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A silvery checkerspot, Charidryas nycteis (Doubleday) (Lepidoptera: Nymphalidae).
Photograph by R. W. Sites, Dept. of Zoology, Southern Illinois University, Carbondale.
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A KEY TO THE BUTTERFLIES OF ILLINOIS
(LEPIDOPTERA: PAPILIONOIDEA)

R. W. Sites and J. E. McPherson

ABSTRACT

A key to the butterflies of Illinois is presented. Included is the general state distribution for each species or subspecies.

Irwin and Downey (1973) presented an excellent checklist of the Illinois butterflies, including distribution maps, but did not include keys. Klots (1951) prepared keys for several groups of the butterflies occurring in North America east of the Great Plains but omitted many others (e.g., Papilionidae, Nymphalidae, Pieridae, Satyridae). Ehrlich and Ehrlich (1961) included keys in their publication on the butterflies of America north of Mexico but the keys, in some cases, are inaccurate. Thus, there are presently no complete and reliable keys which can be used to identify Illinois butterflies. Presented here is a key to the Illinois butterflies, exclusive of the skippers (Hesperiidae), that can be used in conjunction with Irwin and Downey's list. Included are species that are, or were, established residents in this state, and species that stray into our area or pass through on migration routes. Presently known distribution patterns are coded with superscripts after the species names as follows: a, generally distributed throughout the state; b, northernmost counties only; c, primarily northern half of the state; d, primarily southern half of the state; e, southernmost counties only; f, strays or migrates from other areas; g, once occurred in Illinois but may no longer.

The names of several of the Illinois butterflies have changed since publication of Irwin and Downey's list. These changes, suggested by reviewers of this manuscript (see acknowledgments), are listed in Table 1. Also, Anaea aidea (Guerin-Meneville) (Nymphalidae) and Celastrina ebena Clench (Lycaenidae) have been added to the state list. A. aidea has been reported from Massac County (Jeffords 1977), and C. ebena from Jersey County (Wagner and Mellichamp 1978). McDonough County (Hess 1980) and Union County (Sites and McPherson, in press). Finally, Polygonia progne (Cramer), listed by Irwin and Downey from the northern half of the state, has now been recorded from Union County (Sites and McPherson 1979).

Fully labelled, generalized wings (Figs. 1A,B; 2A,B) are provided to aid individuals using the key who are not familiar with lepidopteran wing venation and areas. The anal area is that area of the wing that includes the anal veins(s). Cell-end bars, a characteristic used in identifying some lycaenids, are ventral bar-line markings at the distal end of the discal cell in the fore (Fig. 30A) and hind (Fig. 30B) wings. Fore wing length is measured from the base to the apex.

ACKNOWLEDGMENTS

We would like to thank J. C. Downey, University of Northern Iowa, Cedar Falls; D. F. Hess, Western Illinois University, Macomb; L. D. Miller, Allyn Museum of Entomology, 1Department of Zoology, Southern Illinois University, Carbondale, IL 62901.
<table>
<thead>
<tr>
<th>Current Name</th>
<th>Familya</th>
<th>Irwin &amp; Downey 1973</th>
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<tbody>
<tr>
<td><em>Artogeia napi oleracea</em> (Harris)</td>
<td>PIERIDAE</td>
<td><em>Pieris napi oleracea</em> (Harris)</td>
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<tr>
<td><em>Artogeia rapae</em> (Linnaeus)</td>
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<td><em>Pieris rapae</em> (Linnaeus)</td>
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<td><em>Pontia protodice</em> (Boisduval &amp; Le Conte)</td>
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<td><em>Pieris protodice</em> Boisduval &amp; Le Conte</td>
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<td><em>Zerene cesonia</em> (Stoll)</td>
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<td><em>Colias cesonia</em> (Stoll)</td>
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<td><em>Epidemia helioides</em> (Boisduval)</td>
<td>LYCAENIDAE</td>
<td><em>Lycaena helioides</em> (Boisduval)</td>
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<tr>
<td><em>Gaeides xanthoides dione</em> (Scudder)</td>
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<td><em>Lycaena xanthoides dione</em> (Scudder)</td>
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<tr>
<td><em>Hemiargus isola alce</em> (Edwards)</td>
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<td><em>Hemiargus isola</em> (Reakirt)</td>
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<tr>
<td><em>Hylolycaena hyllus</em> (Cramer)</td>
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<td><em>Lycaena thoae</em> (Guerin-Meneville)</td>
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<tr>
<td><em>Incisalia henrici</em> (Grote &amp; Robinson)</td>
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<td><em>Calliphrys henrici</em> (Grote &amp; Robinson)</td>
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<td><em>Incisalia irus</em> (Godart)</td>
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<td><em>Calliphrys irus</em> (Godart)</td>
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<td><em>Incisalia polios</em> Cook &amp; Watson</td>
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<td><em>Calliphrys polios</em> (Cook &amp; Watson)</td>
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<td><em>Calliphrys gryneus</em> (Hübner)</td>
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<td><em>Parrhasius m-album</em> (Boisduval &amp; Le Conte)</td>
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<td><em>Panthiades m-album</em> (Boisduval &amp; Le Conte)</td>
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<tr>
<td><em>Aglais milberti</em> (Godart)</td>
<td>NYMPHALIDAE</td>
<td><em>Nymphalis milberti</em> (Godart)</td>
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<tr>
<td><em>Anthanassa texana</em> (Edwards)</td>
<td></td>
<td><em>Phyciodes texana</em> (Edwards)</td>
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<td><em>Basilarchia archippus</em> (Cramer)</td>
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<td><em>Limenitis archippus</em> (Cramer)</td>
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<tr>
<td>Basilarchia arthemis arthemis (Drury)</td>
<td>Limenitis arthemis arthemis (Drury)</td>
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<tr>
<td>Basilarchia arthemis astyanax (Fabricius)</td>
<td>Limenitis arthemis astyanax (Fabricius)</td>
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<tr>
<td>Charidryas gorgone carlota (Reakirt)</td>
<td>Chlosyne gorgone carlota (Reakirt)</td>
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<tr>
<td>Charidryas barrassii (Scudder)</td>
<td>Chlosyne harrisi (Scudder)</td>
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<tr>
<td>Charidryas nycteis (Doubeday)</td>
<td>Chlosyne nycteis (Doubeday)</td>
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<tr>
<td>Clossiana bellona (Fabricius)</td>
<td>Boloria bellona (Fabricius)</td>
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<tr>
<td>Clossiana selene nebraskensis (Holland)</td>
<td>Boloria selene myrina (Cramer)</td>
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<tr>
<td>Vanessa cardui (Linnaeus)</td>
<td>Cynthia cardui (Linnaeus)</td>
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<tr>
<td>Vanessa virginia (Drury)</td>
<td>Cynthia virginia (Drury)</td>
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**SATYRIDAE**

| Cylopsis gemma (Hübner) | Euphytica gemma (Hübner) |
| Hermeleptia hermes sosybius (Fabricius) | Euphytica hermes sosybius (Fabricius) |
| Lethe antedon (Clark) | Lethe portlandia antedon (Clark) |
| Megisto cymela (Cramer) | Euphytica cymela (Cramer) |

*Sequence of families follows that of Irwin & Downey (1973).*
Sarasota, Florida; M. C. Nielsen, 3415 Overlea Dr., Lansing, Michigan; Y. S. Sedman, Western Illinois University, Macomb; and J. G. Sternburg and M. E. Toliver, University of Illinois, Urbana, for reviewing the manuscript. We are also grateful to J. C. Downey, L. D. Miller, M. C. Nielsen and M. E. Toliver for suggesting the name changes listed in Table 1. Finally, we wish to thank G. L. Godfrey and D. W. Webb, Illinois Natural History Survey, Urbana, for the loan of specimens; E. G. MacLeod, University of Illinois, for his help and encouragement during the preparation of the manuscript; and S. W. Wilson, Southern Illinois University, Carbondale, for his assistance with the illustrations.

Figs. 1-2. 1. Generalized fore (A) and hind (B) wings with veins and cells labelled. 2. Generalized fore (A) and hind (B) wings with wing areas and margins labelled.
KEY TO ILLINOIS BUTTERFLIES

1. Labial palpi at least as long as thorax, giving the appearance of a long snout (Fig. 3); HW with outer margin serrate (Fig. 4) (LIBYTIDAE) ......................................................... 2
1'. Labial palpi much shorter than thorax; HW with outer margin variable ......... 2
2. FW with R vein 5-branched. M1 not stalked with R (Figs. 5, 6A, 7A, 8A, 9, 10A, 11, 12A, 14A, 15, 16) ................................................................. 3
2'. FW usually with R vein 3-branched (Fig. 29A) or 4-branched (Figs. 22, 23A, 24A, 25, 26, 27, 28) but if 5-branched, then M1 is stalked with R ........................................... 7
3. FW with Sc and Cu veins distinctly swollen basally (Figs. 5, 6A, 7A, 8A, 9); wings with ground color brown (SATYRIDAE) ........................................... 16
3'. FW with Sc and Cu veins not swollen basally, but of uniform thickness or gradually widening basally; wings with ground color variable ........................................ 4
4. HW with one anal vein and with distal tail-like projection that includes vein M3 (Fig. 10B); FW length usually greater than 30 mm (PAPILIONIDAE) .............. 11
4'. HW with two anal veins, with or without tail (Figs. 12B, 13, 14B); FW length variable . 5
5. 3A vein in FW present but short (Fig. 11); large reddish brown butterflies (DANAIDAE) ................................................................. 10
5'. 3A vein in FW lacking (Figs. 12A, 14A, 15, 16) ........................................ 6
6. HW with humeral vein curving toward body (Fig. 12B); wings dorsally with ground color orange-brown; HW ventrally with silver spots, thus appearing similar in color to Speyeria (Nymphalidae); FW narrower than Speyeria (Fig. 12A) (HELICONIIDAE) ........................................ Agraulis vanillae nigrior Michener (Gulf fritillary) 1
6'. HW with humeral vein curving away from body (Figs. 13, 14B), or straight; wings with ground color variable; FW generally wider than in Agraulis (Fig. 14A) (NYMPHALIDAE) .................................................. 63
7. Tarsal claws bifid; wings dorsally with ground color white, yellow, or orange (PIERIDAE) ................................................................. 23
7'. Tarsal claws simple; wings dorsally with ground color variable .................. 8
8. M1 in FW stalked with R beyond discal cell (Fig. 27); HW dorsally with orange ground color confined to outer 1/2 to 3/4 by a dark brown basal area that also extends along costal margin; HW ventrally with several light brownish spots ringed with white (LIPHYRIDAE) .............................................. Feniseca tarquinius (Fabricius) (Harvester) 9
8'. M1 in FW not stalked with R beyond discal cell (Figs. 28, 29A); HW ventrally with ground color variable ...................................................... 9
9. HW with humeral vein (Fig. 32), tails lacking (RODINIDAE) ......................... 2
9'. HW without humeral vein (Fig. 29B), with or without tails (LYCAENIDAE) ...... 37
10. FW dorsally with black along inner margin; wings dorsally with ground color orange-brown .......................................................... Danaus plexippus (Linnaeus) (Monarch) 10
10'. FW dorsally without black along inner margin; wings dorsally with dark reddish brown ground color extending to inner edge ... Danaus gilippus strigosus (Bates) (Queen) 11
11. HW with tails at least as long as abdomen and without distal swelling; wings dorsally with ground color whitish green and with black longitudinal bands; HW ventrally with red longitudinal stripe ....... Eurides marcellus (Cramer) (Zebra swallowtail) 11
11'. HW with tails shorter than abdomen, with or without distal swelling; wings dorsally with ground color yellow, brown, or black; HW ventrally without red longitudinal stripe .................................................. 12
12. Wings dorsally with ground color brown or black, broad yellow band of spots extending from base of HW to apex of FW, extensively yellow ventrally ................... Papilio cresphontes Cramer (Gi ant swallowtail) 12
12'. Wings dorsally with ground color variable but if brown or black, then without yellow band of spots extending from base of HW to apex of FW, may be extensively yellow ventrally .................................................. 13
13. HW dorsally with extensive metallic blue or green (some females may lack metallic
coloration), one row of narrow white submarginal spots which may be reduced, orange spots present ventrally but lacking dorsally .................................................. Battus philenor (Linnaeus) (Pipevine swallowtail)

13'. HW other than above ............................................................ 14

14. Wings ventrally with black submedian stripe, dorsally with ground color yellow, brown, or black ........................................ Papilio glaucus Linnaeus 4 (Tiger swallowtail)

14'. Wings ventrally without black submedian stripe, dorsally with ground color brown or black ............................................................ 15

15. HW dorsally with yellow marginal spots and yellow postmedian spot in cell Sc+R1; wings dorsally with ground color black ........................................ Papilio polyxenes asterius Stoll 3 (Black swallowtail)

---

Figs. 3-8. 3. Head and labial palpi of Libytheana bachmanii. 4. Hind wing of Libytheana bachmanii. 5. Fore wing of male Lethe creola (shape of fore wing of female more closely resembles that of Lethe anthedon, Fig. 6A). 6. Fore (A) and hind (B) wings of Lethe anthedon. 7. Fore (A) and hind (B) wings of Lethe eurydice. 8. Fore (A) and hind (B) wings of Lethe appalachia.
15'. HW dorsally with green or blue marginal spots, and orange, or yellow (many late summer males), postmedian spot in cell Sc+R1; wings dorsally with ground color brown or black ......................... *Papilio troilus* Linnaeus (Spicebush swallowtail)

16. FW dorsally with two eyespots: HW ventrally with numerous short brown lines in basal half .......................... *Ceryyonis pegala olympus* (Edwards) (Wood nymph)

16'. FW dorsally with eyespots variable but if two, then HW ventrally with single brown line in basal half .......................... 17

Figs. 9-12. 9. Fore wing of *Cellapectis gemma*. 10. Fore (A) and hind (B) wings of *Papilio polyxenes asterius*. 11. Fore wing of *Danaus plexippus*. 12. Fore (A) and hind (B) wings of *Agraulis vanillae nigrior*. 
17. FW with base of Sc and Cu strongly swollen (Fig. 9) (Euptychiini) ............... 18
17'. FW with base of Sc and Cu slightly swollen (Figs. 5, 6A, 7A, 8A) (Leihe) ........ 20
18. FW with eyespots lacking dorsally and ventrally; HW ventrally with four black marginal spots between veins Cu1 and M2 ..........................................................

................................................................................................. Cyllopsis gemma (Hübner) (Gemmed satyr)
18'. FW with eyespots present dorsally, ventrally, or both: HW ventrally without four black marginal spots between veins Cu1 and M2 ................................................. 19
19. FW dorsally with two well-developed eyespots ........................................... 

\[ \text{Megisto cymela (Cramer\textsuperscript{b}) (Little wood satyr)} \]

19'. FW dorsally with eyespots absent or greatly reduced .............................................................................. *Helmeuptychia hermes sosybini* (Fabricius* (Carolina satyr)

20. HW with outer margin strongly scalloped (Fig. 6B) ................................................................. 21

20'. HW with outer margin slightly scalloped (Figs. 7B, 8B) ................................................................. 22

21. FW ventrally with postmedian band distinctly irregular between costal margin and vein M3, produced outward in cell M1 (Fig. 5); cell Cu2 usually with submedian eyespot; males with androconial scales on FW dorsally ................................................................. *Lethe creola* (Skinner* (Creole pearly eye)

21'. FW ventrally with postmedian band straight or slightly irregular between costal margin and vein M3, not produced outward in cell M1 (Fig. 6A); cell Cu2 usually lacking submedian eyespot; males without androconial scales .................................................................................... *Lethe anthedon* (Clark* (Pearly eye)

22. HW ventrally with postmedian line deeply zig-zag (Fig. 7B), outer half brown, yellowish brown, or yellowish white ................................................................. *Lethe eurydice* (Johansson* (Eyed brown

22'. HW ventrally with postmedian line slightly wavy (Fig. 8B), outer half violet-brown .................. *Lethe appalachia* R. L. Chemock*

23. HW with humeral vein present; wings dorsally with ground color white (Pierinae) ................. 24

23'. HW with humeral vein usually absent but if present, then never with ground color white; wings dorsally with ground color white, yellow or orange (Coliadinae) ................. 28

24. FW with outer margin concave, apex rounded (Fig. 22), dorsally with an orange patch in males ................................................................................................. *Anthocharis midea* (Hübner* (Falcate orange tip

24'. FW with outer margin convex or straight, apex rounded, orange patch lacking ................. 25

25. HW ventrally with ground color white and with broad irregular olive-colored stripes, dorsally with few or no black markings ......................................................... *Euchloe olympia* (Edwards* (Olympia)

25'. HW other than above ........................................................................................................................................... 26

26. Wings dorsally with ground color white, but without brown or black markings although basal half of FW costal margin may have gray shading .................................................................................... *Artogeia napi oleracea* (Harris* (Mustard white

26'. Wings dorsally with ground color white, and usually with black or brown markings; if markings are lacking, then HW ventrally with ground color yellowish ................. 27

27. FW ventrally with one or two brown or black spots, dorsally with an unbroken black apical patch; HW ventrally with ground color yellowish to white ................................................................................................. *Artogeia rapae* (Linnaeus* (European cabbage butterfly

27'. FW ventrally with more than two spots, dorsally with broken black apical patch; HW ventrally with ground color white but may have extensive zig-zag markings ................................................................. *Ponita protodice* (Boisduval & Le Conte* (Checkered white

28. FW with R vein 3-branched; HW dorsally with dark bar extending outward from base along costal margin; FW length usually 15 mm or less ................................................................. *Natalis isole* Boisduval* (Dainty sulphur

28'. FW with R vein 4-branched; wing coloration variable; FW length usually greater than 15 mm ........................................................................................................................................... 29

29. Arolium and pulvilli present ........................................................................................................................................... 30

29'. Arolium and pulvilli lacking ........................................................................................................................................... 34

30. HW with outer margin produced at vein Cu1; FW dorsally with brown band along outer margin wide, inner edge of band deeply notched in cells Cu1 and M3 ................................................................................................................................. *Eurema mexicana* (Boisduval* (Mexican sulphur

30'. HW with outer margin rounded; FW dorsally with brown band along outer margin wide, narrow or lacking but if wide, then inner margin not deeply notched in cells Cu1 and M3 ................................................................................................................................. 31

31. Wings dorsally with ground color orange; FW dorsally with black border on outer margin; FW length usually 16–25 mm ................................................................. *Eurema nicippe* (Cramer* (Sleepy orange

31'. Wings dorsally with ground color orange, yellow, brownish yellow, or white; FW dorsally with brown border, or series of small dark marks, or without dark markings, along outer margin of wing; FW length variable ................................................................. 32

32. FW dorsally with apical fourth black; wings dorsally with ground color yellow or white; FW length less than 23 mm ................................................................................................. *Eurema lisa* (Boisduval & Le Conte* (Little sulphur
32'. FW dorsally with apical fourth usually not black; wings dorsally with ground color orange, yellow, or brownish yellow; FW length greater than 24 mm. ............ 33

33. FW dorsally with orange bar through discal cell. HW dorsally with band on outer margin. wings dorsally with ground color yellow (males); or FW dorsally with well-developed brown border (Fig. 23A). HW dorsally with marginal spots (Fig. 23B), wings dorsally with ground color dull orange to brownish yellow (females) ........................................ Phoebis philea (Johansson) (Orange barred sulphur)

