In most cases seed was collected from one tree per stand. Although trees of better than average form were sought at the time of collection, no rigid minimum selection criteria were applied. Seedling progeny resulting from these seed collections were planted in 5-tree row plots at a spacing of 1.8 m within rows and 3.7 m between rows, in a randomized complete block design with 10 blocks; upon establishment the plantation contained 2,700 trees.

The progeny test was established in early spring 1973 on a Haymond silt loam on the floodplain of Sexton Creek, on the Shawnee National Forest in Alexander

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The site was an abandoned pasture on which 2.2 m strips were machine sprayed with a simazine, dalapon, 2,4-D mix before planting and spot sprayed in spring 1974 and 1975. In addition the plantation was mowed annually.

In 1976, after three growing seasons, we observed that many of the walnut trees were growing slowly, some were crooked or forked, and a few had died back and sprouted from near the base. We suspected insect attack as a possible cause of the poor form and began surveys for the following insects: Acrobasis juglandis (LeBaron), the pecan leaf casebearer; A. demotella Grote, the walnut shoot moth (Martinat and Wallner 1980); and Xylosandrus americanus (Blandf.), an ambrosia beetle. The ambrosia beetle was later associated with canker and dieback but not with crook or forking problems related to terminal buds (Weber and McPherson 1984).

During 1977-1979, we made two types of surveys, using the same sampling scheme for both. The surveys were done on a pilot basis in 1976 and repeated 1977-1979. First, during the dormant season, we counted 1 mm-long hibernacula containing overwintering larvae of A. juglandis; the hibernaculum is the overwintering case for the first instar larva. First instar larvae develop from eggs hatched in late summer; they spin hibernacula in preparation for overwinter hibernation. Presence of hibernacula on a tree was considered an indicator of susceptibility of the tree to Acrobasis infestation. Second, in the early summer, we counted infested growing shoots as a measure of damage by A. demotella; A. juglandis is a defoliator but does not tunnel into shoots (Martinat and Wilson 1979). One tree per family-row plot per block was randomly chosen (using a random number generator) for infestation evaluation each fall; the same tree was evaluated the following spring. All insect and infested shoot count data were square root transformed to increase normality. Tree height was measured to the nearest cm during the dormant season.

Data were analyzed by analysis of variance and covariance techniques. Family heritabilities were calculated using the $H^2 = (1 - 1/F)$ formula of Kung and Bey (1979) in which $F$ is the variance ratio for families.

RESULTS AND DISCUSSION

During our 4-year evaluation of this plantation, height growth averaged 0.36 m annually. Mean total tree height was 1.5 m in 1976 and the plantation average was 2.9 m by the end of the 1980 growing season. Infestation as reflected by the number of hibernacula per tree increased from 1.4 in 1976 to a high of 2.6 in 1978 and declined to 2.0 hibernacula per tree in 1979. Demographic trends established by Acrobasis hibernacula were mirrored by the number of infested twigs per tree, which increased from 1.3 in 1976 to 2.3 in 1978 and then declined to 1.7 in 1979 (table 1). However, during the 1976-1979 period, the number of trees with hibernacula increased from 20 percent in 1976 to 33 percent in 1977, 44 percent in 1978, and 57 percent in 1979. Similarly, the number of trees with infested twigs increased from 20 percent in 1976 to 30 percent in 1977, 58 percent in 1978, and 63 percent in 1979.

Analyses of variance or covariance failed to disclose any significant family effects. Heritability of susceptibility to infestation by Acrobasis was less than 0.15, confirming that genetic resistance to Acrobasis damage is extremely low or nonexistent. Similarly there was no correlation between either number of hibernacula or number of infested twigs and latitude or longitude of family origin.

Correlation analysis disclosed a pattern of very high positive correlations between the number of hibernacula per tree and the number of infested twigs ($0.87 < r < 0.97$). These high correlations were expected and suggest that the parameters measure similar attributes of Acrobasis infestation and that future surveys probably need to measure only one of these parameters. The high correlations also suggest that overwintering survival rates were high and parasitism/predator levels were low.
Table 1. — Average height (m), height growth (m), number of hibernacula per tree (no.) and number of infested twigs

<table>
<thead>
<tr>
<th>Variable</th>
<th>Mean</th>
<th>Minimum</th>
<th>Maximum</th>
</tr>
</thead>
<tbody>
<tr>
<td>Height 76</td>
<td>1.5</td>
<td>0.2</td>
<td>2.9</td>
</tr>
<tr>
<td>Height 77</td>
<td>1.8</td>
<td>0.4</td>
<td>3.6</td>
</tr>
<tr>
<td>Height 78</td>
<td>2.1</td>
<td>0.4</td>
<td>4.4</td>
</tr>
<tr>
<td>Height 79</td>
<td>2.4</td>
<td>0.6</td>
<td>5.5</td>
</tr>
<tr>
<td>Height 80</td>
<td>2.9</td>
<td>0.8</td>
<td>6.1</td>
</tr>
<tr>
<td>Ht. Gr. 76-77</td>
<td>0.35</td>
<td>-0.63</td>
<td>1.39</td>
</tr>
<tr>
<td>Ht. Gr. 77-78</td>
<td>0.26</td>
<td>-1.15</td>
<td>1.67</td>
</tr>
<tr>
<td>Ht. Gr. 78-79</td>
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<td>-0.90</td>
<td>2.78</td>
</tr>
<tr>
<td>Ht. Gr. 79-80</td>
<td>0.46</td>
<td>-2.55</td>
<td>2.02</td>
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<tr>
<td>Hib. 76</td>
<td>1.4</td>
<td>1.0</td>
<td>7.0</td>
</tr>
<tr>
<td>Hib. 77</td>
<td>2.0</td>
<td>1.0</td>
<td>14.0</td>
</tr>
<tr>
<td>Hib. 78</td>
<td>2.6</td>
<td>1.0</td>
<td>16.0</td>
</tr>
<tr>
<td>Hib. 79</td>
<td>2.0</td>
<td>1.0</td>
<td>3.0</td>
</tr>
<tr>
<td>Infest. 76</td>
<td>1.3</td>
<td>1.0</td>
<td>15.0</td>
</tr>
<tr>
<td>Infest. 77</td>
<td>1.7</td>
<td>1.0</td>
<td>3.0</td>
</tr>
<tr>
<td>Infest. 78</td>
<td>2.3</td>
<td>1.0</td>
<td>14.0</td>
</tr>
<tr>
<td>Infest. 79</td>
<td>1.7</td>
<td>1.0</td>
<td>9.0</td>
</tr>
</tbody>
</table>

Table 2. — Correlations of number of hibernacula and infested shoots with total tree height (m) in current and subsequent years (ns = not statistically significant; ** = significant at P < 0.05)

<table>
<thead>
<tr>
<th>Measurement year</th>
<th>Tree height in current year</th>
<th>Tree height in subsequent year</th>
<th>Tree height in current year</th>
<th>Tree height in subsequent year</th>
</tr>
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<tbody>
<tr>
<td></td>
<td>Number of hibernacula</td>
<td>Number of infested shoots</td>
<td></td>
<td></td>
</tr>
<tr>
<td>1976</td>
<td>0.15 ns</td>
<td>0.18 ns</td>
<td>0.04 ns</td>
<td>0.07 ns</td>
</tr>
<tr>
<td>1977</td>
<td>0.22 **</td>
<td>0.23 **</td>
<td>0.30 **</td>
<td>0.34 **</td>
</tr>
<tr>
<td>1978</td>
<td>0.40 **</td>
<td>0.41 **</td>
<td>0.43 **</td>
<td>0.45 **</td>
</tr>
<tr>
<td>1979</td>
<td>0.08 ns</td>
<td>0.07 ns</td>
<td>0.04 ns</td>
<td>0.05 ns</td>
</tr>
</tbody>
</table>

A more interesting pattern of correlations emerged between number of hibernacula, infestation, and total tree height (table 2) and height growth (table 3). In 1976, the first year of Acrobasis evaluation, there was no statistical correlation (r < 0.2) between either measure of insect attack and growth variables. In 1977, there were low but significant correlations (r > 0.2) of hibernacula and infestation with total tree height. By 1978, both measures of insect attack were significantly correlated with total height and height growth. Furthermore, the magnitude of the correlation between total height and number of hibernacula doubled. However, by 1979 neither measure of Acrobasis infestation was correlated with total height or with height growth.

Such correlation patterns could result from the following circumstances: in 1976, when there was no correlation between infestation and tree size or growth, the
planted plantation was only 3 years old and all trees were in approximately the same size category. The initial infestation was small and the outbreak was distributed among trees of the same size class. By 1977 the outbreak was spreading to the larger trees as evidenced by the positive correlation with tree height; by 1978, positive correlations between infestation and both total height and height growth were present. By 1979, on the basis of average number of hibernacula per tree or mean number of infested twigs per tree, the infestation seemed to be declining. Furthermore, the remaining Acrobasis no longer preferred the larger trees but infested any size trees.

All statistically significant correlations between total height and insect attack were positive, suggesting that Acrobasis preferentially attacks larger trees once an infestation is established in a plantation (table 2). Furthermore, it appears that in 1977 and 1978 Acrobasis also attacked the faster growing trees (table 3). The implication is that, initially, Acrobasis infests the largest trees, those with the largest crowns; however, in subsequent years when most of the trees in the plantation are in larger size classes, insect attacks are of a more random nature.

The lack of negative correlations between infestation in a given year and growth in subsequent years was surprising. Acrobasis damage has been described as resembling frost damage where infested terminals die back. In a large infestation, negative correlations with growth in the following year might be expected. However, in infestations at this population level, negative correlations were not encountered. Instead, positive correlations imply that somehow infestations had little effect on growth and perhaps were even slightly stimulatory. Thus, it is likely that the overall infestation levels were not high enough to affect average growth rates.

By 1979, when the trees averaged 2.4 m in total height (table 1), infestation appeared to be subsiding. Subsequent surveys did not disclose significant infestations. Thus, we hypothesize that Acrobasis is a problem on young trees less than 2.5 m in height; stem form may be affected but growth rate is not. Alternatively, a buildup of populations of natural predators of Acrobasis may have resulted in a declining Acrobasis population; unfortunately, no effort was made to monitor these predators.

**Table 3. — Correlations of number of hibernacula and infested shoots with tree growth (m) in current and subsequent years (ns = not statistically significant; ** = significant at P < 0.05)**

<table>
<thead>
<tr>
<th>Measurement year</th>
<th>Growth in current year</th>
<th>Growth in subsequent year</th>
<th>Growth in current year</th>
<th>Growth in subsequent year</th>
</tr>
</thead>
<tbody>
<tr>
<td>Number of hibernacula</td>
<td></td>
<td></td>
<td>Number of infested shoots</td>
<td></td>
</tr>
<tr>
<td>1976</td>
<td>—</td>
<td>0.14 ns</td>
<td>—</td>
<td>0.08 ns</td>
</tr>
<tr>
<td>1977</td>
<td>0.10 ns</td>
<td>0.13 ns</td>
<td>0.18 **</td>
<td>0.23 **</td>
</tr>
<tr>
<td>1978</td>
<td>0.18 **</td>
<td>0.23 **</td>
<td>0.19 **</td>
<td>0.25 **</td>
</tr>
<tr>
<td>1979</td>
<td>0.08 ns</td>
<td>0.20 **</td>
<td>0.05 ns</td>
<td>0.04 ns</td>
</tr>
</tbody>
</table>

**ACKNOWLEDGMENTS**

We thank David J. Polak, for assistance with statistical analyses. At the time of the study Dave Polak was a computer programmer with NCFES, Carbondale, IL. We also acknowledge the cooperation of the Shawnee National Forest on whose land the plantation was established.


