Newsletter

of the

Alaska Entomological Society

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In this issue:

Announcing the UAF Entomology Club

by Adam Haberski

I am pleased to announce the formation of the University of Alaska Fairbanks Entomology Club. The club was conceived by students from the fall semester entomology course to bring together undergraduate and graduate students with an interest in entomology. Our goals are to provide students with opportunities for research and professional development, and to educate others through community outreach. The club meets biweekly under the guidance of faculty advisor Derek Sikes and has attracted over thirty students from diverse backgrounds.

Highlights of our activities so far include a tour of the University of Alaska Museum Insect Collection and presentations from Derek Sikes and visiting researcher Sydney Brannoch from Case Western Reserve University. This spring, we will be collaborating with the UAF Office of Sustainability to construct pollinator nest boxes to support native bee populations on campus. We will also host movie nights featuring classic "B-movie" horror films. Future plans include an entomophagy bake sale, summer collecting trips, and sending representatives to the International Congress of Entomology in Orlando Florida this September.

The Entomology Club would like to collaborate with members of the Alaska Entomological Society. We are looking for guest speakers, mentors, support for research projects, and opportunities for community outreach. If you would like to become involved, please email me at `ahaberski@gmail.com`.

Officers:
Adam Haberski, Co-President
Megan McHugh, Co-President
Taylor Davis, Secretary
Alan Roos, Treasurer

Bombus occidentalis in Alaska and the need for future study (Hymenoptera: Apidae)

by Megan McHugh[1] and Derek Sikes[1]

Pollinators are important for ecosystem health in Alaska and across the world (Cameron et al., 2011). While all members of this group are important, the role of bumblebees as native pollinators has received considerable recent attention. Bumblebees are especially good at pollination due to their ability to buzz while collecting pollen and nectar from flowers (Cameron et al., 2011). Their tendency to be long distance foragers (Heinrich, 1979) also makes them ecologically important. They pollinate a wide variety of plants, making them a valuable component of ecosystems throughout their range (Hatfield et al., 2015). Bumblebees usually are also among the first insects to pollinate plants that emerge and bloom in early spring (Heinrich, 1979). They are more common than solitary bees in cooler habitats such as closed canopy boreal forest or alpine zones (Armbruster and Guinn, 1989). Bumblebees are important to the success of many agricultural enterprises that are dependent on pollinators, primarily for greenhouse crops (Williams et al., 2012; Pampell et al., 2015). Although agriculture is not a predominant business in Alaska (Koch and Strange, 2012) bumblebees are the primary pollinators of many native berries used by Alaskans.

Half of the species of bumblebees in North America occur in Alaska (Williams et al., 2012). One of these is *Bombus occidentalis* (Greene, 1858), also known as the Western Bumblebee. The range of this species covers the western portion of the continent of North America from Arizona to Alaska, stretching above the Arctic Circle, and as far inland as Nebraska and Saskatchewan (Williams et al., 2012). Although once among the most common *Bombus* species in western North America, since the late 1990s this species has declined and is now considered rare enough to warrant the IUCN's Vulnerable Fed List category (Goulson et al., 2008; Hatfield et al., 2015). Estimates based on the current rate of decline indicate this species will become extinct in 60–70 years (Hatfield et al., 2015). Its distribution and status within Alaska is poorly known but of interest because preliminary findings indicate the species may not be declining in Alaska (Koch and Strange, 2012; Pampell et al., 2015). However, adequate sampling in Alaska of bumblebees, including *B. occidentalis*, has not been performed on a regular enough basis to be confident that the population is stable. Few studies have sampled bumblebee populations in Alaska and these have been in limited areas of the state. These studies have also examined the prevalence of parasites within the colonies of *B. occidentalis* that may be playing a part in the species decline (Koch and Strange, 2012; Pampell et al., 2015). Given the global interest in the status of this species, we thought it would be helpful to summarize what is known about this species in Alaska.

The University of Alaska Museum has 2,619 *B. occidentalis specimens* (see Figures 1 and 2), making it the 4th most prevalent Alaskan bumblebee species in the collection of 23,368 specimens; 1,971 of these *B. occidentalis* specimens were collected during a brief two-year study carried out by R. Pampell working for the USDA ARS in 2009 and 2010. The aim of their study was to document the bumblebee species of Alaska, including the distribution, species composition, seasonal biology, and parasite prevalence of bumblebees in agricultural areas and also to set a baseline for future studies of bumblebees in the state. They focused their collections in Delta, Palmer, and Fairbanks, Alaska. Within this sampling, *B. occidentalis* made up 10.4% of the 16 species collected, indicating that, within Alaska, this species' population might remain healthy (Pampell et al., 2015). This species has been known to carry a high parasite load. It is hypothesized that the parasites may be linked to the population decline and the loss of genetic variation within the species (Pampell et al., 2015). Several authors (Whittington and Winston, 2004; Thorp, 2005; Thorp and Shepherd, 2005; Hatfield et al., 2015) have proposed that the recent catastrophic decline throughout North America of *B. occidentalis* was due to *Nosema*.

A study by Koch and Strange (2012) also found that *B. occidentalis* had a large distribution across Alaska. *Bombus occidentalis* made up 28% of the bumblebees they collected, of a total of 15 species. To understand the parasite influence on *B. occidentalis*, Koch and Strange (2012) also studied the parasites found within the bees they sampled while focusing on the fungus, *Nosema bombi*, a parasite that is dependent on host cells for reproduction and cannot survive outside of the host (Koch and Strange, 2012). *Nosema bombi* is not a native parasite to North America, but made its way from Europe, most likely through the introduction of non-native species from greenhouse contamination (Koch and Strange, 2012). It is unknown if this "spillover," as it is termed, plays a role in the prevalence of *N. bombi* in Alaska because, as previously stated, Alaska does not have much agriculture. Nonetheless, they found that while many species of bumblebees suffer from the parasite, *B. occidentalis* had the highest rate of parasitism, with 44% of the specimens collected containing *N. bombi* within their study.

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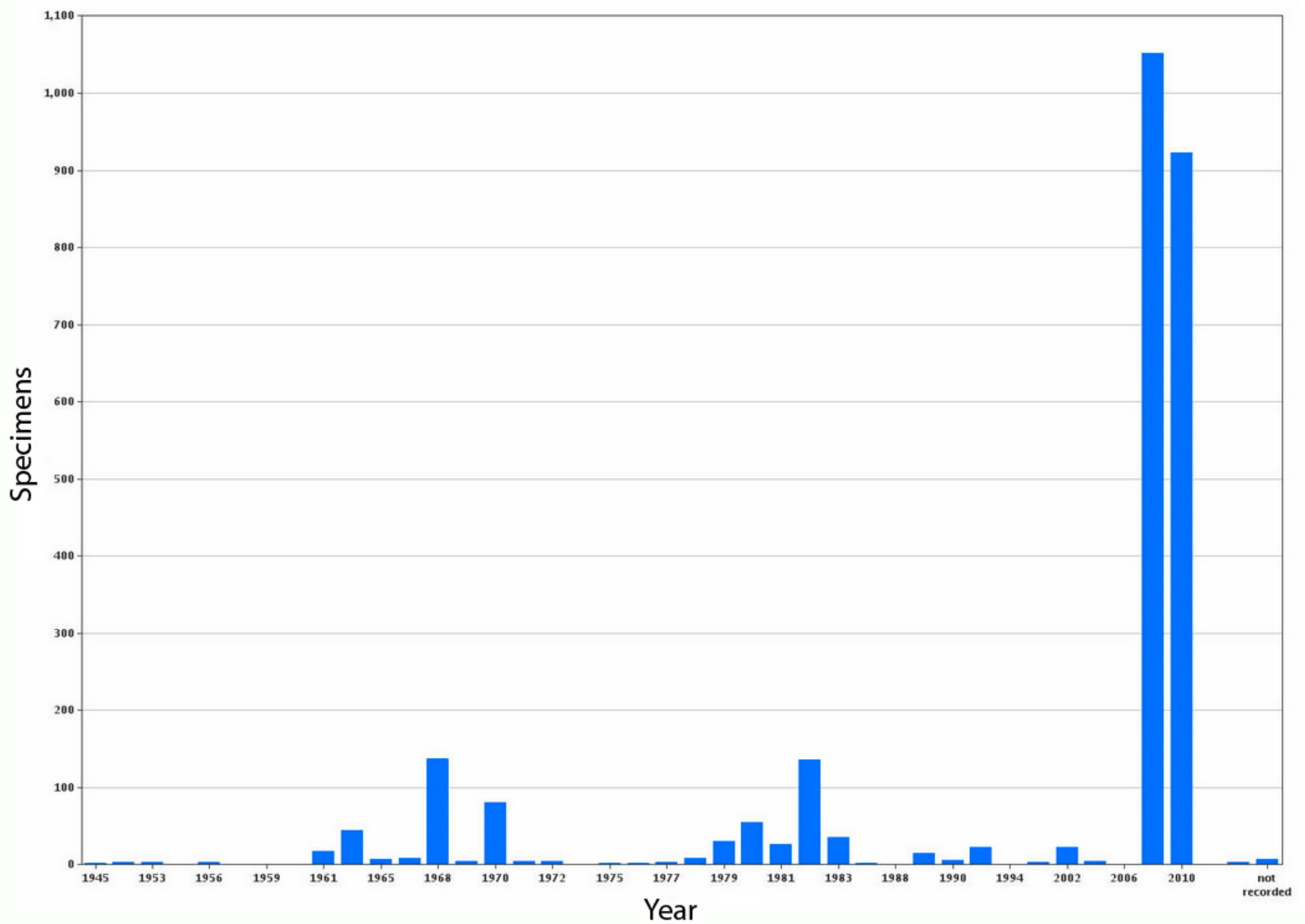

Figure 1: The number of *B. occidentalis* specimens collected over time, archived in the University of Alaska Museum. The large collections made in 2009 and 2010 were from the Pampell et al. (2015) study.

It is important to acknowledge this species' decline as a call for further investigation and monitoring in Alaska, now and in the future. The species is apparently at risk of population collapse due to various factors including parasite spillover, habitat loss, insecticide use, climate change, habitat fragmentation, and introduction of invasive species (Goulson et al., 2008; Williams et al., 2014; Kerr et al., 2015). Accurate representation of the population numbers of *B. occidentalis* in Alaska will allow for an in-depth understanding of this species' prognosis of survival. While yearly sampling on a large scale could negatively affect the species in Alaska, small scale sampling every few years shouldnt, and would help us understand population trends of *B. occidentalis* in Alaska.

Habitat *Bombus occidentalis* prefers meadows, tundra, alpine fields, gardens and other open areas where flowering shrubs grow (Williams et al., 2014). This species pollinates a wide variety of plants but is mostly limited to flowers with short petals due to the short length of its tongue (Williams et al., 2014). Which plant species *B. occidentalis* pollinates most in Alaska is unknown.

Genetics and taxonomy Williams et al. (2012) concluded the species status of *B. occidentalis* is well supported by a number of molecular and morphological diagnostic traits. DNA barcoding supports two subspecies: the northern, long-haired subspecies known as *B. occidentalis mckayi* Ashmead, 1902, which is present in Alaska and the Yukon territory, and a southern short-haired subspecies known as *B. occidentalis occidentalis* Green, 1858, which is present in most states and provinces within the range south of Alaska and the Yukon Territory (Williams et al., 2012, 2014). To date, all known population declines have been restricted to the southern subspecies (Hatfield et al., 2015). However, the southern subspecies has also received much more survey effort than the northern.

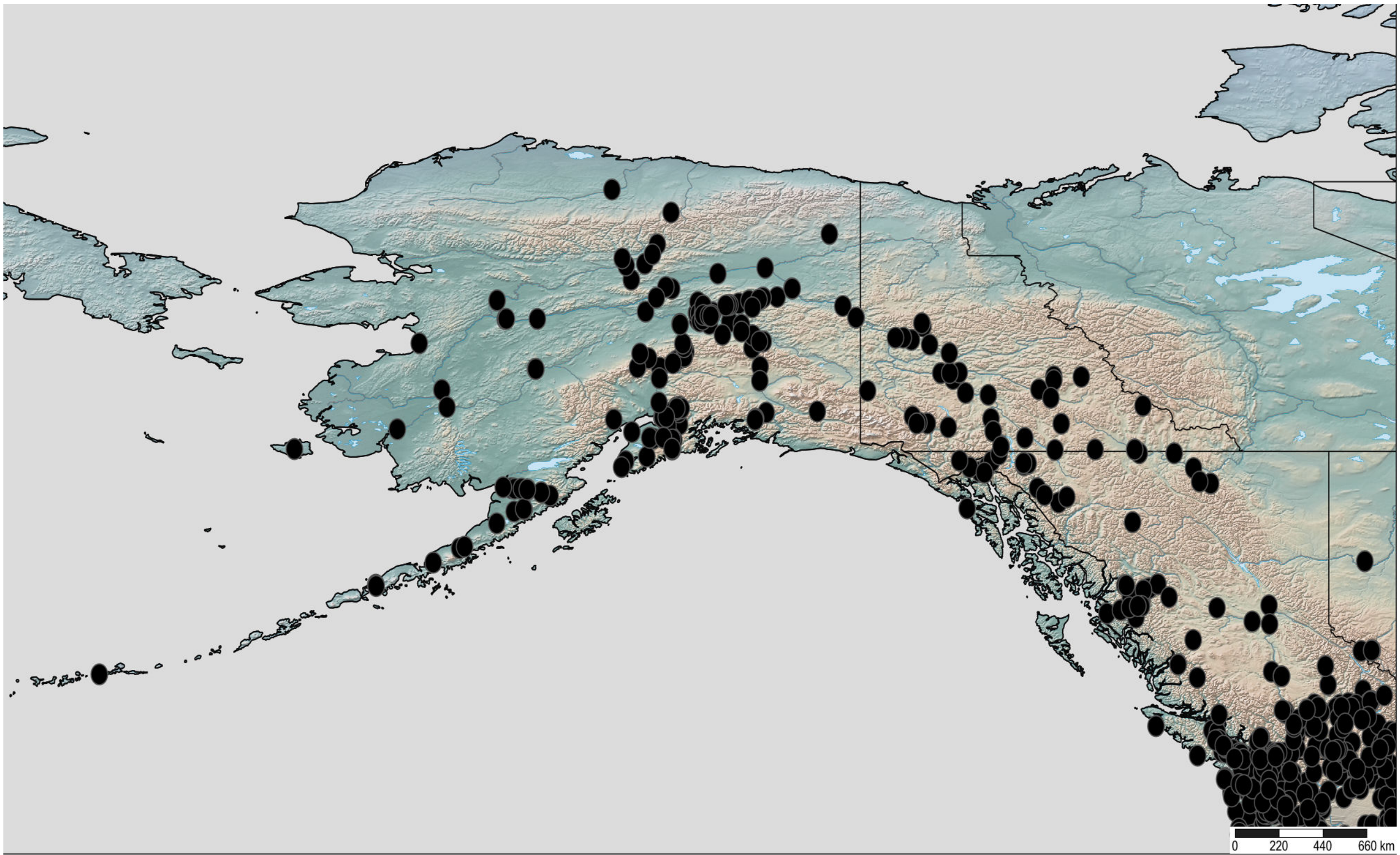

Figure 2: Occurrence data for *B. occidentalis* from GBIF (`http://www.gbif.org`) for Alaska and western Canada, 18.March.2016 (doi:10.15468/dl.tq9ujy). The data mapped originated from the following institutions: Biodiversity Institute of Ontario, USGS PWRC - Native Bee Inventory and Monitoring Lab - US-CA-MX, Canadian National Collection (CNC), Kenai National Wildlife Refuge (KNWR), Snow Entomological Museum Collection (SEMC) at the University of Kansas, the University of Alaska Museum at the Unviersity of Alaska Fairbanks (UAM), the USDA-ARS, and the Yale Peadbody Museum (YPM).

