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In this issue:

Ocellated Emerald dragonfly (*Somotochlora minor*) new to Alaska

by John Hudson[1]

Figure 1: A male Ocellated Emerald at Auke Lake in Juneau. Photograph by John Hudson.

Alaska's dragonfly list grew by one last year with the discovery of a single female Ocellated Emerald (Figure 1) in the Kanuti National Wildlife Refuge. The specimen was netted at a *Sphagnum* bog in the heart of the refuge during a 3-day collecting trip targeting the genus. Since efforts to document the refuge's odonate fauna began in 2004, 25 species have been documented at Kanuti making it one of the top areas for dragonfly diversity in the state. Remarkably, the following month a population of Ocellated Emeralds was discovered at Juneau's Auke Lake, an area that has been intensively surveyed since 1997.

Emeralds (Corduliidae) comprise the most species rich family of dragonflies in Alaska. The common name refers to the brilliant emerald-green eyes and metallic green thorax. Nine species are currently known from the state. They are locally rare, secretive, and difficult to capture. The two species of dragonflies that likely occur here, but have yet to be collected are emeralds: Brush-tipped Emerald (*S. walshii*) and Muskeg Emerald (*S. septentrionalis*).

The Kanuti survey was funded by a Challenge Cost Share grant from the U.S. Fish and Wildlife Service. This year the same grant will fund dragonfly surveys in Kanuti and the Selawick National Wildlife Refuges. To date 35 dragonfly species have been found in Alaska.

[1]Alaska Odonata Survey, Juneau. jhudson@gci.net

European Alder Sawfly Introduced to Alaska

by James J. Kruse[2]

Figure 1: Adult *M. pulveratum*. Photograph by Ken Zogas.

The green alder sawfly, *Monsoma pulveratum* (Figures 1-2), was collected and reared in Alaska by USDA Forest Service Region 10 (R10) Forest Health Protection (FHP) personnel and positively identified by Dr. David Smith at the Systematic Entomology Laboratory. This sawfly is native to Europe, northern Africa, and Turkey to the Caucasus, and this detection in Alaska represents a new U.S. record. Other North American records are from Newfoundland, Canada in 1991 and 1998 with no records in between. This would indicate separate introductions. *M. pulveratum*'s principal larval host in Alaska is Thin-Leaf Alder (*Alnus tenuifolia*), which is found in riparian areas throughout south-central and interior Alaska, joining *Hemichroa crocea* and *Eriocampa ovata* as the major sources of alder defoliation in Alaska. It also joins a growing number of introduced sawflies in Alaska on alder and birch.

In Alaska, *M. pulveratum* is now known from the Kenai Peninsula Borough, Anchorage, Matanuska-Susitna Borough and Fairbanks. It was initially detected in 2004 in an agricultural trapping operation but not reported. By 2007, significant defoliation was recorded in the Palmer Hay Flats where it was not noticed at all in 2006 during the course of an alder insect pest project. By 2008, significant defoliation was noted on the Kenai Peninsula. Defoliation was evident on the Susitna and Little Susitna Rivers (Figure 3), and the first capture in Interior Alaska was made in 2009.

Figure 2: Larva of *M. pulveratum*. Photograph by Ken Zogas.

Figure 3: Defoliation of Thin-Leaf Alder by *M. pulveratum* in the Susitna drainage, July 2009. Photograph by Ken Zogas.

Due to the significance of Thin-Leaf Alder to Alaska salmon spawning stream ecology, R10 FHP is working to energize University and fisheries to research into the potential effects of this insect. Similarly, R10 FHP received Evaluation & Monitoring funds to pursue basic life history studies of the sawfly and is effects on Alaska alder species. USDA APHIS formed a Pest Advisory Group for analysis and recommendations. Currently the mode of introduction is hy-

[2]USDA Forest Service, State & Private Forestry, Forest Health Protection, Fairbanks Unit. jkruse@fs.fed.us.

pothesized to be direct shipping from Europe or eastern Canada to Anchorage. More than 125 ships per year travel to Anchorage via Arctic Ocean shipping routes, a number that will only increase with the increased access and use of the Arctic Ocean in a warming climate.

Safely applying insecticides directly to tree xylem

by Mark E. Schultz[3]

Undoubtedly many of you are familiar with the broadcast pesticide treatment of insects to stop insect outbreaks for agriculture crops and forest trees. There sometimes were environmental consequences to these treatments. One was that beneficial insects or insect predators and parasitoids could be adversely impacted enough that when insect pests recolonized a chemically treated area their population grew much faster. More specific alternatives to broad-spectrum pesticides were developed alongside of more focused Integrated Pest Management (IPM) treatment methodologies. One of these is individual treatment of trees by chemical injections.