HOST PLANT SUITABILITY AND A TEST OF THE FEEDING SPECIALIZATION HYPOTHESIS USING *PAPILIO CRESPHONTES* (LEPIDOPTERA: PAPILIONIDAE)

J. Mark Scriber1 and Robert V. Dowell2

**ABSTRACT**

The concept that host plant favorites would be used for more rapid and/or efficient growth of the "locally adapted" individuals was tested in a preliminary way using the giant swallowtail butterfly, *Papilio cresphontes*. Populations feeding only on northern prickly ash, *Zanthoxylum americanum* (from Wisconsin), primarily (or exclusively) on hoptree, *Ptelea trifoliata* (in Ohio) and on lime prickly ash, *Z. fagara*, or *Citrus*, (in Florida) were compared on alternate hosts and on their actual local hosts under controlled environmental conditions. While the results with final instar larvae generally support the feeding specialization hypothesis with regard to more rapid and/or more efficient growth on local food plant favorites, we are hesitant to extrapolate these results to the species as a whole for several reasons discussed herein.

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2Current address: 1681 Pebblewood Dr., Sacramento, CA 95833.
digestive efficiencies. Subsequent reduction of such efficiencies with respect to ancestral or locally non-used (allopatric) foodplants might lead to what could be called "obligate monophagy" (Smiley 1978). The evidence available for evaluation of this concept is meager (see Fox and Morrow 1981, Scriber 1983, 1986).

The objective of this study was to assess the possibility that *P. cresphontes* populations are differentially adapted to their local foodplant favorites compared to other plants. Our approach was to collect *P. cresphontes* from various geographic locations and carefully bioassay the larval growth performances on their local favorites as well as the allopatric foodplants which are local favorites used by other *P. cresphontes* populations.

**METHODS**

**Sources of Larvae**

Larvae from eggs obtained from *Citrus* spp. in Broward County, Florida were distributed at eclosion (unfed neonates) to the following foodplants: Lime prickly ash, *Zanthoxylum fagara*; Northern prickly ash, *Zanthoxylum americanum*; sweet orange *Citrus sinensis* Osbeck; and hoptree, *Ptelea trifoliata*. In Wisconsin, larvae were obtained from eggs of a field-captured female in western Dane County and a field-captured female in eastern Iowa County and subsequently were randomly distributed to various rutaceous foodplants, including northern prickly ash and hoptree. In these two populations (and nearly all of Wisconsin) prickly ash is the only host plant available. For this study additional Wisconsin *P. cresphontes* were obtained from Richland County where prickly ash, *Z. americanum*, shrubs are the only hosts available. Ohio *P. cresphontes* were obtained from Preble County where the local favorite and dominant host plant is hoptree, *Ptelea trifoliata*.

**Feeding Experiments**

Freshly molted penultimate instar larvae were individually weighed and distributed to 150 x 25 mm petri dishes and maintained on a specific host under standardized rearing conditions (16:8 photo, scoto-phase with a corresponding 23:19-C° thermoperiod, with moistened filter paper in each dish). Plant leaves collected from several trees near the U.W. Arboretum or from our greenhouse were weighed fresh, placed in water-filled floral aquapics® to maintain leaf turgor and subsequently introduced into the petri dishes for standard gravimetric determination of food consumption (Waldbauer 1968). Nutritional indices were calculated based upon the dry weight (biomass) of leaves, feces, and larvae. The mean larval weight during the stadium was estimated by the (initial plus final weight)/2. Indices of larvae performance are reported as in Scriber and Slansky (1981):

\[
\begin{align*}
\text{RGR}, \text{ relative growth rate (mg biomass gained per day per mg larval biomass)} & \\
(RGR = RCR \times AD \times ECD) \\
\text{RCR}, \text{ relative consumption rate (mg biomass ingested per day per mg larval biomass)} & \\
\text{AD, approximate digestibility (also called assimilation efficiency)} & = \frac{\text{Food infested (mg dry wt) - Feces (mg dry wt)}}{\text{Food ingested (mg dry wt)}} \times 100\% \\
\text{ECD, efficiency of conversion of digested food (also called net growth efficiency)} & = \frac{\text{Biomass gained (mg dry wt)}}{\text{Food ingested (mg dry wt) - Feces (mg dry wt)}} \times 100\%
\end{align*}
\]
ECI, efficiency of conversion of ingested food (also called gross growth efficiency) =

\[
\frac{\text{Biomass gained (mg dry wt)}}{\text{Food ingested (mg dry wt)}} \times 100\%
\]

\[\text{ECI} = \text{AD} \times \text{ECD} = \text{(overall efficiency)}\]

Plants, larvae, and feces were frozen and freeze-dried for dry weight determinations. Statistical analyses were performed and where the ANOVA indicated significant differences between the means were analyzed by Tukey's test for unequal sample sizes (Winer 1962, Snedecor and Cochran 1967). The very conservative Tukey's test was used because our sample sizes were not as large as we had wished. We therefore believe that all statistically significant differences represent very real biological differences.

RESULTS

Growth of the Florida *Papilio cresphontes*, which uses only Citrus and lime prickly ash as natural host plants, was significantly faster on lime prickly ash, *Zanthoxylum fagara*, than was growth of Ohio *P. cresphontes* on this Florida plant species for both the penultimate and the final instars (Table 1). The Florida population also had a higher efficiency of processing ingested biomass (ECI) than the Ohio larvae. This central American plant species (*Z. fagara*) occurs only in the southern half of Florida and southern tip of Texas (Fig. 1). As such this Ohio butterfly population gets no closer than 1000 kilometers to the plant.

The congeneric northern prickly ash, *Z. americanum*, occurs sporadically from Georgia and Alabama northward to the Great Lakes States and Canada. While all Wisconsin populations and the Ohio populations are sympatric with northern prickly ash and despite the fact that northern prickly ash is the only host plant for *P. cresphontes* in Wisconsin, larval growth and efficiency of these larvae were no better than for Florida larvae (which are totally allopatric with the plant range; Fig. 2 and Table 2). No consistent differences are observed between the growth performances of the penultimate and final instars of Wisconsin, Ohio, and Florida giant swallowtail larvae. While Florida larvae exhibit the highest overall efficiency of processing *Z. americanum* (ECI) in the penultimate instar, they exhibit the lowest efficiency in the final instar (Table 2). The reverse is true for the consumption rates (RCR).

Ohio populations of *P. cresphontes* primarily use hoptree *Ptelea trifoliata* in the source area (Preble Co., OH) for our experimental larvae (Fig. 3). Growth rates of these Ohio larvae were nearly three times as fast (.215 mg mg⁻¹ d⁻¹) as Wisconsin larvae in the final instar (Table 3). The Ohio efficiencies (ECI and ECD) averaged twice those of Wisconsin larvae in the final instar (eg., ECI = 18.8% versus 8.1% and 8.7% respectively). These patterns were not observed in the penultimate instar, however the digestibility (AD) and consumption rates (RCR) were higher for Ohio than Wisconsin larvae (Table 3). Not enough Florida larvae were available for bioassays on hoptree.

DISCUSSION

Larvae of *P. cresphontes* from Florida, Ohio, and Wisconsin exhibit significant differences in growth rates, consumption rates, and efficiencies of processing three different plant species. For final instars, where the largest amounts of food are consumed, populations in Florida grow faster and more efficiently on the local foodplant, lime prickly ash, than Ohio larvae (Fig. 1). Similarly, populations in
Table 1. — Growth performance of penultimate and final instar *Papilio cresphontes* fed *Zanthoxylum jagara* (lime prickly ash). Data are presented as a mean ± SE.

<table>
<thead>
<tr>
<th>Instar and Source population</th>
<th>(n)</th>
<th>Instar duration (days)</th>
<th>Growth Rate (RGR)</th>
<th>Consumption Rate (RCR)</th>
<th>Efficiencies (A.D.)</th>
<th>Efficiencies (E.C.D.)</th>
<th>Efficiencies (E.C.I.)</th>
<th>Leaf quality (% water)</th>
</tr>
</thead>
<tbody>
<tr>
<td><strong>Penultimate Instar</strong></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Ohio (Preble Co.)</td>
<td>(5)</td>
<td>6.6 ± 0.2</td>
<td>.130 ± .010</td>
<td>1.74 ± .13</td>
<td>24.3 ± 4.4</td>
<td>33.9 ± 4.5</td>
<td>7.5 ± 0.5</td>
<td>69.2 ± 1.4</td>
</tr>
<tr>
<td>Florida (Broward Co.)</td>
<td>(4)</td>
<td>6.1 ± 0.1</td>
<td>.224 ± .010</td>
<td>2.16 ± .17</td>
<td>43.0 ± 4.5</td>
<td>26.0 ± 4.9</td>
<td>10.5 ± 0.9</td>
<td>70.9 ± 2.3</td>
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</tr>
<tr>
<td><strong>Final Instar</strong></td>
<td></td>
<td></td>
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<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Ohio (Preble Co.)</td>
<td>(3)</td>
<td>16.2 ± 1.4</td>
<td>.032 ± .003</td>
<td>1.89 ± .12</td>
<td>25.8 ± 2.9</td>
<td>6.7 ± 0.6</td>
<td>1.7 ± 0.1</td>
<td>66.7 ± 1.7</td>
</tr>
<tr>
<td>Florida (Broward Co.)</td>
<td>(3)</td>
<td>11.8 ± 1.1</td>
<td>.084 ± .006</td>
<td>1.44 ± .41</td>
<td>27.2 ± 3.1</td>
<td>30.4 ± 14.3</td>
<td>11.8 ± 1.1</td>
<td>72.6 ± 1.6</td>
</tr>
</tbody>
</table>

*Significant differences are indicated (* = P ± 0.05, ** = P ± 0.01) ANOVA (Snedecor and Cochran 1967).

Table 2. — Growth performance of penultimate and final instar *Papilio cresphontes* fed *Zanthoxylum americanum* (northern prickly ash).

<table>
<thead>
<tr>
<th>Instar and Source population</th>
<th>(n)</th>
<th>Instar duration (days)</th>
<th>Growth Rate (RGR)</th>
<th>Consumption Rate (RCR)</th>
<th>Efficiencies (A.D.)</th>
<th>Efficiencies (E.C.D.)</th>
<th>Efficiencies (E.C.I.)</th>
<th>Leaf quality (% water)</th>
</tr>
</thead>
<tbody>
<tr>
<td><strong>Penultimate Instar</strong></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>WI (Dane Co.)</td>
<td>(8)</td>
<td>4.7 ± 0.2 bc</td>
<td>.277 ± .011 bc</td>
<td>1.49 ± .06 a</td>
<td>36.0 ± 2.2</td>
<td>53.6 ± 4.2</td>
<td>18.7 ± 0.6 c</td>
<td>71.1 ± 0.9</td>
</tr>
<tr>
<td>Iowa Co.</td>
<td>(8)</td>
<td>4.1 ± 0.2 aab</td>
<td>.321 ± .012 a</td>
<td>1.35 ± .07 ab</td>
<td>38.8 ± 2.1</td>
<td>63.0 ± 3.8</td>
<td>24.0 ± 3.8 ab</td>
<td>70.8 ± 0.7</td>
</tr>
<tr>
<td>Richland Co.</td>
<td>(5)</td>
<td>3.9 ± 0.5 a</td>
<td>.307 ± .016 ab</td>
<td>1.34 ± .15 ab</td>
<td>46.2 ± 3.3</td>
<td>56.2 ± 5.3</td>
<td>23.9 ± 2.5 ab</td>
<td>72.5 ± 0.9</td>
</tr>
<tr>
<td>OH (Preble Co.)</td>
<td>(6)</td>
<td>5.0 ± 0.2 c</td>
<td>.248 ± .017 c</td>
<td>1.30 ± .11 ab</td>
<td>42.3 ± 5.4</td>
<td>51.8 ± 9.9</td>
<td>19.3 ± 0.9 c</td>
<td>74.0 ± 0.5</td>
</tr>
<tr>
<td>FL (Broward Co.)</td>
<td>(9)</td>
<td>4.3 ± 0.2 abc</td>
<td>.293 ± .005 abc</td>
<td>1.14 ± .05 b</td>
<td>39.7 ± 1.3</td>
<td>66.3 ± 3.6</td>
<td>26.0 ± 0.8 a</td>
<td>77.8 ± 0.1</td>
</tr>
<tr>
<td>L.S.D.</td>
<td>(0.7)</td>
<td>(.049)</td>
<td>(.34)</td>
<td>(n.s.)</td>
<td>(n.s.)</td>
<td>(n.s.)</td>
<td>(n.s.)</td>
<td>(4.4)</td>
</tr>
</tbody>
</table>

