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Microarthropods and other soil fauna of Tanana River floodplain soils: a primer

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by Robin N. Andrews¹

Though largely unseen, tiny microarthropods form soils, influence rates of decomposition, and shape bacterial, fungal, and plant communities (Seastedt, 1984; Wall and Moore, 1999; Walter and Proctor, 2013). Difficult to see without a microscope, most microarthropods are between a 0.1 and 2 mm in length. Though they exist much deeper, microarthropods are most abundant in first 5 centimeters of soil where they can reach 70,000 per square meter in early successional alder stages and a million per square meter in mature white spruce stands. These arthropods occupy at least the first couple meters in unfrozen boreal soil decreasing in numbers with depth. We are studying the development of microarthropod communities in three forest stand types along the Tanana River floodplain: early-succession alder, mid-succession balsam poplar, and late-succession white spruce. Preliminary results indicate increasing abundance and richness of microarthropod taxa as succession progresses.

Alaskan microarthropod communities are primarily composed of mites, collembolans, and proturans, but their communities intersect with larger arthropods like myriapods, spiders, and insects. The soil and litter dwelling mites, formerly known as Acari, are a diverse assem-

blage composed of species from the superorder Parasitiformes containing members of order Mesostigmata, and superorder Acariformes composed of the suborders Endeostigmata, Prostigmata, and Oribatida (Krantz and Walter, 2009).



Figure 1: Mesostigmata: Ologamasidae: *Gamasellus* sp.

Soil Mesostigmata, a taxonomic distant cousins of ticks, are primarily predators of nematodes and small arthropods (Walter and Proctor, 2013). The Mesostigmata above (Figure 1) is an undescribed species of genus *Gamasellus* (Ologamasidae) found in abundance in our early-succession alder stands.

¹Ph.D student, Department of Biology and Wildlife, University of Alaska Fairbanks

Endeostigmatan mites often appear little changed from forms seen in the Devonian era, over 400 million years ago, displaying primitive morphological characters like the visible segmentation seen in these Alycidae. The segmentation may make them appear similar to Opiliones (daddy long-legs) but they are only very distantly related (Krantz and Walter, 2009; Dunlop et al., 2014). The Alycidae below (Figure 2) is the mite of the year for 2018. It is purplish, the color of the year. (I informally announce the Mite of the year on New Year's Eve each year beginning in 2016: the year of *Gamasellus*).

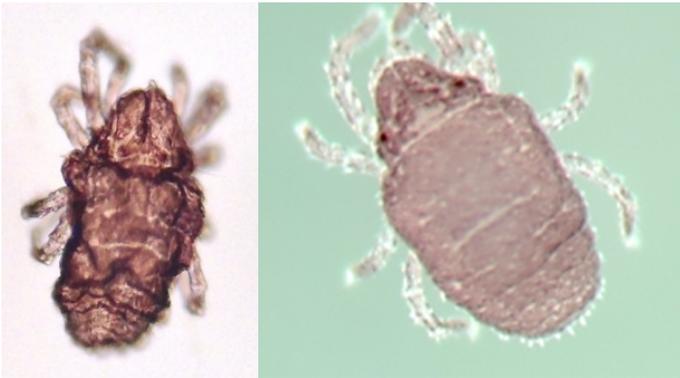


Figure 2: Endeostigmata: Alycidae: *Alycus* sp.

Prostigmata are diverse in form and play many ecological roles among them herbivores, fungivores, detritivores, parasites, and predators. Many have complex life cycles in which they switch their feeding strategy during development, for example from parasites to predators. The Cunaxidae (Figure 3, left) seen here is a predator. The Microdispidae (Figure 3, right) is a fungivore and appears to be carrying a spore, a behavior known from many families of cohort Heterostigmatina. Some Heterostigmatans are thought to farm fungi in a manner similar to ants. (Krantz and Walter, 2009; Walter and Proctor, 2013).



Figure 3: Prostigmata: Cunaxidae (left) and Microdispidae (right)

²See <https://www.humansandnature.org/unseen-commonest-animal>

The suborder Oribatida may be the best-known group of mites and contains some of the most common terrestrial arthropods in the world like *Oppiella nova*². Brachychthoniidae (Figure 4, right) is the most abundant family of Oribatida at our sites. They are early colonizers in the Tanana River's riparian zone while Tectocephidae (Figure 4, left) appear in much lower abundances, mostly in successional stages dominated by balsam poplar and white spruce. In general, oribatid communities are richer and more abundant later in succession in deeper more developed soils.



Figure 4: Oribatida: Tectocephidae (left) and Brachychthoniidae (right)

Collembolans, or springtails, and Proturans are taxonomically Hexapoda closely related to insects (Dindal, 1990; Hopkin, 1997; Pass and Szucsich, 2011). Collembolans are found in all successional stages along the river, but similar to oribatid mites, they increase in abundance and diversity in the balsam poplar and white spruce stages. Collembola are thought to be mostly fungivores but may eat litter, bacteria, nematodes, and scavenge other animal foods (Chahartaghi et al., 2005). Collembolans similar to the white one (Figure 5, left) occur in great abundance in our the balsam poplar stands. Entomobryomorph collembolans like the purple one (Figure 5, right) are found most often in the upper litter layers.