Figs. 23-26. 23. Fore (A) and hind (B) wings of female Phoebis philea. 24. Fore (A) and hind (B) wings of female Phoebis sennae eubule. 25. Fore wing of Zerene cesonia. 26. Fore wing of male Colias philodice.
Figs. 27-32. 27. Fore wing of *Feniseca tarquinius*. 28. Fore wing of *Lycaena phlaeas americana*. 29. Fore (A) and hind (B) wings of *Satyrium calanus falcifer*. 30. Fore (A) and hind (B) wings of *Satyrium carneaorius* showing relative widths of fore wing cell-end bar and hind wing postmedian spot. 31. Tarsal claw of *Everes comyntus*. 32. Hind wing of *Calephelis maticum*. 
33'. Wings dorsally with orange bars and bands lacking (males); or, with thin brown border or reduced spots along outer edge of wings (females) (Fig. 24A,B); wings dorsally with ground color yellow (males and females) ........................................ Phoebis senea eubule (LinnaeusH) (Cloudless sulphur)
34. FW with apex acute, outer margin truncate, dorsally with dark band along outer margin highly irregular along inner edge (Fig. 25); wings dorsally with ground color yellow ........................................ Zerene cesonia (StollP) (Dog face)
34'. FW with apex subacute or rounded, outer margin not truncate, dorsally with dark band along outer margin straight or slightly irregular along inner edge (Fig. 26); wings dorsally with ground color white, yellow, or orange ......................... 35
35. Wings dorsally with ground color yellow and lacking orange areas other than the HW disca l spot .................. Colias philodice GodartB (Clouded sulphur)
35'. Wings dorsally with ground color yellow, white, or yellow with orange patches in addition to orange disca l spot ........................................ 36
36. FW dorsally with ground color orange and without yellow areas basal to the black border .................. Colias eurytheme BoisduvalA (Alfalfa butterfly)
36'. FW dorsally with ground color a yellow and orange mixture, or ground color white and without yellow areas basal to the black border .......... Colias eurytheme or philodiceA
37. FW R vein 4-branched (Fig. 28) ........................................ 38
37'. FW R vein 3-branched (Fig. 29A) (Theclinae) ........................................ 48
38. Tarsal claws with basal tooth (Fig. 31); wings dorsally with ground color usually blue (Plebejinae) ......................... 42
38'. Tarsal claws without basal tooth; wings dorsally with ground color usually brown or orange (Lycaeninae) ............................. 39
39. FW with ground color gray or brownish gray dorsally, white ventrally ........................................ Gaeides xanthoides dione (ScudderF) (Great copper)
39'. FW with ground color orange or brownish purple dorsally, yellow or yellowish orange ventrally ........................................ 40
40. FW ventrally with thin black marginal line; FW length usually greater than 16 mm. ................................ Hyllolcaenae hylitus (CramerA) (Bronze copper)
40'. FW ventrally without black marginal line; FW length usually less than 16 mm ................................. 41
41. FW ventrally with black discal spots ringed with white, gray border present on inner and outer margins ........ Lycana phlaeas americana HarrisA (American copper)
41'. FW ventrally with black discal spots not ringed with white, gray border lacking on inner and outer margins although brownish borders may be present ........................................ Epidemia helioides (BoisduvalF) (Purplish copper)
42. HW ventrally with red or orange spots; with or without tails ........................................ 43
42'. HW ventrally without red or orange spots; tails lacking ........................................ 44
43. HW with tails; ventrally with one or two well-defined red or orange submarginal spots ............... Erthes conmyta (GodartP) (Eastern tailed blue)
43'. HW without tails; ventrally with row of eight red or orange submarginal spots .............. Lycaenides melissa samuelis NabokovA (Karner blue)
44. Wings ventrally with marginal and or submarginal markings, postmedian spots variable ........................................ 45
44'. Wings ventrally without marginal or submarginal markings, postmedian spots black and ringed with white .......... Glacicepsche lydamus couperi GroteF (Silvery blue)
45. HW ventrally with 7-9 irregular brown longitudinal bands that are visible dorsally .................. Leptotes marina (ReakirtG) (Marine blueF)
45'. HW ventrally without 7-9 brown longitudinal bands ........................................ 46
46. FW ventrally with row of black postmedian spots ringed with white .......... Hemiarps isola atce (EdwardsP) (Reakirt's blue)
46'. FW ventrally with row of short brown postmedian-submarginal lines or elongate marks which may be faint ........................................ 47
47. FW dorsally with dark brown outer margin usually narrow, generally enclosing no more

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2Summer migrants of this species may establish breeding colonies but cannot overwinter in Illinois (Howe 1975). They have been recorded only from Cook, Mercer (Irwin and Downey 1973) and Hancock (Sedman 1980) counties.
than outer fourth of vein M3; if enclosing outer third, then specimen was collected during
either summer or fall ..............................................

* Celastrina argiolus pseudoargiolus* (Boisduval & Le Conte)* (Spring azure)

47'. FW dorsally with dark brown outer margin wide, enclosing outer third of vein M3
(females), or with wings entirely dark brown (males), specimens collected only during
spring ....................................................... * Celastrina ebenina* Clench* (Dusky blue)

48. Wings dorsally with ground color iridescent blue which may be restricted to basal
half ................................................................. 49

48'. Wings dorsally with ground color brown, gray, or black .................................. 50

49. Wings ventrally without postmedian line; FW ventrally with red or orange basal spot,
HW ventrally with two red or orange basal spots ..............................................

49'. Wings ventrally with white postmedian line and without red or orange basal spots,
although orange line is present along base of costal margin ..........................

* Parrhasius m-album* (Boisduval & Le Conte)* (White M hairstreak)

50. HW ventrally with clearly-defined red or orange band bordering inner edge of much
narrower white and black postmedian line (evident without magnification) ............

50'. HW ventrally with poorly-defined red or orange band bordering inner edge of wider
white and black postmedian line, or band absent ............................................. 51

51. Wings ventrally with ground color green or greenish brown; FW ventrally with well-
developed white submarginal line; HW ventrally with row of white postmedian marks
forming a broken irregular line, tails present ....................................................

51'. Wings ventrally with ground color variable but if green or greenish brown, then FW
ventrally without well-developed white submarginal line; HW ventrally usually without
row of white postmedian marks forming a broken irregular line, tails present or
absent ................................................................. 52

52. HW ventrally with row of eight red or orange submarginal spots, row of black post-
median spots ringed with white, blue anal spot lacking, ground color brownish gray, tails
absent ......................................................... * Harkenclenus titus* (Fabricius)* (Coral hairstreak)

52'. HW ventrally usually without row of eight red or orange submarginal spots but if present,
then blue anal spot present, ground color variable, tails present or absent ........ 53

53. HW ventrally without red or orange marginal or submarginal spots, although light bands
or scattered red scales may be present, tails present or absent; FW ventrally with ground
color brown, dark brown, or greenish brown .................................................. 54

53'. HW ventrally with red or orange marginal or submarginal spots, tails present; FW
ventrally with ground color variable ................................................................. 57

54. HW ventrally with black and red, or black and brown, stripes basal to postmedian line
giving wing a patterned appearance, tails absent ..................................................

54'. HW ventrally without obvious stripes basal to postmedian line, color usually solid to
base of wing, tails present or absent ................................................................. 55

55. FW ventrally with marginal band of white or light gray; HW without tails ..............

55'. FW ventrally without marginal band of light gray, brown ground color extending to wing
dge; HW with tails .................................................................

56. HW ventrally with black submarginal spot in cell Cu 1; males with long scent pad at end of
discal cell of FW dorsally ....................................................... * Incisialia irus* (Godart)* (Frosted elfin)

56'. HW ventrally without black submarginal spot in cell Cu 1; males without scent
pad ................................................................. * Incisialia henrici* (Grote & Robinson)* (Henry’s elfin)

57. Wings ventrally without cell-end bars ............................................................... 58

57'. Wings ventrally with cell-end bars ................................................................. 59

58. FW ventrally with postmedian line absent in cell Cu 2, ground color light gray ....

............................

* Strymon melinus humuli* (Harris)* (Gray hairstreak)
58. FW ventrally with postmedian line extending into cell Cu2, ground color brown .......... *Euristrum* *ontario* (Edwards) (Northern hairstreak)

59. Wings ventrally with postmedian line broken into well-defined spots ringed with white ............................................. 60

59'. Wings ventrally with postmedian line not broken into well-defined spots, but may be broken into short bars edged on inner and outer margins with white .......... 61

60. Wings ventrally with cell-end bars narrower than postmedian spots and closed at ends, ventral ground color gray .......... *Satyrium* *acacidia* (Edwards) (Acadian hairstreak)

60'. Wings ventrally with cell-end bars wider than postmedian spots and open at ends with brown ground color within, ventral ground color brown ...................... *Satyrium* *edwardsii* (Saunders) (Edward's hairstreak)

61. FW ventrally with four irregular well-defined white lines basal to submarginal line, one of which extends from outer edge of cell-end bar through cell Cu2 .......................... 64

61'. FW ventrally without four well-defined lines basal to submarginal line, and with white line along outer edge of cell-end bar restricted to cells M1 and M2 .......... 62

62. HW ventrally with postmedian spot in cell Sc+R1 ca. 2/3 to 3/4 as wide as FW cell-end bar, or poorly-developed: male genitalia without finger-like projection at base of tegumen .......... *Satyrium* *calanus* *falacer* (Godart) (Banded hairstreak)

62'. HW ventrally with postmedian spot in cell Sc+R1 about as wide as FW cell-end bar (Fig. 30A,B): male genitalia with finger-like projection at base of tegumen ............................. *Satyrium* *caryaevors* (McDunnough) (Hickory hairstreak)

63. Eyes not hairy .......................... 64

63'. Eyes hairy .................................. 65

64. Antennal club with three well-developed ventral ridges that extend down the antennal shaft (Fig. 20) ................................................................. 75

64'. Antennal club with one well-developed ventral ridge that extends down the antennal shaft (Fig. 21) or none .......... 88

65. FW with inner margin concave .......... 66

65'. FW with inner margin straight .......... 69

66. HW ventrally with silver discal marking broken into two parts, thus resembling a question-mark (occasionally the marking is not divided): FW dorsally with dark postmedian spot in cell M2 (Fig. 16): HW with tails usually at least twice as long as broad (Fig. 17) ...................... *Polygonea* *interrogationis* (Fabricius) (Question-mark)

66'. HW ventrally with silver discal marking not broken, thus resembling a comma: FW dorsally without dark postmedian spot in cell M2: HW with tails usually only 1 1/2 times as long as broad (Figs. 18, 19) .......... 67

67. Wings ventrally usually with two rows of green submarginal spots: HW with outer margin extremely irregular (Fig. 18) .......... *Polygonea* *faunus* (Edwards) (Green comma)

67'. Wings (one or both pairs) ventrally with an inner row of small black submarginal spots, and an outer row of black or greenish black submarginal spots that may be continuous to form a zig-zag line: HW with outer margin moderately irregular (Fig. 19) .... 68

68. HW ventrally, in basal half, with broken parallel longitudinal brown and beige lines extending from costal margin to generally beyond base of Cu vein, silver comma mark tapering at both ends .......... *Polygonea* *progne* (Cramer) (Gray comma)

68'. HW ventrally, in basal half, with longitudinal lines generally extending from costal margin to just beyond base of R vein, silver comma mark hooked at one or both ends ............................. *Polygonea* *commi* (Harris) (Comma)

69. Wings dorsally with ground color dark brown or black; FW dorsally with clearly-defined reddish orange bar extending obliquely through discal cell; HW dorsally with clearly defined wide reddish orange band along outer margin .......................... *Vanessa* *ataltana* *rubria* (Fruhstorfer) (Red admiral)

69'. Wings dorsally with ground color variable but if dark brown or black, then FW without clearly-defined reddish orange bar extending obliquely through discal cell; HW dorsally without clearly-defined wide reddish orange band along outer margin .......... 70
70. HW ventrally with a submarginal row of eyespots ........................................... 71
70'. HW ventrally without a submarginal row of eyespots ........................................ 72
71. HW ventrally with two eyespots, dorsally with eyespots in cells Cu1 and M1 larger than those in cells M3 and M2 ... *Vanessa virginiensis* (Drury) (American painted lady)
71'. HW ventrally with four or five eyespots, dorsally with eyespots in cells Cu1 and M1 subequal to those in cells M3 and M2 ... *Vanessa cardui* (Linnaeus) (Painted lady)
72. Wings dorsally with ground color brown or black, with or without a clearly-defined yellow and orange submarginal band .................................................. 73
72'. Wings dorsally with ground color orange or orange-brown, and without a clearly-defined yellow and orange submarginal band ............................................. 74
73. Wings dorsally with broad yellow or yellowish white marginal band and row of blue submarginal spots; FW dorsally without markings in discal cell .................. 75
73'. Wings dorsally with dark brown or black marginal band that may contain a row of white (FW) or blue (HW) spots, and with broad orange submarginal band bordered along inner edge with yellow; FW dorsally with two orange patches in discal cell .......................... 76
74. FW dorsally with two black spots basal to submarginal area in cell Cu1; HW ventrally with well-developed white V-shaped mark in outer area of discal cell .......... 77
74'. FW dorsally with one black spot basal to submarginal area in cell Cu1; HW ventrally with V-shaped mark in outer area of discal cell reduced or absent .................... 78
75. HW margin with tail at vein M3; FW with apex pointed; wings dorsally with ground color reddish brown, orange-brown, or orange ........................................ 79
75'. HW margin without tail, although slight angulation may occur; FW with apex rounded; wings with outer ground color variable .................................................. 80
76. HW with outer margin usually smooth or slightly sinuate; FW dorsally with faint submarginal band or band lacking (males), or with well-defined brownish yellow submarginal band (females) ................. *Anaea andria* Scudder (Goatweed butterfly)
76'. HW with outer margin serrate or sinuate; FW dorsally with well-defined submarginal band (males), or irregular row of yellow submarginal spots (females) ..................... *Anaea aiadea* (Guerin-Meneville) (Tropical leaf wing)
77. HW dorsally with two large eyespots, anterior eyespot about three times size of posterior one; FW dorsally with two orange bands crossing discal cell ...................... 81
77'. HW dorsally without eyespots; FW dorsally with or without orange bands crossing discal cell ............................................................................................... 82
78. HW dorsally with basal ½ uniformly dark brown or black, outer ½ orange or blue; HW ventrally without silver or orange patches in discal cell; FW length usually 42–55 mm .......... *Speyeria diana* (Cramer) (Diana fritillary)
78'. Other than above ...................................................................................... 83
79. HW ventrally with numerous well-developed silver patches basal to submarginal area ........................................................................................................ 84
79'. HW ventrally usually without numerous silver patches basal to submarginal area but if present, they are small and limited to postmedian area .................................. 85
80. HW dorsally with ground color brown, and with white postmedian spots and white or orange submarginal spots ......................... *Speyeria idalia* (Drury) (Regal fritillary)
80'. HW dorsally with ground color orange, reddish orange, or reddish brown but if brown, then without white postmedian spots and white or orange submarginal spots .... 86
81. FW dorsally with black marginal band. *Speyeria altantis* (Edwards) (Atlantis fritillary)
81'. FW dorsally with marginal band of orange subrectangular spots edged with black .................................................................................................................. 87
82. HW ventrally with yellow, yellowish brown, or brown marginal band usually much narrower than yellow submarginal band in cell M3 at its narrowest point; FW dorsally usually without black submedian mark in cell Cu2; wings dorsally with heavy (dark brown) basal suffusion ..... *Speyeria cybele* (Fabricius) (Great spangled fritillary)
82'. HW ventrally with yellow, yellowish brown, or brown marginal band usually wider than yellow submarginal band in cell M3 at its narrowest point; FW dorsally usually with black submedian mark in cell Cu2; wings dorsally with light to moderate (light brown to brown) basal suffusion ...................... Speyeria apirodite (Fabricius) (Aphrodite)

83. Wings dorsally with ground color orange and with lighter postmedian band; FW dorsally with three or four black submarginal spots .............................................. Euptoieta claudia (Cramer) (Variegated fritillary)

83'. Wings dorsally with ground color black, brown, brownish orange, or reddish brown; FW dorsally with one black submarginal spot or none ........................................... 84

84. Wings dorsally with ground color brown or orange-brown, veins not black ...... 85

84'. Wings dorsally with ground color usually blue or black but if orange, then veins black. 86

85. FW dorsally with black submarginal spot in cell Cu1; wings dorsally with ground color brown .......... Asterocampa celits (Boisduval & Le Conte) (Hackberry butterfly)

85'. FW dorsally without black submarginal spot in cell Cu1; wings dorsally with ground color orange-brown. Asterocampa celits (Boisduval & Le Conte) (Tawny emperor)

86. Wings dorsally with ground color orange-brown, veins black .............................................. Basilarchia archippus (Cramer) (Viceroy)

86'. Wings dorsally with ground color black or dark brown, veins concolorous ...... 87

87. Wings dorsally with white postmedian band ................................................................. Basilarchia arthemis arthemis (Drury) (White admiral)

87'. Wings dorsally without white postmedian band ........................................................ Basilarchia arthemis astyanax (Fabricius) (Red spotted purple)

88. FW with outer margin concave at middle; wings dorsally with ground color black or dark brown ..................... Anthanassa texana (Edwards) (Texan crescent)

88'. FW with outer margin straight or convex; wings dorsally with ground color variable . 89

89. Wings dorsally with ground color definitely black, usually with row of orange marginal spots ................................................................. 90

89'. Wings dorsally with ground color orange or orange and dark brown, row of orange marginal spots present or absent ................................................................. 91

90. FW lengths of male and female usually more than 27 and 31 mm respectively, dark ground color tending to obscure the two yellowish or yellowish orange spots in basal half; wings dorsally with yellowish orange marginal spots (spots may be reduced or absent) ................. Euphydryas phaeton ozarkae Masters (Ozark checkerspot)

90'. FW lengths of male and female usually less than 27 and 31 mm respectively, dark ground color generally not obscuring the two orange spots in basal half; wings dorsally with orange marginal spots .............................................................................. 91

91. HW ventrally with prominent silver patches; FW dorsally with row of orange marginal spots .......... Glossiana seleana nebraskensis (Holland) (Silver bordered fritillary)

91'. HW ventrally usually without silver patches but if present, then FW dorsally lacks a row of orange marginal spots ................................................................. 92

92. HW ventrally with basal half orange, outer half reddish brown; wings dorsally with thicker and more numerous black markings in basal half than outer half ................................. Glossiana bellona (Fabricius) (Eastern meadow fritillary)

92'. HW ventrally without basal half orange and outer half reddish brown, but either patterned or with solid ground color; wings dorsally with black markings variable, 93

93. HW ventrally with row of cream-colored submarginal spots and orange marginal band .............................................. Charidrvas harrisii (Scudder) (Harris' checkerspot)

93'. HW ventrally without row of cream-colored submarginal spots and orange marginal band .................................................................................................................. 94

94. HW ventrally with white postmedian band containing a dark zig-zag line, outer angles of zig-zag line on veins (Fig. 13) ................................................................. Charidrvas gorgone cariota (Reakirt) (Gorgone checkerspot)

3This subspecies may intergrade with C. s. myrina (Cramer) near the northern border or Illinois (Kohler 1977).
94'. HW ventrally usually without white postmedian band containing a dark zig-zag line but if present, then outer angles of zig-zag line between veins ......................... 95
95. FW ventrally usually without black median patch in cell Cu2 but if present, then dorsally and/or ventrally with one or more brown or black submarginal spots with white centers ...................... Charidryas nycteis (Doubleday) (Silvery checkerspot)
95'. FW ventrally with black median patch in cell Cu2; HW dorsally and ventrally with submarginal spots entirely brown or black .............................. 96
96. FW ventrally with triangular black patch in anterior part of postmedian area equal to, or larger than, black median patch in cell Cu2 (Fig. 15); HW ventrally with marginal patch dark brown or black .................... Phyciodes tharos (Drury) (Pearl crescent)
96'. FW ventrally with triangular black patch in anterior part of postmedian area smaller than black median patch in cell Cu2; HW ventrally with marginal patch either light brown or lacking .................. Phyciodes batesii (Reakirt) (Tawny crescent) 4

LITERATURE CITED


4One specimen has been recorded from La Salle County, Illinois (Irwin and Downey 1973).
Sphinx poecila, a valid North American Hawkmoth Species (Lepidoptera: Sphingidae)

J. Charles E. Riottel

The correct identification of *Sphinx poecila* has been a matter of concern to lepidopterists for many years. Although clearly described by Stephens (1828) and figured in color by Wood (1839), the species was later not correctly recognized and has been synonymized by different authors to the similar species, *Sphinx gordius* Cramer.

Apparently the first to synonymize *poecila* to *gordius* were Grote and Robinson (1865), although Grote later (1875, 1877) synonymized *poecila* (sic) Stephens with *Sphinx eremitus* (Hübner) to which it has not even a superficial similarity. Then followed with synonymization with *gordius* Butler (1876), Smith (1888), Rothschild & Jordan (1903), Wagner (1913), and Hodges (1971). Smith (1888) pointed out differences in the exterior pattern of *gordius* from different places, but the case rested at that. Clark (1920) described *poecila* as a northern "subspecies" of *gordius*, following the trend of his era, and Cadbury (1931) did the same from a single female creating the "form" *coxeyi*. Hodges (1971) acknowledged the possibility of two taxa treated under the name of *gordius* but he was mistaken in claiming that the "name borealis [i.e., the name of Clark’s northern subspecies] would apply to the northern populations." Apparently he did not see Wood’s (1839) illustration.

This brings us to the question of the availability of a type for *poecila*. Stephens described the species from a single male specimen of unknown origin which had found its way from the collection of a Mr. Wilkin into the collection of a Mr. Vigors where it was placed among indigenous British insects. Stephens pointed out that this was a mistake and that *poecila* as well as "another (Sp.plebeia) were placed in Mr. Wilkin’s cabinet as *Sp. Pinastri*" and that they were accompanied by "a memorandum that one of the two was foreign, the other British." Stephens then showed that both necessarily were of non-European origin and had both been confused with "*Sp.Pinastri."