**Final Instar**

| WI (Dane Co.)               | (3)  | 8.5 ± 1.0 ab           | .128 ± .042       | 1.06 ± .28            | 34.1 ± 5.5 ab       | 34.2 ± 2.4 b        | 11.4 ± 1.2 ab         | 66.8 ± 1.4           |
| Iowa Co.                    | (3)  | 8.1 ± 0.5 ab           | .135 ± .012       | 1.04 ± .08            | 29.4 ± 3.3 b        | 45.4 ± 5.1 a        | 13.0 ± 0.4 a          | 69.9 ± 1.2           |
| Richland Co.                | (3)  | 8.5 ± 0.5 ab           | .107 ± .020       | 1.18 ± .17            | 45.4 ± 2.6 a        | 20.0 ± 0.8 c        | 9.0 ± 0.4 bc          | 69.2 ± 1.4           |
| OH (Preble Co.)             | (4)  | 7.0 ± 0.0 b            | .160 ± .005       | 1.27 ± .05            | 41.7 ± 0.9 ab       | 30.4 ± 1.7 b        | 12.6 ± 0.4 a          | 73.2 ± 0.9           |
| FL (Broward Co.)            | (6)  | 10.5 ± 0.6 a           | .100 ± .009       | 1.67 ± .26            | 41.4 ± 2.7 ab       | 15.2 ± 1.2 c        | 6.3 ± 0.7 e           | 74.4 ± 4.1           |
| L.S.D.                      | (2.8) | (n.s.)                 | (n.s.)            | (14.3)               | (10.4)             | (3.4)              |                      |                      |

*Significant differences (P = 0.05) between the means are indicated by letters (Tukey's test for unequal sample sizes; Winer 1962, Snedecor and Cochran 1967).
Figure 1. Performance of final instar *P. cresphontes* larvae (data are presented as a mean ± se) as a function of geographic source and range (shaded area) of the test plant, lime prickly ash (*Zanthoxylum fagara*).

Ohio grow faster and more efficiently on their local favorite, hoptree compared to Wisconsin populations that prefer northern prickly ash (Fig. 3). Larvae of both Ohio and Wisconsin populations grow faster and more efficiently than Florida larvae on the northern prickly ash *Z. americanum* (Fig. 2).

While these results with final instars lend support to the feeding specialization hypothesis and suggest that biochemical adaptation has occurred with local specialization, the same patterns are not observed in all cases with larvae in their penultimate instar. While Florida larvae grow significantly faster and more efficiently on *Z. fagara* in the penultimate instar (Table 1), Ohio larvae in their penultimate instar grow no faster on their favored *Ptelea trifoliata* (Table 2), and Wisconsin popula-
tion do not grow significantly faster nor more efficiently than Florida larvae on their favorite host Z. americanum (Table 3).

Variable leaf nutritional quality can be very important in determining the larval growth rates and efficiencies (Scriber and Slansky 1981). Different growth performance on different plant species is not unexpected, but there are also significant differences in nutritional quality within a plant species. For example, some leaf quality variability (as indexed by leaf water content) may have been involved in the differential growth performances to some extent in these studies, especially with northern prickly ash where 8-10% differences in leaf water content were detected. These differences may have been largely responsible for the good performance of the Florida populations on northern prickly ash (Table 2).

Another important aspect of local adaptation (not addressed in this study) is the critical first instar survival differences among geographic populations or host races (Diehl and Bush 1984, Futuyma and Moreno 1988). Whether these differences in survival may be related to behavioral (antixenosis) or toxicological (antibiosis) mechanisms, they are of fundamental importance to the ecological success of the

Figure 2. Performance of final instar P. cresphontes on northern prickly ash, Z. americanum as a function of insect population and plant range (shaded).
population. The differences in growth performance detected at the penultimate and final instars for surviving larvae may be of minimal significance compared to the differential survival of the initial cohort of larvae in their neonate stages. However, these later instars are around longer and other defenses appear to be useful (Fig. 4a, 4b).

For example, cryptic resting larvae on the side of the stem (Fig. 4a) could avoid early bird detection and predation (Hirose et al. 1980), whereas osmeterial glands (Fig. 4b) may be effective at deterring foraging ants or wasps as has been shown on related *Papilio* in North America and Japan (Hirose and Tagaki 1980, Damman 1986). In addition, the resting spot for the larger larvae is on the inside of the thorny

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**Figure 3.** Performance of final instar *P. cresphontes* larvae on niptree (*Ptelea trifoliata*) as a function of the plant range (shaded).
Table 3. — Growth performance of penultimate and final instar *Papilio cresphontes* fed *Ptelea trifoliata* (hoptree).

<table>
<thead>
<tr>
<th>Instar and Source population</th>
<th>(n)</th>
<th>Instar duration (days)</th>
<th>Growth Rate (RGR)</th>
<th>Consumption Rate (RGR)</th>
<th>Efficiencies (A.D.)</th>
<th>Efficiencies (E.C.D.)</th>
<th>Efficiencies (E.C.I.)</th>
<th>Leaf (% water)</th>
</tr>
</thead>
<tbody>
<tr>
<td><strong>Penultimate Instar</strong></td>
<td></td>
<td></td>
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<td></td>
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<td></td>
<td></td>
</tr>
<tr>
<td>WI (Dane Co.)</td>
<td>(9)</td>
<td>4.6±0.2 b</td>
<td>.282±.020</td>
<td>1.29±.04 ab</td>
<td>42.8±1.0 b</td>
<td>51.7±4.3 a</td>
<td>21.9±1.6</td>
<td>71.6±0.4</td>
</tr>
<tr>
<td>(Iowa Co.)</td>
<td>(8)</td>
<td>4.3±0.1 ab</td>
<td>.303±.009</td>
<td>1.21±.04 b</td>
<td>47.4±2.0 b</td>
<td>54.0±3.2 a</td>
<td>25.3±1.0</td>
<td>71.6±0.4</td>
</tr>
<tr>
<td>OH (Preble Co.)</td>
<td>(3)</td>
<td>3.8±0.2 a</td>
<td>.312±.030</td>
<td>1.46±.05 a</td>
<td>65.6±3.9 a</td>
<td>32.8±2.7 b</td>
<td>21.3±1.3</td>
<td>72.9±0.4</td>
</tr>
<tr>
<td>L.S.D.1</td>
<td></td>
<td>(0.7)</td>
<td>(n.s.)</td>
<td>(.180)</td>
<td>(7.6)</td>
<td>(16.8)</td>
<td>(n.s.)</td>
<td></td>
</tr>
<tr>
<td><strong>Final Instar</strong></td>
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<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>WI (Dane Co.)</td>
<td>(3)</td>
<td>12.3±1.0 b</td>
<td>.057±.007 b</td>
<td>0.71±.11 b</td>
<td>55.2±4.0</td>
<td>14.9±1.3 b</td>
<td>8.1±0.3 b</td>
<td>69.3±1.2</td>
</tr>
<tr>
<td>(Iowa Co.)</td>
<td>(5)</td>
<td>9.9±1.6 ab</td>
<td>.075±.012 b</td>
<td>0.87±.08 ab</td>
<td>52.8±3.1</td>
<td>17.1±3.0 b</td>
<td>8.7±1.1 b</td>
<td>68.3±1.1</td>
</tr>
<tr>
<td>OH (Preble Co.)</td>
<td>(4)</td>
<td>6.3±0.1 a</td>
<td>.215±.005 a</td>
<td>1.14±.01 a</td>
<td>49.6±3.1</td>
<td>38.4±2.9 a</td>
<td>18.8±0.3 a</td>
<td>72.3±0.5</td>
</tr>
<tr>
<td>L.S.D.1</td>
<td></td>
<td>(5.20)</td>
<td>(.040)</td>
<td>(.31)</td>
<td>(n.s.)</td>
<td>(11.5)</td>
<td>(3.3)</td>
<td></td>
</tr>
</tbody>
</table>

1Significant differences between the means (P = 0.05) are indicated by different letters (Tukey's test for unequal sample sizes: Snedecor and Cochran 1967, Winer 1962). (n.s. = nonsignificant differences via F-test, ANOVA)
Figure 4a-b. a-Final instar *P. cresphontes* resting cryptically on a stem of northern prickly ash, *Zanthoxylum americanum*. b-Final instar "snake-like" osmeterial defensive response of *P. cresphontes* larva to slight agitation. This has been shown for other *Papilio* to be a defensive behavior to ants, wasps, birds or other predators (see text).

thick stems (Figs. 4a, 4b) down and away from the danger of browsing cattle (and possibly deer) which were believed responsible for the pruned outer foliage on many of these hoptree bushes and lower branches in Wisconsin. The use of taller trees of prickly ash would avoid the mammalian grazing damage (i.e., larvae consumed with leaves). However, it has been shown that taller trees of *Zanthoxylum* have nutritionally poorer leaves upon which the *Papilio* growth is slower and mortality is higher (Watanabe 1982). Female *P. xuthus* L. butterflies in Japan prefer these shorter *Zanthoxylum* trees for oviposition (Watanabe 1982), as the *P. cresphontes* in Wisconsin also seem to do. Eggs and larvae of *P. xuthus* on *Zanthoxylum* in Japan in early stages were attacked primarily by smaller predators such as ants, spiders, bugs, and orthopteroids in contrast to birds and *Polistes* wasps that attack larger larvae (Watanabe 1981).

Additional studies would be needed to determine whether the differences in larval growth performance among the different *P. cresphontes* populations have a genetic basis. The phytochemical cues for oviposition and the allelochemical factors making the Rutaceae the primary host family of this insect are certainly of tremendous ecological (Dethier 1941, 1954) and evolutionary significance for the Papilionidae (Hancock 1983, Miller 1987). The specific chemical cues used by *P. cresphontes* for oviposition and enhancement of larval feeding may not be unlike those for Japanese Rutaceae feeders (Nishida 1977, Ichinóshé and Honda 1978). However, chemical similarities in Rutaceae hosts with the Umbelliferae, Lauraceae, Aristolochiaceae, Magnoliaceae, and Asteraceae (see Feeny et al. 1983) make the role of particular allelochemics (and their interactions) in host selection and the ecology of different *Papilio* uncertain at best.
This study reflects significant biological differences in larval performance among different individual larvae on the same host plants under controlled conditions. However, we still lack the extensive geographic replication and intensive (intrapopulation) analysis to detect the extent of genetically based adaptations. We are therefore hesitant to extrapolate the results from any particular host plant or any particular population to the *P. cresphontes* species on the whole. Similarly, we have illustrated that the relative differences in the roles of growth are in some cases instar-dependent. A more rigorous test of the feeding specialization hypothesis must include not only controls for host plant nutritional quality, but should also include a representative sample of genotypes from the entire range of the species of interest. Our studies with the sympatric *Papilio glaucus* group are more extensive and intensive to date, and we have detected several different levels of behavioral biochemical and genetic differences in the adaptation of local populations to favored host plants (Scriber 1986, Scriber et al. 1989, 1990). Yet no single species can be used when evaluating a general ecological concept such as the feeding specialization hypothesis. A comparative approach using different taxa is essential, and thus additional data from other insect/plant systems such as this study are still needed.

ACKNOWLEDGMENTS

This research was supported in part by the Michigan State University Experiment Station (Projects MAES #1644 and 8072) and the College of Agricultural and Life Sciences of the University of Wisconsin at Madison (Hatch #5134) and by a grant from NSF (DEB 7921749). We thank Mark Evans and Nancy Teitelbaum for their help in these studies.

LITERATURE CITED


GEOGRAPHIC DISTRIBUTION OF SIPHONAPTERA COLLECTED FROM SMALL MAMMALS ON LAKE MICHIGAN ISLANDS

William C. Scharf

ABSTRACT

The distribution of ten flea species collected from five small mammal host species on 13 Lake Michigan islands is described. Four new eastern and southern records for *Hystrichopsylla dippiei* Rothschild are given. Speculative suggestions are made regarding dispersal routes of some of the small mammal host species, and the distribution of flea species from *Peromyscus maniculatus gracilis* LeConte is discussed in the context of island biogeography theory.