Tree injection works similarly to hydraulic pumps by forcing pesticides directly into the xylem of trees with the objective of getting protective treatments to leaves and phloem tissue. The injection process utilizes the plumbing of the tree as well as evapotranspiration of leaves to create upward movement of liquids in vessels (angiosperms) and tracheids (gymnosperms). Early injection systems tended to use too much pressure and higher quantities of liquid and, in some cases, caused tree injury (Perry et al., 1991), but contemporary systems use very little and rather non toxic (to the tree), but concentrated, pesticides with lower injection pressures. New injection systems such as Arborjet's QUIK jet system can deliver a small but effective amount of chemical to small injection sites that are sealed and protected with rubber plugs (Figure 1, `http://www.arborjet.com/products/arborplug.htm`)

A relatively new technique of placing powdered pesticides directly, via a cap system, into xylem and using only the transpiration stream of the tree to deliver the pesticide is the strategy of the Acecap system (Figure 2.B, `http://www.treecareproducts.com`)

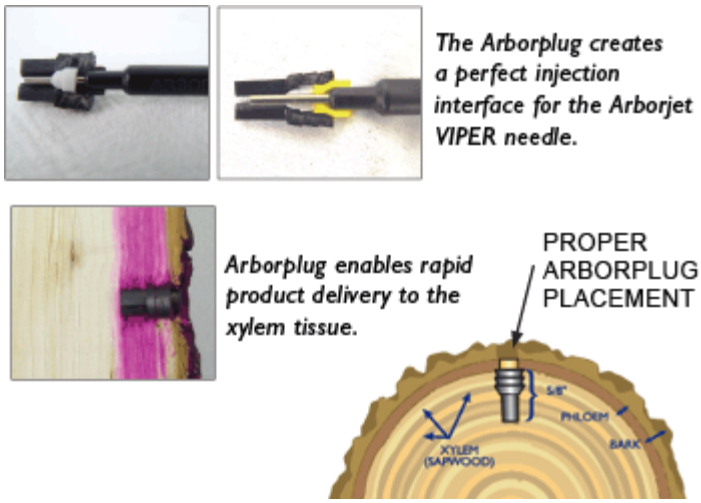

Figure 1: Arborjet injection system (red dye shows the active xylem).

Figure 2: Acecap injection system.

Reference

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[3]USDA Forest Service, Alaska Region, State and Private Forestry, Forest Health Protection. `mschultz01@fs.fed.us`

Bark Beetle Technical Working Group studies on the Northern Spruce Engraver

by Roger Burnside[4], *Chris Fettig*[5], *Chris Hayes*[6], *James J. Kruse*[2], *and Mark Schultz*[3]

Antiaggregation study (2009-2010)

The objective of this study is to determine the effectiveness of verbenone and conophthorin for protecting white spruce slash from colonization by northern spruce engraver (*I. perturbatus*).

The study site is a recently harvested white spruce/quaking aspen stand that is serving as a fire break and logging deck area for biomass that will power a generator at Tok School, Tok, Alaska (63.32°N, 142.98°W; about 515 m elevation). On 18-19 May 2009, 20 slash decks consisting of 15, 4.5 ft. long bolts with large end diameters of 4.0-8.0 in., were cut from freshly felled white spruce near Tok. Half of the piles (n = 10) were treated with two semiochemicals (verbenone and conophthorin). The other piles were left untreated. Attacks and exit holes were recorded 11-13 July and 26-29 August 2009, respectively (Figure 1).

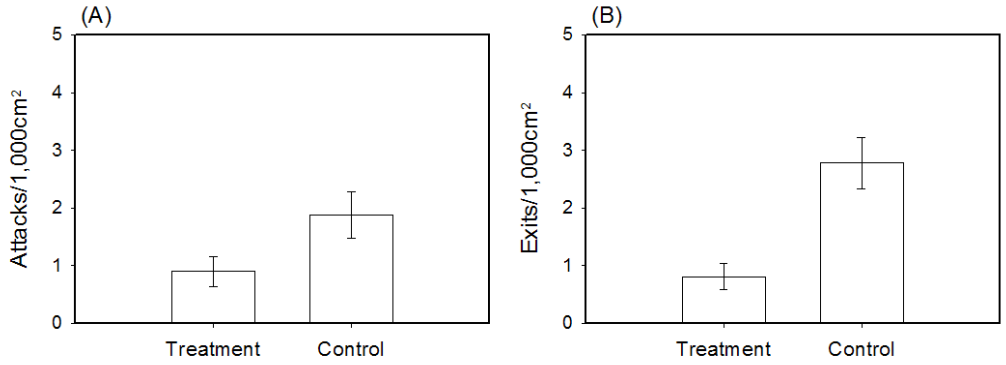

Figure 1: Comparisons of (A) attack and (B) emergence-hole densities between semiochemical treated (verbenone and conophthorin) and untreated bolts. Bars represent mean ± standard error of the mean.

Colonization of slash and residual trees (2009-2011)

A study to assess the effects of various slash management treatments (variables include cutting time, slash arrangement and scoring) on *I. perturbatus* colonization and reproductive success in white spruce is being established in three blocks in interior Alaska (near Fairbanks, Delta Junction, and Tok). Trees were felled in August and September 2009 and will also be felled in May 2010 to complete all treatments. The study design is a randomized complete block with 0.25 acre plots. Dependent variables will include attack and exit hole densities, in addition to mortality of residual leave trees.

[4]State of Alaska, Department of Natural Resources, Division of Forestry, Forest Health Protection Program. roger.burnside@alaska.gov
[5]USDA Forest Service, Pacific Southwest Research Station, Sierra Nevada Research Center. cfettig@fs.fed.us
[6]USDA Forest Service, Pacific Southwest Research Station, Sierra Nevada Research Center. cjhayes@fs.fed.us

Long days, rain, and spiders: a quick survey on Prince of Wales Island

by Joey Slowik[7]

Figure 1: Brown Mountain, Ketchikan.