The University of Alaska Museum has DNA barcoded two Alaskan specimens from their collection (UAM:Ento:187982, UAM:Ento:188350) and the Kenai National Wildlife Refuge collection has DNA barcoded one Alaskan specimen (KNWR:Ento:2800). Unsurprisngly, within the Barcode of Life Database (`http://bins.boldsystems.org`) these all fall into a BIN (Ratnasingham and Hebert, 2013) corresponding to the northern subspecies *B. occidentalis mckayi* (BOLD:ACE8361). It is not clear exactly where in British Columbia the break between the two subspecies occurs (although it is known that the northern subspecies occurs in the northern portion of British Columbia and the southern in the southern portion); how much, if any, gene flow occurs in this region; or if the southern subspecies is expanding its range northward, as would be expected due to climate change.

Historical and current range and distribution patterns
Bombus occidentalis has been collected from Atka in the Aleutian Islands to north of the Brooks Range in Alaska (Figure 2). While most collection has been done along main roadways in Alaska, it is assumed that the species has a broad distribution across the entire state primarily south of the Brooks Range. The niche-modeled range map in Williams et al. (2014) does not correspond well to the known sites of collection in Alaska. We suspect the niche model used was influenced by the large amount of data for the southern subspecies. Future research should establish a niche model focused only on the northern subspecies. This could be used to help predict future range changes under various climate change scenarios.

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New findings of twisted-wing parasites (Strepsiptera) in Alaska

by Molly McDermott[1]

Figure 1: Adult male *Elenchus* sp. collected on Alaska's Seward Peninsula (UAM:Ento:293071). Note front halteres, raspberry-shaped eyes, and forked antennae. Photo by DS Sikes.

Introduction

Strepsipterans are a group of insects with a gruesome life history and an enigmatic evolutionary past. Called 'twisted-wing parasites', they are minute parasitoids with a very distinct morphology (Figure 1). Alternatively thought to be related to ichneumon wasps, Diptera (flies), Coleoptera (beetles), and even Neuroptera (net-winged insects) (Pohl and Beutel, 2013); the latest genetic and morphological data support the sister order relationship of Strepsiptera and Coleoptera (Niehuis et al., 2012). Strepsipterans are highly modified, males having two hind wings and halteres instead of front wings or elytra. Unlike most parasitoids, they develop inside active, living insects who are sexually sterilized but not killed until or after emergence (Kathirithamby et al., 2015).

Strepsipteran life history

Females (in most species) do not undergo a typical pupal phase—after four larval phases, they attain their adult form, lacking eyes, wings, legs, and antennae (Whitfield and Purcell, 2012). They remain embedded in their host throughout their adult life. To attract males, they secrete a pheromone which is detected by elaborate chemical receptors on the male antennae. The sole purpose of a male's 3–6 hour life is to find a female (Kathirithamby et al., 2015). A male mates by rupturing the entrance to the brood canal, located just behind the female's head. This unusual placement allows mating while the female remains within her host (Kathirithamby et al., 2015). A single female can produce 75,000 eggs and 9–10,000 larvae, who move freely in the mother's circulatory system, an arrangement unique to these animals (Kathirithamby, 2002; Whitfield and Purcell, 2012).

Figure 2: Adult female delphacid planthopper collected on Alaska's Seward Peninsula, showing no evidence of parasitism. Genitalia are normal and there are no darkened spots on the abdomen. Photo by M McDermott.

In a 30–40 day process that kills the mother, larvae emerge from the brood canal to seek new hosts. Only these 1st instar larvae and emergent adult males are mobile (Whitfield and Purcell, 2012). Most larvae enter their hosts through the abdomen by secreting an enzyme that softens the host exoskeleton, although there is a recorded case of larvae entering through the host's tarsus (Kathirithamby, 2002; Whitfield and Purcell, 2012). Once inside their host, larvae induce the host to produce a cuticle to protect it from the host immune system, an adaptation that has allowed strepsipterans to parasitize a wide range of hosts (Kathirithamby et al., 2003; Whitfield and Purcell, 2012). The 595 described species of strepsipterans parasitize 34

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insect families, mainly Hemiptera (true bugs) and Hymenoptera (ants, bees and wasps) (Kathirithamby, 2002; Kathirithamby et al., 2015; Whitfield and Purcell, 2012). In a few cases, males and females may parasitize different species, even different orders of insects (Whitfield and Purcell, 2012). The paucity of research on this group makes it likely that there are many undiscovered species (Kathirithamby et al., 2015).

Figure 3: Adult delphacid with two developing strepsipteran males. One parasite (bottom) is close to emergence; the other (top left) is in the pupal phase. Note how the host is prevented from developing any genitalia. Photo by M McDermott.

New Records in Alaska

In the summer of 2013, I worked as a field technician as part of a study conducted by US Geological Survey assessing the distribution and abundance of shorebirds and songbirds in the boreal forest-tundra transition zone of western Alaska (McNew et al., 2013). As an indicator of habitat quality for birds, we collected insects via sweep net from June 28th to July 10th in a variety of subarctic habitats. Upon examination and identification of these insects, Delphacidae (planthoppers, see Figure 2) adults and nymphs were often found to show evidence of strepsipteran parasitism. Evidence ranged from a darkened larval entry point on the abdomen to an adult male emerging head-first (Figure 3). At some sites, delphacids were found to have rates of parasitism as high as 29%. Some hosts had two parasites, engulfing more than half their abdomen (Figure 3). One study site in the interior of the Seward Peninsula had much higher parasitism rates than other sites, although the rate varied greatly along transects (Figure 4).

To determine what ecological variables are related to host prevalence and rates of parasitism, I examined vegetation and landscape characteristics collected at each of the 100 sample points. Delphacid planthoppers were found to be associated with herbaceous plant cover height and abundance, with a slightly higher prevalence on north and northeast facing slopes. Within the Neva Creek study area (Figure 4), higher-elevation sites with gentler slopes and taller grasses and sedges had higher rates of parasitism, even when controlling for planthopper abundance. Interestingly, elevation was a much stronger predictor of parasitism rate than host abundance or herbaceous plant cover.

These results suggest that parasitism rate is not due to host abundance, but varies across the landscape. Considering the extremely short lifespan of the winged male and the miniscule size of mobile larvae, it seems likely that strepsipteran population abundance is restricted by how far they are able to disperse while within their host. It could be that higher elevation sites with gentler slopes are windier, which allows their hosts to travel farther. The high rates of parasitism associated with tall grasses after controlling for host abundance could be explained by a behavioral difference between infected and non-infected hosts, causing infected planthoppers to prefer taller grasses. Alternatively, it is possible that taller grasses provide better conditions for strepsipteran adults to mate or larvae to infect new hosts.

Prior to these collections[2], there were only four records of this order in Alaska, and none from the Seward Peninsula: two in Fairbanks (UAM:Ento:201815, UAM:Ento:217034), one in Delta Junction (UAM:Ento:201812), and one in Sterling (UAM:Ento:100552).

In early 2015, an adult male specimen (Fig. 1) was sent to J. Kathirithamby who identified it as a member of the genus *Elenchus* and possibly a new species. Two adult males were prepared for DNA barcoding this spring, and will be sent out by D. Sikes for sequencing. Although this is an understudied group of insects, there are DNA barcodes published for four described species of *Elenchus* as well as for several unidentified specimens of *Elenchus* (GenBank search, 3/10/16). Genetic results from the Seward Peninsula collections will help to determine if these specimens are one of these four species of *Elenchus* or not, although it will be difficult to determine if this specimen represents a new species or a previously described species that has no genetic data available. Regardless, these collections provide more information about the geographic range and ecology of these fascinating creatures.

[2]UAM:Ento:293071, UAM:Ento:296383, UAM:Ento:296386, UAM:Ento:296387, UAM:Ento:296389, UAM:Ento:296392, UAM:Ento:296393, and UAM:Ento:296396

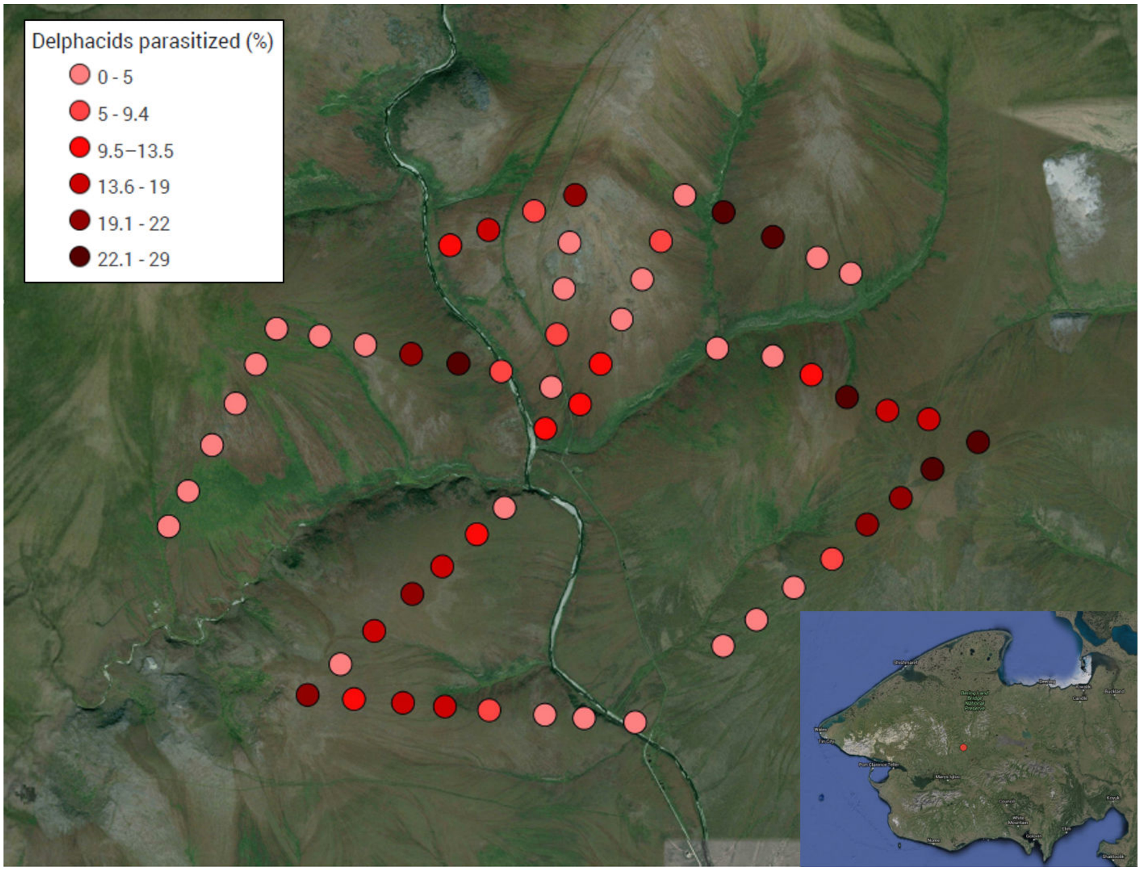

Figure 4: Rates of strepsipteran parasitism around Neva Creek in the interior of Alaska's Seward Peninsula. Each circle represents a plot 500 m in diameter. Within each plot, insects were collected with three randomly placed 50 m transects. Data were also collected on bird abundance, vegetation and landscape characteristics.

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Asian gypsy moths and Alaska

by Jacquelyn Schade[1]

Figure 1: Asian gypsy moth, adult female in Mongolia by John H. Ghent, USDA Forest Service, Bugwood.org, `http://www.forestryimages.org/browse/detail.cfm?imgnum=1241013`

Each year, the Alaska Division of Agriculture conducts Cooperative Agricultural Pest Surveys (CAPS) which are funded through cooperative agreements with the United States Department of Agriculture (USDA), Animal and Plant Health Inspection Service (APHIS).

The CAPS is a National program of APHIS and is active in all 50 states. The purpose of the program is to help safeguard agricultural and environmental resources by early detection of harmful pests and diseases before they are established. The Alaska CAPS Program facilitates the detection of plant pests through surveys, outreach, educational activities and interagency partnering. Alaska has been involved with the CAPS program since the early 1980's.

Since the early 1990's, Alaska has been conducting the Asian Defoliating Moth (AGM) Survey through the CAPS program. This survey primarily searches for the Asian Gypsy moth, but also searches for Rosy Gypsy moth, Siberian Silk moth, and the Nun moth.

Asian defoliating moths are considered among the most important commercial pests in their native range. Their native range extends roughly from Europe to parts of Asia, to include the far east of Russia and other East Asian countries. Outbreaks occur periodically in their native ranges resulting in extensive areas of defoliation. Asian gypsy moths are highly invasive pests to areas where they are not native.

Alaska is considered high risk of AGM introduction due to the high volume of marine traffic we receive from Asia and Russia. During the 2014 shipping season, Custom Border Protection (CBP) and APHIS-Plant Protection and Quarantine (PPQ) Officers intercepted thousands of viable egg masses on vessels arriving from China, Russia, Japan and Korea destined to multiple US Ports. In Alaska, Asian gypsy moth interceptions occurred on vessels in both 2012 and in 2008 near the port of Ketchikan. In 2015, Western Washington intercepted eight Asian gypsy moths and Oregon intercepted two Asian gypsy moths in traps. These interceptions were the first catches in traps since 1999 and demonstrate Alaska's susceptibility to Asian gypsy moth establishment.

Figure 2: Asian gypsy moth, adult male in Mongolia by John H. Ghent, USDA Forest Service, Bugwood.org, `http://www.forestryimages.org/browse/detail.cfm?imgnum=1241014`

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Figure 3: Asian gypsy moth, larva in Mongolia by John H. Ghent, USDA Forest Service, Bugwood.org, http://www.forestryimages.org/browse/detail.cfm?imgnum=1335025

Alaska's forest products, wildlife, and tourist industries represent a significant portion of the state's resources and would be at risk should Asian gypsy moths become established. These resources depend on our healthy forests. Alaska has approximately 129 million forested acres throughout the state that contain host species of these exotic pests, to include species in the genera *Alnus* (alder), *Populus* (aspen), *Betula* (birch), *Salix* (willow), *Larix* (larch), *Sorbus* (mountain ash), *Pinus* (pine), *Picea* (spruce), *Abies* (true fir), and *Tsuga* (hemlock).

Adult male Asian gypsy moths have a wingspan of 1.5 inches, and their wings are greyish-brown. Adult female Asian gypsy moths have a wingspan of 3.5 inches or more, and their wings are white. The females also have distinctive black markings on their wings.

Asian gypsy moths have one generation per year. The female Asian gypsy moth has the ability to fly approximately 25 miles per year. This rapidly increases the Asian gypsy moth's range each year. Female Asian gypsy moths lay 100–1,000 eggs in one egg mass. They are attracted to light and will often deposit their egg masses near light sources on walls, trees, and light poles. They will also lay their eggs in cracks or crevices on rock outcroppings, or slabs of steel that could be bound for export. The Asian gypsy moths lay their eggs between July and September.

Eggs hatch anywhere from late April to early May; where the larvae disperse to host plants by ballooning. Ballooning is the process where the larvae drop on a silk thread from a branch and use air and wind currents to "balloon" to another location. The male Asian gypsy moths generally have 5 instars, while the females have 6. The larvae will feed at night and rest in protected areas on their host during the day. The larval stage is the stage that causes significant damage to host trees and can devastate large forested areas in one generation.

Pupation occurs on the host's foliage or litter and lasts for 10–14 days. When the adults emerge, they do not feed. The male Asian gypsy moths' sole purpose is to mate and lay eggs. Adults only live for 1 to 3 weeks and are active from July to August.

If you suspect that you have Asian gypsy moths in your trap catches or area, contact Jacquelyn Schade at Jacquelyn.Schade@alaska.gov or 907-761-3858.

Figure 4: Asian gypsy moth, eggs in Mongolia by John H. Ghent, USDA Forest Service, Bugwood.org, http://www.forestryimages.org/browse/detail.cfm?imgnum=1335003

Non-marine invertebrates of the St. Matthew Islands, Bering Sea, Alaska

by Derek S. Sikes[1], Dan Bogan[2], Casey Bickford[3], Jozef Slowik[1], and Steve Peek[1]

Abstract

Macroinvertebrates were sampled on St. Matthew and Hall Islands between 31 July–6 Aug 2012. Freshwater invertebrates were processed at the Alaska Center for Conservation Science Aquatic Lab and approximately 4,986 terrestrial invertebrates were processed and curated into the University of Alaska Museum Insect Collection. Although focused on non-marine arthropods, all non-marine invertebrates were also targeted. A list of identifications is provided although some identifications remain coarse. Additionally, all known prior records based on publications or specimens with online data are summarized. Thirty-four specimens of 21 species from this expedition were DNA barcoded (`http://arctos.database.museum/saved/St-Matthew-DNA-barcoded`).