Figure 5: Collembola

Proturans (Figure 6) are common in some of our white spruce stands. Conifers form intimate relationships with ectomycorrhizal fungi. Proturans have been observed to eat mycorrhizal fungi and are often found in proximity to mycorrhizal species (Dindal, 1990; Pass and Szucsich, 2011; Zieger et al., 2017). The possible impact of proturans grazing on mycorrhizal fungi and its consequences to ecosystem functions are unknown.



Figure 6: Protura

Pauropods (Figure 7) are small myriapods related to millipedes and centipedes. Along with many pairs of legs (9-11 pairs in the adult), pauropods have branched antennae. These small myriapods are likely fungivores and are found in the balsam poplar and white spruce stage riparian zones (Dindal, 1990).



Figure 7: Pauropoda

Root Aphids (Figure 8) are a group of small insects from the order Hemiptera. While aphids can look a lot alike, different forms can be found in alder, balsam poplar, and white spruce stages along the river floodplain. Aphids with their piercing, sucking mouthparts are known to be important vectors of plant diseases spreading infections and weakening plant defenses (Dixon, 2012). We found aphids in relatively high abundance at an alder site in-

fectured with a fungal canker but possible relationships between aphids and infected trees remain to be explored.



Figure 8: Hemiptera: Aphidoidea

Finally, we found an exotic worm (*Eiseniella tetraedra*) at one study site along the Tanana River floodplain (see Booyesen et al., 2018). Worms were collected in summers 2016 and 2017 suggesting that they successfully overwintered in the warm floodplain soils. The spread of exotic worms is a major cause for concern in North America (Callaham et al., 2006) especially Canada (Addison, 2009; Langor et al., 2014). Earthworm spread is well known in more southern regions of Alaska (Costello et al., 2011; Saltmarsh et al., 2016) but relatively new to interior Alaska and more sightings are happening yearly (personal observation). Invasive worms have been shown to negatively influence microarthropod communities (González et al., 2003; Eisenhauer, 2010; Cameron et al., 2013; Schlaghamerský et al., 2014) and may affect other soil fauna (Schlaghamerský et al., 2014; Ferlian et al., 2017) and decomposition rates (González et al., 2003). Below (Figure 9) is a native pot worm, an enchytraeid (*Clitellata*). Enchytraeids are distant cousins of the better known earthworms in the family Lumbricidae (Erséus et al., 2010). One feature distinguishing them from earthworms is their much wider segmentation. Segments are much longer relative to their width in enchytraeids than in earthworms (Dindal, 1990, Thomas Peham, personal communication).



Figure 9: Enchytraeidae

Stay tuned for future developments!

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Larger insect collection specimens are not more likely to show evidence of apparent feeding damage by dermestids (Coleoptera: Dermestidae)

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by Joel Stone¹ and Derek S. Sikes¹

Abstract

Dermestids can not only cause damage to museum insect specimens but if left unchecked can ruin museum collections. This study aimed to determine whether larger insect specimens are more likely to show evidence of apparent feeding damage by dermestids than smaller specimens. We examined 366 specimens of various taxa in the Kenelm W. Philip collection, currently housed in the University of Alaska Museum Insect Collection. We measured the size of each specimen and examined each specimen for evidence of dermestid feeding under magnification. The median specimen sizes of the damaged and undamaged groups were compared using a Mann-Whitney *U*-test. We could not reject the null hypothesis ($p = 0.0878$) that all sizes of specimens are equally likely to show apparent feeding damage.

Introduction

Many different fields of research regularly use museum specimens for a variety of topics from ecology to phylogenetics (Suarez and Tsutsui, 2004; Andersen and Mills, 2012). Because of this, it is important to ensure the long-term preservation and protection of these specimens. Dermestidae are a family of beetles (Coleoptera) that feed on protein-rich, dry animal and plant material. In nature, dermestids provide a key ecosystem function as decomposers, but they are commonly considered pests in museums because they feed on specimens and can be difficult to control (Burgess, 1959; Gilberg and Brockerhof, 1991). Many studies have looked at effective ways to protect specimens from this damage (Zaitseva, 1987; Su and Scheffrahn, 1990). But much remains to be learned about the behavior of dermestids that feed on museum specimens. The purpose of this study was to determine if dermestids show a size preference in their choice of specimens. However, be-

cause live dermestids were not used and we had to assume dermestids were the causative agents of the observed damage, we tested the null hypothesis that the median sizes of apparent feeding-damaged and undamaged specimens would not be significantly different (with no specification of the causative agent of the damage).

