A Description of a New Subspecies of *Lycaena phlaeas* (Lycaenidae: Lycaeninae) from Montana, United States, With a Comparative Study of Old and New World Populations

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Abstract: The Palaearctic, Oriental and Ethiopian Region subspecies of *Lycaena phlaeas* are briefly discussed. A more detailed account of the North American subspecies is presented, and a new subspecies, *L. p. weberi*, from the Sweet Grass Hills, Montana is described. The possibility that the eastern United States subspecies *hypophlaeas* was introduced from the Old World is discussed; however no conclusion can be reached with certainty. The relationship between Old World and New World subspecies of *L. phlaeas* is discussed. Evidence presented supports the treatment of New World populations as subspecies of *L. phlaeas*.

Additional key words: Polygonaceae, *Rumex acetosella*, *R. acetosa*, *R. crispus*, *Oxyria digyna*.

INTRODUCTION

*Lycaena phlaeas* (Linnaeus, 1761) is a widespread species with subspecies in Europe, North Africa, Arabia, northern Asia, Japan, North America and tropical Africa. The nominate subspecies occurs in northern Europe (Ackery *et al.*, 1995). Shields & Montgomery (1966) mentioned that European texts list Polygonaceae (*Rumex* and *Polygonum*) as larval foodplants for *L. phlaeas* subspecies. Flight period is April to November, in one to four generations, depending on local conditions; over-wintering is in the larval stage (Tuzov, 2000). Bridges (1988) listed 19 subspecies in his catalogue, not including the North American ones. Miller and Brown (1981) listed five subspecies for North America. Ford (1924) attempted to cover the world-wide geographic races of *L. phlaeas*, but the emphasis was on the Old World taxa. He only discussed two taxa from North America, *hypophlaeas* Boisduval and *feildeni* M’Lachlan. Shields & Montgomery (1966) discussed the distribution and bionomics of *L. phlaeas* subspecies in North America, as did Ferris (1974), with the description of a new subspecies, *arctodon*. Two more recent papers also discussed taxa in *L. phlaeas*. Emmel *et al.* (1998) discussed *hypophlaeas*, with lectotype designation and type locality restriction; and Emmel & Pratt (1998) gave a new name, *alpestris*, to the California population. The Palaearctic, Oriental and Ethiopian Region subspecies will be briefly discussed below. The North American subspecies will receive a more detailed accounting and a new subspecies will be described. There has been speculation that the eastern United States populations were introduced from the Old World by human agency. There has also been speculation by some authors that the North American subspecies are not *phlaeas*, but constitute a different species. These theories will be discussed below.
This section is presented to provide information pertinent to the discussion at the end of this paper and in the hope that more light may be shed on the relationship between the Old World and New World representatives of *L. phlaeas*. It is not intended to be an extensive and complete treatise on the Old World subspecies of *L. phlaeas* and there may be unintended omissions.

**Lycaena phlaeas phlaeas** (Linnaeus, 1761). TL: Westermannia, central Sweden. The nominate subspecies is widespread and common in Europe from south of the Arctic Circle to all of the larger Mediterranean islands and island groups as well as NW Africa (Tolman & Lewington, 1997, plate. 21). Typical specimens are shown in Figs. 1-16 of this paper. Probable synonyms of *phlaeas* are *comedarum* (Grum-Grshimailo, 1890) (East Pamirs); *oxiana* (Grum-Grshimailo, 1890) (Bokhara, Pamirs); *stygianus* (Butler, 1880) (West Pamirs, Baluchistan to Chitrak and Ladak).


**Lycaena phlaeas abbottii** (Holland, 1892). TL: “Eastern Africa”. It is found in northern Malawi, Tanzania and Kenya (Ackery *et al*., 1995), and was treated as a distinct species by Kielland (1990). It is illustrated in D’Abrera (1980, p. 525).

**Lycaena phlaeas baralacha** (Moore, 1884). TL: Baralacha Pass, 4875m, Ladak. It occurs in the outer Himalayas (Kashmir-Kumaon) and Nepal (Shields, 1982).

**Lycaena phlaeas coccineus** (Ford, 1924). TL: Tian-Shan. Illustrated in Ford (1924, pl. LIV, figs. 3-4).

**Lycaena phlaeas chinensis** (C. Felder, 1862). Central China (Bridges, 1988). This subspecies is illustrated in Ford (1924, pl. LIV, figs. 10-11) and Tuzov (2000, pl. 57, figs. 28-30).

**Lycaena phlaeas daimio** Seitz, 1908. TL: Japan (Bridges, 1988). Representative specimens are shown in Figs. 25-28. Also illustrated in Ford (1924, pl. LIV, fig. 9) and Tuzov (2000, pl. 57, figs. 31-33).


**Lycaena phlaeas eleus** (Fabricius, 1798). A representative male specimen is shown in Figs. 17-18.


Figs. 1-20. Old World Lycaena phlaeas ssp. Fig. 1. L. p. phlaeas, Hagieni Forest nr. Mangalia, Romania, 6 June 1984, A. Popescu-Gorj Coll., ♂ dorsal. Fig. 2. Same, ventral. Fig. 3. Same, ♀ dorsal. Fig. 4. Same, ventral. Fig. 5. L. p. phlaeas, Skjeberg, Grimsoy, Norway, 29 July 1990, T.J. Olsen Coll., ♂ dorsal. Fig. 6. Same, ventral. Fig. 7. L. p. phlaeas, Naucelle Lespinassolle a chateau d’eau, 500m, Aveyron, France, 20 July 1990, J. Moonen Coll., ♂ dorsal. Fig. 8. Same, ventral. Fig. 9. L. p. phlaeas, Luberon, Vaucluse, France, 17 July 1983, ♀ dorsal. Fig. 10. Same, ventral. Fig. 11. L. p. phlaeas, Vergato, Italy, 3 May 1984, D. Cappelli Coll., ♂ dorsal. Fig. 12. Same, ventral. Fig. 13. L. p. phlaeas, Barcelona, Spain, 4 March 1980, ♂ dorsal. Fig. 14. Same, ventral. Fig. 15. L. p. phlaeas, Sierra Nevada, 1300m, Spain, 20 June 1988, J. Munoz Sariot Coll., ♀ dorsal. Fig. 16. Same, ventral. Fig. 17. L. p. eleus, Miseb, Malta, 26 May 1986, P. Samut Coll., ♂ dorsal. Fig. 18. Same, ventral. Fig. 19. L. p. lusitanicus, San Roque, Cadiz, Spain, 15 April, 1980, J.L. Torres Mendez Coll., ♂ dorsal. Fig. 20. Same, ventral. All figs. approximately 1.3X life size. Photos by Steve Kohler.
**Lycaena phlaeas flavens** (Ford, 1924). TL: Lhasa, Tibet. The ventral hind wings are of an even shade of lemon-yellow, a unique feature. (Ford, 1924; Bridges 1988).

**Lycaena phlaeas ganalica** P. Gorbunov, 1995. TL: Kamchatka, Russia (Tuzov, 2000).

**Lycaena phlaeas hibernica** Goodson, 1948. TL: Ireland (Bridges, 1988).

**Lycaena phlaeas hyperborea** (Ford, 1924). Arctic Norway (Bridges, 1988). This subspecies is illustrated in Ford (1924, pl. LIV, fig. 6).

**Lycaena phlaeas japonica** (Ford, 1924). TL: Japan (Bridges, 1988). This subspecies is illustrated in Ford (1924, pl. LIV, figs. 2, 16). The type is in the Tring Zoological Museum.

**Lycaena phlaeas kuriliphlaeas** (Bryk, 1942). TL: Kurile Island (Bridges, 1988).

**Lycaena phlaeas lusitanicus** (Bryk, 1940). TL: Portugal (Bridges, 1988). Representative specimens are shown in Figs. 19-22.


**Lycaena phlaeas matsumuranus** (Bryk, 1946). TL: Korea (Bridges, 1988). A representative male specimen is shown in Figs. 23-24.

**Material Studied:** KOREA: Seoul, 30 April 1986, 1♂, 3 May 1986, 1♂.

**Lycaena phlaeas phlaeoides** (Staudinger, 1901). TL: Funchal, Madeira. Found only on Madeira (Tolman & Lewington, 1997). The rich brown, somewhat mottled color and jagged whitish postmedian band of the ventral hind wing on this subspecies are distinctive. It is illustrated in Ford (1924, pl. LIV, figs. 1, 8, 20) and Tolman & Lewington (1997, pl. 21).

**Lycaena phlaeas polaris** Courvoisier, 1911. TL: Norwegian Lappland. Distribution is Arctic Fennoscandia (Tolman & Lewington, 1997). It is illustrated on their plate 21. This subspecies is distinguished by the dove grey ventral hind wing ground color and the whitish postmedian band distally bordering the postdiscal series of black spots.

**Lycaena phlaeas pseudophlaeas** (Lucas, 1866). TL: “Abyssinie”. It is found in the Highlands of Ethiopia (Ackery et al., 1995). It is illustrated in D’Abrera (1980, p. 525).

**Lycaena phlaeas shima** Gabriel, 1954. TL: Yemen: “Jebel Masnab, S. W. of Ma’bar, c. 8400 ft.” It is found in the Highlands of south-western Arabia (Saudi Arabia and Yemen) according to Ackery et al. (1995). It is illustrated in D’Abrera (1980, p. 525).

**Lycaena phlaeas sibiricanus** Kozhanchikov, 1936. TL: Siberia (Bridges, 1988).

Figs. 21-28. Old World Lycaena phlaeas ssp. Figs. 29-40. North American Lycaena phlaeas ssp. Fig. 21. L. p. lusitanicus, San Roque, Cadiz, Spain, 5 March 1982, J.L. Torres Mendez Coll., ♀ dorsal. Fig. 22. Same, ventral. Fig. 23. L. p. matsumuranus, Seoul, Korea, 3 May 1986, ♂ dorsal. Fig. 24. Same, ventral. Fig. 25. L. p. daimio, Koma, Saitama, Japan, 23 April 1977, A. Kitagawa Coll., ♂ dorsal. Fig. 26. Same, ventral. Fig. 27. Same, Iwatsuki, Dairakuteniin, Saitama, Japan, 21 April 1981, S. Ohshima Coll., ♀ dorsal. Fig. 28. Same, ventral. Fig. 29. L. p. arctodon, Beartooth Plateau, Carbon Co., Montana, U.S.A., 15 July 1989, S. Kohler Coll., ♂ dorsal. Fig. 30. Same, ventral. Fig. 31. Same, ♀ dorsal. Fig. 32. Same, ventral. Fig. 33. L. p. arethusa, Hailstone Butte, Alberta, Canada, 24 July 1980, J. Johnstone Coll. ♂ dorsal. Fig. 34. Same, ventral. Fig. 35. Same, Plateau Mountain, Alberta, Canada, 26 July 1980, N.G. Kondla Coll., ♀ dorsal. Fig. 36. Same, ventral. Fig. 37. L. p. weberi, Mount Royal, 6300-6900’, East Butte, Sweet Grass Hills, Liberty Co., Montana, U.S.A., 30 July 2004, S. Kohler Coll., holotype ♂ dorsal. Fig. 38. Same, ventral. Fig. 39. Same, 5 August 2003, allotype ♀ dorsal. Fig. 40. Same, ventral. All figs. approximately 1.3X life size. Photos by Steve Kohler.
NORTH AMERICAN SUBSPECIES

Five subspecies of *L. phlaeas* occurring in North America are recognized. A sixth from the Sweet Grass Hills of Montana is designated below. Each of the subspecies is discussed, and material examined for this study is listed. Forewing length measurements (from the junction with the thorax to the wing apex) are given in millimeters. Ferris (1974, p. 6) used a table to enumerate the differences among the named subspecies, with the characters of dorsal forewing and ventral hind wing black spots; forewing black borders; dorsal and ventral hind wing orange borders; and ground color of dorsal forewing and ventral hind wing. This table was also referred to by Emmel & Pratt (1998). The table of Ferris is reproduced with revisions here as Table 1.

*Lycaena phlaeas hypophlaeas* (Boisduval, 1852). TL: “Nord de la Californie. Il se retrouve dans tout le nord des Etats-Unis”. It was restricted by Shields (1967) to northern California. It was further restricted to vicinity of Boston, Massachusetts by Emmel et al. (1998). The lone syntype specimen is in the U.S. National Museum (Emmel et al., 1998). The name *americana* Harris, 1862 is a junior synonym of *hypophlaeas* Boisduval. This non arctic-alpine subspecies was known for many years by the name *americana*; however, the work of several authors has clarified the correct name. Shields & Montgomery (1966) gave the English translation from Boisduval’s description of *hypophlaeas* in French, which was first given by Wolley Dod (1907) as “North of California. It is found in all the northern United States”. Thus they concluded the type locality is not “California” as listed by Klots (1951) and Comstock & Huntington (1960), and said they did not know of a precise locality for *hypophlaeas*, nor where the type specimen(s) were located. Shields (1967) then said that “north of California” should instead be translated “Northern California”. He also said the probable type locality was “the Sierra Nevada Mountains, California”. Ferris (1974) accepted the reasoning by Shields, and stated, “Boisduval’s type of *hypophlaeas* is in the collection of the United States National Museum. The type was collected by J.M. Lorquin but does not bear exact locality information”. Emmel et al. (1998) questioned the likelihood of *hypophlaeas* being from California. They reasoned: (1) The lone syntype in the U. S. National Museum collection is typical of the eastern United States phenotype and does not resemble any of the high-elevation California populations of *L. phlaeas*; and (2) Even if the type specimen was purported to represent an atypical variant of a California population, it is extremely unlikely that Lorquin collected in any of the current arctic-alpine habitats of this insect. Further, they reasoned that since Boisduval was aware of the presence of this insect in the eastern United States, he undoubtedly already had material from that region and may have assumed that the species occurred in northern California, without any Lorquin specimens to support this assumption. Thus, they concluded that the name *hypophlaeas* was applicable to the *L. phlaeas* populations of the eastern U.S., but not the ones from California. They designated the sole syntype as the lectotype and restricted the type locality to the vicinity of Boston, Massachusetts, an area known to have populations with a phenotype matching the *hypophlaeas* type, and an area which was, at the time, easily accessible to collectors providing material to European lepidopterists. Thus the name *americana* Harris, 1862 becomes a junior synonym of *hypophlaeas* Boisduval.

The subspecies *hypophlaeas* is widely distributed in eastern North America. Ferris (1974) gave its range (as *americana*) as Nova Scotia and The Gaspe west through Canada to central Ontario and Minnesota, south to Virginia and montane northern Georgia, Missouri and Kansas. One historical Cass County, North Dakota record exists (Royer, 2003). It is generally rare or temporary on the Great Plains westward. Marrone (2002) reported only three widely scattered South Dakota records. Layberry et al. (1998) show a record for southern Manitoba. Hooper (1973) mentions one record near Regina, Saskatchewan. Elrod (1906) said that C. A. Wiley found it not rare at Miles City, Montana. It has also been taken in eastern Colorado near Colorado Springs (Ferris & Brown, 1981). Habitat where *hypophlaeas* is most often found is disturbed areas, including vacant lots, weedy pastures, roadsides and lake shorelines.
Klots (1951) reported the larval foodplants *Rumex acetosella* L. (Sheep Sorrel), *R. acetosa* L., and *R. crispus* L. (Curled Dock). Opler & Krizek (1984) described the life history. The pale-green eggs are laid singly on host leaves or stems. The young caterpillars chew holes in the underside of young host leaves and later make longitudinal channels. Development takes about three weeks and pupation is under leaves or rocks. Winter is spent as pupae. The caterpillars are covered with short hairs and are variably colored rose-red to green. There is a red dorsal stripe on some caterpillars. The chrysalis is light brown, tinged with pale yellow-green and spotted with black.
Allen (1997, pl. 33, p. 312) shows a photograph of the larva (as *Americana*). In the northern parts of its range, *hypophlaeas* is bivoltine (June-early July and August-September) and probably has three broods everywhere to the south (mid-April through May, mid-June through July and mid-August through September). The ground color of the dorsal forewing of *hypophlaeas* is bright coppery red-orange, not brassy or brassy-red like the arctic-alpine subspecies (Table 1). Typical adults are shown in Figs. 41-44.

Thirty-two males and 25 females were examined. Average forewing length of males was 12.3 mm, with a range of 11.5 to 14.5 mm. Average forewing length of females was 13.5 mm, with a range of 11.5 to 14.5 mm.


**Lycaena phlaeas feildeni** (M’Lachlan, 1878). TL: Grant Land, Northwest Territories, according to Miller & Brown (1981), who claimed the location of the type was unknown. However, Shields & Montgomery (1966) referencing Tite (1957), stated, “M’Lachlan (1878) described *L. p. feildeni* from two males and one female from ‘Lat. 81° 45’. The British Museum of Natural History contains these three specimens which bear the label ‘Grinell Land west side of Smith Sound, Arctic America 78-83 Lat. (81-45) Capt. Feilden R. N. 77-101’”. They also referenced Wolff (1964) stating that these were collected in 1875 or 1876. Ferris (1974) placed the type locality of *feildeni* as “Ellesmere Island, Lat. 81° 45’N”. He showed the distribution of this subspecies to be the Hayes Peninsula of western Greenland; Ellesmere Island, Banks Island, Baffin Island, Simpson Peninsula, South Hampton Island and District of Keewatin, Northwest Territories, Canada. He also stated that the insect is poorly represented in collections with the few extant specimens placed primarily in the Canadian National Collection and the Natural History Museum (London). Layberry et al. (1998) included the arctic coast of Yukon Territories and Alaska in the distribution of *feildeni*, while Ferris (1974) considered these populations undescribed. The habitat of *feildeni* is tundra and the larval foodplant is *Oxyria digyna* (L.) Hill (Mountain Sorrel) (Ferris, 1974; Layberry et al., 1998). The subspecies *feildeni* is illustrated in Shields & Montgomery (1966, figs. 1 and 2, pp. 232-233); in Ferris (1974, figs. 10-16, p. 12); in Layberry et al. (1998, pl. 10, fig. 1). There is one generation per year. The dull brassy color of the dorsal forewing with smoky washed out aspect and the very small sometimes indistinct ventral hind wing black spots characterize this subspecies (Table 1). No specimens were examined in this study.

**Lycaena phlaeas arethusa** (Wolley Dod, 1907). TL: “nr. Calgary, Alberta” in Miller & Brown (1981). Restricted to the head of Fish Creek, Alberta by Kondla (1996). After giving reasons for restricting the type locality, Kondla (1996) stated, “the locality was near Billings Lumber Mill as evidenced by label data on additional paratypes in the Canadian National Collection, collected on 19 and 20 July 1903. In a brief discussion about *L. phlaeas*, Wolley Dod (1904) stated, ‘About fifteen specimens of this were captured near the spruce bush at the head of Fish Creek in southern Alberta’”. Kondla (pers. com., 2007) has offered new information concerning the types and type locality of *arethusa*. The statement by Shields & Montgomery (1966), “The holotype and allotype are in the U. S. National Museum and six paratypes are in the Canadian National Collection” is not correct, nor is “HT in USNM” in Miller & Brown (1981). Wolley Dod in the original description stated, “Described from five males and eight females. . . . Types, ♂ and ♀ in U.S. National Museum, the rest co-types.” Wolley Dod did not designate one specimen as the name bearing type and so all extant specimens in the type series are syntypes. Also, since one of the syntypes
came from the “south fork of Sheep Creek”, then the type locality should be amended to “the head of Fish Creek and the south fork of Sheep Creek, Alberta”. Syntypes are in the U.S. National Museum and the Canadian National Collection. Layberry et al. (1998) gave the distribution of arethusa as from the Rocky Mountains of Alberta northward to Boreal Zone habitat in southern and central Yukon. A record by James Scott from 1962 (pers. corresp., Scott, 1975)—Logan Pass, Glacier National Park, Flathead/Glacier Counties, Montana is probably referable to this subspecies. Ferris (1974) in discussing the habitat and larval foodplant of arethusa according to J.A. Legge, Jr. and C.D. Bird, stated that “on Plateau Mountain south of Banff, Alberta, it flies in small grassy meadows at 8200’ in association with Oxyria digyna and Rumex alpestris (Scop.)”. The flight period is typically the first two weeks in August. The dull, red-brassy with smoky or dusky cast of the dorsal forewings in most males and the very small ventral hind wing black spots characterize this subspecies (Table 1). Typical adults are shown in Figs. 33-36. A range of phenotypic variation is shown in Fig. 49. For this study, seven males and two females were examined. Average forewing length of males was 13.1 mm, with a range of 12.5 to 14.0 mm. Average forewing length of females was 14.5 mm, with a range of 14.0 to 15.0 mm.


Lycaena phlaeas arctodon Ferris, 1974. TL: E. side Beartooth Pass, Carbon Co., Montana. The holotype is in the Allyn Museum of Entomology, now part of the McGuire Center for Lepidoptera & Biodiversity, Gainesville, Florida. Ferris (1974) gave the distribution of arctodon as “the Beartooth Plateau on the Park Co., Wyoming-Carbon Co., Montana border; the Teton Mountains, Teton Co., Wyoming; Yellowstone National Park on Mt. Washburn; and from the Lemhi Range, Lemhi Co., Idaho”. He also referred specimens from Sweet Grass Co., Montana to this subspecies, and tentatively assigned a single male phlaeas from the Wallowa Mountains, Wallowa Co., Oregon to arctodon. Warren (2005) notes that the original Oregon record was a single male from Matterhorn Mountain and that additional Oregon populations have been found in similar habitats in other parts of the high Wallowas in Wallowa County referencing Pyle (2002). Here the butterfly flies over rockslides and talus slopes above 7500’. Since the description of arctodon by Ferris in 1974, it has been collected in the Wind River Mountains, Sublette Co. and Fremont Co., Wyoming (Harry, 1981), and the Big Horn Mountains, Big Horn Co., Wyoming. New localities in Carbon, Judith Basin, Silver Bow, Gallatin and Stillwater Counties, Montana have also been documented (Fig. 56). Records also exist for the Delano Peak area, Beaver and Piute Counties, Utah (Clyde Gillette, pers. com., 2007). Subspecies arctodon is found in lush moist alpine meadow habitat near or above treeline where the presumed foodplant, Rumex acetosa is found. At the type locality the plants grow in depressions in open meadows where some moisture remains from the spring snow melt. Harry (1981) described the habitat in the Wind River Mountains, Fremont Co., Wyoming as quite different from Beartooth Pass, “Here, the butterfly lives among the rocky slopes like that preferred by Erebia magdalena. This type of habitat is typical of where Oxyria digyna exists”. Harry documented Mountain Sorrel as a larval foodplant at this location on the Bear’s Ears Trail, collecting four larvae from it, and was able to rear one to adult. Later at the same location, he obtained 38 ova from an adult female and reared them to pupa on O. digyna from the Wasatch Mountains, Utah. The suspected foodplant, R. acetosa, at the Beartooth Pass type locality has subsequently been verified by Clyde Gillette (pers. com., 2007). The subspecies closest to arctodon in appearance is arethusa, but arctodon does not have the wide dark dorsal forewing borders exhibited by arethusa nor the smoky cast of the forewings of the males. The appearance of arctodon is much brighter than arethusa, and the dorsal hind wing blue spots are also more prominent. The black spots on the ventral hind wing of arctodon are more distinct than on arethusa (Table 1). Typical adults are shown in Figs. 29-32. A range of phenotypic variation is shown in Fig. 48. Scott (1986, p. 387) applied the subspecies name polaris Courvoisier, 1911 (TL: Norwegian Lappland) to all of the western United States populations, including California, ignoring the name arctodon. This should not be followed, as polaris represents Old World populations distributed in Arctic Fennoscandia that differ from any North
American populations in having extensive whitish spaces on the ventral hind wing distally from the postdiscal series of black spots (Tolman & Lewington, 1997, pi. 21). For this study, 50 males and 29 females of *arctodon* were examined. Average forewing length of males was 12.8 mm, with a range of 11.0 to 14.5 mm. Average forewing length of females was 13.5 mm, with a range of 12.0 to 14.5 mm.


**Lycaena phlaeas alpestris** J. Emmel & Pratt, 1998. TL: north slope of Mt. Dana, 11,200–11,800’, Mono Co., California. The holotype, allotype and nine paratypes are in the collection of the Natural History Museum of Los Angeles County, California. For a long time this subspecies was known as *hypophlaeas*, but as pointed out by Emmel et al. (1998), the lectotype of *Polyommatus hypophlaeas* in the U.S. National Museum does not resemble any California specimens and appears to be a typical example of *L. phlaeas* populations of the northeastern United States (see discussion of *hypophlaeas* above). They restricted the *hypophlaeas* lectotype to eastern U.S. populations of *L. phlaeas* and sunk *mericana* Harris, the name those populations were long known as, to a junior synonym. This left the California populations of *L. phlaeas* without a name, which led to the description of *alpestris* by Emmel & Pratt (1998). The distribution of *alpestris* given by them is “the higher elevations of the Sierra Nevada from Fresno County and Inyo County on the south, north to Sonora Pass on the Tuolumne-Mono County line”. They reference Shields & Montgomery (1966); D. Bauer & K. Davenport (pers. com.). Emmel and Pratt (1998) also recently discovered a population in the White Mountains along the California-Nevada border. This subspecies flies in a single brood from mid-July to mid-August, and the larval foodplant is *O. digyna*. Emmel & Pratt (1998) referred to the table by Ferris (1974) in summarizing the distinguishing characters of *alpestris*. The dorsal forewing ground color is a pale brassy red, often with a dusky aspect. The dorsal forewing spots are prominent and well developed and the outer margin borders tend to be narrow. A typical adult male is shown in Figs. 45-46. For this study, five males were examined. Average forewing length was 13.4 mm, with a range of 11.5 to 14.5 mm.


**Lycaena phlaeas weberi** Kohler, new subspecies

During the winter of 2002-2003, Byron Weber of Missoula, Montana brought to my home a number of pinned butterfly specimens that he had collected in the area of the Sweet Grass Hills in Toole and Liberty Counties, north-central Montana. Looking through this material, I was very surprised to see two male *phlaeas* specimens that Byron had collected on the East Butte, Sweet Grass Hills, Liberty County. My experience with *L. phlaeas* in Montana prior to this had been in high elevation alpine habitats near or above timberline. Needless to say, the two large, very dusky specimens from below 7000’ elevation Canadian Zone habitat on a Prairie Island Range mountain grabbed my attention. Plans were made to return to the area to obtain more specimens and study the population, which Byron and I did in August 2003. I made two additional trips to the area in 2004 and 2005 to accumulate an adequate study series.

**Definition:** Besides the larger size, the most striking characteristic of *weberi* dorsally is the extremely dark, dusky appearance. In many males, the copper ground color of the forewing is almost completely obscured by dark brown, which often obliterates the inner margin of the dark wing border. The dusky brown is also present in many of the females, causing them to appear much more dark and dusky than any *arethusa* females. The dark border of the forewings of both males and females of *weberi* is very wide, more so than any of the other subspecies (excepting possibly *hypophlaeas*), as a percentage of total wing
length (Table 1). The pattern of dorsal forewing blackish spots is also very distinct and heavy (Figs. 37-40). Fig. 47 shows a range of phenotypic variation. Ventrally the ground color of the hind wing of weberi is a darker shade of warm gray than arethusa. This color is continued on the ventral forewing border and wing apex, where it is considerably darker than on arethusa, as well as the orange of the discal portion of the ventral forewing being brighter and more intense than on arethusa. The black spots on the ventral hind wing of weberi are as in arethusa, being very small with distal whitish edging present. However, further distally from these whitish spaces there are spaces that are darker than the rest of the gray ground color of the hind wing of weberi, forming an indistinct darker band and giving somewhat the impression of a two-toned hind wing. There is also darker gray-black in the hind wing tornus area of weberi, being fairly distinct and obvious, but only vaguely present on arethusa. The orange crenulate submarginal line on the ventral hind wing is bright, narrow and distinct on weberi (Figs. 38, 40), but is narrower and sometimes faint on arethusa (Fig. 34). The main differences between this new subspecies and the other named subspecies from North America are outlined in Table 1. Of the North American subspecies, weberi is most similar to arethusa, but larger. Forewing length of male arethusa studied averaged 13.1 mm, with a range of 12.5 to 14.0 mm, while weberi males averaged 14.5 mm, with a range of 12.5 to 15.5 mm. Forewing length of the male holotype is 15.0 mm. Forewing length of female arethusa studied averaged 14.5 mm, with a range of 14.0 to 15.0 mm, while weberi females averaged 15.2 mm, with a range of 13.5 to 16.0 mm. Forewing length of the allotype female is 15.5 mm.

**Etymology:** This subspecies is named for Byron Weber of Missoula, Montana, who discovered the population at the type locality, and whose interest in the Sweet Grass Hills and energy expended in exploring them are inspiring. Byron’s grandfather, Harry Demarest, came to the Sweet Grass Hills from Nebraska around the turn of the 20th century. He worked on ranches and hauled freight with a team of 12 horses and homesteaded in 1910 just north of the town of Whitlash. Today the ranch spreads from East Butte to Middle Butte to the original homestead. As a child and young man, Byron spent his summers on the ranch in the hay fields, but his favorite times were spent alone along the willows of Breed Creek and on the native prairie, identifying wildflowers and birds and quietly observing the mammals. In 1995, he began to seriously study the butterflies of the area and now has several drawers of pinned butterfly specimens.

**Distribution and Phenology:** To date, this subspecies is known only from the type locality (Fig. 56). It flies in a single brood from late-July to mid-August. The adults are found in close association with *Rumex acetosa*, which is the presumed larval foodplant. A preferred nectar source is *Solidago multiradiata* Ait. (Goldenrod). In late July 2004, Byron Weber and I climbed the West Butte, Sweet Grass Hills, Toole County, which is similar in elevation to the East Butte, but much more of the terrain is dominated by rockslides. We did not find weberi, nor did we find the larval foodplant. There is some controversy about whether *R. acetosa* is native to North America. Moss (1983) discussed two subspecies of *R. acetosa* L., ssp. *acetosa*—gardens and waste places, introduced; and ssp. *alpestris* (Scop.) Löve—moist banks and meadows to alpine elevations, native, more or less circumpolar, Alaska, Yukon to Wyoming. This was confirmed by Lesica (2002) discussing *R. acetosa* in Glacier National Park, Montana, “Uncommon in moist meadows and talus slopes, upper montane to alpine; East, West. Our plants are ssp. *alpestris* (Scop.) Löve. Circumboreal south to OR, WY. A closely related ssp. is introduced from Europe and grown as a garden herb”. Thompson & Kuijt (1976) reporting a study of the montane and subalpine plants of the Sweet Grass Hills stated of *R. acetosa*, “Collected from only one area, on the moist north-facing slope of Mount Royal where outcrops of Madison limestone have produced calcareous soils. Although this arctic-alpine species, native to the American Arctic, has been naturalized from Eurasia in the eastern United States, it is believed to occur as a relict in the Sweetgrass Hills rather than as a garden escapee, since it has been reported in the vicinity of Montana only from alpine or subalpine areas in Glacier Park, the Bear Paw Mountains, and the Beartooth Plateau”. They also pointed out that arctic-alpine disjunctions are often correlated with calcareous substrates, and the close association of *R. acetosa* with soils derived from limestone in East Butte suggests its persistence there as an arctic relict.

Deposition of Types: The holotype male, allotype female, two male and two female paratypes will be deposited in the Monte L. Bean Life Science Museum, Brigham Young University, Provo, Utah. Three male and seven female paratypes are in the Weber collection, and the remaining paratypes are in the Kohler collection.

Type Locality: MONTANA: Liberty County: north slope of Mount Royal, East Butte, Sweet Grass Hills, from the summit (6914’) down slope (north) to the saddle (6300’) between Mount Royal and Mount Brown. The upper part is forested with spruce (Picea glauca x engelamnnii), whitebark pine (Pinus albicaulis), limber pine (P. flexilis) and lodgepole pine (P. contorta) (Thompson & Kuijt, 1976), and is fairly steep and rocky in places, but the lower slope towards the saddle is more gentle and open and supports open lush meadow (Figs. 51-53). The north-facing aspect of the slope allows the R. acetosa to grow on the upper parts (Fig. 55). The Sweet Grass Hills in the northern part of Liberty County near the Alberta-Montana border in north-central Montana are unique in that they are the highest isolated peaks in the United States. Of volcanic origin, the Sweet Grass Hills are prominent landmarks, rising nearly 3000’ above the surrounding prairie with rolling hills extending to the north almost to the Alberta-Montana border. They are visible for more than 50 miles and consist of three distinct butte complexes with scattered grassy hills connecting them. The three buttes are West Butte (elevation 6983’, on left); Middle or Gold Butte (elevation 6512’); and East Butte (elevation 6958’, on right) with two smaller features (on far right), Grassy and Haystack Buttes (Fig. 50).

GENITALIC STUDY


There have been few comparative studies of L. phlaeas subspecies in the literature. In Russia, Gorbunov (2001) noted the apical part of the valve of chinensis (C. Felder) was wider than in subspecies phlaeas and ganalica. Ford (1924) in reviewing Ethiopian populations, abbottii (Holland), ethiopica (Poulton) and pseudophlaeas (Lucas) concluded from the genitalic descriptions of T.A. Chapman that “the genitalia of these Ethiopian forms . . . do not differ from those of H. phlaeas phlaeas save in a slight diminution in size, most noticeable in the aedeagus”, signs of geographical variation. Ford (1924) also agreed with Chapman’s conclusions regarding the genitalia of “hypophlaeas (Lapland and N. America) as specifically identical with phlaeas.”
<table>
<thead>
<tr>
<th>Subspecies</th>
<th>FW Length</th>
<th>Black Spots</th>
<th>Black Borders</th>
<th>Orange Borders - HW</th>
<th>Color</th>
</tr>
</thead>
<tbody>
<tr>
<td></td>
<td>males</td>
<td>females</td>
<td>Dorsal FW</td>
<td>Ventral HW</td>
<td>DFW Dorsal Ventral DFW VHW</td>
</tr>
<tr>
<td>hypophlaeas</td>
<td>12.3</td>
<td>13.5</td>
<td>Distinct. CS</td>
<td>Distinct. Postmedian</td>
<td>Wide, up to 15.5% of wing</td>
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<td></td>
<td></td>
<td></td>
<td>frequently</td>
<td>row distally</td>
<td>width, measured along</td>
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<tr>
<td></td>
<td></td>
<td></td>
<td>fused, CES</td>
<td>edged in white.</td>
<td>vein Cu1.</td>
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<td></td>
<td></td>
<td></td>
<td>1-1.5 mm wide.</td>
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<tr>
<td>alpestris</td>
<td>13.4</td>
<td>-</td>
<td>Distinct. CS</td>
<td>Distinct. Postmedian</td>
<td>Moderately narrow, 7.7% of</td>
</tr>
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<td></td>
<td></td>
<td></td>
<td>double or only</td>
<td>row with only</td>
<td>wing width maximum.</td>
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<td></td>
<td>slightly fused.</td>
<td>suggestion of</td>
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<td>CES 1 mm</td>
<td>white edging</td>
<td></td>
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<td></td>
<td></td>
<td></td>
<td>wide.</td>
<td>distally.</td>
<td></td>
</tr>
<tr>
<td>feildenii</td>
<td>-</td>
<td>-</td>
<td>Delicate. CS</td>
<td>Very small &amp;</td>
<td>Narrow, 7.2% of wing width</td>
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<td></td>
<td></td>
<td></td>
<td>double, small &amp;</td>
<td>sometimes</td>
<td>maximum.</td>
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<td></td>
<td></td>
<td>concave, 1 ?</td>
<td>indistinct; distal</td>
<td></td>
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<td></td>
<td></td>
<td></td>
<td>fused, CES</td>
<td>white edging</td>
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<td></td>
<td></td>
<td></td>
<td>less than 1 mm.</td>
<td>present.</td>
<td></td>
</tr>
<tr>
<td>arethusa</td>
<td>13.1</td>
<td>14.5</td>
<td>Distinct. CS</td>
<td>Very small; distal</td>
<td>Wide, up to 14.3% of wing width.</td>
</tr>
<tr>
<td></td>
<td></td>
<td></td>
<td>double &amp; distant.</td>
<td>white edging</td>
<td></td>
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<td></td>
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<td>Generally</td>
<td>present.</td>
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<td>concave out if</td>
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<td></td>
<td>fused, CES 1 mm</td>
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<tr>
<td>arctodon</td>
<td>12.8</td>
<td>13.5</td>
<td>Distinct. CS</td>
<td>Distinct; white</td>
<td>Moderately wide, up to 10.6%</td>
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<tr>
<td></td>
<td></td>
<td></td>
<td>varies; concave.</td>
<td>edging nearly</td>
<td>of wing width.</td>
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<td>inward, outward</td>
<td>obsolete in most</td>
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<td></td>
<td></td>
<td></td>
<td>or double, CES</td>
<td>specimens.</td>
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<td></td>
<td></td>
<td></td>
<td>1 mm wide.</td>
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<tr>
<td>weberi</td>
<td>14.5</td>
<td>15.2</td>
<td>Distinct &amp;</td>
<td>Very small; distal</td>
<td>Wide, up to 21.2% of wing width.</td>
</tr>
<tr>
<td></td>
<td></td>
<td></td>
<td>heavy, CS</td>
<td>white edging</td>
<td></td>
</tr>
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<td></td>
<td></td>
<td></td>
<td>sometimes</td>
<td>present, often with</td>
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<td></td>
<td></td>
<td></td>
<td>double,</td>
<td>dark spaces</td>
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<td></td>
<td></td>
<td></td>
<td>frequently</td>
<td>distally.</td>
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<td></td>
<td></td>
<td></td>
<td>fused, CES 1.5</td>
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<td></td>
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<td></td>
<td>2 mm wide.</td>
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</table>

Northern Alaska & western NWT

Northern populations are like feildenii. Specimens tend to become larger, more heavily and brightly marked in south central Alaska (McKinley National Park). Orange band on both surfaces of the hind wing becomes quite wide and very distinct in McKinley National Park specimens. At least two populations are undescribed (Meade River and Fairbanks, Alaska).

CS=Critical Spot. Character from Ford (1924). This is the spot which appears dorsally in space Cu2 of the forewings.

CES=Cell-End Spot. This is the spot at the end of the cell of the dorsal forewings.

FW=Forewing

HW=Hind Wing

DFW=Dorsal Forewing

VHW=Ventral Hind Wing
Figs. 50-55. Habitat of *Lycaena phlaeas weberi* at type locality. **Fig. 50.** Sweet Grass Hills, Montana, from the southwest, looking north toward Canada; from left, West Butte, Middle Butte (Gold Butte), East Butte, Grassy Butte, Haystack Butte. **Fig. 51.** Near the summit of Mount Royal, East Butte. **Fig. 52.** From the summit of Mount Royal looking north to the saddle and Mount Brown. **Fig. 53.** Looking back toward Mount Royal from the saddle. **Fig. 54.** *L. p. weberi* ♀ taking nectar from a preferred source, *Solidago multiradiata* (Goldenrod). **Fig. 55.** *Rumex acetosa*, presumed larval foodplant of *L. p. weberi*. Photos by Steve Kohler.
Fig. 56. Distribution of *Lycaena phlaeas* ssp. in Montana.

**DISCUSSION**

Opler & Krizek (1984) and Opler & Malikul (1992) suggested that the eastern United States population of *Lycaena phlaeas hypophlaeas* was introduced from Europe in Colonial times, reasoning that it is associated with waste places and introduced foodplants, *Rumex acetosella* (Sheep Sorrel) and occasionally *R. crispus* (Curled Dock) and it resembled European material. Other authors (Ehrlich & Ehrlich, 1961) have also put forth this theory. Layberry et al. (1998) pointed out, however, that unlike European specimens, subspecies *hypophlaeas* (as *americana*) has a pale gray rather than brown ventral hind wing, with larger more sharply defined black spots. They also stated that in Europe second-generation *phlaeas* tends to be duskier in color and have longer tails unlike subspecies *hypophlaeas*. I was not able to find illustrations or specimens of Old World *phlaeas* that completely matched *hypophlaeas* in appearance. Nominate *phlaeas* from Sweden is quite similar, as is subspecies *polaris* as figured by Tolman & Lewington (1997, pl. 21), except that *polaris* has extensive whitish spaces distally from the postmedian series of black spots on the ventral hind wing. Tuzov (2000) figures specimens on pl. 57, p. 337, from the Chita Region and Altais, Russia under the name *L. p. hypophlaeas*, both spring and summer generations. This is a very strange location for something conspecific with the eastern North American population. There are no intermediate populations. Perhaps it represents convergence rather than conspecificity (per. corresp., David Wright, 2007). From the dorsal aspect, these Russian specimens look similar to eastern United States *hypophlaeas*, and the forewings are not dusky in the 2nd generation, nor is there any evidence of long hind wing tails. Ventrally, the hind wing ground color is gray, very similar to Nearctic
hypophlaeas, though the black spots are not quite as large or distinct as in U.S. hypophlaeas. There are Old World populations of L. phlaeas that are similar-enough appearing to U.S. hypophlaeas, that this name has been applied by some authors (Tuzov, 2000), as well as by Wolly Dod (1907), who stated that “In the Staudinger Catalogue, Lapland, northern Scandinavia, Sajan-Geibel (Siberia), Amur and North America are quoted as localities for ‘var. hypophlaeas’, and some that I have bearing labels of some of those Old World localities would pass anywhere as North American specimens, amongst which there is also an occasional tendency to lose the spots, and so assimilate the typical European form”. Some of the driving force for theories that eastern North American populations were introduced is the use of the name hypophlaeas for Old World populations that has persisted through the years. Ford (1924) in discussing hypophlaeas said, “Not only does it occur throughout the Nearctic Region, but it has an extended range in Arctic Europe and Asia. There is a specimen from Siberia in the Hill Museum, Witley, and two from Amurland in the Natural History Museum, South Kensington, while Staudinger also refers to specimens from the later country. There can be little doubt that this form will ultimately be found distributed along the north coast of Asiatic and European Russia, for it is known to occur in Lapland; there is a specimen from this locality in the Tring Zoological Museum (Plate LIV, fig. 21), another in the Hill Museum, together with one labeled ‘Norway’, which although it has no other data, must almost certainly have come from the extreme north-east of that country”. Ford (1924, p. 739) then described subspecies hyperborea from arctic Norway and Lapland, saying that it was not found in Siberia or North America. He stated that specimens of hyperborea are far more frequent in collections than are Palaeartic examples of hypophlaeas, and that some confusion exists in the literature dealing with the Far Northern races of phlaeas. Although some individuals of some Old World populations of L. phlaeas are quite similar to North American hypophlaeas, none match completely the description as translated from the French [from Boisduval 1852] by Ford (1924), “Very near phlaeas, but smaller, with the spots more distinct, the wings more rounded. The under side of secondaries of an ashy whiteness, with the fulvous marginal band well marked”. I am of the opinion that the name hypophlaeas should not be used for any of the Old World populations of L. phlaeas, and that currently there is no conclusive evidence that the North American populations were introduced from Europe. Pratt & Wright (2002) presented an alternate hypothesis to an introduction, positing that the eastern North American populations of hypophlaeas existed endemically in the high elevations of the White Mountains in New England and expanded their range with the introduction of Rumex acetosella. They said, “An expansion of this sort has been observed with alpine populations of L. cupreus and L. editha. Both of these species have broadened their range with the introduction of Rumex acetosella into western North America. Also high altitude California L. phlaeas from 12,000 feet elevation can be experimentally reared on Rumex crispus at 800 feet elevation (and lower), suggesting that the species has the ability to rapidly adapt to lowland conditions. Oxyria digyna is the primary host plant of arctic-alpine L. phlaeas in North America. This plant occurs locally at high elevations on Mount Washington in New Hampshire; the possible existence of high altitude L. phlaeas colonies there and elsewhere in New England has not been studied”. If in the future, such colonies are discovered, it will certainly be a valuable key in solving the introduction question. For the present, I am not able to answer this question with certainty.