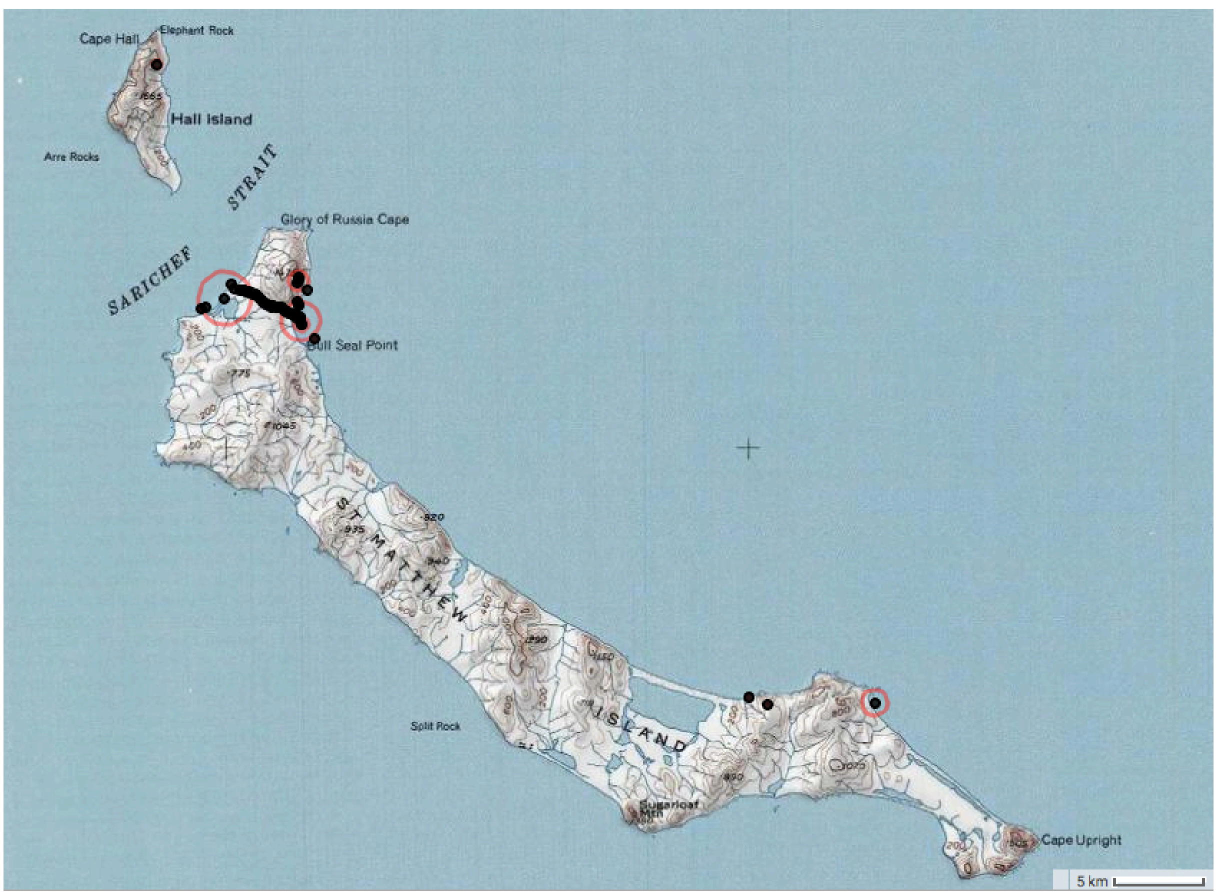

Figure 1: Map of St. Matthew and Hall Islands showing collection sites of terrestrial invertebrates.

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Introduction

St. Matthew, Pinnacle, and Hall Islands comprise the St. Matthew Islands in the Bering Sea, Alaska. These unihabited and remote islands are home to around a million colonial- and ground-nesting birds and were once high elevation points, of volcanic origin, on the Bering Land Bridge which connected Asia to North America (Klein and Kleinleder, 2015; Guthrie, 2004). The land bridge flooded due to rising sea levels and isolated these points as islands between 9,000 and 11,000 years ago (Guthrie, 2004). The largest island, St. Matthew, is about 52 km long (Figure 1) with high points reaching 459 m.

Little prior documentation of the invertebrate fauna of these islands exists. Although the Harriman Expedition landed on St. Matthew and Hall Islands for several hours on July 14 and 15, 1899, their reporting on these islands was restricted to geology. Apparently no invertebrates were collected. Rausch and Rausch (1968), who focused on studying the vole endemic to the islands, listed 18 invertebrate species names and mentioned that more than 70 insect species had been identified from material they collected. However, the identities of these 70+ species have never been published.

The St. Matthew islands have two known vertebrate endemic species, McKay's bunting (*Plectrophenax hyperboreus* Ridgway, 1884) and a singing vole (*Microtus abbreviatus* Miller, 1899), yet, to date, no known invertebrate species endemic to these islands have been described. This is not surprising given the paucity of prior research on the invertebrate fauna of these islands. With the support of the United States Fish and Wildlife Service the first and third authors were invited on an expedition to these islands in 2012 as part of a multidisciplinary research team (Romano et al., 2013). We take this opportunity to present our findings to date. We hope that by making this list of invertebrates available in its current, still incomplete form, we will attract the attention of specialists who would like to borrow and study the as-yet unidentified material.

Methods

Sites

Collection effort was concentrated on the north end of the island between Bull Seal Point on the NE coast and a small lake on the NW coast. The island is approximately three miles wide between these sites (e.g. 60.56504°N, 172.95976°W ± 2 km). Additional collection areas included two on the south end of the island (60.38036°N, 172.50139°W; 60.37816°N, 172.38133°W) and one on Hall Island (60.67879°N, 173.06880°W). Sampling was concentrated in two major areas of the island: 1) the area around North Lake, on the northern end of the island, including ponds and streams near the field camp; and 2) the area around Big Lake, on the southern end of the island (Figure 1).

Habitats

(1) Littoral-beach drift and vegetation, rock outcrops (0–15 m elevation) (2) interior sedge-marsh lowland (10–60 m elevation) with willow hummocks, (3) interior upland (60–150 m elevation) meadows and streams. The interior sedge-marsh lowland vascular vegetation was primarily composed of *Calamagrostis*, *Carex*, *Empetrum*, *Equisetum*, *Eriophorum*, *Luzula*, *Poa*, *Polemonium*, *Potentilla*, *Rhodiola*, *Rubus*, *Rumex*, *Salix*, and *Saxifraga*. Lakes, ponds, (some brackish), and streams were sampled for fresh water invertebrates.

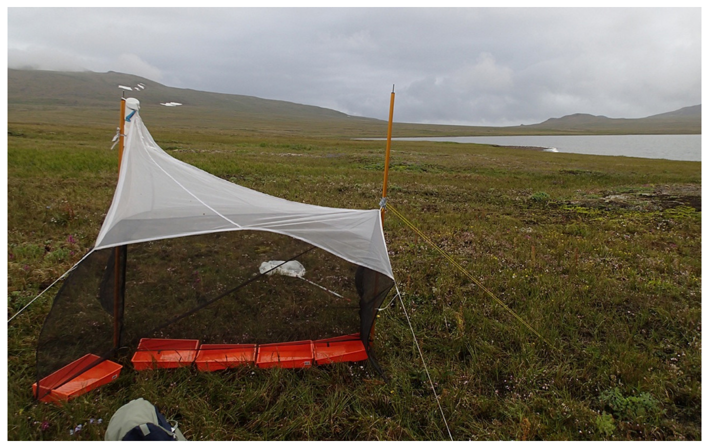

Figure 2: Malaise-Flight Intercept Trap hybrid near the NW coast of St. Matthew.

Collection Methods

Terrestrial collection methods included sweep net, aspirator, forceps, pitfall traps, flight intercept trap (FIT)-Malaise trap hybrids (Figure 2), colored (pollinator) traps, and Winkler extractors. A transect of 33 pitfall traps with two pollinator traps at each was set, spacing traps 100 m apart through the interior sedge-marsh lowland from the NE coast to the NW coast. A Malaise / FIT trap was set at either end of the transect with a third Malaise / FIT trap set in the middle. A grid of 15 large yellow pollinator bowls was set around the NW coast Malaise / FIT trap.

Freshwater aquatic invertebrates were collected primarily by Anthony DeGange and Steve Delehanty with a 500 μm mesh d-frame dip net at four lentic and five lotic sites. Samples at the lentic sites were collected by sweeping the net through targeted littoral habitats. Lotic samples were collected by disturbing approximately two square feet of benthic substrate for approximately two minutes and allowing dislodged organisms to collect in net downstream.

Five locations representing a range of available habitat were sampled at each site. Samples at each of the nine sites were composited and stored in approximately 70% ethanol.

Lab Methods

Terrestrial invertebrate samples were processed in the preparation lab at the University of Alaska Museum. All samples were sorted and had their specimens either mounted on pins or stored in 70% ethanol vials. All pinned specimens and vials were databased. Most specimens have been identified to order or lower, although many taxa have yet to be identified.

Aquatic macroinvertebrate samples were subsampled in the lab to obtain a fixed count of 300 $\pm$20% organisms. Chironomids were grouped by morphotaxa and a subset of each morphotaxa was slide-mounted for microscopic examination. Representatives of each encountered (non-chironomid) taxa were placed in vials containing 70% ethanol and are maintained as a reference collection for this project.

Results and Discussion

Our work, combined with previous efforts, documents seventy-eight species of invertebrates identified to the species level within a total of 258 unique taxon-identifications. Table 1 shows the number of specimens of each identification from this expedition identified, and by whom, to date, including identifications from the collection efforts of Rausch and Rausch (1968).

The terrestrial portions of littoral habitats were the most thoroughly sampled, the interior lowlands were the second most thoroughly sampled, and the interior upland meadows were the least well sampled. Figure 1 shows the sites sampled for terrestrial invertebrates.

A number of littoral, beach- and driftwood-associated taxa that are relatively common on the Pribilofs and the Aleutians were, surprisingly, not found on St. Matthew. These include cybaeid spiders, bristletails, pseudoscorpions, centipedes, and the beetle taxa *Nebria metallica*, *Aegialites*, and *Hypolithus littoralis*. Bumblebees, *Bombus polaris*, were active on the warmest day.

There are two common species of weevil in Alaska in the genus *Lepidophorus*: *Lepidophorus lineaticollis* Kirby, 1837, which is found primarily in interior Alaska; and *Lepidophorus inquinatus* (Mannerheim, 1852), which is found primarly along the coast and the Aleutian chain, incuding the Pribilofs. Both species are well represented in UAM by hundreds of specimens. Despite being an island that receives ample driftwood, the presumed mode of transport for *L. inquinatus*, no *L. inquinatus* were found on St. Matthew. Instead we found a population that keyed to *L. lineaticollis*. We successfully DNA barcoded one of these specimens `http://arctos.database.museum/guid/UAM:Ento:242826` (DNA barcode: `http://bins.boldsystems.org/index.php/Public_RecordView?processid=UAMIC1851-14`) which was found to be 3.76% divergent in its COI sequence from other samples of *L. lineaticollis* and fell into a separate BIN (analogous to species, Ratnasingham and Hebert, 2013). Unfortunately, BOLD has only two partial (based on UAM specimens), but no complete, records of *L. inquinatus*, so the genetic similarity of the St. Matthew population to *L. inquinatus* cannot yet be determined.

The beetle fauna includes some Holarctic species such as *Simplocaria metallica* (Sturm, 1807), *Amara alpina* Paykull, 1790, *Nebria nivalis* Paykull, 1798, and *Pterostichus brevicornis* (Kirby, 1837), while some are restricted to northwestern North America such as *Carabus truncaticollis* Eschscholtz, 1833, or span the boreal-tundra zone of North America such as *Notiophilus borealis* Harris, 1869, and *Pterostichus agonus* Horn, 1880. *Diacheila polita* (Faldermann, 1835) is almost circumpolar but in North America is restricted to the northwest. *Pterostichus empetricola* (Dejean, 1828) and *P. nivalis* (Sahlberg, 1844) are found in the northwest of North America, including the Aleutian chain, and the northeast of Asia (Siberia, Commander Isl.). Distributional information for the Carabidae was obtained from Lindroth (1961, 1963, 1966, 1968, 1969*a*,*b*).

Interestingly, the spider fauna was represented by only the family Linyphiidae. The usually ubiquitous cybaeid (Cybaeidae) and wolf spider (Lycosidae) ground fauna found elsewhere along Alaska's mainland, coasts, and islands were missing. The largest arachnid on these islands was the opilionid *Liopilio yukon* Cokendolpher, 1981. There were several species in which identifications were not possible—one large linyphiine close to the Asian genus *Mughiphantes*, and one erigonine species, (Erigoninae St. Matt 1) of uncertain generic placement but somewhere near *Lophomma*.

The St. Matthew spider fauna is dominated by Beringian and Holarctic species with several of the species, *Collinsia holmgreni* (Thorell, 1871), *Hilaira vexatrix* (O. Pickard-Cambridge, 1877), *Oreonetides vaginatus* (Thorell, 1872), and *Sisicottus nesides* (Chamberlin,1921) being common within the Aleutian fauna.

Current data from UAM specimens, prior samples, and literature are available through the University of Alaska Museum online database Arctos at: `http://arctos.database.museum/saved/St.Matthew-arthropods` that URL will also find all databased samples from the 2012 expedition. Most Arctos records at that link include digital photographs of the collection sites. Additional photos of this expedition can be found at: `http://www.flickr.com/photos/alaskaent/sets/72157631509207664/`.

Table 1: Identifications of St. Matthew and Hall Island inveretrbates from UAM specimens and literature records. All taxa with individual counts of zero are specimens currently in the Alaska Center for Conservation Science Aquatic Lab that have yet to be accessioned into UAM and counted. Records obtained from Rausch and Rausch (1968) are marked R&R. Records identified by C. H. Lindroth are from Lindroth (1961, 1963, 1966, 1968, 1969*a*,*b*), and based on specimens most of which are in the Canadian National Collection and that were presumably collected by Rausch and Rausch.