The original description of *S.poecila*, given by Stephens (1828), is accurate and comprehensible: "Alis subacutis. canis. fusco-nebulosis. anticus puncto medio albo, lineolisque aliquot nigris. posticis fuscis fasciā latā pallidā. abdominis lateribus nigro maculatis. (Exp.alar. 2 uno 9 lin.)." Stephens also gave an original description in English: "Smaller than the foregoing [i.e., *plebeia*] ... anterior wings acute. hoary, clouded with brown, with several longitudinal and oblique black lines, and a zigzag one of the same colour at the apex, a conspicuous white spot on the disc, near the costa, and towards the hinder margin an undulated hoary streak. margined externally with brown; the cilia white, spotted with brown; posterior wings brown, with a broad pale central band; the cilia pure immaculate white; head and sides of thorax hoary; disc of the latter brown changing to hoary posteriorly; abdomen hoary ash, with an obsolete brownish line down the back, and a row of undefined black spots down each side."

Wood (1839) published his catalogue of the Lepidoptera of Great Britain including "doubtful British species." Among those listed is *poecila*. Fortunately, the colored figure of *"S.poecila"* is absolutely unmistakable, for the specimen which served Stephens for the description and Wood for the illustration seems subsequently to have been lost. It is known that the Wilkin’s collection via the Vigors’ collection went "in first choice" directly to the British Museum and that the rest went to the collection of the Zoological Society of London (Horn 1936). No trace, however, could be found of "*S.poecila*"; it is not in the collection of

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1Department of Entomology, B. P. Bishop Museum, Honolulu, HI 96819, and Royal Ontario Museum, Toronto, Ontario, Canada
the British Museum where A. H. Hayes spent much time in trying to locate it for me, and it is also not in the collection at Oxford (in litt.; E. Taylor, Hope Dept. of Entomology, University Museum, Oxford).

Walker (1856) published the sphingid part of his catalogue of the Lepidoptera in the British Museum collection, in which he listed "Anceryx poecila," and referred to "Sphinx poecila Stephens, Ill. Brit. Ent. Haust. I. 122, 8." Walker's description fits, sometimes to the word, the one given by Stephens, and he perhaps had before him the type specimen or he may have looked only at Stephens' description and/or Wood's colored figure. In any case, Walker is witness to the acceptance of poecila as a valid species at that time.

Butler (1876) published his revision of the family Sphingidae, and it is there that the mix-up (apart from Grote & Robinson and Grote) began. Butler listed "Sphinx poecila Stephens" as a synonym under "Sphinx gordius." His reference there for S. poecila is given as "Stephens, Ill. Brit. Ent. Haust. I. p. 222 (1828)," instead of p. 122 as it should have been. He referred previously, however, "Hyloicus poecila. Sphinx poecila, Stephens. Ill. Brit. Ent. Haust. I. p. 122 no. 8 (1828). / Anceryx poecila, Walker, Lep. Het. VIII. p. 229. no. 13 (1856). /—"? [i.e., patria questionable] (Vigors's Coll.). / Like a strong marked female of H. plebeia, which I believe it to be. / Type, B. M." Stephens (1828) also dealt with plebeia, but never mentioned a type. So apparently Butler took one of the plebeia specimens Stephens mentioned and mistook it for the type of poecila which he himself synonymized to gordius only one page later. Somehow Butler seems to have lost track of his representation, or in the time between Walker's publication and Butler's revision the type of the real poecila was lost and Vigors' specimen of plebeia became, in some way unknown to us, connected with the wrong name. That something happened to the specimen which Butler used seems clear, for Hayes found in the British Museum collection the specimen (Fig. 1) of Peratrea plebeia which he thinks (and I concur) to be the specimen Butler used, for his Hyloicus poecila (which then has nothing at all to do with Stephens' Sphinx poecila). The labelling of this specimen shows certain irregularities. One label bears the number (printed) "5957 Vigors' Coll."; this refers, according to Hayes (in litt.), "to our old style registration numbers: i.e., collection 57 to 1859. 170 Lepidoptera are listed with other entries presented by the Zoological Society of London from the Vigorsian Cabinet. However, W. H. T. Tams..."
who has well over fifty years' experience here has never seen a Vigors coll. label until I drew his attention to the specimen under discussion. However, I have traced a Walker type from Vigors coll. (Bearing the same label) in the collection of Hypsidae. The other labels are (printed) "Type" and one (handwritten) "sphinx lineolata," a really tremendous mix-up for "sphinx lineolata" does not make any sense at all. How could this specimen have been substituted for the (lost) type of Stephens' poecila? That Butler was mystified by it all is easily understandable. No author to my knowledge, not even Hodges (1971), has listed Hyloicus poecila as a synonym of Peratreaplebeia (Fabricius), an omission that is corrected herewith.

From the above the most logical conclusion is that the type of Stephens' Sphinxpoecila is lost and that we have only Wood's colored figure. This figure, however, is of such clarity that there is no way to confound it with Sphinx gordius Cramer of which the type is lost also, with only Cramer's figure available. I consider Sphinx poecila Stephens to be the name with priority for the insect later described by Clark, and known to most American lepidopterists, as Sphinx gordius borealis. Clark did not investigate the morphology of this taxon sufficiently well to find out that what he described as a "subspecies" was in reality a separate species which had already been described.

Since 1956 I have studied hundreds of northern specimens of Sphinx usually ascribed to gordius. The study has included extensive rearings and some (negative) hybridization experiments. The result of this investigation has been to confirm the specific identity of Sphinx poecila Stephens and its distinction from gordius. Clearly distinct material from Newfoundland (Hodges 1971) was formerly rare in collections, and this may have been the reason why the species was not recognized as such much earlier. Considerable material, however, has been collected in recent years by the personnel of the Canada Department of Agriculture in St. John's, Newfoundland, and at other localities on the island, and has been made available for this study. Also, on the continent, much more material from many different localities has accumulated, including Labrador and the coastal Sable Island.

**DESCRIPTION**

**Eggs.** In both poecila and gordius, pastel green, oval, slightly flattened, about 1.5 mm long.

**Larva.** (Fig. 2). Bluish-green (Newfoundland) to bright green with a slight yellowish tinge or viridian green (continental), about 65-70 mm long; some larvae may have extended purplish shading laterally and dorsally or be entirely pink; white dots ringed with black all over the body, leaving a narrow unspotted line on dorsum: lateral stripes on body seven, white with crimson followed by black anterior bordering (in Newfoundland stripes of larva often lemon-yellow-white/purple-blackish or dark brownish-purple); spiracles rusty to vermilion; horn black, underside greenish, pointing upwards or straight: (in gordius [Fig. 3]: When mature bluish-green, covered dorsally and laterally with black-ringed, white dots; smooth; lateral stripes from dissolving body-green to white to darker and darker purplish, overlaid in some specimens by a few brown spots [visible under microscope]; horn yellow, black laterally, apex all black: horizontal:) headcapsule (Fig. 5) dark green with lighter green and blackish lateral stripes (Newfoundland: laterally body-green, center with brown overcasting, exterior lateral stripe dark blackish-brown, interior one light pastel green with sometimes some yellowish), laterally straight, not bulging as in gordius (Fig. 6). The latter's headcapsule is body-green, center with brown overcasting; exterior lateral stripe olive-brown, interior stripe yellowish-pastel-green. The headcapsule of S.luscitiosa Clemens (Fig. 7), a very near relative, stands between poecila and gordius, nearer to the former. The thoracic claw segments (Figs. 8-10) are of different shape and chaetotaxis. The claw of gordius is smooth and without teeth; the one of poecila has some irregular teeth; and the one of luscitiosa has three teeth. The mandibles (Figs. 11-13) are symmetrical; in gordius the condyle is very shortly attached and the retinaculum shows two teeth quite separated from one another; in luscitiosa the condyle has a considerably long stalk and the teeth of the

The retinaculum are very nearly arranged to one another; in *poecila* the condyle is still more immediately attached than in *gordius* and also more rounded, the teeth of the retinaculum are not rounded as in the two other species but strongly angled; also the teeth of the mandibles are more strongly developed.

**Foodplants.** In the laboratory the larvae from Newfoundland were initially given blueberry (*Vaccinium* sp.) and *Myrica gale*; they preferred blueberry. Approximately a week later *Spiraea* sp. was given and all larvae went to it. In the last instar, after they apparently rejected *Spiraea*, *Myrica* (which is said to be, with *Comptonia*, the “native” food plant) was re-introduced and immediately taken until pupation. The continental larvae were reared in the laboratory on blueberry and *Myrica gale*, also on *Spiraea* sp. and birch (*Betula* sp.). In the field, we found larvae both on blueberry and *Myrica*. McGugan (1958) reported them also from some coniferous trees (larch, tamarack, white spruce, balsam fir). These records are all from northern localities, from Alberta to Ontario, but there exist many records stating that it is not possible to rear the species on tamarack from the egg, and this is my experience as well. Perhaps, however, displaced larvae can survive on coniferous foodplants. Adults reared from larvae collected on coniferous trees are all very small, as though from undernourished larvae.

Eggs for the rearings of *poecila* were obtained from the Research Station, Canada Department of Agriculture, St. John's, Newfoundland, through R. F. Morris; from the late A. S. Hessel, Washington, Connecticut; and from females caught by us in Algonquin Provincial Park, Ontario, and Chaffeys Locks, Ontario; of *gordius* from J. Müller, Lebanon, New Jersey; of *luscitiosa* from a female caught during summer collecting at Kendal, Ontario.

**Pupa** (Fig. 14). Deep reddish brown, with short detached tongue-case; 35-40 mm long; cremasterial end in lateral view dorsally rounded (more than in *gordius*), ventrally forming an evenly and softly curved line, cremaster slightly bent; bifurcation at apex of cremaster, each apex again minutely bifurcate. The pupa is the safest way to separate *poecila* from *gordius* where they live sympatriically, as in southernmost Ontario, northern New York State, parts of Connecticut and on the prairies (although it should be remembered that the
The pupa of the prairie *gordius* as well as that of the Utah/Colorado population is not yet known. The pupa of *gordius* (Fig. 15) is deep brown, about 40 mm long; cremasterial end semispherical; cremaster strong, slightly bent, quasi the prolongation of the dorsal end-line, arising from the middle of the semisphere; in lateral view dorsally only very slightly rounded, ventrally making an almost right angled turn; apex bifurcate with some ramifications at ends of forks. The pupa of *luscitiosa* (Fig. 16) is deep reddish mahogany brown, with a very short tongue-case which is free but lies closely against the thorax, about 35 mm long; cremasterial end shaped like a sugarloaf, the cremaster proper arising dorsally; no bifurcation but a single sharp pointed end; scar of caudal horn very much expressed. The pupal shape is intermediate between *gordius* and *poecila*.

**Adults.** *S. poecila* (Fig. 17) can be easily recognized, as Smith's remark "rather uniformly grey" is applicable to all specimens, and the species is very often, especially in more northern regions, strongly suffused with dark blackish-brown scales, in distinction from *gordius* (Fig. 18). It is rather somber looking, medium-sized, not rich in contrasts. There is an obvious cline in the color pattern from Newfoundland over Sable Island to Nova Scotia and farther to the west. The insular population is represented by large specimens with distinct submedian, median, postmedian, and marginal transverse bands in burnt umber (not darker); between the latter two a shorter broken intermediate line of the same color; the
postmedian band is especially strongly marked; marginal area not shaded in any way as in *gordius* (much more like in *S. drupifera* J. E. Smith). On the mainland, to the west and south, the ground color becomes lighter and the markings less distinct. The figures in Hodges (1971) show this difference well, Figures 3, 4 and 7 on Plate 5 representing *S. poecila* and Figures 5, 6, 8 and 9 on the same plate representing *S. gordius*.

At apex of forewing a horizontal fingerlike marking, not a compact spot; ground color of forewings bluish-grey, without purplish overtone; costal margin only slightly shaded; discal spot large, elongate, white (in *gordius* small and round); blackish lines and indistinct dashes between the veins and in the cell; veins finely marked with blackish; fringes checkered burnt umber alternating with brownish-white; hindwings brownish-grey (in *gordius* yellowish-grey) with solid burnt umber outer marginal band and a similar median one (in *gordius*, solid blackish outer marginal and median bands; fringes white cut with brownish, individual tendency of suppressing it); underside of both wings with unicolorous burnt umber between bands of hindwing, between median band and angle bluish-grey scales dominant, but only a few along the postmedian band on the forewing and around apex, also very slightly along the costal margin (in *gordius*: underside of forewings bluish-grey-brown; the postmedian and median bands stronger than the upperside; underside of hindwings of the same color; both bands pronounced). Head burnt umber with grey (in *gordius* greyish); disc of thorax a dark
burnt umber, metathoracic tufts light grey (usually extensive) with a few burnt umber scales intermixed (in gordius: blackish-brown with very dark metathoracic tufts, between which are some greyish hairs intermixed with brownish ones). Abdomen brownish-grey, with dorsal black line and broad lateral band interrupted with brownish-grey on four to five segments; ventrally burnt umber with mesial blackish spots indistinct (in gordius: ashy grey, with dorsal black line and broad black lateral band interrupted with whitish-grey on four or five segments, ventrally greyish-brown with distinct, perceptibly elongated blackish mesial spots).

Size. Length of forewing in male 35.2 mm, in female 40.1 mm (16 males, 8 females measured) in the continental populations; 36.8 mm in males, 39.75 mm in females (15 males, 8 females measured) in the Newfoundland population. For colored figures besides those in Hodges (1971) see also Holland (1968), Plate 5, No. 13, and Seitz (1940) Plate 95 b.

Flight period mid-May to second half of July, later in northern regions. In the laboratory we found that early emergence of a partial second generation is possible from larvae reared.
from the egg as well as from mature larvae collected in the field. One larva, for example, was found mature 25 August, pupated 8 September and a male moth emerged 13 October. Larvae usually have a short period of development of about 5.5 to 6.5 weeks, while in gordius the larval development takes about 8 weeks. Pupae may remain dormant for 20 months, but then show high mortality rate (in the laboratory). The life history and biology of S.luscitiosa are well-described by Hodges (1971).

ANATOMY OF ADULTS

Head. Distance between exterior margin of antennae sockets only slightly greater than the distance between lower margins of eyes; this gives the head a squarish appearance (Fig. 19); laterofrontal suture not visible; cranium at vertex strongly arched; long black bristles along inner margin of eyes; labial palpus (Fig. 25) with three segments, the second one the largest, the first and second curved, the third one very small, knobshaped, more elongate in

females. In the center of the cranium four squarely arranged dot-like depressions. In *gordius* the distance between exterior margin of the antennal sockets is about twice as wide as the distance between the lower margins of the eyes; this gives the head a rounded appearance (Fig. 20); labial palpus (Fig. 26) with three segments. differing in proportions from *poecila* and *luscitiosa* (Fig. 27). The first segment of the labial palpus also shows in the three species, different arrangements of sensory bristles (not to be confounded with palpal sensory hairs in the Macroglossinae). The distance between the base of palpus and the apex of the third segment is specifically different: *poecila*=3.368 mm; *gordius*=3.663 mm; *luscitiosa*=2.905 mm. In *luscitiosa* the form of the head is intermediate (Fig. 21), however, the laterofrontal suture is visible making *luscitiosa* a plesiomorph species in the group under discussion.

**Legs.** Epiphysis of anterior legs in male and female present. rounded. basal half wide open; extending in sharp point bent exteriorly; covered with very small bristles: about half as long as tibia; anterior legs without spurs; middle legs with one pair of distal tibial spurs, the exterior one about twice as long as the interior one; the posterior legs with two pairs of tibial spurs, one pair distal, the other at midlength, the exterior spurs about twice as large as the interior ones; paronychium present, one long lateral lobe; pulvillus vestigial. but distinct; empodium with one strong, short bristle. Figured are the epiphyses of all three species for comparison (Figs. 22–24).

**Male genitalia** (Figs. 28, 29). Valve large, usually ellipsoid (in *gordius* distinctly triangular), ventral margin often producing a broad phlange in the angle of the two processes (especially in the Labrador population, and also occasionally in the Newfoundland and Nova Scotia populations); dorsal part of process of sacculus large, the apex usually bent ventrally; basal part with slightly rounded or straight ventral margin, sometimes truncate; there is a tendency to develop asymmetric valves (in *gordius* [Fig. 30]; process of sacculus made up of two parts, a spoon shaped lobe ventrally and a dagger-like part dorsally, the
latter, of almost constant width, rarely narrowing distally, with ventrally bent apex which has many little thorns; basic line of sacculus makes deep notch where it goes over into lobe; aedoeagus more or less sigmoidal distally, sharply pointed conical process; opening of this process faces laterally in the Newfoundland population, length 0.8–0.95 mm, in the continental populations 0.55–0.75 mm; vesica tripartite (in gordius: aedoeagus ending in a straight, solid, sharply pointed conical process; opening of this process laterally 0.5–0.69 mm). Those of luscitiosa (Fig. 31) differ remarkably in that the process of the sacculus is shaped like the jaws of pliers. The fact that the parts are both flat and not elaborated may be taken as another sign of plesiomorphy of the species.

Female genitalia (Fig. 32). Ovipositor valves in lateral view sub-rectangular (in gordius [Fig. 33]: in lateral view regularly rounded): antrum wider than in gordius and on the right side straight while the left side is conspicuously angled (in gordius antrum tapers evenly), sometimes twisted (Newfoundland population); ductus seminalis connected in middle of lateral opening of cephalad end of antrum, before bursa copulatrix is attached in a knoblike structure; bursa copulatrix ovaly rounded (in gordius oblong): signum distinctly V-shaped, the two branches of the V diverging only slightly (in gordius: longer and narrower, branches nearly parallel), granulation finer than in gordius. The female genitalia of luscitiosa (Fig. 34) with their rounded bursa copulatrix and the narrowly V-shaped signum are nearer to poecila than to gordius, although they show in the connection of the bursa copulatrix to the ductus bursae a significant development of their own.

**DISTRIBUTION**

The distributional information is summarized in Figures 35-37. It should be remarked that the Arkansas records could not be verified as to what species is concerned. These records were taken from the literature (Freeman 1938). However, given the circumstances of the fauna of this state, they very probably are poecila. Better collecting should also yield poecila from more midwestern and western states in the U.S.A.

**Material examined:** CANADA: Alberta, British Columbia (Creston only), Labrador, Manitoba, New Brunswick, Newfoundland, Nova Scotia, Ontario, Prince Edward Island,
Sable Island, Quebec, Saskatchewan. UNITED STATES: Connecticut, Maine, Massachusetts, Michigan, Missouri, New Hampshire, New Jersey (no further data; specimen in American Museum of Nat. Hist., New York, Wisconsin. The record for British Columbia is a new one for this province and the one for New Jersey is a single known one.

**Type material.** The type of *poecila* must be considered lost. The type of Clark's *gordius borealis* may be found in the Carnegie Museum, Pittsburgh, Pennsylvania; it is a male from Gull Lake, Ontario. The type of Cadbury's *gordius f. coxeyi* is in the collection of the Academy of Natural Sciences in Philadelphia, Pennsylvania, and was captured “in August” on Greenly Island, Quebec (not Labrador, as Cadbury says in the original description); it is a female.

**DISCUSSION**

There can be no substantial doubt that these taxa form a closely related group. Grote (1875) discussed placing *gordius* and *luscitiosa* together in a separate genus *Lethia* Hübner. Morphological similarities in the adults are sufficiently strong to make this clear. However, distinct differences among these taxa also occur. These distinctions become still more convincing when one takes into consideration the immature stages in the group.

Limits of the taxa treated here were determined as follows. After dissecting roughly 100 specimens of the group it was found that the male genitalia provide distinguishing characters, especially in the aedeagus, while the female genitalia yield useful additional evidence (antrum and shape of ovipositors in lateral view). The form of the antrum and its connection to the bursa copulatrix (a second organ showing interspecific variation in shape)
for two days in the same cage. The males tried to locate their appropriate females but a clear-cut conclusion is possible with completely artificial methods.

Munroe (1956) called "Beringia" the most important and least disputed of the northern refugia and described it briefly. This could well have been the place where lusciriosa survived. Its later distribution would coincide with glacial refugia proposed by biogeographers. Munroe (1956) called "Beringia" the most important and least disputed of the northern refugia and described it briefly. This could well have been the place where lusciriosa survived. Its later distribution would coincide with glacial refugia proposed by biogeographers. Munroe (1956) called "Beringia" the most important and least disputed of the northern refugia and described it briefly. This could well have been the place where lusciriosa survived. Its later distribution would coincide with glacial refugia proposed by biogeographers.

At localities such as McCreary, Manitoba, and Saskatoon, Saskatchewan, gordius occurs together at the same time of the year with poecila, as it does also in the east, e.g., in Washington, Connecticut. A search was made for intermediate specimens in collections from these areas, but none were found. All specimens in collections may be easily assigned to one species or the other.