Do North American populations of Lycaena phlaeas represent a separate species? Evidence to support a single widespread phlaeas species in the Old and New World is available in the literature. Maeki & Remington (1960) showed the haploid chromosome number (n = 24) is the same for three subspecies of L. phlaeas from the Palaeartic (Japan, Finland) and the Nearctic (United States). There is at least as much adult phenotypic diversity among Old World subspecies as there is between the nominate phlaeas and New World subspecies. Even the most phenotypically divergent Old World subspecies, phlaeoides, chinensis, matsumuranus, and daimio are still generally treated as phlaeas subspecies. Genitalic studies also suggest conspecificity between Old and New World populations. Yet these facts may be inconclusive. Many lycaenid complexes have multiple species with identical genitalic and chromosome numbers (pers. com., David Wright, 2007). Keilland (1990) in his recent treatment of the three east African taxa elevated
abbottii from a subspecies of *phlaeas* to full species. The conclusion reached in the present study is that no real evidence exists to contradict the traditional placement of North American subspecies with Old World *phlaeas*. Perhaps future molecular studies will shed more light on how many species are involved.

**ACKNOWLEDGMENTS**

Appreciation is given to David Dyer, Collections Manager, University of Montana Herbarium, Missoula for access to the plant collection, and to Peter Lesica, Assistant Curator, for identification of the larval foodplants of *weberi* and *arctodon*, as well as the foodplants for many other Montana butterflies. Appreciation is also given to Clyde Gillette, Salt Lake City, Utah for information regarding the Utah, Wyoming and Montana *L. phlaeas* populations. Harry Pavulaan, David Wright and Norbert Kondla assisted with the layout and review of this paper.

Special gratitude is given to Byron Weber, Missoula, Montana for freely sharing specimens of *L. p. weberi*, as well as many other Montana butterflies, and information about the Sweet Grass Hills, and for being a pleasant companion on hikes to the summits of the East and West Buttes of the Sweet Grass Hills.

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Natural Life Histories of Alaska *Colias* (Lepidoptera: Pieridae)

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47 San Rafael Court, West Jordan, Utah, USA 84088

**Abstract:** Eight species of the genus *Colias* of Alaska were studied during the years of 2005, 2006, and 2007. Natural life histories were determined, immature stages described, measured, and photographed. Scanning Electron Microscope (SEM) pictures of the micropyle of five species are included.

**Additional key words:** boothii, thula, gigantea, impiyat, hecla, glacialis, nastes, aliaska, palaeno, chippewa, philodice, vitabunda, pelidne, canadensis, christina, kluarvensis, eggs, larvae, pupae, tundra, legume, berm

**Introduction**

The primary purpose of this research project was to determine the life histories of the 6 species of *Colias* known from the north slope of Alaska. The species studied are *C. boothii thula* Hovanitz, *C. gigantea impiyat* Harry, *C. hecla glacialis* M'Lachlan, *C. nastes aliaska* Bang-Haas, *C. palaeno chippewa* W. H. Edwards, and *C. philodice vitabunda* Hovanitz. This study also included four species of *Colias* from the interior of Alaska. These species studied are *canadensis* Ferris, *christina kluarvensis* Ferris, *g. gigantea* Strecker, *philodice vitabunda* Hovanitz. The newly discovered *C. pelidne* Boisduval and Leconte on the north slope is also mentioned.

The north slope is the drainage from the Brooks Range to the Arctic Ocean. All of the north slope is well north of the Arctic Circle and is all tundra. The study was done along the Dalton Highway and along the Trans-Alaska Pipeline at a site 92 miles (147 km) south of Deadhorse (Prudhoe Bay). The study sites are given with each species. Because of the remoteness of the site it was necessary to camp nearby. Fortunately there is a campsite in a lovely flower-covered meadow just far enough from the highway to avoid the dust from the highway and adjacent to the Sagavanirktok River. The mosquitoes and no-see-ums (biting flies) also like this meadow. A tent camp was established each year at this site. The project was started in June of 2005 and at that time it was estimated to take two years. The campsite was occupied in 2005 from June 24 to August 13, in 2006 from May 20 to July 13, and in 2007 from June 1 to July 1. By July 1 of 2007 the research was completed, so all stations were abandoned and all material retrieved.

The study area can also be described as Mile 320.5 to Mile 324.5 Dalton Hwy. There is an access road to the pipeline at the south end of the study area and another at the north end of the study area. Larvae stations were established along the pipeline at both of these access roads and also along the pipeline at a point approximately midway between the north and south study sites. The south site is located at 68°57.17'N and 148°52.49'W, and the north site is located at 69°00.61'N and 148°49.42'W at an elevation of 366 meters. This area is a flat river bottom about 3 meters higher than the river. The pipeline is raised about one meter above ground and there is a gravel berm beneath the pipeline. The legumes that some of the *Colias* use grow in abundance on this berm. They also use the legumes in the natural habitat. The berm also provides nectar sources and access to the non-legume feeders. The pipeline also provides considerable safety from the tourists. Because considerable time would be spent establishing and tending the stations, permission to use the pipeline right-of-way was obtained from Alyeska Pipeline Co. at Pump Station 3.

There was a female grizzly bear that lived near the campsite, thus slightly complicating fieldwork. In 2005 she had two new cubs and in 2006 they were still with her and just about as large as their mother. They were
occasionally observed foraging in the area. The female was known by regular caribou hunters as being very good about leaving people alone. The first day in camp in 2006 they came into camp to welcome me back. When they were discovered in camp they were only 20 feet away. When the mother observed me she just took a quick curious look and then began to wander away. Since she is so non-threatening it was probably good to have her in the area, as she probably kept the males away while she had the cubs. She was not observed in 2007.

Climate and Weather

The weather information herein is mostly temperature data, but a few remarks are made about precipitation. While in camp the daily high and low temperatures were recorded with my own thermometer. Naturally, some days the exact high or low temperatures were missed, but the temperature that was obtained would be a good representation for that day. Also at the campsite is a U.S.G.S. gauging station with a thermometer. Temperature data was obtained for the years previous to the study, 1997 to 2004. Temperature data prior to 1997 and for the year 2000 was not available. Dan Bartu with the U.S.G.S. in Fairbanks obtained the data. Seven years of data is not sufficient to establish an accurate daily average, but it does at least provide some idea of the normal temperature. Temperature data was also obtained for May and June of 2005 and the last half of August when the camp was not occupied. The U.S.G.S. thermometer was acting erratically in 2006 so temperature data for the last half of July and all of August was not obtained. A synopsis of the temperatures is given in Table 1 (page 15).

The spring and summer of 2005 were very dry. It appeared obvious that conditions were dry upon arrival at the campsite in June of 2005. Also, comments from Alyeska personnel indicated the spring had been dry. From June 25 to 29 of 2005 the weather was very warm (24-26°C) and sunny. Then on June 30 a north wind of 8-12 kts started and continued until August 2. This wind caused July to be unusually cold. The first 20 days of July were 3.6°C below average. The last ten days of July were nearly normal, then the first half of August was pleasant. Very little rain occurred during July and August.

In 2006 the temperature from 21 May to 13 July was quite normal. When I arrived at the campsite on May 20 of 2006 the snow had just melted from the flat areas. There were still minor snowdrifts beside the pipeline, etc. There was occasional snowfall until June 9. Thereafter there was much rain and limited sunshine until July 9. During the first half of July the mountains to the south (Brooks Range) were almost constantly cloud covered and there was much rain.

In 2007 the first ten days of June were sunny and exceptionally warm. During that ten-day period the daily high temperature averaged an incredible 10.1°C above average. After that there was a spell of three cool days then the weather was warm for the rest of June.

By comparison, in 1971 there was a cold spell around the first of July that lasted for a couple of days. In 1991 there was no cold spell from June 21 to July 15, and there were only two non-collecting days during that period. The primary flight and egg maturing time frame for Colias is June 20 through July. It is common that there is a cold spell (even with snow) during this period but the spells normally last only two or three days then the temperature returns to normal. The first 20 days of July 2005 were exceptionally cold which delayed egg development and caused many to die.

Eggs and Plants

In 1991 and 1999 there was an extremely good butterfly flight (all species) in the research area and in the Franklin Bluffs area. The Franklin Bluffs construction camp site was at Mile 378 Dalton Hwy, which is 37 miles (59 km) south of Deadhorse and on the coastal plain. From 2000 to 2003 the butterfly flight was moderate to poor. In 2005 and 2006 the butterfly flight at the study site was extremely poor. Except for Boloria frigga, Colias, and the blues there were almost no butterflies and these taxa were very few. It is incredible that the butterfly population could be so depauperate. In 2005 there were a few individuals flying as late as August 5 (including C. philodice and C. gigantea), this was probably because of the cool weather in July. In 2005 the flight at Franklin Bluffs was poor (C. hecla was still flying on August 10) and in 2006 it was moderate, except that Colias boothii had an abundant flight.

At the research site in 2005 there was just barely enough Colias to be able to start the research project, except boothii of which only one female was obtained. Because of the extremely warm first ten days of June in 2007 the flight of most species was very early. However, the Colias flight was only a few days early. There was an extremely abundant flight of all Satyrids and B. frigga. By June 24 there were only a few of these still flying and most of them were worn. The abundance of other taxa was still low.

We always wonder why a particular year had a poor flight, yet another year an abundant flight. Sometimes it seems obvious that weather had been a major factor, either positive or negative. Sometimes predators or parasites
seem abundant, for example, Ken Philip (pers. comm.) has observed an abundant amount of wasps during a particular year and accurately predicted a poor butterfly flight for the following year.

Short cold spells of two or three days on the north slope during July are common, so these probably have no affect on butterflies. Extended cold spells, such as during 2005, do have an adverse affect, when many eggs did not hatch. At the study sites in 2005-2007 there was an abundance of spiders. The majority of these were wolf spiders, but there were some other types as well. These "wolfies" were especially noticeable to the researcher who was paying extra attention to the ground and plants. Even more attention was paid to the larval stations and immediate area around each station. Quite commonly there were spiders on, or associated with the stations. Considering that spiders like to eat larvae, it was disgusting to observe so many spiders when trying to conduct research. It is not known if this abundance of spiders is normal, therefore it is impossible to ascertain if they have had an extra adverse affect in recent years. Some wasps, both small and large, were observed during the study period, 2005-2007. There did not seem to be an unusual abundance so they probably had no more than the usual affect. Interestingly, a few (four or five) larvae under the sleeves were parasitized by Ichneumonidae wasps. They were probably stung while resting on the sleeve. Little birds have been commonly observed in the area, but no extra abundance was observed during the study period.

I had hoped to obtain an accurate determination of how long it takes the eggs to hatch in nature. In 2005 many ova were deposited during the last few days of June. After 10 days the ova were monitored every 2 or 3 days. The majority of these ova did not hatch because the temperature remained unusually cold until July 20. All of the eggs turned orange, so they were fertile. The ova that did hatch took 3 to 4 weeks. On July 9 a hecla ovum was observed being oviposited on a plant that was growing in bare gravel. This was a warmer microhabitat than for most of the ova, so this ovum took only 2.5 weeks to hatch. Throughout the month of July, many more ova were obtained. By the amount of larvae found the next spring it appears that most of these later ova hatched.

In 2006 some boothii eggs that were laid on June 25 began hatching on July 11, so these eggs took 18 days to hatch. The temperature during this period was fairly normal yet a little below normal during the first 10 days of July. The 18 days these eggs took to hatch is probably quite normal. During a year when there is no cold spell during this period the eggs could conceivably hatch sooner.

It was assumed that the first instar could be observed and there would be evidence of eating. However, this did not turn out to be the scenario. Very few pre-diapause first instar larvae of hecla and nastes were observed. Therefore it was uncertain whether there were very many. As it turned out most of them did very little noticeable eating before diapause, however, there were exceptions. It was considered unwise to poke around in the litter and moss at the base of the plants to search for larvae in diapause since they could easily be injured. In the spring when the post-diapause larvae came out of hiding to start eating on the newly growing plant they could be observed. Since many can still be missed, it was never certain how many there were. This is also true for the second, third, and fourth instars since there was no need to poke around the plants. The number of larvae observed would vary from time to time. The number of larvae was never reliably obtained, but this did not matter since the primary purpose was to observe the larvae maturing into adults.

As stated above, the legumes that the Colias use grow in abundance along the pipeline berm. These legumes also grow commonly in natural habitat from this study site north to The Franklin Bluffs area. At the research site the legume feeding Colias fly more abundantly along the pipeline than in the natural habitat. This may be because there are more nectar source flowers along the berm. In the Franklin Bluffs area the Colias fly in the natural habitat as commonly as along the pipeline. The bushy Salix species that gigantea use grow abundantly along the pipeline berm, and in wet and semi-wet areas along side the pipeline, and in many other areas. C. gigantea females were observed ovipositing on Salix in wet areas but were never observed ovipositing (or even interested) on Salix along the pipeline berm. The Vaccinium plants that C. palaeno uses grow abundantly in semi-wet areas beside the pipeline and in natural areas from the research site to Franklin Bluffs. C. gigantea are rather difficult to catch in their natural habitat. Nectar sources (primarily yellow composites) for this species grow abundantly along the pipeline. Therefore, the pipeline provides both nectar sources and access through the preferred habitat for gigantea. The pipeline also provides habitat and/or nectar sources for all of the other butterflies that live in the area. The tent also provided a popular meeting place for a couple species of microlepdoptera.

Most of the plant species on the north slope start to dessicate for winter in early August. The Arctostaphylos alpina L. is bright red during the first half of August. By the middle of August most of the plants that the Colias use are changing color and apparently partially dessicated. Some individual plants of all the species that Colias use are still edible at the end of August. A few larvae were observed eating on August 30, these are noted with the species.
Larval Behavior

*Xolias* larvae almost entirely eat the leaves of the hostplants. Rarely one was observed eating the buds or flowers and even then there was very little eating of the buds or flowers. In general, the first through third instar larvae eat the mesophyll of the leaf and leave the membrane and veins. This varies somewhat, especially with the third instars which sometime eat the entire leaf. This also varies with the different plant species and the age of the growth. The fresh growth in the spring is eaten entirely even by the first instars. The mature leaves of *Salix* and *Vaccinium* are the most difficult to eat, so larvae smaller than fourth instar never eat the entire mature leaf.

The most common resting place for the first through third instar larvae is on the upper side of the leaf along the mid-vein. However, this varies considerably in all species. Sometimes they rest on the underside of the leaf, along the petiole, and occasionally along the stem. Fourth and fifth instar larvae that are eating *Hedysarum*, *Vaccinium*, and *Salix* stay on the plant and rest anywhere, while those eating *Astragalus* and *Oxytropis* like to rest on the ground. These *Colias* larvae overwinter in various instars. The overwintering stages are discussed with each species. There is no evidence anywhere in the world that any *Colias* overwinters as a pupa.

Very soon after the snow has melted the larvae break diapause and become visible while they are waiting for their hostplant to start growing. Some plants started growing very soon while other took a couple of weeks. At a few stations the plant never did grow, which required that the larvae be moved to a new plant. It was quite a nuisance to have to find the first instar larvae so that they could be moved. It was rather amazing how long the first instar larvae remained alive without eating. As time elapsed it became a concern that they would start to die. At three weeks time, without any plant to eat, as many larvae as could be found were moved to a new plant. Every day or two the old site would be visited and sometimes more larvae were found. At 27 days a few larvae were found still alive. Mature fifth instar larvae leave the hostplant and wander about to find a place to pupate. This is usually near the ground on a stem. Those confined under a sleeve would usually pupate on the netting near the ground.

In discussions concerning the elapsed time of development from egg to adult the following terminology is used. The first summer - this is the summer in which the egg was laid. The second summer is one year of elapsed time from egg. The third summer is two years of elapsed time from egg.

Materials and Methods

Net sleeves were placed over the larval hostplants at numerous stations within the study sites. These sleeves were to sequester the females for ovipositing on the larval hostplant and then left in place to protect the larvae. The netting was green polyester netting that BioQuip Products sells for butterfly nets. The netting was supported by two 9-gauge wires. The wires were bent in U shape so that the sleeves would fit over them, and the ends were pushed into the ground. The two wires were placed at 90° to each other to hold the sleeve in place. The bottom of the sleeve was secured by rocks. The rocks had to be placed touching each other to provide a seal to keep predators out. Since the sleeves (stations) are out in nature they are subject to being damaged or destroyed so several stations were established for each species.

The stations were checked every 2 or 3 days and reset when they had been disturbed. When it became apparent that the ova were about to hatch they would be monitored daily to find out when the ova hatched. Thereafter, the contents would be checked occasionally to monitor the progress of the larvae. In 2005 the sleeves were removed on August 30 for the winter and in 2006 the sleeves were removed on September 8. The sleeves were reset on May 24 in 2006 and on June 2 in 2007. Some of the data of the immature stages was obtained at home during the five years prior to 2005. During this project some of the data was obtained in camp and some at home. The larvae that were reared at home were put on potted plants or on cut stems that were placed in water. The potted plants were covered with net sleeves; the cut stems were placed in a plastic container or a bucket with a netting cover. When the larvae stopped eating and were preparing to molt they would be placed in a separate container (still on the leaf). First they would be measured for length and then left to molt. After molting they would be returned to the hostplant and the molted head capsule would be measured. It is rather difficult in the field to monitor the larvae to see when they are getting ready to molt. However, some of the third instar, all of the fourth and fifth instar larvae of *hecla*, *nastes*, and
*philodice* were measured in the field. The first and second instar *nastes* and *palaeno* that were used for measurements were reared in closed plastic containers in camp. *Colias* larvae that are reared in a closed container eventually die (usually third instar), so these were sacrificed.

The larvae were left under the sleeves to pupate. Pupae were collected so that they could be measured. Then the pupae were placed under a sleeve to pupate in natural conditions. Pupae not needed for measurements were put in a communal sleeve near camp for easy monitoring.

Larvae that were reared in the lab were overwintered in the refrigerator. A large plastic container, with the lid on, was used. Four small containers with water for humidity were set in the large container. The larvae were put in small plastic containers that have numerous holes and these containers were put in the large container. No plant material was put in the container with the larvae, since plant material would mold.

Morphological observations and measurements of the eggs, first instar larvae, and all the head capsules were made with the aid of a stereomicroscope and a 0.1 mm scale. Length measurements of the second instar and larger larvae and pupae were made with a mm scale and the aid of 3X reading glasses. Measurements of the head capsule width are of the molted head capsule. Measurements of the length of the larvae were made at pre-molt stage. Length of the fifth instar larva is given for the mature larva during the most common resting position. Their length during resting varies so this measurement is somewhat nebulous. N=10 for all measurements unless otherwise noted. Pupal width is the width of the pupa from side to side. Pupal height is the back to the outside edge of the wing cases.

**General Description of *Colias* Immature Stages**

**Eggs:** Fig. 30. The eggs are typical of the *Colias*. The eggs are fusiform in shape with longitudinal ribs and small transverse ridges. The top is rounded and contains the micropyle. Eggs are creamy white when oviposited and become orange with a creamy tip within 4 or 5 days in the field (2 days in the lab). Prior to hatching the eggs exhibit a black tip, the head of the larvae can be seen through the shell. Eggs hatch in 5 days at room temperature but vary in nature as mentioned above. The size is given under each species.

**Larvae:** All *Colias* have five larval instars. The first instar larvae are similar and are described here rather than repeating under each species. Any exceptions are given with the species.

**First instar:** Head is black with tiny white hairs. Body is green with dark green mid-dorsal stripe. On each side of body there are 3 white hairs on each segment except the first segment which has 5 white hairs. The lateral line is cream colored and there is a cream colored ring around the base of the hairs. Occasionally a few larvae have black hairs on the head and a few on the body. There are many black dots on a yellow body under 20X magnification. The spiracles are black, and the thoracic legs are dark green to black. There is a dark area above the anus on the majority of the larvae but not all of them.

**Second to fifth instar:** These are similar so only the fifth instar is described. The size of each instar is given with each species. The following characters are similar in all species. The head and body are green with many black spots with give rise to black or white hairs. The spiracles are white with a black ring. Sometimes the spiracles exhibit the color of the lower half of the lateral stipe. The dorsal stripe is dark green and the eyes are black. The other features or variations of these features are described with each species.

**Pupae:** Fig. 31. The pupae of all species are similar so they are described here, exceptions are given with the species. Head is green with front darker green, light green horizontal line in middle. Body is green, lighter green posterior. The dorsal stripe is dark green. The sub-dorsal stripes are light green and faint. The lateral stripes are light green or yellow and faintly expressed on wing cases. The spiracles are light green. The sub-lateral stripe is brown on three segments immediately behind wings, there is a small black spot between lateral stripe and sub-lateral on two segments immediately behind wings. The posterior end of the pupa is attached to a silk pad on the substrate and the body is held loosely to the substrate with a silk girdle.
Species Accounts

Colias boothii thula
at north slope research site
Figs. 1-13, 16, 17, 42

Eggs: Length 1.30 mm (range 1.20 to 1.40), width 0.50 mm (range 0.45 to 0.55).

Larvae:
First instar: Length 3.1 mm (range 2.9 to 3.4), head width 0.35 mm (range 0.33 to 0.36).
   Second instar: Length 5.2 mm (range 4.5 to 6.0), head width 0.58 mm (range 0.55 to 0.60).
   Third instar: Length 7.5 mm (range 7.0 to 8.6), head width 0.85 mm (range 0.80 to 0.90).
   Fourth instar: Length 14.8 mm (range 13.6 to 16.2), head width 1.50 mm (range 1.35 to 1.56).
   Fifth instar: The hairs are usually black but white hairs have been observed. Occasionally the dorsal stripe is not apparent. The sub-dorsal stripes are white with red or orange on dorsal side. There is a black patch on lateral side of sub-dorsal stripe on each segment. On some larvae these patches are not present. The lateral stripes are white with red on ventral side. Length 24 mm, head width 2.5 mm (range 2.28 to 2.80).

Pupae: Length 18.2 mm (range 17.0 to 21.0), width 5.0 mm (range 4.5 to 5.5), height 6.4 mm (range 5.5 to 7.0).

Results

C. boothii females have been observed ovipositing on Hedysarum mackenziei Richards (Fabaceae), and Astragalus arcticus Bunge (Fabaceae). The larvae were reared on mackenziei and arcticus in nature at the research site. The larvae were reared on a Thermopsis sp. (Ken Hansen), Hedysarum boreale Nuttall., Thermopsis montana Nutt. in T & G. (Fabaceae), and Astragalus cicer L., in the lab.

In 2005 only one female was collected (on Jun 25) which oviposited 40 eggs under one sleeve. On July 29 four first instar larvae were put under a second sleeve. On August 29 two second instar larvae were observed in hibernation and a third was observed still eating. They overwintered the first winter as second instar. In early June of 2006 the second station was destroyed by an animal and no larvae were found. On June 10 of 2006 eight larvae were observed, these were second and third instar. On July 10 there were 17 larvae, these were third, fourth, and fifth instar. On September 8, ten fifth instars were located; these were all on the sleeve near the ground. It was apparent that they had tried to wander away to hibernate, so had finished eating. If they had been left there after the sleeve was removed they would not have been found the next spring, so they were taken to the lab. Unfortunately, they all died during the winter. Also, on September 8 six fourth instar larvae were observed. In June of 2007 only one of these was found. This one pupated and became an adult the third summer.

In 2006 many ova were obtained from females collected in the Franklin Bluffs area. On June 2 of 2007 there were a few first instar larvae observed. On June 8 there were second, third, and one fourth instar larvae. This shows that they overwintered as first, second, and third instar. Altogether there were about 150 larvae during June. When camp was abandoned on the first of July, 142 fifth instar and two fourth instar larvae were put in containers to take to the lab. A few of these larvae had already diapaused for hibernation until the next spring. These larvae were shipped by express mail from Fairbanks, Alaska to Salt Lake City, Utah. The next spring 84 of these larvae pupated without eating. Those that did not pupate were dead or died without pupating.

Discussion

The larvae vary in that some have the black patch and some do not. The black patches are present in all of the hecla group, including johanseni (Harry, 2005), except canadensis. The larvae of boothii are the only species of the hecla group that exhibit the red along the sub-dorsal and lateral stripes. The adult phenotypes vary considerably, especially the males. Some females appear the same as some nastes females which is not unusual among Colias species.

All of the larvae exhibited a two year life span. The normal behavior is for the larvae to become fully grown sometime during the second summer. Once they become mature fifth instar they diapause, even if they are mature by July 1, until the next spring. They leave the hostplant and wander away to hibernate, and then pupate in the spring without eating.
The larvae that died in the lab were diseased. They probably became diseased during the shipment to Salt Lake City. *Colias* larvae become diseased easily when they have been in a closed container especially when there are many in a container.

*Colias gigantea inupiat*

at north slope research site

Fig. 22

**Eggs:** Length 1.42 mm (range 1.22 to 1.56), width 0.55 mm (range 0.49 to 0.61).

**Larvae:**
- **First instar:** Length 3.1 mm (range 2.7 to 3.2), head width 0.38 mm (range 0.36 to 0.40).
- **Second instar:** Length 5.0 mm (range 4.5 to 5.5), head width 0.56 mm (range 0.50 to 0.65).
- **Third instar:** Length 8.5 mm (range 7.1 to 10.5), head width 0.95 mm (range 0.78 to 1.25).
- **Fourth instar:** Length 14.5 mm (range 13.8 to 15.2) head width 1.5 mm (range 1.46 to 1.65).
- **Fifth instar:** Only black hairs have been observed except that sometimes the hairs are white on the lower half of head and body. The sub-dorsal stripes are yellow and faint. The lateral stripe is white. Length 24mm, head width 2.4 mm (range 2.28 to 2.62).

**Pupa:** Length 19.7 mm (range 18.5 to 21.0), width 5.0 mm (range 4.5 to 5.5), height 6.3 mm (range 5.5 to 7.0).

**Results**

*C. gigantea* females have been observed ovipositing on *Salix lanata* L. (Salicaceae). They probably also use other brushy willows. The larvae were reared on *S. lanata* at the research site. They were reared on *Salix exigua* Nutt. in the lab.

*C. gigantea* was the last *Colias* to start flying in 2005. This was because none had started flying before the cold spell started on June 30. From July 7 to July 15 five females were collected, which did not lay any eggs. Three females were collected on July 21; these laid only four eggs. A few more females and some eggs were obtained in the next few days. The results at the three stations that eventually had larvae are given next.

Station #36: On July 26 and 27 a few eggs were laid. On Aug 28 six larvae were observed. One of these, a second instar, was still eating. In the spring of 2006 this station was submerged and no larvae survived.

Station #37: On July 24 about 12 eggs were laid, these eggs hatched on August 10. No larvae were observed when the sleeve was removed on August 30, they were probably in hibernation in liter at the base of the plant. In the spring of 2006 the station was slightly submerged, but on June 18 four larvae (first and second instar) were observed. On July 4 four larvae (one third and 3 fourths) were observed. These were left in the sleeve when the camp was abandoned on July 13. That was unfortunate because on September 8 when the sleeve was removed, the plant, ground, and sleeve were covered with dust (from nearby road construction) so no larvae were observed.

Station #38: On August 10 of 2005 there were 12 larvae at this station. On August 30 no larvae were observed so they were in hibernation. On June 14 of 2006 three larvae (second instar) were observed. No larvae were observed on Sept. 8.

During early June of 2007 one fourth instar at Station #37 and one fourth instar at Station #38 were found. Both of these larvae became adults in late June.

On June 28 of 2007 one female was collected and about 70 eggs were obtained. These were reared in the lab under constant light and one became an adult on Aug. 9. All of the other larvae diapaused as fourth instar. During July of 2006 approximately 70 to 100 eggs were obtained in three different sleeves. In 2007 there were no larvae in these three sleeves.

**Discussion**

This taxon had the poorest results of all of the *Colias* on the north slope. There were 16 known larvae which resulted in only two larvae in 2007. Therefore all of the known larvae completed development the third summer.
**Colias hecla glacialis**

at north slope research site  
Fig. 18

**Eggs:** Length 1.35 mm (range 1.30 to 1.44), width 0.55 mm (range 0.50 to 0.57).

**Larvae:**
- **First instar:** Length 3.0 mm (range 2.8 to 3.4), head width 0.35 mm (range 0.31 to 0.38).
- **Second instar:** Length 4.7 mm (range 4.3 to 5.0), head width 0.53 mm (range 0.51 to 0.56).
- **Third instar:** Length (N=6) 7.5 mm (range 7.2 to 8.0), head width (N=7) 0.83 mm (range 0.78 to 0.88).
- **Fourth instar:** Length 13.7 mm (range 13.0 to 16.0), head width 1.33 mm (range 1.25 to 1.50).
- **Fifth instar:** Only black hairs have been observed. The sub-dorsal stripes are yellow. There is a black patch on the lateral side of sub-dorsal stripes on each segment. The lateral stripe is white. Length 24 mm, head width 2.3 mm (range 2.18 to 2.48).

**Pupae:** There are a few faint dark streaks on wing cases. Length 18.3 mm (range 17.0 to 20.0), width 5.0 mm (range 4.5 to 5.5), height 6.0 mm (range 5.2 to 6.2).

**Results**

Females have been observed ovipositing on *Astragalus arcticus* and *Hedysarum mackenziei*. Many more observations have been on *A. arcticus* than on *H. mackenziei*, so it appears that *Hedysarum* is used only occasionally.

The larvae were reared on *arcticus* and *mackenziei* at the research site and *Astragalus cicer* in the lab.

Only four females were collected in June of 2005. These females were put in two stations. Many eggs were oviposited at both stations. During July it appeared that very few, if any, of the eggs hatched. From July 15 to August 2 eight more stations were established for females to oviposit. Some of the females were taken at the research site and some at Franklin Bluffs. The next spring approximately 45 post-diapause larvae were found among all the stations. The plants at five of the stations did not grow. Larvae that could be found at these stations were moved to another station.

On August 29 of 2005 five first instar and two second instar larvae were observed. The first instars were still eating and the second instars were in hibernation. On June 12 of 2006 there were first, second, third, and fourth instar larvae found. The fourth instar may have overwintered as third instar. As late as June 18 some first, second, and third instar larvae were found at a station where the plant did not grow. It is amazing how long the first instar can survive while waiting for some food. On June 24 the first pupa was attained, and on July 11 this pupa emerged. All together there were 7 *hecla* (1m, 6f) that became adults in 2006. At one station on July 4 there were two fourth instar and on July 7 they appeared to have diapaused. During June of 2007 only three larvae were found of the 25 that were known in July of 2006. All three of these larvae became adults in June of 2007.

During early July of 2006 three new stations were established with females for oviposition. There were no eggs at one station but many eggs at the other two stations. On June 8 of 2007 there were two fourth instar and six fifth instar larvae. All of these larvae became adults in June of 2007.

**Discussion**

Some larvae had definitely overwintered as first, second, and third instar. Most of the first instars would have had plenty of time to attain second instar but apparently they wanted to remain first instars.

Of the larvae started in 2005 seven became adults in 2006 which means that they completed development in one year. All of the larvae (only 3) in 2007 became adults in June. Of the many eggs that were started in 2006 only three resulted in larvae. These three larvae became adults in 2007.

**Colias nastes alicaska**

at north slope research site  
Figs. 21, 43

**Egg:** Length 1.30 mm (range 1.21 to 1.36), width 0.53 mm (range 0.50 to 0.57).

**Larvae:**
- **First instar:** (N=6) Length 3.1 mm (range 2.7 to 3.4), head width 0.35 mm (range 0.33 to 0.38).
- **Second instar:** Length 4.5 mm (range 4.0 to 4.8), head width 0.50 mm (range 0.49 to 0.53).
- **Third instar:** Length 7.6 mm (range 6.7 to 9.0), head width 0.82 mm (range 0.75 to 0.90).
Fourth instar: Length 12.4 mm (range 9.5 to 13.8), head width 1.32 mm (range 1.12 to 1.44).

Fifth instar: The description is same as hecla. Length 23 mm, head width 2.3 mm (range 2.15 to 2.38).

Pupa: There are a few dark streaks on wing cases. Length 17.3 mm (range 16.0 to 18.0), width 5.0 mm (range 4.8 to 5.0), height 6.0 mm (range 5.8 to 6.5).

Results
Females have been observed ovipositing on *Oxytropis borealis* DC (Fabaceae). There has been no evidence that females have been interested in ovipositing on any other species of plant. Larvae were reared on *Oxytropis borealis* at the research site. In the lab first instar larvae would eat *Thermopsis montana* but did not grow and eventually died, and they would not eat *Astragalus cicer*.

In June of 2005 two stations were established with females for ovipositing. One of these stations was a potted plant. Approximately 30 ova were obtained on the potted plant and 40 to 50 ova at the other station (#15). None of the eggs on the potted plant hatched and only 6 larvae were the most ever observed at #15. More results at this station are discussed because the eggs were laid early and the larvae grew slowly. On August 28 six first instar larvae were observed. On May 23 of 2006, when the sleeve was installed, two first instar larvae were observed and they were apparently eating. On June 8 they were still first instar. On September 8 five third instar larvae were observed. In June of 2007 nine pupae were taken from this station, so there was apparently no loss of larvae at this station.

From July 15 to 25 of 2005 four more stations and one on August 2 were established for females to oviposit. Many eggs were obtained and eventually over 130 larvae. On August 29 five first instar larvae were observed in hibernation. On May 24 of 2006 one third instar larvae was observed which was already eating. On June 13 there were first, second, third, and fourth instar larvae. The first pupa was attained on June 22 and the first adult on July 9. All together there were 10 nastes (2m, 8f) that became adults in 2006.

During July of 2006 there were 134 larvae accounted for. During June of 2007 only 23 larvae were accounted for and all of these became adults in June of 2007. In early July of 2006 three new stations were established for ovipositing and many more ova were obtained. In the middle of June of 2007 55 larvae were accounted for. Only four of them did not become an adult in 2007. One of these three stations had no larvae in 2007.

Discussion
A few of the individuals (10) that were started in 2005 completed development in one year. All of the rest (that were found) completed development in two years. Nearly all of the individuals that were started in 2006 completed development in one year.

*Colias palaeo chippewa*  
at north slope research site  
Figs. 19, 31, 44, 48, 49

Egg: Length 1.37 mm (range 1.25 to 1.53), width 0.49 mm (range 0.45 to 0.54).

Larvae:
- **First instar**: Length 3.1 mm (range 3.0 to 3.3), head width 0.30 mm (range 0.28 to 0.33).
- **Second instar**: Length 5.0 mm (range 4.1 to 5.5), head width 0.53 mm (range 0.46 to 0.59).
- **Third instar**: (N=4) Length 6.9 mm (range 6.3 to 7.4), head width 0.85 mm (range 0.78 to 0.90).
- **Fourth instar**: Length 12.4 mm (range 11.2 to 13.2), head width 1.22 mm (range 1.13 to 1.31).
- **Fifth instar**: Only black hairs have been observed except that sometimes the hairs are white on lower half of head and body. There are no dorsal or sub-dorsal stripes. The lateral stripes are yellow. Length 23 mm, head width 2.1 mm (2.00 to 2.17).

Pupa: There are no sub-dorsal stripes and the sub-lateral stripes are faint. Length 17.5 mm (range 16.5 to 18.5), width 4.7 mm (range 4.5 to 5.0), height 6.0 mm (range 5.5 to 7.0).

Results
Females have been observed ovipositing on *Vaccinium uliginosum* L. (Ericaceae). The larvae at the research site were reared on *V. uliginosum*. The larvae were reared on *V. caespitosum* Michx. in the lab.

From June 29 to July 21 of 2005 five stations were established for females to oviposit and many eggs were obtained. At one station on August 28 there were some first and second instar larvae still eating. On June 17 of 2006
there were second, third, and one fourth instar larvae. In total there were approximately 110 larvae, so it appears that many _C. palaeno_ eggs hatched. This is probably because most of them were laid after the cold spell in July of 2005. On June 22 one fifth instar larva was observed. This fifth instar became an adult on July 13. Some of the third instar larvae that were observed on June 17 diapaused without becoming fourth instar.

During July of 2006 there were 130 larvae accounted for. In June of 2007 there were 47 larvae accounted for and all of these became adults in June of 2007.

On July 9 of 2006 one station was established for females to oviposit and about 30 eggs were obtained. In June of 2007 only one larva was found which became an adult on June 23.

**Discussion**

In 2006 one individual completed development in one year. The rest completed development in two years. In 2007 one individual completed development in one year, this one represented all of those that were started in 2006.

_Colias pelidne_

at north slope research site

Figs. 20, 50-55

In June of 2007 one female was collected on June 28 and one female was collected on June 29. This was only a few days before the research concluded so it was impossible to conduct life history research. The females would not oviposit on _Salix_ or _Hedysarum_. They laid about 125 eggs on _Vaccinium uliginosum_. Most of the larvae died while being transported to the lab so only eight larvae survived to diapause. They were reared on _Vac. caespitum_ and diapaused as fourth instar in the lab. Five of the larvae survived overwintering and became adults in June of 2008. The fifth instar larvae have yellow sub-dorsal stripes and white lateral stripes.

The author has collected on the north slope during 11 different years and has not collected this species previously. This species must have just recently invaded this area (global warming?).

The males of _C. pelidne_ are similar to the males of _C. gigantea_. The outer margin of the hindwing bulges out more than _C. gigantea_, this gives the hindwing a more square appearance. Also, the underside hindwing is a bit more yellow than _C. gigantea_. The females are easy to distinguish from all other species.

_Colias philodice vitabunda_

at north slope research site

Fig. 23

**Egg:** Length 1.36 mm (range 1.30 to 1.44), width 0.49 mm (range 0.47 to 0.53).

**Larvae:**

- **First instar:** Length 3.2 mm (range 3.1 to 3.4), head width 0.36 mm (range 0.32 to 0.38).
- **Second instar:** Length 4.9 mm (range 4.6 to 5.8), head width 0.55 mm (0.52 to 0.59).
- **Third instar:** Length 8.4 mm (range 7.0 to 10.3), head width 0.80 mm (range 0.73 to 0.87).
- **Fourth instar:** Length 13.3 mm (range 11.8 to 14.5), head width 1.34 mm (range 1.21 to 1.43).
- **Fifth instar:** Only white hairs have been observed. The sub-dorsal stripes are yellow and faint or not expressed. The lateral stripe is white with red on ventral side. Length 24 mm, head width 2.4 mm (range 2.25 to 2.52).

**Pupa:** There are no sub-dorsal stripes. Length 18.0 mm (range 17.0 to 18.7), width 4.7 mm (range 4.5 to 5.0), height 5.8 mm (range 5.5 to 6.5).

**Results**

Females have been observed ovipositing on _Hedysarum mackenziei_ and _Astragalus arcticus_. The larvae were reared on both species at the research site. The larvae were reared on _Medicago sativa_ L. (Fabaceae) and _Astragalus cicer_ in the lab.

On June 29 of 2005 one station was established for females to oviposit. Many eggs were obtained but only a few hatched. From July 15 to July 27 five more stations were established and many eggs were obtained and most of these eggs hatched. On August 28 and 29 five second instars were observed in hibernation. Three second instars were still eating and one third instar was still eating. On June 18 of 2006 there were second, third, fourth, and fifth instar larvae observed. On June 22 the first pupa was attained and the first adult emerged on July 9. All total there were 71 adults obtained. All of the larvae pupated and became adults except 8 that became diseased and died as fifth instar or
pupae. On July 13 two fifth instar larvae were left at the research site. When the sleeve was removed on September 8 two dead adults were found. These would have emerged in late July.

Discussion
All of the larvae that were started in 2005 became adults in 2006. Even after a late start in 2005 and a cooler than normal early part of June they completed development in one year. This is obviously the normal habit of this species on the north slope. This results in some individuals flying late in July and early August which was the case in 2005 and 2006.

Colias philodice vitabunda
at Northway

This location is the same as the C. christina location. The station was established 10 meters west of the christina station.
Egg: Length 1.25 mm (range 1.13 to 1.32), width 0.44 mm (range 0.40 to 0.47).
Larvae:
  First instar: Length 3.5 mm (range 3.10 to 3.80), head width 0.35 mm (range 0.32 to 0.38).
  Second instar: Length 5.1 mm (range 4.8 to 5.4), head width 0.55 mm (range 0.50 to 0.57).
  Third instar: Length 8.2 mm (range 7.2 to 10.8), head width 0.88 mm (range 0.79 to 0.93).
  Fourth instar: Length 15.9 mm (range 14.2 to 17.0), head width 1.58 mm (range 1.47 to 1.82).
  Fifth instar: Length 33 mm, head width 2.5 mm (range 2.28 to 2.66).
Pupa: Length 20.4 mm (range 18.5 to 21.0), width 5.3 mm (range 5.0 to 5.5), height 6.8 mm (range 6.5 to 7.0).

Results
Females have been observed ovipositing on Hedysarum mackenziei and Astragalus alpinus. Larvae at the research site were reared on H. mackenziei and in the lab were reared on H. boreale, Astragalus cicer, and Thermopsis montana. On June 17 of 2005 the station was established with two females for ovipositing. As with christina, the researcher had to travel on to the north slope. On August 30 two second instar and one third instar larvae in hibernation were observed. A FSS employee removed the sleeve for the winter at the end of September. On May 15 of 2006 the sleeve was reinstalled. At this time the plant had just barely started to grow. No larvae were observed but no effort was made to find them. On July 15 the station was revisited and 3 dead adults were found.

Discussion
It was demonstrated that C. philodice completes development in one year. Three individuals do not make a thorough scientific study but they indicate that it is normal for C. philodice to complete development in one year. Also, since C. philodice on the north slope completes development in one year it is certain that it does here in the interior where the weather is warmer.

Colias christina kluanensis
at Northway
Figs. 27-29, 45, 46

This location is at the airport at Northway which is 7 miles SW of the Alaska Highway and about 50 miles from the Yukon border. This site is the best known colony of christina in Alaska. The research station was established 25 meters N of the Flight Service Station (FSS) at the location of 62°57.72'N and 141°56.05'W and an elevation of 518 meters. This is a taiga habitat in the interior which has a longer growing season and is warmer than the north slope.

Egg: Length 1.37 mm (range 1.32 to 1.45), width 0.43 mm (range 0.40 to 0.45).
Larvae:
  First instar: Length 3.6 mm (range 3.50 to 3.80), head width 0.35 mm (range 0.32 mm to 0.39).
  Second instar: Length 5.4 mm (range 5.2 to 5.8), head width 0.55 mm (range 0.47 to 0.60).
  Third instar: Length 8.9 mm (range 8.0 to 9.8), head width 0.90 mm (range 0.80 to 0.96).
  Fourth instar: Length 16.1 mm (range 14.8 to 18.0), head width 1.41 mm (range 1.24 to 1.57).
Fifth instar: Some larvae have black hairs and some have white hairs. The sub-dorsal stripes are yellow and on some larvae they are bordered by orange. A few larvae do not express sub-dorsal stripes. The lateral stripes are white with red on ventral side. Length 33 mm, head width 2.6 mm (range 2.30 to 2.75).