The trip started out well enough in usual Southeast Alaskan fashion: pouring rain. Last summer was looking promising, with a very hot dry summer occurring, so I really thought my chances were good for some sun. However, upon my arrival it changed from August to more like October, a constant downpour blowing sideways. I've yet to travel through Ketchikan when it hasn't poured. I only had a few days in Ketchikan so I tried to make the best of it, but the fact is there just aren't that many critters out when it's raining. I stuck to the trees, which if anyone is familiar with Ketchikan roads, tend to be loaded with devil's club as well as bones and carcasses from all the hunters dumping their refuse, another slightly disgusting fact of the area discovered one night while headlamping. I had just three days and two nights to collect. I threw out pit fall traps and headlamped the best I could. I even built a beat sheet out of Tyvek just for the occasion, which I might add worked great even in the rain, although it did sound like a drum. After being soaked for three days and having little to show for it I was looking forward to getting over to Prince of Wales and a hot shower.

I hopped the ferry and met up with a rental car agency on the island. I have to plug this guy, Darren, at Hollis Adventure Rentals because as far as renting on the island goes, he *is* the outfit. I was able to get an older Subaru wagon, "Lulu", with the ability to travel anywhere I could. The other firms all required extra deposits and rental of a truck for that privilege, which would have been much more costly, and used a lot more gas. Also the wagon provided great space for drying things out and even came with a Spot locator in case of an emergency. I met up with Mark Shultz and his seasonal Chris Scott (who oddly enough is a classical music composer in his free time—seasonals always seem to have interesting other lives) of the Forest Service who made arrangements for me to use the Hollis bunkhouse as a staging ground. I wanted to get to as much of the island as I could in the 11 days I had left so my plan was to go north to Memorial Beach, setting pitfall traps and collecting along the way, then head to Polk Inlet to do the same. I laid out the route so the pitfalls would have time to soak while I was running off to some other part of the island. I was also able to finagle a trip with a couple of seasonals (Becky Roberts and Dylan Linet from Thorne Bay Ranger District) to Pete's Moss Cave and Windgate Cave, and a day on Sumez Island with Mark and Chris thanks to Tim Paul of Craig Ranger District. I laid out a very full schedule, even working a couple 20 hour days by the time the headlamping and labelling was done. Thank God for Dead Man's Reach. Also, the weather settled down to more familiar August Southeast weather; i.e. a few clear days, some drizzle, but workable.

On this trip I made the extra effort to collect following genetic preservation practices. This included using 95% propylene glycol in the pitfall traps, and using 100% ethanol for washing and preservation. I also bought a small electric cooler which plugged into the cigarette lighter to keep the samples as cool as possible until I could get them into the freezer, which also worked great for keeping my milk and beer cold. From an arachnologist's position the only drag of collecting for genetics is that when it comes to identifying the samples they are often brittle, causing legs to pop off unintentionally.

I've been on many surveys and collecting trips before, and there is often a period of excitement with every new find, this is usually followed by a long period of redundancy. You begin to find the same animals over and over again and you get the feeling that perhaps you've done enough collecting and there is nothing new to find. Oddly enough, I've found knowing a bit about the fauna beforehand really helps eliminate the redundancy. I've collected a lot in Southeast and I've become aware of the little habitat differences which produce different species. I'm also aware, because of our lack of natural history knowledge on most spider species, that many species appear rare because we don't know where to look for them; of course some may actually be rare as well.

[7]University of Alaska Museum. slowspider@gmail.com

Figure 2: *Antrodiaetus pacificus* burrow.

Figure 3: Some awesome Prince of Wales Island second growth.

In the 14 days of the trip I was able to collect 2,582 invertebrate specimens, of which 1,623 were spiders. Some of the interesting finds include the first male of the species *Pacifiphantes magnificus* (Chamberlin & Ivie) which, thanks to this one male, provides evidence that it is incorrectly placed in *Pacifiphantes* and may even warrant a new genus. I was also able to find a good population of *Antrodiaetus pacificus* (Simon) in the Polk Inlet area. This is Alaska's only mygalomorph species. Mygalomorphs don't have very good dispersal capabilities. Furthermore, this species lives in burrows in rotten logs (Figure 2), often with sequential generations only moving far enough away to make a new burrow. So the question was, how could they have gotten to the island? Only finding *A. pacificus* in the Polk area and around Thorne Bay hints that they may have been introduced with the logging operations. The genetics of the population could be interesting if this is the case. The species also has been reported from Ketchikan but I was unable to find any. I suppose they don't care for the rain.

The effort to preserve specimens for genetics is already paying off. Steve Lew at the University of California Berkley had been pestering me for years for some *Cybaeopsis wabritaska* (Leech) and this is the first time I've been able to produce usable specimens for his analysis. Also Gerry Blagoev of the Biodiversity Institute of Ontario has agreed to do some DNA barcoding of some species to help build up the BOLD database (`http://www.boldsystems.org`). This collection also builds on the ever growing research collection at the University of Alaska Museum curated by Derek Sikes. Currently we list 558 spider species in the state but only have comparison specimens for 304 of those species. With a state as large and diverse as Alaska it will be some time before the holes are filled, but trips like this help.