Methods

One drawer of specimens in the Kenelm W. Philip collection, currently held at the University of Alaska Museum Insect Collection, that had obvious signs of dermestid damaged specimens was used (Figure 1). This drawer had exuvia of dermestid larvae, feeding detritus, and obvious holes chewed in specimens, and contained 366 dried, pinned insect specimens of various sizes and insect taxa (misc. orders, specimens thought to have been collected by W. C. Frohne or at least part of Frohne's collection). Size mea-

¹University of Alaska Museum, Fairbanks, Alaska, 99775-6960, USA

measurements of each were taken from the front of the head (not including mandibles, palps, or antennae) to the end of the abdomen (not including cerci, ovipositor, etc.) using digital calipers in millimeters to the nearest one-tenth of a millimeter for specimens over 3 mm and using an ocular micrometer to the nearest tenth of a millimeter using a Leica M165C stereomicroscope for specimens under 3 mm. Specimens were examined under magnification using this Leica microscope for direct evidence of chew marks or holes left by dermestids (e.g., Figure 2).

If a specimen was missing its head or abdomen the size measurement was estimated to account for the lost body part. Specimens with broken body parts but no evidence of dermestid feeding were considered not feeding damaged (specimens can become broken from a variety of non-

dermestid causes). Each measurement was assigned to one of two groups: specimen feeding damaged or specimen not feeding damaged. The smallest specimens might get eaten entirely, leaving very little evidence, and thus be unavailable for measuring—possibly biasing the results towards an overabundance of specimens too large to eat entirely. The drawer had 12 pins with labels but missing specimens, ten of which were pointed specimens. To account for these potentially feeding damaged and small, but missing specimens, we added 12 randomly generated size values between 1 mm and 5 mm to the damaged data (generated using the R (R Core Team, 2017) command `runif(12, min=1, max=5)`). These added values constitute the last 12 values of damaged specimens in the data. The data are archived at FigShare doi: [doi:10.6084/m9.figshare.5930686](https://doi.org/10.6084/m9.figshare.5930686).



Figure 1: One drawer of the Kenelm W. Philip collection of miscellaneous taxa from various regions including the Philippines, California, Missouri, Ethiopia, and Alaska.



Figure 2: A small parasitoid wasp specimen with a hole presumably chewed by dermestids (arrow). The numbers on the scale bar mark half mm increments.

Results

There were 182 undamaged specimens and 184 feeding damaged specimens. The mean size of the undamaged specimens was 11.83 mm (9.22 mm SD) and the median was 10.47 mm (Figure 3). The mean size of the feeding damaged specimens was 13.97 mm (9.28 mm SD) and the median was 11.32 mm. The Mann-Whitney U -test showed

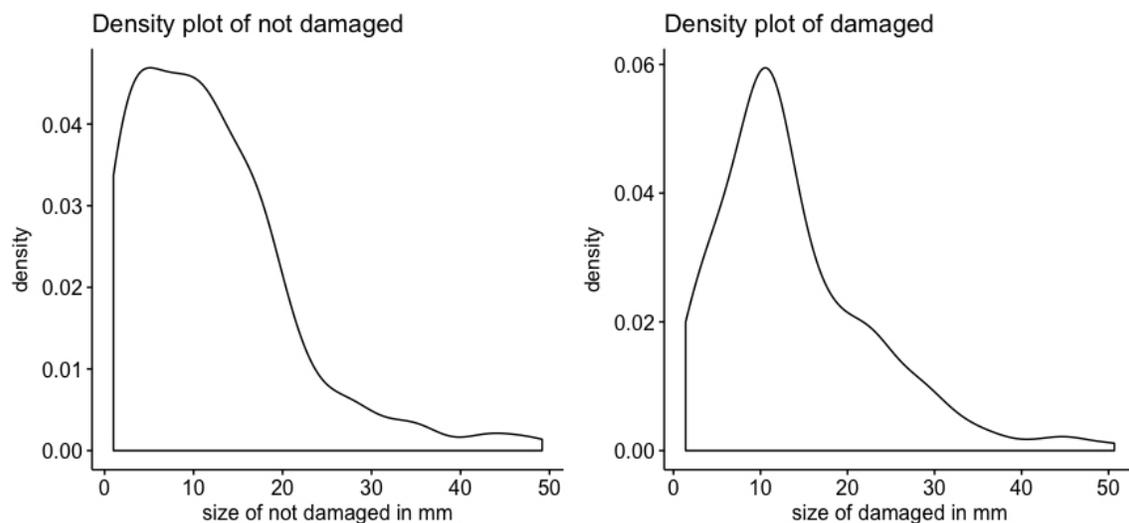


Figure 3: Density plots of not feeding damaged ($n = 182$) and feeding damaged ($n = 184$) specimens.

these medians to be significantly different ($W = 14160$, $p = 0.0107$). However, when this test was re-run with the 12 added damaged values to account for small specimens presumably eaten entirely the medians were not significantly different ($W = 16024$, $p = 0.0878$).