I collected fleas from small mammals on Lake Michigan Islands from 1965 to present. Two other previous studies from Lake Michigan islands (Hatt et al. 1948, and Arnsman unpublished) each reported on flea species from one host species from one island, and agree with my findings. I have verified the Arnsman specimens in the Michigan State University Entomology Department collection. The Hatt et al. specimens, residing in the University of Michigan Museum of Zoology collection were identified by the eminent siphonapterist, Karl Jordan. A small portion of the species listed here are included in Scharf and Stewart (1980), but most are identified there by county and host only.

This listing is intended to shed light on possible distribution, and immigration routes, to the islands by both the small mammals and their fleas beginning from the melting of the Pleistocene ice sheets. I have previously speculated that the possible mechanisms of small mammal distribution were by way of: (1) a land bridge during the Lake Chippewa low water stage; (2) rafting on flotsam; (3) concealment in Indian canoes or early settler's vessels (Scharf 1973, and Scharf and Jorae 1980). While offering little conclusive evidence, the present study offers a few hints of the dispersal pattern of both the hosts and fleas. This distributional evidence is then examined for hypothetical extinctions of fleas due to island size and distance from the mainland in the context of the island biogeography theory of MacArthur and Wilson (1967).

MATERIALS AND METHODS

I collected 590 fleas of 10 species from 5 species of small mammal hosts on 13 islands in northeastern Lake Michigan (Figure 1). Capture was primarily by snap-trapping, but a live-trapping grid on High Island in June of 1986 and 1988 accounted for many fleas from that island. Firearms were used to collect squirrels

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during the hunting season. In most cases the mammals were brushed over a white surface to find the fleas, which were preserved in 70% ethyl alcohol, and mounted on microscope slides in Canada balsam for identification. The flea specimens remain in the private collection of the author. Skins and skulls of the hosts are in the Northwestern Michigan College collection of vertebrates.

The major problem regarding collecting methods remains whether completeness of seasonal, and habitat coverage is achieved, because some flea species show distinct seasonal and habitat abundance. I sampled all of the islands with a trapping pressure of at least 100 trap-nights. Some islands, such as the North and South Manitou
Table 1.—Flea species collected from *Peromyscus maniculatus gracilis* on Lake Michigan Islands. Islands arranged from south to north, east to west and with numbers corresponding to Figure 1.

<table>
<thead>
<tr>
<th>FLEA SPECIES</th>
<th>ISLAND*</th>
</tr>
</thead>
<tbody>
<tr>
<td>Ctenophthalmus pseudagyrti Baker</td>
<td>X X X X X X X X X</td>
</tr>
<tr>
<td>Epetedia w. wenmanni (Rothschild)</td>
<td>X X X X X X X X X</td>
</tr>
<tr>
<td>Hyslrichopsylla dippiei Rothschild</td>
<td>X X X X X</td>
</tr>
<tr>
<td>Orchopeas leucopus (Baker)</td>
<td>X X X X X X X X X</td>
</tr>
<tr>
<td>Peromyscopsylla h. hesperomyx (Baker)</td>
<td>X X X X X</td>
</tr>
</tbody>
</table>

*1—Marion Island; 2—South Manitou Island; 3—North Manitou Island; 4—South Fox Island; 5—North Fox Island; 6—Beaver Island; 7—Hog Island; 8—Garden Island; 9—High Island; 10—Gull Island; 11—Trout Island; 12—Whiskey Island; 13—Squaw Island.

Results and Discussion

The one mammal host species that is common to all of the islands visited is the Woodland Deer Mouse, *Peromyscus maniculatus gracilis* LeConte, although phenetic divergence has been shown between islands (Lederle et. al. 1985). Table 1 shows the distribution of 5 flea species on this host by island and Figure 1 shows the location of the islands keyed to the numbers in Table 1. Noteworthy for its ubiquitous distribution on this mouse is the flea *Orchopeas leucopus* (Baker) which is found on every island.

The occurrence of *Histrichopsylla dippiei* Rothschild from the deer mouse on four islands (Table 1) is an important new range extension. This paper represents the second Michigan record, and a new eastern and southern distribution for the species. While this flea has been noted on deer mice previously, the true host for this species is considered by many to be the Red Squirrel, *Tamiasciurus hudsonicus* (Erxleben), and the closest recorded occurrences of the flea are in Itasca and St. Louis Counties in northeastern Minnesota both on *T. hudsonicus* and *P. maniculatus* (Timm 1975, and Benton 1980), and on *P. maniculatus* on Isle Royale (Nixon and Johnson 1971). Its presence on the four Beaver islands closest to the Upper Peninsula, and its distribution across Canada and the far northern U. S. provides a clue to the route the host took to these islands. The absence of this flea from the southern islands of the Lake Michigan archipelago indicates that small mammal invasion after glacial retreat may have been by more than one route.

The other flea species collected from *P. m. gracilis* are commonly found on this mouse, and widely distributed (Table 1). However, two fleas common on this host in the U. P., *Peromyscopsylla catatina* (Jordan) and *Megabothris quirini* (Rothschild)
Table 2. — Flea species collected from *Tamias striatus* (L.) on Lake Michigan Islands. Islands arranged from south to north, east to west and with numbers corresponding to Figure 1.

<table>
<thead>
<tr>
<th>FLEA SPECIES</th>
<th>2</th>
<th>3</th>
<th>4</th>
<th>6</th>
<th>8</th>
</tr>
</thead>
<tbody>
<tr>
<td><em>Ctenophthalmus pseudagyrtes</em> Baker</td>
<td>X</td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td><em>Megabothris acerbus</em> (Jordan)</td>
<td></td>
<td>X</td>
<td>X</td>
<td>X</td>
<td>X</td>
</tr>
<tr>
<td><em>Tamiophila grandis</em> (Rothschild)</td>
<td>X</td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
</tbody>
</table>

*2—South Manitou Island; 3—North Manitou Island; 4—South Fox Island; 6—Beaver Island; 8—Garden Island;*

are notably absent from the Lake Michigan island mice (Lawrence et al. 1965, Scharf and Stewart 1980, Scharf et al. 1991).

The distribution of fleas from *P. m. gracilis* on the Lake Michigan Islands contradicts both the distance effect, which predicts that on islands of similar size the number of species vary inversely with the distance from the mainland, and the size effect which predicts that species numbers vary directly with the area available for immigration as proposed by the island biogeography theory of MacArthur and Wilson (1967). First, the non-conformity with the distance effect is illustrated in Table 1, by Marion Island, a relatively small island within 3 km of the mainland having as few species of fleas from this host as Squaw and Whiskey Islands, which are similar sized islands, but which are more than 15 km from the mainland. This is further shown by Gull Island, which is the most isolated of the islands, and has one more flea species than the three previous islands. Trout Island, of about the same size and distance from the mainland, is tied for the greatest number of flea species from this host for any of the 13 Lake Michigan islands studied.

Second, the size effect shows a lack of conformity with the theory because the largest islands, North and South Manitou Islands, North and South Fox Islands, and Beaver Island have only three and four species each. This is fewer species than the 5 species of either Hog Island or Trout Island which are considerably smaller. In addition, Garden Island with only 2 species, and one of the largest islands studied has only 2 recorded species of fleas and is within 4 km of Trout and Hog Island which have the most flea species.

I recognize several reasons for caution in using these data from the mouse flea species and islands to measure the validity of the island biogeography theory of MacArthur and Wilson (1967). These are: (1) I have no certainty that I have collected all the flea species from each island; (2) I have no indication of what levels of equilibrium between extinction and immigration are in these islands, much less whether those levels have been reached or not; (3) the area effect, where an increase in area lowers extinction, seems to be unrelated to the sizes of the islands in this study; and (4) the distance effect, lowering immigration in direct proportion to the distance from the mainland, varies depending on whether a person hypothesizes immigration from the Wisconsin side, the Upper Peninsula or the Michigan Mainland.

The distribution of flea species from the Eastern Chipmunk *Tamias striatus* (L.) is given in Table 2. *Megabothris acerbus* (Jordan) is found on all 5 islands where *T. striatus* was collected. *Tamiophila grandis* (Rothschild) is found only on this host on South Manitou Island and South Fox Island, and was recorded from the Fox Squirrel *Sciurus niger* (L.) from North Manitou Island. The only other Michigan records of this flea are from chipmunks on the mainland of Leelanau County (Scharf and Stewart 1980). This indicates that the colonization route may have been from the adjacent mainland for this host on the southern islands of the archipelago. Chipmunks are absent from most of the islands where we failed to collect them with the exceptions of Beaver, Marion, and North Fox islands.

The other four hosts and their fleas are listed here as unremarkable occurrences both because of the limited number of islands inhabited by each mammal species,
and the ubiquitous distribution of their fleas. The Red-backed vole, *Clethrionomys gapperi* (Vigors), harbored *Ctenophthalmus pseudagyrtes* Baker on North Manitou Island, and *Peromyscopsylla h. hesperomys* (Baker) on Beaver Island. The Meadow Vole, *Microtus pennsylvanicus* (Ord) was parasitized by *C. pseudagyrtes* on Marion Island (Scharf 1984). The Fox Squirrel *Sciurus niger* (L.) had *Orchopeas howardii* (Baker) on North Manitou Island in addition to the chipmunk flea noted above. I also collected *Corrodopsylla c. curvata* (Rothschild) from the Masked Shrew, *Sorex cinereus* Kerr, on North and South Manitou Islands.

ACKNOWLEDGMENTS

I thank the William R. Angell Foundation for financial support through their grants to Northwestern Michigan College for island biology studies during over 26 years of Great Lakes island studies. Cooperation from the Michigan Department of Natural Resources, Wildlife Division, on High and Garden Islands, the U. S. Fish and Wildlife Service on Gull Island, and the National Park Service on the Manitou Islands is gratefully acknowledged. The following persons in addition to numerous generations of NMC Natural History of Vertebrates and Ecology students helped trap and collect fleas: T. Allan, B. Busch, M. Fitch, J. Haswell, G. Hansen, M. Hindelang, P. Lederle, L. Osterlin, J. Mason, J. Rogers, G. Shugart, V. Shugart, E. Scharf, K. Scharf, K. Stewart, W. Wagoner, and R. Zillman. I also thank Dr. Omer R. Larson for early encouragement in the study of Siphonaptera.

LITERATURE CITED


Benton, A. H. 1983. An illustrated key to the fleas of the eastern United States. Bioguide No. 3 Marginal Media, Fredonia, N. Y.


ACORNS AS BREEDING SITES FOR *CHYMOMYZA AMOENA* (LOEW) (DIPTERA: DROSOPHILIDAE) IN VIRGINIA AND MICHIGAN

Henretta Trent Band

**ABSTRACT**

*Chymomyza amoena* is the only chymomyzid fly emerging from white oak acorns in Virginia. An average of 2-3 adult flies emerged from a single acorn in July while emergence declined to 0.4 adults/acorns in September. In fall, *Drosophila melanogaster* was also present. The incidence of drosophilid (*Drosophila, Chymomyza*) larvae in parasitized acorns in Virginia (40%) in autumn was significantly greater than in Michigan (14%). The *Chymomyza* larvae present in the parasitized acorns in Michigan most likely were *C. amoena*, from the known adaptation of this species in Michigan to frass-breeding.

Williams (1989a) has compiled a list of North American nut-infesting insects and their host plants. *Chymomyza amoena* (Loew) was among the insects contributing to northern red oak (*Quercus rubra*) acorn decay in Illinois (Winston 1956) and larvae were found in acorns of mixed oak *Quercus* species in West Virginia (Dorsey et al. 1962). Drosophilids do not everywhere use the same breeding substrates. For instance, *Drosophila pseudoobscura* Frolowa and *D. persimilis* Dobzhansky and Epling breed in slime fluxes at Mather, California (Carson 1951, Carson et al. 1956) but not at Blodgett Forest 127 km distant (Spith 1987). The rarity of slime fluxes led Spith (1987) to discover that drosophilids could breed in California black oak *Quercus kelloggii* acorns, if moist. *Drosophila subobscura* Collin in England breeds in rowan berries but not elderberries; the reverse occurs in Switzerland (Burla et al. 1987).