Figure 4: *Hyptiotes gertschi* Chamberlin & Ivie.

Assessing larch mortality and regeneration after a landscape level sawfly outbreak in interior Alaska

Forest Health Monitoring Evaluation Monitoring grant #WC-EM-08-03

by Roger Burnside[4] *and Mark Schultz*[3]

The larch sawfly (LSF, *Pristiphora erichsonii*), is an invasive defoliator of larch (*Larix laricina*) in Alaska. Based on aerial survey data, it is estimated that 600,000-700,000 acres of larch forest in interior Alaska have been impacted by a LSF outbreak that began in 1999 and continues to a lesser degree to the present time. The mortality of larch affected by the LSF has been documented, largely from aerial surveys, to reach 80%. However, due to the majority of interior Alaska larch stands being inaccessible (unroaded) and the difficulty of obtaining accurate assessments of associated mortality agents (e.g., larch beetle, LSF, *Armillaria* root disease, etc.), concerns have been expressed that many stands are not regenerating and that factors unrelated to insect agents are the primary forces affecting larch stand succession, regeneration success and larch sustainability.

An evaluation project was previously conducted (2006 & 2007) to resolve mapping inaccuracies that estimated the range of larch distribution in interior Alaska. In addition, it also assessed the extent of healthy larch within the area of heaviest mortality, LSF caused or otherwise. The lack of any landscape level remote sensing data coverage for this area makes it difficult to easily delineate the extent, basal area or % composition of larch in stands that are mostly composed of black spruce (*Picea mariana*) in interior Alaska. Forest health risk assessments are continually challenged by the inability to cost effectively access stands for current and repeat data collection. The current project was initiated to better determine the primary source of larch mortality within LSF-impacted stands, to add to the limited body of information on stand-level larch dynamics, to establish permanent plot points for subsequent analysis/stand delineation, and to better assess larch regeneration. This information allows for more informed determinations of forest health risk factors associated with larch stand establishment after repeated insect and/or abiotic disturbance.

Between July-August of 2008 and 2009, ground plots were established along a series of transects and stand assessments were conducted at 14 sites across the mapped extent of larch in interior Alaska. Stand exams were conducted along the existing road system and at more remote areas accessible only by float-plane. Estimates of stand composition, tree and shrub regeneration, presence/absence of cone-bearing trees, stocking, basal area, stand age, and cause(s) of recent larch mortality were recorded. Conclusions were drawn as to relative site quality, actual mortality causal agent(s), future of larch succession and regeneration potential for the sites evaluated.

A poster about this project is available at `http://www.akentsoc.org/doc/Burnside_R_et_al_2010.pdf`.

A second Alaskan bristletail

by Matthew L. Bowser[8]

Until recently, *Petridiobius arcticus* (Folsom, 1902) was the only species of bristletail (Microcoryphia) known from Alaska. *P. arcticus* is widely distributed in southern coastal Alaska, inhabiting rocky places from the Aleutians to Southeast Alaska.

Schultz and De Santo (2006) collected bristletails of the genus *Pedetontus*, subgenus *Verhoeffilis* in tree bole intercept traps on Prince of Wales Island, the first record of this genus in Alaska. They were apparently the same species reported by Mendes (1992) and Sturm (2001) from British Columbia. Mendes (1992) noted that the only difference between his specimens and Silvestri's (1911) description of *Pedetontus submutans* (Silvestri, 1911) from Washington and Oregon was the length of the ovipositor. In *P. submutans*, the ovipositor extends abound 1 mm beyond the apices of the terminal spines of the stylets of the 9th abdominal segment; in Mendes' specimens, the ovipostor only reached the bases of these terminal spines. Mendes (1992) commented that this could have been merely a difference in development.

In order to resolve the identity of the Alaskan specimens, I sought topotypical specimens for comparison, borrowing bristletails from the following collections: Maurice T. James Entomological Collection, Washington State University, Pullman, Washington (WSU); the Burke Museum of Natural History and Culture, University of Washington, Seattle, Washington (UWBM); the Spencer Entomological Collection, Vancouver, British Columbia (UBCZ); the Univerity of Alaska Museum, Fairbanks, Alaska (UAM); and

[8]US Fish & Wildlife Service, Kenai National Wildlife Refuge, Soldotna, Alaska. `Matt_Bowser@fws.gov`

the Juneau USDA Forest Service collection, Juneau, Alaska (FSAJ).

In the loaned material were numerous specimens of *Pedetontus* subg.*Verhoeffilis* from Juneau, Alaska to Galliano Island, British Columbia, but none from Washington or Oregon. All of these specimens, including a female with an ovipositor extending slightly beyond the terminal spines of the 9th stylets, appeared to be the same species. Due to a lack of topotypes, I could not determine whether *Pedetontus* specimens from Alaska and British Columbia were conspecific with *P. submutans*.