Discussion

It was a long-held assumption of DSS that larger insect specimens are more likely to show feeding damage by dermestids. These results demonstrate this impression is most likely due to feeding damage being easier to see on larger specimens. Supporting this view are the results of a prior analysis of this question in which feeding damage was assessed without magnification. Those data strongly rejected the null hypothesis and indicated larger specimens were more likely to show feeding damage ($W = 4543.5$, $p < 0.00001$). However, when magnification was used to assess damage it was possible to see damage on the smaller specimens (eg., Figure 3) that had been previously missed without magnification. These results support a random-encounter model of dermestid feeding with no apparent choice or preference based on specimen size.

Acknowledgments

We thank Kathryn Daly for her tireless work curating the Kenelm W. Philip collection and for help in R. We also thank the students in the University of Alaska Fairbanks Entomology course who performed class project ideas of this nature. We thank Nina Sikes for taking the photograph used for Figure 1.

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DNA barcoding Alaskan willow rosette gall makers (Diptera: Cecidomyiidae: *Rabdophaga*)

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by Matt Bowser¹, Apphia M. Bowser¹, Ethan L. Bowser¹, Miriam D. Bowser¹, Dominique M. Collet², and Tracy Melvin³

Introduction

Members of the *Rabdophaga rosaria* group form conspicuous rosette galls on a variety of willow (*Salix* spp.) hosts (Collet, 2002; Amendt, 2003) and have a holarctic distribution. Gall formation halts elongation of willow stems and alters the morphology and chemical makeup of host tissues (Gailite et al., 2005; Samsone et al., 2011). Ecologically, these flies are a keystone species for a community of insects associated with rose galls including multiple parasitoid, hyperparasitoid, and commensal species (Van Hezewijk and Roland, 2003; Collet, 2006; Skuhrová and Thuróczy, 2007). The larvae serve as food for chickadees and tits, which pick them out of galls in winter (Van Hezewijk and Roland, 2003; Nyman et al., 2011, Figure 1). The galls themselves are avoided by moose (Kenai National Wildlife Refuge staff, 1981; Ford et al., 1995; Rea, 2012) and snowshoe hares (Ford et al., 1995).



Figure 1: A black-capped chickadee dismantling a gall induced by *Rabdophaga strobiloides* in central Michigan, December 30, 2017 (<https://flic.kr/p/232e1Yn>). Image © J. D. Sommer. Used with permission.

¹US Fish & Wildlife Service, Kenai National Wildlife Refuge, Soldotna, Alaska, Matt_Bowser@fws.gov

²Homer, Alaska, dominique.m.collet@gmail.com

³Michigan State University, East Lansing, Michigan, swemtrac@msu.edu

The taxonomy of North American members of this group is problematic. Although Gagné (1989) provided a key and illustrations to some members of this group, the only descriptions available are the original descriptions of Osten Sacken (1862), Walsh (1864), Osten Sacken (1878), and Packard (1869), which do not enable separation of the species except through willow host species and gall morphology. Also, North American species have not been compared to Palearctic species.

Through a literature search for species that form rosette or rosette-like galls on willows we found a total of nine named *Rabdophaga* species: *Rabdophaga clavifex* (Kieffer, 1891); *Rabdophaga jaapi* Rübsaamen, 1916; *Rabdophaga rosaria* Loew, 1850; and *Rabdophaga rosariella* (Kieffer, 1897) from the Palearctic and *Rabdophaga salicisbrassicoides* Packard, 1869; *Rabdophaga saliscoryloides* Osten Sacken, 1878; *Rabdophaga salicisgnaphaloides* Osten Sacken, 1878; *Rabdophaga salicisrhodoides* Osten Sacken, 1878; and *Rabdophaga strobiloides* Osten Sacken, 1862 from the Nearctic.

We sought to determine whether one polyphagous species or multiple species of *Rabdophaga* caused rosette galls on southcentral Alaskan willow species using DNA barcodes. We also wanted to relate these entities to described *Rabdophaga* species where possible.

Methods

Initially as a homeschool science project, the first four authors collected willow rosette galls from *Salix sitchensis* Sanson ex Bong. and *Salix barclayi* Andersson to determine whether or not these willows were galled by distinct species of *Rabdophaga*. We later expanded our sampling to a variety of willow hosts in southcentral Alaska, using the illustrations and text of Collet (2002) as a guide to these types of galls. Alaskan willow specimens that could not be identified with confidence by MLB were identified by DC. Additional specimens were obtained by TM from Kodiak Island and Michigan. A map of collection localities is given in Figure 2.