Pupa: Length 22.4 mm (range 21.0 to 24.0), width 5.7 mm (range 5.0 to 6.0), height 7.0 mm (range 6.5 to 7.5).

Results

Females were observed ovipositing on *Hedysarum mackenziei* and *Astragalus alpinus*. Larvae were reared on *mackenziei* at the research site and on *H. boreale, A. cicer, and Thermopsis montana* in the lab. On June 17 of 2005 the station was established with two females for ovipositing. The researcher had to travel on to the north slope so it was not known until August 30 if there were any larvae. On August 30 three second instar larvae in hibernation were observed. At the end of September the sleeve was removed for the winter by a Federal Aviation Employee (FAA) at the FSS. On May 15 of 2006 the sleeve was replaced. At this time the plant had not begun to grow. One larva was observed which was waiting for food. The station was revisited on 15 July and three dead *christina* adults were found.

Discussion

The larvae that were used for measurements were reared in the lab during 1999, 2000, and 2007. It was demonstrated that *C. christina* completes development in one year. Three individuals do not make a thorough scientific study but they indicate that it is normal for *C. christina* to complete development in one year.

Colias christina X canadensis

at Northway

Figs. 14, 15

On June 10 of 1999 a female *C. canadensis* was collected at this location. Eggs were obtained from this female and the larvae were reared to diapause by Ken Hansen. The post diapause larvae were reared by Ken Hansen, Jacque Wolfe, and J. Harry. 16 adults were obtained and all of these adults display characters that demonstrate they are hybrids of *C. christina* and *C. canadensis*. The fifth instar larvae and adults are described.

Fifth instar: Like christina with red along the stripes.

Adults, males: Upper surfaces ground color is orange with the orange completely covering the wing as in *canadensis*. The dark border is wide as in *C. christina*. Undersurfaces appear to be a combination of the two species. Size varies from *C. canadensis* to *C. christina*. The males that are the size of *C. christina* are the largest all orange *Colias* from Alaska, since the orange does not cover the entire surface of *christina*.

Adults, female: Variable like *C. canadensis* and *C. christina* but large like *C. christina*. Undersurfaces appear to be a combination of the two species.

Colias gigantea gigantea

at Northway

Figs. 39-41, 47

Egg: Length 1.44 mm (range 1.32 to 1.65), width 0.49 mm (range 0.46 to 0.53). The SEM pictures of the micropyle demonstrate that different eggs of the same species have a different number of cells around the micropyle.

Larvae:

First instar: Length 3.6 mm (range 3.5 to 3.8), head width 0.37 mm (range 0.35 to 0.38).

Second instar: Length 5.9 mm (range 5.2 to 6.5), head width 0.59 mm (range 0.57 to 0.61).

Third instar: Length 9.3 mm (range 8.6 to 11.8), head width 0.98 mm (range 0.88 to 1.18).

Fourth instar: Length 16.5 mm (range 14.3 to 19.0), head width 1.46 mm (range 1.24 to 1.78).

Fifth instar: Only white hairs on head and body have been observed. The sub-dorsal stripes are yellow. The lateral stripes are white with red on ventral side. Length 33 mm, head width 2.7 mm (range 2.30 to 2.84).

Pupa: Length 23.3 mm (range 22.0 to 25.0), width 6.0 mm (range 5.5 to 6.5), height 7.0 mm (range 6.0 to 8.0).

Results

Females were observed ovipositing on a *Salix* sp. Larvae were reared on *Salix exigua* in the lab.
Discussion
The natural life history was not determined in interior Alaska. The larvae that were used for measurements were reared in 2003 and 2007.

*Colias canadensis*

at Northway

Fig. 26

**Egg:** Length 1.30 mm (range 1.20 to 1.40), width 0.51 mm (range 0.48 to 0.60).

**Larvae:**
- **First instar:** Length 3.3 mm (range 2.9 to 3.5), head width 0.36 mm (range 0.32 to 0.37).
- **Second instar:** Length (N=5) 5.4 mm (range 5.0 to 5.8), head width (N=6) 0.59 mm (range 0.55 to 0.61).
- **Third instar:** Length 9.0 mm (range 8.3 to 9.7), head width 0.92 (range 0.80 to 1.00).
- **Fourth instar:** Length 15.6 mm (range 14.5 to 17.3), head width 1.50 mm (range 1.38 to 1.65).
- **Fifth instar:** Larvae are mostly without sub-dorsal stripes, only 4 of 52 exhibited a hint of sub-dorsal stripes. Length 28 mm, head width 2.6 mm (range 2.25 to 2.85).

**Pupa:** Length 17.4 mm (range 16.5 to 19.0), width 5.25 mm (range 5.0 to 5.5), height 6.2 mm (range 5.5 to 6.5).

Results
Females have not been observed ovipositing at this location. The females readily oviposit on *Lupinus arcticus* S. Wats (Fabaceae), but will not oviposit on *Astragalus alpinus* in captivity. Larvae were reared on ornamental lupine (russel hybrid) and *Hedysarum boreale* in the lab. All larvae became mature fifth instar the first summer and then diapaused. The larvae pupated the next spring without eating.

Discussion
The natural life history was not studied at this location. The adults start flying in early June which is 20-30 days earlier than the other *Colias*.

*Colias canadensis*

at Mile 110 Dalton Hwy

Figs. 24, 25

This location is 0.9 km west of the Dalton Highway at Mile 110. The location is 9 km south of the Arctic Circle at 66°29.36’N and 150°43.46’W at the elevation of 690 meters. This location is about 120 meters above treeline on a mixed wet and dry hillside.

Results
Females have been observed ovipositing on *Lupinus arcticus* S. Wats. Larvae were reared on *L. arcticus* at the research site, and in the lab on *Hedysarum boreale* and ornamental lupine (russel hybrid). On June 21 of 2005 five stations were established with females for ovipositing. One female was put in each station. On August 14 four second instar larvae were observed among the five stations. These were at base of the plants so they had already diapaused even though the plants were still in good condition. On June 21 of 2006 the sleeves were removed for the winter. On May 20 of 2006 the sleeves were installed. At this time the plants had already started to grow with one plant having grown 1.5 centimeters. On July 13 fourteen fifth instar larvae were observed. On September 8 when the sleeves were removed sixteen fifth instar larvae were found. They all had left the plant and had hibernated on the sleeve near the ground. Since they left the plants and tried to crawl away they did not intend to eat in the spring. If they had been left there they would not have been found in the spring so they were taken to the lab. Five of these larvae pupated the next spring without eating. The other larvae died during overwintering.

Discussion
The location at Mile 94 was a much better location than Mile 110. The Mile 94 location is 1.5 miles west of the highway on a flat dry ridge. In 1999, 2001, and 2003 there was an abundant number of adults flying. In 2000 and 2002 there were only a few flying. It was apparent that *C. canadensis* has a two year life cycle at this location and the research verifies this. The majority of adult males taken at this location are *C. canadensis* but a few *C. boothii*
have been collected. Some of the larvae are like *C. canadensis* at Northway, some are like *C. boothii*, and some are intermediate. The two larvae in Figs. 24, 25 are from the same female. It appears that *C. canadensis* and *C. boothi* commonly hybridize at this location or are conspecific. It was hoped to do further research at this location but the area burned in 2004 and there were no adults flying in 2005. In June of 2001 females were sleeved on *Lupinus* plants at Mile 94 Dalton Highway. When the researcher returned 11 days later the eggs were just beginning to hatch. These larvae were reared on *Hedysarum boreale* and ornamental lupine in the lab.

The larvae that died during overwintering had become diseased. They probably became diseased while being transported home.

**Concluding Discussion**

During the period of July 2005 to June 2006 seven *C. hecla*, ten *C. nastes*, one *C. palaeno*, and all (71) *C. philodice* completed development. This proves that even during an unfavorable time span some individuals complete development in one year. Then all of the remaining individuals completed development in 2007 (two years time span). Unfortunately, there was considerable loss of larvae the second winter so the numbers were low that completed development in two years.

Nearly all of the *C. nastes* and all of the *C. palaeno* (only one) that were started from eggs in 2006 completed development in 2007. This demonstrates that during a favorable time span most individuals (except *C. boothii*) will complete development in one year.

If a good supply of *C. gigantea* had been obtained it is probable that some individuals would have completed development in one year.

There was a huge loss of larvae from Sept. 8 of 2006 to June 1 of 2007. The cause can only be speculated but all species were affected. It would seem that the most likely cause would be the weather. Whatever the cause was it sure didn’t inhibit the Satyrds from having an abundant flight in 2007.

The first winter larvae of all species overwinter in different instars (first, second, third). It is obvious that the larvae do not progress at the same rate in nature. The second winter larvae of *gigantea, hecla, nastes*, and *palaeno* overwintered as fully grown fourth instar. Anytime one of these become fifth instar they complete development without diapausing.

During the second summer the larvae of *boothi* become fully grown fifth instar and then diapaused. *C. boothi* is the only species on the north slope that diapauses as fifth instar. This is the reason that they fly 7-10 days earlier than the other species. However, there are fresh *boothi* individuals that fly with *hecla* and *nastes*.

*Colias canadensis* and *boothi* are the only Colias in Alaska that diapause as fifth instar larvae. They both diapause as mature fifth instar.

The larvae of some species of *Colias* vary in the expression of patterns and colors. In Utah some larvae of *philodice* and *eurytheme* exhibit sub-dorsal stripes and some do not exhibit the sub-dorsal stripes. In Alaska some larvae of *christina* exhibit the sub-dorsal stripes and some do not. Also, some *christina* larvae have the orange along the sub-dorsal stripes and some do not. However, all observations indicate that *boothi* is the only species in the *hecla* group that the larvae have the orange along the sub-dorsal stripes.

Quite often when stations were monitored there would be Arachnids on the sleeve or among the rocks at the base of the sleeve. A few times a spider was observed in the process of eating a larva that was resting on the sleeve. Occasionally a dead larva which had been eaten was found stuck to the sleeve. Probably other larvae had been eaten and the carcass fell to the ground. A few larvae were parasitized by Ichneumid wasps and Diptera. The parasites must have stung the larvae while resting on the sleeve.

The larvae that were reared in the lab were under constant light. It is assumed that the constant light would have no affect on the north slope larvae since they have constant light naturally. However one larvae of *gigantea* in 2007 went straight through to adults which was quite unexpected.

Many of the natural oviposition observations were made in years previous to the start of the study. All of the plant species on which ovipositions have been observed were used in this research. All of the *Colias* species were successfully reared to adults on all the corresponding plant species.
Acknowledgements

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References

Harry, Jack L.

Table 1. Daily high temperatures (deg. C) at research site.

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Fig. 1 - *C. boothii thula* holotype
Mead River, Alaska, USNM

Fig. 2 - *C. boothii boothii* lectotype
Boothia Peninsula, Nunavut, USNM

Fig. 3 - aberrant *C. boothii* male
Dalton Highway, north slope, Alaska

Fig. 4 to 9 - dorsal views of male variation in *C. boothii*, Dalton Highway, north slope, Alaska

Figure 4
Figure 5
Figure 6

Figure 7
Figure 8
Figure 9

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Figure 10
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Figure 13

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Figure 47: side of *C. gigantea* egg with 2 micrometer holes
Figure 48 – white male *C. palaeno* mile 323 Dalton Highway, Alaska

Figure 49 – *¼* gynandromorph *C. palaeno* mile 94 Dalton Highway, Alaska

Figure 50 – reared *C. pelidne* male dorsal view Mile 323 Dalton Highway, Alaska

Figure 51 – reared *C. pelidne* male ventral view Mile 323 Dalton Highway, Alaska

Figure 52 – wild caught *C. pelidne* female dorsal view Mile 323 Dalton Highway, Alaska

Figure 53 – wild caught *C. pelidne* female ventral view Mile 323 Dalton Highway, Alaska

Figure 54 – wild caught *C. pelidne* female dorsal view Mile 323 Dalton Highway, Alaska

Figure 55 – wild caught *C. pelidne* female ventral view Mile 323 Dalton Highway, Alaska
Notes on the natural life history of *Papilio machaon* in Alaska

**Resume:** The results of a brief study of the natural life history of *Papilio machaon alaska* Scudder on the north slope of Alaska in 2005 are annotated.

The site for this research on the north slope of Alaska is 92 miles (147 km) south of Deadhorse (Prudhoe Bay). This is the same research site as that of the *Colias* research in this publication. The eggs and larvae of *P. machaon* were found on *Petasites hyperboreus* Rydb. (coltsfoot) (Asteraceae) at Sagwon Hills which are 30 miles (48 km) north of the camp site. The eggs and larvae were sleeved on *hyperboreus* near the camp. The materials and methods are also the same as the *Colias* research.

On July 17 of 2005 eight eggs and one first instar larva were put in the first station. On July 18 six first and one second instar larvae were put in the second station. On August 10 there was one fifth and four fourth instar larvae in the first station. Also, on August 10 there were three fifth instar larvae in the second station. It is unknown what happened to the missing larvae. On August 28 the sleeve was removed from the first station for the winter. At this time there were three pupae which were attached to a dwarf birch stem near the ground. On August 28 in the second station there was one pupa, one prepupa, and two fifth instar larvae. The pupa was attached to dwarf birch stem near the ground. The prepupa was attached to the sleeve about 3 centimeters above the ground. The sleeve was reset to another plant with the prepupa and the two larvae.

On May 24 of 2006 the pupae in the first station were missing. It is probable that they were eaten by a vole. At the second station the one pupa not under the sleeve was found. A sleeve was put over this pupa and on June 19 a female adult emerged. There was one pupa attached to the sleeve and 2 dead larvae carcasses. The pupa that was attached to the sleeve died or may have already been dead on May 24. The larvae did not mature and pupate before cold weather stopped them or the food plant became inedible.

The two larvae that did not pupate, died during the winter. Although results from two larvae do not constitute a rigorous scientific study, they indicate that *P. machaon* larvae cannot survive the winter. The larvae and one pupa were under a sleeve so they may not have had proper conditions. A short cold spell during July or August with mild freezing temperatures and snow are not unusual so they must be able to survive these conditions. This demonstrates that *P. machaon* larvae pupate the same season as the eggs are laid.
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http://groups.yahoo.com/group/TILS-leps-talk/
ABSTRACT: Rearing techniques are discussed for 375 different butterfly and skipper taxa from Utah and beyond.

Additional keywords: ova, larvae, pupae, over wintering, obtaining and caring for immatures

INTRODUCTION

The authors of this paper, Jacque Wolfe, Jack Harry, and Todd Stout, with contributions from Dale Nielson have over 100 years combined experience collecting and rearing butterflies. This publication includes natural and lab host plants. We hope that this information will help you avoid some of the mistakes and losses we have experienced. We also hope that this publication will encourage someone who has only collected adults to give rearing a try.

For those new to rearing we encourage starting small. Not only can rearing provide perfect specimens but also provide knowledge regarding the life histories of butterflies, which includes how to find caterpillars or how to entice live females to lay eggs. The advantages justify the time and effort it requires.

Another advantage of rearing is that some species, like *Papilio indra* and *Megathymus* species, are difficult to collect as adults. Therefor, rearing them can be much easier. For example, collecting larvae or netting a single live female can result in obtaining a nice series of perfect specimens.

Remember not to be discouraged when you have setbacks. There is a learning curve involved with rearing that this paper will help to accelerate. Good luck.
GENERAL METHODS OF OBTAINING IMMATURES

For most species, it is both faster and easier to collect gravid females and confine them on their host plant to obtain ova. For larger butterflies, such as *Limenitis*, *Adelpha*, and *Papilio*, we use a 12 to 24-inch cubic cage constructed with a frame made with thin strips of wood or aluminum and covered with nylon window screen (Fig. 7). With a screen lid and no bottom it can be placed on a flat surface. For Lycaenids a 6-inch cubic cage works well.

Another type of container that is used for larger females is a 5-gallon plastic bucket pail with a screen top. Our favorite container for all but the largest butterflies is a 3-quart plastic container with a chiffon top. Place a 2-inch thick piece of foam rubber that fits tightly in the bottom. A 1½-inch diameter hole is made in the center so that a 1½-inch diameter by 2½-inch tall water container can be inserted. These containers, which are used to hold pills, can be purchased at many pharmacies. The foam rubber keeps the host plant from falling, therefore creating a very portable arrangement. It can ride on the dashboard or seat of your vehicle and sit in filtered sunshine while you are out collecting. Place a small container that has a honeywater soaked sponge in it set on the foam rubber for females to nectar (Figs. 1-2).

The proper amount of host plant placed in the container that houses a live female is critical. There needs to be sufficient plant but the females must also have room to fly. Most butterflies will oviposit under artificial light. Use a 60 or 100-watt bulb placed about 6 inches from the container. We use a 24-hour timer and have it set for 3 hours on and then 3 hours off during the day and then switched off overnight.

Using indoor light has several advantages. First, the females won't overheat, which is always a danger when using direct sunshine that passes through a window. Second, you can get eggs on an overcast day. If you do use direct sunlight take precautions to prevent overheating. Place the container in partial shade or filtered sunlight through a window. Venetian blinds can be very helpful to filter sunlight. Having 3 or 4 females in a cage or container is ideal as they keep each other active. Sometimes a solitary female will not be very active without other stimuli. Sometimes females become active with a change of light from overcast to sunshine and vice versa.

For some groups of butterflies large numbers of ova or larvae may be found. With a few taxa, field collecting of immatures is preferable to searching for elusive females. Actual methods will be discussed in each genus or species account.

Searching for eggs or larvae in the field can be advantageous for several reasons. First, it can extend the productivity of a day of collecting in the field. Not only can you check host plants for immatures from sunrise until the time of day when the adults begin to fly, but also from the time of day they stop flying until dusk. At the same time, if overcast weather keeps the adults grounded, you can still spend a full day in the field looking for immatures. Second, if you are unable to collect adult butterflies during one of their flight periods, you can always look for immatures either before or after their main flight depending upon the species.

For species that diapause as larvae you can collect these in the early spring. This not only gives you a wider timeframe for having immatures, but also extends your collecting season, which can be beneficial after a long winter. Even in the dead of winter there are collecting opportunities. For example, you could have an enjoyable and productive day looking for *Limenitis hibernacula* adjacent to river courses or *Hypaurotis crysalus* eggs near the buds of oak trees until the new growth of spring makes them too hard to find.

Collecting post-diapause larvae means you should have some adults without having to overwinter your larvae. A word of caution is that many species of checkerspot larvae, if smaller than 4th instar, may feed for an instar and then re-diapause. There are many possible explanations for this. If the larvae are somehow overcrowded or if the quality or quantity of hostplant is somehow restricted, these checkerspot larvae can diapause until later that spring or for another year. Also exposing the larvae to extra warm temperatures can cause re-diapause.
GENERAL REARING

Lab rearing necessitates that larvae be protected from predators, such as spiders, earwigs, etc. The only size requirement for containers and cages is that they must be large enough to hold sufficient host plant without crowding the larvae. For rearing in larger numbers, common 3 or 5-gallon pails or 10-gallon terrariums (Fig. 14) can be effective. When larvae are small, many can be placed in a container, but at most 15 to 20 larger larvae. Overcrowding must be avoided.

As stated earlier, these containers or cages must have chiffon, fine mesh, or screen covering. Chiffon is preferable as the porous top allows light and air into the container and allows moisture to escape. This is also essential as it keeps caterpillar frass dry. Frass should be removed from the rearing environment frequently and deteriorating host plant should be replaced regularly. Overexposing larvae to their own wet frass or deteriorating plant can cause disease in many varieties of butterflies and should be avoided.

Place the food plant stems into a small bottle of water through the neck of the bottle. There must be no spaces for the larvae to crawl or fall into the water through the neck of the bottle. Wrapping the stems with the correct amount of plastic wrap is an effective way to seal the opening. Also, an effective method to create an understory where the larvae can hide, rest or pupate is to place sections of white paper towel loosely around the bottle.

As stated earlier, food plant cuttings deteriorate and lose nutritional value and need to be replaced regularly. When larvae are small do not wait until all the plant has been consumed before changing it. The amount of time that the hostplant remains useable depends on the plant species itself. Good rules of thumb to follow is to replace host plant cuttings every 3 days in your cage or container and obtain a fresh supply of refrigerated cuttings about every week or so. Again, the timing on these replacements needs to be fine tuned depending upon the plant species.

Disinfection of any cage or container is also important. A mild bleach and water mixture applied for ten minutes is effective. Lysol or any other 2-long-chain quat, sold in an aerosol can, can also be effective. Avoid handling the larvae as much as possible, but when necessary, a small larva can be picked up with a camel's-hair brush dipped in water. A toothpick dipped in water or teasing needle also works.

Most larvae can be transferred by laying the section of plant that they are attached to onto a new plant. After the larvae crawl onto the fresh plant the old pieces can be removed. Larvae that are set to molt from one instar to the next NEVER should be forced off their host plant which is why cutting them away from the host as described above is always advisable.

Using potted hostplant is much less labor intensive and the plant is always fresh (Fig. 15). We recommend 1½ to 2-gallon pots. They are small enough for easy handling and large enough for a good supply of plant. To contain the larvae bend two 4-foot long 9-gauge wire lengths into a U-shape. Push the ends of the wire loop into the soil with the loops at 90 degrees to each other. Place a mesh sleeve over the wire and secure to the pot with twine or elastic cord. 5-gallon paint strainers from home improvement stores are ideal for this.

An effective method to keeping plants watered is to place the potted plant in a plastic pan with about 2 inches of water. If you keep plants outside replace water regularly as mosquitos can reproduce in standing water. With this method the plant is watered from the bottom. Place your plants in the sun for 3 or 4 hours a day and then back in the lighted lab before it gets dark. If left alone for 3-4 days, the plants will be okay inside.
A similar technique to potting plants is to place a mesh or chiffon sleeve around a live branch or branches that contain larvae. Naturally, the plant must be in a secure area. Make sure that the netting is sealed tightly around the branch. This technique is not recommended when rearing larvae that need to be exposed to 24-hour photoperiod or reared in a different manner. All special handling will be listed in the individual accounts.

Pupae should be placed in a container with a paper towel or cardboard lining so that emerging adults can climb to the mesh top to expand their wings. Pupae should be regularly misted with water until emergence or until they are put in diapausing containers to be placed under an overwintering process.

After the freshly emerged adults have expanded their wings they should be put in an individual paper-lined container and kept in subdued light or a dark closet for 1½-2 hours. If they are not crowded, 3 or 4 that emerge at the same time can be placed in the same container. They should then be put in the refrigerator for a minimum of 24 hours (5-6 days for Euphydryas). After being refrigerated they can be put in the freezer. Specimens should be briefly thawed and placed in glassine envelopes and stored in an airtight container and kept in the freezer. They will stay fresh enough to spread for a few months.

**OVERWINTERING TREATMENT OF DIAPAUSING IMMATURES**

It is necessary to have a refrigerator that is used exclusively for overwintering immatures. Put a thermometer inside so the temperature can be monitored. Adjust the cold until the temperature is 30-32 degrees. We use the largest plastic storage container that will fit in our large refrigerator. We place 2-4 small plastic containers, containing water, into the large container, which provides 100 percent humidity in the large container. The immatures are put in plastic containers that either have small holes poked in them or simply a chiffon top. Some loosely wadded white paper towels can be put in a container for the larvae to cling to but it is not necessary. The immatures should be kept in the winter conditions for at least 4 months but longer is okay. After the immatures begin diapause, during the summer, we simply put them in the containers and store in the lab until fall. They must be misted every other day or so until put in the refrigerator.

**Post-winter treatment of immatures:**

**Ova:** Overwintering ova should only be brought out of cold conditions and exposed to room temperature when suitable hostplant is available. Specific strategies will be discussed in the species accounts.

**Larvae:** After overwintering, the larvae should be placed in the rearing container, on their host, just like the one used to rear them to diapause. You can however, until diapause is broken, have many more larvae in a container. Until the larvae begin to feed they must be sprayed 2-3 times each 24 hours to prevent dessication. As is true with rearing pre-diapausal larvae, when plant condition starts to deteriorate it must be changed. Keep the larvae under 24-hour photoperiod. This procedure may be modified for larvae that were reared to diapause using a special method. This too will be covered in their account.

**Mature larvae:** Mature larvae finish all feeding prior to overwintering. Therefore, they need to be treated exactly the same way pupae are treated both before and after they pupate.

**Pupae:** For the first 7 days, pupae should be kept moist. Soaking with a spray bottle quite often can do this. For less attention, cover with a few layers of white paper towel and spray until soaked. This is necessary during the transition time from being exposed to colder temperatures to the lab with a long photoperiod. After the first week they can just be misted 2-3 times a day. Pupae that need different handling will be covered in the genus or species accounts.
Adults: In spite of efforts, we have not as of yet, created a suitable overwintering technique for those groups of butterflies, i.e; Polygonia, Nymphalis, Zerene, Phoebis, Eurema, etc. that overwinter or spend the dry season as adults.

Please see http://raisingbutterflies.org for updated content from this article, which also includes text, photos, and videos on raising butterflies. This site is collaborative and will invite others to share their best practices as well.

ARRANGEMENT OF SPECIES/SUBSPECIES ACCOUNTS

Host(s): Larval food plants as documented in nature by one of the authors. Documentation is defined as finding larvae or eggs on a species of plant and rearing them on that species of plant.
Lab host(s): Plants we have used in the lab to obtain ova or rear the larvae.
Other's host(s): Ova or larvae were collected by persons other than the three authors but were reared by one of the authors.
Remarks: Special handling or collecting methods, behavior, etc.

Parnassius smintheus savi
Host: Sedum lanceolatum
Remarks: Post-diapause larvae are easy to see as they feed out in the open. Larvae can be seen as one walks, so many plants can be checked in a short time. This is preferable to getting eggs from a female because you have adults one year sooner. This is one genus where wing-caught specimens may be more desirable than reared ones because reared females do not have a sphragis. Females have been observed ovipositing on rocks adjacent to host Sedum. Ova hibernate.

Parnassius eversmanni thor
Remarks: In nature, the females oviposit near the probable host, which is Corydalis pauciflora. In captivity the females readily oviposit on material near the Corydalis plants.

Papilio
Remarks: Papilio eggs perish if kept in an airtight container. They should be kept on white paper towel in a plastic container with a chiffon top and misted occasionally. Larvae can be transferred to the host with a camel's-hair brush or teasing needle as they hatch, or when hatching is near the eggs can be put on the host.

Papilio machaon group females normally oviposit readily in captivity. Papilio glaucus group females are rather fussy about ovipositing in the lab. Females that were reared on Foeniculum vulgare, after mating, will oviposit on F. vulgare.

Papilio coloro
Host: Thamnosma montana
Lab hosts: Foeniculum vulgare, Ruta graveolens
Remarks: Adult abundance is greatly determined by sufficient and timely moisture. The best plants to find larvae on are those growing on the shoulder of the road or other places where the road was bulldozed, stimulating fresh growth (or germination) of the plants. Due to run-off, available moisture is much greater here, resulting in the new growth on which the females love to oviposit. To be sure of getting both adult color forms, it is best to obtain ova from captive females. Since Thamnosma plants do not have much food growing on them, the lab hosts are preferable for rearing. In Utah, black adults (form clarki) seem to increase in abundance during the summer months. They are never common in Utah.
**Papilio bairdii** (including ssp. oregonius)

**Hosts:** *Artemisia dracunculus, Cymopterus duchesnensis, Lomatium grayi grayi* (In years with a population explosion, we have found larvae on *Lomatium junceum*).

**Lab host:** *Foeniculum vulgare*

**Remarks:** Population numbers can drastically fluctuate, producing large numbers in some years and very few in other years. Checking plants that are growing near a watercourse can be very productive where population size warrants. Where yellow (*brucei*) and black (*bairdii*) forms fly together, obtaining eggs from captive females is a good way of obtaining numbers of both forms. *P. bairdii* is multiple-brooded so the pupae usually emerge in the same year.

**Papilio brevicauda bretonensis**

**Lab hosts:** *Daucus carota, Lomatium dissectum, Foeniculum vulgare*

**Other's host:** *Lingusticum scoticum*

**Remarks:** Keep watch on pupae for a month or so, as some may break diapause. Most larvae reared on *Lomatium dissectum* will die as large fifth-instar or prepupa.

**Papilio zelicaon** (including form nitra)

**Hosts:** *Lomatium dissectum, L. grayi grayi, Zizia aptera, Sphenosciadium capitellatum*

**Lab host:** *Foeniculum vulgare*

**Other's host:** *Musineon tenuifolium*

**Remarks:** The surest way to get both forms, in good numbers, is to get ova from captive females. Larvae can be found.

**Papilio machaon alaska**

**Host:** *Petasites sagittatus*

**Papilio machaon pikei**

**Host:** *Artemisia dracunculus*

**Lab host:** *Foeniculum vulgare*

**Remarks:** With both machaon subspecies, some larvae can be found.

**Papilio polixenes asterius**

**Lab host:** *Foeniculum vulgare*

**Papilio indra indra** (includes western Nebraska ssp.)

**Hosts:** *Lomatium graveolens, Cymopterus terebinthinus*

**Other's host:** *Musineon tenuifolium*

**Remarks:** *P. indra* colonies in Salt Lake and Davis counties use *Lomatium graveolens*. To the north in Cache County, they use *Cymopterus terebinthinus*. *Papilio indra* in Cache County die when reared on local *Lomatium graveolens* but do fine on *Lomatium graveolens* from Davis or Salt Lake County. This inconsistency is puzzling. It may have to do with the soil where it is growing. We have found that *L. graveolens* and *C. terebinthinus* may be used to rear many indra subspecies. We have had many losses on *L. graveolens* and none on *C. terebinthinus*. The older stalks on the periphery of each *Lomatium* should be discarded when gathering plant to feed larvae. The stalks in the center are fresher and last longer. Unlike most plants, *Lomatium* and *Cymopterus* spp. will stay fresh longer if they are not put in a tub or vase of water. Put the cuttings in a plastic garbage bag and place in an ice chest. Refrigerated plant is safe to use for 3 days but no longer. Eggs from a female are good of course but females of all subspecies can be hard to locate at times. Eggs and larvae can be collected in good numbers. Plants that are growing against rocks are the most productive. Eggs and small larvae will usually be found on the plant's periphery. Larger larvae may be hiding in the center of the plant, or off the plant entirely.
**Papilio indra minori**
**Hosts:** Lomatium junceum, L. eastwoodii, L. parryi, Cymopterus terebinthinus  
**Lab hosts:** Lomatium graveolens  
**Remarks:** Population numbers vary greatly due to parasitism and rainfall. Keep pupae under long photoperiod for a month or more, watch closely, and mist daily. Some might emerge.

**Papilio indra (UT west desert segregate)**
**Host:** Lomatium grayi var. depauperatum  
**Lab hosts:** Lomatium graveolens, L. junceum, Cymopterus terebinthinus calcicola.

**Papilio indra calcicola**
**Hosts:** Lomatium parryi, L. scabrum  
**Lab host:** Lomatium junceum, Cymopterus terebinthinus  
**Remarks:** A highly variable taxon.

**Papilio indra shastensis**
**Host:** Cymopterus terebinthinus, Lomatium macrocarpum  
**Lab host:** Lomatium graveolens  

**Papilio indra pergamus**
**Hosts:** Taushia parishi, T. arguta  
**Lab host:** Lomatium graveolens  

**Papilio indra fordi**
**Host:** Cymopterus panamintensis  
**Lab host:** Cymopterus terebinthinus  
**Remarks:** Late instar larvae feed fine on Lomatium parryi but first-instar do not do as well on it.

**Papilio canadensis**
**Lab hosts:** Populus fremontii, P. tremuloides

**Papilio rutulus**
**Hosts:** Populus tremuloides, P. lombardii, Salix spp.  
**Lab host:** Populus fremontii  
**Remarks:** Immatures can be found.

**Papilio glaucus**
**Host:** Prunus serotina  
**Lab host:** Salix exigua  

**Papilio multicaudatus**
**Hosts:** Fraxinus pennsylvanica, Prunus virginiana  
**Remarks:** Frequently large numbers of larvae can be collected. Try places of new construction where it has been landscaped with many small host trees. Also, seek out isolated host plants with new growth. We have also found larvae in numbers in Evanston, Green River, Rock Springs, and Rawlins, Wyoming, where cultivated ash grows in corporate areas of these cities.

**Papilio eurymedon**
**Hosts:** Ceanothus velutinus, Prunus virginiana  
**Lab host:** Prunus serotina  
**Remarks:** On several occasions we have had a small number of pupae remain in diapause until the second spring.
**Papilio cresphontes**  
**Host:** *Citrus* spp.  
**Remarks:** Larvae can be found in good numbers on isolated plants. Always seek out new growth especially suckers coming out from near the trunk of the mature tree. Larvae do not feed on older leaves.

**Papilio xuthus**  
**Host:** *Citrus* spp.

**Battus philenor philenor**  
**Host:** *Aristolochia watsoni*  
**Lab hosts:** *Aristolochia tomentosa, A. druior, A. fimbriata*  
**Remarks:** *Aristolochia watsoni* is a very prostrate plant. It would usually go uncut if the area were mowed. Many larvae can be collected in the late afternoon, however. As soon as the sunlight hits at a sharp angle many larvae crawl up on tall plants. They can be easily seen from quite a distance. By carefully searching the understory below the larvae that you see you will usually find its host. More often than not there will be more larvae on it. You can collect until it is too dark to see or for a while later if you have a good lantern.

**Battus philenor hirsuta**  
**Host:** *Aristolochia californica*  
**Remarks:** Pupae can be found in the winter, often in good numbers. The host is a large vine usually growing around a tree or shrub. Pupae can be found on the vine or nearby trees and posts. The pupae on the trees or posts are usually no more than 3 feet above the ground. They are easy to see when there is no foliage to block your view. In season, larvae should be easy to find.

**PIERIDAE:**  
**Remarks:** Monitor hatching ova or separate ova, as hatching larvae will often eat other ova.

**Colias:**  
**Remarks:** For large numbers of immatures of all *Colias*, it is best to get ova from females. The larvae of *Colias eurytheme, Colias alexandra, and Colias philodice* will not diapause if reared with a 24 hour photoperiod. It is best to use a 24 hour photoperiod for all species of *Colias*. Constant light accelerates larval development and, with some individuals or species, prevents diapause. Any taxon that might not diapause (that we are aware of) will be mentioned in the species account. *Colias* are very hard to rear as they are highly prone to disease. The easiest and best way to rear all *Colias* is to use potted plants.

*Colias* larvae that always diapause will not leave drying plant in search of fresh food, but will just diapause on the plant. Potted plants stay fresh and the larvae will grow to their maximum diapausing instar. If only cut plant is available the containers must be designed so they can be placed in the sun for a few hours a day and then put in the lighted lab before sundown. Ultraviolet rays greatly reduce the chance of disease. Once a day carefully remove the sleeve and check the plant for freshness and add water to the water bottle. When the plant freshness starts to decline transfer each larvae to the leaf of a fresh plant.

**Colias eurytheme**  
**Hosts:** *Medicago sativa, Lupinus argenteus, Astragalus lentiginosus*  
**Lab host:** *Thermopsis montana*  

**Colias philodice eriphyle**  
**Hosts:** *Medicago sativa, Astragalus cicer, A. lentiginosus, Lupinus argenteus*  
**Lab host:** *Thermopsis montana*
**Colias interior**  
Lab hosts: Vaccinium caespitosum, V. angustifolium, Vaccinium sp.  
Remarks: Females fly in open timber. When shadows are long few females are flying.

**Colias pelidne skinneri**  
Lab host: Vaccinium myrtilloides  
Remarks: In timbered areas, females do not fly much when sunlight comes at too low of an angle.

**Colias gigantea gigantea**  
Lab host: Salix exigua, Salix sp.  
Remarks: An occasional larva will not diapause when reared with 24-hour photoperiod. These have always been females.

**Colias gigantea inupiat**  
Host: Salix lanata  
Lab host: Salix exigua

**Colias gigantea harroweri**  
Host: Salix spp.  
Lab host: Salix exigua, Salix sp.

**Colias scudder**  
Hosts: Vaccinium caespitosum, V. myrtilloides, Salix planifolia  
Lab host: Salix exigua

**Colias boothi thula**  
Hosts: Hedysarum mackenziei, Astragalus arcticus

**Colias canadensis**  
Host: Lupinus arcticus  
Lab hosts: Hedysarum boreale, Lupinus (russell hybrids)

**Colias hecla glacialis**  
Hosts: Astragalus arcticus, Hedysarum mackenziei  
Lab Host: Astragalus cicer

**Colias nastes nastes**  
Remarks: Oviposition observed on Astragalus arcticus

**Colias nastes alaska**  
Host: Oxytropis borealis

**Colias nastes streckeri**  
Lab hosts: Astragalus cicer, Astragalus sp.

**Colias palaeno chippewa**  
Host: Vaccinium uliginosum  
Lab host: Vaccinium caespitosum

**Colias mossi**  
Remarks: Oviposition observed on Astragalus uniflora.
**Colias lesbia**

**Host:** Medicago sativa

**Colias dimera**

**Remarks:** Oviposition observed on Medicago sativa, Trifolium sp.

**Colias weberbaueri**

**Remarks:** Oviposition observed on Trifolium sp. and Medicago sativa.

**Colias vaithieri vauthieri**

**Host:** Trifolium sp.

**Lab host:** Medicago sativa

**Colias vaithieri cunninghami**

**Remarks:** Oviposition observed on Astragalus sp.

**Colias flaveola mendoza**

**Remarks:** Oviposition observed on Astragalus sp.

**Nathalis iole**

**Host:** Bidens cermua

**Lab host:** Tagetes sp.

**Eurema nicippe**

**Hosts:** Sena hirsuta var. gluterna, S. covesii

**Zerene eurydice**

**Host:** Amorpha californica

**Remarks:** Larvae refuse Medicago sativa and Melilotus officinalis in the lab. Larvae that fed on Trifolium repens in the lab perished either because of lab host incompatibility or disease.

**Pieris napi macednoughi**

**Pieris napi sequoia**

**Pieris napi reicheli**

**Pieris napi venosa**

**Pieris napi marginalis**

**Hosts:** Descurainia richardsonii

**Lab hosts:** Brassica nigras, Cardaria draba, Dentaria californica, Isatis tinctoria, Arabis holboelli, Arabis sp., Rorippa nasturtium-aquaticum

**Remarks:** If larvae are reared with an 8-hour photoperiod the adults will be the dark spring form. A long photoperiod produces the light summer form. Rorippa nasturtium-aquaticum is a usable lab host from early spring to fall.

**Pieris rapae**

**Hosts:** Cardaria draba, Isatis tinctoria, Arabis sparsflora

**Remarks:** Wherever unsprayed crucifers grow in cities, suburbs, or disturbed areas, larvae and ova can be found.

**Pieris occidentalis**

**Hosts:** Thlaspi montanum, Descurainia californica, Cleome serrulata, Brassica nigras

**Lab hosts:** Brassica nigras, Isatis tinctoria, Cardaria draba
**Pieris protodice**
*Hosts:* Brassica nigra, Sisymbrium officinale, Stanleya pinnata, Cleome serrulata, Cardaria draba, Sisymbrium altissimum
*Lab host:* Cardaria draba

**Pieris beckerii**
*Hosts:* Stanleya pinnata, Brassica nigra, Cleome serrulata, Descurainia pinnata, Sisymbrium altissimum
*Lab host:* Isatis tinctoria
*Remarks:* Occasionally larvae can be found in large numbers. Last instar larvae provide conspicuous feeding damage as they strip the inflorescence of the host.

**Pieris sisymbrii**
*Hosts:* Arabis microphylla, A. sparsiflora, A. perennans, Isatis tinctoria, Descurainia pinnata, Cardaria draba
*Lab hosts:* Sisymbrium altissimum

**Pieris sisymbrii nigravenosa**
*Hosts:* Streptanthus cordatus, Descurainia pinnata, Arabis holboelli
*Lab hosts:* Arabis perennans, Cardaria draba, Isatis tinctoria, Sisymbrium altissimum

**Anthocharis lanceolata lanceolata**
*Lab hosts:* Arabis holboelli, A. sparsiflora
*Other's host:* Streptanthus tortuosus

**Anthocharis cethura pima**
*Hosts:* Streptanthella longirostris, Descurania pinnata, Caulanthus lasiophyllum var. utahensis, Sisymbrium irio
*Lab host:* Arabis perennans
*Remarks:* We have had mixed results using Sisymbrium irio as a lab host even though females occasionally oviposit on it in nature. Pupae can diapause for up to 11 years in the lab and still produce healthy adults.

**Anthocharis sara sara**
*Host: Dentaria californica*
*Lab hosts:* Arabis sparsiflora, Isatis tinctoria

**Anthocharis sara pseudothoosa**
*Hosts:* Descurainia pinnata, Arabis perennans (Fig. 4)
*Lab hosts:* Arabis glabra, Isatis tinctoria

**Anthocharis thoosa thoosa**
*Hosts:* Arabis perennans, Arabis holboelli, Descurainia pinnata, Streptanthella longirostris, Isatis tinctoria
*Lab hosts:* Any species of Arabis will serve as a lab host. Larvae eventually die on Streptanthus cordatus and Brassica nigra.

**Anthocharis thoosa colorado**
*Hosts:* Descurainia pinnata, Arabis spp.
*Lab hosts:* Any species of Arabis will serve as a lab host.

**Anthocharis thoosa inghami**
*Hosts:* Arabis perennans, Descurainia pinnata, Streptanthella longirostris
*Lab hosts:* Any species of Arabis will serve as a lab host.

**Anthocharis julia julia**
*Hosts:* Arabis glabra, A. holboelli, A. perennans, Descurania pinnata, Streptanthella longirostris
*Lab hosts:* Isatis tinctoria
**Anthocharis julia browningi**

**Hosts:** Arabis glabra, A. perennans, A. sparsiflora var. subvillosa, Arabidopsis thaliana, Descurainia pinnata

**Lab hosts:** Arabis microphylla, Isatis tinctoria, Streptanthella longirostris. (Note: Any species of Arabis will serve as a lab host. Larvae accept but perish on Sisymbrium altissimum, Cardaria draba, and Chorispora tenella. Larvae refuse Capsella bursa-pastoris and die.

**Anthocharis julia stella**

**Hosts:** Arabis perennans, Descurainia pinnata

**Lab hosts:** Isatis tinctoria

**Remarks:** Females from the Anthocharis julia stella TL near Marlette Peak, Carson City, Nevada, do not oviposit on the inflorescence as do many other taxa within the Anthocharis sara complex. They oviposit on the center stalk towards the middle. This likely happens because deer or other animals consume inflorescences.

**Anthocharis julia sulfuris**

**Hosts:** Descurainia pinnata, Arabis drummondii, Arabis sp.

**Lab host:** Isatis tinctoria

**Remarks:** Females of Anthocharis julia sulfuris from Boise County, Idaho oviposit near the inflorescence of Descurainia pinnata whereas females of Anthocharis thoosa thoosa oviposit more towards the middle to the upper two thirds of the plant.

**Anthocharis julia flora**

**Lab hosts:** Arabis glabra, Isatis tinctoria

**Anthocharis julia alaskensis**

**Lab host:** Arabis glabra

**Remarks:** Pupae develop to produce adults approximately 6 days earlier than do Colorado Anthocharis julia julia under identical lab conditions.