Hybridization experiments were conducted with freshly emerged pairs of gordius from Lakehurst, New Jersey, and poecila from Algonquin Provincial Park, Ontario, kept together for two days in the same cage. The males tried to locate their appropriate females but seemed to be so confused that no copulation resulted at all. Hand pairing was not tried as no clear-cut conclusion is possible with completely artificial methods.

The distributions for these three species, as shown on the maps (Figs. 35-37), coincide well with glacial refugia proposed by biogeographers. Munroe (1956) called "Beringia" the most important and least disputed of the northern refugia and described it briefly. This could well have been the place where luscitiosa survived. Its later distribution would coincide with that of many other insect species. Munroe (1956) further suggested another refugium in the northeast, quoting a number of authors on this controversy in the opinion that, while theories of migrating biota may be largely correct, they do "not, however, account for the general occurrence of endemics and disjuncts in the Gulf [i.e., of St. Lawrence] region, at low levels as well as at high, both as localized forms and as radiants in Labrador and elsewhere in the Northeast. An eastern refugium separate from the general southern glacial-

Fig. 36. Distribution map of S. gordius

is quite different from species to species. Using it as a criterion it is possible to define three distinct populations: the one in the north (poecila), the one in the south and on the prairies (gordius) and the one sympatric with both (luscitiosa).
margin strip is required. Data presented by Flint (1940) suggest that isostatic depression probably did not seriously affect the Grand Banks and Sable Island banks. Howden (1970) added that "the validity of the above is contingent upon the geological history of the Sable Island and Grand Banks area." Lindroth (1963) considered that some carabids of larger than usual size may have survived the last glaciation on or near Newfoundland. *Sphinx poecila* may well have done the same.

Shapiro (1970) stated, "The Omaha, Nebraska area seems to have been a xerothermic refugium. . . The Omaha area was well suited as a refugium because of its ravines and valleys, deepened in Wisconsin time and later partly filled with alluvium." Nebraska, it may be noted, is the center of the range of the western populations of *gordius*. Shapiro (1970) also mentioned the relict area of the coastal plain at the North Carolina-Virginia border which may well have served as the refugial territory of the eastern population of *gordius*. This would perhaps account for the slight differences between eastern and western populations of that species (Hodges 1971).

A combination of morphological and zoogeographic evidence may be used to suggest a possible phylogeny for the three species discussed in this paper. The present situation can be explained by the existence of two preglacial species: *luscitiosa* and a pre-*gordius*. During glaciation, *luscitiosa* was reduced to a single population in a northern refugium and has altered little during postglacial times, remaining a primarily northern species. The widespread pre-*gordius* species, however, was split into three different refugial populations by the advancing glacier: Newfoundland, Omaha-Nebraska, and the eastern coastal plain. The northeastern population was isolated longest and upon renewed post-glacial contact with its
sister groups proved specifically distinct (poecila). The two southern populations (gordius), however, were separated for only a relatively short interval and free gene flow between them apparently resumed after secondary contact had been established. This hypothesis is also supported by the fact that poecila and gordius are the more apomorphic species in the group.

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A study of 100 cocoons of *Antheraea polyphemus* (Cramer) collected on and around isolated foodplants in southern Michigan reveals that their formation is various. Some remain attached to the foodplant, some fall and overwinter on the ground, and others are spun directly at ground level among grasses close to the foodplant axis. Ninety-six of the cocoons were clearly associated with foodplants, involving 16 species belonging to nine families. Species of oak (*Quercus*) were most productive.

The polyphemus moth, *Antheraea polyphemus* (Cramer), is the most widespread giant silkworm moth in North America, ranging from Canada to Mexico and coast to coast. During the 1800's its cocoon was considered as a possible source of commercial silk (Trouvelot 1868). Popular everywhere, this insect is readily raised in culture and is widely used for demonstrations in teaching. The larvae are reported to feed on a lengthy list of foodplants. Nevertheless we found considerable confusion in the literature regarding the mode of cocoon construction, and we therefore investigated the foodplants and cocoons of this moth in southern Michigan.

The North American giant silkworm moths have two categories of cocoons, those with "valves" through which the moth emerges (*Hyalophora, Callosamia, Samia, Rothschildia, Eupackardia*) and those which lack such valves, emergence being accomplished by secretion of substances that enable ready separation of the silk to make an orifice at one end (*Actias, Antheraea*). Those of the former type tend to be lanceolate in outline, the latter elliptic. Most species tend to form their cocoons in characteristic locations and attach them in characteristic ways, as will be discussed below.

The situation with respect to polyphemus, however, is much confused in past reports. For example, Holland (1968) wrote that the cocoon “is spun among the leaves and falls in the autumn to the ground” (italics added). On the contrary, Essig (1926) described the cocoons as “tightly fastened to the branch with silk to prevent the natural drop in the autumn. The large suspended cocoons are readily discernible in fruit orchards during the winter months and are readily gathered and destroyed by the growers” (italics added). Borror et al. (1976) stated, in contrast, that the cocoon “is formed in a leaf on the ground” (italics added). Which is correct?

To answer this question, and also to find out what foodplants are preferred by polyphemus in southern Michigan, we carried out an extensive search for the cocoons during the months of January through April 1980. In spite of the abundance of the adult moth, which is taken routinely at lights everywhere in this region, its cocoons are considered by most collectors to be difficult to find. We examined the branches and the ground below several thousand

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trees and shrubs, mostly in western Washtenaw County and the adjacent townships of Jackson and Livingston counties. Almost all of the plants we examined were solitary and in the open, along roads, hedgerows, and fields. Only in this way could we be confident of the foodplant associations, and we have found that the incidence of cocoons is greater in open-grown, isolated plants than in large colonies of foodplants in the forest. Most of the oak, hickory, and willow trees were 15 to 30 m tall, the birches and hawthorns 5 to 10 m tall, and the shrubs 2 to 4 m tall.

We set an arbitrary limit of finding 100 cocoons. Of these, 60 were "living" and 40 "dead." Table 1 gives an analysis of the positions in which the cocoons were found and their covering materials. Seventeen cocoons, approximately one in six, were found still hanging in the trees. The rest were found lying on the ground. These belonged to four categories of covering materials: surrounded by broad leaves of dicotyledonous woody plants, surrounded by both broad leaves and grass leaves, surrounded by grass leaves only, and covering unidentified (owing to eroding away on cocoons left over from past years). The presence of grass, whether or not in association with broad leaves, was considered to be a sure sign that the cocoon was actually constructed on the ground. Using this criterion, we found that about half of the total cocoons were formed away from the foodplant. In the category of cocoons covered only by broad leaves, we can only guess where they were formed. Some were, no doubt, formed upon the foodplant and dropped to the ground. However, this does not preclude that others were constructed in fallen broad leaves on the ground.

Of particular interest is the fact that polyphemus cocoons were positioned in all of the ways reported by previous authors, hanging on the foodplant, falling to the ground, and built on the ground below the foodplant. Only Eliot and Soule (1902) and Robertson-Miller (1949), of 20 sources consulted, gave the state of affairs correctly. Polyphemus proved to be the most versatile of our silkworm moths in the construction of its cocoons, essentially covering the gamut shown by our other North American species. The different conditions are enumerated below:

(a) Cocoon attached longitudinally, as in the genus *Hyalophora* (Figure 1a). The larva spins a longitudinal connection to the twig which holds the cocoon in place and prevents it from falling during the winter. There may or may not be leaves still attached to the cocoon. The longitudinal connection differs from that of *Hyalophora* in being generally weaker and commonly not entirely parallel to the axis of the cocoon.

(b) Cocoon attached apically, as in *Callosamia promethea* Drury, *Samia*, *Rothschildia*, *Eupackardia*, by a distinct silken peduncle attached to the twig (Fig. 1b). This type is nicely illustrated by Lutz (1921). Sometimes there is a double peduncle, but usually it is a single string-like attachment that follows the petiole of one of the subtending leaves. Generally the peduncles are weaker than those found, say, in *Callosamia promethea* or *Rothschildia orizaba* Westwood.

Table 1. Late-winter positions and covering materials of cocoons of *Antheraea polyphemus*.

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<th>On plant</th>
<th>Broad-leaf covering</th>
<th>Broad-leaf + grass</th>
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aCocoons which, when shaken, appeared to have living contents.
bEmpty cocoons, or contents hard and dried out.
Fig. 1. A. Two cocoons on left attached longitudinally along the side: one on the right merely wrapped in leaves but still remaining on tree. All three on *Betula alba*. B. Cocoons attached by peduncles; (left) on *Crataegus* sp. spine; (middle and right) on *Betula alba* twigs.

(c) Cocoon formed arboreally among leaves, but without definite attachment and falling to the ground, as in *Callosamia angulifera* Walker (Fig. 1a, cocoon on right; Fig. 2a). Surprisingly these cocoons, which are not attached to any stems but only leaf blades, may remain on the plant until March. We have, however, experimentally shaken them off by swinging the supporting branches. Cocoons found on the ground which are surrounded only by broad leaves are probably in large part of this category.
Fig. 2. A. Cocoons found upon the ground and wrapped entirely in tree leaves. B. Cocoons found upon the ground and wrapped partially in tree leaves (Quercus) and partially in grass leaves.

(d) Cocoon formed terrestrially, as in Actias luna L. (Fig. 2b; Fig. 3a). These are spun among fallen leaves and ground-growing grasses and herbs. They are readily separated from Actias cocoons because of their much thicker walls and light tan, rather than dark brown, color.

Unlike Callosamia angulifera, the cocoons of which are widely scattered under its food-plant (Liriodendron), sometimes as much as 5 m from the trunk, Antheraea polyphemus cocoons are practically always within 1 m of the foodplant trunk. Once this is realized it
Fig. 3. Cocoons covered by grass leaves only.

makes it easy to find them. Indeed, the majority of polyphemus cocoons occurred within 0.2-0.5 m of the trunk, indicating that the caterpillars crawl down the bark to the ground and there pupate, without making extensive travels to the cocoon-building site.

We found cocoons associated with 16 species of foodplants (Table 2). The inclusion of Corylus in Table 3 is based upon finds in years prior to 1980. A total of 55 plants yielded cocoons. The most productive genera were hickories (Carya), birches (Betula), and oaks (Quercus). A smaller number of cocoons was found with hawthorn (Crataegus) and willow (Salix), and even fewer cocoons on sycamore (Platanus) and basswood (Tilia). Nielsen (pers. comm.; see table 3) reports several other genera.

Members of the same genus are not necessarily chosen equally by polyphemus. We found that the cocoons were much more commonly associated with the shagbark hickory (Carya ovata) than with false shagbark or pignut hickory (C. glabra). We found far more cocoons under trees of the white oak group (Q. alba, Q. macrocarpa) than the black oak group (Q. velutina).

All of the birches that yielded cocoons were cultivated plants; one of them, Betula pendula, not a native species. It is interesting to note that Nielsen (pers. comm.) discovered two polyphemus cocoons in Iron County, Michigan, where they were hanging on the shrubby bog birch (B. pumila). A reason for our paucity of records from sycamore (Platanus occidentalis) is probably the fact that it is a relatively uncommon tree in this region, and we were therefore able to examine only a small number of individuals.

We consider all of the plants reported here from our field studies to be primary larval foodplants, ones upon which the eggs were actually laid by the female and upon which the larvae actually fed; as opposed to secondary foodplants, those to which larvae later migrated and fed; or false foodplants, to which larvae migrated but did not feed, merely using them as places for temporary support or to construct cocoons and undergo pupation. Clearly we would be in error to consider the various grasses in which polyphemus cocoons were spun at the bases of trees as primary or even secondary foodplants. These grasses are false food-
Table 2. Association of cocoons with isolated foodplants, February-April 1980. For each plant species, the total of cocoons found associated with it is given, together with the number (in parentheses) of plants on which, or under which, they were found.

<table>
<thead>
<tr>
<th>Plant Species</th>
<th>Number of Cocoons</th>
</tr>
</thead>
<tbody>
<tr>
<td>Acer saccharinum L.</td>
<td>2 (1)</td>
</tr>
<tr>
<td>Betula alba L.</td>
<td>8 (6)</td>
</tr>
<tr>
<td>Betula pendula Roth.</td>
<td>5 (1)</td>
</tr>
<tr>
<td>Carya glabra (Mill.) Sweet</td>
<td>1 (1)</td>
</tr>
<tr>
<td>Carya ovata (Mill.) K. Koch.</td>
<td>21 (19)</td>
</tr>
<tr>
<td>Cornus racemosa Lam.</td>
<td>3 (2)</td>
</tr>
<tr>
<td>Crataegus sp.</td>
<td>3 (3)</td>
</tr>
<tr>
<td>Platanus occidentalis L.</td>
<td>1 (1)</td>
</tr>
<tr>
<td>Quercus alba L.</td>
<td>14 (5)</td>
</tr>
<tr>
<td>Quercus X bebbiana Schneid.</td>
<td>6 (1)</td>
</tr>
<tr>
<td>Quercus bicolor Wild.</td>
<td>2 (1)</td>
</tr>
<tr>
<td>Quercus macrocarpa Michx.</td>
<td>21 (6)</td>
</tr>
<tr>
<td>Quercus velutina Lam.</td>
<td>4 (4)</td>
</tr>
<tr>
<td>Salix babylonica L.</td>
<td>2 (2)</td>
</tr>
<tr>
<td>Salix fragilis L.</td>
<td>2 (1)</td>
</tr>
<tr>
<td>Tilia americana L.</td>
<td>1 (1)</td>
</tr>
<tr>
<td>Uncertain, a</td>
<td>4 (?)</td>
</tr>
</tbody>
</table>

*aPossible migrants from other species.

Table 3. Polyphemus foodplants from Tietz (1952) arranged by taxonomic relationships following Cronquist (1968).

<table>
<thead>
<tr>
<th>PINOPSIDA</th>
<th>Vitaceae</th>
<th>Platanaceae</th>
</tr>
</thead>
<tbody>
<tr>
<td>Pinidae</td>
<td>Vitis</td>
<td>Platanus</td>
</tr>
<tr>
<td>Pinaceae</td>
<td>Juglandaceae</td>
<td>Ulmus&lt;sup&gt;a&lt;/sup&gt;</td>
</tr>
<tr>
<td>Pinus</td>
<td>Juglans</td>
<td></td>
</tr>
<tr>
<td>MAGNOLIOPSISIDA</td>
<td>Carya</td>
<td>Dilleniidae</td>
</tr>
<tr>
<td>Rosidae</td>
<td>Cornaceae</td>
<td>Salicaceae</td>
</tr>
<tr>
<td>Rosaceae</td>
<td>Cornus</td>
<td>Populus</td>
</tr>
<tr>
<td>Rosa</td>
<td>Hamamelidae</td>
<td>Salix</td>
</tr>
<tr>
<td>Amelanchier</td>
<td>Hamamelidaceae</td>
<td>Tilia</td>
</tr>
<tr>
<td>Crataegus</td>
<td></td>
<td>Tiliae</td>
</tr>
<tr>
<td>Cydonia</td>
<td>Betulaceae</td>
<td>Arbutus</td>
</tr>
<tr>
<td>Malus</td>
<td>Alnus</td>
<td>Astereidae</td>
</tr>
<tr>
<td>Pyrus</td>
<td>Betula</td>
<td></td>
</tr>
<tr>
<td>Amygdalus</td>
<td>Carpinus</td>
<td></td>
</tr>
<tr>
<td>Prunus&lt;sup&gt;a&lt;/sup&gt;</td>
<td>Corylus</td>
<td></td>
</tr>
<tr>
<td>Saxifragaceae</td>
<td>Ostrya</td>
<td></td>
</tr>
<tr>
<td>Ribes</td>
<td>Fagaceae</td>
<td></td>
</tr>
<tr>
<td>Fabraceae</td>
<td>Castanea</td>
<td></td>
</tr>
<tr>
<td>Gleditsia</td>
<td>Fagus</td>
<td></td>
</tr>
<tr>
<td>Acrecaceae</td>
<td>Quercus</td>
<td></td>
</tr>
</tbody>
</table>

<sup>a</sup>Confirmed as a foodplant in Michigan by Nielsen (pers. comm.), who also has unreported records on Fraxinus (Astereidae: Oleaceae) plus several horticultural genera,
<sup>b</sup>See text regarding inclusion of this genus.

Plants, merely ones in which the caterpillars chose to spin up their cocoons. We found a large, healthy cocoon in Ann Arbor attached to a barberry bush (Berberis thunbergii); however, the position of this bush in close proximity to such potential foodplants as Juglans nigra and Carya ovata makes its candidacy as a true foodplant questionable.

It is interesting to note that the only attached cocoons were found on birches, hawthorns, and the foregoing barberry bush, the plants with the shortest leaves. In previous years we have also observed them hanging on gray dogwood (Cornus racemosa) and willows (Salix spp.). It is also interesting to note that polyphemus cocoons fall off of shrubs just as readily as...
trees. We found cocoons under *Cornus racemosa* on the ground between the upright stems from which they had fallen.

In Table 3, the foodplants for polyphemus listed by Tietz (1952) are arranged in subclasses (following mainly the taxonomy of Cronquist [1968]). In our study we found examples in every subclass except for the Pinidae and Asteridae, both of which seem to be quite isolated from the other subfamilies on which polyphemus is known to feed. It seems to us, therefore, that further confirmation of natural feeding of this moth on pines (*Pinus*) is called for, as well as on red pepper (*Capsicum annuum*) (given by Tietz as *Capricltm annum* L., presumably a typographical error).

**SUMMARY AND DISCUSSION**

The confusion that exists in the literature regarding the nature of cocoon construction in the polyphemus moth is resolved by our study of 100 cocoons found in winter and early spring in southern Michigan. The cocoons may remain attached to the foodplants, may fall off, or may be constructed on the ground. The moth is the most versatile in cocoon construction of North American giant silkmoths; its various cocoon forms match all four different types found among the other genera and species. Reports of foodplants for polyphemus in the past include 17 families. We found examples in nine of these families in southern Michigan, and probably others will be found. Of past reports we consider as dubious the families Pinaceae and Solanaceae. It is possible that these are actually false foodplants, to which caterpillars had merely crawled from the true foodplants.

Our study brings up a number of questions. Among them is what causes the destruction of so many cocoons. One in three were torn open by agents other than normal emergence of the moths. The holes made by the ichneumonid wasp *Enicospilus americanus* Christ (*E. macrurus* of authors), resemble very closely some of the smaller openings we found on the torn-open cocoons. From one of our “living” cocoons a specimen of this large wasp emerged, but not one of our torn-open ones showed the characteristic wasp cocoon within; all were either empty or contained remains of the pupal skin. We can only speculate at this time that the holes and tears found on so many cocoons were caused by birds, white-footed mice, and/or shrews. Such animals may peck or bite open the cocoons to eat the contents within. One torn cocoon we encountered in March still had remains of the pupa within; it had been eaten almost entirely, leaving most of the pupal skin with some flesh attached to it, which ants were eating.

In our work we have found 16 species of definite foodplants, although earlier authors have listed three times this number. It seems highly probable that each area has its distinctive combinations of foodplants which are preferred by polyphemus. There are foodplants still to be discovered, species within previously reported genera, and perhaps even other genera. Also, the type of cocoon construction and overwintering position may differ from region to region. Studies like the present one should therefore be made in other parts of the range of this moth.

**ACKNOWLEDGMENTS**

We thank David J. Bay for making the photographs, and Joseph Beitel, Dennis Brown, Michael Gumina, Robert Masta, and Robert Stewart for their help in the field. Henry Townes and Thomas Webb provided us with a number of suggestions. We especially wish to acknowledge the aid of Mogens Nielsen, whose friendly and enthusiastic assistance to us and other students of Michigan Lepidoptera has been a source of steady inspiration.

**LITERATURE CITED**


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ABSTRACT

A North Carolina specimen of *Euschistus servus* (Say) with midventral abdominal spots is reported.

The genus *Euschistus* is found throughout North America and contains some of the more common species of stink bugs. These are generally yellow, brown, or black in color, sometimes suffused or flecked with red or green (Rolston 1974), and often very similar in external appearance. Two of the more widespread species are *Euschistus tristigmus* (Say) and *E. servus* (Say).

*E. tristigmus* and *servus* share several external features including black spots on the hemelytral membrane, spiracles concolorous with supporting plates, a black spot in the lateral incisure of each ventral abdominal segment, and others. As a result, they often key out in the same or nearby couplets. The character often used to separate them is the presence of one to four midventral abdominal spots in *tristigmus*, and their absence in *servus* (e.g., Blatchley 1926, Froeschner 1941, Furth 1974, Hoffman 1971, McPherson 1970, Rolston 1974, Slater and Baranowski 1978, Torre-Bueno 1939); occasionally, the spots may be lacking in *tristigmus* and a faint trace of a spot may be present in *servus*.