*Chymomyza amoena* breeds in a variety of fruits in Michigan, including domestic (commercial) apples *Malus pumila*, in apples in Virginia and in black walnuts *Juglans nigra* in Michigan (Band 1988a,b,c,d, 1989a). Larvae have been found to overwinter in a variety of substrates in Michigan (Band and Band 1984, 1987) and in apples in Virginia (Band 1988a). The question arises; does this species also breed in acorns in the two states? Here I demonstrate *C. amoena* emergence from acorns in Virginia. Larval adaptation to frass breeding arising from female exploitation of parasitized substrates for oviposition (Band 1988a,b,d, 1989a) suggests that *C. amoena* is also the chymomyzid in acorns in Michigan.

**MATERIALS AND METHODS**

Studies in Virginia were carried out at Mountain Lake Biological Station (MLBS), elev. approx 1200 m, Giles Co. Virginia, during the summer and fall of 1989 and at

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two sites in Michigan in the fall of 1989. The 600 acre MLBS site with mixed deciduous and evergreen forests is adjacent to the 100,000 acre Jefferson National Forest. Other Chymomyza and Drosophila species occur in Giles Co., Virginia (Band 1988c,d, 1989b). Chymomyza eggs characteristically have a crown of short filaments (Sturtevant 1921, Throckmorton 1962, Schumann 1987). Larvae can be distinguished by the posterior spiracular region (Hackman et al. 1970). To date, C. amoena is the only chymomyzid consistently recorded in frass or frassy interiors (Dorsey et al. 1962, Band 1988a).

Study sites in Michigan were stands of oaks. In the northern lower peninsula the site was between Gaylord and Grayling adjacent to US Interstate route 75 and in mid-Michigan the site was a stand of oaks near a housing development in Meridian Twp., Ingham Co. off Grand River Ave. Chymomyza procnemoides has also been collected in Michigan as well as Virginia (Wheeler 1952) but its breeding sites are unknown.

Virginia. White oak acorns identified as Quercus alba were collected at two locations near the Station on 12 July and 25 July 1989. The acorns collected were rejects among piles of consumed acorns abandoned by squirrels or were unpiled acorns lying 1–2 m adjacent to a foot trail through the woods. All acorns were microscopically inspected for the presence of Chymomyza larvae and/or eggs and tentatively identified as C. amoena. Larvae were reared in a glass population jar over moist paper toweling. Some of the emerging C. amoena adults were transferred to apple + frass to determine if these acorn flies were comparable to C. amoena emerging from other substrates (Band 1988a,b) and could utilize apples as a food source; the rest were maintained on high protein laboratory medium (Band 1988a).

On 30 September 1989 acorns were more rigorously collected on the ground in order to assess the frequency of those damaged and those infested by drosophilid larvae. All were transported back to Michigan State University where acorns with holes were separated, dissected and inspected for the presence of drosophilid larvae or eggs.

Michigan. On 16 October 1989 large numbers of red oak acorns were collected at a site in northern lower (NL) Michigan in Crawford Co. and on 20 November in mid-Michigan in Ingham Co. (EL). Both groups were sorted, and examined following the same protocols as those for the Virginia Fall season collection.

Data analysis. G-statistics have been calculated according to Sokal and Rohlf (1969).

RESULTS

Virginia. Chymomyza amoena was the only chymomyzid to emerge from the infested acorns. Eleven of 16 acorns collected in mid-July contained 31 C. amoena larvae and 16 of 20 acorns collected in late July had 133 eggs and larveae. Eggs were found in the vascular elements at the base of the acorn or inside on frassy pulp, as found by Spieth (1987) for drosophilid oviposition in acorns in California. However, no other drosophilids were detected in summer. Table 1 records the numbers of adults emerging from the July and September MLBS acorn collections. Adults emerging readily oviposited on apples + frass; 76 adults emerged and produced an F2. Ten acorn emergents showed the ability to switch to a fruit substrate and retain fertility, as expected (Band 1988a). As observed for C. amoena egg counts in apples (Band, 1988a), egg totals in acorns exceeded the numbers of adults emerging.

Twenty-two percent of the acorns collected in September in Virginia were damaged (n = 216) of which 40% had drosophilid larvae inside. No acorns collected in September contained eggs on the outer surface of damaged or undamaged acorns. Only parasitized acorns were used for drosophilid oviposition in September among the newly fallen acorns but the initial parasite, probably a curculionid larva, had
Table 1.— *Chymomyza amoena* emergence from damaged white oak acorns in Virginia in July and September 1989.

<table>
<thead>
<tr>
<th>Month</th>
<th>No. infested acorns</th>
<th>Emerged species</th>
<th>No. adults emerged</th>
<th>Ave. no. adults emerged per acorn</th>
</tr>
</thead>
<tbody>
<tr>
<td>mid-July</td>
<td>11</td>
<td><em>C. amoena</em></td>
<td>24</td>
<td>2.2</td>
</tr>
<tr>
<td>late July</td>
<td>16</td>
<td><em>C. amoena</em></td>
<td>50</td>
<td>3.1</td>
</tr>
<tr>
<td>September</td>
<td>17</td>
<td><em>D. melanogaster</em></td>
<td>4</td>
<td>0.2</td>
</tr>
<tr>
<td></td>
<td></td>
<td><em>C. amoena</em></td>
<td>7</td>
<td>0.4</td>
</tr>
</tbody>
</table>

Table 2.— Comparison of acorns infested with *Chymomyza amoena* and other drosophilid larvae from Mt. Lake Biological Station (MLBS), VA, Northern Lower (NL) Michigan and East Lansing (EL), MI in Fall, 1989.

<table>
<thead>
<tr>
<th>State</th>
<th>Location</th>
<th>Total</th>
<th>Undamaged (%)</th>
<th>Acorns</th>
<th>No. Damaged</th>
<th>No. larvae</th>
<th>With larvae (%)</th>
<th>(%) Total</th>
</tr>
</thead>
<tbody>
<tr>
<td>Virginia</td>
<td>MLBS</td>
<td>216</td>
<td>(79%)</td>
<td></td>
<td></td>
<td>29</td>
<td>19 (40%)</td>
<td>48</td>
</tr>
<tr>
<td>Michigan</td>
<td>NL</td>
<td>210</td>
<td>(95%)</td>
<td></td>
<td></td>
<td>7</td>
<td>3 (30%)</td>
<td>10</td>
</tr>
<tr>
<td>Michigan</td>
<td>EL</td>
<td>327</td>
<td>(92%)</td>
<td></td>
<td></td>
<td>22</td>
<td>2 (8%)</td>
<td>24</td>
</tr>
</tbody>
</table>

Comparison among damaged acorns: d.f. = 2; G = 8.72; P 0.05

exited. Five *Drosophila* larvae and 28 *C. amoena* larvae were counted in the 19 acorns; 4 *D. melanogaster* and 7 *C. amoena* adults emerged.

**Michigan.** Six percent of the total red oak acorns collected (n = 537) were damaged with 14% of these containing drosophilid larvae (Table 2). These infestation rates are significantly lower than that encountered in Virginia. The intervening month between collecting in northern lower Michigan and in mid-Michigan did not increase the rate of drosophilid acorn infestation in mid-Michigan despite area wide *C. amoena* overwintering/substrate utilization studies in past years which established its presence in the greater Lansing/East Lansing area and adjacent communities (Band and Band 1984).

Michigan *C. amoena* females also oviposited only in parasitized acorns. As in Virginia in September, no eggs or remnants of egg cases were detected in vacular elements at the base of the acorns. There were 1 *Drosophila* and 4 *C. amoena* larvae in the 3 NL acorns, 5 *C. amoena* larvae in the 2 EL acorns.

**DISCUSSION**

Sturtevant (1921) was the first to report that *C. amoena* bred in a variety of fruits and nuts: Banks bred it from acorns, Shannon from walnut and butternut husks, he and Metz obtained it from apples and bananas (p. 61). Winston (1956) reared all larvae to adulthood, thereby determining that *C. amoena* bred in parasitized and decaying acorns. He labeled them fungal feeders and scavengers. Dorsey et al. (1962) found that curculionid larvae were the primary acorn pests in West Virginia forests and listed *C. amoena* among the secondary invaders. Sampling over a year's time, beginning in September, Winston (1956) found that larval-infested acorns were rejected by squirrels. Weckerly et al. (1989) discovered that intact acorns from which curculionid larvae had not emerged were consumed by squirrels. Winston (1956) also found that moisture was important, as Spieth (1987) did later.

Since discovering that *C. amoena* in mid-Michigan was using such substrates as unripe firm apples for larval development in summer (Band 1988a,b) and firm
native crabapples *Malus coronaria* for overwintering (Band and Band 1984), my interests have included comparative substrate use and how females are able to oviposit in firm substrates. Numbers emerging from acorns versus initial numbers of eggs and larvae show a sharp reduction as in apples (Band 1988a). Exit holes made by initial pest larvae (Moffett 1989, Williams 1989b) provide an entrance way, analogous to pest larvae breaking the surface in a developing apple. For Virginia, these findings indicate that the surrounding forest may be the reservoir from which *C. amoena* can invade fallen and un fallen apples yearly. Two major summer sites which have larval-infested apples lack fallen apples by late Fall (Band 1988c). The ability to invade and breed in acorns damaged by primary insect attackers indicates an inter-species dependency that may contribute to the survival of this species in forested and urban areas. The Virginia (reported here), West Virginia (Dorsey et al. 1962) and Illinois (Winston 1956) studies also indicate *C. amoena* was a forest/woodland species where it evidently was breeding in such native nuts and fruits as acorns, black walnuts, and endemic crabapples before the colonists arrived.

The significantly lower numbers of infested Michigan acorns as compared with Virginia may reflect a lower population density and/or totally migrant female oviposition. Neither site is immediately adjacent to areas in northern lower Michigan or mid-Michigan used in past studies on overwintering/substrate utilization (Band and Band 1982, 1984). Biotic factors as availability of substrates and abiotic factors as altitude, latitude and past glaciation history influence species composition. Oviposition behavior on fruits in summer has been strikingly similar in Michigan and Virginia (Band 1989a).

The presence of *D. melanogaster* in acorns in Virginia is not unexpected. *Drosophila* species can breed in parasitized acorns in California (Spieth 1987) and have emerged from frass in Hawaii (Heed 1968). Three species have adapted to the nephritic gills of crabs (Carson 1974). To date however, *C. amoena* has been the only chymomyzid in parasitized acorns, a female oviposition behavior and larval developmental adaptation that may have enabled this species to follow other pests into unripe parasitized domestic fruits. Breeding sites and breeding behavior of this species in Europe (Bächli and Rocha Pité 1982, Schumann 1987) remain unknown. Breeding sites for other forest *Chymomyza* in the East remain unknown.

**NOTE ADDED**

In a letter dated 16 Oct. 1990, Dr. Hans Burla, Zoological Museum, University of Zurich, reported that he had bred 10 females and 14 males of *C. amoena* from 100 chestnuts, *Castanea sativa* (= *vulgaris* = *vesica*), collected in a forest in the Ticino Canton of Switzerland. The chestnuts were old, and had been collected in the forest, not far from the edge. The shipment also contained crabapples, apples, pears and plums. Thirteen *C. amoena* adults emerged from the 13 apples.

Dr. Burla verified that this species of *Castanea* is native to Europe. This again revives the comment initially made by Bächli and Rocha Pité (1981) after *C. amoena* was collected in eastern Europe, that this species may be introduced, and capable of spreading under favorable conditions as *C. procnemis* did in Japan (Okada 1976) or may indicate that *C. amoena* has a Holarctic ditribution.

**ACKNOWLEDGMENTS**

Appreciation is gratefully extended to Jim Murray, Director, University of Virginia's Mountain Lake Biological Station for providing research space in summer, 1989. I thank Spencer Tomb for identifying the *Quercus alba* acorns and Jon Beaman the
Quercus rubra acorns. Reviewers' comments have been helpful in revising the manuscript.