Order	Family	Scientific Name	Identified By	Count
Enchytraeida	Enchytraeidae	Enchytraeidae	Tarmo Timm	1
		Oligochaeta	Dan Bogan	0
Canalipalpata	Sabellidae	*Manayunkia* sp.	Dan Bogan	0
		Annelida	J. Williamson	90
Acari	Lebertiidae	*Lebertia* sp.	Dan Bogan	0
Acari	Pionidae	*Piona* sp.	Dan Bogan	0
Acari	Sperchonidae	*Sperchon* sp.	Dan Bogan	0
Acari	Unionicolidae	*Huitfeldtia* sp.	Dan Bogan	0
Acari		Acari	Dan Bogan	0
Acari		Acari	D. S. Sikes	2
Acari		Acari	J. Williamson	533
Acari		Brachypylina	Dan Bogan	0
Acari		Macropylina	Dan Bogan	0
Acari		Oribatida	Dan Bogan	0
Araneae	Linyphiidae	*Agyneta jacksoni*	J. A. Slowik	1
Araneae	Linyphiidae	*Bathyphantes eumenis*	J. A. Slowik	1
Araneae	Linyphiidae	*Bathyphantes pallidus*	H. Levi	R&R
Araneae	Linyphiidae	*Collinsia holmgreni*	J. A. Slowik	41
Araneae	Linyphiidae	*Erigone arctica*	H. Levi	R&R
Araneae	Linyphiidae	*Erigone arctica*[4]	A. Holm	2
Araneae	Linyphiidae	*Erigone arctica*	J. A. Slowik	34
Araneae	Linyphiidae	*Erigone arctophylacis*	J. A. Slowik	62
Araneae	Linyphiidae	*Erigone cristatopalpus*	J. A. Slowik	3
Araneae	Linyphiidae	*Erigone psychrophila*	J. A. Slowik	2
Araneae	Linyphiidae	*Erigone tirolensis*	J. A. Slowik	2
Araneae	Linyphiidae	Erigoninae St. Matt 1	J. A. Slowik	6
Araneae	Linyphiidae	*Hilaira vexatrix*	J. A. Slowik	29
Araneae	Linyphiidae	*Improphantes complicatus*	J. A. Slowik	13
Araneae	Linyphiidae	*Islandiana cristata*	J. A. Slowik	1
Araneae	Linyphiidae	*Islandiana falsifica*	J. A. Slowik	9
Araneae	Linyphiidae	Linyphiidae	J. A. Slowik	140
Araneae	Linyphiidae	*Mughiphantes whymperi* ?	J. A. Slowik	2
Araneae	Linyphiidae	*Oreoneta arctica*	J. A. Slowik	16
Araneae	Linyphiidae	*Oreonetides vaginatus*	J. A. Slowik	49
Araneae	Linyphiidae	*Porrhomma convexum*	J. A. Slowik	1
Araneae	Linyphiidae	*Sisicottus nesides*	J. A. Slowik	26
Araneae		Araneae	J. A. Slowik	2
Mesostigmata	Laelapidae	*Haemogamasus ambulans*	G. P. Holland	R&R
Mesostigmata	Laelapidae	*Laelaps kochi*	G. P. Holland	R&R
Opiliones	Phalangiidae	*Liopilio yukon*	Matt Bowser	10
Diplostraca	Chydoridae	*Eurycercus lamellatus*	M. S. Wilson	R&R
Harpacticoida	Canthocamptidae	*Paracamptus reggiae*	M. S. Wilson	R&R
		Ostracoda	Dan Bogan	0
Coleoptera	Byrrhidae	*Simplocaria metallica*	P .J. Johnson	2

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[4]http://mczbase.mcz.harvard.edu/guid/MCZ:IZ:84407

Order	Family	Scientific Name	Identified By	Count
Coleoptera	Carabidae	*Amara alpina*	C. H. Lindroth	1
Coleoptera	Carabidae	*Amara alpina*	D. S. Sikes	17
Coleoptera	Carabidae	*Amara alpina*	F. Hieke	1
Coleoptera	Carabidae	*Bembidion arcticum*	D. R. Maddison	13
Coleoptera	Carabidae	*Carabus truncaticollis*	D. S. Sikes	1
Coleoptera	Carabidae	*Carabus truncaticollis*	G. Ball	1
Coleoptera	Carabidae	*Diacheila polita*	C. H. Lindroth	1
Coleoptera	Carabidae	*Diacheila polita*	D. S. Sikes	23
Coleoptera	Carabidae	*Nebria nivalis*	C. H. Lindroth	1
Coleoptera	Carabidae	*Nebria nivalis*	D. S. Sikes	203
Coleoptera	Carabidae	*Notiophilus borealis*	D. S. Sikes	8
Coleoptera	Carabidae	*Pterostichus* (*Cryobius*) sp.	D. S. Sikes	2
Coleoptera	Carabidae	*Pterostichus agonus*	D. S. Sikes	4
Coleoptera	Carabidae	*Pterostichus brevicornis*	C. H. Lindroth	1
Coleoptera	Carabidae	*Pterostichus brevicornis*	G. Ball	1
Coleoptera	Carabidae	*Pterostichus brevicornis* ?	D. S. Sikes	25
Coleoptera	Carabidae	*Pterostichus empetricola*	D. S. Sikes	55
Coleoptera	Carabidae	*Pterostichus empetricola* ?	D. S. Sikes	14
Coleoptera	Carabidae	*Pterostichus nivalis*	G. Ball	1
Coleoptera	Carabidae	*Pterostichus nivalis* ?	D. S. Sikes	6
Coleoptera	Carabidae	*Pterostichus parasimilis*	C. H. Lindroth	1
Coleoptera	Carabidae	*Pterostichus parasimilis*	G. Ball	1
Coleoptera	Carabidae	*Pterostichus pinguedineus*	C. H. Lindroth	1
Coleoptera	Carabidae	*Pterostichus pinguedineus*	G. Ball	1
Coleoptera	Carabidae	*Pterostichus similis*	C. H. Lindroth	1
Coleoptera	Carabidae	*Pterostichus ventricosus*	C. H. Lindroth	1
Coleoptera	Carabidae	*Pterostichus ventricosus*	D. S. Sikes	18
Coleoptera	Carabidae	*Pterostichus ventricosus*	G. Ball	1
Coleoptera	Curculionidae	*Lepidophorus lineaticollis*	D. S. Sikes	4
Coleoptera	Curculionidae	*Sthereus ptinoides*	D. S. Sikes	25
Coleoptera	Dytiscidae	*Agabus* sp.	Dan Bogan	0
Coleoptera	Dytiscidae	*Liodessus* sp.	Dan Bogan	0
Coleoptera	Dytiscidae	*Sanfilippodytes* sp.	Dan Bogan	0
Coleoptera	Hydrophilidae	*Helophorus* sp.	Dan Bogan	0
Coleoptera	Scarabaeidae	*Agoliinus* sp.	D. S. Sikes	1
Coleoptera	Staphylinidae	Aleocharinae	D. S. Sikes	1
Coleoptera	Staphylinidae	*Atheta* (*Dimetrota*) sp.	J. Klimaszewski	4
Coleoptera	Staphylinidae	*Atheta terranovae* ?	J. Klimaszewski	1
Coleoptera	Staphylinidae	*Boreostiba frigida*	J. Klimaszewski	1
Coleoptera	Staphylinidae	*Eucnecosum brachypterum*	M. Thayer	76
Coleoptera	Staphylinidae	*Eucnecosum brunnescens*	M. Thayer	13
Coleoptera	Staphylinidae	*Lathrobium* sp.	D. S. Sikes	1
Coleoptera	Staphylinidae	*Micralymma brevilingue*[5]	M. Thayer	5
Coleoptera	Staphylinidae	*Olophrum consimile*	M. Thayer	5
Coleoptera	Staphylinidae	*Olophrum latum*	M. Thayer	4
Coleoptera	Staphylinidae	*Olophrum rotundicolle*	M. Thayer	15
Coleoptera	Staphylinidae	Staphylinidae	D. S. Sikes	1
Coleoptera	Staphylinidae	*Tachinus apterus*	A. Newton	2
Coleoptera	Staphylinidae	*Tachinus apterus*	D. S. Sikes	65
Coleoptera		Coleoptera	J. Williamson	1

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[5]May refer to *Micralymma dicksoni* (Mäklin, 1881) based on DNA barcode results (Sikes et al., in press)

Order	Family	Scientific Name	Identified By	Count
Collembola	Sminthuridae	Sminthuridae	Dan Bogan	0
Collembola		Collembola	Dan Bogan	0
Collembola		Collembola	J. Williamson	1188
Diptera	Anthomyiidae	Anthomyiidae	S. Peek	194
Diptera	Anthomyiidae	Anthomyiidae ?	J. Williamson	59
Diptera	Anthomyiidae	*Botanophila rubrigena*	S. Peek	23
Diptera	Anthomyiidae	*Botanophila* sp.	S. Peek	14
Diptera	Anthomyiidae	*Delia pilifemur*	S. Peek	1
Diptera	Anthomyiidae	*Delia* sp.	S. Peek	42
Diptera	Anthomyiidae	*Fucellia* sp.	S. Peek	2
Diptera	Bibionidae	*Bibio* sp.	S. Peek	1
Diptera	Calliphoridae	Calliphoridae ?	J. Williamson	3
Diptera	Calliphoridae	*Cynomya cadaverina*	unknown	R&R
Diptera	Calliphoridae	*Cynomya mortuorum*	S. Peek	43
Diptera	Calliphoridae	*Cynomya mortuorum*	unknown	R&R
Diptera	Ceratopogonidae	Ceratopogonidae	J. Williamson	7
Diptera	Ceratopogonidae	Ceratopogonidae ?	J. Williamson	5
Diptera	Chironomidae	*Nanocladius* (*Plecopteracoluthus*) sp.	Dan Bogan	0
Diptera	Chironomidae	*Acricotopus* sp.	Dan Bogan	0
Diptera	Chironomidae	*Chaetocladius dentiforceps* group	Dan Bogan	0
Diptera	Chironomidae	*Chaetocladius sp.?*	Dan Bogan	0
Diptera	Chironomidae	Chironomidae	J. Williamson	5
Diptera	Chironomidae	Chironomidae	S. Peek	46
Diptera	Chironomidae	Chironomidae ?	J. Williamson	3
Diptera	Chironomidae	Chironominae	Dan Bogan	0
Diptera	Chironomidae	*Chironomus* sp.	Dan Bogan	0
Diptera	Chironomidae	*Constempellina* sp.	Dan Bogan	0
Diptera	Chironomidae	*Corynoneura lobata*	Dan Bogan	0
Diptera	Chironomidae	*Corynoneura scutellata*	Dan Bogan	0
Diptera	Chironomidae	*Cricotopus* (*Isocladius*) sp.	Dan Bogan	0
Diptera	Chironomidae	*Cricotopus* or *Orthocladius*	Dan Bogan	0
Diptera	Chironomidae	*Cricotopus* sp.	Dan Bogan	0
Diptera	Chironomidae	*Cricotopus tremulus* group	Dan Bogan	0
Diptera	Chironomidae	*Cryptochironomus stylifera*	Dan Bogan	0
Diptera	Chironomidae	*Diamesa* sp.	Dan Bogan	0
Diptera	Chironomidae	*Eukiefferiella brehmi* group	Dan Bogan	0
Diptera	Chironomidae	*Eukiefferiella claripennis* group	Dan Bogan	0
Diptera	Chironomidae	*Eukiefferiella coerulescens* group	Dan Bogan	0
Diptera	Chironomidae	*Eukiefferiella gracei* group	Dan Bogan	0
Diptera	Chironomidae	*Heterotrissocladius maeaeri* group	Dan Bogan	0
Diptera	Chironomidae	*Hydrobaenus conformis* group	Dan Bogan	0
Diptera	Chironomidae	*Hydrobaenus lapponicus* group	Dan Bogan	0
Diptera	Chironomidae	*Hydrobaenus* sp.	Dan Bogan	0
Diptera	Chironomidae	*Metriocnemus eurynotes* group	Dan Bogan	0
Diptera	Chironomidae	*Metriocnemus fuscipes* group	Dan Bogan	0
Diptera	Chironomidae	*Micropsectra* sp.	Dan Bogan	0
Diptera	Chironomidae	*Oliveridia* sp.	Dan Bogan	0
Diptera	Chironomidae	Orthocladiinae	Dan Bogan	0
Diptera	Chironomidae	*Orthocladiinae* genus "Greenland"	Dan Bogan	0
Diptera	Chironomidae	*Orthocladius* (*Eudactylocladius*) sp. 1	Dan Bogan	0
Diptera	Chironomidae	*Orthocladius* (*Eudactylocladius*) sp. 2	Dan Bogan	0
Diptera	Chironomidae	*Orthocladius* (*Euorthocladius*) sp. 1	Dan Bogan	0

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Order	Family	Scientific Name	Identified By	Count
Diptera	Chironomidae	*O.* (*Mesorthocladius*) cf. *frigidus*	Dan Bogan	0
Diptera	Chironomidae	*Orthocladius* (*Mesorthocladius*) sp. 1	Dan Bogan	0
Diptera	Chironomidae	*Orthocladius* (*Orthocladius*) cf. *dorenus*	Dan Bogan	0
Diptera	Chironomidae	*O.* (*Orthocladius*) cf. *obumbratus*	Dan Bogan	0
Diptera	Chironomidae	*O.* (*Orthocladius*) cf. *rubicundus*	Dan Bogan	0
Diptera	Chironomidae	*Orthocladius* (*Orthocladius*) sp. 1	Dan Bogan	0
Diptera	Chironomidae	*Orthocladius* (*Pogonocladius*) sp. 1	Dan Bogan	0
Diptera	Chironomidae	*Paratanytarsus* sp.	Dan Bogan	0
Diptera	Chironomidae	*Psectrocladius limbatellus* group	Dan Bogan	0
Diptera	Chironomidae	*Psectrocladius psilopterus* group	Dan Bogan	0
Diptera	Chironomidae	*Psectrocladius sordidellus* group	Dan Bogan	0
Diptera	Chironomidae	*Psectrocladius* sp.	Dan Bogan	0
Diptera	Chironomidae	*Pseudokiefferiella* sp.	Dan Bogan	0
Diptera	Chironomidae	*Pseudosmittia* sp.	Dan Bogan	0
Diptera	Chironomidae	*Psilometriocnemus* sp.	Dan Bogan	0
Diptera	Chironomidae	*Smittia* sp.	Dan Bogan	0
Diptera	Chironomidae	*Thienemanniella* sp.	Dan Bogan	0
Diptera	Chironomidae	*Tvetenia* sp.	Dan Bogan	0
Diptera	Coelopidae	*Coelopa* sp.	S. Peek	16
Diptera	Dolichopodidae	*Chrysotus* sp.	S. Peek	4
Diptera	Empididae	*Clinocera* sp.	Dan Bogan	0
Diptera	Empididae	Empididae	J. Williamson	3
Diptera	Empididae	*Empis* sp.	S. Peek	21
Diptera	Empididae	*Heleodromia* sp.	S. Peek	1
Diptera	Empididae	*Rhamphomyia* sp.	S. Peek	36
Diptera	Ephydridae	Ephydridae ?	J. Williamson	16
Diptera	Fanniidae	*Fannia* sp.	S. Peek	6
Diptera	Heleomyzidae	*Aecothea* sp.	S. Peek	5
Diptera	Heleomyzidae	*Neoleria* sp.	S. Peek	1
Diptera	Muscidae	Muscidae	S. Peek	2
Diptera	Muscidae	Muscidae ?	J. Williamson	18
Diptera	Muscidae	*Phaonia bidentata*	S. Peek	1
Diptera	Muscidae	*Phaonia consobrina*	S. Peek	85
Diptera	Muscidae	*Phaonia* sp.	S. Peek	38
Diptera	Muscidae	*Spilogona* sp.	S. Peek	60
Diptera	Mycetophilidae	*Boletina borealis*	J. Salmela	1
Diptera	Mycetophilidae	*Boletina* sp.	S. Peek	1
Diptera	Mycetophilidae	*Coelosia* sp.	S. Peek	3
Diptera	Mycetophilidae	*Mycetophila* sp.	S. Peek	9
Diptera	Mycetophilidae	Mycetophilidae	J. Williamson	1
Diptera	Mycetophilidae	Mycetophilidae	S. Peek	11
Diptera	Mycetophilidae	Mycetophilidae ?	J. Williamson	69
Diptera	Phoridae	*Megaselia* sp.	S. Peek	4
Diptera	Phoridae	Phoridae	S. Peek	30
Diptera	Piophilidae	*Parapiophila* sp.	S. Peek	1
Diptera	Piophilidae	Piophilidae ?	J. Williamson	2
Diptera	Rhagionidae	*Chrysopilus* sp.	S. Peek	10
Diptera	Scathophagidae	*Gimnomera* sp.	S. Peek	1
Diptera	Scathophagidae	*Microprosopa pallidicauda*	S. Peek	3
Diptera	Scathophagidae	*Microprosopa* sp.	S. Peek	10
Diptera	Scathophagidae	*Scathophaga* sp.	unknown	R&R
Diptera	Scathophagidae	*Scathophaga* sp.	S. Peek	495