I recently found a peculiar male *Euschistus* among pentatomoid specimens collected 26 May 1978 by window trapping in a black walnut (*Juglans nigra* Linnaeus) plantation near Asheville, North Carolina. It seems to possess all characters of *servus* except for the presence of a row of midventral abdominal spots (Fig. 1). Since nothing, other than the spots, suggests it is a hybrid, it must simply be labeled an aberrant specimen.

The specimen is deposited in the Entomology Collection, Southern Illinois University Zoology Research Museum.

ACKNOWLEDGMENTS

I wish to thank Ms. Barbara C. Weber, USDA North Central Forest Experiment Station, Carbondale, for donating the Asheville pentatomoid collection to Southern Illinois University, Carbondale, and Dr. Richard C. Froeschner, U.S. National Museum, Washington, D.C., for examining the *E. servus* specimen.

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Fig. 1. Dorsal (upper row) and ventral (lower row) views of typical _E. serus_ (left), aberrant _E. sercus_, and typical _E. tristigmus_ male specimens.

**LITERATURE CITED**


LIFE HISTORY OF THE AMBROSIA BEETLE XYLEBORUS AFFinis (COLEOPTERA: SCOLYTIDAE) FROM IN VITRO CULTURE


ABSTRACT

In vitro cultures of the wood-boring beetle X. affinis were fed on their symbiotic fungi. All the various life stages of the beetle developed more rapidly at 29°C than at 22–24°C. Head capsule measurements indicated the development of three larval instars. In 84.8% of the cultures only a single progeny adult male was found. Observations indicated that the progeny male was the first to develop in each brood. X. affinis had an adult female: male sex ratio of 8.5:1 from in vitro cultures.

Ambrosia beetles attack weakened, wind thrown, or cut trees. Adult females of Xyleborus species bore tunnel systems into the xylem in which they cultivate their symbiotic fungi and oviposit. The larvae feed exclusively on the fungi lining the walls of the tunnel system. Adults of Xyleborus are sexually dimorphic with adult males being smaller, flightless, haploid, and reproduced parthenogenetically; while the females are diploid (Norris 1972). Saunders and Knoke (1967) were the first to successfully culture in vitro an ambrosia beetle Xyleborus ferrugineus Fabricius. French and Roeper (1972) have cultured X. dispar Fabricius. Roeper et al. (1980) described the initiation of X. affinis Eichhoff with its symbiotic fungi into culture in various media from Michigan-collected beetles. This study describes aspects of the development of life stages, larval instars, and sex ratios of affinis from in vitro culture.

METHODS

Adult females of affinis with their symbiotic microbes from stock cultures were individually placed in tubes of sterile culture medium “D” which consisted of sucrose (20 g), vitamin-free casitone (10 g), yeast extract (10 g), wheat germ (5 g), corn oil (10 ml), T-19 mineral solution (40 ml), ground aspen wood (150 g), agar (40 g) and 1000 ml of distilled water (Roeper et al. 1980). All culture tubes were maintained in the dark at either 22–24°C or 29°C. Two cultures with active broods from each temperature control were harvested daily for 35 days to obtain life stages. Specimens were examined directly after harvesting or after being preserved in 70% ethyl alcohol. Head capsule widths and body lengths were measured with an ocular micrometer.

RESULTS AND DISCUSSION

Initial tunneling activity by the adult female occurred prior to oviposition (Roeper et al. 1980). Eggs were off-white, shiny, and oblong with a mean length of 0.718 mm ranging from 0.618 to 1.045 mm (n = 30). Groups of two to four eggs were laid in short horizontal

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branching tunnels from the main vertical tunnel. From portions of the tunnels exposed to the side of the culture tubes the adult females were observed tending the eggs and cropping the fungal growth. When exposed to a strong light the maternal adult female would move the eggs to other tunnels of the gallery system. Adult female tending behavior seemed important to progeny development. Numerous attempts to culture eggs independently from the maternal female failed in tubes or petri dishes of culture media preinoculated with the symbiotic fungi of *affinis*. At 22–24°C the first eggs were observed on the 11th day after the introduction of the adult female and were last seen on the 27th day. At 29°C eggs were laid at three days and were last observed on the 12th day.

White, slightly curved, legless larvae were first observed after 14 days and a few encountered at 35 days at 22–24°C; while at 29°C larvae developed at seven days and were last observed at 16 days. Upon hatching larvae moved freely about in the tunnel system and were occasionally found on the upper surface of the culture medium. They actively fed upon the symbiotic fungi and did not appear to consume the culture medium. Some larvae moved out of the tunnels and occupied a space which occurred between the slightly dehydrated medium and the side of the culture tube. These larvae usually died.

Head capsule measurements of *affinis* larvae indicated three distinct larval instars (Table 1). This corresponds with three larval instars recorded in *ferrugineus* from in vitro cultures (Peleg and Norris 1973). Discrimination between haploid male larval and diploid female larvae was not made, but it can be assumed female larvae dominated the cultures based on the sex ratio of resulting adults. Peleg and Norris (1973) found only the male and female 3rd instar larvae could be confidently separated. The greatest deviation in head capsule widths were found in the 3rd instar larvae of *affinis*.

Pupae were first observed at 21 days and a few were still present at 35 days at 22–24°C. At 29°C the first pupae were observed at 11 days and were not observed after 23 days. Pupae remained stationary but were observed reflexing their abdominal section. Pupae were white, then pigmented to light brown just prior to the emergence of the callow adult. Male pupae would be distinguished by a blunt projection on their anterior dorsal section and their length which ranged from 1.9 to 2.2 mm with a mean of 2.0 mm (n = 12). Female pupae measured 2.5 to 2.9 mm in length with a mean of 2.7 mm (n = 32).

Callow adults were seen at 27 days and dominated cultures after 35 days at 22–24°C. At 29°C the first adult was found at 18 days and all progeny were adult at 35 days. Newly emerged callow adults were easily distinguished by their light pigmentation. Normal adult pigmentation, light reddish brown, generally occurred after a week. Progeny adults moved extensively through the tunnel system and fed on fungal growth lining the walls. Adult females measured 2.5–2.9 mm in length with a mean of 2.7 mm (n = 42) and males, with their characteristic blunt point on their pronotum, measured 1.8–2.0 mm in length with a mean of 1.9 mm (n = 18). These measurements correspond to field collected specimens (Bright 1968). Of a total of 145 culture tubes harvested between 35–45 days a single male progeny was found in 123 tubes (84.8%), two adults were found in nine tubes (6.1%), and 13 tubes (9.0%) had all male progeny. A fertilized adult female appeared generally to lay only one male egg per brood. Culture tubes with all male progeny were probably parthenogenetically reproduced from virgin females as observed by Norris (1972) in *ferrugineus* cultures. The

### Table 1. Head capsule widths in micrometers (µm) of *X. affinis* mixed male and female larvae from in vitro culture.

<table>
<thead>
<tr>
<th>Instar</th>
<th>No. of measured larvae</th>
<th>Width (µm)</th>
<th>Mean ± SD</th>
</tr>
</thead>
<tbody>
<tr>
<td></td>
<td></td>
<td>Least</td>
<td>Greatest</td>
</tr>
<tr>
<td>1st</td>
<td>23</td>
<td>224</td>
<td>241</td>
</tr>
<tr>
<td>2nd</td>
<td>46</td>
<td>270</td>
<td>331</td>
</tr>
<tr>
<td>3rd</td>
<td>68</td>
<td>390</td>
<td>469</td>
</tr>
</tbody>
</table>
female:male sex ratio of 8.5:1 was recorded for \textit{affinis} cultures. This female:male sex ratio compares with in vitro cultured reports of 2:1 for \textit{dispar} \cite{French_and_Roeper_1972} and 35:1 for \textit{ferrugineus} \cite{Saunders_and_Knoke_1967}.

Culture tubes were sampled daily to determine the order of emergence of the sexes from the pupae stage to that of the new progeny adult. Results indicate that in general the first emerged adult was a male. In cultures when a single progeny adult in a brood had emerged (n = 4 culture tubes) that adult was always a male. When two progeny adults had emerged (n = 5 culture tubes), one of the adults was a male and when three progeny adults had emerged (n = 5 culture tubes), one of the adults was always a male. The significance of these observations is that if the haploid male were the first adult to emerge, then an adult male would be always available to inseminate sibling diploid female adults as they emerged. Peleg and Norris \cite{Peleg_and_Norris_1973} found the male and female larvae of \textit{ferrugineus} developed at the same rate. This suggests the first egg oviposited by \textit{affinis} was the haploid male egg, but since eggs were always found in groups in culture tubes and eggs could not be cultured independently from the maternal female, this hypothesis could not be fully tested.

**ACKNOWLEDGMENT**

The support for this study came in part from a National Science Foundation Undergraduate Research Participation Grant to Alma College.

**LITERATURE CITED**


OBSERVATIONS OF THE AMBROSIA BEETLE
XYLEBORUS SAYI (COLEOPTERA SCOLYTIDAE)
INFESTING SUBCANOPY MAPLES IN MICHIGAN

Charles R. Hazen and Richard A. Roeper

ABSTRACT

Xyleborus sayi Hopkins was found to have a single generation infesting subcanopy maples. The beetle constructed simple branch gallery systems in which the life stages of beetle developed during the summer. Two larval instars were recorded. The female adult was found to have an intersegmental pouch type of mycangium between the pro- and mesonotum containing Ambrosiella hartigii Batra as its symbiotic fungus.

Members of the genus Xyleborus attack weakened, wind-thrown, or timbered trees in which they bore tunnel systems within the xylem. The adult female beetles disseminate and cultivate symbiotic fungi which grow on the tunnel walls and are consumed by larvae and progeny adults. Xyleborus sayi Hopkins has been recorded throughout eastern United States and Canada and ranges from Georgia to Illinois (Bright 1968). Known as X. obesus variety minor Swaine and Anisandrus sayi Hopkins, Bright (1968) synonymized these to Xyleborus sayi Hopkins. This study reports upon the biology of sayi attacking subcanopy maples (Acer) in Michigan.

METHODS

Segments of Acer rubrum L. and A. saccharum Marsh infested with beetles were collected from 1973-1977 in Montcalm County in central Michigan and Antrim County in the Northern Lower Peninsula of Michigan. These host timbers were split open upon return to the laboratory and measurements of tunnel systems and tree diameters were taken. Beetle life stages were either studied directly upon removal or preserved in 70% ethyl alcohol for later study. Measurements of body sizes and head capsule measurements were made with an ocular micrometer.

Isolations of the symbiotic fungus of sayi were made from the fungal transmitting organ (mycangium) of the beetles and brood tunnels and cultured on 3% malt extract and 1% yeast extract agar, and/or potato dextrose extract agar using techniques described by Batra (1967).

OBSERVATIONS AND DISCUSSION

Mycangium and Symbiotic Fungus. The mycangium was located by careful dissection of all external body sections and by microscopic examination (450x) using trypan-blue stain which discriminated between fungal cells and insect tissue. Only females of X. sayi were found to have an intersegmental pouch type mycangium between the pronotum and mesonotum. This mycangium was similar but smaller than the mycangium of adult females of X. dispar Fabricius (Franke-Grosman 1956). By removing the mycangium and streaking the fungal material on culture medium only the imperfect fungus Ambrosiella hartigii Batra was

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isolated. Repeated isolations of *A. hartigii* were also made from the tunnel walls of the gallery system which contained actively feeding beetle larvae. This fungus has been previously associated with *X. dispar* (Batra 1967) and *X. obesus* (Roepfer, Hazen, et al. 1980).

**Characteristics of Host Trees.** Bright (1968) listed *Acer, Betula, Carya, Fagus, Fraxinus, Juglans, Sassafras* and *Tilia* as tree hosts of *X. sayi*. In Michigan *X. sayi* was found most commonly in *Acer rubrum* (*n* = 42) and *A. saccharum* (*n* = 18). Collections were also made of *A. saccharinum* L. (*N* = 2), *Quercus rubra* L. (*n* = 1) and *Carpinus caroliniana* (Marsh) Fern. (*n* = 1). *X. sayi* probably can attack most deciduous trees within its range.

Spring attacks by *X. sayi* on *A. rubrum* and *A. saccharum* occurred on standing, small diameter trees with breast height diameter between 21–49 mm (mean = 39.7 mm; *SD* = 4.1; *n* = 60). These subcanopy maples appeared to be of low vitality. The leaves of the host trees were already wilted and/or yellowed when the initial attacks of *X. sayi*. only a day or two old, were observed. No sign of host tissue deterioration was evident upon dissection. The percentage of subcanopy maple attacks never exceeded more than 2.0% of the trees in any stand studied.

**Beetle Attack and Gallery Characteristics.** Only the adult female beetle attacked and initiated brood galleries in host trees. Single entrance holes are bored on surface scars on lenticels in the smooth bark of the small maples. The density of attack, measured by the number of entrance holes per surface area (dm²), averaged 0.95 entrance holes per dm² (*SD* = 0.32; range = 0.35–2.44 holes per dm²; *n* = 24). *X. sayi* appeared to space their entrance holes on the bark surface. The mean distance between an entrance hole and the nearest neighboring entrance hole was 133.9 mm (*SD* = 34.5; range 6–246 mm; *n* = 203). Only 12 entrance holes were closer than 50 mm in distance from each other. The single entrance tunnels were bored perpendicular to the bark surface to a depth of 7.0–10.0 mm into the xylem. Xylem borings were ejected from the entrance hole and usually did not accumulate on the bark surface. Lateral branch tunnels were then initiated perpendicular to the entrance tunnel on a horizontal plane. The lateral tunnels curved parallel to bark surface generally following a xylem growth ring. A lateral branch tunnel was completed with growth of the symbiotic fungus and oviposition before a second lateral branch was started in opposite direction beginning at the deepest penetration of the entrance tunnel. Lateral branch tunnels ranged from 13 to 22 mm in length (x̄ = 15.4 mm; *SD* = 3.1; *n* = 56). The ends of the lateral tunnels never met. In completed gallery systems additional short tunnels (4–6 mm) were bored from the main lateral branches. Completed systems (entrance tunnel and all lateral tunnels) from trees collected in September were longer in length with wider maple trunk diameters (linear correlation coefficient *r* = 0.803; *n* = 9). The shortest gallery length was 28 mm with a 26 mm tree diameter. The longest gallery length was 49 mm in a tree diameter of 54 mm. A tunnel width of 1.1 mm was constant throughout the gallery system.

**Life History of the Beetle.** In Michigan, *sayi* overwinter as adults within their galleries and in the spring the female adults fly to infest new tree hosts. The earliest spring attack was found in Montcalm County on 27th April. The latest initial attack was 3rd July in northern Antrim County. *X. sayi* was found to have only single generation a year in Michigan.

Eggs were whitish, translucent, oblong, and averaged 0.70 mm (*SD* = 0.01) long by 0.36 mm (*SD* = 0.01) wide (*n* = 12). Eggs were found from mid-May to late July. The adult female appears to lay eggs in groups since the lowest number of progeny found in gallery was four. Eggs were found usually at the end of lateral tunnels in association with luxuriant growth of the whitish ambrosia fungus which proliferated from the tunnel walls.

Larvae were white with brownish mandibles, legless, and slightly curved and usually fed actively on the fungal growth. Larvae were located throughout the gallery system except in the entrance tunnel and were present in the galleries from early June to mid-August. Two larval instars were identified based on two groupings of head capsule widths ranging from 0.284–0.347 mm (x̄ = 0.310 mm; *SD* = 0.023; *n* = 40) and from 0.441–0.504 mm (x̄ = 0.477; *SD* = 0.017; *n* = 58). Peleg and Norris (1973) and Roepfer, Treeful et al. (1980) recorded three larval instars for other *Xyleborus* species using in vitro culture techniques. It is possible that an earlier instar of *sayi* was missed since Peleg and Norris (1973) found the first instar of *X. ferrugineus* Fabricius existed for only 24 to 48 h after emergence from the egg.

Pupae were white and became tan with age. They were first observed by mid-July and a
few were found in galleries collected in September. The dimorphic sex characteristic of *Xyleborus* species could be recognized by the size differences. Female pupae averaged 2.7 mm (SD = 0.01; range 2.5-2.9; n = 23) in length, while male length averaged 1.4 mm (SD = 0.02; range 1.3-1.6; n = 13). Pupae were found located throughout the gallery system and seemed motionless unless moved by other larvae or adults.

Progeny adults as described by Bright (1968) were first observed in late July. These adults appeared inactive except for some feeding on the depleted fungal material on the tunnel walls. Live wingless adult males readily copulated with sibling females in petri dishes when removed from galleries in late August and September. This indicates insemination occurred prior to the overwintering period. The presence of dead males (n = 5) suggested that males do not always survive the overwintering period. Galleries sampled in September averaged 22.5 (SD = 3.2; range 14-25; n = 14) progeny per gallery. The number of progeny adults in each mature gallery system were found to increase as the length of gallery system increased (linear correlation coefficient $r = 0.779$; n = 9). A tunnel of 28 mm had 14 progeny, while the maximum number of 25 progeny was found in a gallery of 49 mm in length. The adult female:male sex ratio was 3.6:1. The maximum number of males found in any single gallery was five.

Known previously only from taxonomic description, host tree records, and distribution (Bright 1968), the habits of *X. sayi* appeared typical of other temperate distributed *Xyleborus* species. *X. sayi* had the same symbiotic fungus, *A. harfigii*, as found in closely related species of *X. dispar* and *X. obsesus* which had been placed in the beetle genus Anisandrus (Bright 1968). The observation of a single generation of *X. sayi* in Michigan does not exclude the possibility of multiple generations in the southern states from which it has been recorded.

ACKNOWLEDGMENT

The support for this study came from a National Science Foundation-Undergraduate Research Participation Grant to Alma College.

LITERATURE CITED


ABSTRACT

Alfalfa weevil larvae, *Hypera postica* (Gyllenhal), were collected from selected counties in Illinois to determine the incidence of infection from the fungus, *Entomophthora phytonomi* Arthur.

The fungus, *Entomophthora phytonomi* Arthur, has been recognized for many years as the most important, naturally occurring, beneficial control agent of the clover leaf weevil, *Hypera punctata* (Fabricius) (Anon. 1956). Harcourt et al. (1974) were the first to observe this fungus in larvae of the alfalfa weevil, *H. postica* (Gyllenhal) in Ontario. *E. phytonomi* was later reported from Missouri and Nebraska (Puttler et al. 1978). Reported here is the distribution of this fungus in Illinois in alfalfa weevil larvae.

MATERIALS AND METHODS

Alfalfa weevil larvae were collected from selected counties throughout the state. The alfalfa was searched for obviously infected larvae, which were immediately placed in petri dishes containing nonnutrient agar. Larvae that produced a conidial shower after 24 h were considered positive for *E. phytonomi*.

Large numbers of larvae also were collected from each field with sweepnets and placed in paper cartons. One hundred large larvae were selected for observation and were reared on greenhouse alfalfa in the laboratory. In many instances only one field was sampled per county.

RESULTS AND DISCUSSION

Figure 1 shows Illinois counties positive for *E. phytonomi* infecting alfalfa weevil populations. *E. phytonomi* was found as early as 8 May in the southern counties and as late as 20 June in the northern counties. The fungus was present in northern and southern bordering counties and also eastern and western, suggesting that the fungus occurred throughout the state. Absence of the disease in some counties was probably due to the timing of collection or the small sample size.

Larval mortality, as determined by sweepnet collection, ranged from 10 to 93%. This was

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1 This research has been financed in part with Federal funds from the Environmental Protection Agency under grant number L 800146; and in part by the Illinois Agricultural Experiment Station and the Illinois Natural History Survey. The contents do not necessarily reflect the views and policies of the Environmental Protection Agency nor does mention of trade or commercial products constitute endorsement or recommendation for use.

2 Illinois Natural History Survey and Illinois Agricultural Experiment Station, Urbana, IL 61801.

3 South Agland Coop, Olney, IL.
Fig. 1. Known distribution of *Entomophthora phytonomi* in *Hypera postica* from counties sampled in Illinois, 1979.

similar to the report by Harcourt et al. (1974) who observed larval mortalities as high as 90%. Harcourt et al. (1977) stated that *E. phytonomi* was the key factor responsible for the population decline of larvae from 1974 to 1976 in test plots in Ontario.

**LITERATURE CITED**


MORE ACCURATE ESTIMATORS FOR SPRUCE BUDWORM
EGG MASS DENSITY (LEPIDOPTERA: TORTRICIDAE)

Gary W. Fowler and Gary A. Simmons

ABSTRACT

Two adjusted estimators were considerably less biased and more accurate than counts not corrected for overlooked egg masses of the spruce budworm, Choristoneura fumiferana (Clemens). The two adjusted estimators compared quite favorably with the unbiased estimator where no egg masses were missed. The adjusted estimator based on a counter's percentage accuracy (efficiency) may have wider applicability than the adjusted estimator based on a counter's average difference between egg masses found and egg masses actually present.