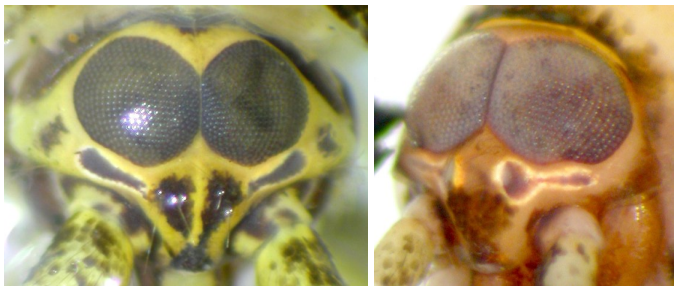

Figure 1: Frontal view of heads of *Petridiobius arcticus* (left) and *Pedetontus* subg.*Verhoeffilis* (right) from Sitka, Alaska (UAM).

Figure 2: *Pedetontus* subg. *Verhoeffilis* ♂ from Prince of Wales Island, Alaska (FSAJ). The caudal filaments are missing in this specimen.

Judging from label data, this *Pedetontus* is a forest species. They were found on tree boles, on the forest floor, in moist logs, in moss, and on trunks of Sitka spruce (*Picea sitchensis*), western hemlock (*Tsuga heterophylla*) and red alder (*Alnus rubra*).

The two Alaskan species can be separated by the generic characters of the lateral ocelli as in the key of Mendes (1990). In *Pedetontus*, the lateral ocelli are sole-shaped, extending well into the fronto-ocular area, sometimes nearly touching; the lateral ocelli of *Petridiobius* are constricted little if at all medially and are shorter, not extending onto the fronto-ocular area (Figure 1). The color and general appearance of the two species also differ. In preserved specimens, *P. arcticus* has more and darker integumental pigmentation than the Alaskan *Pedetontus*. The scales of *P. arcticus* are mostly dark gray (in alcohol), where the scales of the Alaskan *Pedetontus* are brown. The legs and maxillary palpi of *P. arcticus*, especially males, are also more robust than those of the Alaskan *Pedetontus*.

I thank Karen Needham (UBCZ), Richard S. Zack (WSU), Rod Crawford (UWBM), Derek Sikes (UAM), and Mark Schultz (FSAJ) for the loans of material.

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Rearing experiment to determine the willow host range of *Rabdophaga* spp. in Alaska

by Dominique M. Collet[9]

Introduction

Most of the species of the genus *Rabdophaga* (Diptera: Cecidomyiidae) induce the growth of galls on willows (*Salix* spp.) Few of the more than 5,451 species of the gall midges live up to their name by inducing the growth of galls on plants. Most of the other Cecidomyiids are fungivores or predators of soft-bodied insects such as aphids (Gagné, 2004).

Rabdophaga larvae, by feeding on specific sites on their host plants induce predictable distortion of the plant organ they target. Species of *Rabdophaga* can be sorted into discrete species groups based on the observation this distortion as well as other morphological and ethological characters shared among the members of each groups.

Within a species group, it may be difficult to find consistent morphological difference to separate the species. Some authors attribute a type of gall affecting a dozen or more willow species to a single *Rabdophaga* species while others will argue that several gall midges species are involved. Attempts to resolve the question solely by comparative morphological studies were not entirely convincing (Stelter, 1970). More definite results are obtained by controlled breeding experiments supported by morphological evidence and DNA sequencing (K. Harris and K. Yukawa, *personal communication*). Such a study of host range of *Rabdophaga rigidae* was done by isolating a willow and the breeding *Rabdophaga* species in the same enclosure (Smith et al., 1983). Similarly, Nematines *Pontania*, *Euura*, and *Phyllocolpa* (Tenthredinidae: Hymenoptera) willow host species were determined by providing mature female Nematines which had emerged from galls on identified willow hosts with selected willow species grown in confined pots (Kopelke, 1999). The following rearing trials were done with species of the *Rabdophaga rosaria* species group to determine the host range for each species.

Methods

In 2009, I used cages to isolate a few stems of identified willow growing in its natural habitat and introduced inside the cage a dozen or so galls of the previous year collected from identified willows of a known species. Adult gall midges are very fragile and cannot be handled directly without risking damage. Therefore, whole galls of the previous year, still containing the immature insects, were handled and introduced in the cages.

Not all the willow shrubs from the same willow species are galled to the same extent. It is not uncommon to see two individual willows of the same species growing side by side, one bearing multiple galls, the other free of galls. For this reason, only willow shrubs already bearing galls were used for these experiments. The galls were clipped off before the stems were introduced into the cages.

Figure 1: Internal cube of fine mesh netting.

The cages measured about a foot on all sides. Five faces were made out of rigid, soldered and vinyl-coated wire mesh with 1/2 in. spacing. The sixth side was made out of 1/8 in. plexiglass with a 4 in. circle cut out in the middle. The sides of the cube were lashed together with plastic wire ties and metallic wire. Out of fine meshed fiberglass hardware cloth I sewed a cube with the outside dimensions equal to the inside dimensions of the rigid wire cube (Figure 1). One side of the cloth cube had a 4 in. hole cut out in the middle to which a sleeve 4 in. in diameter and 6 in. long was sewn in extension of the hole. The fine meshed cube was then stuffed inside the rigid cube, the 6 in. long

[9]Sterling, Alaska. rabdophaga@gmail.com

sleeve was lined up with the hole in the plexiglass, and was extend outside of the cube. Plastic ties at each corner of the fabric cube held the liner in place inside the rigid cube.