Continued on next page…

Order	Family	Scientific Name	Identified By	Count
Diptera	Scathophagidae	Scathophagidae ?	J. Williamson	10
Diptera	Sciaridae	*Lycoriella* sp.	S. Peek	5
Diptera	Sciaridae	Sciaridae	J. Williamson	8
Diptera	Sciaridae	Sciaridae	S. Peek	51
Diptera	Sciaridae	Sciaridae ?	J. Williamson	65
Diptera	Simuliidae	*Prosimulium* sp.	Dan Bogan	0
Diptera	Simuliidae	*Prosimulium ursinum*	unknown	R&R
Diptera	Simuliidae	Simuliidae	Dan Bogan	0
Diptera	Simuliidae	*Simulium* sp.	Dan Bogan	0
Diptera	Sphaeroceridae	*Copromyza* sp.	S. Peek	2
Diptera	Sphaeroceridae	Sphaeroceridae	S. Peek	30
Diptera	Sphaeroceridae	Sphaeroceridae ?	J. Williamson	1
Diptera	Syrphidae	*Helophilus* sp.	S. Peek	1
Diptera	Tipulidae	*Dicranota* sp.	Dan Bogan	0
Diptera	Tipulidae	Limoniinae	S. Peek	1
Diptera	Tipulidae	*Ormosia* sp.	Dan Bogan	0
Diptera	Tipulidae	*Tipula* sp.	Dan Bogan	0
Diptera	Tipulidae	*Tipula* sp.	S. Peek	12
Diptera	Tipulidae	Tipulidae	Dan Bogan	0
Diptera	Trichoceridae	Trichoceridae ?	J. Williamson	2
Diptera		Diptera	C. Coon	10
Diptera		Diptera	J. Williamson	81
Diptera		Diptera	S. Peek	1
Hemiptera	Aphididae	Aphididae	J. Williamson	10
Hemiptera	Saldidae	*Chiloxanthus stellatus*	D. S. Sikes	2
Hemiptera		Heteroptera	D. S. Sikes	1
Hymenoptera	Apidae	*Bombus polaris*	P. H. Williams	11
Hymenoptera	Apidae	*Bombus polaris*	unknown	1
Hymenoptera	Ichneumonidae	Ichneumonidae	D. S. Sikes	2
Hymenoptera		Apocrita	D. S. Sikes	24
Hymenoptera		Apocrita	J. Williamson	9
Hymenoptera	Cynipoidea	Cynipoidea	D. S. Sikes	1
Hymenoptera		Hymenoptera	D. S. Sikes	91
Hymenoptera		Hymenoptera	J. Williamson	20
Hymenoptera		Hymenoptera	S. Peek	6
Hymenoptera		Symphyta	D. S. Sikes	1
Hymenoptera		Symphyta	J. Williamson	21
Lepidoptera	Erebidae	*Pararctia subnebulosa*	K. W. Philip	1
Lepidoptera	Noctuidae	*Xestia alaskae*	C. D. Ferris	2
Lepidoptera		Lepidoptera	D. S. Sikes	4
Lepidoptera		Lepidoptera	J. Williamson	14
Plecoptera	Capniidae	*Capnia* sp.	Dan Bogan	0
Plecoptera	Capniidae	Capniidae	Dan Bogan	0
Plecoptera	Nemouridae	*Nemoura arctica*	K. Stewart	2
Plecoptera	Nemouridae	*Nemoura arctica* ?	D. S. Sikes	43
Plecoptera	Nemouridae	*Nemoura* sp.	Dan Bogan	0
Plecoptera	Nemouridae	*Nemoura* sp.	K. Stewart	1
Plecoptera	Nemouridae	Nemouridae	Dan Bogan	0
Plecoptera	Nemouridae	Nemouridae sp1	Dan Bogan	0
Plecoptera	Nemouridae	*Podmosta* sp.	Dan Bogan	0
Plecoptera		Plecoptera	D. S. Sikes	30
Plecoptera		Plecoptera	J. Williamson	1

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Order	Family	Scientific Name	Identified By	Count
Siphonaptera	Ceratophyllidae	*Amalaraeus penicilliger*	G. P. Holland	R&R
Siphonaptera	Ceratophyllidae	*Megabothris groenlandicus*	G. P. Holland	R&R
Trichoptera	Apataniidae	*Apatania* sp.	Dan Bogan	0
Trichoptera	Brachycentridae	*Brachycentrus* sp.	Dan Bogan	0
Trichoptera	Brachycentridae	*Micrasema scissum*	Dan Bogan	0
Trichoptera	Lepidostomatidae	*Lepidostoma* sp.	H. H. Ross	R&R
Trichoptera	Limnephilidae	*Dicosmoecus* sp.	Dan Bogan	0
Trichoptera	Limnephilidae	*Glyphopsyche* sp.	H. H. Ross	R&R
Trichoptera	Limnephilidae	*Grensia* sp.	H. H. Ross	R&R
Trichoptera	Limnephilidae	*Grensia praeterita*	unknown	1
Trichoptera	Limnephilidae	*Grensia* sp.	Dan Bogan	0
Trichoptera	Limnephilidae	Limnephilidae	Dan Bogan	0
Trichoptera	Limnephilidae	Limnephilidae sp1	Dan Bogan	0
Trichoptera	Limnephilidae	Limnephilus	H. H. Ross	R&R
Trichoptera	Limnephilidae	*Limnephilus picturatus*	unknown	1
Trichoptera		Trichoptera	C. Coon	1
Trichoptera		Trichoptera	J. Williamson	2
		Insecta	A. Haberski	3
		Insecta	J. Williamson	121
Amphipoda	Crangonyctidae	*Crangonyx* sp.	Dan Bogan	0
Isopoda	Chaetiliidae	*Saduria entomon*	M. S. Wilson	R&R
Isopoda	Chaetiliidae	*Saduria* sp.	D. S. Sikes	1
Calanoida	Diaptomidae	*Diaptomus pribilofensis*	M. S. Wilson	R&R
Calanoida	Temoridae	*Eurytemora gracilicauda*	M. S. Wilson	R&R
Cyclopoida	Cyclopidae	*Cyclops kolensis*	M. S. Wilson	R&R
		Copepoda	Dan Bogan	0
Podocopida	Candonidae	*Candona subgibba*	M. S. Wilson	R&R
Anthoathecata	Hydridae	*Hydra*	unknown	R&R
Veneroida	Sphaeriidae	*Pisidium lilljeborgi*	unknown	R&R
Veneroida	Sphaeriidae	Sphaeriidae	Dan Bogan	0
Basommatophora	Planorbidae	*Gyraulus deflectus*	unknown	R&R
Basommatophora	Planorbidae	*Gyraulus parvus*	Dan Bogan	0
Basommatophora	Planorbidae	Planorbidae	Dan Bogan	0
Heterostropha	Valvatidae	*Valvata* sp.	Dan Bogan	0
Stylommatophora	Succineidae	*Succinea strigata*	unknown	R&R
		Gastropoda	J. Williamson	18
		Mollusca	J. Williamson	1
		Nematoda	Dan Bogan	0
Neorhabdocoela	Typhloplanidae	*Mesostoma platygastricum*	unknown	R&R
		Animalia	J. Williamson	1

Acknowledgments

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Food review: *Urocerus flavicornis* (Fabricius) (Hymenoptera: Siricidae)

by Matt Bowser[1]

Figure 1: Larvae from spruce wood, 8.March.2015.

Last March as my family was working on putting up wood at my household in Kasilof, there was a time when we came across quantities of large larvae in spruce (*Picea ×lutzii* Little) rounds from a tree that had died in the last couple of years. We usually feed these to our chickens, but we found enough at this time that a meal for a person could be considered.

Most of these I am pretty confident were *Urocerus flavicornis* (Fabricius), which we have collected here (KNWR:Ento:8613), but some of the smaller siricid larvae could have been *Sirex nitidus* (T. W. Harris), also known from our property (KNWR:Ento:7175). There were also a few of what may have been beetle larvae.

I sautéed all of these larvae as if they were shrimp in a scampi sauce of butter, garlic, lemon juice, and crumbled, dried celery leaves. They cooked to a firm texture not unlike cooked shrimp and had a surprisingly sprucy flavor like spruce tips, spruce gum, or pine nuts. This seemed a little out-of-place in the scampi, but it was not bad. The terminal spines were surprisingly sharp and poky. It would have been best to chop off the hind ends of these larvae after they had been cooked.

In future, I think *U. flavicornis* larvae might be best served roasted, then chopped into a fresh pesto, where that sprucy taste might be a welcome, cheaper alternative to pine nuts.

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The spruce aphid, a non-native species, is increasing in range and activity throughout coastal Alaska

by Elizabeth Graham[1] *and Jason Moan*[2]

Figure 1: Sitka spruce trees heavily infested with spruce aphid in Halibut Cove. Photo Credit: Garret Dubois

Issue

The spruce aphid originated in continental Europe and became a major pest in the 1800s and 1900s after widespread planting of Sitka spruce. It eventually made its way to North America, likely via nursery stock, first reported in 1915 in Vancouver, British Columbia. Spruce aphid, *Elatobium albietinum* Walker, was first reported in Alaska in 1927 in the southeast town of Wrangell, but outbreaks did not become frequent, extensive, or severe until 1967 where it was found injuring ornamental spruce in Sitka. It has since been a reoccurring pest of Sitka spruce throughout the coastal areas of Southeast Alaska, such as Juneau, Ketchikan, Craig, and especially Sitka. During heavy outbreak years, over 40,000 acres of damage was attributed to spruce aphid during aerial detection surveys. It has previously been detected in the Prince William Sound area, most notably in 2005 when it was mapped on over 4500 acres, but significant damage was not reported outside Southeast Alaska again until 2015 when it was found on the Kenai Peninsula. Concerned homeowners on the Kenai Peninsula reported numerous Sitka spruce trees with brown needles (Figure 1).

Entomologists from Forest Health Protection and AK Division of Forestry identified the causal agent as the spruce aphid, confirming a considerable expansion of the aphid's known range in Alaska (Figure 2). In maritime environments, spruce aphid outbreaks are directly correlated with mild winters. Spruce aphids begin actively feeding in late winter, and research indicates that actively feeding aphids cannot tolerate temperatures below -7°C for prolonged periods or frost events at even lower temperatures (Day and Kidd, 1998; Powell and Parry, 1976). Aphids become inactive during prolonged periods of cold and eventually starve to death. Alaska is on the forefront of climate change; over the last 60 years temperature has increased an average of 6°F state-wide (Chapin et al., 2014). The warming climate may have triggered a spruce aphid range expansion or alternatively increased chance of establishment and outbreak after separate introduction(s) into these areas.

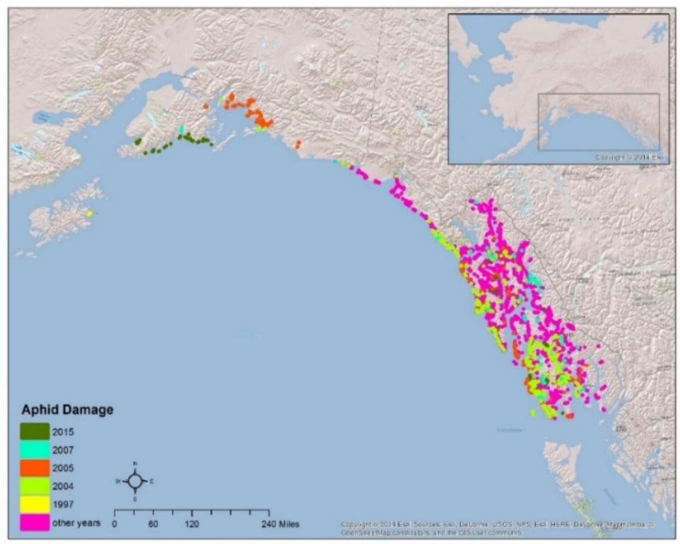

Figure 2: Spruce aphid damage mapped during aerial detection survey since 1987. Years with heavy aphid outbreaks are distinguished by color. Spruce aphids were confirmed on the Kenai Peninsula for the first time in 2015.

Aphid feeding is restricted to mature (one year old and older) needles, with new foliage being nutritionally inadequate. Feeding damage causes the needles to turn yellow then brown and fall off, leaving the crown looking thin (Figure 3). Repeated years of heavy defoliation can lead to mortality or subject the tree to secondary damage agents, such as spruce beetle.

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Current situation

Spruce aphid activity is on the rise in 2016 with confirmed activity on the western side of the Kenai Peninsula. Concerned citizens from Homer reported damage associated with these aphids, with severe defoliation observed in some of the infested trees. A site visit conducted by FHP and Alaska Division of Forestry specialists in late February 2016 found scattered infested trees throughout the Homer area, varying in severity of defoliation. Damage appeared to be confined to trees in close proximity to the coast. FHP staff also surveyed for spruce aphid in the towns of Seward, Whittier, and Hope, as well as along Turnagain Arm, the Hope Highway, and the Portage area but no signs of aphid activity were observed.

Figure 3: Spruce aphids feeding on the needles of Sitka spruce.

The mild winter temperatures in 2016 promoted aphid activity. There is concern that the aphids may gradually move into higher elevation forests or further inland. There is also concern about the potential susceptibility of spruce aphid-weakened spruce to spruce beetle on the Kenai Peninsula. Spruce beetles preferentially attack and kill weakened trees. The combination of aphid and beetle activity could lead to increased mortality.

To answer some of these questions and concerns a long-term plot network will be established in 2016 to monitor the impact and range of spruce aphid throughout coastal Alaska. FHP entomologists will be working in collaboration with an entomologist from the Rocky Mountain Research Station to establish a risk assessment model for predicting aphid outbreaks. High-value trees can be treated with insecticides to prevent or limit aphid attack and the proposed risk assessment model can serve as an indicator of when treatment may be warranted and as a warning system to land managers prior to an outbreak.

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A history and update of the Kenelm W. Philip Collection, currently housed at the University of Alaska Museum

by K. M. Daly[1]

Introduction

Dr. Kenelm W. Philip, founder of the Alaska Lepidoptera Survey, was the owner of the largest private Arctic Lepidoptera collection in the world with over 111,000 individual specimens. I have had the honor of caring for his collection since Dr. Philip's sudden death two years ago, and felt it time to share a brief history along with an update on work that has occurred since the collection arrived at the University of Alaska Museum (UAM). Dr. Ken Philip died in March of 2014 at the age of 82, after calling Fairbanks, Alaska home since October of 1965. He came to Fairbanks to work as a professor of physics with the Geophysical Institute, having previously worked as a research staff astronomer at the Yale University Observatory in New Haven, Connecticut. He began collecting and studying Lepidoptera as a child; though he never formally taught entomology in a classroom he inspired hundreds of people to take interest in these animals through his genuine enthusiasm for knowledge.

Citizen science: the Alaska Lepidoptera Survey

Specimens were acquired across eastern Russia, Alaska and western Canada through the help of over 600 volunteers of the Alaska Lepidoptera Survey. Dr. Philip prepared and mailed collecting equipment free of charge to anyone who was willing to capture butterflies and moths on his behalf. Many volunteers were researchers headed to remote areas of the state for field projects, but anyone who expressed interest was supplied with a collecting kit. Housewives and children who caught specimens in backyards and even pipeline workers who were stationed at camps along the Dalton Highway were involved, along with National Park Service employees who obtained permits to collect for Dr. Philip. He stored the entirety of his collection, what I've counted to be over 127,000 specimens, in a private custom laboratory he had built in his home in Fairbanks.

Support and scientific appointments

Spread specimens were protected through storage in cabinets provided by the Smithsonian Institution, who supported this effort with needed supplies to curate the collection. Dr. Philip became a research associate of both the Smithsonian and the University of Alaska Museum in 1971, and from 1985 he also maintained an appointment as a research scientist for the Institute of Arctic Biology at the University of Alaska Fairbanks. Alaska's National Park Service supported Dr. Philip though issuing permits for both himself and interested volunteers to collect within Park boundaries beginning in the late 1960's. He made dear friends from Toolik Field Station on the North Slope of Alaska to Kluane Lake Field Station in the Yukon Territory and everywhere between; his mission to document Lepidoptera fauna led him across Alaska and western Canada for almost 50 years, along with four trips he took to collect specimens around eastern Russia.

Dr. Philip served as Vice President of the Lepidopterists Society for six years. He also served on their Executive Council and Editorial Committee, and for decades he prepared the annual Season Summary of Lepidoptera activity in Alaska and western Canada as the Zone Coordinator of the Far North region each year. In these positions, Dr. Philip maintained professional correspondence with researchers across the globe. He was truly a phenomenal scientist because he treated new students of Lepidoptera with the same respect and encouragement for our shared joy of knowledge.