**Anthocharis midea annickae**

**Lab host:** Arabis holboelli, A. sparsiflora, A. perennans

**Other's host:** Arabis glabra

**Euchloe hvantis lotta**

**Hosts:** Sisymbrium altissimum, Descurainia pinnata, Stanleya pinnata, Streptanthus cordatus, Caulanthus lasiophyllum utahensis

**Lab host:** Isatis tinctoria

**Remarks:** Can occasionally find larvae in good numbers. Females oviposit towards the inflorescence of the host. Adults fly usually after Pieris sisymbri nigravenosa and Anthocharis thoosa thoosa have reached their peak flight.

**Euchloe hvantis hvantis**

**Lab hosts:** Arabis holboelli, Arabidopsis spp.

**Other's host:** Streptanthus tortuosus

**Remarks:** Larvae accept but die on Cardaria draba in the lab.

**Euchloe ausonides coloradensis**

**Hosts:** Arabis sparsiflora, A. perennans, Isatis tinctoria, Descurainia pinnata, Cardaria draba

**Lab hosts:** Arabis holboelli, Arabidopsis spp.

**Other's host:** Streptanthus tortuosus
**Euchloe olympia**

**Hosts:** Arabis glabra, Boechera fendleri, Descurainia pinnata,

**Lab hosts:** Arabis sp., Isatis tinctoria

**Remarks:** Larvae are similar to *Euchloe ausonides coloradensis* at early instars but look darker at later instars. Like other Euchloini larvae prefer fruits and flowers.

**Ascia monuste monuste**

**Ascia monuste phileta**

**Hosts:** Capparis sp.

**Lab host:** Tropaeolum majus

**Other's host:** Lepidium virginicum

**Remarks:** Length of photoperiod does not appear to affect the darkness of adults.

**Neophasia menapia menapia**

**Hosts:** Pinus edulis, P. ponderosa

**Remarks:** Overwinters as ova. Larvae camouflage well against its host. Before years of heavy flights, post-diapause late instar larvae can be found if portions of tree are mist sprayed with water because the larvae jerk back and forth violently. This suggests that the mist may be perceived as some sort of predator making the larvae very conspicuous.

**Daunus plexippus**

**Host:** Asclepias speciosa (Fig. 3)

**Remarks:** Females seem to prefer plants that are growing among scattered trees or shrubs.

**Daunus gilippus thersippus**

**Hosts:** Sarcostemma cynanchoides, Asclepias speciosa, A. erosa

**Agraulis vanillae incarnata**

**Host:** Passiflora sp.

**SATYRIDAE:**

**Remarks:** Overall, female satyrids will oviposit liberally in the lab. If you feel comfortable dealing with potted grasses and sedges rearing many multivoltine taxa of satyrids is not too difficult. The problem arises with some univoltine taxa that are difficult to force through to adult under lab conditions. Diapause for many satyrids is not the rigid concept it can be for many other species of butterflies, except skippers. For example, some species of satyrids, i.e., *Coenonympha tullia brenda*, will feed through to 4th instar and then slow down its feeding and growth rate to the point that larvae will not grow nor progress to the next instar. It is not always clear as to when these larvae should be placed into diapause or attempted to be forced through to adults under a 24-hour light scheme.

*Megisto cymela, Erebia magdalena,* and *Neominois ridingsi* are other examples of satyrids which feed extremely slow under lab conditions. Their feeding rate seems to slow down even further as they approach mature fifth-instars, suggesting that some *Neominois* may overwinter in that stage.

**Coenonympha tullia ampelos**

**Coenonympha tullia brenda**

**Coenonympha tullia eryngii**

**Lab Host:** Poa pratensis

**Remarks:** Obtain ova from live females. Females prefer to oviposit on dead blades. Larvae will feed on many species of potted grass. It is not generally difficult to push larvae of multivoltine populations of the *Coenonympha tullia* complex through to adult under lab conditions. However, univoltine populations either diapause at 3rd or 4th instar, or slow down their feeding rate as to continue feeding without growing or molting to the next instar.
Cercyonis spp.
Remarks: Use a similar rearing strategy for Cercyonis pegala, C. sthenele, C. meadi and C. oetus. Set up live females in a small cage. Females seem to prefer to oviposit on dead blades of any bunch grass, including Poa pratensis. In the lab ova postpone hatching for roughly 17 days and unfed 1st instar larvae diapause. This is oddly similar strategy as compared to Speyeria spp. Larvae can be forced out of diapause by placing them on fresh grass blades and placing them under 24 hours of light. Once larvae start feeding it is advisable to then rear them through the fall/winter months on any convenient species of potted grass.

Cercyonis meadi mexicana
Lab hosts: Bouteloua gracilis, Poa pratensis, Most species of grasses will serve as a lab host.

Megisto cymela
Lab host: Bromus inermis
Remarks: Both Megisto cymela and Megisto rubricata larvae have taken a long time to rear through to adults under lab conditions (4-5 months) either due to the usage of problematic grasses or artificial photoperiod problems.

Megisto rubricata cheneyorum
Lab host: Bromus inermis
Remarks: Like many satyrids, larvae will accept many species of grasses in the lab. Mature larvae will aestivate and not pupate until exposed to treatments of mist spraying, simulating summer monsoons in the desert southwest.

Neonympha areolata areolata
Lab host: Poa pratensis
Remarks: Females will oviposit on grasses as well as sedges. Larvae will accept grasses in the lab but generally do better on sedges.

Satyrodes appalachia appalachia
Hosts: Carex stricta
Lab host: Cyperus esculentus
Remarks: Females will oviposit on sedges. Larvae will accept many varieties of sedges in the lab. Finding larvae in the field can be difficult.

Erebia magdalena magdalena
Lab host: Poa pratensis
Remarks: In the field females oviposit haphazardly around rocks and talus near its native grasses. Under lab conditions, females do not necessarily even need grasses or a lot of sunlight to oviposit. Young instar larvae are extremely wary and should be raised on potted grasses surrounded by nylon netting or chiffon to avoid escape (Fig. 15). Young instars that wander off into water have proven to be drown-resistant. Larvae take many months in the lab to feed from first to fifth-instar. Larvae likely diapause as mature 5th instar; but this is not proven.

Neominois ridingsi dionysius
Lab host: Pseudoroegneria spicata
Remarks: Caterpillars grow very slowly in the lab and likely diapause as 5th instar.

Neominois wyomingo
Host: Pseudoroegneria spicata
Remarks: Unfed 1st instars diapause; however, larvae will feed on fresh grasses if provided. Larvae grow very slowly in the lab.
**Asterocamva clvton texana**

**Host:** Celtis reticulata  
**Lab host:** Celtis occidentalis  
**Remarks:** Larvae and pupae can be found.

**Asterocamva clvton clvton**

**Host:** Celtis reticulata  
**Lab host:** Celtis occidentalis  
**Remarks:** Larvae and pupae can be found. New growth on isolated plants may be best.

**Asterocamva clvton flora**

**Host:** Celtis reticulata  
**Lab host:** Celtis occidentalis  
**Remarks:** Larvae can be found. New growth on isolated plants may be best.

**Asterocamva celtis celtis**

**Host:** Celtis reticulata  
**Lab host:** Celtis occidentalis  
**Remarks:** Larvae can be found. New growth on isolated plants may be best.

**Asterocamva celtis montis**

**Host:** Celtis reticulata  
**Lab host:** Celtis occidentalis  
**Remarks:** Larvae can be found. New growth on isolated plants may be best.

**Asterocamva celtis antonia**

**Host:** Celtis reticulata  
**Lab host:** Celtis occidentalis  
**Remarks:** Larvae can be found. New growth on isolated plants may be best.

**Asterocamva leilia**

**Host:** Celtis pallida  
**Remarks:** Larvae can be difficult to find. The best strategy is to look for new growth on isolated plants. 1st instar larvae refuse Celtis reticulata in the lab

**Nymphalis antiopa**

**Hosts:** Salix exigua, Celtis reticulata, Ulmus pumila  
**Remarks:** Occasionally larvae can be collected in large numbers.

**Nymphalis californica**

**Hosts:** Ceanothus velutinus, C. martini  
**Remarks:** Large numbers of larvae can be found, sometimes in varying sizes in close proximity. They can defoliate their host.

**Nymphalis milberti**

**Host:** Urtica dioica  
**Remarks:** Larvae are easily collected in large numbers. Larvae are gregarious at early instars and then tend to scatter somewhat at later instars and make nests similar to Vanessa atalanta.

**Vanessa atalanta**

**Host:** Urtica dioica  
**Remarks:** Occasionally larvae can be found in good numbers. Larvae create nests.

**Vanessa cardui**

**Hosts:** Cirsium undulatum, Carduus nutans, Lupinus argenteus, L. sericeus, Helianthus annuus  
**Remarks:** Larvae can be found in good numbers. It can be a common urban dweller.

**Vanessa annabella**

**Hosts:** Malva neglecta, Sida hederacea  
**Remarks:** Larvae and ova can be collected in good numbers on isolated plants. Late summer, until a hard freeze, is the most productive time. Host Malva neglecta is somewhat cold weather resistant. Normally adults overwinter but larvae at any instar can survive the winter as well. Instead of diapausing larvae can feed at a very slow rate during the winter when temperatures are above freezing.

**Vanessa virginieensis**

**Host:** Anaphalis margaritacea, Gnaphalium palustre  
**Remarks:** Can find ova and larvae in good numbers.
**Precis coenia**
Hosts: *Plantago major, P. lanceolata, Antirrhinum majus*
Remarks: Larvae can be found on host in agricultural areas. Adult phenotypes can vary in the lab when larvae are subjected to variable photoperiod and temperature.

**Precis nigrosulcata**
Lab host: *Antirrhinum majus*
Other's host: *Mimulus sp.*
Remarks: Larvae can be found.

**Anartia jatrophae guantanamo**
Host: *Lantana sp.*
Lab host: *Plantago major*

**Polygonia spp.**
Remarks: Some *Polygonia* species have two seasonal forms. Larvae of these reared with 24-hour photoperiod produce the non-hibernating form. Those reared with 8-hour photoperiod produce the hibernating form.

**Polygonia faunus arcticus**
**Polygonia faunus ceneray**
Host: *Salix scouleriana*
Lab host: *Salix sp.*
Remarks: Larvae can be found.

**Polygonia satyrus** (coastal and inland forms)
Host: *Urtica dioica*

**Polygonia gracilis zephyrus**
Hosts: *Ribes montigenum, R. viscossissimum, R. cereum*
Lab host: *Ribes oxyacanthoides*
Remarks: Last instar larvae tend to feed and strip leaves towards the ends of branches on shrubs. Larval strip pattern is noticeable and last instar larvae can be found if timing is correct. Larvae that feed on *Ribes montigenum* can camouflage themselves well.

**Polygonia oreas oreas**
**Polygonia oreas threatfuli**
Lab hosts: *Ribes oxyacanthoides, Ribes sp.*
Other's host: *Ribes divaricatum*
Remarks: Females will often hover next to the host. They prefer using scattered bushes interspersed in an open clump with other shrubs or small trees.

**Polygonia interrogationis**
Lab host: *Ulmus pumila*

**Adelpha eulalia**
Hosts: *Quercus turbinella, Q. gambelii*
Lab hosts: *Quercus robur, Q. alba*
Remarks: Can find an occasional larva. Most females are reluctant to lay in captivity. In nature females have been observed ovipositing on leaves that are more concealed within the body of the bush, making the finding of immatures more difficult. Similar to *Limenitis*, 1st instar larvae construct a perch extending the vein of the leaf.
**Limenitis lorquini lorquini**
**Limenitis lorquini burrisonii**
**Limenitis lorquini pallidafacies**

**Hosts:** *Salix* spp., *Populus angustifolia*, *Amelanchier alnifolia*

**Lab hosts:** For all *Limenitis*, *Populus fremonti*, *Salix exigua*

**Remarks:** Most *Limenitis* that are reared with a 24-hour photoperiod will not diapause. Freshly molted 2nd instars are especially sensitive to monitoring photoperiod. 1st instars do not. In habitats where the hosts are scattered or confined ova and small larvae can be collected. During the late fall or winter when most of the leaves have fallen it is not too difficult to spot hibernacula. As is true with other *Limenitis* species, females oviposit well in a cage with high humidity and exposed to filtered sunlight. Immatures of all *Limenitis* can be found.

**Limenitis lorquini x Limenitis weidemeyeri**

**Hosts:** *Prunus virginiana*, *Salix* sp.

**Remarks:** A very occasional hibernacula can be found along a willow-lined stream. Less than a hundred feet up on the dry hillside, short, scattered chokecherries were growing. Nearly every *Prunus* plant had a hibernaculum and sometimes as many as six. This is a typical scenario in the dry southwest.

**Limenitis weidemeyeri weidemeyeri**
**Limenitis weidemeyeri oberfoelli**
**Limenitis weidemeyeri angustifascia**
**Limenitis weidemeyeri latifascia**

**Hosts:** *Amelanchier alnifolia*, *Populus tremuloides*, *P. angustifolia*, *Salix* sp., *Prunus virginiana*

**Limenitis archippus archippus**
**Limenitis archippus obsoleta**
**Limenitis archippus lahontani**
**Limenitis archippus floridensis**

**Hosts:** *Salix exigua*, *S. laevigata*, *Populus fremontii*

**Remarks:** Females prefer to lay on branches that are hanging over water

**Limenitis archippus obsoleta x Limenitis astyanax arizonensis**

**Host:** *Salix* sp.
**Lab host:** *Populus fremontii*

**Limenitis astyanax astyanax**
**Limenitis astyanax arizonensis**

**Hosts:** *Prunus serotina*, *P. virginiana*, *Salix* sp., *Populus* sp.

**Limenitis arthemis rubrofasciata**

**Host:** *Salix* sp.
**Lab hosts:** *Populus balsamifera*, *P. tremuloides*

**Euphydryas phaeton phaeton**
**Euphydryas phaeton ozarke**

**Hosts:** *Chelone glabra*
**Lab hosts:** *Castilleja chromosa*, *Penstemon cyananthus*

**Remarks:** Post diapause larvae have been found on *Castilleja coccinea* and *Virburnum recognitum*.
**Euphydryas gilletti**

**Hosts:** *Lonicera involucrata, Veronica wormskjoldi*

**Remarks:** This taxon has a 2-year life cycle; therefore, the larvae overwinter for two winters. After the first winter the second diapause can be avoided by rearing with a 24-hour photoperiod. Egg clusters and small larvae are easy to locate. Females like scattered plants near a stream or on a dry hillside above it. Post-diapause larvae can be found in good numbers in leaf litter under the host or on branches in the plants interior.

**Euphydryas anicia anicia**
**Euphydryas anicia maria**
**Euphydryas anicia alena**
**Euphydryas anicia bernadetta**
**Euphydryas anicia windi**
**Euphydryas anicia macyi**
**Euphydryas anicia veaziae**
**Euphydryas anicia wheeleri**
**Euphydryas anicia hermosa**

**Hosts:** *Penstemon gloriosus, P. utahensis, P. cyananthus, P. palmeri, Penstemon sp., Castilleja chromosa, Lonicera involucrata, Symphoricarpus oreophilus*

**Lab hosts:** All *Euphydryas* that use *Castilleja* or *Penstemon* will switch from one to the other.

**Other's host:** *Bessaya wyomingensis*

**Remarks:** Egg clusters and prediapause larvae can be found. Prediapause larvae feed gregariously in silk nests and disperse to plants up to 500 feet away before diapause and can be found as post-diapause larvae on these plants the next year. An individual female will normally lay her eggs in a relatively small area. *Euphydryas* break diapause early and can be found as soon as their host has usable growth. Even if no larvae are visible on a plant, search all the understory and debris nearby. Post-diapause larvae that feed on shrubs are not sensitive to dry and warm conditions and may be taken directly to the lab for rearing. Post-diapause larvae like to rest on dead foliage. Post-diapause larvae can rediapause if overcrowded. However, larvae that have re-diapaused can break diapause yet again the same year if exposed to another cold treatment of one month.

**Euphydryas colon nevadensis**
**Euphydryas colon sperryi**

**Hosts:** *Symphoricarpus oreophilus*

**Lab hosts:** *Penstemon cyananthus, Symphoricarpus sp.*

**Remarks:** Post diapausal larvae can be found in good numbers. They are easiest to see in early spring when the host has just leafed out. *Penstemon* and *Castilleja* grows very common among the *Symphoricarpus* but no larvae were found on them.

**Euphydryas chalcedona chalcedona**
**Euphydryas chalcedona olancha**
**Euphydryas chalcedona klotsi**
**Euphydryas chalcedona maclashani**

**Hosts:** many *Penstemon* spp., *Keckiella antirrhinoides*

**Lab hosts:** *Penstemon cyananthus, Penstemon palmeri*

**Euphydryas editha lehmani**
**Euphydryas editha colonia**
**Euphydryas editha baroni**

**Hosts:** *Castilleja chromosa*

**Lab hosts:** *Penstemon cyananthus, Collinsia sp.*

**Other's host:** *Penstemon sp., Plantago lanceolata*
**Phyciodes mylitta mylitta**

**Hosts:** *Cirsium undulatum, C. vulgare*

**Remarks:** Occasionally larvae can be found in good numbers. When reared with a 24-hour photoperiod most *Phyciodes* will not diapause. Females will sometimes oviposit on young *Cirsium vulgare* basal rosettes in the fall where the larvae overwinter under the leaves.

**Phyciodes orseis orseis**

**Phyciodes orseis ssp. (CA)**

**Lab host:** *Cirsium undulatum*

**Other's host:** *Cirsium cymosum*

**Remarks:** At least a few larvae will not diapause when reared with a 24-hour photoperiod. In some populations, most of the larvae will not diapause. Immatures can be found with regularity. Females like to use plants that are growing against a rock, tree or windfall.

**Phyciodes pulchella pulchella**

**Phyciodes pulchella shoshone**

**Phyciodes pulchella Camillus**

**Phyciodes pulchella ssp. (CA)**

**Host:** *Aster foliaceus*

**Lab Hosts:** *Aster chilensis, Aster sp.*

**Remarks:** Larvae can be found.

**Phyciodes pallida pallida**

**Phyciodes pallida barnesi**

**Lab host:** *Cirsium undulatum*

**Phyciodes cocyta cocyta**

**Phyciodes cocyta selenis**

**Lab hosts:** *Aster foliaceus, A. chilensis, Aster sp.*

**Phyciodes texana texana**

**Lab hosts:** *Dicliptera resupinata, Beloperone guttata*

**Remarks:** Immatures have been found.

**Poladryas arachne arachne**

**Hosts:** *Penstemon utahensis, P. humilis*

**Lab host:** *Penstemon cyananthus*

**Remarks:** Occasionally post-diapause larvae can be found. Post-diapause larvae refuse *P. cyananthus* but larvae obtained from ova thrive on it. Most larvae reared from ova will not diapause when reared with 24-hour photoperiod.

**Thessalia leanira leanira**

**Thessalia leanira wrighti**

**Thessalia leanira alma**

**Thessalia leanira elegans**

**Thessalia leanira oregonesis**

**Hosts:** *Castilleja chromosa, C. sulphurea*

**Lab host:** *Castilleja spp.*

**Other's host:** *Castilleja affinis*
Remarks: Depending on sufficient moisture, large numbers of post-diapause larvae can be collected. In areas where *Euphydryas* are sympatric, *leanira* will be found after most *Euphydryas* have finished. The reason for this is that post-diapause larvae will molt before resuming feeding in the spring, whereas *Euphydryas anicia* complex larvae will feed immediately, placing *Euphydryas* larvae ahead of *Thessalia* larvae by approximately 1-2 instars. Most larvae will be found in the understory and surrounding vegetation. Larvae can be found as much as 3 feet from the host. Larvae of *elegans* will go through to adult if reared with a 24-hour photoperiod. The other subspecies diapause. Larvae of *wrighti* that have re-diapaused will resume feeding 1 month later if separated and placed on fresh host after a cold treatment.

*Thessalia theona thekla*
Hosts: *Castilleja lanata, C. laxa*
Lab host: *Castilleja chromosa*
Remarks: Post-diapause larvae can be found in good numbers. Larvae of *T. fulvia* and *T. cyneas* may be found in the same area. The larvae lack bright coloration.

*Thessalia fulvia fulvia*
*Thessalia fulvia coronado*
*Thessalia fulvia ssp.* (mostly AZ)
Lab host: *Castilleja chromosa*
Other's hosts: *Castilleja laxa, C. lanata, Castilleja sp.*
Remarks: Immatures can be found in good numbers.

*Thessalia cyneas*
Lab host: *Castilleja chromosa*
Other's host: *Castilleja laxa*
Remarks: Post-diapause larvae can be found in limited numbers.

*Thessalia chinatiensis*
Host: *Leucophyllum minus*
Remarks: Larvae can be found in numbers

*Chlosyne Ianais*
Host: *Anisacanthus wrighti*
Remarks: The host is a low spreading shrub that grows in dense colonies. Often larvae of all instars can be found as well as pupae. When the plants are bathed in sunshine the larvae are hiding in shady interior branches or the understory. Larvae are up on the plant feeding in early morning and evening.

*Chlosyne californica*
Host: *Viguiera deltoides*
Remarks: When moisture has been sufficient larvae can be found in good numbers.

*Chlosyne lacinia* form *adjustrix*
*Chlosyne lacinia* form *crocale*
Host: *Helianthus annuus*
Remarks: Larvae of all sizes can be found. Early spring larvae usually produce form *adjustrix* adults. Larvae collected mid-summer will produce both forms.

*Chlosyne gorgone gorgone*
Lab host: *Helianthus annuus*
**Chlosyne harrisi harrisi**

**Chlosyne harrisi liggetti**

**Chlosyne harrisi albimontana**

**Lab host:** *Aster engelmannii*

**Other's host:** *Aster umbellatus*

**Remarks:** Have obtained ova and reared larvae on *A. engelmannii*. *C. harrisi liggetti* post-diapause larvae can be found in good numbers on *Aster umbellatus* near Spruce Knob, West Virginia.

**Chlosyne nycteis nycteis**

**Chlosyne nycteis drusius**

**Lab host:** *Helianthus annuus*

**Other's host:** *Helianthus annuus*

**Chlosyne palla palla**

**Chlosyne palla eremita**

**Chlosyne palla flavula**

**Chlosyne palla ssp. (3-CA)**

**Chlosyne palla ssp. (MT)**

**Host:** *Aster engelmannii*

**Lab Host:** For all subspecies: *Aster engelmannii*

**Other's hosts:** *Aster* sp.

**Remarks:** When reared with a 24-hour photoperiod only an occasional larva will not diapause. Rearing post-diapause larvae with constant light is best. With a natural photoperiod about 50 percent of the larvae will rediapause. With 24 hours of light, 30 percent or less will rediapause.

**Chlosyne hoffmani segregata**

**Lab host:** *Aster engelmannii*

**Other's host:** *Aster brickellioides*

**Remarks:** With 24-hour photoperiod only an occasional larva will rediapause.

**Chlosyne gabbii gabbii**

**Lab hosts:** *Aster chilensis, Corethrogynoe filginifolia*

**Remarks:** Four females laid 5 egg clusters (one was very small) on *Aster engelmannii* as no natural host was available. Upon hatching the larvae fed for about 30 days before eventually dying. Females would not lay on *A. chilensis* but larvae thrived on it. When reared with 24 hour photoperiod an occasional larva will not diapause.

**Chlosyne sterope sterope**

**Chlosyne sterope dorothyi**

**Chlosyne sterope acastus**

**Hosts:** *Chrysothamnus viscidiflorus, C. greenei, Machaeranthera canescens, Pyrrocoma radiatus*

**Lab host:** *Aster engelmannii*

**Remarks:** Ova and larvae can be found in good numbers including an occasional pupa. Prediapause larvae of *C. sterope acastus* have always diapaused when reared under constant light. Of approximately 300 *C. sterope dorothyi* larvae reared from collected ova and 1st instar larvae, 92 went through to adults. 17 larvae of *C. sterope sterope* went through to adults (all females) and 87 larvae diapaused.

**Chlosyne neumoegeni**

**Host:** *Machaeranthera tortifolia*

**Remarks:** As with all desert species abundance is determined by sufficient timely moisture. Larvae can be collected in good numbers. They can be seen at a distance but a stealthy approach is advised. The slightest vibration of the host plant causes them to “jump ship” and drop to the understory.
**Chlosyne damoetas ssp. (UT, WY)**

**Chlosyne damoetas ssp. (MT)**

**Lab host:** *Aster engelmanni*

**Remarks:** These taxa are associated with *Solidago multiradiata* and *Erigeron leiomeris* the likely larval host plants. Females from all locations have laid egg clusters and the larvae have been reared on *Aster engelmanni*. Obtained ova from the UT and WY populations in 2001 and reared the 162 larvae to diapause. The majority diapaused as large 2\(^{nd}\) instars, 14 molted to 4\(^{th}\) instars, which fed very briefly and diapaused. The remainder diapaused as 3\(^{rd}\) instars. In 2002, raised post-diapause larvae under 24-hour photoperiod where all but two larvae grew an instar and rediaapaused. The two that went through to adult emerged as females. In nature, I think all would have diapaused. 2003 was a repeat of 2002. Two more female adults emerged and the rest diapaused as either large 3\(^{rd}\) or 4\(^{th}\) instars. In 2004 two males and one female emerged. All of the larvae diapaused as 4\(^{th}\) instars. Sometime in the fall of 2004 all of the diapausing larvae died. In 2006 we obtained 200-300 eggs from the distinctive Montana subspecies and 13 larvae went through to adults; 5 males and 7 females. 31 diapaused as large 4\(^{th}\) instars and the rest as large 3\(^{rd}\) instars. In 2007 fifteen went through to adults. About 30 percent of the larvae diapaused as 4\(^{th}\) instars; the rest as 3\(^{rd}\) instars. In 2008 only a few larvae were alive when spring arrived, all died without breaking diapause.

**Boloria spp.**

**Remarks:** There is an alien species of *Viola* that is toxic to *Boloria* and *Speyeria* larvae. This *Viola* is frequently grown in gardens.

**Boloria kriemhild**

**Host:** *Viola* sp.

**Remarks:** Larvae diapause at mid-instars when reared in the lab.

**Boloria frigga sagata**

**Lab host:** *Salix exigua, Salix* sp., *Viola* sp.

**Remarks:** If reared with 24-hour photoperiod a good percentage will not diapause. The females oviposited on *Salix* sp. Larvae do well on *Salix* or *Viola*.

**Boloria eunomia ursadentis**

**Host:** *Viola adunca*

**Speyeria diana**

**Lab Host:** *Viola* sp.

**Other's host:** *Viola papilionacea*

**Remarks:** *Speyeria diana* can be reared on potted *Viola tricolor* but some do not do well on it. Unfed 1\(^{st}\) instars will diapause. If you have *Viola* available you can break the larval diapause. Place the newly hatched larvae in a petri dish with new growth *Viola* leaf. Use 24-hour photoperiod and make sure the *Viola* is always fresh. This method gives you adults several months sooner and avoids the danger of your diapausing larvae dying. *Speyeria* females will oviposit well if placed in a paper grocery bag with a couple of *Viola* leaves. Fold the top shut and hold it closed with clothes pins or large paper clips.

**Speyeria cybele charlotti**

**Speyeria cybele leto**

**Speyeria cybele letona**

**Speyeria cybele ssp. (MT)**

**Lab hosts:** *Viola adunca, V. tricolor*

**Other's host:** *Viola papilionacea, Viola* sp.

**Speyeria nokomis apacheana**

**Lab host:** *Viola* sp.
Speveria idalia
Lab host: Viola sp.
Other's host: Viola papilionacea, V. pedatifida

Speveria mormonia luski
Lab host: Viola sp.
Other's host: Viola adunca, Viola sp.

Speveria atlantis nausicaa
Lab host: Viola sp.
Other's host: Viola sp.

Speveria edwardsi
Lab host: Viola sp.
Other's host: Viola nuttallii

Euptoieta claudia
Lab host: Viola sp.
Other's host: Passiflora sp.

Apodemia mormo mormo
Apodemia mormo virgulti
Apodemia mormo deserti
Apodemia mormo cythera
Apodemia mormo mejicana
Apodemia mormo durvi
Apodemia mormo nigrescens
Hosts: Eriogonum inflatum, E. corymbosum, E. fasciculatum, E. brevicaule, E. umbellatum, E. wrightii, Krameria glandulosa
Remarks: Larvae can be collected in good numbers. Larva usually live in a nest of leaves silked together. Larva using Eriogonum inflatum chew a hole into the inflated pod when it is green. They hide and eventually pupate in the dried pod. If the hole in the pod is silked over you know it is occupied. The larger the larvae you collect the better because they grow very slowly.

Apodemia palmeri
Hosts: Prosopis glandulosa, Prosopis pubescens
Remarks: When rearing in the lab, small larvae are reluctant to crawl from unusable plant if its cutting is laid on fresh plant. The larvae should be transferred with a camel's-hair brush.

LYCAENIDAE:
Remarks: When possible remove prepupae from rearing containers. Larvae are highly cannibalistic and will often eat prepupae or pupae.

Theclini:
Remarks: Almost all of the Theclini can best be reared by obtaining ova from females. The only reared Theclini that were obtained by other methods are Habrodais grunus, Hypaurotis crysalus citima, Satyrium californicum, Satyrium tetra, Sandia mcfarlandi, Incisalia fottis, Callophrys comstocki, and Strymon bazochi. These will be discussed with each species. Habrodais grunus and Hypaurotis crysalus are notorious for refusing to oviposit in captivity. To rear, they must be obtained as ova or larvae. Both species overwinter as ova. All Satyrium overwinter as ova. All Callophrys, Incisalia, and Mitoura overwinter as pupae.
**Habrodais grunus**  
**Host:** Quercus chrysolepis  
**Remarks:** Larvae can be collected in good numbers. Larvae can be found by inspecting the foliage or by placing a tarp under the branches and beating them with your net handle. If most of the larvae are small, beating will be more profitable. Some larvae may be *Satyrium auretorum*. There will also be a good supply of spiders, earwigs, moth larvae, etc.

**Hypaurotis crysalus citima**  
**Host:** Quercus gambelii  
**Remarks:** During the winter and spring following a good flight, eggs can be found in good numbers. Eggs are deposited at the base of next year’s leaf buds. Cuttings of host do not stay fresh very long.

**Harkenclenus titus immaculosis**  
**Lab host:** Prunus virginiana  
**Remarks:** To obtain ova confine females on cuttings that have cracks and imperfections in the stems. Few leaves are needed. Also lay some twigs at the base of the cuttings to construct an understory.

**Satyrium californica**  
**Hosts:** Purshia tridentata, Cercocarpus montanus, Amelanchier utahensis, Ceanothus velutinus, Prunus virginiana  
**Remarks:** All attempts to obtain ova from females in captivity have been unsuccessful. Oviposition has been observed in nature (although this is very time consuming) and the ova were taken. Larvae are usually difficult to find. However, in one good colony we were able to find a good supply of larvae by looking for attendant ants. Racemes of *P. virginiana* or *C. velutinus* are the best to use for rearing. We have found pupae in good numbers on two occasions.

**Satyrium sylvinus putnami**

**Satyrium sylvinus megapallidum**  
**Host:** Salix exigua  
**Remarks:** Females lay best on cuttings with cracks, holes and other imperfections. If each cutting has twig stubs, that helps also. Females like to tuck all their eggs in such places. Very few leaves are needed.

**Satyrium saepium**  
**Hosts:** Ceanothus velutinus, Ceanothus sp.  
**Remarks:** Small larvae prefer flowers. Larvae were obtained by beating the hostplant branches adjacent to the flowers, and allowing them to fall into a net or other container.

**Satyrium behri behri**  
**Hosts:** Purshia tridentata, *P. mexicana*, Cercocarpus montanus, C. ledifolius

**Satyrium fuliginosum semiluna**  
**Hosts:** Lupinus sericeus, *L. argenteus*  
**Remarks:** Ants, in some colonies, hollow out areas under most of the plants where *S. fuliginosum* flies and the larvae hide in these holes, tended by the ants. Often there are 3 or 4 larvae in a hole. The ants not only protect them from 6 and 8-legged predators but also from each other. They are very cannibalistic and must be reared separately. If you check the same holes a month earlier, you will find *Plebejus icarioides* larvae. Occasionally you find a late finisher when the *fuliginosum* larvae are quite small. Females oviposit in the ground at the base of the Lupine plants. To obtain ova from females the base of the plant must have some soil surrounding it. It probably also helps to have a few twigs and leaves on the soil.

**Satyrium auretorum**  
**Host:** Quercus chrysolepis
Satyrium tetra
Host: Cercocarpus betuloides
Remarks: S. tetra larvae were obtained by the beating method. Many were parasitized.

Satyrium liparops aliparops
Lab host: Prunus virginiana

Satyrium calanus godarti
Host: Quercus gambeli

Atlides hailesus estesi
Hosts: Phoradendron juniperinum, P. californicum
Remarks: Host will last for days in the lab with the stem of the mistletoe hostplant in water. If only raising a few larvae, using a closed container works when you replace host plant and container daily.

Sandia mcfarlandi
Host: Nolina erumpens
Remarks: Larvae can be found in good numbers, and can be found in the blossoms and resting at the base of the plant. Also, ova have been obtained from females. Four different color morphs of larvae were found on the same plant.

Incisalia eryphon eryphon
Incisalia eryphon purpurascens
Lab host: Pinus monophylla
Other's hosts: Pinus contorta, P. monophylla
Remarks: Larvae like the new growth of spring. Females prefer to oviposit on new growth and refuse to oviposit on older growth.

Incisalia augustinus
Hosts: Ceanothus velutinus, Purshia tridentata, Arctostaphylos uva-ursi, Prunus virginiana
Remarks: Oviposition observed on Cuscuta sp. Larvae have been collected from Ceanothus velutinus using the beating method.

Incisalia mossii
Host: Sedum lanceolatum

Incisalia fotis fotis
Host: Purshia mexicana
Remarks: Larvae can be found in good numbers. Since larvae can be found, no attempt has been made to obtain ova from females. Adults will emerge soon after pupae are put into a warm room in winter or spring. Some have even emerged in an unheated garage (or even in a refrigerator) in February.

Mitoura spinetorum
Hosts: Arceuthobium americanum, A. divericatum

Mitoura johnsoni
Lab host: Arceuthobium americanum
Other's host: Arceuthobium campylopodum

Mitoura siva siva
Mitoura siva chalcoviva
Hosts: Juniperus osteosperma, J. scopulorum
**Mitoura loki**
Lab host: *Juniperus californica*

**Mitoura loki thornei**
Lab host: *Cupressus forbesii*

**Callophrys sheridani neoperplexa**
Hosts: *Eriogonum heracleoides, E. racemosum, E. corymbosum, E. umbellatum, E. brevicaule*
Remarks: Females will oviposit in a squat tub closed container with host lying at the bottom (Fig. 8). Females respond well to artificial light that is utilized intermittently, turn the light on and off every 20 minutes or so.

**Callophrys affinis affinis**
Hosts: *Eriogonum umbellatum, E. elatum, E. racemosum, E. heracleoides*
Remarks: Larvae can be found.

**Callophrys apama apama**
Hosts: *Ceanothus fendleri, Eriogonum racemosum, E. alatum*

**Callophrys comstocki comstocki**
Hosts: *Eriogonum corymbosum, E. hermannii, E. fasiculatum*
Remarks: Larvae can be found in good numbers on the dorsal side of host leaves. Ova have been obtained from females, but most of our rearing was done by finding larvae.

**Callophrys dumetorum**
Host: *Lotus scoparius, Eriogonum fasiculatum*

**Strymon melinus franki**
Hosts: *Eriogonum racemosum, E. kearnyi, E. alatum, E. heracleioides, E. umbellatum, E. corymbosum, Astragalus utahensis, Malva neglecta, Malvella leprosa, Hibiscus mocheutos, Althea rosea, Medicago sativa, Hedysarum boreale, Lupinus argenteus, Sidalcea oregana, Iliamna rivularis*
Remarks: Larvae can be very abundant. If you are collecting other Lycaenid larvae and you find one with a double row of dashes down the back, you have a *melinus*. They have many color morphs. Larvae are very cannibalistic so must be reared separately.

**Strymon bazochii**
Host: *Hyptis pectinata*
Lab host: *Lantana camara*
Remarks: Ova and larvae have been found in good numbers. Therefore, no attempt has been made to obtain ova from females.

**Erora laeta**
Lab host: *Salix exigua*
Other's host: *Corylus cornuta*

**Erora quaderna**
Lab host: *Salix exigua*

**Lycaena spp.**
Remarks: In the Rocky Mountains, all Lycaena overwinter as eggs except *L. cupreus* and *L. phlaeas*. To obtain ova of the species that use *Rumex, Oxyria, and Polygonum* (except *cupreus* and *phlaeas*). It is necessary to have soil surrounding the base of the plant because females oviposit in the soil. A few twigs and leaves on the soil help. *L. cupreus* and *L. phlaeas* females oviposit on the leaves of the host plant.
**Lycaena gorgon**
*Host:* Eriogonum nudum
*Remarks:* Larvae and ova can be collected in good numbers. Large larvae can be seen at a distance, eating the very top of the flower stem.

**Lycaena mariposa penroseae**
*Lab host:* Vaccinium caespitosum, Vaccinium sp.

**Lycaena phlaeas arctodon**
*Host:* Oxyria digyna

**Lycaena phlaeas weberi**
*Lab hosts:* Rumex acetosella, Oxyria digyna (reared by Dale Nielsen), R. acetosa alpestris
*Remarks:* Obtained ova on Rumex acetosa alpestris from the colony and Oxyria digyna. Larvae refused Rumex acetosella hybrid that Lycaena cupreus and Lycaena editha do well on. Larvae accepted but perished soon thereafter on Rumex crispus. Reared under natural photoperiod but in a warm lab many larvae pupated and emerged. Some of the ova diapaus was. As day length lessened; pupae and half-grown larvae diapaus.

**Lycaena phlaeas feildeni**
*Lab hosts:* Oxyria digyna, Rumex acetosa

**Lycaena xanthoides xanthoides**

**Lycaena xanthoides dione**
*Lab host:* Rumex crispus

**Lycaena rubidus sirius**
*Host:* Rumex crispus
*Remarks:* Larvae can be found in large numbers. If the plant is growing in hard-packed soil with no understory there will be no larvae. Larvae are found in loose understory, plant debris or gravel. Larvae may be 4 or 5 inches below the surface and 7 or 8 inches away. Once 92 larvae were found under one small plant.

**Lycaena hyllus**
*Host:* Rumex maritimus
*Lab host:* Rumex crispus
*Remarks:* Larvae can be found in large numbers. Look in the understory as with prior species. Females will also oviposit on plants growing in water. On these, the larvae will be on the Rumex.

**Lycaena helloides helloides**

**Lycaena helloides mesaloceras**
*Lab hosts:* Rumex crispus, R. paucifolius
*Remarks:* Obtained ova of megaloceras on Potentilla concinna. Larvae were offered two other species of Potentilla besides concinna, and they refused all.

**Lycaena heteronea heteronea**

**Lycaena heteronea northi**
*Hosts:* Eriogonum umbellatum, E. racemosum, E. corymbosum
*Lab host:* Eriogonum heraclioides

**Lycaena nivalis browni**
*Lab hosts:* Rumex crispus, Rumex sp., Polygonum douglasii
*Remarks:* Some larvae eat Rumex but only a few lived. Most females will not oviposit on Rumex. An occasional female will lay but usually only a few eggs. Females oviposit well on Polygonum douglasii and larvae flourish on it. It is best to pot thick clumps of douglasii. It takes several individual plants to rear a single larva.
Lycaena arota schellbachi
Host: *Ribes leptanthum*
Lab host: *Ribes* spp.
Remarks: As soon as it is warm and sunny in the morning the females are active. Look for yellow composites near the host.

Lycaena editha montana
Lab host: *Rumex acetosella* hybrid
Remarks: Larvae will feed on other *Rumex* spp. besides their known hosts but most will die in the late instars.

Lycaena cupreus cupreus
Lab hosts: *Rumex acetosella* hybrid, *Rumex paucifolius*
Remarks: Most larvae diapause at 3rd instar; but some will go through to adult in the lab. Most females are reluctant to oviposit in captivity.

Lycaena cupreus snowi
Host: *Oxyria digyna*
Remarks: Larvae can be found but normally only a few. Females oviposit readily on leaves of *digyna* in a closed container.

Brevhidium exilis
Hosts: *Salsola iberica*, *Atriplex canescens*, *Portulaca oleracea*
Remarks: Put cardboard or a dark tarp under plant and beat with net handle. Larvae and pupae can be collected in good numbers using this technique.

Agriades sp. (MT)
Other's host: *Douglasia montana*
Remarks: Post-diapause larvae can be found. Females will oviposit well in the lab.

Lampides boeticus
Lab host: *Pisum sativum*
Remarks: Reared on fresh snow peas purchased at the supermarket.

Hemiargus ceraunus gyas
Host: *Melilotus officinalis*
Lab host: *Pisum sativum*
Remarks: Female oviposited on *Melilotus officinalis* but larvae accepted fresh snow peas purchased at the supermarket.

Celastrina ladon echo
Host: *Eriogonum racemosum*, *E. wrightii*, *Ceanothus velutinus*
Remarks: Larvae and ova can be found. Larvae found on *Eriogonum racemosum* in Sevier County, Utah, were oddly not tended by ants.

Celastrina ladon lucia
Host: *Ledum palustre*
**Everes amyntula**  
**Host:** Vicia americana  
**Lab host:** Lathyrus lanszwertii, L. latifolius, L. odorata  
**Remarks:** Young larvae burrow into flower buds and remain there for as long as the bud will support them. Larvae turn brown before diapausing or pupating. Some larvae diapaused and some did not, when reared with 24-hour photoperiod.

**Glaucopsyche lygdamus lygdamus**  
**Glaucopsyche lygdamus oro**  
**Hosts:** Astragalus sp., Lupinus sericeus, L. argenteus, Hedysarum boreale, H. mackenziei, Eriogonum wrightii  
**Remarks:** Larvae can be collected in good numbers. Look for ants tending larvae on inflorescens.

**Glaucopsyche piasus daunia**  
**Hosts:** Lupinus sericeus, L. argenteus  
**Remarks:** Where **lygdamus** and **piasus** fly together, **lygdamus** larvae are usually in mid to late-instar when **piasus** is found as eggs or small larvae. The majority of the larvae will usually be **lygdamus**. Females are reluctant to oviposit in captivity.

**Plebeius acmon acmon**  
**Plebeius acmon lutzi**  
**Hosts:** Eriogonum umbellatum, E. kearneyi, E. racemosum, E. wrightii  
**Remarks:** Have found larvae usually when looking for Callophrys or Euphilotes larvae. Females oviposit well on E. umbellatum. Most lutzi larvae that feed on leaves in the lab do not diapause (John Emmel, pers. comm.).

**Plebeius melissa melissa**  
**Host:** Medicago sativa

**Plebeius icarioides ardea**  
**Host:** Lupinus sericeus  
**Remarks:** Find larvae in ant holes beneath the plant. Mid-instar larvae usually diapause in nature but larvae can be forced through to adult if ova are obtained from live females in the lab.