LITERATURE CITED


LIST OF THE LEPIDOPTERA OF BLACK STURGEON LAKE, NORTHWESTERN ONTARIO, AND DATES OF ADULT OCCURRENCE

C.J. Sanders

ABSTRACT

From May to September each year from 1960 through 1968, a collection of Lepidoptera was made at Black Sturgeon Lake, northwestern Ontario, from specimens captured in a light trap and from specimens netted during the day. A total of 564 species was recorded from 70 families. A list of the species with dates of capture is presented.

From 1960 through 1968, a 15-watt black-light trap was operated each year at a Forestry Canada field station at Black Sturgeon Lake, northwestern Ontario. The trapping program was initiated by R.E. Fye in 1960 and was taken over by C.J. Sanders in 1965. Each year from 1960 through 1968, the light trap was set up when the station was opened in early May and it was operated continually until the station was closed in mid-to late September. In 1969 the research program at the Black Sturgeon Lake field station was curtailed, and operation of the light trap was discontinued.

Black Sturgeon Lake is located about 100 km from the northwestern shore of Lake Superior, 110 km northeast of Thunder Bay at latitude 49°20'N, longitude 88°54'W, at an altitude of 250 m, in the Superior (B9) Boreal Forest Region of Rowe (1972) (Fig. 1). The corresponding Universal Transverse Mercator coordinates are 1636546. The physical, environmental and vegetational characteristics of the study site have been described by Lethiecq and Regniere (1988). The site lies in the Black Sturgeon District of the Lake Nipigon Site Region, which is characterized by: "Flat topped basaltic ridges with deeper deposits—(i) granitic and clay forming sand, (ii) silt, and (iii) limy clay, in the depressions." (Hills 1959). The vegetation is boreal mixedwood, composed of fairly uniform stands of balsam fir (Abies balsamea), white spruce (Picea glauca), black spruce (P. mariana), white birch (Betula papyrifera), and trembling aspen (Populus tremuloides). Understory species include hazel (Corylus cornuta), alders (both Alnus crispa and A. rugosa), and mountain maple (Acer spicatum).

Continuous weather records are not available for the Black Sturgeon Lake area, so instead information is presented for Cameron Falls, which is 42 km to the southeast of Black Sturgeon Lake (49°09'N, 88°21'W, 229 m altitude), at the southern end of Lake Nipigon (Anon. 1980). The mean annual air temperature is 1.7°C. The mean low temperature for January is -16.6°C, with the lowest recorded temperature -46.1°C. The mean high for July is 17.0°C, with the highest temperature recorded 38.9°C. The mean annual rainfall is 559.1 mm, and the snowfall 233.6 cm.

Forestry Canada, Ontario Region, P.O. Box 490, Sault Ste Marie, Ontario, Canada, P6A 5M7.
The primary purpose of the light trap was to monitor the numbers of pest species such as spruce budworm (*Choristoneura fumiferana* [Clem.]), jack pine budworm (*C. pinus* pinus Free.) and forest tent caterpillar (*Malacosoma disstria* Hbn.), but the opportunity was taken to assemble a collection representative of the Lepidoptera in the area. The contents of the light trap were examined each morning, and representative specimens of each species were collected. Particular attention was paid to obtaining the first specimens that were captured in the light trap each year. Additional specimens of each species were collected thereafter throughout the flight period, but no attempt was made to collect them in proportion to the numbers that were collected in the trap. The collection is therefore an indication of the period over which the insects were captured, and is not an indication of their abundance. In addition to the light-trap material, specimens of day-flying Lepidoptera were collected by net.

All collected specimens were pinned and spread, and then sent for identification to the Biosystematics Research Centre (Agriculture Canada, Ottawa, Ontario, Canada) or, after a reference collection had been assembled, identifications were made by comparing specimens with the previously identified voucher specimens. Repre-
sentative specimens have been retained in the Canadian National Collection, in the collection of the Forest Insect and Disease Survey (Forestry Canada, Ontario Region, Sault Ste. Marie, Ontario) and in the personal collection of the author. Insects in the following listing are catalogued according to Hodges et al. (1983). Nomenclature has not been updated beyond that of Hodges et al. with the following two exceptions: Anavitrinella (checklist #6590) is incorrectly spelled by Hodges et al. and has been corrected to Anavitrinella (e.g., McGuffin 1977). Cydia youngana (Kft.) (checklist #3466) is now considered to be a junior synonym of C. strobilella (L.) (Brown and Miller 1983), and has been changed accordingly.

In several cases, entries at the subspecific level are accompanied by entries at the corresponding specific level. In these instances, specimens under the species label have not been identified to the subspecies level, and may therefore contain representatives of one or more subspecies, including the accompanying subspecies that is listed.

ANNOTATED LIST OF LEPIDOPTERA

HEPIALIDAE

19 Sthenopis purpurascens (Pack.) 15-30 Jul
20 Sthenopis quadriguttatus (Grt.) 15 Jul
29 Hepialus novigannus B. & Benj. 20 Aug-20 Sep

TINEIDAE

261 Nemagogon acapnopennella (Clem.) 25 Jun-5 Jul
264 Nemagogon defectella (Zell.) 15 Aug
421 Monopis spilotella Tengström 5 Jun-30 Jun

LYONETIIDAE

560 Bucculatrix canadensisella Cham. 20 Jun

GRACILLARIIDAE

587 Caloptilia alnivorella (Cham.) 15-30 May, 30 Jul-5 Aug, 20 Sep
601 Caloptilia coronellia (Clem.) 10-30 May
609 Caloptilia invariabilis (Braun) 10 May
655 Parectopa pennsylvaniella (Engel) 25 Jun

OECOPHORIDAE

911 Bibbarambla allenella (Wlsm.) 30 Jun
913 Semioscopia merriccella Dyar 20 May-5 Jun
914 Semioscopia inornata Wlsm. 25 Aug
916 Semioscopia aurorella Dyar 20 May

BLASTOBASIDAE

1144 Geradina caritella Bsk. 30 Jul-25 Aug

MOMPHIDAE

1458 Mompha unispeciella (Cham.) 30 Jun
<table>
<thead>
<tr>
<th>Family</th>
<th>Species</th>
<th>Dates</th>
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<tbody>
<tr>
<td>Yponomeutidae</td>
<td>Swammerdamia caesiella (Hbn.)</td>
<td>15-20 Jun, 15 Jul</td>
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<td>Sesiidae</td>
<td>Synanthedon acerni (Clem.)</td>
<td>25 Jun-5 Jul</td>
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<td>Choreutidae</td>
<td>Choreutis diana (Hbn.)</td>
<td>20 Jul-10 Aug</td>
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<td>Cossidae</td>
<td>Acossus centerensis (Lint.)</td>
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<td>Tortricidae</td>
<td>Endothenia albolineana (Kft.)</td>
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<td>Endothenia impudens (Wism.)</td>
<td>20-30 Jul</td>
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<td>Apotomis funerea (Meyr.)</td>
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<td>Apotomis spinulana (McD.)</td>
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<td>Apotomis dextrana (McD.)</td>
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<td>Olethreutes connectus (McD.)</td>
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<td>Olethreutes constellatana (Zell.)</td>
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<td>Olethreutes astrologana (Zell.)</td>
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<td>Olethreutes bipartitana (Clem.)</td>
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<td>Petrova albicapitana (Bsk.)</td>
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<td>Eucosma dorsiSIGNatana similana (Clem.)</td>
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<td>Notocelia culminana (Wism.)</td>
<td>20 Jul-10 Aug</td>
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<td>Griselda radicana Heins.</td>
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<td>Hopobota unipunctana geminana</td>
<td>(Steph.) 20-25 Jul</td>
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<td>Epinotia stroemiana (F.)</td>
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<td>Epinotia medioviridana (Kft.)</td>
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<td>Epinotia rectiplicana (Wism.)</td>
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<td>Epinotia solitaria (Wlk.)</td>
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<td>Ancylis mediofasciana (Clem.)</td>
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<td>Grapholitha packardi Zell.</td>
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<td>Cydia strobilella (L.)</td>
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<td>Croesia albicollana (Clem.)</td>
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<td>Acleris macdunnoughi Obr.</td>
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<td>Acleris siemiania (Rob.)</td>
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<td>Acleris celiana (Rob.)</td>
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<td>Chloristoneura fraxinellana (Clem.)</td>
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<td>Chloristoneura parallela (Rob.)</td>
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<td>Chloristoneura rosea (Harr.)</td>
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<td>Chloristoneura conficta (Wlk.)</td>
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<td>Chloristoneura fumiferana (Clem.)</td>
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<td>Chloristoneura pinus Free.</td>
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<td>Archips argyrospila (Wlk.)</td>
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<td>Archips purpurana (Clem.)</td>
<td>5-15 Aug</td>
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3661 Archips cerasivorana (Fitch) 30 Jul-10 Aug
3664 Archips striana (Fern.) 25 Jun-20 Jul
3665 Archips alberta (McD.) 10-30 Aug
3672 Syndemis affliclana (Wlk.) 20-25 Jun
3681 Clepsis persicina (Fitch) 5-20 Jul
3684 Clepsis melaleuca (Wlk.) 30 Jun
3686 Plycholoma perilana (Clem.) 20-25 Jul
3687 Plycholoma virescana (Clem.) 20-25 Jun
3697 Sparganothis lycopodiana (Kft.) 15 Aug
3706 Sparganothis xanthoides (Wlk.) 10-20 Jul
3720 Sparganothis reticulatana (Clem.) 30 Jul-10 Aug, 20 Sep
3740 Sparganothis idaeusalis (Wlk.) 25 Jul

COCHYLIDAE
3787 Carolella vilellinana (Zell.) 25-30 Jun
3802 Hyslerosia riscana (Zell.) 15-20 Jul

HESPERIIDAE
3910 Thorybes pylades (Scudder) 10 Jul
3945 Erynnis icelus (Scudder & Burgess) 20 Jun
4041 Polites nemesis (Scudder & Burgess) 20 Jul
4105 Amblyscirtes virescens (Edw.) 20 Jun

PAPILIONIDAE
4176 Papilio glaucus L. 30 May-20 Jun

PIERIDAE
4195d. Artogeia napi oleracea (Harr.) 15 Jun, 30 Jul-5 Aug
4197 Artogeia rapae (L.) 10 Jul
4210 Colias eurytheme Bdv. 30 Jun, 20 Aug
4220 Colias interior Scudder 15 Jul

LYCAENIDAE
4362 Everes amyntula (Bdv.) 15-20 Jun
4372 Gláscopusche lygdamus (Doubleday) 15 Jun

NYMPHALIDAE
4423 Polygonia faunus (Edw.) 5-25 Aug
4429 Polygonia progne (Cram.) 5 Aug, 25 Aug, 20 Sep
4430a. Nymphalis vaux-album j-album (Bdv.& Leconte) 5 Aug, 5-15 Sep
4432 Nymphalis antiopa (L.) 30 Jun, 5 Sep
4433 Aglais milbertii (Godt.) 5 Aug
4435 Vanessa cardui (L.) 25 May-5 Jun
4459 Speyeria atlantis (Edw.) 20-30 Jul
4564f. Clossiana seinea arocostalis (Huard) 30 Jun, 30 Jul-5 Aug
4465a. Clossiana bellona toddi (Holl.) 20 Jun
4481 Phycides tharos (Drury) 30 Jul-30 Jul
4490 Charidryas nycteris (Doubleday) 25 Jun
4522 Basilarchia arthemis (Drury) 30 Jun-15 Jul