2011: My first Alaska summer

I had the pleasure of corresponding with Dr. Philip beginning in 2011, when he received and identified my photographs of caterpillars and butterflies from Toolik Field Station on the North Slope. I traveled to Alaska for my first time to spend that summer at the field station as an undergraduate student. I wanted to learn about the caterpillars that were prevalent within an established snowmelt experiment run by Dr. Heidi Steltzer, my biology undergraduate advisor at Fort Lewis College in Durango, Colorado. I initially sent my photos to Dr. Derek Sikes at the University of Alaska Museum, who then shared them with Dr. Philip. He kindly encouraged me to keep sending him whatever photos I could capture, and shared his own photos of live and pinned specimens. Dr. Philip had initially planned to come to Toolik that summer, but weather patterns led him to travel down the Dempster Highway instead so I didn't get a chance to meet him in person before I left back to Colorado. I was disappointed, for everyone who had met him

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at Toolik had told me what a congenial and knowledgeable person he was.

2013: return to Alaska forever

I knew Alaska was where I wanted to be, and was fascinated by what Dr. Philip had shared with me about the Lepidoptera of the North Slope. In 2013, I was ecstatic when I was offered a chance to return again to Toolik for another summer. This time, I was a field technician for PhD entomology student Ashley Asmus. I loved sharing butterfly images again with Dr. Philip in my free time. At the end of that summer, he invited me to visit the collection any time I had the chance. I was thrilled to have the opportunity and when I was offered a temporary research assistant job at UAF that fall, it seemed my destiny to move to Fairbanks. I couldn't wait to meet Dr. Philip in person; his humble and friendly demeanor made viewing his magnificent collection all the more astounding to me.

I also took my first tour of the University of Alaska Museum in September of 2013, where Dr. Sikes offered me Lepidoptera specimens to take to Dr. Philip. That winter, I learned how to prepare and spread the specimens, and to take the time that Dr. Philip did to straighten each head and curve antennae into the most natural bend. His mastery of this art was but one part of his life where he excelled; aside from his career as an astronomer & physicist, he also studied fractals of the Mandlebrot equation and created a range mapping software program (one of the first of its kind) that sold globally. He seemingly had a photographic memory for knowledge and could summon any bit of information with a smile, along with a relevant quote or pun as he saw fit. His willingness to welcome me into his life that winter to patiently teach me about Arctic Lepidoptera was a tremendous gift. He was one of the most remarkable people I have ever had the pleasure of knowing. Many people have shared the same thought with me since his passing. He clearly impacted thousands of people through his diverse interests and efforts.

March 2014

When he died suddenly in March of 2014, Dr. Philip left behind legacies in both science and the arts; his immense collection of classical music was donated to KUAC radio station and his donated book collection quadrupled the inventory of the Literacy Council of Alaska. His Lepidoptera collection is a priceless resource that will certainly influence generations of future research. The majority of the collection was willed to the Smithsonian, with the remainder to be split with the University of Alaska Museum.

The entire collection is currently still at UAM but it is likely the split will begin in 2017. The National Park Service, who supported Dr. Philips work over the years, recognized the immediate need to tend to the collection after his sudden death. They provided funding for one year of work to inventory it and to database specimens that he collected within Alaskas National Parks. As Dr. Philip's last student, I was offered this position by Dr. Sikes and officially began that work in June of 2014. However, it was immediately critical to secure the collection for transfer to the University of Alaska Museum directly after Dr. Philip's death.

We opened almost 500 drawers to push down every specimen pin to ensure they were safe for transport. The drawers were laid carefully into low stacks in the back of a Uhaul truck, using plenty of blankets for packing. Drawers and boxes lined the back hallway of the museum as we packed away shelf after shelf of Dr. Philip's laboratory. The smell of naphthalene was so powerful that museum employees went home ill, and the Children's Museum (which is no longer at UAM) closed for two days!

The drawers and also boxes containing paperwork, correspondence and specimens lined the entire hallway, stacked shoulder high in some places. Miscellaneous containers of all shapes and sizes were found to be filled with specimens sent in by collectors. Dr. Philip had a sponsorship with the Sucrets cough drop company for their signature metal containers, which are perfect for safely storing glassine envelopes filled with specimens. He also supplied his volunteer collectors with Amphora tobacco canisters. Those, along with Kodak film boxes, BandAid tins, cigar boxes, fishing tackle boxes and Tupperware tubs were dutifully returned to him by his Alaska Lepidoptera Survey volunteers, packed with glassine envelopes containing butterflies and moths from across Alaska. Dr. Philip would then tape an index card to the container, and using a black marker, he would enscribe the year, locality, and collector's last name in his hasty scrawl.

Cleaning, inventory and databasing

My first task once I began work in June of 2014 was to spend over a month scraping white naphthalene crystals out of grooves carved in the sides of each drawer. I worked in a fume hood, and placed the specimens in a different drawer while doing this. While the crystals are long gone now, the drawers still carry this scent! After cleaning, each drawer of specimens was placed in a -40°F freezer for two days to kill any pests. After the freezer, I brought each load downstairs to the collection range where I inventoried each container.

I counted total numbers of specimens and attempted to decipher Dr. Philip's handwriting so that I could enter whatever information was on the container into a spreadsheet. Each container received a unique numerical code, based on its place in the collection, so that I could find it again. I also searched for specimens which were on loan to Dr. Philip from the Smithsonian and other institutions. I learned how to database these and also National Park specimens using ARCTOS, a collection management program used by UAM. Time passed quickly down in the collection range, studying each container and wondering about the places where each creature had flown.

2015: First visiting lepidopterists arrive for the collection

May of 2015 brought the first of several researchers to UAM to use the collection. The longest visitor, Dr. Zdenek F. Fric, was a Fulbright scholar from the Czech Republic. He and his family spent the entire summer in Alaska collecting butterflies along all the major road systems to analyze genetic patterns across Beringian species. Dr. Fric kindly identified specimens from Dr. Philip's collection along with butterflies which were brought to the museum by citizens of Fairbanks. Several people who collected for the Alaska Lepidoptera Survey have continued to send in or hand deliver specimens to UAM.

Dr. Fric helped in my curation efforts immensely when he translated the labels of drawers that Dr. Philip had obtained from eastern Russia, written in Cyrillic. At the end of the summer, Dr. Fric also generously donated several hundred butterfly specimens which he collected to UAM. All the 2015 Lepidoptera donations are still being curated but once complete those data will be publicly accessible in ARCTOS. He collected butterfly legs off these specimens and others from Dr. Philip's collection for DNA analysis in his home country. Once his research is complete, I will share notice of his findings through the AKES's listserv and website.

Dr. Andrew Warren, senior collections manager at the McGuire Center for Lepidoptera and Biodiversity (MGCL) in Florida, visited the collection in June of 2015 to examine butterflies in the genus *Oeneis*. He was particularly interested in viewing *Oeneis chryxus* specimens, as he had discovered within the MGCL collection a series of butterflies collected along the Tanana River which were darker and larger than the *Oeneis chryxus* of western Canada and elsewhere. When we looked through the drawers together, I saw how Dr. Philip had placed a determination of *Oeneis chryxus* ? on some of these specimens.

Upon returning from his visit, Dr. Warren dug into this mysterious creature by examining hundreds more Alaskan specimens from public and private collections. He found both morphological and genetic differences in this animal and has since designated the butterfly a new species, *Oeneis tanana*. The paper describing these findings (Warren et al., 2016) was published 15 March 2016 in *The Journal of Research on the Lepidoptera*. Each of these projects is certain to bring increased attention to Alaska's butterfly species, a mission which Dr. Philip initiated upon his arrival here in 1966.

Import of the Alaska Lepidoptera Survey databases

In late September of 2015; the entirety of the Alaska Lepidoptera survey databases were made publicly accessible in ARCTOS, our museum collection management system. Dr. Philip maintained several databases of butterflies which he or ALS volunteers had collected until 2005. The databases were imported after months of work by Matt Bowser, Derek Sikes and myself. We had discovered that there were collecting events within the volunteer database that had no geographic coordinates. To determine precisely where that volunteer had collected, each locality was searched on Google Maps, Google Earth, and occasionally even in Orth (1971).

As a relative newcomer to Alaska, I was only somewhat familiar with the North Slope before moving to Fairbanks. It was incredible to search for each of these localities on a map to gain a deeper appreciation for the vast reaches of the Alaska Lepidoptera Survey. I am happy to report that the vast majority of localities were found, with only a handful remaining which were too ambiguous to confidently decipher. I maintain hope that when the time comes, further information on these localities may be revealed as Dr. Philip's specimens are curated and his paperwork is organized.

Butterflies of Alaska, first edition

After all the georeferencing was complete, the ALS databases were uploaded to ARCTOS and it finally became possible to create dot maps portraying the range of every species. During our time of working on the georeferencing of these data, Dr. Cliff Ferris, one of Dr. Philip's best friends, had undertaken writing the first *Butterflies of Alaska* field guide. This project was one that Dr. Philip intended to complete himself but died before that could be actualized. He did manage to capture live photographs of almost every species of Alaska's butterflies, and those photos accompany pinned specimen images taken by Dr. Ferris. The guide (Philip and Ferris., 2015) was authored by Dr. Philip (posthumously) and Dr. Ferris. It became available for sale in December, 2015. Bioquip sold copies online, and several booksellers across the state purchased orders. In Fairbanks, the guide is sold at the University of Alaska Museum, Gullivers Book Store and Creamer's Field. The furthest order for copies came from Japan, from a bookseller and butterfly collecter who had traded specimens with Dr. Philip! There were only 500 copies printed, and these sold out by the end of January, 2016. A second edition is being printed soon to include the new species, *Oeneis tanana*. It will be available before butterfly season, 2016—keep an eye on the AkEntoNet-L listserv for an announcement!

Curation efforts continue

Coming into butterfly season of 2016, it feels as though the past two years have gone by in a flash. It has been my sincere pleasure to care for Dr. Philip's collection, and I look forward to my coming months of work with it. I am happy to share that I am now the curatorial assistant to Dr. Derek Sikes and a graduate student at the University of Alaska Fairbanks. I began in September of 2015 and am using the collection to ask several research questions. These will be detailed in a future article in the *AKES Newsletter*. This semester, I am currently training 16 undergraduate students to link Dr. Philip's specimens to their digital records. Another undergraduate student is working to photograph every drawer in the collection. Once complete, these will be publicly accessible and also will go on display in an exhibit planned for 2017 at UAM. It will be wonderful to share the life and legacy of Dr. Ken Philip and his Alaska Lepidoptera Survey with museum visitors. Until then, I look forward to sharing future news of work on his collection with the Alaska Entomological Society community.

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The Blackberry Skeletonizer, *Schreckensteinia festaliella* (Hübner) (Lepidoptera: Schreckensteiniidae) in Alaska

by Matt Bowser[1], Matt Goff[2], and Kristin DuBour[3]

Figure 1: Photo of *Schreckensteinia festaliella*, Sitka, Alaska, 29.June.2008 by Matt Goff (http://bugguide.net/node/view/197612).

The Blackberry Skeletonizer, *Schreckensteinia festaliella* (Hübner, 1819), now appears to be present in eastern Alaska. A Palearctic species, *S. festaliella* was first reported in Canada by Pohl et al. (2005) and has more recently been documented from as close to Alaska as British Columbia (Pohl et al., 2015). This species had not reported from Alaska by Ferris et al. (2012). Although no specimens are available for definitive confirmation, two recent observations are consistent with its presence in the state.

The first was a moth photographed by Matt Goff in Sitka on June 29, 2008 (BugGuide record 197612) that was later tentatively identified from the photographs as *S. festaliella*. Derek Sikes made a corresponding observation record on Arctos: UAMObs:Ento:234757.

The second record is from a Forest Inventory and Analysis Program pilot project (Andersen et al., 2015) on Tetlin National Widlife Refuge. Sweep net samples from twenty-six sites had been sent to Research and Testing Laboratory, Lubbock, Texas, for next-generation sequencing on an illumina MiSeq using the *ZBJ-ArtF1c/ZBJ-ArtR2c* primer set (Zeale et al., 2011) targeting COI, yielding a

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157 bp COI fragment. Sequencing and analysis methods are available from Research and Testing Laboratory (2014). A cluster of 21,348 reads of a sequence matching *S. festaliella* was detected at one site. Collection data: USA: Alaska, Tetlin National Wildlife Refuge, USDA Forest Inventory and Analysis plot 36354, hills above Cheslina River, 62.626853°N, 142.644882°W ±50 m, 10.June.2014, Jeff Horoshak. Collection data are available via Arctos record UAMObs:Ento:235210 and GenBank BioSample record SAMN04532700; sequence data are available through GenBank SRA submission SRR3212095. See, for example, run SRR3212095, spot id 26431.

Larve of *S. festaliella* feed on *Rubus* (Pohl et al., 2005), including raspberry (*Rubus idaeus* L.) on which it can be "very damaging" according to Alford (2014). Johansen and Kobro (1996) described a rather severe outbreak of *S. festaliella* on cloudberry (*Rubus chamaemorus* L.) in Norway.

Figure 2: Map of Alaska records of *Schreckensteinia festaliella* from Arctos as of 10.March.2016.

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Northern spruce engraver monitoring in wind-damaged forests in the Tanana River Valley of Interior Alaska

by Jason E. Moan[1]

In September of 2012, a large scale wind event occurred in the Tanana River Valley which resulted in an estimated 1.4 million acres of forest damage. The damage consisted of numerous broken, uprooted, and leaning spruce and birch. The details of this wind event were first reported in FS-R10-FHP (2013) and subsequent Forest Health Conditions in Alaska reports have provided updates on northern spruce engraver (*Ips perturbatus*) damage and monitoring in the wind-impacted areas (FS-R10-FHP, 2014, 2015).

Northern spruce engraver activity is generally found in scattered pockets of weakened or damaged spruce such as those along the edges of wildfires, rivers, or impacted by weather events. Often, windblown trees dry out quickly and become less suitable for the beetles. Many of the wind-damaged spruce stands in the Tanana River Valley, however, were left leaning but alive, potentially providing a persistent supply of weakened host trees for the beetles.

In the year following the windstorm, minor amounts of northern spruce engraver-caused mortality (~115 acres) were observed during the annual forest health aerial detection surveys conducted by Alaska Division of Forestry and FHP staff. By 2014, an estimated 425 acres of scattered mortality was mapped in the area. Observed mortality increased to nearly 900 acres of scattered damage in 2015. The damage observed thus far has been widely scattered individual trees or small groups of trees. To date, no extensive contiguous areas of northern spruce engraver-caused mortality have been observed in the wind-impacted area.

In response to the windstorm, local natural resource managers between Delta Junction and Tok have been actively working to mitigate bark beetle activity, using various techniques including those outlined in recent northern spruce engraver research (Fettig et al., 2013). Numerous monitoring projects have also been initiated. Current efforts include a Division of Forestry-led monitoring project in the Quartz Lake area near Delta Junction, being conducted with assistance from Fairbanks FHP staff. Stands in the Quartz Lake area were impacted by the 2012 windstorm and then again in late 2013 by a smaller scale windstorm.

Northern spruce engraver monitoring in the Quartz Lake area was initiated in 2014, with the installation of 15 monitoring sites distributed across a range of wind-damage severities; 21 sites were monitored in 2015. Based on data from previous monitoring efforts (N. Lisuzzo, pers. obs.), the 2014 Quartz Lake trapping data suggested that beetle populations were elevated in roughly half of the sites monitored[2]. The 2015 monitoring data is currently being finalized, but initial indications are that elevated northern spruce engraver populations are present in a majority of trap locations. Aerial observers also noted scattered northern spruce engraver-caused mortality in residual spruce in the Quartz Lake area during the 2015 aerial surveys. A more detailed description of this monitoring effort is available in FS-R10-FHP (2016).

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[2] Ideally, monitoring traps should be placed prior to the initial beetle flights each spring. Installation of the 2014 traps, however, appeared to coincide with the initial northern spruce engraver flight, presumably resulting in overall lower trap catches.