**Plebeius shasta minnehaha**  
**Host:** Astragalus sp.  
**Remarks:** Larvae can be found in fair numbers.

**Philotes sonorensis**  
**Host:** Dudleya cymosa  
**Remarks:** Larvae can be found.

**Euphilotes spaldingi**  
**Host:** Eriogonum racemosum  
**Remarks:** Larvae can be found in good numbers. Examine food plant with a magnifying glass when you get to the lab. Often you will find small larvae. Rear separately. 1st and 2nd instar larvae, if reared under 24-hour photoperiod, will pupate and emerge the same year. The pupae must have the long photoperiod also. All of these methods apply to all Euphilotes we have reared. Diapausing Euphilotes pupae must be kept outside and monitored until emergence time (this coincides with the blooming of their hostplant). Without natural photoperiod and temperature they will not emerge.
**Euphilotes pallescens pallescens**  
**Euphilotes pallescens ricei**  
**Euphilotes pallescens arenamontana**  
**Host:** Eriogonum kearneyi  
**Remarks:** Larvae can be found in good numbers.

**Euphilotes enoptes ancilla**  
**Hosts:** Eriogonum heraclioides, E. umbellatum  
**Remarks:** Females tend to oviposit only on the open flower petals of Eriogonum heraclioides. Finding larger larvae on the host plant can be difficult because they camouflage or hide themselves quite easily on the flowers or seed pods of the host plant.

**Megathyminae:**  
**Remarks:** The "Mega" included in this list (and possibly all Megas) can be reared with good results. Megathyminae use only small to medium size plants and the larva usually kill the plant. To rear Megathyminae Yucca plants were planted in the yard. Over a period of several to 25 years the crop of Yucca harrimaniae spread by rhizomes to constitute a sizable and dense patch of Yucca plants. The plants become too dense to use and must occasionally be thinned by cutting the stems at ground level, also plants that become large must be cut off at ground level. Yucca angustissima plants spread only sparingly by rhizomes; however, individual plants would continue to grow and develop a large root. These plants also must be cut off at ground level when they become large. After being cut off, the root would grow two new stems, each one on opposite sides of the root. When there is a large root the larva does not kill the plant and after a few years the plant is usable again.

Agathyminus use Agave plants of any size in Utah. Agave plants were transplanted to the yard in the spring when they were going to be used. Most of the plants did not live permanently but would survive until the larvae pupated. When transplanted, the plants must be cleaned by removing the dead leaves and washing out the remaining good leaves. The dead leaves and rubbish harbor unwanted critters. It is easy to obtain ova from Agathyminus females but very few of the newly hatched larvae become established in a leaf. It was quite successful to harvest first and second instar larvae in nature and transplant to plants in the yard. Plants or group of plants must be covered with screen to keep the parasites and predators away from the larvae. Even with a screen cover an occasional larva would get parasitized or spiderized.

Megathyminae ova are glued to a leaf and are quite easy to find. Agathyminus ova are dropped in or near a plant and are nearly impossible to find.

**Megathyminae yuccae**  
**Hosts:** Yucca angustissima, Y. bacata, Y. harrimaniae  
**Remarks:** Larvae build a silk tent in the center of the plant. This tent can easily be found and many M. yuccae pupae can usually be found. The pupae can be harvested shortly before flight time. Larvae finish eating long before flight time but diapause until shortly before flight time to pupate. If larvae are found within a couple months of flight time they can be kept in their silk tube and will eventually pupate or a paper tube can be used.

**Megathyminae streckeri**  
**Hosts:** Yucca harrimaniae, Y. angustissima, Y. glauca  
**Remarks:** Larvae build their tent in the ground near the plant. The top of the tent is at ground level so it is very difficult to find the pupae. This taxon is best obtained by rearing. Many ova can be found easily after the females have been flying a few days.

**Agathyminae neumoegeni**  
**Host:** Agave scabra  
**Remarks:** Pupae can be found by spotting the trap door in the Agave leaf or by spotting the frass.
**Agathymus mariae**  
**Host:** Agave lechugilla  
**Remarks:** Pupae can be found by spotting the trap door in the Agave leaf or by spotting the frass.

**Agathymus alliae**  
**Host:** Agave utahensis  
**Remarks:** Larvae make their trap door on the under side of the leaf. It is possible to find pupae in the field but searching can be a long arduous process with limited results. To get a good series it is necessary to rear this taxon.

**Pyrginae, Hesperinae:**  
**Remarks:** Larvae of most hesperine and pyrgine (spread-wing skippers) construct nests. These skipper larvae differ from most moth larvae in that they exit in order to launch frass whereas most moths that make nests do not. In other words, finding larval nests on plants with frass means you have found a moth nest not a skipper nest. Females of most hesperine skippers will oviposit on and larvae will usually accept a far greater variety of grasses than is normally utilized in nature making lab rearing not too difficult. Rearing of some hesperine skippers from humid areas of the Midwest and Eastern U.S. requires the use of humidity (mist spraying) both for unhatched ova and for unfed first-instars in order to stimulate hatching and feeding.

Similar to Megathyminae, many (but not all) pyrgine and hesperine skippers pupate in the same nest they fed as larvae but seal up the entrance of the nest with silk. This extra layer of white silk is very noticeable in hesperine skippers. This increases the possibility of finding pupae in the wild.

Larvae of the Hesperine genera *Hesperia, Polites, Pseudocopaeodes, Hylephila,* and *Atalopedes* make nests down at the bases of bunch grasses and are nearly impossible to locate. Larvae of *Amblyscirtes, Atrytonopsis, Lerodea, Panoquina, Ochlodes, Poanes, Piruna, Thymelicus, Wallengrenia, Copaeodes* and *Oarisma* feed on taller, wide-blade species of grasses higher where they can be much more conspicuous. Some "skipperling" hesperine larvae make smaller nests or none at all.

Finding larvae of many pyrgine skippers is not too difficult when their population numbers are significantly high and hostplants are not too prevalent.

**Epargyreus clarus**  
**Hosts:** Glycyrrhiza mendota, G. lepidota, Robinia pseudoacacia, Lotus crassifolius  
**Remarks:** Larvae construct a nest among the leaves of the hostplant and are easily found in any instar. This is especially true when the host is Glycyrrhiza mendota. They are easiest to rear by finding 5th instar larvae so that they don’t have to be fed very long. When the pupae are put into a warm room in the spring to emerge they must be moist for some time (to adjust to the warmth) or they will desiccate. This is easily accomplished by placing them on wet paper towels and covered by wet paper towels. Use lab host Robinia pseudoacacia over natural host Glycyrrhiza lepidota in an open terrarium setup. The problem with using cuttings of Glycyrrhiza lepidota is that cuttings will wilt in an open terrarium. The only way to keep cuttings erect is to increase humidity in the setup similar to a closed terrarium setup recommended for Limenitis. However, this increased humidity in the closed terrarium does not allow frass to dry and larvae can die as pupae. Robinia pseudoacacia is a much drier host. Larvae use this host naturally in the Eastern U.S. An open terrarium setup is fine with this host and larvae stay healthy. For multivoltine populations such as Epargyreus clarus californica keeping pupae humid is not necessary.

**Polygonus leo savignyi**  
**Host:** Pisidium piscipula  
**Remarks:** Larval nests can be found on new growth of host. Larvae will pupate in nest.
**Phocides pigmaion okeechobee**  
**Host:** Rhizophora mangle  
**Remarks:** Larval nests can be found on host. Larvae will pupate in nest and pupae can be occasionally found. It is not advisable to use cuttings of the host, as larvae do not do well on cuttings. Try and obtain R. mangle (red mangrove) starts from a nursery and rear on potted plants. Use saline solution for potted plants.

**Thorybes pylades pylades**  
**Host:** Melilotus officinalis  
**Lab Hosts:** Trifolium repens, Medicago sativa  
**Remarks:** Mature larvae hibernate. Larvae make nests. Females have oviposited on Astragalus cicer in nature but first-instar larvae refused to feed.

**Thorybes mexicana nevada**  
**Lab host:** Medicago sativa, Melilotus officinalis  
**Remarks:** Mature larvae hibernate. For some reason pre-diapausal 5th instars can easily die during the wintering process.

**Thorybes diversus**  
**Lab host:** Trifolium repens  
**Remarks:** Mature larvae hibernate.

**Erynnis icelus**  
**Hosts:** Populus tremuloides, Salix sp.  
**Remarks:** Mature larvae hibernate. Pupae do not hibernate. Occasionally, lab reared larvae in the fall will pupate and will emerge a few weeks later. This also applies to other species of Erynnis.

**Erynnis brizo burgessi**  
**Hosts:** Quercus gambellii, Q. turbinella  
**Lab host:** Quercus alba  
**Remarks:** Seek out seedlings to find nests in the late summer or early fall. Mature larvae hibernate. Females oviposit only on very young tender new growth of leaves. Ova are white, usually turning orange 24 hours later.

**Erynnis telemachus**  
**Host:** Quercus gambellii  
**Lab host:** Quercus alba  
**Remarks:** Finding late instar larvae in late summer can be a challenge even during larger flights. Finding isolated seedlings growing along roads or fresh cuts can improve chances of finding immatures. Larvae take 3-4 months to mature to last instar in the lab.

**Erynnis meridianus**  
**Host:** Quercus turbinella  
**Lab hosts:** Quercus alba, Quercus gambellii  
**Remarks:** Double-brooded in southwest Utah. The key to finding immatures is to seek out ova on very fresh succulent new growth of the host during the summer brood in mid to late August. New growth is usually hard to find on the host during this time of the year, therefore, when you do find it you usually will find an ova. Timing is important because finding smaller instar larvae on the same new growth is usually much less productive as young instars can be quickly consumed by predation.

**Erynnis pacuvius lilius**  
**Host:** Ceanothus velutinus  
**Remarks:** Larvae can be found.
**Erynnis persius fredericki**  
**Host:** Lupinus argenteus  
**Remarks:** Larvae can be found.

**Erynnis afranius**  
**Host:** Hedysarum boreale  
**Lab host:** Lupinus argenteus  
**Remarks:** There are up to 3 broods at low elevations. Late instar larvae are not too difficult to locate. Poor-quality natural host in late summer produces smaller larvae and hence smaller adults for the first flight in the spring. Second brood adults are almost as large as *Erynnis telemachus*.

**Heliopetes ericetorum**  
**Host:** Sphaeralcea sp.  
**Lab hosts:** Malva neglecta, Sida hederacea, Althaea rosea  
**Remarks:** Larvae of all instars overwinter.

**Pyrgus scriptura**  
**Hosts:** Sphaeralcea ambigua, Sida hederacea  
**Lab host:** Althaea rosea  
**Remarks:** Larval nest is somewhat distinctive to *Pyrgus communis*. In the same habitat; larvae that are found on short hosts in dry areas are *P. scriptura*. *P. communis* larvae prefer healthier plants in wetter areas. Pupae overwinter.

**Pyrgus communis**  
**Hosts:** Sphaeralcea ambigua, Sida hederacea, Malva neglecta  
**Remarks:** Females will oviposit in a small cage. Finding larvae in disturbed or suburban areas is not too difficult where the adults fly. Larvae change color from green to brown when overwintering.

**Pyrgus ruralis ruralis**  
**Hosts:** Potentilla glandulosa, Fragaria vesca  
**Lab Host:** Potentilla fruticosa  
**Remarks:** Females will oviposit fairly well in captivity on the hostplant. Finding immatures in the wild can be very difficult. Larvae create nests but do not pupate in their nest. Pupae overwinter.

**Pyrgus centaureae loki**  
**Host:** Potentilla diversifolia  
**Lab Hosts:** Potentilla glandulosa, Fragaria vesca  
**Remarks:** Larvae accept *Fragaria vesca* in the lab but attempts to get pupae to emerge the following spring have failed. Current overwintering techniques that work for *Pyrgus ruralis ruralis* have not worked for *Pyrgus centaureae loki*.

**Systasea zampa**  
**Host:** Abutilon abutiloides  
**Lab host:** Alcea rosea  
**Remarks:** Conspicuous larval nests are not too hard to find.

**Pholisora catillus**  
**Hosts:** Chenopodium album, Amaranthus retroflexus  
**Remarks:** Larvae can be found.
**Hesperopsis libya libya**
*Host:* *Atriplex canescens*
*Remarks:* Larvae can be found in areas where the host is not overly abundant. Look on plants along the periphery of the population. Larvae create nests and will pupate in nests.

**Hesperopsis libya confertiblanca**
*Host:* *Atriplex confertifolia*
*Lab Host:* *Atriplex canescens*
*Remarks:* Look on isolated plants. Sometimes multiple larvae can be found on one plant. 3rd through 4th instar larvae will semi-aestivate during the hot summer months and will not usually pupate until mid to late-July. Therefore, it is advisable to sleeve larvae on local *Atriplex canescens* then harvest the late instars or pupae later.

**Hesperopsis alpheus oricus**
*Host:* *Atriplex canescens*
*Remarks:* Larvae can be found. Like *Erynnis*, *Thorybes*, and *Amblyscirtes* mature last instar larvae overwinter and pupate the following spring.

**Hylephila phyleus phyleus**
*Host:* *Cynodon dactylon*
*Lab host:* *Poa pratensis*
*Remarks:* Females will oviposit fairly well on Bermuda grass. Under lab conditions the amount of time between the 1st instar larvae and pupae is roughly 9 weeks, which is much quicker than many species of *Hesperia* and *Polites*.

**Atalopedes campestris campestris**
*Host:* *Cynodon dactylon*
*Lab host:* *Poa pratensis*

**Atalopedes campestris huron**
*Lab host:* *Distichlis spicata*
*Remarks:* Females will oviposit on *Distichlis spicata*. Larvae are generalists and will accept many species of grass in the lab.

**Polites vibex**
*Lab host:* *Poa pratensis*
*Remarks:* Larvae feed quickly from hatchling 1st instar to pupa in about 9 weeks.

**Polites origenes rhena**
*Lab host:* *Bromus inermis.*
*Remarks:* Raising this skipper can be difficult. Exposing larvae to 24 hours of light does not guarantee that they will go through and pupate the same year. Many larvae slow down their metabolism at late instars neither completely diapausing nor growing. Some overwintered larvae resume feeding at a normal rate, pupate and emerge. Whereas others continue feeding at a very slow rate and eventually die.

**Polites themistocles**
*Lab hosts:* *Poa pratensis, Phalaris arundinacea*
*Remarks:* Females oviposit somewhat sparingly on Kentucky Blue Grass. Larvae will accept *Phalaris arundinacea* in the lab and will go through to adults the same year.

**Polites sabuleti sabuleti**
*Host:* *Poa pratensis*
*Remarks:* Last instar larvae can burrow at the base of the plant to make a nest. Rearing larvae on potted grass is advisable.
**Polites sabuleti chusca**
*Host*: Distichlis spicata  
*Remarks*: Females will not oviposit on Poa pratensis in the lab.

**Polites peckius**
*Lab host*: Poa pratensis, Phalaris arundinacea

**Polites sonora utahensis**
*Lab host*: Sedges  
*Remarks*: Females oviposit very sparingly on grasses, if at all.

**Hesperia juba**
*Host*: Distichlis spicata  
*Lab Host*: Phalaris arundinacea  
*Remarks*: As is the case with many Hesperia, larvae grow at a fairly slow rate, about 2-3 months from ova to adult.

**Hesperia pahaska nr. martini**
*Host*: Bouteloua gracilis  
*Lab Host*: Phalaris arundinacea  
*Remarks*: As is the case with many Hesperia, larvae grow at a fairly slow rate, about 2-3 months from ova to adult. Rear larvae under 24 hours of light and provide fresh host to avoid diapause.

**Hesperia uncas lasus**
*Host*: Bouteloua gracilis  
*Lab Host*: Phalaris arundinacea  
*Remarks*: Females will oviposit on Distichlis spicata in the lab. As is the case with many Hesperia, larvae grow at a fairly slow rate, about 2-3 months from ova to adult. Rear larvae under 24 hours of light and provide fresh host to avoid diapause.

**Hesperia nevada nevada**
*Lab Host*: Phalaris arundinacea  
*Remarks*: Females prefer not to oviposit on Poa pratensis. Other natural bunchgrasses should be better.

**Pseudocopaeodes eunus eunus**
*Host*: Distichlis spicata  
*Remarks*: To get a series, try and get as many females as is possible. Compared to other hesperine skippers, ova of eunus are very large and females are limited to how many ova they can oviposit.

**Poanes taxiles**
*Host*: Bromus inermis  
*Lab host*: Phalaris arundinacea  
*Remarks*: Larvae attempt diapause (or a feeding slowdown) at 4th to 6th instar. If larvae are given fresh host regularly they will feed through and produce adults the same year.

**Poanes zabulon**
*Lab hosts*: Bromus inermis, Phalaris arundinacea.  
*Remarks*: Unlike Poanes taxiles, Poanes zabulon larvae only have five instars. Throughout much of its range, *P. zabulon* is multivoltine. Therefore, if larvae are given fresh host regularly they will feed through to pupae and produce adults the same year.
**Pomona hobomok**
Lab hosts: *Bromus inermis, Phalaris arundinacea.*
Remarks: Larvae have 6 instars and seem to insist on diapausing at the 4th instar.

**Ochlodes yuma yuma**
Host: *Phragmites australis*
Remarks: Larvae and pupae can be found. Ova have been found on *Sorghum halepense.* See http://utahbutterflies.ning.com/video/finding-yuma-skipper-larval for a video tutorial on how to find larval nests on *Phragmites.*

**Ochlodes sylvanoides napa**
Hosts: *Phalaris arundinacea, Bromus inermis*
Remarks: Larvae can also be found. Larval nests are conspicuous. Last instar larvae diapause for roughly 2-6 weeks before finally pupating, producing a late summer flight. Unfed 1st instars hibernate by silking the tips of the leaves.

**Amblyscirtes vialis**
Host: *Bromus inermis*
Lab Host: *Phalaris arundinacea*
Remarks: Females will oviposit in the lab. Mature last instar larvae hibernate. In the lab some mature last instar larvae will pupate, producing adults soon thereafter.

**Amblyscirtes fimbriata**
Host: *Phalaris arundinacea*
Remarks: Larvae can be found. Mature last instar larvae hibernate. However, in the lab many last instars will pupate and produce adults soon thereafter.

**Amblyscirtes eos**
Host: *Sorghum halepense*
Lab Host: *Phalaris arundinacea*
Remarks: Larvae can be found.

**Lerema accius**
Host: *Sorghum halepense*
Lab Host: *Phalaris arundinacea*
Remarks: Larval nests can be found.

**Lerodia eufala**
Host: *Sorghum halepense*
Lab Host: *Phalaris arundinacea*
Remarks: Larval nests can be found. Larval nests, coloration, and feeding patterns are oddly similar to *Thymelicus lineola.*

**Wallengrenia egeremet**
Host: *Panicum sp.*
Lab Host: *Phalaris arundinacea*
Remarks: Females will oviposit on a variety of grasses. Ova seem to need humidity to hatch.

**Oarisima garita**
Host: *Bromus inermis*
Lab Host: *Phalaris arundinacea*
Remarks: Larvae can be found. Using potted grasses is advisable for rearing this taxon.
*Thymelicus lineola*

**Host:** *Phalaris arundinacea*

**Remarks:** It is not too hard to find larval nests in the field. However, late instars use nests to a lesser degree or not at all. Larvae pupate on grass blades and are not too difficult to locate. This skipper overwinters as ova.

*Ancyloxypha numitor*

**Remarks:** It’s not too hard to find larval nests in semi-wet habitats. Larvae pupate in grass blades in such a tight fashion as to making their removal quite problematic. It is advisable to leave the nest intact for emergence purposes.

*Ancyloxypha areneg*

**Lab Host:** *Phalaris arundinacea, Bromus inermis*

**Remarks:** Populations are very local in wetland areas. Larvae pupate in grass blades in such a tight fashion as to making their removal quite problematic. It is advisable to leave the nest intact for emergence purposes.

*Copaeodes aurantiacus*

**Host:** *Cynodon dactylon*

**Lab Host:** *Phalaris arundinacea*

**Remarks:** To get a series, try and get as many females as is possible. Compared to other skipperlings ova are very large and females are limited to how many ova they can oviposit. Larvae show interesting similarities to some Satyrids.

*Copaeodes minima*

**Lab Host:** *Phalaris arundinacea*

**Remarks:** Females will oviposit in the lab.

*Piruna pirus*

**Host:** *Bromus inermis*

**Lab Host:** *Phalaris arundinacea*

**Remarks:** Females seem to be fussy about ovipositing in the lab in numbers. Larvae feed through rapidly to adult in the lab even though they diapause in nature.
Fig 1-2. *A. julia browningi* and *P. eurymedon* females nectar on honey water. **Fig 3.** *D. plexippus* female oviposits on *Asclepias speciosa*. **Fig 4.** *A. sara* female oviposits on *Arabis perennans* (Dennis Walker). **Fig 5.** Ovum of *A. julia browningi* on *Arabis* sp. **Fig 6.** Ovum of *M. siva chalcosiva* on *Juniperus osteosperma*. **Fig 7.** Female *A. eulalia* set up in a screen cage with cuttings of *Quercus gambelii*. **Fig 8.** Female *C. sheridani neoperplexa* set up to oviposit in a squat tub with *Eriogonum racemosum*. **Figs 9-12.** Last instar caterpillars of *C. affinis affinis*, *P. bairdi*, *P. indra minori*, *A. cethura pima*, and *T. leanira wrighti*. **Fig 13.** *L. lorquini burrisoni* 3rd instar caterpillar crawls out of its hibernaculum. **Fig 14.** Open terrarium setup for *Colias meadi* larvae under 24 hours of light (Nicky Davis). **Fig 15.** Potted grass setup for *E. magdalena* larvae. **Fig 16.** *P. eurymedon* pupa.
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Observations on *Anthocharis julia browningi* and *Anthocharis thoosa thoosa*
Including Tension Zones near Nephi, Juab County, Utah

**TODD STOUT**

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**ABSTRACT:** Two visibly distinct taxa of the sara orangetip fly sympatrically and synchronically in a zone northeast of Nephi, Juab County, Utah. This paper discusses how these two taxa interact based upon six character sets—adult phenotype, adult male behavior, distribution, habitat, larval coloration, and pupal shape.

**Introduction**

There currently are different schools of thought regarding how many species there are in the *Anthocharis sara* complex (sara orangetip). The purpose of this report is to discuss a small piece of that puzzle by sharing observations in the distribution, habitat, behavior, adults, and immature stages of *Anthocharis julia browningi* and *Anthocharis thoosa thoosa* (Fig. 1.) More importantly, this report will also provide a historical account of tension zones between these two taxa in Northern Utah as well as present a closer in-depth study carried out during the spring of 2009 near Nephi, Juab County.

Because of my research in progress on much of the *Anthocharis sara* complex, *browningi* is currently treated as a subspecies of the southern rocky mountain orangetip (*Anthocharis julia*) based upon larval and pupal characters as well as adult blend zones amongst this and other races of *A. julia*. At the same time, the southwestern orangetip (*Anthocharis thoosa*) is treated as a distinct species consistent with Opler (1999), because of adult, larval, and distributional differences with *A. sara*.

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*Fig 1. Adult reared series of A. julia browningi (left) and A. thoosa thoosa (right) from Northern Utah.*
**Anthocharis julia browningi in Utah**

**General:**

The Type Locality of *Anthocharis julia browningi* is City Creek Canyon, Salt Lake County, Utah; Skinner, 1905. Topotypical *A. j. browningi* is phenotypically very similar to other Wasatch Front populations of this butterfly. In both sexes, the dark markings are reduced or washed out as compared to *A. julia stella* and *A. julia julia*. The background color in males is off white with a slight yellowish tint that can be more pronounced in some individuals in Cache County populations. The background color in females is a light yellow. The discal cell bar is narrow, offset, and frequently disconnected with the bottom black apical border. This subspecies has been recognized as being a race of *A. sara*. However, consistent larval differences between this and other races of *A. sara* and *A. thoosa* coupled with consistent larval similarities with ssp. *julia, sulfuris, stella, flora,* and *alaskensis,* place *browningi* as a subspecies of *julia.*

**Utah Distribution and Habitat:**

In Utah, *browningi* flies throughout most of the Wasatch Range from Box Elder to Juab County as well as the Bear River Range found in the extreme northeastern corner of the state. Intermediates between *browningi* and nominotypical *julia* have been taken by Alan Myrup in the Uinta Mountains. *A. j. browningi* also flies in the San Pitch Mountains of Juab County and in Carbon County in Price Canyon.

**Bionomics:**

Host plants for *browningi* include many rock cresses including *Arabis sparsiflora* var. *subvillosa, Arabis perennans, Arabis microphylla, Arabis drummondii,* and *Descurainia pinnata* (tansy mustard.) Oviposition has also been observed on *Arabidopsis thaliana.* Larvae will readily accept *Isatis tinctoria* (dyars woad) in the lab. With the exception of dyars woad, most of these larval host plants can be found either between rocks (*Arabis* spp.) or taking refuge under trees (*Descurainia pinnata.*)

The ova are white turning orange after 24 hours; hatching in about 4-5 days. First instar larvae are cannibalistic and will consume other pierid ova if it finds them. The young first instar larva is light colored with a dark head. Second and third instar larvae of *browningi* are greenish. The fifth instar larva is light green and is pictured below (Fig. 2) and darkens as it approaches pupation. Hibernation is as pupa.

![Last instar larva of Anthocharis julia browningi from the Type Locality located at City Creek Canyon, Salt Lake City, Salt Lake County, Utah.](image)

Fig 2. Last instar larva of *Anthocharis julia browningi* from the Type Locality located at City Creek Canyon, Salt Lake City, Salt Lake County, Utah.
**Anthocharis thoosa thoosa in Utah**

**General:**

The Type Locality of *Anthocharis thoosa thoosa* is Mokiak Pass, Mojave County, Arizona; Scudder, 1878. The amount of dorsal yellow coloration in females seems to be greater with southern Utah populations as compared to northern ones. This butterfly is at home in great basin pinyon-juniper habitat where males patrol up and down ravines or dry washes occasionally leaving the gully to investigate nearby *Juniperus osteosperma* trees in search of females. Females can be found flying in association with Juniper trees either resting or ovipositing on one of its larval host plants, *Descurainia pinnata*, which take refuge under or near the same Juniper trees. Females also oviposit on rock cresses from the genus *Arabis*.

**Utah Distribution and Habitat:**

The general distribution of *thoosa* in Utah ironically forms a general U shape surrounding and circumventing the Wasatch Range and the distribution of *browningi*. In the northwestern part of the state, *A. thoosa* flies in Utah’s Great Basin and West Desert Ranges south to Utah’s Dixie located in the southwestern corner of the state. Populations then extend east towards the four corners region of the state and then north again towards the East and West Tavaputs Plateau. Eastern Utah populations are designated as *Anthocharis thoosa colorado*. See Scott and Fisher (2008).

Current studies of adults, larvae, and pupae of topotypical *colorado* from McElmo Creek, Montezuma County, Colorado, may place it as a junior synonym to nominotypical *thoosa*. To date, my furthest documented northeastern population of *thoosa* is Slaughter Canyon, near Sunnyside, Carbon County, Utah. As you head north, populations of this butterfly from Eastern Utah are replaced by *Anthocharis julia* in the Uinta Range in Summit and Daggett counties in the NE section of the state.

**Bionomics:**

Host plants for *A. t. thoosa* include *Descurainia pinnata, Arabis perennans, Arabis holboelli*, and other species of *Arabis*. The young first instar larva is light orange colored with a dark head. Second and third instar larvae of *A. thoosa* are greenish. The fifth instar larva is green and is pictured below (Fig 3). Hibernation is as pupa. Because the butterfly flies in more or less xeric habitat, most pupae will bypass emergence after one year and will emerge during the second, third, or fourth year of diapause.

![Last instar larva of Anthocharis thoosa thoosa](image)

Fig 3. Last instar larva of *Anthocharis thoosa thoosa* from the Type Locality located at Mokiak Pass, Mohave County, Arizona.
Differences in Immatures

**Larvae:**

Because last instar caterpillars of many species of the pierid tribe Euchloini change color as they advance through their last instar, it is imperative that any larval coloration comparisons between taxa be made from the same timeframe after a fourth instar larva molts to fifth instar. For my larval comparison studies, I compare larvae that have been fifth instars for 54 - 60 hours because this interval demonstrates the most visible, consistent differences to the naked eye.

Fifth instar browninsi larvae have a broader white lateral stripe and the ground color is lighter green as compared to thoosa (Fig. 4.) At the same time, to the naked eye, the transitional color change from the white lateral strip to the green base color of a browninsi larva is much more subtle as compared to thoosa larvae. A. t. thoosa larvae show larger green pinacula surrounding the setae or tubercle giving the larva the appearance of being a much darker green to the naked eye. The pinacula of browninsi fifth instar larvae also enlarge and darken as the larva progresses towards pupation; but do so 12 to 24 hours later.

These consistent differences between last instar caterpillars of these two taxa are also applicable when comparing other subspecies of A. thoosa to A. julia. (This is not just a consistent phenomenon in comparing thoosa to browninsi.) Interestingly, coloration of fifth instar larvae of topotypical A. sara sara and topotypical A. sara pseudothoosa are consistently a much darker green above the white lateral stripe than those of A. julia or A. thoosa; irrespective of the darkening of the pinacula surrounding the setae.

**Pupae:**

The shape of browninsi pupae are consistently different to those of thoosa even though color differences, whether green or tan are not reliably consistent. The main difference is that the browninsi pupal cone bends back whereas those of thoosa are erect and upright (Fig. 4.) These pupal differences are only consistent on a population or subspecific level; but, not necessarily a specific level. For example, pupae of A. julia sulfuris and A. julia flora tend to have pupal cones that bend back and are longer than those of A. julia julia, A. julia browninsi, and A. julia Stella. (Pupal cones of A. julia alaskensis are erect; similar to A. thoosa.) At the same time, pupal cones of A. thoosa inghami are similar in shape to those of A. thoosa thoosa; but, the cone is slightly longer and sometimes slightly tilted back.

**Cross section Comparison of Fifth Instar Larvae**

| A. thoosa thoosa | A. julia browninsi |

**Comparison of Pupae**

| A. julia browninsi | A. thoosa thoosa |

Fig. 4. Comparison of the mid segments of same-age fifth instar larvae (left) and pupae (right) of A. t. thoosa and A. j. browninsi. Oddly enough, color larval differences appear more noticeable to the naked eye or when you squint your eyes—which is how I noticed their differences in the first place.
Differences in Adult Characters

There are a few contact or tension zones of different taxa within the *Anthocharis sara* complex including but not limited to *A. sara* and *A. julia stella* near Fresno Dome, Madera County, California (Ken Davenport, personal communication), *A. sara sara* and *A. julia stella* in Northern California—see Geiger and Shapiro (1986), *A. sara* Siskiyou segregate and *A. julia* nr. *stella* at Klamath River Canyon, Klamath County, Oregon (Andy Warren, personal communication), as well as *A. julia* and *A. thoosa colorado* near the Four Corners Region—see Scott and Fisher (2008). Scott also references several other contact zones in California, Colorado, and New Mexico.

The best opportunity to visibly note possible intermediates between two sympatric taxa was to select those that visibly looked the most distinct from each other. *A. julia browningi* and *A. thoosa thoosa* seemed to fit these criteria. (See table and Figure 5 below.)

<table>
<thead>
<tr>
<th>Distinguishing Adult Characters</th>
<th><em>Anthocharis julia browningi</em></th>
<th><em>Anthocharis thoosa thoosa</em></th>
<th><em>A. julia browningi</em> x <em>A. thoosa thoosa</em></th>
</tr>
</thead>
<tbody>
<tr>
<td>Ground Color in Males</td>
<td>Off White</td>
<td>White</td>
<td>White</td>
</tr>
<tr>
<td>Ground Color in Females</td>
<td>Light Yellow</td>
<td>White with some Yellow</td>
<td>Unknown</td>
</tr>
<tr>
<td>Thickness of Discal Cell Bar</td>
<td>Narrow</td>
<td>Wide</td>
<td>Intermediate</td>
</tr>
<tr>
<td>Coloration of Orange Tip</td>
<td>Bright orange</td>
<td>Brighter Orange</td>
<td>Intermediate</td>
</tr>
<tr>
<td>Ventral Hindwing Mottling</td>
<td>Medium Green</td>
<td>Dark Green/Gray</td>
<td>Intermediate</td>
</tr>
<tr>
<td>Dorsal Forewing Apical Black Borders</td>
<td>Somewhat faded</td>
<td>Dark</td>
<td>Dark</td>
</tr>
<tr>
<td>Alignment of Orange Tip to Discal Cell bar</td>
<td>Offset</td>
<td>Slightly Offset (but hard to tell based upon thickness of bottom black margin)</td>
<td>Offset</td>
</tr>
<tr>
<td>Connecting of Bottom Black Margin to Discal Cell Bar</td>
<td>Usually Disconnected</td>
<td>Usually Connected</td>
<td>Barely connects / Intermediate</td>
</tr>
</tbody>
</table>

Fig 5. Dorsal and ventral examples of an *Anthocharis julia browningi* male (left), *Anthocharis thoosa thoosa* male (middle) and hybrid male (right) from 2009 study area in Juab County, Utah.
Previous Northern Utah Contact Zones

Willow Creek; 3.0 miles ESE of Mona, Juab County

My personal interest in discovering contact zones of *A. j. browningi* and *A. t. thoosa* was piqued on 7 May 1997, when Bob Hardbarger discovered both taxa flying together at Willow Creek; 3.0 miles east of Mona, Juab County, Utah. Three days later, Bob, Steve Sommerfeld and I returned to Willow Creek and confirmed Bob’s finding when I vouchered several *Anthocharis* males—some of those were *thoosa* and the rest were *browningi*. (I do not have exact numbers as those specimens were later sent to Paul Opler for research and study.) However, we did not locate any apparent intermediates or hybrids on that day.

Subsequent trips to Willow Creek in 2006, 2007, and 2008 were somewhat frustrating as the dominant taxon was *A. j. browningi*. All males and females collected during this timeframe were *browningi*. However, I also collected and reared several eggs and caterpillars to adult—all *browningi*; except two were *thoosa*. I obtained no intermediates from reared material.

Deep Creek Canyon; 3.6 miles South of Levan, Juab County

On 27 May 1999, Steve Spomer, Jim Reiser, and I found mostly *A. t. thoosa* and a few *browningi* flying together at Deep Creek Canyon, Juab County. Of the males I collected that day, one appeared to have intermediate characters between *thoosa* and *browningi* (Fig. 6.) The rest were parentals. We also found a population further up the canyon where patrolling males were *browningi*.

![Fig 6. Male of an apparent hybrid of *Anthocharis thoosa thoosa* x *Anthocharis julia browningi* taken on 27 May 1999 at Deep Creek Canyon, Juab County, Utah.](image)

Gardner Creek; 2.7 miles NNE of Nephi, Juab County

Because *A. j. browningi* was the dominant taxon at Willow Creek in 2007, I decided to investigate Gardner Canyon—which was typical “thoosa” habitat located 2.5 miles south of Willow Creek and 2.7 miles NNE of Nephi. On 14 Apr 2007, because it was a relatively dry year for that region, I only collected two *thoosa* males, 1 *thoosa* female, six ova on *Descurainia pinnata* (which were reared to adult and turned out to be *thoosa*) and one apparent hybrid (Fig 7); but did not observe or collect any parental *browningi*.

In a tension zone, the key to finding *thoosa* and *browningi* flying sympatrically may have a lot to do with habitat. The mouth of this canyon, even though officially part of the Wasatch Mountains, was typical Great Basin habitat ubiquitous with *Artemisia tridentata*, *Juniperus osteosperma* and *Purshia mexicana*. 
Fig 7. Male of a possible hybrid of *Anthocharis thoosa thoosa* × *Anthocharis julia browningi* taken on 14 Apr 2007; Gardner Canyon; 2.7 miles NNE of Nephi, Juab County, Utah.

**Rock Canyon; 2 miles East of Provo, Utah County**

On 17 April 2004, after a Utah Bug Club meeting held at the Monte L. Bean Life Science Museum at BYU, I took students on a field trip and collected several males of *A. j. browningi* as well as 2 males of *A. t. thoosa* flying in what was previously considered to be a “browningi-only” population in Rock Canyon, just east of BYU in Provo, Utah County, Utah. These two *thoosa* males were examined and appeared to be pure parental *thoosa* without any visible evidence of gene exchange with *browningi*.

**2009 Study Area**

During the winter of 2009, because I located two study areas within 2.5 miles of each other where either one taxa or the other dominated in northeastern Juab County, I decided to take a closer look at other canyons, draws, and/or gullies between these two areas where I might find both taxa flying sympatrically and synchronically.

I decided to study three small accessible canyons—Birch Creek, Little Birch Creek, and an unnamed draw several hundred feet south of Little Birch Creek. These study areas (Fig. 11) were almost nestled between Willow Creek to the north (where *browningi* dominates) and Gardner Creek to the south (where *thoosa* dominates.)

**Birch Creek; 3.7 miles SSE Mona; 4.0 miles North of Nephi, Juab County**

On 20-21 Apr 2009, I visited Birch Creek and found several *A. t. thoosa* males patrolling along the base of the canyon flying in typical *Juniperus osteosperma* habitat at an elevation of 5400 feet (Fig. 11.) As I hiked about 1,000 feet to the east up Birch Creek to an elevation of 5585 feet, I noted the subtle difference in habitat change where oaks and maples replaced Juniper trees. It was here where I collected three patrolling *browningi* males and one apparent hybrid male.

About fifty feet away from where the males were flying, I collected a female (Fig. 8.) My initial impression was that she was a hybrid based upon her proximity to *browningi* males, smaller discal cell bar than typical *thoosa*, faded dark markings (which turned out to be the result of age), and weak flight. However, of the 25 eggs she laid; 16 were reared to pupae where larvae and pupae conformed to *thoosa*; without showing any noticeable *browningi* characters.
Little Birch Creek; 4.2 miles SSE Mona; 3.6 miles North of Nephi, Juab County

Although I was unable to locate *A. t. thoosa* and *A. j. browningi* flying in the exact same spot at Birch Creek, I was able to collect 4 male *browningi*, 9 male *thoosa*, and 4 apparent male hybrids patrolling sympatrically and synchronically at Little Birch Creek at an elevation of 5400 feet (Fig. 9.) This might have been the case because this ravine was so narrow that it didn’t really separate *browningi* from *thoosa* habitat.

It was interesting to note the behavioral differences in the patrolling males at Little Birch Creek. *A. j. browningi* males tended to fly much slower than *A. t. thoosa* males. Also, *thoosa* males left the ravine from time to time to investigate nearby Juniper trees in search of females; before scampering back to the ravine to patrol. Oddly enough, it seemed to me that the behavior of the hybrid males was either slow and deliberate (*browningi*) or fast and scampering (*thoosa*).

Approximately 250 feet below the ravine, I collected two female *thoosa* flying amongst the Juniper trees which laid eggs in the lab on native host Descurainia pinnata. Both the larvae and pupae of those immatures were analyzed as *thoosa* without any visible evidence of *browningi* immature traits.

I had hoped to find ova on several plants of *Arabis perennans* growing in the outcroppings near the ravine where the males of both species were flying; but didn’t find any pierid immatures on them; including those of *Pontia sisymbri nigra venosa*. 

Fig 8. Three *A. j. browningi* males (left); one hybrid male and one faded/worn female *A. thoosa thoosa* (right) collected at Birch Creek on 20 Apr 2009.
Fig 9. I collected a total of four *browningi*, nine *throosa*, and four apparent hybrid males on 20-21 April, 2009 at Little Birch Creek, Juab County, Utah. (If you have an electronic copy of this file, you can examine these specimens more closely by zooming in.)

**Unnamed Draw: 4.3 miles SSE Mona; 3.4 miles North of Nephi, Juab County**

I didn’t spend as much time in the draw located roughly 0.1 miles south of Little Birch Creek; but, I was able to collect 1 male *A. thoosa thoosa* and 1 male *A. julia browningi* flying there (Fig 10.)

Fig 10. Male *browningi and thoosa* collected at the unnamed draw located 0.1 miles south of Little Birch Creek
Review

During my 2009 study of contact zones of *Anthocharis julia browningi* and *Anthocharis thoosa thoosa* between Mona and Nephi, Juab County, Utah, I noted that of the 23 collected males, 43.5 percent were parental *A. t. thoosa*, 34.8 percent were parental *A. j. browningi*, and 21.7 percent showed apparent intermediate traits. I also collected three females who were parental *thoosa* where all offspring and emerged adults showed notable *thoosa* characters; although at press time, these have not yet been spread.

The fact that the percentage of hybrids in this study area is roughly 22 percent with the rest showing parental traits coupled with a narrow overlap in distribution, consistent differences in habitat preference, male adult flying behavior, larval coloration characters and pupal shape characters, suggest that subspecies *thoosa* and *browningi* belong to different species.

Also, outside of noticeable adult characters, there may be other factors that might show a higher percentage of gene exchange using better available technologies such as electrophoresis (Geiger & Shapiro, 1986), or nuclear dna studies.
Acknowledgments

Special thanks are given to Jon Pelham, Dr. Andy Warren, Paul Opler, and Harry Pavulaan for reviewing this paper and to Norbert Kondla for his help with plates. Also, because of information shared with James A. Scott in Papilio #18, I did not reference Scott who was referencing me regarding larval studies; but have referenced original material from James A. Scott. Acknowledgment also goes to COL. Clyde F. Gillette, who has proven records of either browningi or thoosa in every county in Utah.

Bibliography


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The Nomenclatural Status of Ten Names in the Genus *Pieris* (Lepidoptera: Pieridae)

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Abstract: The nomenclatural status of ten names in the genus *Pieris* are reviewed. The dates and locations of publication of *pseudobryoniae* Verity, 1908 and *adalwinda* Fruhstorfer, 1909 are corrected; they are infrasubspecific unavailable names. The names *pseudobryoniae* Barnes and McDunnough, 1916 and *arctica* Barnes and McDunnough, 1916 are newly identified as available species-group names with at least four and six syntypes respectively. A lectotype of *pseudobryoniae* Barnes and McDunnough is selected, with the type locality being Nulato, Alaska; *browni* Eitschberger, 1983 is a subjective synonym. A lectotype of *arctica* Barnes and McDunnough is designated, with the type locality being northern Norway. The name *arctica* Verity, 1911 is an infrasubspecific unavailable name. The date and location of publication of *pseudonapi* Verity, 1909 is corrected; it is an available name with *pseudonapi* Barnes & McDunnough, 1916 a primary homonym. The name *macdunnoughii* Remington, 1954 is the correct and original spelling; Miller and Brown (1981) provide the incorrect subsequent spelling *mcduanoughi*. The name *passosi* Warren, 1968 is an available species-group name. A lectotype for *passosi* is designated; the name *meckiae* Eitschberger, 1983 is a subjective synonym. A lectotype is designated for *pallidissima* Barnes and McDunnough, 1916. The name *angelika* Eitschberger, 1981 is a nomen nudum, but *angeliuka* Eitschberger, 1983 is an available name. We suggest placing two taxa as subspecies of *P. angelika*, resulting in the combinations *P. angelika shelutzikoi* Eitschberger, 1983 and *P. angelika schinflmeisteri* Eitschberger, 1983. The publication date of Ulf Eitschberger’s *Systematische untersuchungen am Pieris napi-bryoniae-komplex (s. I.) (Lepidoptera, Pieridae)* is determined to be sometime in December 1983, nominally placed as 31 December 1983.

Additional key words: butterfly, nomenclature, margined white, arctic white.