Examination of balsam fir, Abies balsamea L., foliage for spruce budworm, Choristoneura fumiferana (Clemens), egg masses is tedious and subject to personal error. Errors include miscounting insects, forgetting the count before it is recorded, making no distinction between new and old egg masses, and overlooking egg masses (Morris 1955). Overlooking egg masses is the most common error and results in an underestimation of egg mass densities.

The percentage accuracy or efficiency in finding egg masses is related to the population level and nature of foliage (amount of defoliation, amount of new needles, straightness of needles, condition of the tree, and tree species). It varies from counter (worker) to counter, and it is not constant from plot to plot or from year to year (Morris 1955). For these reasons, Morris suggested there was little hope that a simple correction factor can be applied to eliminate checking. Correction factors would be different for different counters, plots, population levels, years, and tree species. The percentage accuracy of counters would probably be affected by prior knowledge of checking.

Governmental organizations in Canada and the United States involved with egg mass density estimation for the spruce budworm and the western budworm, Choristoneura occidentalis Freeman, were contacted to find out how they currently handle the problem of overlooking egg masses.

The following organizations do not check for egg mass counting accuracy: (1) USDA Forest Service—Regions 1, 2, 4, and 6; (2) State natural resource organizations—Idaho, Maine, Minnesota, New York, Oregon, Vermont, Washington, and Wisconsin; (3) Canadian Forestry Service—Provinces of British Columbia and Newfoundland. In the province of New Brunswick (including Nova Scotia and Prince Edward Island), the Canadian Forestry Service counts 80% of the branches twice and uses correction factors for branches not checked. In the Quebec Department of Lands and Forests, all branches are counted at least twice. In the Province of Ontario, the Canadian Forestry Service counts all branches twice and spot checks 10% of the branches a third time; no correction factors are based on the spot checks.

Present practices of egg mass density counting largely ignore the overlooking of egg masses. Correction factors based on egg mass counting accuracy have found limited use. The objective of this paper is to present two estimators based on the average accuracy of each counter involved. Both of these estimators are less biased and more accurate than the
unadjusted count or estimate. All estimators are compared using egg mass counts from a sample of balsam fir branches. Guidelines are presented for developing the adjusted estimators.

METHO DS

Data for the Study. Data for this study were obtained from two balsam fir stands in the Ottawa National Forest in Michigan's Upper Peninsula during August 1979. One stand was judged to have light defoliation and the other moderate defoliation. In each stand, five cluster centers were randomly chosen along and 10 m from a section of a secondary truck trail running through each stand. For each cluster, the five closest trees to the cluster center that were 10–20 m tall, had no top kill, and could be sampled with a pole pruner at midcrown were selected.

Five branches were clipped from the midcrown of each tree and cut back to 70 cm. Twenty-five branches were sampled from each cluster, and 125 branches were sampled from each stand. Each branch was placed in a separate plastic bag and identified as to stand, cluster, tree, branch number, and quadrant. We then counted the number of egg masses on each branch in the laboratory (Dixon et al. 1978). Each branch was cut into 5 to 15 cm-long twig segments before it was examined. The needles on which egg masses were found were removed from the branch and placed in a petri dish for count verification after the branch was completely examined. For each branch, the number of new and old egg masses, number of next year's bud tips, maximum foliated branch width, and foliated branch surface area (grid method) were determined.

Twenty branches representing the range of branch surface area and the nature of foliage encountered were checked for egg mass counting accuracy. Such checks were made without the counter knowing he was being checked and at different times during the workday. We were the two egg mass counters in this study, and each of us checked 10 branches for each other to estimate egg mass counting accuracy. The number of new egg masses found by the original counter plus those found by the checker was called the actual number of egg masses present on a branch.

Estimators with Improved Accuracy. A random sample of five of the 10 branches for each counter was taken to form an estimation set of 10 branches. The number of actual ($x_A$) and observed ($x_0$) egg masses per 1000 cm$^2$ on the five branches for each counter are shown in Table 1.

The estimator based on the observed egg mass density is biased because it underestimates actual egg mass numbers. It is given by:

$$\bar{x}_0 = \frac{1}{n}\sum_{i=1}^{n} x_{0i}$$

Table 1. Estimation data set showing the number of observed ($x_0$) and actual ($x_A$) egg masses per 1000 cm$^2$ for the five branches for each counter.

<table>
<thead>
<tr>
<th>COUNTER 1</th>
<th>COUNTER 2</th>
</tr>
</thead>
<tbody>
<tr>
<td>Branch No.</td>
<td>Egg Mass Density (No. Egg Masses/1000 cm$^2$)</td>
</tr>
<tr>
<td></td>
<td>Observed ($x_0$)</td>
</tr>
<tr>
<td>2</td>
<td>12.24</td>
</tr>
<tr>
<td>4</td>
<td>15.53</td>
</tr>
<tr>
<td>6</td>
<td>16.67</td>
</tr>
<tr>
<td>8</td>
<td>17.01</td>
</tr>
<tr>
<td>10</td>
<td>11.67</td>
</tr>
</tbody>
</table>
where \( x_{0i} \) is the number of egg masses observed per \( 1000^2 \) for the \( ith \) branch, regardless of what counter examined the \( ith \) branch. This is simply the average of the observed counts.

We examined the potential usefulness of two additional estimators which are less biased than the average of observed counts. The first proposed estimator is

\[
(2) \quad \bar{x}_1 = \frac{1}{n} \sum_{i=1}^{n} x_{1i}
\]

where \( x_{1i} = x_{0i} \cdot \hat{P}_j \) and \( \hat{P}_j \) is the proportion of accuracy of the \( jth \) counter. \( \hat{P}_1 = 0.8398 \) and \( \hat{P}_2 = 0.9154 \) from the estimation data set.

Our second proposed estimator is

\[
(3) \quad \bar{x}_2 = \frac{1}{n} \sum_{i=1}^{n} x_{2i}
\]

where \( x_{2i} = x_{0i} + \bar{D}_j \) and \( \bar{D}_j \) is the average difference between actual and observed egg mass density for the \( jth \) counter. \( \bar{D}_1 = 2.85 \) and \( \bar{D}_2 = 0.78 \) from the estimation data set.

Neither the \( \hat{P}_j \)'s nor the \( \bar{D}_j \)'s were pooled for the two counters as it was determined earlier that counter accuracy was significantly different between the two counters (t-test, \( p<0.01 \)).

The remaining 10 branches (5 for each counter) in our original sample of 20 branches were used to form a prediction data set to validate the two proposed estimators.

**RESULTS AND DISCUSSION**

**Biases of Various Estimators.** The observed \( (x_0) \), adjusted \( (x_1 \) and \( x_2) \), and actual \( (x_A) \) number of egg masses per \( 1000 \) cm\(^2\) for the five branches for each counter (prediction data set) are shown in Table 2. If the two egg mass counters never missed any egg masses when examining a branch, the unbiased estimator

\[
(4) \quad \bar{x}_A = \frac{1}{n} \sum_{i=1}^{n} x_{Ai}
\]

yields the unbiased estimate \( \bar{x}_A = 13.98 \) for \( n = 10 \) (Table 2).

The biased estimator \( x_0 \) yields the biased estimate \( \hat{x}_0 = 11.84 \) with estimated bias \( \hat{B}_0 = \bar{x}_0 - \bar{x}_A = -2.14 \).

Table 2. Prediction data set showing the observed \( (x_0) \), adjusted \( (x_1 \) and \( x_2) \), and actual \( (x_A) \) number of egg masses per \( 1000 \) cm\(^2\) for the five branches for each counter.

<table>
<thead>
<tr>
<th>COUNTER 1</th>
<th>COUNTER 2</th>
</tr>
</thead>
<tbody>
<tr>
<td><strong>Egg Mass Density</strong>&lt;br&gt;(No. Egg Masses/1000 cm(^2))&lt;br&gt;<strong>Branch No.</strong>&lt;br&gt;Observed</td>
<td><strong>Egg Mass Density</strong>&lt;br&gt;(No. Egg Masses/1000 cm(^2))&lt;br&gt;<strong>Branch No.</strong>&lt;br&gt;Observed</td>
</tr>
<tr>
<td>x(_0)</td>
<td>x(_1)</td>
</tr>
<tr>
<td>1</td>
<td>14.24</td>
</tr>
<tr>
<td>3</td>
<td>6.77</td>
</tr>
<tr>
<td>5</td>
<td>17.59</td>
</tr>
<tr>
<td>7</td>
<td>6.25</td>
</tr>
</tbody>
</table>
The two proposed estimators \( \hat{x}_1 \) and \( \hat{x}_2 \) are also biased because the adjustments (\( \hat{P}_1 \)'s and \( \hat{D}_1 \)'s) were based on sample estimates (estimation data set). However, the bias associated with these estimators is considerably less than that of \( x_0 \) making them more accurate than \( x_0 \). The biased estimator \( \hat{x}_1 \) based on adjusting \( x_0 \) by dividing by \( \hat{P}_1 \) yields the biased estimate \( \hat{x}_1 = 13.48 \) with estimated bias \( \hat{B}_1 = \hat{x}_1 - \bar{x}_A = -0.50 \). The biased estimator \( \hat{x}_2 \) based on adjusting \( x_0 \) by adding \( \hat{D}_1 \) yields the biased estimate \( \hat{x}_2 = 13.65 \) with estimated bias \( \hat{B}_2 = \hat{x}_2 - \bar{x}_A = -0.33 \).

**Efficiencies of Various Estimators.** The efficiencies of the four estimators above can be compared by looking at their sample mean square errors (Cochran 1977):

\[
\text{MSE}_{\hat{x}_0} = \frac{s_0^2}{n} + \frac{\hat{B}_0^2}{n} = \frac{18.3711}{n} + 4.6010
\]

\[
\text{MSE}_{\hat{x}_1} = \frac{s_1^2}{n} + \frac{\hat{B}_1^2}{n} = \frac{23.6368}{n} + 0.2550
\]

\[
\text{MSE}_{\hat{x}_2} = \frac{s_2^2}{n} + \frac{\hat{B}_2^2}{n} = \frac{17.8455}{n} + 0.1089
\]

\[
\text{MSE}_{\hat{x}_A} = \frac{s_A^2}{n} = \frac{26.4128}{n}
\]

\( s_0^2, s_1^2, s_2^2, s_A^2 \) are the pertinent sample variances based on the \( 10 \) \( x_0, x_1, x_2 \) and \( x_A \) values, respectively, in the prediction data set (Table 2). Notice that \( s_2^2 < s_0^2 < s_1^2 < s_A^2 \). \( x_0, x_1, x_2 \) are 0.847, 0.964 and 0.976 of \( x_A \) with relative biases (\( B/x_A \)) of \(-0.153, -0.036 \) and \(-0.024 \), respectively.

The relative efficiency (Lindgren 1962) of an estimator \( \hat{\theta}_1 \) to an estimator \( \hat{\theta}_2 \) is

\[
e(\hat{\theta}_1, \hat{\theta}_2) = \frac{\text{MSE}_{\hat{\theta}_2}}{\text{MSE}_{\hat{\theta}_1}}
\]

If \( e(\hat{\theta}_1, \hat{\theta}_2) < 1 \), \( \hat{\theta}_2 \) is a more efficient (accurate) estimator than \( \hat{\theta}_1 \) with the reverse being true when \( e(\hat{\theta}_1, \hat{\theta}_2) > 1 \).

Table 3 shows sample mean square errors for the four estimators and associated relative efficiencies for selected values of \( n \). The two proposed estimators are considerably more accurate than the estimator \( x_0 \), especially for larger sample sizes. The unbiased estimator \( \hat{x}_A \) becomes more efficient than the two proposed estimators for larger sample sizes with \( \hat{x}_2 \) comparing more favorably with \( \hat{x}_A \). Overall, both \( \hat{x}_1 \) and \( \hat{x}_2 \) are considerably more efficient than \( x_0 \) and compare quite favorably with \( \hat{x}_A \).

Once the proposed estimators have been constructed and validated with the estimation and prediction data sets, both sets should be pooled to construct the final proposed estimators. For our 20-branch sample, \( \hat{P}_1 = 0.8170 \) and \( \hat{P}_2 = 0.9095 \) for \( \hat{x}_1 \) (equation 2) and \( \hat{D}_1 = 2.74 \) and \( \hat{D}_2 = 1.22 \) for \( \hat{x}_2 \) (equation 3).

**Sample Sizes Needed for a Specified Precision.** Using the sample variance \( s^2 \), the sample size necessary to yield a 75% error bound based on Tchebycheff's Inequality (Lindgren 1962, Mendenhall et al. 1971) is:
where \( d \) is the desired error bound half-width. Using the sample variances from the prediction data set and assuming no bias for any of the estimators, \( n_0 = 8, n_1 = 11, n_2 = 8, \) and \( n_A = 12 \) for \( d = 3 \) egg masses per 1000 cm\(^2\).

To construct the two proposed estimators \( \hat{x}_1 \) and \( \hat{x}_2 \), the sampler must decide how accurately \( \hat{P}_1 \) and \( \hat{D}_1 \) need to be estimated for each counter. For our 20 branch sample, \( \hat{P}_1 = 81.70\% \) with associated variance \( s^2 = 70.7533 \) and \( \hat{P}_2 = 90.95\% \) with associated variance \( s^2 = 36.7983 \). To estimate percentage of accuracy with \( d = 2\% \) for the 75\% error bound, the desired sample size is 71 and 37 for counters 1 and 2, respectively. For our 20-branch sample, \( \hat{D}_1 = 2.74 \) with \( s^2 = 1.0776 \) and \( \hat{D}_2 = 1.22 \) with \( s^2 = 2.1332 \). To estimate average differences between observed and actual egg mass density per 1000 cm\(^2\) with \( d = 0.25 \) egg masses per 1000 cm\(^2\), the desired sample size is 69 and 137 for counters 1 and 2, respectively.

**Problems Associated with Biased Estimators.** Bias distorts probability statements about population parameters (Cochran 1977). The distortion depends on the ratio \( |B|/V(\hat{\theta}) \) where \( B \) is the bias and \( V(\hat{\theta}) \) is the variance of the biased estimator \( \hat{\theta} \). For a biased estimator with the bias unknown, the actual \( \alpha \) used to construct a \((1 - \alpha)\%\) confidence interval for the population parameter \( \Theta \), assuming normality, becomes larger than the desired nominal value \( \alpha \) and the actual confidence coefficient \((1 - \alpha)\) becomes smaller than the nominal one as \( |B|/V(\hat{\theta}) \) increases. For \( \alpha = 0.05 \), the actual value of \( \alpha \) is 0.0509, 0.0604, 0.0790, and 0.1700 for \( |B|/V(\hat{\theta}) = 0.10, 0.30, 0.50, \) and 1.00 respectively. For \( \hat{\theta}_0, \hat{\theta}_1, \) and \( \hat{\theta}_2 \) based on \( n_0 = 8, n_1 = 11, \) and \( n_2 = 8 \) observations, respectively,

\[
\frac{|\hat{\beta}_0|/s_0^2/n_0} = 1.41, \frac{|\hat{\beta}_1|/s_1^2/n_1} = 0.34, \text{ and } \frac{|\hat{\beta}_2|/s_2^2/n_2} = 0.22.
\]

\( \hat{x}_1 \) and \( \hat{x}_2 \) compare very favorably with \( \hat{x}_0 \) with relatively small probability distortion.

It is not strictly correct that two estimators that have the same MSE are equally accurate (Cochran 1977). The frequency distribution of errors \((\hat{\Theta} - \Theta)\) will not be the same for the two estimators if they have biases of different sizes. However, if \( |B|/V(\hat{\theta}) < 0.5 \), the two frequency distributions are almost identical with respect to absolute errors \(|\hat{\Theta} - \Theta|\).

**Implications of These Studies.** Spruce budworm workers should be aware that estimators of egg mass densities (not accounting for the missing of egg masses by counters) are negatively biased. This bias increases as counting accuracy decreases. Besides having a negative bias, the consequences of using such an estimator includes: (1) distortion of probability statements used to make inferences about population parameters, and (2) underestimation of optimum sample sizes needed to yield specified error bound half-widths.

Spruce budworm samplers have recognized the seriousness of missing egg masses and have suggested 50\% to 100\% checks of all egg mass counters (Morris 1955). We believe that this solution to the problem will be cost prohibitive in most situations. However, it does alleviate the problem and should be used whenever feasible.

In those cases where extensive checking is not practical, we suggest use of either esti-

**Table 3. Sample mean square errors (MSE) for estimators \( \hat{x}_0, \hat{x}_1, \hat{x}_2, \) and \( \hat{x}_A \) and associated relative efficiencies for selected values of \( n \).**

<table>
<thead>
<tr>
<th>( n )</th>
<th>( \hat{x}_0 )</th>
<th>( \hat{x}_1 )</th>
<th>( \hat{x}_2 )</th>
<th>( \hat{x}_A )</th>
<th>( e(\hat{x}_0, \hat{x}_1) )</th>
<th>( e(\hat{x}_0, \hat{x}_2) )</th>
<th>( e(\hat{x}_1, \hat{x}_2) )</th>
<th>( e(\hat{x}_A, \hat{x}_1) )</th>
<th>( e(\hat{x}_A, \hat{x}_2) )</th>
<th>( e(\hat{x}_0, \hat{x}_A) )</th>
</tr>
</thead>
<tbody>
<tr>
<td>5</td>
<td>8.28</td>
<td>4.98</td>
<td>3.68</td>
<td>5.28</td>
<td>0.60</td>
<td>0.44</td>
<td>0.74</td>
<td>0.94</td>
<td>0.70</td>
<td>0.64</td>
</tr>
<tr>
<td>10</td>
<td>6.44</td>
<td>2.62</td>
<td>1.89</td>
<td>2.64</td>
<td>0.41</td>
<td>0.29</td>
<td>0.72</td>
<td>0.99</td>
<td>0.72</td>
<td>0.41</td>
</tr>
<tr>
<td>20</td>
<td>5.52</td>
<td>1.44</td>
<td>1.00</td>
<td>1.32</td>
<td>0.26</td>
<td>0.18</td>
<td>0.70</td>
<td>1.09</td>
<td>0.76</td>
<td>0.24</td>
</tr>
<tr>
<td>50</td>
<td>4.97</td>
<td>0.73</td>
<td>0.47</td>
<td>0.53</td>
<td>0.15</td>
<td>0.09</td>
<td>0.64</td>
<td>1.38</td>
<td>0.89</td>
<td>0.11</td>
</tr>
<tr>
<td>100</td>
<td>4.78</td>
<td>0.49</td>
<td>0.29</td>
<td>0.26</td>
<td>0.10</td>
<td>0.06</td>
<td>0.58</td>
<td>1.88</td>
<td>1.12</td>
<td>0.05</td>
</tr>
</tbody>
</table>
mator based on adjusting egg mass counts by counter accuracies. Such estimators, while still being biased, will be considerably more accurate than the biased estimator not accounting for missing egg masses. Both of the proposed estimators can be based on (1) multiple linear regressions of counter accuracy on such factors as number of egg masses found, foliage surface area of branch, and the time since counter started examining foliage for each counter or (2) average counter accuracies for each counter. Even if there are no significant multiple linear regressions, the sampler may want to develop an average counter accuracy for light, moderate, and heavy infestations for each counter rather than an overall average if necessary to obtain the accuracy desired.

Guidelines for Implementation. To construct either of the proposed estimators, we suggest the following steps:

1. Determine the range of egg mass densities expected in the survey.
2. A different sample of 50–100 branches should be examined by each counter used in the survey and checked by another counter to obtain counter accuracy for each branch. To alleviate the problem of advance knowledge of checking, the sample used to determine the accuracy of each worker can be obtained by having the survey supervisor choose branches for checking as work progresses. The sample should cover the range of foliated branch surface areas, egg mass densities, foliage type, and time since the counter started examining foliage.
3. Determine if there are any significant multiple linear regression relationships of counter accuracy (accuracy proportion and average difference between observed and actual egg mass densities) on the factors listed in (2). If there are strong relationships for each counter, predictions from these equations should be used in constructing the estimators. If not, the sampler can develop separate averages for low, moderate, and high densities, or an overall average of counter accuracy for each counter. These averages can be used in constructing the estimators as was done for the 20-branch sample.
4. Randomly divide the sample of branches for each worker into an estimation set to construct the estimators and a prediction set to validate the estimators.
5. Assuming that the best estimator has been chosen and validation indicates that the desired accuracy can be obtained, pool the estimation and prediction sets for each worker and construct the final estimator based on the final counting accuracy of each counter.

In choosing between the two proposed estimators, accuracy proportion is probably more constant than differences between observed and actual egg mass densities. For this reason, the estimator based on accuracy proportion may be preferred in many cases.