An overview of ongoing research: Arthropod abundance and diversity at Olive-sided Flycatcher nest sites in interior Alaska

by Adam Haberski[1], Megan McHugh[1], Julie Hagelin[2], Derek Sikes[1]

Introduction

The Olive-sided Flycatcher (*Contopus cooperi*) is an aerial insect specialist that nests in the boreal forest of Alaska each summer. Population declines over the last four decades in North America have elevated conservation concern for this species in many places, including Alaska (Sauer et al., 2014; Hagelin et al., 2015). In 2013, the Alaska Department of Fish and Game (ADF&G) began investigating the migratory habits and breeding biology of Olive-sided Flycatchers. In other populations, aerial arthropods are positively associated with foraging rate of adults and negatively associated with nest failure (Meehan and George, 2003). For some species of passerine, the abundance and diversity of arthropod prey are primary factors in habitat selection (Brown et al., 2011).

A previous study conducted by ADF&G in the 1990s (Wright, 1997) has provided us with the location of historical Olive-sided Flycatcher nests. These locations were resurveyed for three consecutive years (2013–2015) and found to be unoccupied. Comparing arthropod biomass and diversity at these historical sites with current breeding sites will allow us to correlate insect prey with bird occupancy. We hypothesize that (1) occupied sites will have higher arthropod biomass than historical sites and (2) occupied sites will have greater taxonomic diversity than historical sites. This study is amongst the first to document aerial insect communities in black spruce forests and has already produced new state records.

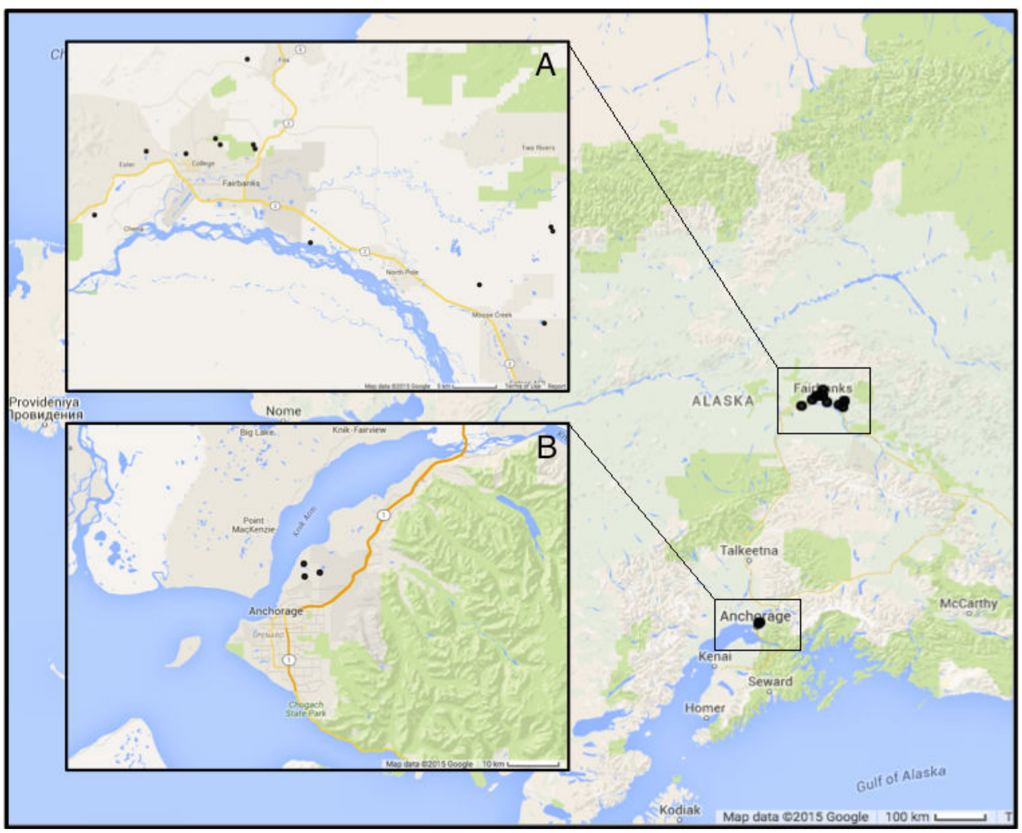

Figure 1: Collection sites of the 2013 sampling year in both Fairbanks (A) and Anchorage (B).

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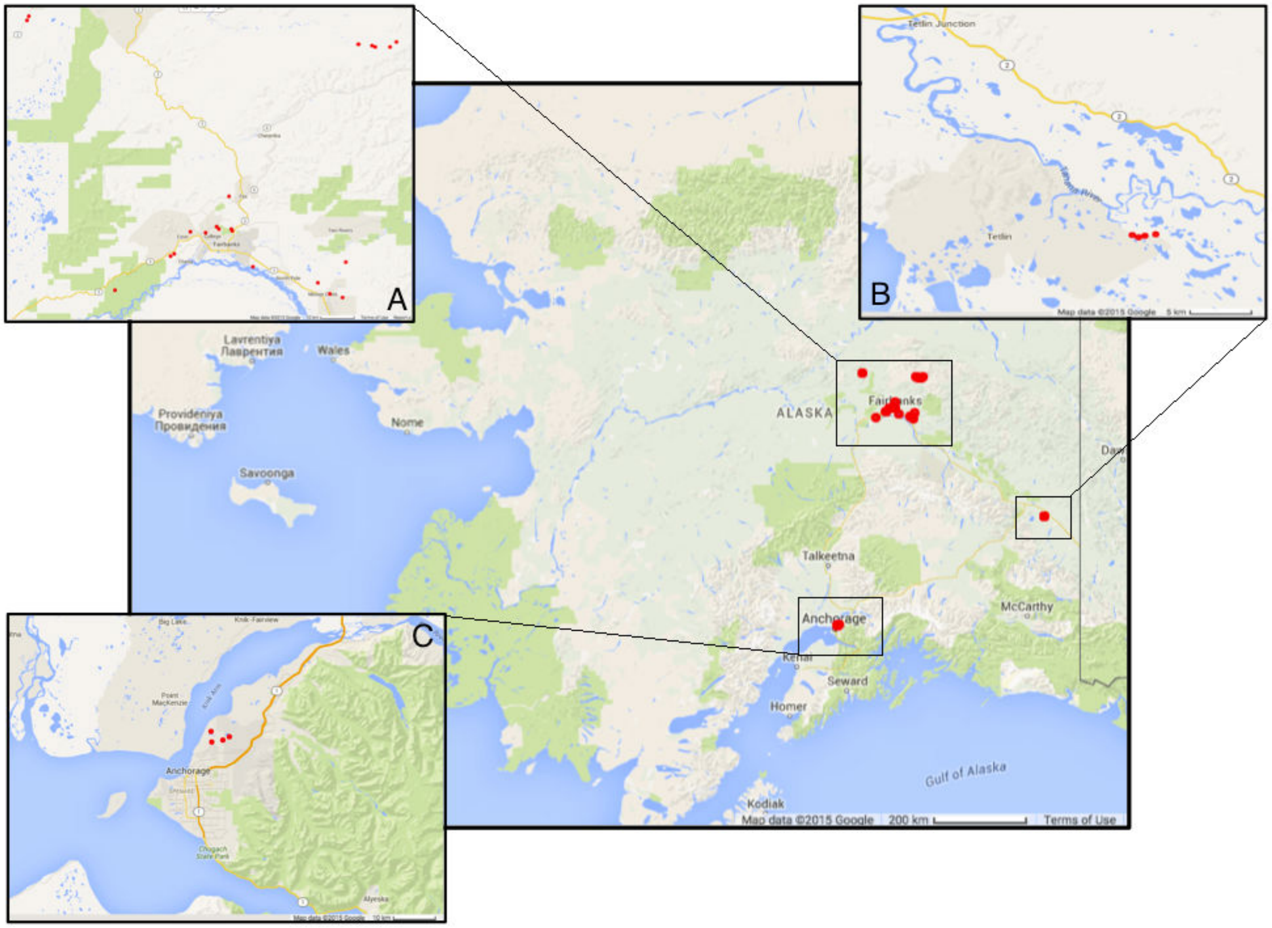

Figure 2: Collection sites of the 2014 sampling year in Fairbanks (A), Tetlin National Wildlife Refuge (B) and Anchorage (C). 2015 sampling locations were similarly located.

Study sites

Olive-sided Flycatchers nest predominantly on the edge of forest openings, including those caused by clear cutting or seasonal burns. They prefer to build nests on tall conifers with singing perches, to ensure their songs will carry (Altman and Sallabanks, 2012). In Alaska, nest sites occur in wet, boggy environments dominated by muskeg, spruce and birch.

Study sites were concentrated in three locations; the greater Fairbanks area, Joint Base Elmendorf-Richardson in Anchorage, and the Tetlin National Wildlife Refuge (Figure 1, 2). We studied 17 nest sites in 2013 (9 occupied and 8 historical); 31 sites in 2014 (19 occupied and 12 historical), and 32 sites in 2015 (20 occupied and 12 historical). Nest sites were located by ADF&G and were designated occupied or unoccupied following a series of bird song surveys during peak singing times.

Insect collection

Insect collection took place between May and July. Hanging Malaise traps were placed within 50m of known nest locations, approximately 5m from the ground. Hanging Malaise traps represent the upper story foraging level of Olive-sided Flycatchers. In 2014 and 2015, pollinator vane traps were deployed in addition to the hanging Malaise traps. Pollinator vane traps represent the near ground foraging level of flycatchers and attract insects, particularly *Bombus* spp., which are infrequently caught in hanging Malaise traps. Samples were collected every two weeks.

After collection, samples were sorted, databased into Arctos (the University of Alaska Museum's online database), and labeled for incorporation into the Insect Collection. These records are accessible via an Arctos project link[3].

[3] http://arctos.database.museum/project/olive-sided-flycatcher-contopus-cooperi-habitat-quality-study

Diversity calculations

Taxonomic order richness will be calculated by comparing the number of insect orders present at each collection site. The number of individual specimens per order was totaled for each sample site and these totals will be used to compare taxonomic order evenness between occupied and historical sites.

Biomass calculation

Specimens were measured from head to abdomen (excluding wing length) and biomass was determined using the calibrated formulae determined by Rogers et al. (1977). Formulae are provided for Araneae, Coleoptera, Lepidoptera, Hemiptera, Homoptera and Diptera. Other arthropods will be grouped within the aforementioned orders based on morphological similarity (Rogers et al., 1977). Although this may leave room for error, we feel it is important to include all specimens in the calculation. Arthropods smaller than 3mm were excluded based on the assumption that they are too small to be viable prey for Olive-sided Flycatchers. Biomass will be standardized by number of trap-days to correct for differences in trap deployment time. A comparison of total biomass will be made between occupied and historical sites for all years.

Moving forward

Insect collection is scheduled to continue during the summer of 2016, in order to test our hypotheses related to how insect patterns are associated with Olive-sided flycatcher breeding occupancy. Additional specimens will be collected in 2016 for stable isotope analysis. Stable isotope analysis of insects relative to bird blood will give us a better understanding of Olive-sided Flycatcher diet, which is not well understood, and potentially a limiting factor to successful reproduction. Isotope work will also reveal whether certain taxa of insects which are under-represented in our traps, such as grasshoppers and dragonflies, may constitute a significant portion of the flycatcher's diet.

Acknowledgements

We thank Chris Barger, Jayce Williamson, Ian MacDougal, Sarah Meierotto, Renee Nowicki, the ADF&G field technicians, and all of the University of Alaska Museum volunteers. Funding was provided through a State Wildlife Grant (SWG) administered through the Alaska Department of Fish and Game's Threatened, Endangered and Diversity program.

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Glocianus punctiger (Sahlberg, 1835) (Coleoptera: Curculionidae) common in Soldotna

by Matt Bowser[1]

Figure 1: Larvae of *Glocianus punctiger* in inflorescence of *T. officinale*, June 10, 2015 (http://www.inaturalist.org/observations/1620773).

Glocianus punctiger (Sahlberg, 1835), an exotic weevil of Palearctic origin, has been known in Alaska from two specimens in the University of Alaska Museum Entomology collection: UAM:Ento:24180 from Fairbanks and UAM:Ento:113229 from Anchorage. These specimens, identified by C. W. O'Brien, are the basis of the Alaska record of *G. punctiger* in Bousquet et al. (2013). Larvae of *G. punctiger* feed on floral and seed tissues of common dandelions (*Taraxacum officinale* F.H. Wigg.), also exotic to Alaska.

On June 10–11, 2015, I observed larvae in inflorescences of *T. officinale* at two localities in Soldotna. The larvae were quite common in inflorescences when seeds were maturing, after the petals had fallen and before the inflorescences reopened to release the wind-dispersed seeds. From the back lawn of the Kenai National Wildlife Refuge's headquarters building (60.465°N, 151.073°W) on June 11 I collected a handful of inflorescences at the seed maturation stage and placed them in a jar of water, arranging them so that the flowers were positioned over a plate. The larvae had dropped out of the flowers onto the plate by June 15. I placed the larvae in a jar of soil, where they quickly burrowed into the substrate. Twelve adults had eclosed between July 2 and July 6. These specimens now reside in the Kenai National Wildlife Refuge's entomology collection (KNWR:Ento:10799–KNWR:Ento:10810).

Figure 2: *Glocianus punctiger* specimen KNWR:Ento:10806, lateral view.

It is unlikely that the presence of *G. punctiger* in Alaska will meaningfully reduce the spread and persistence of common dandelions. The abundance of *G. punctiger* tends to be lower at high latitudes than at lower latitudes (Verhoeven and Biere, 2013). Even where *G. punctiger* is more abundant, the larvae consume only a small proportion of the plants' total seed production (McAvoy et al., 1983; Honek and Martinkova, 2005). Adults also consume dandelion foliage, but they cause little damage (McAvoy et al., 1983). With exceptions of the aphid *Myzus persicae* (Sulzer, 1776) and the slug *Deroceras reticulatum* (O. F. Müller, 1774), most of the other known herbivores of *T. officinale* listed by Stewart-Wade et al. (2002) appear to be absent from Alaska.

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I wonder if *G. punctiger* is breeding in any of Alaska's native dandelion species. In particular, I would like to know if horned dandelions (*Taraxacum ceratophorum* (Ledeb.) DC.) are affected. This species has seldom been collected on the lowlands of the western Kenai Peninsula. I know of only three records: a dot on the map in the Niskiski area in Hultén (1968), one specimen in the Kenai National Wildlife Refuge's herbarium from Skilak Lake collected in 1951 (KNWR:Herb:573), and a specimen held by the Pratt Museum in Homer with label data as follows:

> **Locality** AK; Kenai Borough; Ninilchik; 1200 Rd., 18 mile
> **Date** 7/15/96
> **Habitat** area above and adjacent to muskeg, abundant microrelief
> **Associated Species** *Cladina stellaris*, *Vaccinium caespitosum*, *Pleurozium schreberi*
> **Collected By** Chris Reidy

If *T. ceratophorum* should be a species of conservation concern here, then it seems likely that genetic assimilation by *T. officinale* (see Brock, 2004) would be more of a threat to this species than seed consumption by *G. punctiger*. Still, I would like to know if *G. punctiger* feeds on *T. ceratophorum* or any of Alaska's other *Taraxacum* species.

I thank Savanna Bradley of the Pratt Museum for locating and photographing the *T. ceratophorum* specimen. Derek Sikes (University of Alaska Museum) and Matt Carlson (Alaska Center for Conservation Science) provided helpful comments on this note.

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Figure 3: Larva of *G. punctiger* in inflorescence of *T. officinale*, June 11, 2015 (`http://www.inaturalist.org/observations/1620902`).