INTRODUCTION

The North American populations of butterflies in the genus *Pieris* have had numerous species- and subspecies-level names applied to them through history. This paper focuses on the nomenclatural status of ten names potentially applicable to North American *Pieris*, and establishes the date of publication of Ulf Eitschberger’s book *Systematische untersuchungen am Pieris napi-bryoniae-komplex (s. I.) (Lepidoptera, Pieridae)*. A few minor taxonomic issues are also addressed. The “available” names identified in this paper will be used during future taxonomic revisions, as part of determining the valid name for each taxon.

The provisions of the *International Code of Zoological Nomenclature* (International Commission on Zoological Nomenclature (ICZN) 1999; henceforth referred to as the “Code”) are the basis for nomenclatural conclusions. The Code is not “law”, but is the carefully codified recommendations of a respected worldwide group of taxonomists. In this review we apply Code provisions to ensure that future scientific discussions can be based on internationally accepted criteria.
The topics addressed in this paper, with their page numbers, are:

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2. **ADALWINDA FRUHSTORFER, 1909** .......................................................... 4
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12. **SUMMARY OF CONCLUSIONS** ................................................................. 16

**ANALYSIS OF THE NAMES**

1. **PSEUDOBRYONIAE VERITY, 1908**

Kudrna (1983) catalogued (p. 60) this name as:

**pseudobryoniae (fm) – Pieris napi frigida pseudobryoniae Verity, 1908 – 010:**


where the “(fm)” indicates that Kudrna considered the taxon to be an infrasubspecific form. We agree with Kudrna’s conclusion but the nomenclatural saga of the word “pseudobryoniae” does not end there, as detailed below.

Verity described *Pieris napi* var. *frigida* form *pseudobryoniae* on page 146 [publication date 31 Jan 1908] of Verity (1905-1911), with the phrase

“certain exemplaires se rapprochent cependant de *bryoniae* par leurs nervures larges et diffuses et meritent le nom de *pseudobryoniae* (fig. 36 et 37).”

This can be translated to English as

“Certain examples nevertheless compare themselves to *bryoniae* by their wide and diffuse veins and merit the name of *pseudobryoniae* (fig. 36 and 37).”

The species level taxonomy of “*Pieris napi*” and the variety “*frigida*” are clearly stated by Verity, and he indicates that the name *pseudobryoniae* applies only to certain individual examples of var. *frigida*. The name *pseudobryoniae* Verity, 1908 is therefore infrasubspecific, and is not available under the Code, Article 10.2:

> **10.2. Availability of infrasubspecific names.** An infrasubspecific name is not available [Art. 45.5] from its original publication, unless it was published before 1961 for a “variety” or “form” and is deemed to be available under Art. 45.6.4.1. If an author uses a name, previously published at infrasubspecific rank, in a way which makes it available for a species or subspecies, that author thereby establishes it as a new name and it takes his or her authorship [Art. 45.5.1] (see also Articles 23.3.4 and 50.3.1)

Several other Articles are cited in Article 10.2. Article 45.5 includes the statement that “A fourth name published as an addition to a trinomen automatically denotes an infrasubspecific entity”. Article 45.6.4.1 is only applicable if the name is deemed by the Code to not be infrasubspecific under Article 45.6.4, which is not the case for *pseudobryoniae* Verity, 1908. The application of Articles 45.5.1, 23.3.4 and 50.3.1, which deal with the consequences of a later author using an infrasubspecific name at the subspecies or species level, are not relevant to the availability of

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1 Publication dates for parts of Verity (1905-1911) are provided by Kudrna (1983), which is in part based on Verity (1914).
pseudobryoniae Verity, 1908, as discussed in the next section. Therefore, the name pseudobryoniae Verity, 1908 is an infrasubspecific name not available for taxonomic use under the Code.

Verity’s figures of two specimens of pseudobryoniae, with the associated figure captions were published on 30 Apr 1909 as Plate XXXII Figures 36, 37. Neither specimen of pseudobryoniae Verity, 1908 was identified as the “type” in the original description. However, Verity (1905-1911) starts with an Index to all taxa in the work; the Index has the publication date of 31 Oct 1911. The explanatory heading on page XIII of the Index is shown in Figure 1, and the entry on page XXVIII for the species Pieris napi, subspecies frigida, race arctica, form pseudobryoniae is shown in Figure 2. The explanatory heading for the Index (Fig. 1) states that an asterisk, as in “XXXII, 37*”, specifies that figure 37 is of the “type” specimen. Therefore, this index entry is the designation, by Verity in 1911, of the specimen illustrated in his Plate XXXII Figure 37 (Fig. 4, 5) as the “type” of form pseudobryoniae.

INDEX SYSTÉMATIQUE ET TABLEAU SYNOPTIQUE
DE LA VARIATION ET DE LA DISTRIBUTION GÉOGRAPHIQUE

<table>
<thead>
<tr>
<th>GENRE</th>
<th>espèce</th>
<th>sous-espece</th>
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<th>forme (morpha)</th>
<th>aberration</th>
<th>aberration abortive ou strictement pathologique</th>
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Figure 1. Heading to the Index (Verity 1905-1911).

Figure 2. Index entry for pseudobryoniae (Verity 1905-1911).
[the phrase “[XLVII, 16 *-17” pertains to the previous line of the legend, taxon arctica]

Verity’s index + text + illustrations that were published on 31 October 1911 are all part of the same work, therefore the index is part of the original descriptions of the new taxa that were named at that time. Use of the asterisk to indicate "type" is “holotype” designation for the new taxa published 31 October 1911. However, this particular “type” was designated in 1911, three years after the 1908 description of the taxon. Hence it would be the designation of a “lectotype” by Verity, if the Code applied to an infrasubspecific name (which it does not). This “lectotype” is a specimen from “Finmark, Scandavnie”, which is the type locality. According to Kudrna (1983), “Finmark” corresponds to northern Norway. However, this “lectotype” designation by Verity was actually without meaning because it was preceded by a “lectotype” designation by Fruhstorfer (1909), as discussed below. In any case the “lectotype” designation is moot, because pseudobryoniae Verity, 1908 is an infrasubspecific name and hence is unavailable under the Code; hence terms such as “lectotype” are not actually applicable.
2. **ADALWINDA FRUHSTORFER, 1909**

Fruhstorfer (1909), after seeing Plate 32 of Verity (1905-1911) that was published on 30 Apr 1909 (see above), determined that Verity’s two illustrated specimens represent two separate taxa:

"Unter dem Namen pseudobryoniae vereigt Verity pag. 146 und t. 32 f. 36 und 37 zwei heterogene Rassen aus Alaska (Type) und Finnmarken. Letztere ist viel großer und steht natürlich der alpinen bryoniae O., namentlich der f. obsoleta Rober viel naher als der nordamerikanischen Polarform. Für unsere nordische Rasse, charakterisiert durch seissliche Grundfarbe und im distalen Teil der Vorderflügel viel dunklere braune Flecke führe den Namen adalwinda ein."

This can be translated to English as:

“Under the name *pseudobryoniae* Verity, page 146 and Plate 32 Figures 36 and 37, is combined two heterogeneous races from Alaska (Type) and Finland. The latter is much larger and stands naturally much closer to the alpine *bryoniae* O., namely *f. obsoleta* Röber, than to the North American polar form. For our northern race, characterized through whitish ground colour and in the distal part of the forewings much darker brown spots, is given the name *adalwinda*."

Fruhstorfer (1909) designates the specimen illustrated in Verity’s Figure 36 (from Nulato, Alaska) as the “type” of *pseudobryoniae* Verity, 1908. This “type” is the first “lectotype” of *pseudobryoniae*, with publication date priority over the “lectotype” designation of Verity (1911), which was discussed above. This is of no importance, given that *pseudobryoniae* Verity, 1908 is an unavailable infrasubspecific name.

Fruhstorfer (1909) called *pseudobryoniae* Verity a “race” from Alaska, and he clearly considers *pseudobryoniae* Verity to have the same taxonomic rank as what he names as race *adalwinda*. A “race” named at that time is equivalent to “subspecies” under Article 45.6 of the Code unless there is clear evidence to the contrary. There are two pieces of information that provide such contrary evidence. First, in the remainder of his paper Fruhstorfer (1909) proceeds to describe four new subspecies in the genus *Pieris*, using the headings “*Pieris napi leovigilda* nov. subspec.”, “*Pieris napi nesis* nov. subspec.”, “*Pieris rapae micipsa* nov. subspec.” and “*Pieris rapae lysicles* nov. subspec.”.
subspec.” He therefore clearly and consistently used the genus-species-subspecies trinomial concept, with explicit use of the term “subspecies” in contrast to his previous use of “race”. Second, Fruhstorfer references race *pseudobryoniae* Verity in the same taxonomic context as when he names race *adalwinda*, and hence the taxonomic placement of *pseudobryoniae* and *adalwinda* is that used by Verity – as a quadrinomial infrasubspecific taxon. These two lines of evidence demonstrate that Fruhstorfer deliberately and consistently uses the term subspecies in a trinomial name, and used “race” for *pseudobryoniae* and *adalwinda* as a quadrinomial infrasubspecific taxon.

The names *pseudobryoniae* and *adalwinda* Fruhstorfer, 1909 are therefore not available as species-group names because they are infrasubspecific names. We leave it to other taxonomists to decide whether to apply to the International Commission on Zoological Nomenclature to take action to conserve the name *adalwinda* Fruhstorfer, 1909 in the interest of nomenclatural stability, as recommended by Kudrna (1986).

### 3. *PSEUDOBRYONIAE* BARNES AND MCDUNNOUGH, 1916

Barnes and McDunnough (1916) reviewed the North American *Pieris*, and stated:

“In the extreme north [of North America] three distinct forms are separable; in the inland Arctic region (Barren Plains) we have the form arctica Verity with strongly blackish marked veins on the underside in both sexes and on the upper side in the ♀; there is however no suffusion of black and the markings are clear cut; we figure a ♂ and ♀ from Chatanika, Alaska (Figs. 6, 7). Along the Alaskan coast we meet with the form *pseudobryoniae* Verity which is what has been considered until recently to be *bryoniae* Ochs., a race now restricted to the Alps of Europe; Wright’s figures (Butt. W. Coast Pl. VI, Fig. 43b and 42bb) are typical of the variation of the ♀. On the numerous islands of the Behring Sea and Alaskan coast the form *hulda* Edw., is found in which the secondaries on the under side are almost totally suffused with greenish in the ♂ sex, leaving only dashes of yellowish ground color; the ♀’s are usually less suffused and on the upper side are intermediate between arctica and pseudobryoniae; we figure a ♂ underside and ♀ upperside (Figs. 8, 9).”

They do not illustrate *pseudobryoniae*, however in the figure captions they treat *arctica* and *hulda* as subspecies names. Furthermore they state that “along the Alaskan coast” Wright’s figures “are typical of the variation [of *pseudobryoniae*]”. They clearly considered *pseudobryoniae* to be a geographic subspecies that occurs along at least part of the Alaskan coast, at the same taxonomic level as subspecies *arctica* and subspecies *hulda*. In this paragraph they are using the word “form” with the generalized meaning of “phenotype”, not “form” in the taxonomic sense.

This is relevant in relation to Code Article 45.5.1:

> **45.5.1** A name that has infrasubspecific rank under the provisions of this Article cannot be made available from its original publication by any subsequent action (such as "elevation in rank") except by a ruling of the Commission. When a subsequent author applies the same word to a species or subspecies in a manner that makes it an available name [Arts. 11-18], even if he or she attributes authorship of the name to the author of its publication as an infrasubspecific name, that subsequent author thereby establishes a new name with its own authorship and date.”

The first sentence means that an infrasubspecific name can only be elevated in rank through a ruling of the Commission.

(a) therefore *pseudobryoniae* Verity cannot be “elevated in rank” by Barnes and McDunnough (1916); and

(b) the action taken by Barnes and McDunnough (1916) is the establishment of a new name, not the elevation of Verity’s name.

The second sentence says that:

(a) the same word that was used for the infrasubspecific name can be used by a new author, to establish a new name;

(b) the establishment of the new name must conform to Articles 11-18; and

(c) it is irrelevant whether the new author thinks he is using someone else’s species-group name.

The name *pseudobryoniae* Barnes and McDunnough, 1916 meets the requirements of Articles 11-18, of which Articles 13-18 are not relevant. All the provisions of Article 11 are met. For Article 12, Barnes and McDunnough (1916) do not provide even the slightest hint of a description or definition. However, Barnes and McDunnough
provide a clear indication by bibliographic reference to specific illustrations in a specific publication by Wright (Article 12.2.7); therefore the specimens represented by those illustrations, reproduced in Figure 3, are syntypes of *pseudobryoniae* Barnes and McDunnough, 1916 and the name is available through that indication. The relevant parts of Article 12 are:

**Article 12. Names published before 1931.**

**12.1. Requirements.** To be available, every new name published before 1931 must satisfy the provisions of Article 11 and must be accompanied by a description or a definition of the taxon that it denotes, or by an indication.

**12.2. Indications.** For the purposes of this Article the word "indication" denotes only the following:

12.2.1. a bibliographic reference to a previously published description or definition even if the description or definition is contained in a work published before 1758, or that is not consistently binominal, or that has been suppressed by the Commission (unless the Commission has ruled that the work is to be treated as not having been published [Art. 8.7])

12.2.7. the proposal of a new genus-group name or of a new species-group name in association with an illustration of the taxon being named, or with a bibliographic reference to such an illustration, even if the illustration is contained in a work published before 1758, or in one that is not consistently binominal, or in one that has been suppressed by the Commission (unless the Commission has ruled that the work is to be treated as not having been published [Art. 8.7])

The reference to “*pseudobryoniae* Verity” in the above quotation is not a clear indication in itself. However, the first line of the Barnes and McDunnough’s (1916) treatment of *Pieris napi* states: “Verity has lately (Rhop. Pal. Vol. I) dealt at considerable length with the various races and forms of this species; we offer the following remarks as to the arrangement of our North American races as it is probable that Verity’s work is inaccessible to the majority of American entomologists”. Hence, Barnes and McDunnough gave an adequate bibliographic reference for *pseudobryoniae* Verity was based (Figure 4) are also syntypes of *pseudobryoniae* Barnes and McDunnough, 1916 and the name is also available through that indication.

Barnes and McDunnough (1916) also imply that they examined other specimens that they considered to be *pseudobryoniae*, if so, those specimens are also syntypes (Article 72.4.1). The location of these syntypes, if they are identifiable, is unknown to us.

Therefore *pseudobryoniae* Barnes and McDunnough, 1916 is an available name, and the syntypes are the two specimens illustrated by Verity (1905-1911), plus the two specimens illustrated by Wright (1905), plus any other specimens (identity and location unknown) that Barnes and McDunnough examined and considered to be *pseudobryoniae* (Article 72.4.1).

The type series of *pseudobryoniae* of Barnes and McDunnough likely contains more than one taxon (Eitschberger 1983), therefore a lectotype needs to be designated to provide both taxonomic clarity and foster nomenclatural stability. We therefore designate the specimen illustrated in Plate 32 Figure 36 of Verity (1905-1911), reproduced here in Figure 4, as the lectotype of *Pieris napi pseudobryoniae* Barnes and McDunnough, 1916, with the type locality being Nulato, Alaska. This is consistent with historical, although irregular, use of the name *pseudobryoniae*, with various authors attributed to it.

In our opinion, *Pieris marginalis browni* Eitschberger, 1983 (Type Locality: Seward Peninsula, Alaska) is a junior subjective synonym of *Pieris napi pseudobryoniae* Barnes and McDunnough, 1916. Eitschberger (1983, p. 349) recognized that Plate 32 Fig. 36 represented his taxon *browni*, but did not recognize the availability of the name *pseudobryoniae* Barnes and McDunnough, 1916.

After Barnes and McDunnough (1916), other authors also used the word *pseudobryoniae* as a subspecies-level name and, through indication to Verity (1905-1911), met the Code requirements for to make the name available with their new authorship and date (e.g. dos Passos 1965). These later uses of the name are not available names, because the author of a name is the person who first publishes it (Article 50); in this case Barnes and McDunnough (1916).
4. **Arctica Verity, 1911**

Kudrna (1983) catalogued (p. 60) this name as:


where the “(ra)” indicated that Kudrna considered the taxon to be an infrasubspecific race. We agree with Kudrna’s conclusion but, as with pseudobryoniae, the nomenclatural saga of the word “arctica” does not end there.

Verity described *Pieris napi* var. *frigida* race *arctica* on page 334 [publication date 31 Oct 1911] of Verity (1905-1911), with the sentences:

“Je crois qu’il serait utile de distinguer la race arctique d’Europe par le nom de arctica afin d’éloigner une fois pour toutes la confusion engendrée par le fait que cette race n’a pas été distinguée de celle des Alpes. Mes figures 32 et 33 (Pl. XXII [sic – actually XXXII] auxquelles j’ajoute deux autres types de ♂♀ (Pl. LXVII, fig. 16 et 17) donneront une idée exacte de cette race et la comparaison du ♂ (fig. 32) avec le ♂ alpin (fig. 25) l’en distingue bien.”

This can be translated to English as

“I believe that it would be useful to distinguish the Arctic race of Europe by the name of arctica in order to dispel for once the whole confusion generated by the fact that this race has not been distinguished from the one of the Alps. My figures 32 and 33 (Pl. XXII [sic – actually XXXII]), to which I add two other typical ones (Pl. LXVII, fig. 16 and 17), will give an exact idea of this race and its comparison (Fig. 32) with the alpine one (Fig. 25) will distinguish it well.”

The species level taxonomy of “*Pieris napi*” and the variety “frigida” are clearly stated by Verity, and he indicates that the name *arctica* applies to a race within variety *frigida*. Variety *frigida* is of subspecies rank, by Article 45.6.4 of the Code. The name *arctica* Verity, 1911 is therefore infrasubspecific. This interpretation agrees with the listing in the *Index* (with the same publication date), which also has *arctica* as a fourth order name (Fig. 2, 4). The “holotype” is designated in Verity’s *Index* as Plate XXXII Figure 32, with three “paratypes” (Verity’s Plate XXXII Fig. 33 and Plate LXVII Figs. 16, 17).

A possible concern with this interpretation is Verity’s Plate LXVII and its legends for figures 16 and 17 (Fig. 6 below), which appears to create the name *P. napi* var. *arctica* Verity, 1911. In isolation this *arctica* name is specifically defined as being of subspecies rank by the Code (Article 45.6.4), however the Article includes the qualifying phrase “unless the author also expressly gave it infrasubspecific rank, or the content of the work unambiguously reveals that the name was proposed for an infrasubspecific entity, in which case it is infrasubspecific.” Verity’s *Index*, the text on page 334, and Plate LXVII were all published together on 31 October 1911; hence they constitute a single work. The *Index* and the text on page 334 provide the required evidence that Verity was giving a fourth order (infrasubspecific) rank to the name *arctica*.

The above evidence demonstrates that the name *arctica* Verity, 1911 is infrasubspecific and therefore is not available under the Code (Article 10.2).

| 17. Id. ♀ (Laponie) [e coll. Leech] |

**Figure 6. Plate LXVII legends for arctica** (Verity 1905-1911)

5. **Arctica Barnes and McDunnough, 1916**

Barnes and McDunnough (1916) reviewed North American *Pieris*, and stated:

“In the extreme north [of North America] three distinct forms are separable; in the inland Arctic region (Barren Plains) we have the form *arctica* Verity with strongly blackish marked veins on the underside in both sexes and on the upper side in the ♀; there is however no suffusion of black and the markings are clear cut; we figure a ♂ and ♀ from Chatanika, Alaska (Figs. 6, 7).”
The name *arctica* Barnes and McDunnough, 1916 meets the requirements of Articles 11-18, of which Articles 13-18 are not relevant. All the relevant provisions of Article 11 are met. For Article 12, Barnes and McDunnough (1916) provide a definition and figures of two syntypes from Chatanika, Alaska. However, they also provide an indication to Verity’s original description, including the figures, through the same mechanism discussed above in Section 3 for *pseudobryoniae*. Therefore the specimens represented by the Verity’s illustrations for his *arctica* are also syntypes of *arctica* Barnes and McDunnough, 1916.

Barnes and McDunnough (1916) also imply that they examined other specimens that they considered to be *arctica*; if so, those specimens are also syntypes (Article 72.4.1). Their identity and present location is unknown.

Therefore *arctica* Barnes and McDunnough, 1916 is an available name, and the syntypes are the four specimens illustrated by Verity (1905-1911), plus the two specimens illustrated by Barnes and McDunnough (1916), plus any other specimens (identity currently unknown) that Barnes and McDunnough examined and considered to be *arctica* (Article 72.4.1).

The locations from which the six known syntypes of *arctica* Barnes and McDunnough originate range from Scandinavia to Alaska, and more than one taxon is likely represented; hence designation of a lectotype is required for taxonomic clarity and to stabilize the nomenclature for on-going revisions of the genus *Pieris*. Barnes and McDunnough (1916) considered the name *arctica* to be represented by the “types” of the unavailable name *arctica* Verity. Therefore, we designate the lectotype of *arctica* Barnes and McDunnough, 1916 to be the specimen represented by Plate XXXII Figure 32 of Verity (1905-1911). This is the specimen designated as the “holotype” of *arctica* Verity and shown above (Fig. 4), with the type locality being “Norvège sept.”, which is “Scandinavia: N. Norway” according to Kudrna (1983).

It could be argued that the “holotype” of *arctica* Verity is automatically the holotype of *arctica* Barnes and McDunnough. This would certainly be the case if *arctica* Verity was an available name being replaced by another name due to homonymy. However, the circumstance of a name being available through indication of the description of an infrasubspecific name is not addressed by the Code. Hence, designation of a lectotype that is the same as the putative holotype under an alternative interpretation of the Code (with which we disagree) achieves the objective of nomenclatural stability.

The name *arctica* Barnes and McDunnough, 1916 is the available name for the European populations to which the unavailable name *adalwinda* Fruhstorfer, 1909 is presently applied.

After Barnes and McDunnough (1916), other authors also used the word *arctica* as a subspecies-level name and, through indication to Verity (1905-1911), met the Code requirements for to make the name available with their new authorship and date (e.g. dos Passos 1965). These later uses of the name are not available names, because the author of a name is the person who first publishes it (Article 50); in this case Barnes and McDunnough (1916).

### 6. PSEUDONAPI VERITY

Kudrna (1983, p. 60) catalogued this name as:

**pseudonapi (ra) – Pieris melete melete pseudonapi Verity, 1911 – 010:330 – Japan:**

*Yezo [= Hokkaido]: Ichikiri.*

where the “(ra)” indicated that Kudrna believed that Verity had described the taxon with the rank of “race”, and that the taxon is infrasubspecific and the name is not available (the reasoning behind this conclusion is doubtful in the context of the Code, but does not need to be discussed here). This conclusion suggested that the name *pseudonapi* Barnes and McDunnough, 1916 had been incorrectly determined to be a homonym by Remington (1954), who replaced it with the name *macdunnoughi*. However, Kudrna’s conclusion was incorrect because the text on page 330 of Verity (1905-1911) was not the actual original description of *pseudonapi*.

Verity’s Plate LIX and the associated figure legend (Figures 7 and 8 below) were published on 31 Jan 1911. The plate and figure legend together are a valid original description (Code Articles 12.1 and 12.2.7), and that description has date priority over the 31 Oct 1911 text description on page 330 of Verity. The legend for Plate LIX Figures 13-17 used the term “var.” (variety) to indicate the rank of *pseudonapi*, which the Code specifically states must be considered to be equivalent to subspecies rank (Article 45.6.4), in the absence of clear evidence to the contrary – as
in this case. The name \textit{pseudonapi}, Verity, 1909 is therefore available, and the specimens represented by Verity’s Plate LIX Figures 13-17 are the syntypes.

The name \textit{pseudonapi} Barnes & McDunnough, 1916 is therefore a primary homonym of \textit{pseudonapi}, Verity, 1911, validly replaced by \textit{macdunnoughii} Remington, 1954.

\section*{Figure 7. Copy of the figures of the syntypes of \textit{P. melete} var. \textit{pseudonapi} (Verity, 1905-1911, Plate LIX)\footnote{Figure numbers re-typed for clarity. Figure legends shown above (Figure 7).}}

\begin{tabular}{c}
14. \textit{Id}. \textit{♂} Revers (Ichikishiri, Yesso, Japon) \\
15. \textit{Id}. \textit{♀} (Ichikishiri, Yesso, Japon) \\
16. \textit{Id}. \textit{♀} (Ichikishiri, Yesso, Japon) \\
17. \textit{Id}. \textit{♀} Revers (Ichikishiri, Yesso, Japon)
\end{tabular}

\section*{Figure 8. Copy of the figure captions for \textit{P. melete} var. \textit{pseudonapi} from Verity’s Plate LIX}

\begin{itemize}
\item 13. MACDUNNOUGHII REMINGTON, 1954
\end{itemize}

The action of Remington (1954) in replacing the North American name \textit{pseudonapi} McDunnough, 1916 was correct, because of homonymy, although he cited the wrong date and location (1911, p. 330) for the original description of \textit{pseudonapi} Verity (1 Jan 1911, Plate LIX Figures 13-17; see above). Regardless of the publication date, \textit{pseudonapi} McDunnough, 1916 is a primary homonym of the available name \textit{pseudonapi} Verity, 1911, and hence a new name such as \textit{macdunnoughii} was required to replace it.

The spelling \textit{macdunnoughii}, used by dos Passos (1964), was exactly the same as appeared in the original description by Remington (1954). It therefore could not be an “unjustified emendation” (Code Article 33.2.3) as stated by Miller and Brown (1981) in the note for their checklist entry “\textit{mcdunnoughi}”: “\textbf{[Note] 259. Unjustifiably emended to “macdunnoughi” [sic] by dos Passos, Mem. Lepid. Soc. (1): 40 (1964).}”

Miller and Brown (1981) use the spelling \textit{mcdunnoughi}, which has two spelling changes from the original description – a change of “\textit{mac}” to “\textit{mc}” and a change of double “\textit{ff}” to single “\textit{f}”. The relevant sections of the Code are:

\begin{itemize}
\item \textbf{33.2. Emendations.} Any demonstrably intentional change in the original spelling of a name other than a mandatory change is an "emendation", except as provided in Article 33.4.33.2.1. A change in the original spelling of a name is only to be interpreted as "demonstrably intentional" when in the work itself, or in an author's (or publisher's) corrigenda, there is an explicit statement of intention, or when both the original and the changed spelling are cited and the latter is adopted in place of the former, or when two or more names in the same work are treated in a similar way.
\end{itemize}
33.2.2. The correction of an incorrect original spelling in accordance with Article 32.5 is a "justified emendation", and the name thus corrected retains the authorship and date of the original spelling [Art. 19.2].

33.2.3. Any other emendation is an "unjustified emendation"; the name thus emended is available and it has its own author and date and is a junior objective synonym of the name in its original spelling; it enters into homonymy and can be used as a substitute name, but

33.2.3.1. when an unjustified emendation is in prevailing usage and is attributed to the original author and date it is deemed to be a justified emendation.

33.3. Incorrect subsequent spellings. Any subsequent spelling of a name different from the correct original spelling, other than a mandatory change or an emendation, is an "incorrect subsequent spelling"; it is not an available name and, like an incorrect spelling [Art. 32.4], it does not enter into homonymy and cannot be used as a substitute name, but

33.3.1. when an incorrect subsequent spelling is in prevailing usage and is attributed to the publication of the original spelling the subsequent spelling is deemed to be a correct original spelling.

33.4. Use of -i for -ii and vice versa, and other alternative spellings, in subsequent spellings of species-group names. The use of the genitive ending -i in a subsequent spelling of a species-group name that is a genitive based upon a personal name in which the correct original spelling ends with -ii, or vice versa, is deemed to be an incorrect subsequent spelling, even if the change in spelling is deliberate; the same rule applies to the endings -ae and -aæ, -orum and -iorum, and -arum and -iarum.

The change to a single "i" was clearly an incorrect subsequent spelling under Article 33.4. The change from "mc" to "mac" in "macdunnoughii" was not a "demonstrably intentional change" (Article 33.2), because there was not an explicit statement of intention" through the (incorrect) statement by Miller and Brown that the original spelling using "mac" in macdunnoughii was incorrect. That could be a reasonable assumption to be deduced from the (incorrect) reference to an unjustified emendation, but they do not explicitly make that statement – they could have been referring only to the use of the double "ii". The spelling changes by Miller and Brown were therefore an "incorrect subsequent spelling" under Code Article 33.3, unless the "incorrect subsequent spelling is in prevailing usage". We have seen no evidence in the literature that the incorrect spelling is in prevailing usage.

However, there is the question of whether the spelling macdunnoughii was a deliberate change of "Mc" to "mac" when Remington based the spelling on the surname McDunnough. The relevant part of the Code reads:

32.5. Spellings that must be corrected (incorrect original spellings).

32.5.1. If there is in the original publication itself, without recourse to any external source of information, clear evidence of an inadvertent error, such as a lapsus calami or a copyist's or printer's error, it must be corrected. Incorrect transliteration or latinization, or use of an inappropriate connecting vowel, are not to be considered inadvertent errors.

32.5.1.1. The correction of a spelling of a name in a publisher's or author's corrigendum issued simultaneously with the original work or as a circulated slip to be inserted in the work (or if in a journal, or work issued in parts, in one of the parts of the same volume) is to be accepted as clear evidence.

Examples. If an author in proposing a new species-group name were to state that he or she was naming the species after Linnaeus, yet the name was published as nimnaei, it would be an incorrect original spelling to be corrected to linnaei. Enigmophyllum is not an incorrect original spelling (for example of Enigmatophyllum) solely on the grounds that it was incorrectly transliterated or latinized.

It can be argued that, in parallel to the Code example, the statement in the paper that the taxon was named after McDunnough is sufficient evidence of a lapsus calami when the name was spelled “macdunnoughii”. However, in the example given by the Code for a species named after Linnaeus, the incorrect spelling was not part of latinization but was a simple typographical error. In contrast, the changing of “Mc” to “mac” is part of the latinization of the word “McDunnough” to “macdunnoughii”, as indicated by a 1964 Code (International Commission on Zoological Nomenclature 1964) recommendation (Appendix D, p. 109):
Personal names bearing prefixes should be treated as follows in forming zoological names:

(a) The prefixes "Mac", "Mc", or "M" should be spelled "mac" and united, as in maccooki (McCook), macoysi (M'Coy)."

Incorrect latinization is specifically stated to be not considered an inadvertent error, and in this case the spelling change was clearly correct latinization by the standard of the day. As a point of interest, the same requirement is still present in the modern International Code of Botanical Nomenclature. The requirement has been dropped from the modern Zoological Code, but there is also nothing forbidding such latinization. In any case, the original spelling was clearly the result of latinization at the time; hence Article 32.5.1 forbids considering it to be an inadvertent error.

The spelling macdunnoughii Remington, 1954 is therefore the correct, as well as the original, spelling.


Warren (1968) described *Pieris passosi* as a “hybrid species”, consisting of a population that he considered to be the result of hybridization between *P. oleracea* Harris, 1829 and *P. hulda* W.H. Edwards, 1869. A small number of “pure” examples of the parental species were also present at the type locality, with one specimen of each being identified as such by Warren. The type specimens are syntypes, because Warren explicitly chose not select a holotype, and comprise 17 males and 9 females in the collection of C.F. dos Passos (now part of the American Museum of Natural History collection). Warren incorrectly called the type specimens “paratypes”, rather than “syntypes”. The type locality is Palmer, Alaska, which is south of Anchorage. The relevant Code articles to determine the availability of the name *passosi* are:

**Article 1. Definition and Scope**

1.3 Exclusions. Excluded from the provisions of the Code are names proposed
1.3.3 for hybrid specimens as such (for taxa which are of hybrid origin see Article 17.2).

**Article 17. Names found to denote more than one taxon, or taxa of hybrid origin, or based on parts or stages of animals or on unusual specimens.** The availability of a name is not affected even if
17.1. it is found that the original description or name-bearing type specimens(s) relates to more than one taxon, or to parts of animals belonging to more than one taxon; or
17.2. it is applied to a taxon known, or later found, to be of hybrid origin (see also Article 23.8);

**Article 23.8. Application to species-group names established on hybrids.** A species-group name established for an animal later found to be a hybrid [Art. 17] must not be used as the valid name for either of the parental species, even if it is older than all other available names for them. Such a name may enter into homonymy. For names based on taxa which are of hybrid origin see Article 17.2.

Definitions:

as such. Being strictly what has been cited (e.g. "a photograph as such" is an illustration on light-sensitive paper, not one printed in a work).

hybrid, n. The progeny of two individuals belonging to different taxa. For the treatment of names given to hybrids and to taxa of hybrid origin see Articles 1.3.3, 17, 23.8.

The interpretation of Code Article 1.3 is critical, because it determines whether the name *passosi* is covered by the provisions of the Code, or excluded. Excluded from the provisions of the Code are “names proposed for hybrid specimens as such”; included in the Code are all names proposed for specimens that the author did not consider to be hybrids as defined by the Code. Warren did not apply the name *passosi* to specimens that he considered were “the progeny of two individuals belonging to different taxa”, in this case *Pieris oleracea* and *P. hulda*. He considered that hybridization between *oleracea* and *hulda* had resulted in the new species *passosi*, and that the parents of the specimens he examined were other individuals of the species *passosi*. In modern genetic language, the specimens were not F1 hybrids (progeny of different taxa), but were F2, F3, etc. hybrids (progeny of other hybrids, not different taxa). A name falls under the Code, and is not excluded through the provisions of Code Article 1.3, if the author explicitly treats the population as a taxon, not just as a number of individual specimens (Philip Tubbs, International Commission on Zoological Nomenclature, pers. comm.).
Note that Articles 17.2 and 23.8 provide for the case where the author treated the specimen(s) as representing a taxon, not being hybrid(s), but it is later determined that the types are hybrid specimens. In that case the name is an available name under the Code, but, quite logically, cannot be used as a valid name for either parent taxon because it equally represents both.

Extracts from the original description of *P. passosi* demonstrate that Warren treated the specimens as representing a taxon:

“We know several long established hybrid species of *Pieris*, but *P. passosi* gives us one such hybrid in what must be a relatively early stage of development, for the parental species are still present.”

And further on he describes his interpretation of reproduction in the population:

“... hybrids mating with other hybrids of differing characters, on occasions back-crossing to one or other parent race only to be back-crossed again to some hybrid form.”

Code Article 1.3 excludes individual hybrid *specimens* (= individuals) from the mandate of the Code; they are the equivalent of aberrations or other abnormal individuals that do not form a self-perpetuating population. A *taxon* of hybrid origin is specifically not excluded, because it forms a self-perpetuating population. Article 17 explicitly states that names applied to a *taxon* “known to be of hybrid origin” can be available, which reinforces this interpretation.

The name *passosi* B. Warren, 1968 therefore falls under the provisions of the Code, and, because the provisions of Articles 11 and 12 are met, is an available name.

The type series of *Pieris passosi* includes at least three taxa – *P. oleracea* (T. Harris, 1829), *P. marginalis meckyae* Eitschberger, 1983, and *P. angelika* Eitschberger, 1983 (Eitschberger 1983). We borrowed the syntypes of *Pieris passosi* from the American Museum of Natural History (AMNH), together with a nearly equal number of nonsyntypes from near Palmer, Alaska that were also from the dos Passos collection, for a total of 41 specimens. One of the syntypes of *Pieris passosi*, illustrated in Figure 6 of Warren (1968), is missing. There are 15 males and 12 females either labeled as paratypes (= syntypes) of *passosi* or identifiable as having been figured by Warren, rather than the 17 males and 9 females stated by Warren. There is strong sexual dimorphism in the wing pattern of *Pieris* from the Palmer area; hence it is unlikely that Warren incorrectly determined the sex of any specimens. However, about half of the paratype labels were apparently attached at a later date, and two of Warren’s figured “paratypes” lack paratype labels; hence some of the specimens labeled as paratypes may not have been actually part of Warren’s type series. In addition to the three species already mentioned, one specimen appears to be *P. marginalis hulda*, although this may be an extreme variant of *P. meckyae*. The type series of *P. passosi* consists of specimens of at least three taxa; hence Warren’s confusion is hardly surprising.

We designate as lectotype of *Pieris passosi* B. Warren, 1968 the specimen shown with its labels in Figure 9, to promote nomenclatural stability and taxonomic clarity. This is the “paratype” (= syntype) specimen represented by Plate IV Figure 5 of Warren (1968); the lectotype is demonstrably from the syntype series because it was illustrated by Warren. This specimen is the same taxon as *Pieris marginalis meckyae*, and hence *meckyae* Eitschberger, 1983 is a subjective synonym of *passosi*. The remaining syntypes are now paralectotypes that no longer have any nomenclatural significance; paralectotype labels have not been attached due to the uncertainty of the original syntype status of some of the specimens. The lectotype has been returned to the AMNH.

![Figure 9. Syntype of Pieris passosi](image)

Scale larger than life-size; forewing span = 42 mm.
9. **PALLIDISSIMA** Barnes & McDunnough, 1916

Barnes and McDunnough (1916) described the taxon *pallidissima* with the words:

“In Utah we meet with a second generation (July, August) which is extremely pale, being practically immaculate in both sexes on both sides; the underside is tinged with pale yellow on secondaries and apex of primaries and the ♀ on the upperside of primaries shows faint traces of upper black spot; it is a further development of *casteria* apparently differing from both this form and *pallida* in the reduction of the black spots in the ♀; we propose the name PALLIDISSIMA for the race and figure the type ♀ and ♦ from Provo, Utah (Figs. 4, 5, 10).”

The captions for the figures of *pallidissima* are clearly in trinomial form, with *pallidissima* treated as a subspecies (Plate VI, Figs. 4, 5, 10), as shown in Figure 10.

Figure 10. captions for the figures of *Pieris napi pallidissima*

The phrasing of the text could be misinterpreted, without close examination, to indicate that *pallidissima* was named as the summer form of the Utah populations; this was the interpretation of Remington (1954). However, the use of the word “race”, combined with the format of the captions of the figures, makes it clear that *pallidissima* was named as a geographically defined group of populations — a subspecies — that is characterized by the appearance of the summer generation.

Miller and Brown (1981) and Pelham (2008) asserted that there is a holotype of *pallidissima*. However this is impossible because (1) no holotype was explicitly designated in the original description, and (2) the name is based on more than one specimen and hence holotype designation by monotypy does not apply (Article 73.1). The specimens in the type series therefore are all syntypes.

If the taxonomic decision is made that the Utah and Colorado populations of *Pieris marginalis* are the same taxon, as suggested by authors such as Remington (1954) and Warren (1968), then *macdunnoughii* Remington, 1954 may become a subjective synonym of *pallidissima* Bames & McDunnough, 1916. Given the taxonomic uncertainty related to *Pieris* populations in western North America; we consider it essential to have a clear and objective standard of reference for the name *pallidissima*. We therefore designate the specimen illustrated in Plate VI Figure 4 of Barnes and McDunnough (1916) to be the lectotype of the name *pallidissima*.

10. **ANGELIKA** Eitschberger, 1983

The name *Pieris angelika* was first proposed by Eitschberger (1981). When translated to English, the original text reads:

“4) *Pieris angelika angelika* n. spec.

This species so far has gone under the unjustified name of *Pieris napi pseudobryoniae* auct. (not VERITY, 1908) …. The populations from Alaska and Northwestern Canada I hereby call *Pieris angelika angelika* n. spec. after the name of my wife, who not only suffers Entomology, but is rather actively involved in promoting and supporting my work. Of this species, there are so far, from diverse localities in the above named regions nearly 200 males and females in the coll. EITSCHBERGER-STEINIGER. Further material is at hand from various private and museum collections. Exact analysis and description of this species follows in the earlier mentioned revision. But in order to already determine the species at this time, a few black and white photographs shall be shown here. These specimens, as well as all other taxa of the *napi-
bryoniae group, will later be shown on a number of color plates in the revision. Even though there is no recognizable subspecific tendency in the available material, and therefore all examples at hand are treated as species material (exact listing follows later), I would like to designate the vicinity of Elsa and Keno (Canada, Yukon) as the type locality. With this, possible arguments and uncertainties should be avoided in case angelika n. spec. is not monotypic.”

The Code requires that, for a name proposed after 1930, a description, definition or bibliographic reference must be provided (Article 13). Eitschberger (1981) provided illustrations, but did not provide even a single phrase of description, definition, or bibliographic reference. The name angelika Eitschberger, 1981 is therefore a nomen nudum, as stated by Kudrna & Geiger (1985), Pelham (2008) and others. The relevant Code Articles are:

<table>
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<tr>
<td>13.1. Requirements. To be available, every new name published after 1930 must satisfy the provisions of Article 11 and must</td>
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<tr>
<td>13.1.1. be accompanied by a description or definition that states in words characters that are purported to differentiate the taxon, or</td>
</tr>
<tr>
<td>13.1.2. be accompanied by a bibliographic reference to such a published statement, even if the statement is contained in a work published before 1758, or in one that is not consistently binominal, or in one that has been suppressed by the Commission (unless the Commission has ruled that the work is to be treated as not having been published [Art. 8.7]), or</td>
</tr>
<tr>
<td>13.1.3. be proposed expressly as a new replacement name (nomen novum) for an available name, whether required by any provision of the Code or not.</td>
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However, two years later Eitschberger (1983) did provide a description of Pieris angelika, and met all the Code requirements to make the name available. The name angelika Eitschberger, 1983 is therefore an available name, as stated by Kudrna & Geiger (1985), Pelham (2008) and others. The original description is too long to repeat here.

It is worth noting that two Siberian subspecies attributed to Pieris bryoniae by Eitschberger (1983) are, in our opinion, actually subspecies of Pieris angelika. The species is therefore a “Beringian” species that during the last glacial period was likely spread through much of the ice-free area that extended from the Yukon and Alaska into eastern Siberia (= Beringia), with a land connection where the Bering Strait is now present. The synonymy is:

**Pieris angelika** Eitschberger, 1983

a. ssp. *angelika* Eitschberger, 1983
   TL: Keno (el. 4600 feet), Yukon, Canada
b. ssp. *schinlmeisteri* Eitschberger, 1983
   TL: Jakutia, Tommot, Russia
c. ssp. *sheljuchkoi* Eitschberger, 1983
   TL: Omsukchan, Magadan Gebeit, Russia

Code Article 24.1 requires that the name *angelika* Eitschberger, 1983, proposed as a species-level name, take precedence over *sheljuchkoi* Eitschberger, 1983 and *schinlmeisteri* Eitschberger, 1983 which were both proposed as subspecies.

### 11. PUBLICATION DATE OF EITSCHBERGER’S BOOK SYSTEMATISCHE UNTERSUCHUNGEN AM PIERIS NAPI-BRYONIAE-KOMPLEX (S. L.) (LEPIDOPTERA, PIERIDAE)

Ulf Eitschberger described many new taxa in his monumental two volume work *Systematische untersuchungen am Pieris napi-bryoniae-komplex (s. l.) (Lepidoptera, Pieridae)*. The book is dated “1983”, and, according to the author (U. Eitschberger, pers. comm.), all copies were printed, bound and ready for distribution by mid-December, 1983. In mid-December, 1983 Eitschberger placed one copy on display in a public university library, and distributed additional copies to his brother and to E. J. Reissinger (U. Eitschberger, pers. comm.). Further distribution of the book started in February 1984. The issue is whether distributing three copies constitutes “publication”, or whether publication occurred upon recommencing further distribution of the book in February 1984.
Reissinger (1986) summarized this as:

1. Publication datum (pp. 47-48). Eitschberger's Publikation is zwar für 1983 datiert, ist Ende dieses Jahres auch schon in der Zoologischen Universität Bonn zur Einsicht öffentlich ausgelegt worden, die Abgabe und der Versand aller beiden Bände und damit einer allgemeinen Öffentlichkeit zugänglich gemacht, erfolgte ab 4.II.1984. Dies ist somit das gültige Datum – the true date of publication according to the International Code of Zoological Nomenclature (ICZN), Articles 21 and 22.