In April, well before the willows unfurled their leaves, live *Rabdophaga* galls and stems from an identified willow were introduced inside the cage through the sleeve which was then sealed shut by several wraps of wire. The cage was stabilized on the willow by guy wires tied to the adjoining vegetation (Figure 2). The seal was kept throughout the growing season. The cage was removed in the fall and the stems were examined for the presence of galls containing live last instar larvae. The previous-year galls were also dissected and the number of successfully emerged gall midges was counted.

Figure 2: Cage #3 installed on a *Salix fuscescens* in the field.

On April 14, 2 cages were installed in the Arc Loop wetlands in Soldotna: One cage (#1) enclosed *Salix pulchra* stems, and 25 galls of *R. rosaria* species group induced on *Salix barclayi*. One "D" *rosaria* species group gall had been clipped off the *S. pulchra* stems. The other cage (#2) included stems from *Salix commutata* and 16 rosette galls from *S. barclayi*. One *R. rosaria* sp. group and one *R. rigidae* have been clipped off from the *S. commutata* stem.

On April 16 2009, one cage (#3) was installed in the wetlands near Ames Bridge, including *Salix fuscescens* stems and 17 galls of *R. rosaria* species group induced on *S. barclayi*.

On April 17, the two last cages were installed in Sterling, on Corey street. The first cage (#4) included *S. bebbiana* stems and 19 *R. rosaria* species group galls induced on *S. barclayi*. One old *R. rigidae* gall was noted on the stems included in the cage.

The last cage (#5) was a control which included *S. barclayi* stems and 14 *R. rosaria* group induced galls on *S. barclayi*.

The test showed that the same *Rabdophaga* species induced galls on both *S. commutata* and *S. barclayi* (Table 1). The negative result of the control demonstrated that, even when the host is known to be susceptible that there is not complete control of the experiment and that a negative result does not necessarily indicate that the willow being tested is not a potential host. These tests must be repeated. This experiment will be continued in 2010.

I want to thank Matthew Bowser for constructive criticism and help during the experiment and for the review of the manuscript. I want to thank also Nichola Lisuzzo for taking on the experiments in the Fairbanks area.

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Table 1: Numbers of donor galls, successfully emerged adult *Rabdophaga* midges, and new galls on five *Salix* species in enclosure cages seeded with *R. rosaria* species group galls from *S. barclayi*. *In this cage the base of the stem was severed and the stems died inside the cage.

Cage	Recipient Species	Donor Galls	Adults Emerged	New Galls
#1	*S. pulchra*	25	10	0
#2	*S. commutata*	16	11	10
#3	*S. fuscescens*	17	9	0
#4	*S. bebbiana*	19	15	0*
#5	*S. barclayi*	14	8	0

Distribution of Carabid beetles along an elevational gradient

by Matthew L. Bowser[8]*, Edward E. Berg*[10]*, Al Magness, and Dominique M. Collet*[9]

We examined the distributions of Carabid beetle species along an elevational gradient on the Kenai National Wildlife Refuge, Kenai Peninsula, Alaska, as candidate metrics for climate change monitoring. Carabid beetles tend to distribute themselves based on their climatic preferences (Ashworth, 2001) so paleoclimates can be inferred through Carabid beetle assemblages (Atkinson et al., 1987). If Carabid species were found to be stratified by elevation, then potential changes in species' elevational distributions may be attributed to changing climatic conditions as they move up- or down-slope tracking their climatic preferences.

On June 18, 2003 we installed 24 pitfall traps along a section of the Skyline Trail (60.53°N, 150.17°W). The trail is situated on a south-facing slope in the Mystery Hills on the western edge of the Kenai Mountains. The elevation on the transect ranged from 139 m to 790 m. Most of the stations were in mixed mature white spruce (*Picea glauca*) and paper birch (*Betula papyrifera*) forest with a substantial alder (*Alnus viridis*) understory, with the top five stations in mountain hemlock (*Tsuga mertesiana*), alder, or alpine tundra.

Pitfall traps consisted of open, white, plastic 480 mL cottage cheese containers with holes punched in the bottom for drainage. A 25 mm square piece of Hot Shot No-Pest® insecticidal strip was placed in each trap. We serviced the traps at two-week intervals (July 2, July 16, July 29, August 12, and August 27). Voucher specimens were identified by Foster Purrington (Ohio State University). All specimens were deposited in the arthropod collection of the Kenai National Wildlife Refuge (coden: KNWR). We sent our Carabid distribution data to Dr. Scott Elias (University of London), who applied the Mutual Climate Range (MCR) method (Atkinson et al., 1987) to infer climate from the observed assemblages at each point.

The traps yielded 182 Carabid beetles: mostly *Scaphinotus marginatus* (Fischer von Waldheim, 1822) and *Calathus advena* (LeConte, 1848) (Table 1). Other species were collected much less frequently.

We did not observe clear elevational stratification of Carabid beetle species (Figure 1). The two commonly collected species, *S. marginatus* and *C. advena*, were present across the entire elevational gradient from 139 m to 790 m. Less frequently encountered species were scattered over the range of elevations sampled.