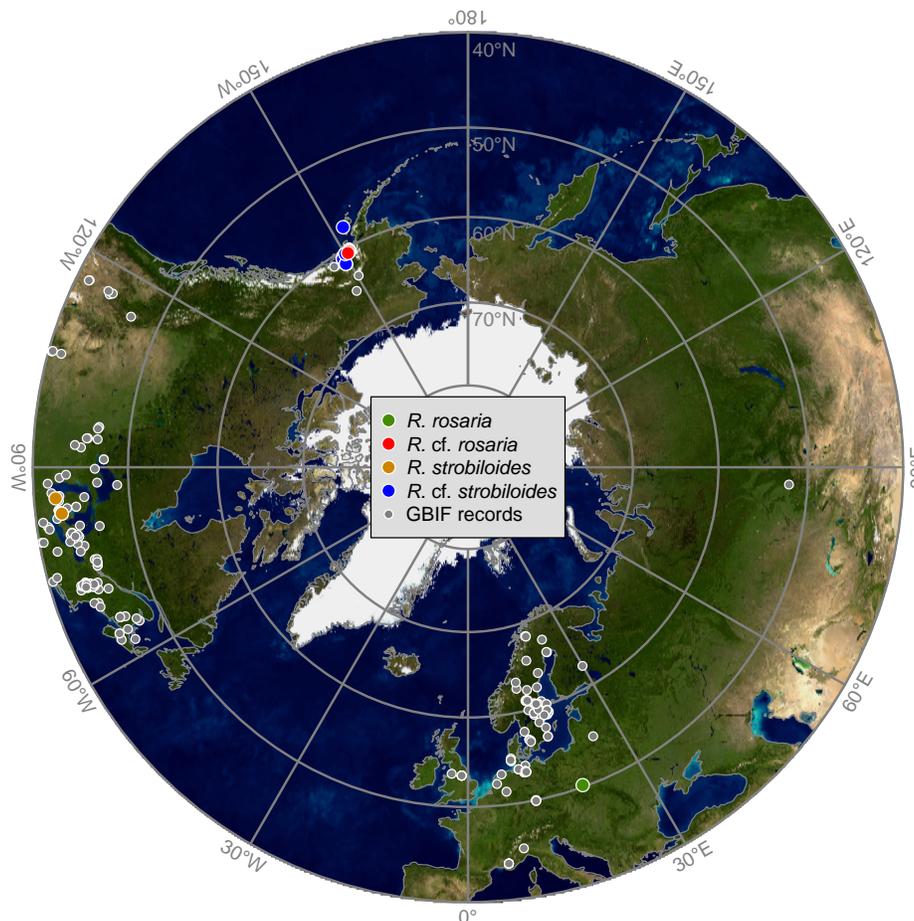


Figure 2: Polar map of collection localities color-coded as in clades in Figure 4. See the Methods section for details on the GBIF records.