This can be translated to English as:

1. Publication date (pp. 47-48). Eitschberger's publication is dated 1983; it was already displayed for public viewing at the end of the year in the Zoological University of Bonn. The delivery and shipping of both volumes, and thus general public accessibility, took place starting 4.II.1984. This is therefore the valid Datum – the true date of publication according to the International Code of Zoological Nomenclature (ICZN), Articles 21 and 22.

The 1964 edition of the Code, which was in force when Reissenger published his opinion, states:

**Article 9. What does not constitute publication.** – None of the following acts constitutes publication within the meaning of the Code:

(6) mere deposit of a document in a library.

Therefore, under the 1964 Code, if only the one copy had been deposited in the library, it would not have constituted publication. That restriction no longer exists in the Code, although it suggests how to interpret parts of the current Code. In any case we now know that there were additional copies distributed to the public and hence the restriction actually never applied. However the issue is compliance with the present Code, and it is the ‘date of publication’ that is relevant; not the act of ‘publication’. The present Code glossary states:

**Definition:**

**Date of publication, n.** Of a work (and of a contained name and nomenclatural act): the date on which copies of the work become available by purchase or free distribution. If the actual date is not known, the date to be adopted is regulated by the provisions of Article 21.2-7.

The criteria of ‘delivery and shipping’ and ‘general public accessibility’ used by Reissinger (1986) are not Code criteria for establishing date of publication. The Code only requires that copies ‘become available’, and it has been established that copies were in fact available in December 1983. Distribution commenced in December 1983, with three copies actually distributed, and the commencement of distribution is the normal ‘proof’ that a book is available for distribution. However, it is worth noting that the Code does not require actual distribution for date of publication; in theory at least, a publication could be available for distribution but no-one chooses to obtain a copy.

Ulf Eitschberger (pers. comm. Jan. 2009) does not remember the exact date, after 25 years, on which he commenced distribution of the book with the first three copies, other than it was well before the end of December. Therefore the date of availability is here set as 31 December 1983, consistent with Article 21.3.1.

**21.2. Date specified.** The date of publication specified in a work is to be adopted as correct in the absence of evidence to the contrary.

**21.3. Date incompletely specified.** If the day of publication is not specified in a work, the earliest day on which the work is demonstrated to be in existence as a published work is to be adopted as the date of publication, but in the absence of such evidence the date to be adopted is

21.3.1. the last day of the month, when month and year, but not day, are specified or demonstrated, or

21.3.2. the last day of the year when only the year is specified or demonstrated.

We are not aware of any other taxonomic publication regarding the genus *Pieris* in December 1983; therefore further refinement of the date is unnecessary, even if possible.
Notes regarding Eitschberger (1983):

1) Kudrna and Geiger (1985), Shapiro (1985), and Ferris (1989) claimed that some of the many names proposed by Eitschberger (1983) were *nomen nuda*, on the grounds that they failed to meet the Code requirement for a “definition or description”. We have examined the text for all of the new taxa in Eitschberger (1983), and there is clearly a description accompanying every name. The names all meet the remaining requirements of the Code, and hence are all available names. This has generally been accepted by this date, and hence we do not further discuss the issue.

2) The taxon *tremblayi* Eitschberger was spelled incorrectly as “tremblay” by Kudrna and Geiger (1985), which is an “incorrect subsequent spelling” and is not an available name under Code Article 33.3.

3) Eitschberger stated in a footnote that his taxon *guppyi* was a patronym for “Cyril S. Guppy”, this was an error for “Crispin S. Guppy”.

4) Eitschberger did not include the collector’s name for many of the specimens he listed as having examined. Most of the British Columbia, Yukon, and Alaska specimens for 1976 and 1977 listed as being in the Eitschberger-Steiniger collection were collected by Crispin S. Guppy. In many cases Guppy retained part of the series collected at each site; for new taxa these retained specimens are not paratypes because Eitschberger did not examine them.

12. SUMMARY OF CONCLUSIONS

1. The name *pseudobryoniae* Verity, 1908 is an unavailable infrasubspecific name.

2. The name *adalwinda* Fruhstorfer, 1909 is an unavailable infrasubspecific name, with the publication date corrected from 1911 to 1909.

3. The name *pseudobryoniae* Barnes and McDunnough, 1916 is newly identified as an available species-group name, with at least four syntypes. A lectotype is designated, with the type locality being Nulato, Alaska. The name *browni* Eitschberger, 1983 is a subjective synonym of *pseudobryoniae* Barnes and McDunnough, 1916.

4. The name *arctica* Verity, 1911 is an unavailable infrasubspecific name.

5. The name *arctica* Barnes and McDunnough, 1916 is newly identified as an available species-group name, with at least six syntypes. A lectotype is designated, with the type locality being northern Norway. It is the available name for the populations to which the unavailable name *adalwinda* Fruhstorfer, 1909 is presently applied.

6. The name *pseudonapi* Verity, 1909 is determined to be an available species-group name. The correct original description is identified and the date of publication corrected to 1909, from 1911. There are five syntypes from the type locality of Jesso, Japan (= Hokkaido). The name *pseudonapi* Barnes & McDunnough, 1916 is therefore a primary homonym of *pseudonapi* Verity, 1909, validly replaced by *macdunnoughii* Remington, 1954.

7. The name *macdunnoughii* Remington, 1954 is the correct and original spelling as it appears in Remington (1954); the spelling *macdunnoughii* by dos Passos (1964) is correct and is not an “unjustified emendation” as asserted by Miller and Brown (1981). Miller and Brown (1981) provide the incorrect subsequent spelling *mcduhnoughii*.

8. The name *passosi* Warren, 1968 is an available species-group name, with 26 syntypes from the type locality of Palmer, Alaska. The syntypes consist of specimens from at least three species. A lectotype is designated to fix the name to a specific taxon, with *meckyae* Eitschberger, 1983 a subjective synonym of *passosi*.

9. The name *pallidissima* Barnes & McDunnough, 1916 is an available species-group name. A lectotype is designated – the specimen illustrated in Plate VI Figure 4 of Barnes and McDunnough (1916) – to fix the name to a specific specimen as the basis for a later taxonomic decision regarding possible synonymy of *pallidissima* and *macdunnoughii* Remington, 1954.

10. The name *angelika* Eitschberger, 1981 is a *nomen nudum*, but *angelika* Eitschberger, 1983 is an available name. Two Siberian taxa are suggested to be subspecies of *P. angelika*, rather than being subspecies of *P. bryoniae*, resulting in the combinations *P. angelika sheljuzhkoii* Eitschberger, 1983 and *P. angelika schintmeisteri* Eitschberger, 1983.
11. The publication date of Eitschberger’s *Systematische untersuchungen am Pieris napi-bryoniae-komplex (s. l.) (Lepidoptera, Pieridae)*, and hence the date of publication of the new taxa described within, is 31 December 1983. Three copies of the book were distributed during December 1983, demonstrating the Code requirement of availability for distribution on or before that date. All the new names proposed in Eitschberger (1983) have descriptions sufficient to meet the requirements of the Code for available names, contrary to Kudrna and Geiger (1985), Shapiro (1985), and Ferris (1989).

ACKNOWLEDGMENTS

We thank Jonathan Pelham for reviewing drafts of the paper and providing stimulating discussions regarding interpretations of the Code (although he may disagree with some of the conclusions), Philip Tubbs of the International Commission on Zoological Nomenclature for being helpful with respect to interpreting the Code Articles regarding hybrids, Kenelm Philip for providing color illustrations from old literature, Joe Beliceck for excellent review comments, and Ulf Eitschberger for clarifying the dates of publication and distribution of his book.

REFERENCES


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Larval host plants of *Enodia anthedon*, *Satyrodes appalachia* and *S. eurydice* in Vermont, USA

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Abstract: Field observation and captive rearing was used to clarify larval host plant use in *Enodia anthedon*, *Satyrodes appalachia* and *S. eurydice* in Vermont, USA. In nature *S. appalachia* larvae were found on *Carex lacustris*, *C. lupulina*, *C. gracillima* and *C. tuckermanii*. *E. anthedon* in nature was found to oviposit on grass and on *Carex lacustris*. Larvae of *E. anthedon* were found on *C. lacustris* and *C. lupulina*. In captivity, all but two *E. anthedon* larvae chose *Carex* over grass. Both *E. appalachia* and *E. anthedon* larvae thrived on a diet of *C. lacustris* and *C. lupulina* in captivity. Early instar captive larvae refused *C. sparganioides* but late instar captive larvae accepted said plant. Grass may be an acceptable alternate host for Vermont *S. eurydice*. Miscellaneous observations on early instars are reported.

Additional key words: Lepidoptera, Nymphalidae, Satyridae, life cycle, habitat

INTRODUCTION

Various grass species and *Carex* sedges are listed respectively as host plants for *Enodia anthedon* and *Satyrodes appalachia* by many authors, including Howe (1975). Scott (1986) wrote: [anthedon] “Hostplants grasses” and [appalachia] “Larvae refused grasses in the lab.” Handfield (1999) lists eight grasses for *E. anthedon*, and *Carex* for *S. appalachia leeuwi*. Nielsen (1999) and Douglas & Douglas (2005) similarly list various species of grasses for *anthedon* vs. sedges for *appalachia*. O’Donnell, et al. (2007) list grass (*Dactylis glomerata*) for *anthedon*, and sedges (incl. *Carex stricta*) for *appalachia*. Allen (1997) wrote: “Grasses are the primary host for [anthedon]” and “Third instar larvae from the last brood overwinter in a rolled grass blade tied together with silk.” Allen describes *appalachia* as overwintering in a similar way, but adds, “In West Virginia the Appalachian Brown also uses grasses as hosts.”

METHODS

I searched for ovipositing females and larvae in natural habitats during 2007, 2008, 2009 and 2010. I searched the range of habitats used by *E. anthedon* and *S. appalachia* in the Grand Isle area. Larvae were raised, in containers, in locations receiving partial sunlight – somewhat imitating wooded habitat. A screened narrow porch on the west side of the house provided partial late afternoon sunlight; the duration of which was limited by trees to the west. Indoor locations were used when high winds were thrashing the sedges and when close monitoring of larvae was desired (also in cold months!): windows provided partial direct morning and afternoon light, with only a distant 60w bulb extending light hours throughout each evening.
Figures 1-3. Host – *Carex* sedges. Fig. 1. 31-May-2009, small sixth-instar *E. anthedon* larva eating container-grown sedge. Fig. 2. 30-Jun-2009: Typical habitat at the swamp, a 1.75 acre (0.71 hectare) vernal pool. Fig. 3. *E. anthedon* male at swamp.

Figures 4-12. Life cycle of *E. anthedon*. Fig. 4. *E. anthedon* larvae hatching. Fig. 5. Recently hatched *E. anthedon* larvae after first meals of sedge. Fig. 6. First-instar larvae: *E. anthedon* (one) & *S. appalachia* (two), 19-Jul-2009. Fig. 7. Second-instar *E. anthedon*. Fig. 8. Third-instar *E. anthedon*. Fig. 9. Fourth-instars: *E. anthedon* (left) & *S. appalachia* (right), on grass, 26-Aug-2009. Fig. 10. Sixth-instar *E. anthedon*, after molt from fifth-instar as collected on *Carex* sedge, 4-Jun-2009. Fig. 11. Late sixth-instar *E. anthedon*. Fig. 12. *E. anthedon* female, ex-pupa 30-Jun-2009.
Figures 13-15. *S. appalachia*. Fig. 13. Female (54.5mm), ex-ovum Jul-2008; ex-pupa 12-Jun-2009. Fig. 14. Various-aged *S. appalachia* larvae and molted head capsules illustrating the often prominent dark line extending from the base of the horns to the eyes. Fig. 15. Male (D/V), ex-larva collected at swamp, ex-pupa 24-Jun-09.

Figures 16-19. *S. appalachia*. Fig. 16a. Male, ex-pupa 26-Aug-2008; from ovum laid 30-Jun or 01-Jul-2008. Fig. 16b. Female, netted at swamp 02-Sep-08. Fig. 16c. Male, netted at lake site 03-Sep-08. Fig. 16d. Male, from swamp 05-Sep-08. Fig. 17. Newly hatched larva 01-Sep-10, from unassisted pairing of second-flight *appalachia*. Fig. 18. A third-instar *appalachia* larva (upper right), from the second flight, in diapause mid-Oct-2010 to Apr-2011; and two first-flight diapausing *E. anhedon* larvae (lower left). Fig. 19. Phenograms illustrating partial second flight of *appalachia*.

**RESULTS AND DISCUSSION**

**Field Observations**

At a woodland swamp, I located ca. 20 post-diapause *S. appalachia* larva during May-June 2007, 18 post-diapause larvae in 2008, and 38 in 2009. While most larva were feeding on *C. lacustris* (Willd.) and C. lupulina (Muhl. ex Willd), a few were found on *C. gracillima* (Schwein), and one fed successfully on a fourth sedge species, *C. tuckermanii* (Dewey), an observation repeated in 2010. One of three first-instar *S. appalachia* larva found July 2007 was feeding on grass, an unexpected host, as were two ex-ova larvae raised at home and two of >20 larva observed in the woodland swamp Aug.-Sept. 2009.

Grass might also be an acceptable alternate host for Vermont *S. eurydice* – one of which laid an egg on grass in an all-grass habitat on July 5, 2007.
An *E. anthedon* was observed ovipositing on grass, the expected host, in the woodland near the swamp on July 27, 2008. However, in the same swamp, on Sept. 25, I collected three small *anthedon* larvae from *Carex* sedges in two locations. These three larvae were stored over winter in my refrigerator, along with two dozen *appalachia* larvae. Due to mold encroachment, some larvae were prematurely transferred to container sedge in late January, too early for extended larval survival – and in late-February, when newly released *appalachia* larvae shortly resumed robust growth. However, the two surviving *anthedon* larvae soon died – only one eating but one meal on Mar. 4, 2009. Room temperature was 14°C (57°F).

Subsequent searches of the woodland swamp, May 30 to June 14, 2009, revealed ten post-diapause sedge-eating *anthedon* larvae. Another was found on June 4, 2009, in a lakeside wet woodland 10k (5.4 miles) away. Two larvae were monitored at the swamp; the other nine were collected, the smallest of which died; it appeared to be deformed, or possibly was injured when collected. Another larva escaped from its container. Although two of the larvae had been feeding on a fine-leaf grass, they were situated in sparse vegetation where they had initially eaten sedge. The two undoubtedly would have returned to sedge – the available grass was insufficient to provide many meals. When collected and offered a choice of sedge or the same grass species, the two larvae chose sedge. These seven surviving container-bound larvae successfully matured on the sedge diet; the first two emerged from their pupae on June 25, the same day that the first flight of *anthedon* (*n* = 4) was observed. The two uncollected larvae also pupated successfully on their host sedges.

From early Sept. through Oct. 5, 2009, twelve pre-diapause *anthedon* larvae were found eating sedge at the swamp - some of these larvae were found again in May of 2010.

The two woodland study sites (44.75000N, -073.30556W and 44.69391N, -073.34210W) for *E. anthedon* were chosen for the presence of colonies of *S. appalachia* with which the *anthedon* associated. The nearby drier site, a former pasture reverting to shrubs, was at 44.69548N, -073.34010W. Observed flight numbers in 2009 for *anthedon* were low, perhaps related to frequent rainy weather; the flight seemed to be delayed a week or two. However, the one-day maximum of 120-140 *appalachia* was higher at the lake site than numbers observed in previous years - in 2010, an early first flight peaked at 250. The lake site is dominated by young ash (*Fraxinus pennsylvanica*) and elm (*Ulmus americana*) with a dense carpet of sedge. The woodland swamp is a 1.75 acre (0.71 hectare) vernal pool shaded by red maples (*Acer rubrum*), and surrounded by other mature trees and patchy forest-floor vegetation.

An interesting observation was made: At the dry upland site, up to 15 male *anthedon* clustered together on shrubbery, instead of engaging in territorial aerial pursuit as observed in the two wet habitats. A few years previously, I had observed similar communal behavior at the lake site. These Vermont sites fall within the “contact zone” of *E. a. anthedon* and *E. a. borealis* – taxa showing behavioral differences (Grkovich & Pavulaan, 2003).

**Captive Rearing**

In 2009, I collected five female *E. anthedon*, and a few female *S. appalachia* to obtain ova. Two of the *anthedon* were collected from the woodland swamp, two from the lakeside woodland (one was ovipositing on sedge), and the fifth from a nearby dry upland habitat. I later noted no discernible differences among the *anthedon* larvae.

Of >65 *E. anthedon* and *S. appalachia* eggs, most were deposited on sedge (it being the dominant plant in each container), one egg on grass, one on twine, and several inside the plastic cap of a support stake and on the sides of the Rubbermaid® containers.

Larvae were free to chose between sedge and grass – all but two of the *anthedon* larvae opting for sedge. By late-Aug., three *anthedon* larvae were feeding on grass – two having moved from sedge onto grass, while one of the previous two had abandoned the grass. On Sept. 8, four were on grass, but some of the *anthedon* were already idle, in diapause, as were most of the *appalachia* larvae sharing the containers. Two of the *S. appalachia* larvae were also feeding on grass. One quit its grass diet for sedge in mid-Aug; the second *appalachia* remained on grass, molting to a
normal straw-colored 4th instar in late-Aug. All appalachia (n = 45-50) entered diapause as 4th instar larvae by Sept. 14, while six anthedon were still feeding. Larvae of both species ate at any hour of the day, the daytime feeding being especially noticeable with the appalachia. Conversely, Cech and Tudor (2005) wrote, “[appalachia] Caterpillars feed at night, hiding near hostplant base by day.”

Whereas most authors state that larval diapause occurs with the third or fourth instar, Layberry, et al. (1998) wrote, “[anthedon] larvae overwinter in the first instar.” Watching and measuring each anthedon larva, I attempted to confirm that every larva molted from third to fourth instar prior to diapause, and am confident that all larvae did. By Sept. 14, the six ex-ova anthedon larvae still eating sedge (five) and grass (one) had reached their maximum instar length: 18.5-19mm. The two smallest of the five larvae collected from the swamp molted to fourth instar on Sept. 13 and 17. The lack of frost in September ensured that all larvae had time to enter the fourth instar. In most years, the surrounding Lake Champlain buffers Grand Isle from early frost. The average first frosts normally arrive in mid-Oct.

In all years, none of the diapausing larvae, anthedon or appalachia, made any attempt to create a shelter of rolled leaves as reported by Allen (1997), although in 2008 some appalachia did hide under dried deciduous leaves, rather than stay on sedge stems and leaves. I did notice that occasionally a cut section of refrigerated sedge leaf, while drying, by chance would partially curl around the attached larva – the larva’s silk track having no influence on the direction of the curl. It was also noted that anthedon (and appalachia) larval length shrinks during diapause.

When appalachia larvae resumed growth in springtime, they required two more molts prior to pupation. In 2008, the one container-raised appalachia larva that skipped diapause, apparently did skip one late larval instar; I saw no evidence of the missing (and necessarily short-timed) molt. In 2009, one exceptional appalachia larva unexpectedly molted to a large, boldly-striped SEVENTH-instar, but the ensuing pupa was slightly deformed and eclosion failed.

From my limited observations of May-June anthedon larvae, I noted only two molts occurring post-diapause – a total of six larval instars (final max. length 41-43.5mm) – the same sequence as the local appalachia. Again in early 2010, the same two post-diapause molts were observed for anthedon larvae which had survived fourth-instar diapause in my refrigerator, and which successfully eclosed as adults, after feeding on sedge. Additionally, in 2010, a warm year in which appalachia and anthedon had early (mid-August) second flights, I obtained some ova from a first flight anthedon female, as well as from some appalachia. Three of nine surviving anthedon larvae skipped diapause, eclosing in late August and early September.

Although the majority (50) of my first-brood appalachia larvae also skipped diapause in 2010, twenty-two entered diapause at fourth-instar as I had anticipated. However, from a mated pair of the second-flight appalachia, six of the seven ex-ova larvae entered diapause earlier, at third-instar; only one underwent one more molt.

Both species were again raised on C. lacustris and/or lupulina, with access to a very limited amount of grass in one of the four containers of sedge. Larvae of both species were observed eating sedge and grass, with no apparent preference. Also present in two of the containers were C. sparganioides (Muhl. ex Willd) and an unidentified common wide-leaf sedge. Both of the latter sedges were refused by early-instar larvae, thus unlikely to be utilized in the wild – although both, especially the sparganioides, were accepted by late-instar larvae.

Phenotypically, my anthedon larvae match published descriptions. However, larvae of this Vermont population of appalachia leeuwi differ slightly from descriptions – the black stripe on the Vermont leeuwi head-capulse horns typically extends beyond the base of the horns to the eyes (the stripe may vary in intensity, but is often very bold). Cardé, et al. (1970) and other authors indicate that for appalachia, unlike S. eurydice, the dark stripes stop at the base of the horns.

CONCLUSION

Although sedges are not currently listed as host plants for E. anthedon, Carex species are accepted by, and may be the preferred host of, this northern New England population of anthedon. Oviposition by anthedon on C. lacustris, and
on a grass species, was observed, and larvae were found feeding on *C. lacustris* and *C. lupulina*. *E. anthedon* larvae raised from ova thrived on a diet of *C. lacustris* and *C. lupulina*. Host sedges for larvae of *S. appalachi* in Grand Isle include *C. gracillima*, *C. lacustris*, *C. lupulina* and *C. tuckermanii*.

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**REFERENCES**


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A case of sympatric *Celastrina ladon* (Cramer), *Celastrina lucia* (W. Kirby) and *Celastrina neglecta* (Edwards) (Lycaenidae: Polyommatinae) in Northern Virginia, with additional records of *C. lucia* in Virginia

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**ABSTRACT.** A case of fully sympatric *Celastrina ladon*, *C. lucia* and early spring brood *C. neglecta* is documented at a site in northern Virginia. Observations indicate that all three species occupy the same habitat, fly during the same flight period and utilize the same host plant with no evidence of hybridization. *C. ladon* and *C. lucia* are obligate univoltines while *C. neglecta* is multivoltine. A later flight (second brood) of *Celastrina neglecta* at the same site utilizes eriophyid mite-induced leaf galls on the very same host tree species. Additional Virginia records of *C. lucia* are documented.

**Additional key words:** sympatry, androconia, *Prunus serotina*, leaf galls, *Cornus florida*

**INTRODUCTION**

*Celastrina* systematics has long remained in flux. Following the separate descriptions of *C. ladon* (Cramer, 1780), *C. lucia* (W. Kirby, 1837) and *C. neglecta* (W. H. Edwards, 1862), the notion of how many species of *Celastrina* inhabit North America has fluctuated from author to author and has been a source of endless debate and confusion. Disagreement among checklist authors continues to the present day. For example, Pelham (2008) lists nine *Celastrina* species north of Mexico, thus adopting species-rank for the three species recorded in this study. On the other hand, the North American Butterfly Association’s most recent checklist as of this writing (NABA, 2001), recognizes only three *Celastrina* species, relegating *neglecta* to subspecies status under *C. ladon*, and does not recognize *lucia* at any rank. Until the 1990’s, most authors traditionally treated *neglecta* as a summer form of *C. ladon* (e.g. Iftner et al., 1992). Since that time, the majority of newly published regional-level guides have treated *C. neglecta* at full-species rank. Wright and Pavulaan (1999) identified a unique primary dorsal wing scale character that differentiates *C. ladon* from *C. neglecta* and all other North American *Celastrina* (except *C. nigra*) as a full-species taxon (Figs. 1 & 2). This character breeds true within *C. ladon* from annual generation to generation without variation. It is also expressed in lab-produced "false summer generation" adults and never appears in lab-reared *C. neglecta*. This unique character is used as a convenient method to differentiate *C. ladon* from both *C. lucia* and *C. neglecta* at a northern Virginia site in the present study.

The name *Celastrina lucia* has been applied to a broad grouping of phenotypically similar, though apparently distinct, *Celastrina* populations spanning the northern portion of the North American continent and extending southward in the Appalachian and Rocky Mountain regions. The taxonomic standing of these various populations is under current review and will likely be revised to include two or more sibling species once it can be determined precisely which population Kirby described as *lucia*. The Appalachian *lucia* population reported here is tentatively retained as a member of the *lucia* species-group until further

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research resolves the type locality issue and clarifies the relationship of continental populations currently treated as lucia. All references given here to the name lucia are tentative and follow the line of reasoning given above. [As an alternative, lucia in Virginia may be referable to as "lucia Auct." until its taxonomic standing can be resolved.]

Fig. 1: Scanning electron micrograph (SEM 640X) of dorsal forewing of C. neglecta showing androconia between blue scales. Specimen taken September 17, 1987, Harleysville, Montgomery Co., PA. Fig. 2: (SEM 640X) of dorsal forewing of C. ladon showing long overlapping scales and lack of androconia. Specimen taken April 23, 1992, Green Ridge State Forest, Allegany Co., MD. Photos by David M. Wright.

DISCUSSION

A distinctive hilltop site known as Old Knob located near Gore, VA, Frederick County, was discovered in June, 2005. This hill (elevation 1,300 ft.) is not impressive by regional standards, but the summit area contains an exceptional array of butterfly species. During a multi-year study of the local colony of Papilio (Heraclides) cresphontes and summit stand of Ptelea trifoliata (Wafer Ash, Hop Tree), an unexpected assembly of sympatric Celastrina species (ladon, lucia, neglecta) was uncovered.

A substantial colony of Celastrina neglecta was first observed on the summit of Old Knob on June 6, 2006. It was noted that Prunus serotina (Black Cherry) comprised a significant portion of the forest understory. Many of these trees were infested with leaf galls formed by eriophyid mites, attributed to Phytophys Cerasicrumena. Several second-brood adult C. neglecta females were observed ovipositing on the leaf galls and many eggs were located, but not collected. Eriophyid mite leaf galls are the primary larval host of C. serotina throughout much of the northeastern United States (Pavulaan & Wright, 2005). Oviposition by C. neglecta on P. serotina leaf galls was previously observed in the area surrounding Big Meadows Recreational Area of Shenandoah National Park in Page County, VA in 1985. C. neglecta has been observed to utilize hosts from a broad range of plant families (Pavulaan and Wright, 2005), thus this behavior at Old Knob was not deemed unusual. At both the Old Knob and Big Meadows sites, mound-building ants were very common (Fig. 27); the ants built huge mounds and defended their turf against intruders rather aggressively. The ants at Big Meadows were of an unidentified type of large stinging ant, while the ants at Gore did not sting, but inflicted painful bites in large numbers as experienced by the author. Throughout the study the ants vigilantly guarded and protected larvae against predators in exchange for larval honeydew secretions. The presence of ant colonies likely contributed to the longevity of the associated Celastrina colonies in this region.
On April 23, 2007 adults of both the *Celastrina lucia* (Figs. 3, 9, 15) and *C. ladon* (Figs. 4, 10, 16) populations were collected on the summit of Old Knob. No early spring brood individuals of *C. neglecta* were recorded on Old Knob in 2007. The *C. lucia* males were easily distinguished from *C. ladon* males by dissimilarity of their forewing scale characters (Figs. 1 and 2). Furthermore, it was immediately surmised that *C. lucia* and *C. ladon* resided in sympatry at this northern Virginia site without evidence of interbreeding and were capable of retaining their separate species-level identities. It was further observed that both species were found in close proximity to a potential host tree *Prunus serotina* and associated ant mounds. No attempt was made to obtain ova on this date. Further exploration demonstrated *C. ladon* was also found in considerable numbers on the south slope of the hill below the summit along Knob Road at approximately 700-860 ft. elevation. Despite the relatively short distance from the summit, no *C. lucia* individuals were found below the summit along Knob Road. This suggests that *C. lucia* may have a stronger inter-dependency on mound-building ants on the summit than *C. ladon*.

Subsequent early-spring surveys on Old Knob on April 11, 2008 and April 18, 2009 revealed the same pattern, with only *C. ladon* and *C. lucia* evident at the summit and only *C. ladon* along the roadway at lower elevations. In 2008, several ova were obtained from an unidentified female confined with *Prunus serotina* flower buds. The larvae were first reared on flower buds, but were later switched to eriophyid mite galls. These produced a “false (lab-induced) second generation” of *C. ladon* adults (Figs. 7, 13, 19) with all male specimens displaying the unique dorsal scale character of *ladon* forewings. It was subsequently observed that both *C. ladon* and *C. lucia* utilize *Prunus serotina* flower buds and eriophyid mite galls at the summit site.

On April 6, 2010 at the summit, several adults of what appeared to be spring form of *C. neglecta* (Figs. 5, 11, 17) were found flying with *C. ladon* and *C. lucia*, and also in close association with *Prunus serotina*. Several females of the three distinct phenotypes were confined with *Prunus serotina* buds in separate containers and many ova were obtained. Unfortunately, the eggs of only one female successfully emerged, while the rest failed to emerge. It is unclear why the remainder did not emerge; mold or fungal infection is suspected or the females may not have mated. The surviving larvae from the single female yielded typical summer form *C. neglecta* adults (males distinguished by typical *neglecta* dorsal forewing scale structure) (Figs. 8, 14, 20), which emerged over the period from May 20 to 29. An additional trip was made April 14, 2010, but no females were found. A trip to Old Knob on May 27, 2010 detected a second (summer) brood of *C. neglecta* was flying. This confirmed a spring flight (April) and subsequent late May flight of *C. neglecta* occurred at Old Knob. Both broods apparently utilized *Prunus serotina*. Even later summer broods of *C. neglecta* also occurred at Old Knob. These have not been studied other than the collection of specimens to confirm the presence of additional broods of *C. neglecta*.

On May 5, 2011, a worn female of an unidentifiable phenotype was collected on Old Knob and confine on developing flower buds of *Prunus serotina*. Several eggs were obtained and larvae were reared to maturity on both flower buds and leaf mite galls. Many larvae perished during the course of rearing due to cannibalism. Two “false second generation” adults of *C. lucia* (Figs. 6, 12, 18) eclosed on June 6, 2011. The male displayed a dorsal forewing scale structure identical to spring brood individuals.

The fact that three different *Celastrina* populations occupy the same ecological niche in Virginia without apparent hybridization or intergradation is strong evidence of reproductive barriers maintaining species-level distinctness. It is surmised that a small isolated *C. lucia* population on the summit of Old Knob would have been obliterated long ago by natural hybridization, if this were to have occurred. A fourth species, *C. neglectamajor*, was also recorded on Old Knob, but not at the study site. Two individuals were collected on the lower portion of Knob Road on May 12, 2009. Due to the close proximity of the *neglectamajor* colony, there is a high likelihood that stray individuals briefly traveled into the study site. The host plant (*Cimicifuga racemosa*) of *C. neglectamajor* (Pavulaan & Wright, 2001) was not present on the summit.
**Celastrina lucia.** Fig. 3:♂ (d), 4/14/2010, Old Knob, Gore, Frederick Co., VA. Fig. 9:♀ (d), 4/6/2010, Old Knob, Gore, Frederick Co., VA. Fig. 15: typical spotted form ♂ (v), 4/6/2010, Old Knob, Gore, Frederick Co., VA. Fig. 6: lab-reared false second-generation “summer” form ♂ (d), ex-ova, em: 6/6/2011, Old Knob, Gore, Frederick Co., VA. Fig. 12: lab-reared false second-generation “summer” form ♀ (d), ex-ova, em: 6/6/2011, Old Knob, Gore, Frederick Co., VA. Fig. 18: lab-reared false second-generation “summer” form ♂ (v), ex-ova, em: 6/6/2011, Old Knob, Gore, Frederick Co., VA. Fig. 21: ♂ (d), 4/11/2010, George Thompson WMA, Markham, Fauquier Co., VA. Fig. 22:♀ (d), 5/14/1987, Tanners Ridge (part of Blue Ridge), near Stanley, Page Co., VA. Fig. 24: f. marginata ♀ (v), 4/6/2010, Old Knob, Gore, Frederick Co., VA. Fig. 25: f. lucia ♂ (v), 4/18/2009, Old Knob, Gore, Frederick Co., VA. Fig. 26: f. lucia ♀ (v), 5/3/2014, Great North Mountain, near Hayfield, Frederick Co., VA. *Celastrina ladon.* Fig. 4:♂ (d), 4/23/2007, Old Knob, Gore, Frederick Co., VA. Fig. 10:♀ (d), 4/23/2007, Old Knob, Gore, Frederick Co., VA. Fig. 16:♂ (v), 4/23/2007, Old Knob, Gore, Frederick Co., VA. Fig. 7: lab-reared false second-generation “summer” form ♂ (d), ex-ova, em: 5/27/2008, Old Knob, Gore, Frederick Co., VA. Fig. 13: lab-reared false second-generation “summer” form ♂ (d), ex-ova, em: 5/26/2008, Old Knob, Gore, Frederick Co., VA. Fig. 19: lab-reared false second-generation “summer” form ♀ (v), ex-ova, em: 5/27/2008, Old Knob, Gore, Frederick Co., VA. Fig. 23: margined form ♂ (v), 4/23/2007, Old Knob, Gore, Frederick Co., VA. *Celastrina neglecta.* Fig. 5: spring form ♂ (d), 4/6/2010, Old Knob, Gore, Frederick Co., VA. Fig. 11: spring ♀ (d), 4/6/2010, Old Knob, Gore, Frederick Co., VA. Fig. 17: spring form ♂ (v), 4/6/2010, Old Knob, Gore, Frederick Co., VA. Fig. 8: lab-reared summer form ♂ (d), ex-ova, em: 5/20/2010, Old Knob, Gore, Frederick Co., VA. Fig. 14: lab-reared summer form ♀ (d), ex-ova, em: 5/27/2010, Old Knob, Gore, Frederick Co., VA. Fig. 20: lab-reared summer form ♂ (v), ex-ova, em: 5/20/2010, Old Knob, Gore, Frederick Co., VA. Dorsal = (d), Ventral = (v).
COMPARISON OF CELASTRINA TAXA

A comparison of the three taxa including the summer form of C. neglecta is presented here. All three sympatric species display a similar phenotype to the naked eye during their early spring flight periods. Only C. ladon males are easy to distinguish from C. lucia and spring form C. neglecta.

**Celastrina ladon.** Adults (Figs. 4, 10) at the study location are typically of the spotted ventral hindwing phenotype (Fig. 16), showing no tendency to develop darkened ventral hindwing margins or dark ventral hindwing discal patches, which are more frequent in the northern portions of the species’ range. The spotted form is referred to as form "violacea" (W.H. Edwards, 1866), which is technically a species-level junior synonym of the name ladon. One individual of the dark-margined form [form "marginata" of authors] was collected here (Fig. 23). Males of C. ladon are easily distinguished from all other blue Celastrina species by their unique male wing scale structure (Fig. 2). Adults of C. ladon tend to be slightly more violet-blue in color than either C. lucia or C. neglecta, thus the name violacea.

C. ladon is univoltine throughout its range. In lab rearing, an artificial summer phenotype can be produced (Figs. 7, 13, 19), which bears the unique ladon male dorsal wing scale structure and has only a superficial resemblance to the natural summer form of C. neglecta (which does not bear the unique male dorsal wing scale structure). No adults resembling the lab-reared false summer form of C. ladon have ever been found at the study site.

**Celastrina lucia.** Adults (Figs. 3, 9) at the study location are generally of the spotted ventral hindwing phenotype (Fig. 15), with some individuals displaying darkened ventral hindwing margins [described as form marginata (W. H. Edwards, 1883)] or rarely the dark ventral hindwing discal patch (form "lucia"), which is characteristic of C. lucia in northern latitudes. The ventrally-spotted form has been incorrectly referred to as form "violacea" of authors, however the name violacea technically applies only to the spotted form of C. ladon. Two interesting individuals are figured from here, one is form "marginata" (Fig. 24) and the other is form "lucia" (Fig. 25). Males of C. lucia are easily distinguished from males of C. ladon which bear the unique wing scale structure (Fig. 2). Adults of C. lucia tend to be noticeably more metallic blue in color than C. ladon when fresh, but have a peculiar tendency to become more violet when flight-worn with age. Males of C. lucia can be distinguished from males of spring form C. neglecta by the lack of very distinct white wing veins along the leading forward edge of the dorsal side of the forewing, which are characteristic of spring form C. neglecta. Also individuals of spring form C. neglecta bear clear hindwing fringes, while in C. lucia they are either darkened or checkered black and white. Females are generally difficult to distinguish, as both C. lucia and C. neglecta females tend to be very similar in appearance, both being noticeably lighter blue than females of C. ladon. In spread series, C. lucia females from the Appalachian region are noticeably smaller than C. neglecta females and have narrower black outer margins on the dorsal forewing.

C. lucia is univoltine throughout its range, and is thus presumed to be univoltine at the study site. In the lab, an artificial summer phenotype can be produced (Figs. 6, 12, 18), which bears no resemblance to the natural summer form of C. neglecta. No naturally occurring individuals resembling the lab-reared false summer form have ever been found at the study site.

**Celastrina neglecta spring form.** Adults (Figs. 5, 11) at the study site are typically of the distinctly-spotted ventral hindwing phenotype (Fig. 17), showing no tendency to darkened ventral hindwing margins or dark ventral hindwing discal patches. The ventrally-spotted form has traditionally been referred to as form "violacea" by authors, but the name violacea technically applies only to the ventrally-spotted form of C. ladon. Males of C. neglecta spring form are easily distinguished from males
of *C. ladon* which have the unique forewing scale structure (Fig. 2). However, in all other respects, they are extremely similar to *C. ladon* and very difficult to distinguish by ventral markings alone. Adults of both sexes of *C. neglecta* spring form tend to be bluer in color than *C. ladon*, but similar in color to *C. lucia*. Males of *C. neglecta* spring form can often be distinguished from sympatric males of *C. lucia* by the presence of distinct white veins along the leading edge of the dorsal forewing. This character state is useful when examining fresh individuals, but is not always reliable as veins become subdued with age. Also *C. neglecta* spring form individuals have clear white fringes on the hindwing edge. In *C. lucia* these fringes tend to be darkened or checkered and in *C. ladon* they also tend to be darkened, but may appear light in some individuals. Females are generally difficult to distinguish from *C. lucia*, but in general they are larger and have broader black outer margins on the dorsal forewing.

*Celastrina neglecta* summer form. The summer form of *neglecta* is uniquely different from the three spring phenotypes, in that the venter is very white, and dark markings are reduced to mere dashes and dots (Fig. 20). On the dorsum, the males and females both display characteristic distribution of white insuffusion on the hindwings, arranged in rays between the wing veins (Figs. 8, 14). This phenotype has been produced in reared offspring of spring form females throughout the range of *neglecta*.

The summer form of *C. neglecta* was recorded during surveys conducted on the following dates: June 6, 2006 and May 27, 2010 (associated with mite galls on both dates); June 7 & 23, July 1, and August 29 in 2010.

**ADDITIONAL NOTES ON CELASTRINA LUCIA IN VIRGINIA**

In addition to the Old Knob study site, *C. lucia* has been confirmed from six additional sites in Virginia, totalling four counties (Fig. 28). It is interesting to note that all of these populations are closely-associated with *Prunus serotina* with the exception of the Great North Mountain population:

**Great North Mountain, near Hayfield, Frederick County, VA.** A visit to the top of this ridge on April 21, 2014 found a colony of *C. lucia* flying in an area of remnant Pitch Pine/Scrub Oak barren at 2300 ft. elevation. Most of the forest canopy now consists of various Oak species with an understory of Scrub Oak (*Quercus ilicifolia*) and Mountain Laurel (*Kalmia latifolia*). Interestingly, unlike the other Virginia *C. lucia* sites, *Prunus serotina* was not observed to be a primary component of the ridgetop forest. Rather Blueberries (*Vaccinium* sp.) were very common everywhere especially along the roadside edges and are suspected to be the host. Five males were collected and an additional 15 individuals were observed in the same area, presumably all *C. lucia*. During a return trip on April 24, 2014, six males were collected and an additional 12 individuals were observed. A final trip on May 3, 2014, following several days of damp rainy weather, only found one worn male and three fresh females were found. One of the females was form “*lucia*” with the distinctive ventral hindwing patch (Fig. 26).

**Cacapon Mountain, north of Cross Junction, Frederick County, VA.** A visit to the top of this ridge, literally within a few hundred feet of the very northern border of Virginia on May 3, 2014 produced a single male specimen of *C. lucia*. *C. lucia* occurs more commonly northward along the top of this same ridge on the West Virginia side of the border.

**Lake Thompson area, lower east slope of Blue Mountain, George Thompson Wildlife Management Area, north of Markham, Fauquier County, VA.** Few *C. lucia* adults have been documented at this location among thousands of spring-flying *Celastrina* individuals that were either collected, examined in-hand (net/release) or observed (resting only) at very close range. All *C. lucia* specimens have been collected toward the end of their flight period with most adults being faded from age, thus leading one to suspect they have flown in from some distance, likely from somewhere on Blue Mountain or along the Blue Ridge. All specimens were collected along woodland trails at the fairly low elevations of 970-
1200 ft. Interestingly, no C. lucia specimens have ever been collected on top of Blue Mountain. These specimens are based on adult phenotype (Fig. 21) and are a close match to C. lucia found to the north in central Pennsylvania or on the Allegheny Plateau in West Virginia. Prunus serotina is present in the woodlands here and is the only host tree on which ovipositions were observed. Blueberries (Vaccinium sp.) are uncommon in the dense mixed Appalachian woodland of Blue Mountain; thus they are likely not utilized. Collection dates: April 10, 1999; April 29, 2000; April 13, 2002; April 27 & May 3, 2003; April 17, 2004; April 11, 2005; April 13 & 14, 2006; May 17, 2008; April 17, 2009; April 11 & 23, 2010.

Tanners Ridge (part of the Blue Ridge), near Big Meadows Recreational Area, Shenandoah National Park, Page County, VA. A single female specimen of form "marginata" was collected at this location [under permit], just off Skyline Drive at an elevation of 3387 ft. In general, Celastrina are noticeably scarce along the top of the Blue Ridge, except in late May and early June when C. neglectamajor flies. Prunus serotina is common on the crest of the Blue Ridge and several sightings of unidentified Celastrina adults around P. serotina suggest this might be the primary host for C. lucia or other Celastrina species on the Blue Ridge. Blueberries (Vaccinium sp.) are also very common in open places at higher elevations, but apparently not utilized by Celastrina. Over several years (1985-2008), I have carefully scanned forest clearings, roadside edges and a power line cut in the forest around the Big Meadow, but have seen no Celastrina associated with Vaccinium. Collection date: May 5, 2001.