Both *S. marginatus* and *C. advena* were most frequently collected early in the summer (Figure 2). Catch rates for *S. marginatus* were highest (0.12 individuals/trap day) in early July, after which time they gradually decreased to 0.025 individuals/trap day in late August. Catch rates for *C. advena* were highest in late June (0.10 individuals/trap day), dropped to zero in late July, and increased to 0.0083 individuals/trap day in late August.

Due to the paucity of stenothermic species in our samples, a reasonable climate estimate could only be estimated by MCR methods at the highest point (790 m), where *S.marginatus*, *C. advena*, and *A. alpina* were observed. The MCR climatic estimates for this point were July temperatures of 9.5-10.5°C and mean January temperatures of -11.5 to -8°C. At lower sites, the few species collected were eurythermic, with broadly overlapping temperature preferences, conveying relatively little information about climate.

[10]US Fish & Wildlife Service, Kenai National Wildlife Refuge, Soldotna, Alaska. Edward_Berg@fws.gov

Table 1: Quantities of Carabid beetle species collected.

Species	Quantity
Scaphinotus marginatus (Fischer von Waldheim, 1822)	125
Calathus advena (LeConte, 1848)	45
Pterostichus adstrictus Eschscholtz, 1823	2
Pterostichus brevicornis (Kirby, 1837)	3
Pterostichus cf. *brevicornis* (Kirby, 1837)	1
Pterostichus riparius (Dejean, 1829)	1
Trachypachus holmbergi Mannerheim, 1853	2
Trechus tenuiscapus Lindroth, 1961	2
Amara alpina (Paykull, 1790)	1

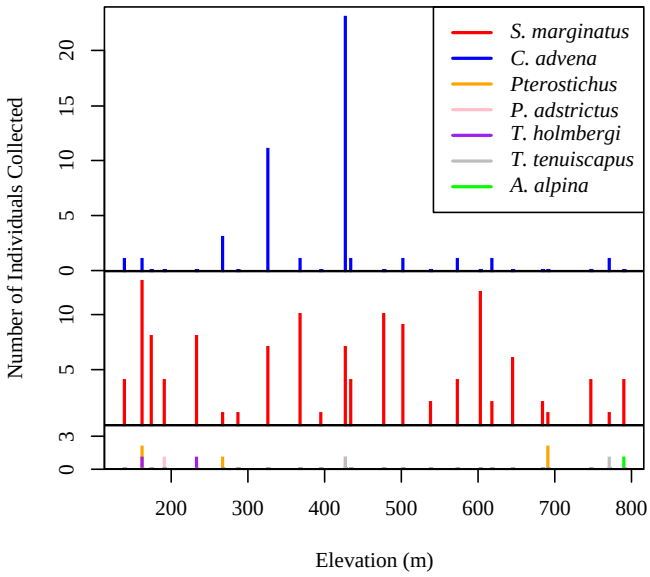

Figure 1: Elevational distribution of Carabid beetle species.

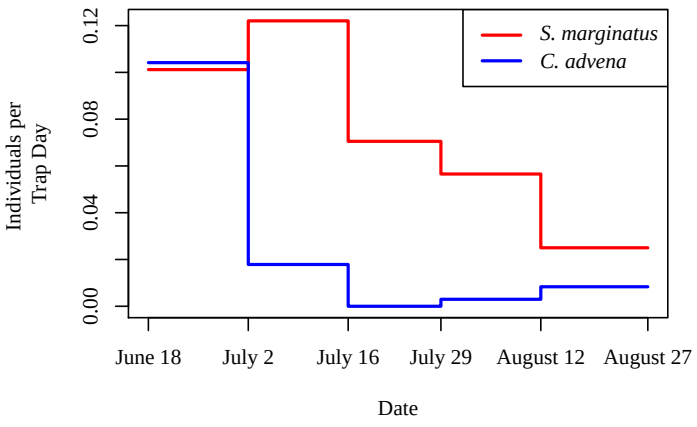

Figure 2: Seasonal activity of *S. marginatus* and *C. advena*.

The distributions of Carabid beetle species along this transect did not appear to be useful as a baseline for future monitoring because no clear elevational stratification was observed for frequently collected species. The relatively short length of the transect may have been partially to blame, however, even shorter elevational transects have yielded distinct carabid assemblages Butterfield (1996). Likely reasons why we observed a nearly uniform fauna are that the transition between boreal forest to the alpine occurred at the highest points in our study and that the summer temperature gradient along this south-facing slope was probably less dramatic than temperature gradients along slopes of other aspects.

A better way to design a study with similar goals would be to sample a set a of sites along a larger gradient of climate, preferably over a climatic gradient where change is expected. In addition, temperature data should be gathered at each site, e.g. by deploying inexpensive temperature data loggers, so that Carabid activity patterns and assemblages can be directly related to weather and climate.

We thank Foster Purrington and Scott Elias for their help with this project.

Supplementary data from this pilot study are available: the full dataset in CSV (Comma Separated Values) format (`http://www.akentsoc.org/doc/AKES_newsletter_2010_I_Carabids_data.csv`) and a map of collection sites in KML (Keyhole Markup Language) format (`http://www.akentsoc.org/doc/AKES_newsletter_2010_I_Carabids_sites.kml`).