Review of the ninth annual meeting

by Logan Mullen

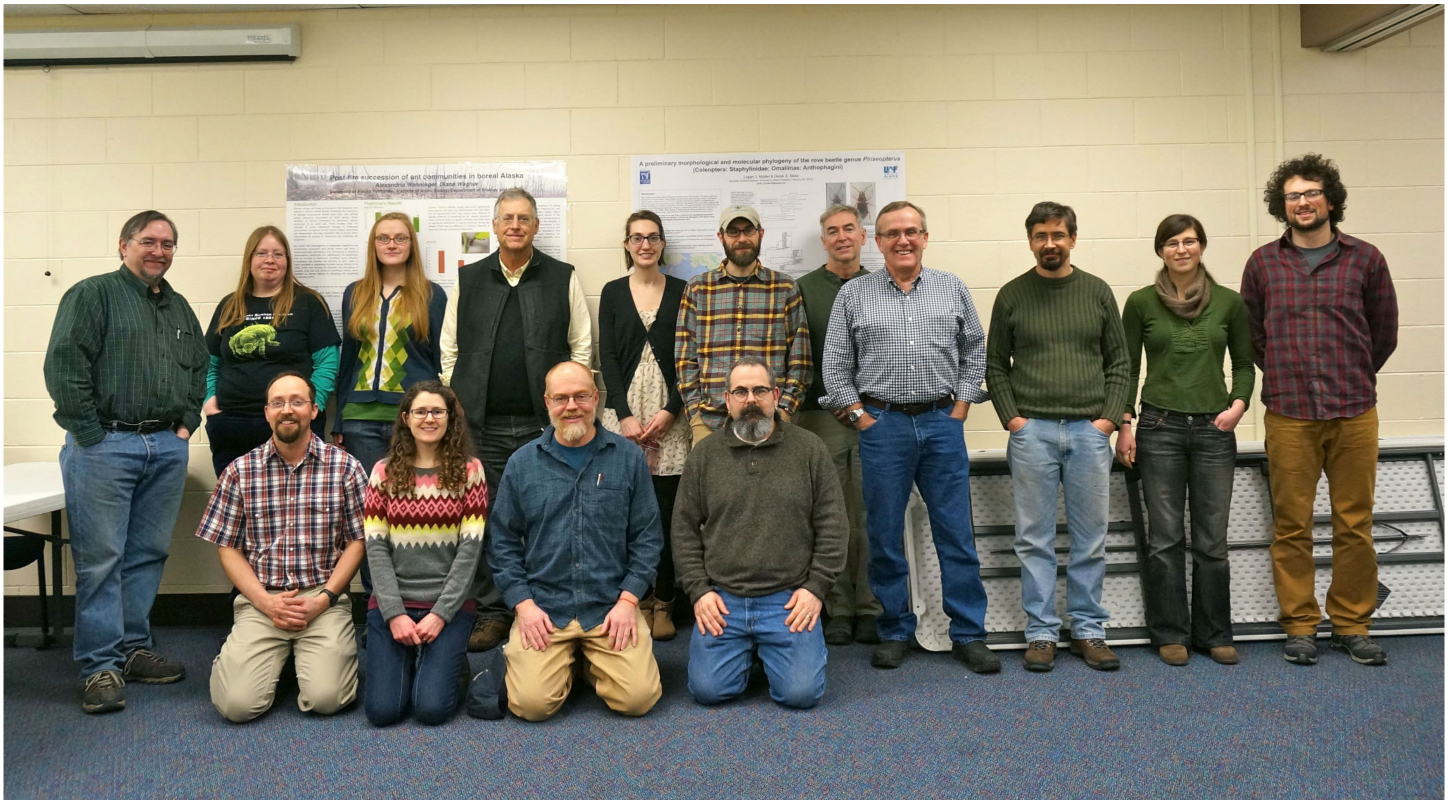

Figure 1: Members present at the end of the meeting. Back row, from left: Michael Baldwin, Robin Andrews, Kathryn Daly, John Lundquist, Alexandria Wenninger, Jason Moan, Dan Bogan, Roger Burnside, Derek Sikes, Molly McDermott, and Logan Mullen. Front row, from left: Matt Bowser, Jessie Moan, Steve Swenson, and Garret Dubois.

The ninth annual meeting of the Alaska Entomological Society was held at the Anchorage Cooperative Extension Service office on January 30th, 2016. We are very grateful to Jessie Moan for offering to host the meeting.

Presentations

We had many excellent presentations this year. In the morning, Jacque Shade presented on "CAPS program in Alaska", followed by Matt Bowser's "Inventorying arthropods on Tetlin National Wildlife Refuge by next generation sequencing" and "Building a DNA barcode library of Alaskan nonmarine arthropods" by Derek Sikes.

The student talks consisted of two presentations: "Response of arthropod communities to shrub expansion in Western Alaska" by Molly McDermott, and "An update on the Kenelm W. Phillip Lepidoptera collection at the University of Alaska Museum" by Kathryn Daly. Both student talks were excellent, making the job of the student award committee particularly challenging, and Molly McDermott was selected as this year's Student Presentation Award recipient. Congratulations Molly!

In a first for society, a pest control expert, Ken Perry of Paratex Pied Piper Pest Control presented "What is pestering Alaska?", providing intriguing anecdotes and insights into the world of pest control in Alaska. Wrapping up the presentations, John Lundquist presented "Highlights from the FHP program in Alaska" and Robin Andrews spoke on "Soil mites of interior Alaska".

Posters

Alexandria Wenninger and myself presented research posters this year, the former presenting a poster entitled "Post-fire succession of ant communities in boreal Alaska", and myself presenting "A preliminary morphological and molecular phylogeny of the rove beetle genus *Phlaeopterus* (Coleoptera: Staphylinidae: Omaliinae: Anthophagini)."

Business items—highlights

- Butterflies of Alaska, published by the society, was printed in December 2015, has sold out of its first printing as of January 2016! The second edition should be available soon.
- The Alaska Entomological Society will begin to maintain a list of "species of interest" to raise awareness of species of particular value (rare species, invasive species, species of commercial or agricultural importance, etc.) for collection or observation in Alaska. The list will be maintained on the society's website.
- Election Results: Logan Mullen (president), Alexandria Wenninger (vice president), Jill Stockbridge (secretary), Roger Burnside (treasurer)
- Next year the student presentation award will include a category for posters as well as for oral presentations

The minutes from the business meeting are available on our website.

Presentation and poster abstracts

Soil Mites of Interior Alaska *Robin Andrews*
This photo-rich presentation introduces common soil mites present in the greater Fairbanks region of Interior Alaska. Collembola, Protura, and small myriapods, spiders, and insects are covered briefly to provide perspective on the greater soil arthropod community. The role of worms and protists in the soil environment and concerns about invasive worms are briefly mentioned. An overview of basic Acari taxonomy is given. Mesostigmata, Endeostigmata, Prostigmata, and Oribatida groups are introduced. Some very basic techniques used to determine family level taxonomy are presented. Problems of Acari preservation and identification are discussed.

Forest Health Protection in South Central Alaska—2015 Highlights *J. E. Lundquist, S. Swenson, and G. Dubois*
Southcentral Alaska is situated at an intersection of the Boreal and Maritime forests. It is an area rich in topography, vegetation, climate, native and non-native cultures, the ecosystem services provided by its forested areas, and the 'community-think' expressed by people living in the various villages and towns within this region. Because it is the most populated region in Alaska, its forests are more accessible than elsewhere in the state. This region has a deep history of insect infestations, some of the most notably include: the spruce beetle outbreak on the Kenai Peninsula during the 1990s, the amber-marked birch leaf miner infestations of birch during the early 2000s, and the widespread geometrid moth and leaf roller defoliations in succession during the early and mid-2010s. Notable occurrences during 2015 were the expanded range of spruce aphid to Homer and the reemergence of the Amber-marked birch leaf miner on the outskirts of Anchorage. Apart from monitoring annual insect pest conditions, South Central FHP entomology activities during 2015 included inspecting for insect pests and diseases the tree chosen to be the Capitol Christmas Tree before it travelled to Washington DC, contributing to the Climate Change Vulnerability Committee as part of the Chugach National Forest Plan revision effort, distributing and managing approximately $300,000 of the Western Bark Beetle Initiative grants, composing a forest health kiosk at the Alaska Botanical Gardens, establishing of a Forest Health Treatment Area on the Kenai Peninsula, conducting various field visits at the requests of stakeholders, and collaborating in a variety of forest health oriented projects with colleagues from the PNW Research Station and other USFS research stations, various universities and Alaskan tribal entities.

Response of arthropod communities to shrub expansion in Western Alaska *Molly McDermott*
Shrub thickets have increased in coverage across the Arctic in recent decades as temperatures have risen and permafrost has thawed. This is affecting the distribution, abundance and phenology of arctic arthropods. Arthropods are sensitive indicators of temperature change, provide a variety of ecosystem services, and comprise the majority of biodiversity in arctic environments. Previous research in Arctic Alaska has found that overall arthropod abundance increases with shrub dominance and is strongly predicted by NDVI and snowmelt timing, however, taxon-specific responses to shrub dominance and changes in phenology remain understudied components of arctic ecology.

My research focuses on testing the hypotheses that 1) the timing of food availability is earlier in tundra habitats due to earlier snowmelt and 2) plant height heterogeneity increases arthropod abundance across a gradient of habitats in the boreal-Arctic transition zone of northwestern Alaska. I use a combination of pitfall traps and sweep net sampling in shrub and tundra habitats, focusing on arthropods used by migratory songbirds. Here I present findings indicating that arthropod biodiversity and abundance increase with shrub height, but important songbird prey groups are most abundant between shrub thickets and open tundra, supporting the second hypothesis. Understanding the ecological drivers of spatial and temporal patterns of arthropod abundance in these rapidly shifting habitats will help scientists model how Arctic ecosystems may respond to climate-mediated changes.

A preliminary morphological and molecular phylogeny of the rove beetle genus *Phlaeopterus* (Coleoptera: Staphylinidae: Omaliinae: Anthophagini) *Logan Mullen*
The omaliine rove beetle genus *Phlaeopterus* Motschulsky 1853 contains 14 species, which are known from the northwestern United States, western provinces of Canada, and Alaska. These beetles are largely confined to the edges of alpine snowfields and streams, habitats that are particularly sensitive to the impacts of climate change. Here, I present preliminary Bayesian and maximum likelihood phylogenetic analyses of the genus *Phlaeopterus* using morphology and molecular data. Species hypothesis of Campbell (unpublished), Hatch (1957), Fauvel (1878), and Motschulsky (1853) are tested for the first time with modern phylogenetic methods. My analyses support the addition of multiple undescribed species to the genus *Phlaeopterus*, as well as the synonymizations of several current species.

Post-fire succession of ant communities in boreal Alaska
Alexandria Wenninger and Diane Wagner
Research suggests that climate warming will cause an increase in fire frequency and severity in Alaskan boreal forests, increasing the proportion of younger successional forests over time, and shifting forests previously dominated by black spruce to forests dominated by deciduous species. These changes in post-fire succession have the potential to cause widespread changes in arthropod communities throughout boreal interior Alaska. We predict that heterogeneity in understory vegetation and microclimate associated with young forests will foster a diverse prey base, promoting a rich community of predatory Hymenoptera, specifically ants (Formicidae). Additionally, we hypothesize that an increase in deciduous trembling aspen (Populus tremuloides) will increase the diversity of ants; aspen is highly palatable in comparison to black spruce, which may increase the diversity and abundance of available prey, and also produces extrafloral nectar, which mediates an indirect defense by attracting and nourishing ants. The objectives of this project are 1) to characterize changes in the boreal Hymenoptera community during post-fire succession and 2) to test the hypothesis that aspen fosters higher abundance and diversity of ants than black spruce. Ants are sampled across successional time (young, intermediate, and mature aged post-fire regenerating forests) and between two successional trajectories (shift to aspen and return as black spruce). Field collection of ants includes pitfall trapping and sweep netting. The data are used to characterize and compare the Hymenoptera communities among successional ages and trajectories. Preliminary results suggest that ants are more abundant and speciose in young successional forests compared to intermediate aged forests, regardless of successional trajectory. The ant communities of young successional forests are also compositionally dissimilar to the intermediate aged forests. This work will contribute to understanding how climate change will impact boreal insect communities.

Upcoming Events

Klondike Gold Rush National Historical Park BioBlitz, May 13, 2016

Klondike Gold Rush National Historical Park is hosting an aquatic and terrestrial invertebrate BioBlitz on May 13 to engage the public in science discovery and data collection. Anyone is welcome to participate in this fun, hands on event. For more information, contact Jami Belt at jami_belt@nps.gov.

Goals Contribute to science of biodiversity, inform KLGO about a group of organisms that is little-known, provide baseline indexes of organisms for long-term monitoring, and engage the community in science discovery.

Focus taxon aquatic and terrestrial invertebrates. Scope: 1-day sampling event on May 13. Pre-visit classroom sessions are available for participating schools.

Sites Dyea, Skagway, Chilkoot Trail, possibly also White Pass if it can be accessed safely.

Target participation and audience Approx 60–100 total including approximately 40–60 Middle and High-school students from Skagway and Klukwan.

Figure 1: Klondike Gold Rush National Historical Park.

Anaktuvuk Bioblitz, May 20–22, 2016

As part of the National Park Services (NPS) centennial celebration during 2016, Gates of the Arctic National Park will be participating in a National Geographic and NPS sponsored BioBlitz effort this summer.

Over 110 National Parks across the country are participating in the centennial BioBlitz, including four parks in Alaska (Gates of the Arctic, Bering Land Bridge, Denali, and Klondike Gold Rush). The Gates of the Arctic event, which will take place in the village of Anaktuvuk Pass during May 20-22, has the distinction of serving as the Arctic Regional Showcase Bioblitz, and will receive special attention in Washington, D.C. for contributing its unique collection of Arctic faunal diversity to the national species inventory.

Figure 2: Graphic from Anaktuvuk Bioblitz postcard.

Scientists from the NPS and the University of Alaska Fairbanks are coordinating with the Simon Paneak Memorial Museum to lead Nunamiut School students as they identify and learn about a wide range of taxa in the Anaktuvuk Pass area within Gates of the Arctic National Park. Investigations will include camera trap, track and scat surveys to detect large mammals such as ungulates and carnivores, a live-trapping effort to capture small mammals such as rodents and shrews, point counts to detect birds, lake and stream surveys in search of fish and frogs, sampling to detect both aquatic and terrestrial invertebrates, and DNA barcoding to identify soil microbes. Participants will also learn traditional Inupiaq names, trapping techniques, and uses for many taxa from local subsistence experts.

Photos, locations and identities of taxa detected during this three-day event will be uploaded via the iNaturalist smartphone app and automatically added to the National BioBlitz species list. These georeferenced records will then be archived in the Global Biodiversity Information Facility (GBIF) where they will contribute to a growing international database of faunal records that are freely available for download and use in subsequent research. Additionally, all activities will be documented by professional photographers and student videographers as part of the larger national BioBlitz celebration.

Anyone interested in volunteering or participating is encouraged to contact Andy Baltensperger at 907-455-0623 or andrew_baltensperger@nps.gov.

Serpentine Hot Springs Bioblitz, July 7, 2016

As part of the National Park Services (NPS) centennial celebration in 2016, Bering Land Bridge National Preserve will participate in a National Geographic and NPS sponsored BioBlitz effort this summer. The Bering Land Bridge BioBlitz event will take place from July 7–10 at Serpentine Hot Springs. NPS scientists and staff will lead students from the surrounding communities on a multiday science fieldtrip to inventory, sample, and observe a wide range of taxa surrounding Serpentine Hot Springs. Investigations include samples of aquatic and terrestrial invertebrates, including butterflies and dragonflies, observation of beaver ecology and its influence on passerines, waterfowl, water quality, and fish. All inventoried species will be documented on the iNaturalist application which will add to a growing international database of plant and animal species. Additionally, this event will be part of the nationwide Dragonfly Mercury Project (DMP), a community-driven endeavor that measures mercury levels in dragonfly larva as an indicator of overall ecosystem health.

This much anticipated event will allow parks to grow their natural resource knowledge base while making science fun for youth in the surrounding villages. If you are interested in learning more about the Bering Land Bridge Bioblitz please contact Guadalupe Zaragoza at Guadalupe_Zaragoza@nps.gov / 907-443-6120 or Kathleen Cullen at Kathleen_Cullen@nps.gov / 907-443-6127.

Figure 3: Serpentine Hot Springs.

Tenth Annual Meeting, January 2017

The tenth annual meeting of the Alaska Entomological Society will take place in Fairbanks in January 2017. Check for updates on our events page as the meeting date approaches.