Tanners Ridge (part of the Blue Ridge), along Route 682, near Stanley, Page County, VA. Early in this study I explored a location on the west slope of the Blue Ridge just outside the National Park, where one could collect fairly high in elevation (up to 2884 ft.). Several female Celastrina were collected along this road in May, 1987, which for several years I kept in a papered series of C. ladon from the location. Only upon examination of these specimens, which were mounted for this report, did I discover three specimens with features distinctly those of C. lucia, i.e. very light blue dorsal color with narrow black dorsal forewing margins (Fig. 22). Collection dates: May 14 & 16, 1987.

Shenandoah Mountain, west of Briery Branch, Rockingham County, VA. Several individuals were collected along the upper portion of State Road 924 (at elevations of 2062 ft.-3467 ft.) and also along Forest Road 85 (approximately 3845 ft.) going north along the ridge top, which delineates the Virginia-West Virginia border. A female captured on April 29, 2001 was confined with Prunus serotina flower buds in a rearing container; eggs laid in confinement subsequently produced several "false summer brood" adults on June 1, 2001. These individuals resembled lab-produced false brood adults from C. lucia populations in West Virginia, Pennsylvania and New Jersey. Collection dates: April 29, 2001; May 4 & 6, 2006.

HISTORICAL RECORD

In The Butterflies of Virginia, under Cyaniris argiolus pseudargiolus, Clark & Clark (1951) wrote: "We have taken the form lucia only in western Frederick County in Virginia." It is presumed that the authors were referring to the ventral hindwing dark-patched form. However, since the patched form has been recorded in all Celastrina species in the eastern region of the United States, it is not known whether the authors collected C. lucia, C. ladon or C. neglecta. Assuming they did have a specimen of C. lucia in hand, that would be the first record of C. lucia in Virginia. The present study would corroborate the older Frederick County record.
As an extension of this study, an effort was attempted to corroborate previous host intolerance findings, specifically that spring Celastrina neglecta females do not oviposit on and neonate C. neglecta larvae do not utilize Cornus florida (Flowering Dogwood), a common C. ladon host. On April 21, 2014, a survey of the Old Knob study site found no C. lucia, but two females of C. ladon and one female of C. neglecta were captured. These females were separately confined in containers with cuttings of C. florida flower buds. The two C. ladon females readily oviposited on C. florida within the first day, whereas the C. neglecta female refused to oviposit on the same plant under identical conditions. On the third day of confinement, cuttings of Viburnum prunifolium (a documented C. neglecta host in this region) were added to the container containing the female C. neglecta. She immediately oviposited approximately 50 eggs on V. prunifolium within 24 hours, while still ignoring C. florida. Subsequently, individual flower buds containing C. neglecta eggs were removed and strategically placed onto cuttings of C. florida flower buds so that newly-hatched larvae would have the direct choice of feeding on Cornus florida. Newly hatched larvae were also transferred from the V. prunifolium buds to C. florida, thus leaving them no choice but to feed on C. florida. By May 3, 2014, most of the C. neglecta larvae had hatched and ignored the C. florida, subsequently starving and leaving only shriveled corpses on the container sides. A few remaining larvae attempted to feed on C. florida, not on the flower buds but rather boring into the base of the underside of the white bracts or into the basal portion of the flower buds. By May 8, 2014, all C. neglecta larvae had died. A previous attempt in 2013 at getting C. neglecta larvae to accept C. florida similarly failed, with all larvae preferring to starve rather than to eat C. florida. An earlier 2012 observation of ovipositional behavior by captive females also found that C. neglecta females refused to lay eggs on C. florida. This finding demonstrates that Cornus florida is not acceptable to C. neglecta, and the plant likely has toxic properties to certain Celastrina species.

**CONCLUSION**

Based on observations made during this study and reported here, the following statements are presented as clear succinct conclusions:

(1) The three taxonomic entities Celastrina ladon, C. lucia and C. neglecta behave as distinct full species in sympatry in northern Virginia.

(2) The following species arrangement is hereby confirmed:

*Celastrina ladon* (Cramer, 1780)
*Celastrina lucia* (W. Kirby, 1837)
*Celastrina neglecta* (W. H. Edwards, 1862)

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I wish to sincerely thank David M. Wright who critically reviewed multiple drafts of this manuscript and offered several helpful suggestions.

**LITERATURE CITED**


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*Fig. 27:* Ant mounds and young *Prunus serotina* trees on summit of Old Knob (elev. 1300 ft.), near Gore, VA. Photo taken on April 6, 2010 shows very early spring vegetative progression, with only *Prunus* showing flower bud development, necessary for oviposition activity by *Celastrina.*

*Fig. 28:* Map showing known county distribution of *Celastrina lucia* in Virginia, as of June 16, 2014.
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**Cercyonis pegala agawamensis** (Satyridae): A new butterfly subspecies from the coastal salt marshes of the northeastern United States of America

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**Abstract:** A new subspecies of the Common Wood Nymph (*Cercyonis pegala*) from the New England coastline in the northeastern region of the United States is described on the basis of phenotype, behavior, habitat, seasonality, flight period, and phenology. *Agawamensis* is univoltine as in all *pegala*. The newly described subspecies occurs almost entirely in coastal salt marshes and estuarine meadows, quite atypical as understood for *Cercyonis pegala*. The newly described subspecies *agawamensis* predominately prefers and occurs within large open salt marshes but can also be found in similar habitat along smaller tidal rivers, coastal inlets and streams.

**Additional key words:** phenotype, behavior, habitat, seasonality, flight period, phenology, coastal salt marshes.

**INTRODUCTION**

A new subspecies of the Common Wood Nymph is described from the New England coastline in the northeastern region of the United States. The new subspecies is described on the basis of phenotype, behavior, habitat, and voltinism. As noted above, *agawamensis* is univoltine as in all *pegala*. Within the scope of this paper we have not as of yet examined the early stages or analyzed the DNA, in order to determine if further splitting of this subspecies from *Cercyonis pegala* is warranted. We point out, however, that there is precedence for such an assumption: Two rather restricted taxa occurring locally in similar salt marsh habitats in New Brunswick and adjacent coastal Gaspe, Quebec have been recently awarded specific status on the basis of similar criteria to those noted above. These taxa are the Maritime Ringlet (*Coenonympha nipisiquit* McDunnough) which has been split from the Common Ringlet (*Coenonympha tulii inornata* W. H. Edwards), and the Dos Passos Copper (*Lycaena dospassosi* McDunnough), which has been split from the Dorcas Copper (*Lycaena dorcas* W. Kirby) or more specifically from the Clayton’s Copper (*L. dorcas claytoni* A. E. Brower) - known only from a few calcareous bog localities in northern Maine and in adjacent New Brunswick.

**OVERVIEW OF NAMED TAXA OF “WOOD NYMPHS” OCCURRING IN EASTERN NORTH AMERICA:**

This paper is not intended as an in-depth investigation of taxonomic issues involving the *Cercyonis pegala* group in the East, but is intended to only highlight the following discussion in deference to future anticipated publications on this subject. According to Klots (1951), there have been three recognized subspecies of *Cercyonis pegala* in New
England. These are C. p. alope (Fabricius), C. p. maritima (W. H. Edwards) and C. p. nephele (W. Kirby). In addition to the above three subspecies, a number of subspecies of Cercyonis pegala have been recognized and described from the Eastern United States. These include the nominate subspecies C. p. pegala (Fabricius), C. p. abbotti F. M. Brown, C. p. carolina (F. Chemock & R. Chemock) and C. p. pegala texana (W. H. Edwards).

The following subspecies (some in debatably infrasubspecific forms) were described from the eastern United States and southern Canada (in part from Klots 1951):

pegala (Fabricius, 1775): TL “America”, defined as vicinity of Charleston, South Carolina (F. M. Brown, 1966). Klots (1951) listed this as occurring from “Florida and Gulf States n. to North Carolina and (blend zone with alope) New Jersey in coastal plain; northern limits in Mississippi Valley uncertain; large, brightly colored, with tendency to reduce or lose lower ocellus in orange FW eye patch [in males]; ocellus of HW above prominent”.

Abbotti F. M. Brown, 1969: TL Chipley, Washington Co., Florida. Like pegala but with a tendency to reduce or lose the lower ocellus in FW eye patch in both males and females (Mather, 1966; Sourakov, 1995). Florida, but apparently occurs along the Gulf Coast to at least Mississippi.

alope (Fabricius, 1793): TL “India”, corrected to Burke-Screven-Bulloch counties, Georgia (F. M. Brown 1966). Klots listed this as occurring from “Virginia (mountains) and New Jersey n. to Maine and Quebec (coastal plain) and New York (inland); blends with maritima along its eastern limits, with nephele northward, with carolina and pegala southward; smaller and not so richly colored as nominate pegala, both ocelli in orange of FW well developed”. The HW ocellus is typically smaller. Harry Pavulaua (pers. comm.) describes alope as “...a population of mutts occurring between coastal pegala and carolina of the southern Appalachians...” that “...constitutes an essentially highly variable southern Piedmont group of populations...” The FW eye patch is more typically yellow in alope. What is further confusing about alope, which also warrants study and probable further revision is the fact that the TL of alope is located only two counties west of the TL of pegala in the State of Georgia.

maritima (W. H. Edwards, 1880): TL Oak Bluffs, Martha’s Vineyard, MA. Like alope, but smaller, darker, and with FW eye patch deeper orange. Klots (1951) defined the range as: “Coastal edge, Maryland to Maine”, but this phenotype apparently extends west along the northern limits of the lighter (yellowish) eye-patched southeastern populations of C. pegala at least to western Pennsylvanina (see description of ochracea below) also into Ohio and southern Michigan and perhaps as far west as Illinois.

nephele (W. Kirby, 1837): TL “Canada”, defined as “the extreme western end of North Channel, Lake Huron...in Ontario, Canada” (F. M. Brown 1966). Klots (1951) describes nephele as: “FW dark, lacking yellow orange patch (but ocelli present); New York and New England (mountains) n. and w. through northern Mississippi Valley and Canada to Hudsonian Zone”.

texana (W. H. Edwards, 1880): TL “Texas”, defined as Bastrop, Bastrop Co., Texas (F. M. Brown 1964). Klots (1951) provides the range description as: “Southwest, Texas n. to Kansas; like [nominate] pegala but markings beneath more clear cut and distinct; paler colored”.

carolina (F. Chemock and R. Chemock, 1942): TL Conestee Falls, North Carolina. Southern Appalachian Mountains and inner Piedmont, north to about the Washington D. C. region. Klots (1951) described: “Paler brown than in alope and pegala, a minor form”. The FW eye patch is creamy white. Apparently intergrades with alope to the east (central North Carolina), maritima toward the northeast (central Maryland), nephele to the north (western Pennsylvania), and with olympus to the northwest.

olympus (W. H. Edwards, 1880): TL “...between New York and Illinois...” F. M. Brown limited the TL to Chicago, IL. This subspecies has been considered as occurring across the Upper Austral and Transition Zones of the upper Midwest, west to the eastern slopes of the Rocky Mountains. Olympus is suggestive of nephele except that the FW eye patch, typically lacking in nephele, is suffused by dark orange-red or reddish brown scales,
sometimes tending toward yellowish, with a definite reddish or purplish-red iridescence on the dorsal surfaces of the wings especially in fresher individuals. The dorsal FW ocelli as well as the ventral ocelli on both wings are well developed. The ground color is very dark. We have taken specimens that are probably best referred to as *olympus* in southwestern Ontario (Essex Co.) and adjacent southeastern Michigan, in old dry shrubby fields bordering deciduous woodlands. Populations further east into southern and eastern Ontario (i.e., Elgin Co. and eastward across the northern shores of Lake Erie) are perhaps also referable to *olympus*, while we have taken specimens in southwestern Maine (i.e., Streaked Mountain, Oxford Co., el. 900 feet) which could be referred to as either *olympus* or *maritima*. Specimens along the north shore of Lake Ontario feature a brown ground color with the FW eye patch yellow; these intergrade northward to *nephele*, southward and southeastward they appear to intergrade to *alope*.

*ochracea* (F. Chemlock & R. Chernock, 1942): TL Washington Park, Providence, Providence Co., R. I. This was originally described as a form of *alope*. Chemlock and Chernock (1942) noted: “...on examination of Edwards’ type material, the name *maritima* cannot be applied to this form”. However, specimen series from both Rhode Island and coastal islands of Nantucket and Martha’s Vineyard (TL of *maritima*) show a similar range of variation. Presently, it is probably best considered as a form (or synonym) of *maritima*. The relationship and comparative distributions of *ochracea* and *maritima* warrant review.

Howe (1975) retained the use of the name *abbotti* to describe the “large and beautiful subspecies” occurring from Florida and Georgia and along the Gulf Coast to Mississippi, but sunk, however, each of the various forms occurring north of the range of *abbotti* in the East into *pegala*, referring to them as “phenotypes of *pegala*”. He stated that the typical form of *pegala* is restricted to the southeast coast, and stated that this form is “probably the same as *maritima*”, pointing out that *pegala* “…tends to have only one ocellus whereas *maritima* has two and is usually smaller in size”. He further stated that “these coastal forms with a dark orange-yellow postmedian eye patch on the forewings intergrade with the inland form with a bright yellow patch which is called ‘*alope*’; and that “in turn in the north Atlantic states *alope*” blends with a form that lacks the yellow patch and is called ‘*nephele*’ (probably incorrectly!). Howe also referred to *olympus* as a synonym, presumably of *pegala*, and recognized *carolina* as a “…larger paler form from the southern Appalachians.”

While there is probably some merit to Howe’s conclusions, it is our opinion that the above is somewhat oversimplified. Certainly *maritima* is not the same thing as *pegala*, at least from the coastal Carolinas and Virginia, and that the name *nephele* probably does need to be retained as a valid taxon from the northern, Canadian Zone regions of the range of the species.

Perhaps, rather predictably, Scott (1986) condensed his treatment of eastern *C. pegala* even farther than Howe (1975). Scott recognized only TWO subspecific names: Those being *nephele*, consisting of patchless populations ranging from “Nova Scotia, c Penn., and Ill. north and west...” and *pegala* - which he ascribed to ALL yellow and/or orange patched eastern populations south of his stated range of *nephele*.

Most currently, the Butterflies of America website (http://www.butterfliesofamerica.com) offers a rather reasonable treatment of eastern *C. pegala* based upon the landmark ‘Catalogue of the Butterflies of the United States and Canada’ (Pelham, 2008), which is updated frequently on the website to reflect recent taxonomic changes. Pelham recognized the following subspecies: *pegala* (southeastern - presumably Florida to Massachusetts), *texana* (southwestern - Texas to Kansas), *alope* (regions north and west of *pegala*, including *carolina* and *maritima* as synonyms), *olympus* (Upper Midwest, including the synonym *borealis*) and *nephele* (north). While certainly more comprehensive than the rather scant treatments rendered by Howe and Scott, these treatments also appear to have apparent flaws but are based upon current literature. For example, *maritima* and *abbotti* are certainly not the same thing biologically and cannot both be synonyms of *pegala*; likewise *ochracea* probably can be listed as a synonym of *maritima*.

Klots (1951) also noted that “…the above forms [that Klots refers to] represent merely averages. Intermediates not only occur abundantly in blend zones but also may occur anywhere in an area. Sometimes, within blend zones, local colonies of one form or another seem to be segregated out...” Caution should be taken here not to
misinterpret and expand the concept of intermediates to distant ends of the various clines seen in pegala, as many authors have. Intermediates appear primarily in blend zones. For example, there are no known records of “nephele” forms in the southeastern U.S. and conversely no known records of “abbotti” forms in Canada. Many authors interpret the presence of intermediates in bland zones as applying species-wide, which we suggest is an incorrect application of the concept of intermediates. Klots also suggested “...a terrific amount of work is needed on the distributional problems of this clinal species...”

At this point, we would like to point out - in response to the treatment of some authors such as Cech & Tudor (2005) and Bouseman & Sternburg (2001) - the exact parallel between the relationship of Cercyonis pegala/nephele (and/or maritima/nephele) and Limenitis arthemis/astyanax in New England. In virtually all butterfly field guides since the time of Klots (1951), astyanax has been treated as a subspecies - and not a form - of arthemis; while in at least the publications referred to above, nephele has been defined as merely a form of pegala. This, despite the fact that there exist in BOTH cases, within arthemis/astyanax and pegala/nephele, extensive and broadly overlapping subspecies ranges or zones of hybridization of each of the two pairs. In New England, for example, the zone of hybridization between arthemis and astyanax extends from almost the south coast of Connecticut, Massachusetts and Rhode Island - where traces of the white bands along with other subtle characters of arthemis can be seen in specimens of astyanax - to at least the latitude of the Kancamagus Highway of northern New Hampshire (we have actually taken a partially white-banded hybrid specimen that otherwise resembles astyanax even further north of this at the Scott Bog region of extreme northern New Hampshire only a few miles from the Quebec border at an elevation of 2,000 feet, well into the Canadian Life Zone). In terms of the Cercyonis, we have similarly found specimens of nephele with partial to full development of the orange forewing eye patch at least as far north as the vicinity of Streaked Mountain near Buckfield, Oxford Co., Maine at elevation of 950 feet. At the same time we have also found totally dark specimens of nephele well southward into west central Massachusetts in the vicinity of Hawley in Franklin Co. at elevation of approximately 800 feet, and the nephele phenotype has been found in Rhode Island; while fine examples of alope with fully developed yellow forewing eye patches, in some cases with the lower eyespot reduced, have been taken substantially north of this in the hilly regions west of Brattleboro, Windham Co., Vermont.

In conclusion of this discussion, we can state that in New England, among the previously recognized subspecies, only nephele in the more northern Canadian Zone regions appears to be more or less well distinguished, while alope and maritima have been widely subject to confusion and appear to be rather poorly differentiated within the more southern and central New England regions. We have, however, somewhat frequently noted individuals in southeastern (or even occasionally in northeastern) Massachusetts and in southeastern Vermont featuring an extensive yellow FW eye patch with the LOWER eyespot reduced in size in males, suggestive of the nominate southeastern pegala; or specimens in western Massachusetts which are essentially brown, lacking the FW eye patch entirely, but with the FW ocelli large and pupiled in bright blue scales.

*Cercyonis pegala agawamensis* Arey and Grkovich, new subspecies

**ETYMOLOGY.** The type specimens (holotype and allotype) are designated from the former historical settlement and territory of the Native American Agawam Tribe in northeastern Essex County, Massachusetts (ca. 1638) that has historically encompassed the current towns of Ipswich, Rowley, Essex, Newbury, Byfield and Georgetown. We propose the subspecies scientific name as *agawamensis* and the common name as the *Salt Marsh Wood Nymph*.

**DESCRIPTION.** Male somewhat distinct from nearby coastal and inland populations of alope and maritima; female quite noticeably distinct. Both sexes demonstrate a general brighter “southern look” in comparison to their neighboring inland populations occurring outside of salt marsh habitats.

**Male:** FW length 25-26 mm. **Upperside:** Ground color brown. FW apex typically somewhat more pointed than other pegala subspecies. FW eye patch fairly broad (width of patch 10 mm along costal margin), yellow to slightly yellow-orange; eyespots well developed, somewhat small but without tendency in most specimens for lower
eyespot to be reduced; occasional specimens however do have lower eyespot reduced or even absent. Pupils sharply defined and centered in light blue. Upper eyespot sometimes reduced in size. HW eyespot well developed, small but with well-defined outer circle of lighter orange-brown; pupiled in light blue. HW submarginal dark line prominent and well defined. **Underside:** Ground color lighter brown, particularly on outer half of HW. FW yellow eye patch prominent and wide, lower border at inner margin of FW wide and straight. Eyespots well-defined and round in shape, smaller with sharp edges not diffuse as in nearby alope and maritima populations. Dark lines bordering inner and outer edges of FW eye patch prominent; eyespots well-defined and sharply pupiled in light blue. HW eyespots smaller than in typical southeastern Cercyonis but well-defined. Striations sharp and crisp; median and submarginal dark lines prominent.

**Female:** FW length 29–31 mm. **Upperside:** Ground color lighter brown than in male; a lighter shade along outer half of HW beyond cell area. FW apex slightly rounded but FW broad and squarish. FW eye patch broad (12–15 mm along costal margin), extending nearly to (and sometimes into) the cell area, color of patch yellow to slightly orange-yellow; yellow patch extends into marginal area in some specimens; margins of patch very sharply defined and not diffuse. FW eyespots as in male, sharply defined without diffuse edges, pupiled in blue; upper eyespot round, lower eyespot somewhat squarish and only somewhat larger. [In nearby alope and maritima both FW eyespots are larger, rounded with diffuse edges, lower eyespot is often significantly larger than upper.] HW eyespot generally smaller than in nominate pegala but with well-defined orange-brown outer circle; submarginal dark line prominent; occasional second smaller eyespot above the main HW eyespot sometimes present. **Underside:** Ground color lighter brown and somewhat brighter, striations heavy and well-defined; FW eye patch very broad and bright yellow, eyespots as on the above surface, widely pupiled in light blue. Small third eyespot on FW below the lower main eyespot is present. There is no apparent tendency for lower eyespot to be reduced. HW eyespots are typically prominent, but apparently not quite as well developed as in the male.

**Phenotypical comparison between C. p. agawamensis and C. p. maritima:** We examined a series of males and females of agawamensis from the coastal salt marshes of northeastern Massachusetts and southern New Hampshire, in order to compare these to males and females of maritima from nearby higher “dry old fields”. Males of agawamensis average somewhat distinct from males of maritima; while females of agawamensis are particularly distinct from females of maritima.

Males of agawamensis are somewhat larger and average somewhat lighter above than in maritima; agawamensis males are also somewhat two-toned on the hindwing with the postmedian dark hindwing line often present. Males of maritima have the ground color above darker and more uniformly colored. The orange-yellow forewing eye patch is typically wider and more rectangular in agawamensis males, with the borders of the patch tending to be more clear-cut. The two forewing eyespots are smaller and tend toward equal size in agawamensis. Beneath, the above described characters are essentially repeated: agawamensis males tending to be more two-toned on the hindwing making the hindwing eyespots slightly more prominent; the forewing eye patch tends to be more yellow with less orange tint than in maritima.

Females of agawamensis are, as noted above, particularly distinct in comparison to females of maritima, and are suggestive of smaller examples of females of texana. Females of agawamensis are significantly larger than those of maritima, have much broader wings and have the forewing eye patch very broad. The ground color above is lighter and brighter with the hindwing tending toward two-toned as in the males. The forewing eyespots are rather clear cut and distinct and tend toward being equal in size; while in maritima the eyespots are larger with indistinct edges especially in the lower eyespot. Females of agawamensis have the anal eyespot above small in comparison to females of pegala but always well-defined and circled in orange; maritima females typically lack this eyespot altogether. Beneath, agawamensis females are rather brightly and robustly colored, again suggestive of texana, and differ conspicuously from females of maritima. Agawamensis females below have the forewing eye patch very large, broad and clear cut. The patch typically extends from the postmedian to the submarginal dark line and often extends beyond the submarginal line almost to the outer margin. The eyespots tend to equal size and, as above, are clear-cut with well-defined blue-white pupils. The ventral hindwing is distinctively two-toned with clearly defined postmedian dark line; the eyespots are smaller but at least 4 or 5 are present and fairly well-defined. Maritima females are less noticeably two-toned on the hindwing, the postmedian line is less distinct and the hindwing
eyespots are typically absent except for the two near the anal angle, even these are inconspicuous. The forewing eye patch in *maritima* is narrower and does not extend beyond the submarginal dark line; the eyespots are larger and less clear-cut.

**VOLTINISM & PHENOLOGY.** Single brooded, diapausal (hibernal) first or second instar larva. The flight period of *agawamensis* commences during the latter part of the flight period of sympatric *C. pegala* alope or *maritima*; and as currently understood extends from late July through mid September. The peak flight time for *agawamensis* as noted in surveyed colonies in northeastern Massachusetts and southern New Hampshire is from 28 July to 20 August. Extreme dates of *agawamensis* as we have observed are 17 July and 26 September. Comparative phenology for *C. pegala* alope or *maritima* are very late June through mid August, with peak flights occurring from 12 July to 30 July. Extreme dates for *C. pegala* alope or *maritima* are 25 June and 8 September. These phenology and voltinism differences have strong similarity to the recent taxa split of another Satyrid genus *Megisto* (Hübner) where a late spring flight of *Megisto eurytis* (Fabricius, sensu Gatrell) or formerly called *M. cymela viola* (Maynard) is split from the early summer flight of *M. cymela* Cramer (Gatrell, 2005) and these flights often do overlap as does the *P. pegala* alope / *P. pegala* agawamensis complex.

**HABITAT AND BEHAVIOR.** Newly described subspecies *agawamensis* inhabits coastal salt marshes and estuarine meadows, which are quite atypical as understood for *Cercyonis pegala*. It predominately prefers and occurs within large open salt marshes but can also be found in similar habitat along smaller tidal rivers, coastal inlets and streams. Adults occasionally wander into adjacent habitats such as old fields, hayfields, wet meadows and open woodland margins and hammocks where they then mingle with nearby populations of *C. pegala* alope. Dominant halophilic salt marsh plant species in the northeastern U.S. are Cordgrasses (*Spartina alterniflora, S. patens, S. pectinata*), Common Reed (*Phragmites australis*), Spike Grass (*Distichlis spicata*), Black Grass (*Juncus gerardii*), Sea Lavender (*Limonium nashii*), High-Tide Bush (*Iva frutescens*), Salt Marsh Goldenrod (*Solidago sempervirens*), Siff-Leaved Quackgrass (*Agropyron pungens*), Sea Myrtle (*Baccharis halimifolia*), Seaside Gerardi (*Agalinus martima*), Glassworts (*Salicornia sp.*), Seaside Plantain (*Plantago maritima*), Salt Marsh Aster (*Aster tenuifolius*), Widgeon Grass (*Ruppi a maritima*) and Switchgrass (*Panicum virgatum*). Larval host plant(s) is (are) likely one or more types of the salt marsh grass species.

However, if habitat - along with phenotype - serves to distinguish *agawamensis*, then we must also discuss behavior to further distinguish it and differentiate it from its closest relatives. Both sexes fly in open estuarine salt marsh or along tidal waterways. Unlike almost all other populations of *C. pegala*, *agawamensis* will often seek nectar from flowering plants, both within the habitat or in nearby adjacent habitat. The adults are more likely to stray from the salt marsh habitat and into tidal marshes in the latter part of their flight period. Most frequented nectar sources include Purple Loosestrife (*Lythrum salicaria*), Joe-Pye-Weed (*Eupatorium purpurea*), Common Milkweed (*Asclepias syriaca*), Swamp Milkweed (*A. incarnata*), Red Clover (*Trifolium sp.*), Goldenrod (*Solidago sp.*), Asters (*Aster sp.*) and other composites in adjacent habitat. Nectar sources utilized within the estuarine biomes include Salt Marsh Goldenrod (*Solidago sempervirens*), Sea Lavender (*Limonium nashii*) and bordering thickets of Purple Loosestrife.

*Agawamensis*, unlike either *aloep* or *maritima* which can and do occur in significant numbers in a relatively small area of their typically DRY old field and meadow habitats, is noticeably communal and is frequently observed perching communally on salt marsh Cordgrasses and associated vegetation in large numbers over a small area. When approached, one or more individuals will take flight, followed in rapid succession by nearly all individuals making up the group. We believe that this behavior serves to confuse a potential predator by making it difficult for the predator concentrate on any one particular individual as prey. As it is perceived that the danger has passed, they will once again perch in similar fashion. While we have never observed this communal behavior in North American *Cercyonis*, we have observed similar behavior of Neotropical Satyrids (i.e. *Taygetis, Cissia, Hermeuptychia* etc.), especially toward dusk when their typically brown ground colors effectively conceal them and render this evasive and confusing behavior even more effective. The flight characteristic itself of *agawamensis* appears to differ from eastern (if not all) *Cercyonis*, in that its flight as we have observed in the salt marshes is not as “bouncy” or as “jerky” as typical *Cercyonis*, but is quicker and more straight-ahead 12 to 18 inches above the substrate, suggestive of a typical *Erebia* or *Oeneis* rather than of a typical *Cercyonis*. 

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As noted elsewhere, *agawamensis* also differs from all eastern *Cercyonis* - except for unusual populations of *C. pegala* “nephele” occurring in northern Coos Co., New Hampshire - in that it nectars freely in its salt marsh habitat. These “nephele” which we suspect should be further studied, also differ from typical *nephele* in that they also occur in rather boggy wet meadows and roadsides, nectar freely and frequently on Joe Pye Weed, Everlastings, etc. and also have a fast and rather straight-ahead “unbouncy” flight. We have observed these very blackish populations with very conspicuous ocelli in early to late August at such elevated locations as Pinkham Notch (Mount Washington, 2100 ft.) and at the Scott Bog, East Inlet Road, etc. (above 1900 ft.) near the New Hampshire/Quebec border.

**GEOGRAPHIC RANGE AND DISTRIBUTION.** Range confirmation for this subspecies is so far restricted to coastal estuarine habitat from southern Essex County, Massachusetts (vicinity of Salem) extending northward to southern York County, Maine (near Ogunquit). The greatest and most continuous concentration of this insect occurs in the more extensive coastal salt marshes north of Cape Ann in Massachusetts (Ipswich River, Parker River and Merrimack River drainages) to the New Hampshire seacoast approximately 12 to 15 Km. south of the Piscataqua River. This encompasses the coastal towns/cities of Essex, Ipswich, Rowley, Newbury, Newburyport and Salisbury in Massachusetts (Essex Co.), and the salt marshes in Seabrook and Hampton (Rockingham Co.) of the southernmost coastline in New Hampshire. This subspecies has also been confirmed in less extensive habitat south of Cape Ann in the Danvers River / North River drainage in the vicinity of Pope’s Landing (Danvers) and the tidal flats within the Forest River drainage system in Salem. Another population occurs in a more extensive estuarine meadow located in Ogunquit, Maine (Piscataqua and Kennebunk River coastal watersheds), which is speculated to be the northernmost range of *agawamensis*. Habitats south of Essex County, MA, especially in Plymouth, Barnstable and Bristol Counties, and even coastal Rhode Island have yet to be explored. However, there is a strong likelihood that they do occur in the more extensive estuaries and tidal river basins in much of this region. *Agawamensis* should also be compared to the geographically restricted island populations of subspecies *maritima* that occur specifically on the islands of Nantucket, Martha’s Vineyard, Block Island (R.I.) and Naushon Island (M.A.) located in Buzzards Bay. *Maritima* is also very common in dry weedy open field habitats on mainland Rhode Island, on Long Island, and also near Gettysburg, PA.

**TYPE LOCALITY AND TYPE SERIES.** TL: Estuarine salt marsh (open), Boston Road, Newbury (Parker River watershed), Essex Co., Massachusetts. The holotype specimen will be deposited in the McGuire Centre for Lepidoptera and Biodiversity, Gainesville, Florida. Holotype (female), allotype paratype (male) and all paratypes are identified as follows:

**MASSACHUSETTS:**

**ESSEX CO., Newbury (Parker River watershed):**

- NB001 (female): Boston Road, estuarine salt marsh (open), 7/21/2007 (HOLOTYPE).
- NB002 (male): Boston Road, estuarine salt marsh (open), 7/21/2007 (ALLOTYPE).
- NB003-NB004 (2 males): Boston Road, estuarine salt marsh (open), 7/21/2007.
- NB005-NB006 (2 females): Boston Road, estuarine salt marsh (open), 7/21/2007.
- NB011-NB012 (2 males): Boston Road, estuarine salt marsh (open), 7/20/2008.
- NB013-NB014 (2 males): Boston Road, estuarine salt marsh (open), 8/2/2008.
- NB015 (female): Boston Road, estuarine salt marsh (open), 7/20/2008.
- NB016-NB018 (3 females): Boston Road, estuarine salt marsh (open), 7/28/2013.
- NB019-NB020 (2 females): Boston Road, estuarine salt marsh (open), 8/2/2008.
- NB021 (1 male): Plum Island, salt marsh, 8/14/1999.
- NB022-NB023 (2 females): Plum Island, salt marsh, 8/14/1999.
- NB024-NB029 (6 males): Boston Road, estuarine salt marsh (open), 7/28/2010.
- NB030-NB033 (4 females): Boston Road, estuarine salt marsh (open), 7/28/2010.
- NB034-NB038 (5 males): Boston Road, estuarine salt marsh (open), 7/24/2011.
- NB039-NB042 (4 females): Boston Road, estuarine salt marsh (open), 7/24/2011.
- NB043-NB045 (3 females): U.S. Route 1, salt marsh, 8/12/2011.
- NB051-NB053 (3 females): U.S. Route 1, salt marsh, 8/8/2013.

ESSEX CO., Essex (Essex River watershed):
EX001-EX004 (4 males): Route 133, salt marsh, 7/26/2008.
EX005-EX007 (3 females): Route 133, salt marsh, 7/26/2008.

ESSEX CO., Ipswich (Ipswich River watershed):
IW003-IW004 (2 males): Argilla Road, estuarine salt marsh (open), 7/22/2007.
IW005 (female): Argilla Road, estuarine salt marsh (open), 7/22/2007.
IW006 (female): Argilla Road, old field (coastal), 7/22/2007.
IW007 (male): Argilla Road, estuarine salt marsh (open), 7/9/2013.
IW008 (female): Argilla Road, estuarine salt marsh (open), 7/9/2013.
IW009-IW011 (3 males): Argilla Road, estuarine salt marsh (open), 8/5/2013.
IW012 (female): Argilla Road, estuarine salt marsh (open), 8/5/2013.

ESSEX CO., Newburyport (Merrimack River watershed):
NP001 (female): Plum Island Turnpike, coastal salt marsh (open), 7/31/1983.

ESSEX CO., Rowley (Rowley River watershed):
RW001 (male): Route 1A, estuarine salt marsh (open), 7/27/2007.
RW002-RW005 (4 males): Route 1A, estuarine salt marsh (open), 7/19/2013.

ESSEX CO., Salem (Forest River watershed):

ESSEX CO., Salisbury (Merrimack River watershed):
SB003 (female): U.S. Route 1A, estuarine salt marsh (open), 7/20/2008.
SB004 (female): U.S. Route 1A, estuarine salt marsh (open), 7/31/2008.
SB005-SB007 (3 males): U.S. Route 1A, estuarine salt marsh (open), 7/30/2009.

MAINE:

YORK CO.; Ogunquit (Stevens Brook watershed):
OQ001 (1 male): Furbish Road, estuarine salt marsh (open), 8/1/2007.
OQ002 (1 female): Furbish Road, estuarine salt marsh (open), 8/1/2007.
OQ003 (1 male): Furbish Road, estuarine salt marsh (open), 8/1/2007.

NEW HAMPSHIRE:

ROCKINGHAM CO.; Seabrook (Blackwater River watershed):
SK001-SK004 (4 males, dark type): Route 286, estuarine salt marsh (open), 7/24/2007.
SK005-SK007 (3 males): Route 286, estuarine salt marsh (open), 8/6/2007.
SK008-SK010 (3 females): Route 286, estuarine salt marsh (open), 8/6/2007.
SK016-SK017 (2 females): Route 286, estuarine salt marsh (open), 8/6/2007.

Additional locations where C. p. agawamensis have been vouchered:


Locations where C. p. alope have been vouchered for comparison to C. p. agawamensis:


Locations where C. p. maritima have been vouchered for comparison to C. p. agawamensis:

Locations where *C. p. nephele* have been vouchedered for comparison to *C. p. agawamensis*:

**MASSACHUSETTS:** HAMPIONEIR CO.: West Hawley. **MAINE:** AROOSTOOK CO.: Sherman Mills Twp. **KENNEBEC CO.:** Augusta, Vassalboro. **OXFORD CO.:** Streaked Mountain. **PENOBSCOT CO.:** Dixmont. **WALDO CO.:** Belfast, Troy, Unity. **WASHINGTON CO.:** Addison, Cherryfield, Columbia, Columbia Falls, Harrington. **NEW HAMPSHIRE:** 

**COOS CO.:** Pinkham Notch, Pittsburg, Scott Brook Road, East Inlet Road, Clarkston. **GRAFTON CO.:** Franconia Notch. **VERMONT:** RUTLAND CO.: Pittsford. **GRANDE ISLE CO.:** Grande Isle.

**DISCUSSION OF THE EVOLUTIONARY ORIGINS OF SUBSPECIES AGAWAMENSIS**

We hypothesize that the “*agawamensis*” subspecies developed and evolved due to the result of ecological isolation within the estuarine biomes following the last glaciation of the Pleistocene approximately 12,000 to 15,000 years ago: A post-Pleistocene ‘coastal refugium’.

As has been discussed above, *Cercyonis pegala* subspecies *alope* and *maritima* occur along the New England coastal areas in proximity to, if not in actual association with, *agawamensis*. While we have observed *maritima* and/or *alope* in drier meadows near the *agawamensis* habitats, we have never observed either of the two subspecies WITHIN the typical *agawamensis* salt marsh habitat. *Alope* is described as having a yellow FW eye patch and a somewhat lighter ground color, while *maritima* is generally understood as having a deeper orange-yellow or orange-red FW eye patch, and a somewhat darker ground color. Intergrades between *agawamensis* at, and along, the “tension zone” between the salt marshes and the adjacent wet meadow habitats appear to be chiefly if not entirely with the *alope* phenotype; while *maritima* appears to occur in the drier meadows somewhat more inland. These observations could be the result, however, of the expression of the more yellow FW eye patch of *agawamensis* into the intergrades, rather than the expression of more frequent contact between *agawamensis* and *alope*. Outside of the salt marshes and in adjacent and nearby open meadows along the coast, however, the *alope* phenotype with the yellow FW eye patch does appear from our observations to be the dominant form.

Further inland and upland in the Transition Zone (i.e. at Streaked Mountain, Oxford Co., Maine), it is possible that the darker, more reddish forms of *C. pegala* (*maritima*) that occur there can be referred to as *olympus*, which may range across the Upper Midwest and through the Transition Zone of central New England.

As the glaciers retreated northward, the tundra, and then temperate grasslands followed, and the typical associated insect fauna soon followed as the Northern Hemisphere gradually warmed. At this latitude, forests rapidly took hold shortly thereafter, and therefore ‘open habitat’ species such as *C. pegala* were confined to the few vestiges of scattered prairies, margins along inland waterways, open hilltops and coastal estuaries. As the forests matured further, most of the *C. pegala* populations were pushed into more and more isolation at the immediate but relatively more expansive coastline, as other favorable habitats along riverbanks, on hilltops and other features became closed in by encroaching woodland. The bulk of the *C. pegala* population likely collapsed and retreated west of the densely wooded Appalachians - except for those still existing in the coastal marshes. These populations were both ecologically and geographically isolated and became adapted to the coastal marine environment and likewise probable host-plant shifts from inland prairie grasses to more coarse halophilic estuarine grass species. This further led to changes in phenology, flight behavior and adult food preferences and other ecological requirements. They also began to differ morphologically. These populations were hypothetically in the process of becoming a distinct species...but this process was then interrupted by another abrupt change.

The first Native American peoples arrived in the Northeast approximately 8,000 years ago as predominately nomadic hunter-gatherers. Agricultural practices and techniques adopted from tribes of the Southwest and Central America arrived soon thereafter. The dense forests were beginning to be cleared for crops, and thinned in order to attract large game animals such as deer, elk and wood bison.

Thousands of years later in the early 17th Century, came the arrival of the first Europeans. With the European settlers came the sawmill, the plow and herds of livestock. The ecological impact brought forth by the Europeans was unprecedented, and over a period of several decades, the forests in Eastern North America were cleared for
agriculture, grazing and valuable timber. The decimation of the forests allowed for prime open habitat consisting of grasses, wildflowers and other herbaceous plants. *C. pegala alope* and/or *maritima* soon re-colonized the vast new tracts of required grassland habitat. The “inland” type *C. pegala alope* and/or *maritima* then began to mingle with the coastal populations of *C. p. agawamensis*, and so began intergrading. This genetic introgression combined with coastal development and alteration of habitat has led this subspecies to now exist in the few large tracts of open salt marsh that are still present today and now protected. Introgression is however suspected to be still ongoing in areas where both grassland ecotones meet and the flight of *C. p. agawamensis* overlaps with the “inland” populations of *C. pegala*.

Evidence of this introgression has turned up in a number of vouched specimens, especially in areas where both types of habitat are in close proximity to one another. One intriguing example of this is evident in a few examples collected along the far northeast coastline in Maine (Washington Co.) in salt marsh habitat along a tidal river. At this latitude, the typical southern *C. pegala (maritima?)* featuring the orange FW eye patches, are replaced by a northern dark phenotype suggestive of *nephele* totally lacking the eye patch. A few male specimens were vouched having a residual amount of the FW patch, and wing shape similar to male specimens of *C. p. agawamensis* occurring further south of the Canadian Zone. It can be suggested, based upon the occurrence of these dark male *agawamensis*-like populations, that *agawamensis* may have historically occurred at these latitudes but may have been eliminated by periods of much colder atmospheric conditions, and/or that these northern agawamensis populations may have been almost completely genetically “washed out” by the strong influx of *C. p. nephele* at the periphery of these coastal habitats. Another hypothesis is that these northern populations do indeed represent non-patched *C. p. agawamensis* and are in fact exhibiting a type of convergent evolution or non-aposematic mimicry in coexistence with the dark *C. p. nephele*, and are therefore a “dark phase” *agawamensis*, in which the deep orange-red or reddish scales have invaded the FW yellow patch. Intensive future exploration of the Maine coastline is needed to determine the status of these mysterious northern Canadian Zone coastal populations, with observations of behavior required along with voucher specimens of both sexes.

ACKNOWLEDGMENTS

We thank Harry Pavulaan and Norbert Kondla for their help in getting the manuscript ready for publication.
Figure 1. Holotype of *Cercyonis pegala agawamensis*. Female, wingspan 54 mm.

Figure 2. Allotype of *Cercyonis pegala agawamensis*. Male, wingspan 51 mm.

Figure 3. Male topotype of *C. pegala maritima*, wingspan 49 mm.
Manuel Correllus S.F., Oak Bluffs, Dukes Co. MA, (Martha’s Vineyard Is.), July 24, 2010 M. Arey leg.

Figure 4. Female topotype of *C. pegala maritima*, wingspan 55 mm.
Manuel Correllus S.F., Oak Bluffs, Dukes Co. MA, (Martha’s Vineyard Is.), July 24, 2010 M. Arey leg.
Figure 5. Male *C. pegala nephele*, wingspan 47 mm

Figure 6. Male *C. pegala alope*, wingspan 49 mm
Witch Hollow Farm, Boxford, Essex Co. MA, July 4, 1998 M. Arey leg.

Figure 7. Female *C. pegala alope*, wingspan 53 mm
Powerline cut – Rt 97 S Georgetown, Essex Co. MA, July 23, 2002 M. Arey leg

Figure 8. A large female *C. pegala agawamensis*, wingspan 60 mm
Figure 9. Possible female intergrade between *nephele* and *agawamensis*, wingspan 54 mm

Figure 10. Male *C. pegala agawamensis*, wingspan 51 mm
Salt marsh – Rt. 286 E Seabrook, Rockingham Co. NH, August 6, 2007 M. Arey leg.

Figure 11. Female *C. pegala agawamensis*, wingspan 57 mm
Salt marsh – Rt. 286 E Seabrook, Rockingham Co. NH, August 6, 2007 M. Arey leg.
Figure 12. Habitat at type locality - Boston Rd. Newbury, Essex Co. MA; June 6, 2014

Figure 13. Habitat at type locality - Boston Rd. Newbury, Essex Co. MA; June 6, 2014
REFERENCES


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