SATYRIDAE
4568 Enodia portlandia (F.) 15-20 Jul

LIMACODIDAE
4652 Tortricidia testacea Pack. 30 Jun-25 Jul

PYRALIDAE
4703b. Gesneria centuriella caecalis (Wlk.) 10-15 Jul
4716 Scoparia biplagialis Wlk. 30 Jun, 20 Jul-5 Aug
4737 Eudonia lugubralis (Wlk.) 15-20 Jun, 5 Jul
4748 Munroesia icciusalis (Wlk.) 15-30 Jul
4759 Parapoynx maculalis (Clem.) 10-20 Jul, 10 Aug
4760 Parapoynx obscuralis (Grt.) 20-25 Jul
4897 Evergestis pallidata (Hufn.) 10-25 Jul
4935 Saucrobotys fumoferalis (Hulst) 20-30 Jun
4936 Saucrobotys fuitalis (Led.) 10-20 Jul
4950 Fumibotys fumalis (Gn.) 20-25 Jul, 25 Aug-10 Sep
4953a. Phlyctaenia coronata tertiata (Gn.) 15-25 Jun
4956a. Nealgedonia extraialis dional (Wlk.) 20-25 Jun
4957 Muturaauia mysippusalis (Wlk.) 25-30 Jun
5060a. Pyrausta subequalis borealis Pack. 10 Jun
5079 Udea rubigalis (Gn.) 30 May, 15 Jun, 30 Jun
5156 Nomophila nearctica Mun. 10 Jun-20 Jul, 20 Sep
5159 Desmia funeris (Hbn.) 30 Jun-10 Jul
5262 Framinghamia helvalis (Wlk.) 25 Aug
5275 Herpetogramma pertexalis (Led.) 10-25 Aug
5339 Crambus pascuellus (L.) 30 Jun-5 Jul, 20 Jul, 20 Aug
5341a. Crambus alienellus labradoriensis Christoph 10 Jul
5343a. Crambus perlellus innotateillus Wlk. 15 Jul-5 Aug
5344 Crambus unistriaeillus Pack. 25 Jul-10 Aug
5357 Crambus leachellus (Zinck.) 10 Aug-5 Sep
5362 Crambus agitatiellus Clem. 20 Jul
5379 Crambus luteoellus Clem. 15-30 Jun
5380 Crambus zeellus Fern. 20 Jul
5391 Chrysoteuchia topariella (Zell.) 30 Jun-20 Jul
5403 Agriphila vulgivagella ( Clem.) 10-20 Aug
5408 Catoptria lattradiella (Wlk.) 25 Jul-15 Aug
5413 Pediasia trisecta (Wile.) 10-20 Aug
5416 Aglossa caprealis (Hbn.) 5-30 Jul
5421 Herculia thymetusalis (Wile.) 30 Jun-15 Jul
5429 Herculia thymetusalis (Wile.) 30 Jun-15 Jul
5430 Acrobasis tricolorella Gtr. 15-30 Jun
5438 Acrobasis betulella Hulst 30 Jun-10 Aug
5439 Myelopsis coniella (Rag.) 10 Aug
5443 Myelopsis subtelricella (Rag.) 5-20 Jun
5447 Apomyelois bistriatella (Hulst) 15 Aug, 10 Sep
5453 Glyptocera consobrinella (Zell.) 25 Jun
5455 Nephopterix carneella (Hulst) 20-30 Jun
5460 Telethusia ovalis (Pack.) 15 Aug
5466 Eulogia ochrifrontella (Zell.) 30 Jun-10 Jul, 15 Aug
5470 Dioryctria reniculelloides Mutun. & Mun. 10 Jul, 10-15 Jul, 20-25 Jul
5478 Euchlaena obtusaria (Hbn.) 30 Jun-5 Jul
5480 Euchlaena madusaria (Wlk.) 5-20 Jul
5483 Euchlaena marginaria (Minit.) 30 May-15 Jun
5488 Orthojidonia tinctaria (Wlk.) 5-10 May, 15 Jun
5495 Orthojidonia tinctaria (Wlk.) 5-10 May, 15 Jun
5498 Anacamptodes ephyraria (Wlk.) 30 Jun-20 Jul
5501 Anacamptodes ephyraria (Wlk.) 30 Jun-20 Jul
5506 Anacamptodes ephyraria (Wlk.) 30 Jun-20 Jul
5509 Anacamptodes ephyraria (Wlk.) 30 Jun-20 Jul
5512 Anacamptodes ephyraria (Wlk.) 30 Jun-20 Jul
5516 Euchlaena obtusaria (Hbn.) 30 Jun-5 Jul
5521 Euchlaena madusaria (Wlk.) 5-20 Jul
5526 Euchlaena marginaria (Minit.) 30 May-15 Jun
5531 Euchlaena marginaria (Minit.) 30 May-15 Jun
5536 Euchlaena marginaria (Minit.) 30 May-15 Jun
5541 Euchlaena marginaria (Minit.) 30 May-15 Jun
5546 Euchlaena marginaria (Minit.) 30 May-15 Jun
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5816 Euchlaena marginaria (Minit.) 30 May-15 Jun
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<tr>
<th>Species</th>
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<tr>
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8005  Schizura ipomoeae  Doubleday  5-20 Jul
8007  Schizura unicornis  (J. E. Smith)  15 Jul-5 Aug
8011  Schizura leptirodes  (Grt.)  20 Jun-25 Aug

ARCTIIDAE

8045.1  Crambidia pallida  Pack.  25 Aug
8051  Crambidia casta  (Pack.)  20 Aug-10 Sep
8090  Hypoprepia fuscata  Hbn.  10 Aug
8098  Clemensia albata  Pack.  10-25 Aug
8111  Haploa lecontei  (Guer.-Ménéville)  30 Jun-20 Jul

8114a.  Holomelina laevis  treatii  (Grt.)  25 Jun-15 Jul
8123  Holomelina ferruginosa  (Wlk.)  25 Jun-20 Jul, 5 Aug
8127  Paraseis plantaginis  (L.)  25 Jun-10 Jul
8134  Spilosoma congener  Wlk.  5-20 Jun
8136  Spilosoma dubia  (Wlk.)  20 Jun
8137  Spilosoma virginica  (F.)  20-30 Jun
8158  Phragmaphobia assimilans  (Wlk.)  25 May-10 Jun
8162  Platharchia parthenos  (Harr.)  20 Jun-7 Jul
8166  Arctia caja  (L.)  15-30 Aug
8175  Apanites virguncula  (W. Kirby)  25-30 Jul
8176  Apanites anna  (Grt.)  25 Jul
8186  Apanites williamsonii  (Dodge)  5-15 Jul
8187  Apanites celia  (Saund.)  20 Jul
8196  Apanites parthenice  (W. Kirby)  15 Jul-20 Aug
8197  Apanites virgo  (L.)  15 Jul
8214  Lophocampa macula  (Harr.)  20-30 Jun
8262  Ctenucha virgulica  (Esp.)  10 Jul-5 Aug
8267  Cisseps fulvicollis  (Hbn.)  10-20 Jul

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8322  Idia americalis  (Gn.)  25 Jul, 10 Aug
8334  Idia luridalis  (Gey.)  25 Jul-10 Aug
8338  Phalaenopha pyramidalis  (Wlk.)  10 Jun
8341  Zanclognatha theralis  (Wlk.)  5-20 Jul
8349  Zanclognatha protumulus  (Wlk.)  5 Jul
8352  Zanclognatha jacchusalis  (Wlk.)  15 Jul
8356  Chrytolita petrealis  Grt.  20-30 Jun
8362  Phalaenosta leuconoealis  (Wlk.)  5-25 Jul
8370  Blepina caradrinialis  Gn.  10 Jul, 20 Aug
8397  Palithis angularis  (Hbn.)  15-25 Jun
8413  Mysterothra inexplicata  (Wlk.)  10-20 Jul
8443  Bomolocha biuncialis  (Wlk.)  25 Jul
8444  Bomolocha palparia  (Wlk.)  5 Jul
8455  Lomaleites eductalis  (Wlk.)  25 Jul
8461  Hyposia humuli  Harr.  20 Aug
8479  Spargaloma sexpunctata  Grt.  30 Jun-10 Jul, 5 Aug
8490  Pangrapha decoralis  Hbn.  30 Jun