References

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Review of the Third Annual Meeting

by Matthew L. Bowser[8]

The third annual meeting of the Alaska Entomological Society took place at the University of Alaska Museum (UAM) in Fairbanks on January 30, 2010. In addition to engaging talks and a productive business meeting, a highlight was the tour of the recently upgraded new carriages, insect cabinets, and wet collection room of the UAM entomology collection given by **Derek Sikes**.

Presentations

Derek Sikes spoke about recent major improvements to the University of Alaska Musuem Insect Collection and gave an update on the Arthropods of Alaska checklist. I introduced Alaska's two bristletail species in a short talk (see page 7 of this issue). **Rehanon Pampel** presented on work she has started on species composition, distribution, and seasonal abundance of the tribe Bombini. **Brent Mortensen** gave a presentation on his work examining the relationships of aspen, aspen leaf miners, and predators at three scales. **Joey Slowik** described the phylogenetic study of the *Pardosa groenalndica* species complex that he is pursuing. **Brandi Fleshman** gave a talk about her work on myrmecomorphy in *Micaria* spiders, including memorable video footage of a wolf spider nabbing a *Micaria*. **Dominique Collet** showed us how he is complementing morphological taxonomy in the difficult genus *Rabdophaga* through rearing experiments (see page 9 of this issue). **Jim Kruse** updated us on invasive insects in Alaska, focusing in on *Monosoma pulveratum* (see page 2 of this issue). He gave a separate talk on 2009 forest health conditions in Alaska. **Kenelm Philip** provided an update on the Butterflies of Alaska project and showed off beautiful photographs of each of Alaska's butterfly species, all photographs of live animals in their natural habitats. **Alberto Pantoja** presented on the insects associated with agricultural production in Alaska.

Business Items

- A motion passed at the 2008 meeting altering the membership categories and dues schedule was reversed, maintaining the original categories and dues.
- We set a tentative date for the fourth annual meeting: late January, 2011.
- A slate of officers was nominated, voted on, and passed: **M. Bowser** as president, **D. Sikes** as vice president, **J. Kruse** as secretary, and **R. Burnside** retained as treasurer.

Much of the business meeting focused on membership issues. We opted to move toward an electronic system for accepting membership applications. In addition, several changes were requested for the web site (http://www.akentsoc.org). We also discussed ways to expand our educational outreach beyond college students to public schools and home school groups.

The student award committee awarded the **Outstanding Student Presentation Award** to **Brent Mortensen** for his presentation, "Scales of predator mediated defense against an aspen (*Populus tremuloides*) specialist".

Upcoming Events

Third Annual Alaska Dragonfly Day, June 19

The Third Annual Alaska Dragonfly Day will be held at Creamer's Field in Fairbanks June 19, 2010. As in previous years, the event will feature dragonfly arts and crafts, live dragonfly larvae and other aquatic invertebrates, dragonfly study tools-of-the-trade, information on wetlands, and the popular dragonfly walks where participants net, hold, and identify adult dragonflies. For more details, contact Joanna Fox with the Kanuti National Wildlife Refuge at 907-456-0330 or joanna_fox@fws.gov.

Young and old alike net dragonflies at a temporary pond during the 2008 Dragonfly Day held at Creamer's Field in Fairbanks. Photograph by Robert Armstrong.

Southeast Alaska BioBlitz, June 26-27

Although summer is still a long way off, please mark your calendars and plan to come join us on June 26-27, 2010 at Eaglecrest in Juneau for the first ever Southeast Alaska Bioblitz. The goal of the Bioblitz is to count as many species as possible in a 24 hour period from the alpine to the ocean. The event is being sponsored by ADF&G, USFS, USFWS, NOAA, and CBJ.

At the moment we are recruiting scientists and naturalists for the taxon teams—information and a schedule of events for the general public will follow. If you would like to participate, please contact the appropriate taxon leader: **Terrestrial Invertebrates**—John Hudson (USFWS), john_hudson@fws.gov, 780-1169; **Freshwater Invertebrates**—Mark Wipfli (UAF), mark.wipfli@uaf.edu, 474-6654.

Please feel free to forward this announcement to any other scientists or naturalists you know who might be interested. For more information, visit the Southeast Bioblitz webpage at http://www.wildlife.alaska.gov/index.cfm?adfg=education.bioblitz or contact Karen Blejwas (465-4328; karen.blejwas@alaska.gov).

Dragonfly Day, July 9

Dragonfly day will be at the Kenai National Wildlife Refuge Environmental Education Center (Ski Hill Road, Soldotna) on July 9, 2010. There will be dragonfly and damselfly arts and crafts, games, life cycle information, etc. John Hudson and Robert Armstrong will be guest speakers. Guided walks may take place off-site on July 10. For more information, contact Michelle Ostrowski at 907-260-2839 or Michelle_Ostrowski@fws.gov.

Fourth annual meeting, January 2011

Plans for the 2011 annual meeting of the Alaska Entomological Society are underway, tentatively scheduled for late January in Anchorage. Details will be announced on the Alaska Entomology Network e-mail list (https://lists.uaf.edu:8025/mailman/listinfo/akentonet-l). For more information, write the AKES secretary at secretary@akentsoc.org.