The proposed estimators based on averages can yield estimates that are within 5% of the unbiased estimates with no missing egg masses. The amount of checking needed to develop these estimators will be considerably less than that needed for 50% to 100% checks. Once the estimators have been constructed, they can be continually validated over time, different stands, and different population levels as the sampler deems necessary by selecting other prediction data sets. If the desired accuracy is obtained in these data sets, they should be pooled with the data sets already collected to get more accurate estimates of each counter’s accuracy.

ACKNOWLEDGMENTS

Thanks are due to Dr. J. A. Witter, University of Michigan, for his helpful comments. We gratefully acknowledge the assistance of Mr. J. M. Maher in contacting the various natural resource organizations regarding their current procedures of handling egg mass counting accuracy. Special thanks are due to the officials of the natural resource organizations contacted who kindly gave us the information on current procedures of handling egg mass counting accuracy. Work leading to this publication was funded by a USDA Forest Service sponsored program entitled Canada/United States Spruce Budworms Program administered through the USDA Science and Education Administration (Grant 904-15-11).
This paper discusses the development of statistically reliable sampling procedures for two stages of the alfalfa blotch leafminer, *Agromyza frontella* (Rondani), a relatively new pest of alfalfa in eastern North America. For prepupae, population estimates were based on counts of the mature larvae that dropped from the alfalfa canopy into 22 x 22 cm pans containing ethylene glycol. For puparia, estimates were based on the contents of 16 x 16 cm quadrats of soil, 5 cm deep. Analysis of sampling variability showed that 40 pans and 50 quadrats per field gave adequate precision (<10% of the mean with confidence probability 80%) for population estimation of the two stages. The pattern of counts for both stages was over-dispersed, but conformed to the negative binomial distribution in 84% of the cases.

The alfalfa blotch leafminer (ABL), *Agromyza frontella* (Rondani), is a relatively new pest of alfalfa in eastern North America. Of European origin, it was first discovered on this continent in Massachusetts in 1968 (Miller and Jensen 1970). Since that time it has dispersed rapidly throughout the northeastern United States and adjacent parts of Canada and now occurs in the lower Great Lakes region on both sides of the international border.

The life history of the leafminer has been described by several authors (e.g., Bremer 1976, Hendrickson and Barth 1978). The eggs are deposited within the alfalfa leaflets, and larvae feed between the epidermal tissues, developing a characteristic blotch-type mine during the course of three instars. The mature larvae emerge from the mined leaflets and drop to the ground where they pupate just below the soil surface. There are three to four generations a year.

In 1977, a detailed study of the population dynamics of the leafminer was begun in eastern Ontario. The purpose was to develop a management strategy for the pest based on a thorough understanding of its life system and of the role of intrinsic and extrinsic factors in causing seasonal and annual changes in the species population. This is the second paper in a series dealing with sampling procedures for natural populations. The first (Harcourt and Binns 1980) reported on a sampling system for the eggs and feeding larvae. This paper gives sampling procedures for the two stages that span the subsequent period during which the insect is soil-borne: prepupae, defined here as the interval between leaf exit and soil entry, and puparia, the resting stage during which adult metamorphosis occurs.

**EXPERIMENTAL PLOTS**

The work was carried out during July and August 1977–1979, in conjunction with life table studies of the pest in two fields of vernal alfalfa at the Central Experimental Farm, Ottawa. The fields, each of which comprised 0.4 ha (1 ac) of pure stand, were situated in flat terrain.

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1Ottawa Research Station Contribution No. 594, Engineering and Statistical Research Institute Contribution No. 1-184.
2Ottawa Research Station, Agriculture Canada, Ottawa KIA OC6, Canada.
3Engineering and Statistical Research Institute, Agriculture Canada, Ottawa KIA OC6, Canada.
and contained an average of 645 stems per m\(^2\) during the course of the study. Cutting schedules were manipulated so as to avoid the catastrophic effects of harvesting on ABL populations (Harcourt and Binns 1980).

**SAMPLING METHODS**

In the three years, numbers of each stage were estimated on a total of 16 occasions. Prior to sampling, an area measuring \(30 \times 30\) m \((100 \times 100\) ft\) was marked off within the fields and divided into four blocks of equal size. These were further subdivided into four equal plots. In preparation for sampling, four sites were selected at random from within each plot, making a total of 64.

The prepupae were sampled just before harvest when mines were being vacated. The sampling device was a \(22 \times 22\) cm aluminum cake pan filled with ethylene glycol to a depth of 1 cm and placed on the ground beneath the alfalfa canopy. At sampling, the contents of each pan were poured into glass jars and brought to the laboratory for counting. A total of 6 man-minutes was required to collect and process a single sample unit.

For counts of puparia, taken just after harvest, the sample unit was a \(16 \times 16\) cm quadrat of soil, 5 cm deep. Preliminary studies showed that 93\% of the puparia occurred in this profile. The quadrats were delineated by a metal frame and the soil was removed by trowel, bagged, and brought to the laboratory for processing. The samples were first screened to remove debris and then placed in beakers of water and stirred to float out the puparia. After the puparia were skimmed off, the soil was washed through a 24 mesh screen to remove any specimens that remained. A total of 12 man-minutes was required to collect and process a single quadrat.

Numbers of the leafminer stages were recorded for each pan or quadrat, the totals for individual sample units and means per population sample (64 units) ranging as follows: pans (prepupae), totals 0–839, means 1.7–485.1; quadrats (puparia), totals 1–1062, means 20.1–424.8.

**STATISTICAL ANALYSIS**

**Detection of the spatial pattern.** Using the Fortran program of Gates & Ethridge (1972), the Poisson \((\sigma^2 = \mu)\) and negative binomial \((\sigma^2 = \mu + \mu^2/k)\) distributions were fitted to the 32 sets of data and tested by \(\chi^2\). When Poisson distributions were fitted to the observed distributions, discrepancies between observed and expected values were significant in 29 cases. However, the frequencies of all counts approximated the negative binomial series much better, and deviations between observed and expected values were significant in just five cases. Individual \(k\) values ranged from 1.47 to 54.58. Common values of \(k\) as determined by the regression method of Bliss (1958) were 12.15 for the prepupae and 3.52 for the puparia.

Variance-mean relationships for the 32 counts are illustrated in Figure 1. The over-dispersed nature of the data is clearly shown by the plotted values, which depart noticeably from the 45-degree line of Poisson expectation to attain slopes of 1.60 and 2.02 respectively for the two stages.

**Analysis of variance.** The statistical methods used in this study follow those of Harcourt and Binns (1980). The data were analyzed using a nested analysis of variance (among blocks, among plots in blocks, and among quadrats in plots). In these analyses the data were stabilized by the transformation \(\log(x + 1)\), where \(x\) is equal to the observed count. The analysis of variance is illustrated (Table 1) using one of the 32 sets of data.

Analysis of the 32 sets of data showed that variation between blocks and plots was significant in five and six cases, respectively. Although this implied that variation between sample units comprised most of the sampling variance, it also indicated that some heterogeneity occurred throughout the field. Because of this and the fact that the ratio of costs to select a site and to count the individuals at a site is small, it was deemed adequate to present the sampling of prepupae and puparia as a single stage sampling procedure. Hence, a
Fig. 1. (Left) Variance-mean relationships for 16 counts of *A. frontella* prepupae taken from pans of ethylene glycol placed beneath the alfalfa canopy. Each point plotted is based on a sample of 64 pans. (Right) Variance-mean relationships for 16 counts of *A. frontella* puparia taken from quadrats of soil. Each point plotted is based on a sample of 64 quadrats.
### Table 1. Results of analysis of variance for counts of prepupae of the alfalfa blotch leafminer, 1 September 1977

<table>
<thead>
<tr>
<th>Source of variation</th>
<th>df</th>
<th>Observed mean square</th>
<th>F</th>
</tr>
</thead>
<tbody>
<tr>
<td>Blocks</td>
<td>3</td>
<td>636.8</td>
<td>0.280</td>
</tr>
<tr>
<td>Plots within blocks</td>
<td>12</td>
<td>2272.0</td>
<td>1.508</td>
</tr>
<tr>
<td>Quadrats within plots</td>
<td>48</td>
<td>1507.1</td>
<td></td>
</tr>
</tbody>
</table>

*Transformed scale*

*sampling plan for either age interval should provide for a number of sample units well spread out over the field.*

**Sampling precision.** Table 2 lists the estimates of population density together with their standard errors in the untransformed scale. As a rule, the latter were within 10% of the mean. They exceeded it on only three occasions.

### OPTIMUM ALLOCATION OF SAMPLING RESOURCES

**Prepupae.** To avoid interpretive problems associated with presenting sampling recommendations in a transformed scale, raw data were used to investigate the sample size for predetermined confidence limits. The inter-quadrat coefficients of variation (CV) were derived as 100s/m. For prepupae, these ranged from 25 to 79 (Table 2).

For insects with populations as dense as *A. frontella* (see also Harcourt and Binns [1980]), it is reasonable to assume that mean population values will be approximately normally distributed (Snedecor and Cochran 1967). Hence, the number of sample units corresponding to a confidence interval of width p% of the mean (m) at a probability level of 1°-α is given by

\[
N_s = \left( z_\alpha \times CV/p \right)^2
\]

where CV is the percent coefficient of variation, p is the level of precision, and \(z_\alpha\) is the (1-α/2) significant value for the standard normal distribution.

Using this equation, values for \(N_s\) were obtained for confidence probabilities of 0.8 and 0.9, and a 10% level of precision (Table 2). The data were then averaged to determine the precision corresponding to certain given values of \(N_s\), 1-\(\alpha\), and CV of 45%: the latter was chosen by taking its rounded arithmetic mean over dates. Precisions corresponding to sample sizes of 30, 40, 50, and 60 quadrats, with confidence probabilities of 0.8 and 0.9 are given in Table 3. From these results, it is evident that an acceptable confidence probability (0.8) and level of precision (10%) for life table studies on ABL can be obtained with 40 pans per field. To obtain a greater or lesser degree of precision, numbers of pans should be increased or decreased accordingly.

**Puparia.** Estimates of mean density, coefficients of variation, and sampling requirements for the puparia are listed in Table 2. The coefficients of variation ranged from 34 to 75 with an arithmetic mean of 54. Using this number and pertinent values for \(z\), the precisions corresponding to four sample sizes are given in Table 3 which shows that an acceptable confidence probability and level of precision would require 50 quadrats per field.

### DISCUSSION

In determining the sample size for target insect stages, it is important to evaluate the costs of sampling. In these terms, only 240 min (= 4 h) would be required to collect and process the contents of 40 pans for prepupal larvae. To collect and process 50 quadrats of soil for puparia would require a much more substantial 600 min (= 10 h). However, where
Table 2. Estimates of the numbers of sample units required for two confidence probabilities and a 10% level of precision in sampling for two stages of the alfalfa blotch leafminer

<table>
<thead>
<tr>
<th>Sample</th>
<th>Mean number per quadrat</th>
<th>CV</th>
<th>Confidence probability</th>
</tr>
</thead>
<tbody>
<tr>
<td></td>
<td></td>
<td></td>
<td>0.80^a</td>
</tr>
<tr>
<td>Prepupae</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>1</td>
<td>316.7 ± 19.8</td>
<td>35</td>
<td>20</td>
</tr>
<tr>
<td>2</td>
<td>49.4 ± 4.3</td>
<td>49</td>
<td>39</td>
</tr>
<tr>
<td>3</td>
<td>485.1 ± 25.7</td>
<td>30</td>
<td>15</td>
</tr>
<tr>
<td>4</td>
<td>58.8 ± 4.3</td>
<td>41</td>
<td>28</td>
</tr>
<tr>
<td>5</td>
<td>18.8 ± 2.3</td>
<td>68</td>
<td>76</td>
</tr>
<tr>
<td>6</td>
<td>400.5 ± 17.6</td>
<td>25</td>
<td>10</td>
</tr>
<tr>
<td>7</td>
<td>137.3 ± 7.4</td>
<td>31</td>
<td>16</td>
</tr>
<tr>
<td>8</td>
<td>101.2 ± 5.7</td>
<td>32</td>
<td>17</td>
</tr>
<tr>
<td>9</td>
<td>70.0 ± 3.0</td>
<td>35</td>
<td>20</td>
</tr>
<tr>
<td>10</td>
<td>33.3 ± 1.9</td>
<td>45</td>
<td>33</td>
</tr>
<tr>
<td>11</td>
<td>87.4 ± 5.0</td>
<td>46</td>
<td>35</td>
</tr>
<tr>
<td>12</td>
<td>23.6 ± 1.1</td>
<td>38</td>
<td>24</td>
</tr>
<tr>
<td>13</td>
<td>33.6 ± 2.2</td>
<td>53</td>
<td>46</td>
</tr>
<tr>
<td>14</td>
<td>10.0 ± 0.8</td>
<td>66</td>
<td>72</td>
</tr>
<tr>
<td>15</td>
<td>27.0 ± 3.3</td>
<td>54</td>
<td>48</td>
</tr>
<tr>
<td>16</td>
<td>1.7 ± 0.2</td>
<td>79</td>
<td>102</td>
</tr>
<tr>
<td>Puparia</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>1</td>
<td>51.9 ± 3.7</td>
<td>40</td>
<td>26</td>
</tr>
<tr>
<td>2</td>
<td>33.6 ± 2.8</td>
<td>48</td>
<td>38</td>
</tr>
<tr>
<td>3</td>
<td>100.4 ± 7.9</td>
<td>63</td>
<td>66</td>
</tr>
<tr>
<td>4</td>
<td>57.5 ± 3.6</td>
<td>51</td>
<td>43</td>
</tr>
<tr>
<td>5</td>
<td>20.1 ± 1.8</td>
<td>70</td>
<td>80</td>
</tr>
<tr>
<td>6</td>
<td>86.0 ± 5.3</td>
<td>50</td>
<td>41</td>
</tr>
<tr>
<td>7</td>
<td>37.0 ± 1.6</td>
<td>34</td>
<td>19</td>
</tr>
<tr>
<td>8</td>
<td>274.6 ± 21.4</td>
<td>63</td>
<td>66</td>
</tr>
<tr>
<td>9</td>
<td>424.8 ± 28.1</td>
<td>53</td>
<td>46</td>
</tr>
<tr>
<td>10</td>
<td>124.1 ± 7.2</td>
<td>46</td>
<td>35</td>
</tr>
<tr>
<td>11</td>
<td>112.8 ± 4.9</td>
<td>35</td>
<td>20</td>
</tr>
<tr>
<td>12</td>
<td>221.0 ± 15.6</td>
<td>57</td>
<td>53</td>
</tr>
<tr>
<td>13</td>
<td>268.5 ± 15.5</td>
<td>46</td>
<td>35</td>
</tr>
<tr>
<td>14</td>
<td>62.5 ± 5.4</td>
<td>69</td>
<td>78</td>
</tr>
<tr>
<td>15</td>
<td>79.5 ± 6.5</td>
<td>62</td>
<td>63</td>
</tr>
<tr>
<td>16</td>
<td>100.0 ± 9.3</td>
<td>75</td>
<td>92</td>
</tr>
</tbody>
</table>

^aZ = 1.2816
^bZ = 1.6449

resources are limited a lower level of precision could be adopted for puparia, e.g. Table 3 shows that just 30 quadrats would yield a population estimate that provides a probability of 0.8 and a level of precision of 13%.

In the foregoing statistical appraisal, untransformed data were used to set the sample size for specified precision. In many cases, sampling plans based on log transformed data tend to underestimate the sampling requirement; therefore, in situations where log transformed data are appropriate, a plan based on non-transformed data should ensure adequate precision.
Table 3. Levels of precision corresponding to certain values of $N_s$ and $1-\alpha$ for two stages of the alfalfa blotch leafminer

<table>
<thead>
<tr>
<th>$N_s$</th>
<th>Confidence probability ($1-\alpha$)</th>
<th>Percent precision</th>
<th>Prepupaeb</th>
<th>Pupariae</th>
</tr>
</thead>
<tbody>
<tr>
<td>60</td>
<td>0.8</td>
<td>-</td>
<td>9</td>
<td></td>
</tr>
<tr>
<td></td>
<td>0.9</td>
<td>10</td>
<td>12</td>
<td></td>
</tr>
<tr>
<td>50</td>
<td>0.8</td>
<td>8</td>
<td>10</td>
<td></td>
</tr>
<tr>
<td></td>
<td>0.9</td>
<td>11</td>
<td>13</td>
<td></td>
</tr>
<tr>
<td>40</td>
<td>0.8</td>
<td>9</td>
<td>11</td>
<td></td>
</tr>
<tr>
<td></td>
<td>0.9</td>
<td>12</td>
<td>14</td>
<td></td>
</tr>
<tr>
<td>30</td>
<td>0.8</td>
<td>11</td>
<td>13</td>
<td></td>
</tr>
<tr>
<td></td>
<td>0.9</td>
<td>14</td>
<td>16</td>
<td></td>
</tr>
</tbody>
</table>

\[ a_p = \frac{z \cdot CV}{\sqrt{N_s}} \]
\[ bCV = 45\% \]
\[ cCV = 54\% \]

LITERATURE CITED

NEW RECORDS OF SIPHONAPTERA FROM NORTHERN MICHIGAN

William C. Scharf and Kevin R. Stewart

Since 1965 we have collected 701 fleas from birds, mammals, and their associated nests in 13 counties in both the Northern Lower and Upper peninsulas of Michigan. The results of these studies are tabulated for 27 host species and 29 flea species and subspecies. All specimens are slide mounted in the personal collections of the authors.

New Michigan records and new Lower Peninsula records are indicated in the list. These have been determined by comparison with the findings of Lawrence et al. (1965), which are the only previous records from the western Upper Peninsula and overlap our collections only in Houghton County, and Hatt et al. (1948) whose records from Trout Island, Charlevoix County constitute the only published records for the Lower Peninsula. Also, after examining all pertinent literature, university, and museum collections, A. H. Benton (in litt.; in press) considers the rest of our records not covered by the two publications cited above to be new distribution records for the counties indicated.

Noteworthy range extension records are a new southern locality for Cercopis gallinaria from Harbor Island, Lake Huron, within 8 km of the Canadian border; Hystrichopsylla dippiei recorded from a new more easterly location on Hog Island, Lake Michigan; and Atyploceros bishopi in Benzie County, more westerly than usual (Benton, in press).

Mammal names are according to Hamilton and Whitaker (1979). Bird names follow the Checklist of North American Birds (1957). Most Siphonaptera were identified using Holland (1949) and his nomenclature is followed except for Promyscopsylla which follows the revision of Johnson and Traub (1954), and the use of the names Atyploceros bishopi and Neuctropsylla genitalis genitalis which follows the accepted nomenclature of Hopkins and Rothschild (1953).

SIPHONAPTERA AND HOSTS
(* = new to Michigan, = new to Lower Peninsula)

* Ceratophyllum celsus celsus Jordan: Zonotrichia leucophrys (Forster), Leelanau.
* C. gallinae (Schrank): Passer domesticus (L.), Grand Traverse.
* C. idius Jordan and Rothschild: Iridoprocne bicolor (Vieillot) Grand Traverse, Leelanau.
* C. riparius Jordan and Rothschild: Riparia riparia (L.), Grand Traverse.
* Cediopsylla simplex (Baker): Taxidea taxus (Schreber), Grand Traverse; Sylvilagus floridanus (J. A. Allen), Grand Traverse; Leelanau, Oceana.
* Chaetopsylla lotorii (Stewart): Taxidea taxus Grand Traverse; Vulpes vulpes L., Grand Traverse, Lake.

Corrodopsylla curvata curvata (Rothchild): Sorex cinereus Kerr, Leelanau.

* Ctenocephalides canis (Curtis): Canis familiaris, Leelanau.
Ctenocephalides pseudagyrtes Baker: Scalopus aquaticus (L.), Charlevoix; Sorex cinereus, Leelanau; Blarina brevicauda (Say), Chippewa, Grand Traverse; Spermophilus tridecemlineatus (Mitchill). Antrim; Tamias striatus (L.), Leelanau; Sciurus carolinensis Gmelin, Grand Traverse; Peromyscus maniculatus gracilis LeConte, Charlevoix, Chippewa, Grand Traverse, Leelanau; Cletthromys gapperi (Vigors), Charlevoix, Leelanau; Microtus pennsylvanicus (Ord), Grand Traverse.

Doratopsylla blarinae C. Fox: Condylura cristata (L.), Grand Traverse; Blarina brevicauda, Chippewa, Grand Traverse.

1Division of Science and Mathematics, Northwestern Michigan College, Traverse City, MI 49684.
* M. quinrii (Rothchild): Peromyscus maniculatus gracilis. Chippewa.


* Stenopiaena americana (Baker): Peromyscus maniculatus gracilis. Chippewa.