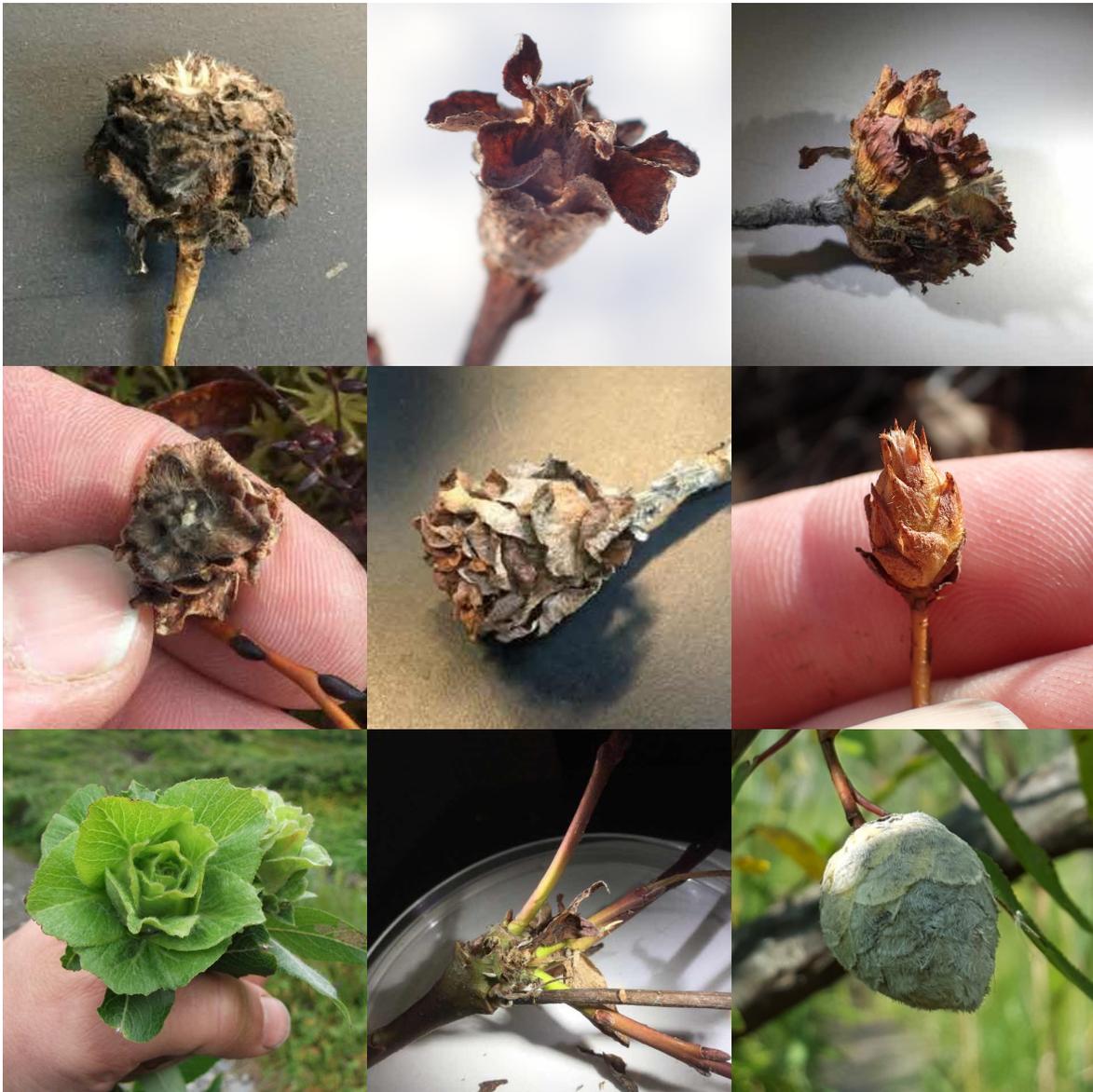


Figure 3: Representative photographs of galls sampled. **Top left:** Rosette gall on *Salix barclayi*, Alaska, Kenai Peninsula, Summit Creek, collected February 14, 2016. Arctos GUID: KNWR:Ento:11156. Image CC0 Matt Bowser (<http://arctos.database.museum/media/10510773>). **Top center:** Rosette gall on *Salix bebbiana*, Alaska, Soldotna, Ski Hill Road, February 20, 2018. Arctos GUID: KNWR:Ento:11281. Image CC0 Matt Bowser (<https://www.inaturalist.org/photos/14507792>). **Top right:** Rosette gall on *Salix commutata*, Alaska, Kenai Peninsula, Turnagain Arm, collected February 14, 2016. Arctos GUID: KNWR:Ento:11114. Image CC0 Matt Bowser (<http://arctos.database.museum/media/10522732>). **Middle left:** Rosette gall on *Salix fuscescens*, Alaska, Kenai Peninsula, Soldotna, Ski Hill Road, April 11, 2016. Arctos GUID: KNWR:Ento:11124. Image CC0 Matt Bowser (<http://arctos.database.museum/media/10508965>). **Center:** Rosette gall on *Salix hookeriana*, Alaska, Kenai Peninsula, Turnagain Arm, collected September 23, 2016. Arctos GUID: KNWR:Ento:11179. Image CC0 Matt Bowser (<http://arctos.database.museum/media/10518370>). **Middle right:** Rosette gall on *Salix pulchra*, Alaska, Kenai Peninsula, Sterling, Moose River Drive, September 26, 2016. Arctos GUID: KNWR:Ento:11169. Image CC0 Matt Bowser (<http://arctos.database.museum/media/10508962>). **Bottom left:** Rosette gall on *Salix richardsonii*, Alaska, Kenai Peninsula, Palmer Creek, August 13, 2016. Arctos GUID: KNWR:Ento:11115. Image CC BY Matt Bowser (<https://www.inaturalist.org/photos/4583073>). **Bottom center:** Rosette gall on *Salix sitchensis*, Alaska, Kenai Peninsula, Jims Landing, collected on December 7, 2015. Arctos GUID: KNWR:Ento:11097. Image CC0 Matt Bowser (<http://arctos.database.museum/media/10522737>). **Bottom right:** Willow pinecone gall on *Salix nigra*, Michigan, Kalamazoo County, Al Sabo Preserve, Portage Creek, September 16, 2017. Arctos GUID: KNWR:Ento:11280. Image CC BY-NC Tracy Melvin (<https://www.inaturalist.org/photos/13524200>).