8500  Metalectra quadrifasciata  (Wlk.)  30 Jun-10 Jul, 25 Jul
8554  Alabama argillacea  (Hbn.)  5 Sep
8694  Zale aeruginosa  (Gn.)  25 May-15 Jun
8697  Zale minerea  (Gn.)  5-15 Jun
8697a.  Zale minerea nuda  (Sm.)  25 Jun-5 Jul
8703  Zale duplicata  (Bethune)  10-15 Jun
8716  Zale unicolor  (Grt.)  25 May-5 Jun
8731  Euclidia cuspida  (Hbn.)  10 Jun-5 Jul
8738  Caemergina crassiuscula  (Haw.)  25 Jun, 30 Jul, 15 Aug
8803  Catocala relicta  (Wlk.)  20 Aug-5 Sep
8805  Catocala unijuga  (Wlk.)  5-20 Aug
8817  Catocala briseis  Edw.  20 Aug-20 Sep
8821  Catocala semirelictta  Grt.  20 Aug
8833  Catocala concumbens  Wlk.  30 Aug-10 Sep
8846  Catocala sordida  Grt.  5 Aug
8857  Catocala ultronia  (Hbn.)  20 Aug-15 Sep
8867  Catocala blandula  Hulst  30 Jul-25 Aug
8896  Diachrysia aeroides  (Grt.)  25 Jul-10 Aug
8897  Diachrysia ballica  Gey.  25 Jul-15 Aug
8899  Pseudeva purpurigera  (Wlk.)  30 Jul-10 Aug, 25 Aug
8904  Chrysarctia forbesia  (Grt.)  5 Jul-15 Aug
8908  Autographa precationis  (Gn.)  15 Jun, 15-20 Jul, 25 Aug, 20 Sep
8909  Autographa rubida  Ottol.  15 Jun-5 Jul
8911  Autographa bimaculata  (Steph.)  25 Jul-25 Aug
8912  Autographa mappa  (G. & R.)  30 Jun-5 Aug
8913  Autographa pseudogamma  (Grt.)  5-15 Jul
8916  Autographa flagellum  (Wlk.)  25-30 Jul, 5 Sep
8923  Autographa ampla  (Wlk.)  15 Jul-15 Aug
8924  Anagrapha falcinera  (Kby.)  25 May, 15-30 Jun, 25 Aug-10 Sep
8926  Syngrapha octoquadra  (Grt.)  10 Jul, 5-25 Aug
8927  Syngrapha epigaea  (Grt.)  15-25 Aug
8928  Syngrapha selecta  (Wlk.)  15 Jul-20 Aug
8939  Syngrapha alisa  (Ottol.)  25 Jun, 10 Aug
8942  Syngrapha rectangula  (W. Kirby)  5 Jul, 25 Jul, 15-25 Aug
8945  Syngrapha montana  (Pack.)  30 Jun
8946a.  Syngrapha microgemma  neartica  Fgn.  30 Jun
8950  Plusia putnami  Grt.  30 Jun-10 Sep
8953  Plusia venusta  Wlk.  30 Jul-5 Aug
8969  Baileya doubledayi  (Gn.)  30 Jun
8970  Baileya ophthalmica  (Gn.)  5 Jun
8974  Charocama nilotica  (Rogenhofer)  25 Jun, 30 Jul
8946  Lithacodia bellicula  Hbn.  10 Jul
8948  Lithacodia albida  (Gn.)  20-25 Jun
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9053 *Lithacodia carneola* (Gn.) 30 Jun-15 Jul
9059 *Capis curvata* Grt. 10-20 Jul
9090 *Tarachidia candeformis* (Hbn.) 15 Sep
9177 *Panthea acronyctoides* (Wlk.) 20 Jun-5 Jul
9183 *Panthea pallescens* McD. 30 Jun, 25-30 Jul
9183a *Panthea pallescens centralis* McD.
9185 *Colocasia propinquilinea* (Grt.) 30 May-15 Jun, S Jul
9193 *Raphia frater* Grt. 15-20 Jun
9203 *Acronicta dactylina* Grt. 20-30 Jun, 5 Sep
9205 *Acronicta lepusculina* (Gn.) 20 Jun
9207 *Acronicta innotata* (Gn.) 5-25 Jun, 10-20 Jul, 10-20 Aug
9212 *Acronicta grisea* Wlk. 20 lun-10 Jul
9226 *Acronicta superans* (Gn.) 25-30 Jun
9229 *Acronicta hasta* (Gn.) 25 Jun
9241 *Acronicta fragilis* (Gn.) 20-25 Jun, 10-15 Jul, 15 Aug
9257 *Acronicta impleta* Wlk. 20 Jun
9259 *Acronicta noctivaga* Grt. 10-20 Jul
9286 *Harrisimemna trisignata* (Wlk.) 30 Jun-5 Jul
9326 *Apamea verbascoldes* (Gn.) 20 Jul-S Sep
9344 *Apamea plutonla* (Grt.) 25 Jul-5 Aug
9351 *Apamea alia* (Gn.) 25 Jun, 15 Jul
9359 *Apamea commoda* (Wlk.) 5-30 Jul
9360 *Apamea noctivaga* Grt. 10-20 Jun
9367 *Agroperlna dubilans* (Gn.) 30 Jun-15 Jul
9367a *Agroperlna dubitans cogitata* (Sm.) 10-15 Jul, 30 Jul, 25 Aug
9374 *Protargotis niveivenosa* (Grt.) 5-10 Aug
9382 *Cymodes devastator* (Brace) 10 Aug-5 Sep
9391 *Luperina passer* (Gn.) 10 Aug
9415 *Oligia bridghaml* (G. & R.) 25 Aug
9419 *Oligia maculata* (Gn.) 20 Jul, 20 Aug-15 Sep
9420 *Oligia ilicola* (Wlk.) 20-30 Jul, 25 Aug
9431 *Parastictis discivaria* (Wlk.) 15 Aug-5 Sep
9437 *Hypocoena inquinata* (Gn.) 25 Aug
9439 *Hypocoena basistigma* (McD.) 5-20 Sep
9449 *Archanae oblonga* (Grt.) 5-10 Sep
9450 *Archanae sutflava* (Grt.) 5 Sep
9453 *Helotropha reniformis* (Grt.) 5-20 Sep
9454 *Amphipoea velata* (Wlk.) 20 Aug
9457 *Amphipoea americana* (Speyer) 5 Aug-10 Sep
9472 *Papaipema harrisii* (Grt.) 20 Aug-20 Sep
9474 *Papaipema sauzalitae* (Grt.) 30 Aug-5 Sep
9480 *Papaipema pterisii Bird* 20 Sep
9509 *Papaipema unimoda* (Sm.) 5-25 Sep
9520 *Achatodes zea* (Harr.) 30 Jul, 25 Aug
9523 *Bellura gortynoides* f. diffusa (Grt.) 25 Jun
9525 *Bellura obliqua* (Wlk.) 20 Jun-20 Jul
9545 *Euplexia benesimilis* McD. 25-30 Jun
9546 *Philogophora iris* Grt. 20-30 Jun
9547 *Philogophora periculosa* Grt. 30 Jul-25 Aug
9549 *Enarga decolor* (Wlk.) 15 Aug-20 Sep
9555 *Ipimorpha pleonectusa* Grt. 15 Aug-20 Sep
9556 *Chytonia palliaticricula* (Gn.) 20 Jun-15 Jul, 15-25 Aug
9560 *Dypterygia rozmami* Berio 10-20 Jul
9564 *Andropolia contacta* (Wlk.) 15-30 Aug
9578 *Hypa xylinoides* (Gn.) 20 Jun-20 Jul, 5 Sep
9583 *Callipistria cordata* (Ljungh) 25 Jun-20 Jul
9587 *Magusa orbifera* (Wlk.) 10 Sep
9587 *Proxenus miranda* (Grt.) 20-30 Jun
9589 *Proxenus mendosa* McD. 20 Jul
9573 *Platyperiogia mutifera* (Wlk.) 10 Aug-10 Sep
9566 *Spodoptera frugiperda* (J. E. Smith) 10-20 Sep
9568 *Elaphria festivoides* (Gn.) 5 Jun-10 Jul
9573 *Xylena supera* (Lint.) 5 May
9574 *Xylena curvimacula* (Morr.) 20 May-5 Jun, 20 Sep
9576 *Xylena cinerita* (Grt.) 10-25 May, 30 Aug-20 Sep
9584 *Litholomia napea* (Morr.) 20 Aug-25 Sep
9589 *Lithophane petulca* Grt. 20-25 May, 10-25 Sep
9591 *Lithophane amanda* (Sm.) 5 Jun
9592 *Lithophane baileyi* Grt. 15-25 Sep
9593 *Lithophane georgii* Grt. 10 May
9596 *Lithophane unimoda* (Lint.) 15 May-5 Jun, 25 Sep
9597 *Anathix ralla* (G. & R.) 5 Sep
9596 *Anathix puta* (G. & R.) 15 Aug, 5-20 Sep
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9967 Hilla iris (Zett.) 5-15 Sep
9974 Fuchsia entreaxa Grt. 10-25 Sep
9976 Platypodia anceps (Steph.) 5-20 Sep
9980 Xylo type acadia B. & Benj. 30 Aug-25 Sep
9990 Sulyna profunda (Sm.) 25 Aug-20 Sep
9998 Brachylomia algens (Grt.) 10 Sep
10005 Feralia jocosa (Gn.) 15 May-10 Jun
10007 Feralia major Sm. 15-20 May, 5 Jun
10008 Feralia comstocki (Grt.) 25 May-10 Jun
10111 Brachionycha borealis (Sm.) 5 May-30 Jun
10059 Homohadena badisriga (Grt.) 30 Jul-30 Aug
10065 Homohadena infixa (Wlk.) 10 Jul-20 Aug
10123 Oncocnemis piffardi (Wlk.) 10 Sep
10 198 Cucutlia postera (Gn.) 15 Jul
10223 Discesa trifolii (Hufn.) 10 Sep
10265 Sideridis rosea (Harv.) 10-20 Jun
10275 Polia nimbosa (Gn.) 20 Jul-10 Aug
10276 Polia imbrifera (Gn.) 20 Jul-5 Aug
10280 Polia purpurissata (Grt.) 15 Jul-20 Aug
10288 Polia detracta (Wlk.) 10 Jul
10288a. Polia detracta neoterica (Sm.) 15-25 Jul
10290 Polia obscura (Sm.) 15-25 Jun
10292 Melanchra adjuncta (Gn.) 10-25 Jun, 10-25 Jul
10294 Melanchra pulverulenta (Sm.) 15-30 Jun
10295 Melanchra assimilis (Morr.) 30 Jun-15 Jul
10296 Lacinipolia lustralis (Grt.) 10 Jun-5 Jul, 15 Aug
10297 Lacinipolia radix (Wlk.) 5-15 Jun
10300 Lacinipolia grandis (Gn.) 5 Aug
10301 Lacinipolia latra (Gn.) 10-25 Jun
10303 Lacinipolia tacoma (Stkr.) 10-25 Jun
10310 Papestra quadrala (Gn.) 30 May-5 Jun
10312 Papestra cristifera (Wlk.) 5-30 Jun
10370 Lacinipolia lastralis (Grt.) 20 Jun-10 Jul
10372 Lacinipolia anguina (Grt.) 5-15 Jun
10397 Lacinipolia renigera (Steph.) 25 Jul-30 Aug
10405 Lacinipolia lorea (Gn.) 30 Jun-10 Jul
10436 A/etia oxygala (Grt.) 20 Jul-10-25 Aug
10438 Pseudaletia unipuncta (Haw.) 10 Jun
10446 Leucania mutilinea (Wile.) 10-20 Aug
10447 Leucania commoides (Gn.) 5-30 Jun
10449 Leucania insueta (Gn.) 15 Jun
10471 Stretchia plusi aiformis Hy. Edw. 15-20 May
10471a. Stretchia p. coloradica Strand. 5-20 May
10490 Orthosia revicta (Morr.) 10-25 May
10513 Egera dolosa (Grt.) 25 May-10 Jun
10563 Protorhodes oviduca (Gn.) 10-25 Jun
10578 Pseudorhodes ve Scors (Gn.) 30 Jun-5 Jul
10587 Orthodes cynica Gn. 10 Jun, 15 Jul
10644 Agrotis mollis Wlk. 25 Jul
10651 Agrotis venerabilis Wlk. 20 Aug-15 Sep
10659 Agrotis volubilis Harv. 25 Jun
10660 Agrotis obliqua (Sm.) 15 Jul
10663 Agrotis ipilon (Hufn.) 5-25 Sep
10670 Feltia jaculifera (Gn.) 30 Aug-15 Sep
10676 Feltia helis (Grt.) 25 Jul-10 Sep
10702 Euxoa diversgens (Wlk.) 30 Jun-30 Jul
10715 Euxoa scandens (Riley) 25-30 Jul, 10-15 Sep
10738 Euxoa mimallonis (Grt.) 5 Aug-5 Sep
10755 Euxoa declara (Wlk.) 5-25 Aug
10801 Euxoa ochrogaster (Gn.) 5-15 Aug
10891 Octochropla plecta (L.) 5-15 Jul, 25 Aug
10915 Peridroma saucia (Hbn.) 15 May-20 Jun, 5-25 Sep
10917 Diasia rubifera (Grt.) 30 Jul-20 Aug
10918 Diasia distocata (Sm.) 30 Jul-15 Aug
10919 Diasia jucunda (Wlk.) 15 Jul-10 Sep
10928 Graphiphora haruspic a (Grt.) 30 Jul-25 Aug
10929 Eurois occulta (L.) 5 Jul, 10-25 Aug
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10942 Xestia adela Franc. 5 Aug-10 Sep
10943 Xestia normaniana (Grt.) 20 Jul-20 Aug
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10947 Xestia oblata (Morr.) 5 Jul-10 Aug
10951 Xestia tenuicula (Morr.) 5 Aug
10954 Xestia collaris (G. & R.) 25 Aug
10957 Anomogyna atrata (Morr.) 25-30 Jun
10962 Anomogyna perquiritata (Morr.) 10-30 Aug
10967 Anomogyna elinita (Gn.) 10 Aug-10 Sep
10969 Anomogyna dilucida (Morr.) 5 Aug
10988a. Eugraphe subrosea opacifrons (Grt.) 20 Aug
10992 Paradiarsia littoralis (Pack.) 5-15 Jul
10996 Metaledes saltarum (Wlk.) 15-25 May
10997 Metaledes fishii (Gt.) 30 Jun
10999 Aplectoides condita (Gn.) 20-25 Jun, 20 Jul
11001 Aplectoides pressus (Grt.) 5 Jul-10 Sep
11003 Chersotis juncta (Grt.) 25 Jul-15 Aug
11004 Protoplampa rufpectus (Morr.) 5-25 Aug
11008 Euereiacrotis persatena (Grt.) 5 Jul
DISCUSSION

Black Sturgeon Lake is in a relatively remote area, where only limited collecting has been carried out in the past. In his review of the state of knowledge on the Lepidoptera in Canada, Munroe (1979) lists a number of locations from which faunal samples are needed. Among these he names "the Lakehead district of Ontario, and the whole region from there to the Lake of the Woods, along the U.S. border." Black Sturgeon Lake is situated at the eastern end of this area, and this listing is therefore a contribution to meeting this lack of knowledge.

In all, 564 species of Lepidoptera plus 8 subspecies were collected at Black Sturgeon Lake during the years 1960–1968, representing 36 families, 5 butterflies, 1 skipper and 30 moths. This compares with 4692 species from 70 families listed by Munroe (1979) for the whole of Canada.

It is probable that the listing for Black Sturgeon Lake is incomplete. The collections were made by staff trained in forest entomology, but with no taxonomic expertise, who may have overlooked rarer species, especially if they were inconspicuous or resembled other, more numerous, species. However, it is unlikely that many species were overlooked. The stated objective was to make a representative collection, and so the collectors were continually on the look out for new species. The listing may then be considered fairly reliable, but absence from the listing cannot be taken as conclusive evidence that the species is absent from the area.

ACKNOWLEDGMENTS

I thank members of the Biosystematics Research Center (Agriculture Canada) for the identification of numerous specimens, and all those at Forestry Canada, Ontario Region, who worked on the assembly, typing and proofreading of the information in this report. In particular, Doug Lawrence, George Lucuik, Suzanne Palmer, Elizabeth Chudoba and Corrine Motluk should be thanked; without their care, diligence, and patience, the work would never have been completed. As well, my thanks to Dr. Paul Syme (Forestry Canada, Ontario Region) and Dr. Kevin Barber (Forestry Canada, Forest Pest Management Institute) for their careful and thorough review and for their advice on the taxonomy and nomenclature of many of the species and subspecies.

LITERATURE CITED


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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.

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