ACKNOWLEDGMENTS

We thank the following persons: A. H. Benton and O. R. Larson helped with some species determinations; T. A. Allan, M. Fitz. A. Kurta, J. Mason, J. H. Rogers, E. W. Scharf. K. G. Scharf. G. W. Shugart, S. Slater, G. Stider. S. Westphal and R. A. Zillman provided host specimens from which parasites were gleaned. This paper is the culmination of a special topics course taken by Stewart at Northwestern Michigan College. Many specimens reported in this paper were collected by Scharf on class field trips supported by grants from the William R. Angell Foundation or while working on research contracts to the college by the U.S. Fish and Wildlife Service.

LITERATURE CITED


It is generally accepted that the mating activity of tiger beetles occurs most often during daylight hours. In fact, Lengerken (Zeitschr. Wiss. Zool. 135:1-162, 1929) stated that it occurs only during periods of hot sunshine. However, Shefford (J. Morph. 22:551-618, 1911) mentioned observing Cicindela tranquebarica Herbst mating and ovipositing in the laboratory on cloudy days. Willis (Univ. Kansas Sci. Bull. 47:145-313, 1967) reported seeing mounted pairs of Cicindela circumpicta (ssp. johnsoni Fitch) after dark while the soil was still warm from the day's heat.

On 25/26 July 1979, two mounted pairs of Cicindela lepida Dejean were observed by flashlight between 2430 h (25 July) and 0115 h (26 July) by Mr. Reginald Webster and myself. This activity took place in Yankee Springs Township (T3N.R10W. Sec. 13), Barry County, Michigan.

The earlier hours of the evening had been spent blacklighting and baiting for Lepidoptera. We decided to stop at a sand blowout in Section 13 to search for lepida, as this species had been taken there on previous occasions during daylight hours, and is known to be taken at light.

The air temperature had declined steadily since sunset (from 29° to 18°C), and by midnight both the air and the soil were quite cool. The soil was also damp from a 0.12 mm rainfall earlier in the evening.

The area was scouted by flashlight for beetle activity prior to setting up the blacklight. Two mating pairs and a single individual were spotted within 15 min. One of the mounted pairs was moving, but stopped when exposed by the flashlight beam. Reaching down to take them by hand caused the male to dismount, but both individuals were readily captured. A blacklight was set upon a white sheet spread on the ground. During the next half hour three more lepida were taken, two beneath the edge of the sheet, and one lurking at the shadowy edge of the lighted area.

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ENTOMOLOGICAL NOTES

THE FIRST REPORT OF THE OCCURRENCE OF BROCHYUMENA CAROLINENSIS IN MISSOURI (HEMIPTERA: PENTATOMIDAE)

The phytophagous genus Brochymena is found throughout North America and contains species and subspecies that are usually associated with trees and shrubs. Ruckel (Entomol. Amer. 26:143–238, 1946) revised the genus and included geographical ranges and host plants for the various taxa. He reported that B. carolinensis (Westwood) occurs along the eastern seaboard from New England to Florida and then west across the Gulf States to Texas. He also stated that in the northern part of its range, it did not occur west of the Appalachian Mountains although Furth (Bull. Ohio Biol. Surv. [N.S.] 5[1]:1-60, 1974) has since reported it from Ohio and Indiana. Ruckel listed species of pine and oak as food plants. He also reported that B. marginella Stål was a closely related species and definitely known only from Texas. Although he treated marginella as a valid species, Stål (Kongliga Svenska Vetenskaps-Akad. Hand. 10[4]:1-159, 1872, p. 16) was unsure of its status and R. C. Froeschner (pers. comm.) reports that specimens housed in the U.S. National Museum Collection appear to fully bridge Ruckel’s key characters to separate the two. Thus, for the purposes of this paper, I am treating marginella as a junior synonym of carolinensis.


The Texas County specimens were collected from the bark of pine (probably Pinus strobus Linnæus); late instar nymphs were observed but not collected.

The specimens collected by D. D. Kopp and M. E. Rice are deposited in the personal collection of D. B. Thomas, Department of Entomology, University of Missouri; the remainder in the University of Missouri Entomology Museum, Columbia.

ACKNOWLEDGMENTS

I wish to thank Mr. E. G. Riley, Department of Entomology, University of Missouri, Columbia, for the loan of the specimens and for the biological information, and Dr. R. C. Froeschner, U.S. National Museum, Washington, D.C., for his preliminary opinion of the status of B. marginella.

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BENTHIC MACROINVERTEBRATES FOUND ON THE FRESHWATER SPONGE SPONGILLA LACUSTRIS

James D. Matteson and Gerald Z. Jacob

ABSTRACT

Benthic macroinvertebrates were quantitatively collected from sponge and natural substrate in the Plover River, Wisconsin. A total of 37 taxa of invertebrates including several families and genera of Trichoptera, Plecoptera, Ephemeroptera, Coleoptera, and Diptera was present on sponge. Those occurring in significant numbers included Chironomidae, Hydropsyche, Cheumatopsyche, Chimarra, Baeatis, and Climaci a areolata.

Symbiotic relationships between marine and freshwater sponges and other invertebrates have been observed by Hyman (1940), Pennak (1953), and Steffan (1976). Brown (1952), Roback (1968), and Lehmkuhl (1970) have identified specific faunal parasites or predators on freshwater sponge. To our knowledge, no studies have quantitatively assessed benthic macroinvertebrates which utilize sponge as a substrate. Freshwater sponges, contributing to the standing biomass in ponds (up to 62 g wet weight m⁻² [Frost 1978]) and up to 5% of the bottom coverage in streams (Jacob, unpubl. data), could support a high density of benthic macroinvertebrates. The purpose of this study was to investigate macroinvertebrate composition and density on natural substrate and different sized sponge colonies (i.e., surface area covered and volumetric displacement).

METHODS AND MATERIALS

An area 25 m below Jordan Pond Dam on the Plover River, a tributary of the Wisconsin River, 6 km northeast of Stevens Point, Portage County, Wisconsin, was selected as the study site. This well-aerated stream section supported a large assemblage of freshwater sponge Spongilla lacustris L. on and within the interstices of rubble and boulder-sized granite rock. Physical and chemical parameters in the collection area on 20 October 1974 were: water temperature 7°C, ambient current velocity 0.5 m/sec., water depth 0.25 to 0.75 m, dissolved oxygen 15.4 mg/litre, total CaCO₃ hardness 190 mg/litre, and pH 8.5.

Specimen collections were on the following: (1) Natural substrate (230 cm²) lacking sponge but adjacent to a sponge colony (No. 2); (2) Sponge approximately 0.6 cm high covering an area of 230 cm², many developed gemmules present; (3) Sponge composite approximately 2.5 cm high covering an area of 160 cm², volumetric displacement 200 cm³; few gemmules present; (4) Sponge composite approximately 2.5 cm high covering an area of 260 cm², volumetric displacement of 350 cm³; many developed gemmules present; (5) Composite sample of several sponge colonies ranging between 0.6-3.0 cm high and 6.3-116.2 cm² with a total displacement of 500 cm³; few gemmules present.

A 6.4 mm (0.25 inch) wire mesh screen was used to measure the natural substrate area and basal area covered by each sponge colony. Macroinvertebrates and sponge, after being carefully scraped and lifted from the substrate, were preserved in 70% alcohol. Volumetric measurements of sponge were estimated by water displacement in a graduated cylinder.

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In the laboratory, the sponge was washed on a USA Standard Testing Sieve No. 80 (0.177 mm aperture). Water pressure from a faucet forced sponge tissue, silt, and fine particles through the sieve with only spicules and macroinvertebrates remaining.

*S. lacustris* was identified by the spicules which were prepared by the concentrated nitric acid method suggested for the preparation of diatoms (Patrick and Reimer 1966). Jewell (1939) reported *lacustris* as common to this and adjacent Wisconsin watersheds.

**RESULTS AND DISCUSSION**

Commensal algae, some of which gave the sponge a green color, included *Cladophora, Spirogyra, Gomphonema, Cocconeis, Navicula, Fragilaria, Cymbella, Rhopalodia*, and *Hantzschia*. It is not known whether algae were intertwined in the spicules and surface detritus or were on sponge epithelium; studies by Frost (1976) indicated the former. We also did not investigate macroinvertebrate gut contents to see if algae, sponge tissue, or both were selected as food items.

A total of 37 taxa of macroinvertebrates was found on sponge: 30 were insects representing seven orders. Eight taxa, in four orders of aquatic insects, were found on the adjacent natural substrate and were common to sponge (Table I). Sponge harbored a diverse fauna (H=2.68) as well as a high density of organisms (78,500/m² in collection No. 4).

Table 1. Taxa and numbers of benthic macroinvertebrates collected on natural substrate and sponge (*Spongilla lacustris* L.) below Jordan Pond, on the Plover River, Portage County, Wisconsin.

<table>
<thead>
<tr>
<th>Taxa</th>
<th>Collection</th>
</tr>
</thead>
<tbody>
<tr>
<td></td>
<td>natural</td>
</tr>
<tr>
<td></td>
<td>1</td>
</tr>
<tr>
<td>Trichoptera</td>
<td></td>
</tr>
<tr>
<td>Hydropsychidae</td>
<td></td>
</tr>
<tr>
<td><em>Hydropsyche</em></td>
<td>171</td>
</tr>
<tr>
<td><em>Cheumatopsyche</em></td>
<td>14</td>
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<tr>
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<td></td>
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<tr>
<td><em>Chinarra</em></td>
<td>2</td>
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<tr>
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<tr>
<td>Brachycentridae</td>
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<tr>
<td>Helicopsychidae</td>
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</tr>
<tr>
<td><em>Hydropsyche borealis</em></td>
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</tr>
<tr>
<td>Hydrotiliidae</td>
<td></td>
</tr>
<tr>
<td><em>Hydropila</em></td>
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</tr>
<tr>
<td>Molannidae</td>
<td></td>
</tr>
<tr>
<td><em>Molanna</em></td>
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</tr>
<tr>
<td>Plecoptera</td>
<td></td>
</tr>
<tr>
<td>Perlodidae</td>
<td></td>
</tr>
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<td><em>Isoperla</em></td>
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<tr>
<td>Perlidae</td>
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<td><em>Acronia</em></td>
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<tr>
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<td><em>Taeniopteryx</em></td>
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Table 1. Continued

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<th>Family</th>
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<td><em>Nemoura</em></td>
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</tr>
<tr>
<td>Pteronarcidae</td>
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<tr>
<td><em>Pteronarcys</em></td>
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<td><em>Ephemera</em></td>
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<tr>
<td>Sisyridae</td>
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<tr>
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<td><strong>Megaloptera</strong></td>
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<tr>
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<tr>
<td><em>Corydalus cornutus</em></td>
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</tr>
<tr>
<td><strong>Coleoptera</strong></td>
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</tr>
<tr>
<td>Elmidae</td>
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</tr>
<tr>
<td><em>Stenelmis</em> (adult)</td>
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</tr>
<tr>
<td><em>Stenelmis</em> (larvae)</td>
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</tr>
<tr>
<td><em>Dubitaphia</em> (adult)</td>
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<tr>
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<tr>
<td><em>Agabus</em> (adult)</td>
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<tr>
<td><em>Phylla</em></td>
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</tr>
<tr>
<td><strong>Pelecypoda</strong></td>
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<tr>
<td><em>Sphaerium</em></td>
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<tr>
<td><em>Musculium</em></td>
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Table 1. Continued

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<th>Turbellaria</th>
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<th>UC</th>
<th>UC</th>
<th>UC</th>
</tr>
</thead>
<tbody>
<tr>
<td>Oligochaeta</td>
<td>0</td>
<td>UC</td>
<td>A</td>
<td>3</td>
<td>A</td>
</tr>
</tbody>
</table>

| Total Numbers | 204 | 153 | 355 | 204 | 1477 |
| Total Taxa    | 8   | 10  | 14  | 31  | 27  |
| Diversity Index | \( H = - \sum p_i \log_2 p_i \) | 0.99 | 1.86 | 2.68 | 2.08 | 2.29 |

\( a \) Common, \( A \) abundant, \( UC \) uncommon. Relative abundance not counted.

\( b \) One other taxon present but not represented numerically.

\( c \) Three other taxa present but not represented numerically.

\( d \) Two other taxa present but not represented numerically.

Significant occurrences on sponge were noted for Trichoptera: *Hydropsyche, Cheumatopsyche*, and *Chinarra*; Ephemeroptera: *Ephemerella, Heptagenia, Steunema*, and *Baetis*; Neuroptera: *Climacia aerolaris*; Coleoptera: *Stenelmis* (L.); and Diptera: *Atherix*. A dominant group, the family Chironomidae, was not keyed further but probably included numerous species as indicated in Roback (1968). In addition, other organisms (Bryozoa, Turbellaria, and Oligochaeta) were present but not counted on the sponge.

Sponge itself may be acknowledged as being present in invertebrate surveys, but its role as a significant substrate for other benthic macroinvertebrates should not be overlooked.

ACKNOWLEDGMENT

We wish to thank Steven J. Oppenheimer for his suggestions.

LITERATURE CITED


ILLINOIS WATER MITES OF THE GENUS ARRENURUS

Joseph A. Beatty¹ and Brent D. Opell²

ABSTRACT

Distributional records for 25 species of *Arrenurus* are presented, including 11 species not previously reported from Illinois.

*Arrenurus* is the largest in number of known species of all water mite genera (Wilson 1961) and includes almost 150 described species in North America (Cook 1976). Individuals of the genus are abundant, making up, in one study (Cook 1954a), 25 percent of all water mite specimens collected. Thus they are encountered frequently by students of fresh water organisms, especially in habitats where aquatic plants are abundant.

In contrast with most other aquatic mites, the species of *Arrenurus* bear distinctive structures of sufficient size to permit their identification at relatively low magnifications, without the necessity of mounting them on slides. At present only males are identifiable in most species, principally on the basis of the secondary sexual modifications of body form. However, association of females with known males has begun (Cook 1976, Mullen 1976) and in the future determination of females may also be possible.

Studies of the genus in North America are fairly numerous, and consist principally of descriptions of new species and regional faunal studies. Some ecological and life history data have also been published (Cook 1954a; Lavers 1945; Mitchell 1959; Mullen 1976; Münchberg 1951, 1953; Wilson 1961; Smith and Oliver 1976). Among the more important references for taxonomic and distributional data are Piersig (1904), Marshall (1908, 1910, 1940, 1944), Mullen (1976), Mitchell (1954), Lavers (1945), Cook (1954a, 1954b, 1955, 1976), and Wilson (1961). The latter three authors treat the *Arrenurus* fauna of Washington, Michigan and middle Tennessee, respectively.

The *Arrenurus* fauna of Illinois is relatively little known, despite abundance of the animals and the history of extensive investigation of aquatic organisms in Illinois. Literature reports the occurrence of only 15 species of *Arrenurus* in the state (Cook, op. cit.; Marshall 1908; Wilson 1961), as compared with 63 from Michigan (Cook, op. cit.) and 31 from middle Tennessee (Wilson 1961).

In our collections of *Arrenurus* from 24 counties of southern Illinois (Fig. 1) we have taken all but one (*americanus*) of the species previously reported from the state and 13 additional species, two of them undescribed. One undescribed species from Gallatin County is similar to *manubriator* and another, found in 12 counties, is similar to *intermedius* and *marshallae*. Distributional data for the 25 named species collected are presented below. For four widespread and common species only the number of county localities is given. Collecting dates for 19 species found on more than one occasion are summarized in Figure 2. Although this figure suggests seasonal differences in species maturity, it is not the result of a quantitative sampling study and should be interpreted cautiously. The collection includes 1911 *Arrenurus* specimens, of which 967 are males. Specimens are housed in the Research Museum of Zoology, Department of Zoology, Southern Illinois University at Carbondale, except for a few in the second author’s collection.

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²Department of Biology, Virginia Polytechnic Institute and State University, Blacksburg, VA 24061.
Fig. 1. Map of southern Illinois counties. *Arrenurus* specimens are reported from all but Hardin and Pulaski counties.

ACKNOWLEDGMENT

We wish to thank David R. Cook for determining some specimens, confirming identifications of others, and for providing specimens of *A. cardiacus* and *A. intermedius*.

RECORDS OF *ARRENURUS* FROM SOUTHERN ILLINOIS
(* indicates a new Illinois record)


*apetiolatus* Piersig. 18 counties, over 200 males.

*bartonensis* Cook. Franklin Co.: Prairie Creek Lake, 1.6 km SW of Zeigler. 5 males. Jackson Co.: Lake Murphysboro State Park, 1 male; Carbondale. SIUC campus. 3 males. Carbondale, McLafferty Road, 1 male. Lawrence Co.: Sam Dale Lake State Park, 1 male. Williamson Co.: Pond E of Lakewood Park, 1 male.


*crenellatus* Marshall. Franklin Co.: Prairie Creek Lake, 1.6 km SW of Zeigler. 2 males. Jackson Co.: Carbondale, SIUC campus, 2 males.
Fig. 2. Summary of collecting dates for 19 *Arrenurus* species found on more than one occasion. Depending on its position, a black dot indicates that a species was collected during the first, middle, or last third of the month.

<table>
<thead>
<tr>
<th>JAN</th>
<th>FEB</th>
<th>MAR</th>
<th>APR</th>
<th>MAY</th>
<th>JUN</th>
<th>JUL</th>
<th>AUG</th>
<th>SEP</th>
<th>OCT</th>
<th>NOV</th>
<th>DEC</th>
</tr>
</thead>
<tbody>
<tr>
<td>ACUTUS</td>
<td>APETIOLATUS</td>
<td>BARTONENSIS</td>
<td>BIRGEI</td>
<td>GRESELLATUS</td>
<td>EXPANSUS</td>
<td>FALCICORNIS</td>
<td>FLABELLIFER</td>
<td>GENNADUS</td>
<td>INFUNDIBULARIS</td>
<td>LATICAUDATUS</td>
<td>LATICORNIS</td>
</tr>
</tbody>
</table>

*expansus* Marshall. Franklin Co.: Prairie Creek Lake, 1.6 km SW of Zeigler, 16 males. Jackson Co.: Carbondale, fish culture pond, Pleasant Hill Road, 3 males; pond 0.8 km W of Carbondale post office, 1 male.

*falcicornis* Marshall. Franklin Co.: Prairie Creek Lake, 1.6 km SW of Zeigler, 23 males.

flabellifer Marshall. Franklin Co.: Prairie Creek Lake. 1.6 km SW of Zeigler, 2 males. Jackson Co.: Carbondale, SIUC campus, 2 males; Lake Murphysboro State Park, Little Lake, 1 male.

*genadus* Cook. Franklin Co.: Prairie Creek Lake. 1.6 km SW of Zeigler, 5 males. Jackson Co.: Lake Murphysboro State Park, Little Lake, 1 male; Carbondale, SIUC farms, 1 male.

infundibularis Marshall. Franklin Co.: Prairie Creek Lake, 1.6 km SW of Zeigler, 5 males. Williamson Co.: pond E of Lakewood Park, 5 males.

laticaudatus Marshall. Franklin Co.: Prairie Creek Lake, 1.6 km SW of Zeigler, 1 male. Jackson Co.: Carbondale, pond 0.8 km W of post office, 1 male; SIUC campus, 1 male. 2.4 km S of Boskydell, 2 males. Williamson Co.: pond 0.8 km E of Lakewood Park, 1 male.

laticornis Marshall. Alexander Co.: Horseshoe Lake, 1 male. Franklin Co.: Prairie Creek Lake, 1.6 km SW of Zeigler, 3 males. Jackson Co.: Lake Murphysboro State Park, 1 male. Carbondale, SIUC campus, 2 males; SIUC farms, 1 male; Carbondale city reservoir, 1 male. Lawrence Co.: Red Hills State Park, 1 male. Pope Co.: ca 0.8 km W of Golconda, 1 male. Williamson Co.: 0.8 km NE of Lakewood Park, 1 male.

*lyriger* Marshall. Alexander Co.: Horseshoe Lake, 1 male. Franklin Co.: Prairie Creek Lake, 1.6 km SW of Zeigler, 1 male Jackson Co.: Carbondale, McLafferty Road, 1 male.

magnicaudatus Marshall. Alexander Co.: Horseshoe Lake, 1 male. Franklin Co.: Prairie Creek Lake, 1.6 km SW of Zeigler, 30 males.
major Marshall. Alexander, Franklin, Jackson, Johnson, Lawrence, Massac, St. Clair, and Williamson counties, 123 males.


marshallae Piersig. 22 counties, 225 males.


*mutchkowskii* Marshall. Franklin Co.: Prairie Creek Lake. 1.6 km SW of Zeigler. 1 male. Nov.

*neobirgei* Cook. Jackson Co.: Carbondale. SIUC campus. 6 males. Massac Co.: Mermet Lake, 1 male. Pope Co.: Lake Glendale. 1 male.

*platyrotund cuspidator* Münchberg. Franklin Co.: Prairie Creek Lake. 1.6 km SW of Zeigler, 9 males.


LITERATURE CITED


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