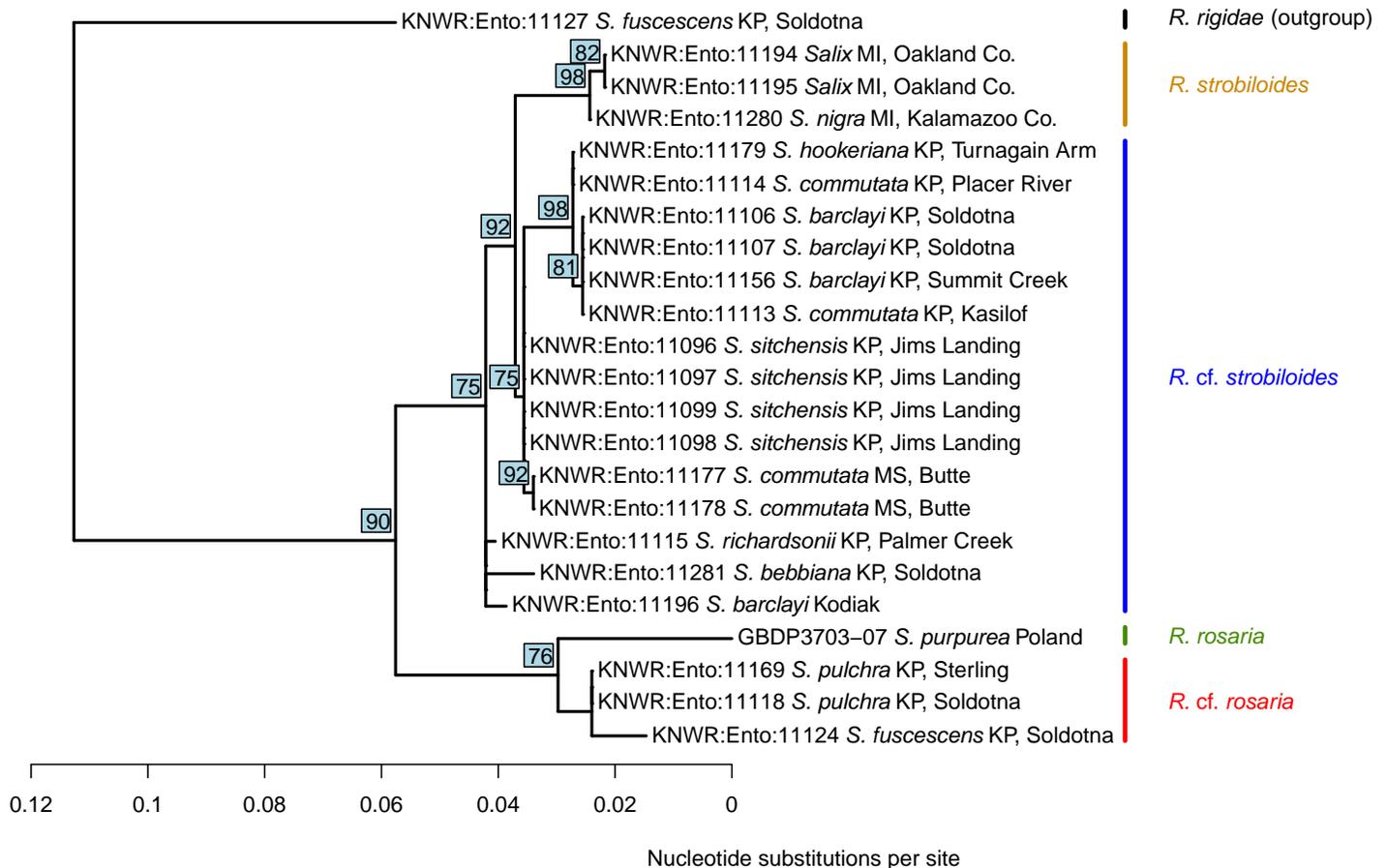


Figure 4: Phylogram of *Rabdophaga* sequences generated by PhyML. Node support values are given as percentages in blue boxes where support values were greater than 50%. KP: Kenai Peninsula, Alaska; MI: Michigan; MS: Matanuska-Susitna Borough, Alaska.

We dissected *Rabdophaga* larvae from galls and submitted whole specimens for DNA barcoding using LifeScanner kits (<http://www.lifescanner.net/>). Sequences from the DNA barcoding region of the COI gene were obtained by the Center of Biodiversity Genomics using proprietary methods. We downloaded from BOLD (Ratnasingham and Hebert, 2007) one sequence of *Rabdophaga rosaria* from Poland obtained by Sato and Yukawa (2006). We included a sequence of *Rabdophaga rigidae* (Osten Sacken, 1862) as an outgroup.

Sequences were aligned using Clustal Omega (Sievers et al., 2011) on EMBL-EBI (McWilliam et al., 2013; Li et al., 2015), accepting default parameters. A phylogenetic tree was generated from the aligned sequences using PhyML (Lefort et al., 2017) accepting default parameters.

For making a map we searched GBIF (<https://www.gbif.org/>) for all records of *Rabdophaga* on April 25, 2018. These data are available as a GBIF Occurrence Download (doi:10.15468/dl.luzr8v). For mapping we included only the nine *Rabdophaga* species of willow rosette gall mak-

ers listed above. We also added a single observation record from from Russia (<https://www.inaturalist.org/observations/7431914>).

Results

We obtained DNA barcode sequences of rosette gall makers from the following willow hosts: *Salix barclayi*, *Salix bebbiana* Sarg., *Salix commutata* Bebb, *Salix fuscescens* Andersson, *Salix hookeriana* Barratt ex Hook., *Salix pulchra* Cham., *Salix richardsonii* Hook., and *Salix sitchensis* from southcentral Alaska; *S. barclayi* from Kodiak Island; and *Salix nigra* Marshall from Michigan (Figure 3).

Specimen data are available on Arctos (<http://arctos.database.museum>) via a saved search (doi:10.7299/X7PG1S1N). Sequence data are available on BOLD.

In the phylogenetic analysis, rosette gall makers were placed in two main clades (Figure 4) with distances be-

tween members of the clades of 0.05–0.09 nucleotide substitutions per site.

One clade included *Rabdophaga rosaria* from Poland and gallers from *S. pulchra* and *S. fuscescens* hosts with distances among members of 0.00–0.04 nucleotide substitutions per site. Distances between the Alaskan specimens and *R. rosaria* were 0.04 nucleotide substitutions per site. We tentatively identified Alaskan members of this clade as *Rabdophaga cf. rosaria*.

The other clade included *Rabdophaga strobiloides* from Michigan and Alaskan specimens from *S. barclayi*, *S. bebbiana*, *S. commutata*, *S. hookeriana*, and *S. richardsonii*. Distances between members of this clade were 0.00–0.03 nucleotide substitutions per site. The smallest distance between *R. strobiloides* and Alaskan members of this clade was 0.01 nucleotide substitutions per site. We tentatively identified Alaskan members of this clade as *Rabdophaga cf. strobiloides*.

Our sequences were organized into corresponding clades by BOLD's BIN algorithm (Ratnasingham and Hebert, 2013). Members of the clades including *R. rosaria*, *R. strobiloides*, and *R. rigidae* were placed in BINs BOLD:ADA9342 (doi:10.5883/BOLD:ADA9342), BOLD:ACZ0652 (doi:10.5883/BOLD:ACZ0652), and BOLD:ADB1445 (doi:10.5883/BOLD:ADB1445), respectively. BINs BOLD:ADA9342 and BOLD:ACZ0652 were separated by 3.75% (*p*-dist); maximum within BIN distances were 0.00% to 2.01%.

Sequences from three California specimens identified as *Rabdophaga salicisbrassicoides* have not yet been publicly released⁴, but the sequences are available for searching via BOLD's Identification Engine. They are 95% and 96% similar (*p*-dist) to Alaskan *R. cf. strobiloides* and *R. cf. rosaria*, respectively.

Discussion

Both our analysis and BOLD's BIN algorithm placed Alaskan *Rabdophaga cf. strobiloides* with *Rabdophaga strobiloides* from Michigan. The Alaskan specimens may be conspecific with *R. strobiloides*, consistent with identifications of this species on *S. scouleriana* in British Columbia (Rea, 2012) and on *S. bebbiana* in Alberta (Van Hezewijk and Roland, 2003).

We found Alaskan members of the *R. cf. strobiloides* group induced galls on five species of willows in subgenus *Vetrix*, clade C2 of Lauron-Moreau et al. (2015). Our finding that members of this group galled *S. barclayi* and *S. commutata* corroborates the rearing experiment of Collet (2010), who found that midges reared from *S. barclayi* formed rosette galls on *S. commutata*. Elsewhere in North America, *R. strobiloides* has been recorded from *Salix cordata*

Michx., *Salix eriocephala* Michx., *Salix humilis* Marshall, *Salix nigra* Marshall (Wilson, 1968; Gagné, 1989), and *Salix scouleriana* Barratt ex Hook. (Rea, 2012), members of multiple subgenera of *Salix* in clades C1 and C2 of Lauron-Moreau et al. (2015).

Interestingly, even though *R. strobiloides* parasitizes *Salix scouleriana* in British Columbia (Rea, 2012), rosette galls on *S. scouleriana* appear to be extremely rare on the Kenai Peninsula. DC is aware of a single example of a rosette gall on *S. scouleriana* from Sterling, Alaska. MLB has not seen a rosette gall on *S. scouleriana*.

Alaskan specimens of *Rabdophaga cf. rosaria* were different enough from the one Old World sequence of *R. rosaria* that they may represent separate species. The Alaskan specimens, all taken from wetlands, were collected from *S. fuscescens* and *S. pulchra*, both facultative wetland species (Collet, 2002) and both members of the C2 clade of Lauron-Moreau et al. (2015). It seems more likely that the Alaskan *R. cf. rosaria* is a specialist on these willow species rather than a specialist on the wetland habitat because *R. cf. strobiloides* co-occurs with *R. cf. rosaria* on separate willow species in the same wetlands.

It remains to be resolved whether some of the genetic structure within these two groups is correlated with specialization on willow host species, geographic separation, or morphological differences. We could not conclusively answer our initial question of whether rosette makers on *S. sitchensis* and *S. barclayi* were distinct because the observed differences could have been due to geographic patterns. In the future we would like to obtain sequences from *R. cf. strobiloides* gall midges on *S. sitchensis* from other localities to help resolve this question. DC and MLB have observed that at some localities where both *S. barclayi* and *S. sitchensis* are present, rosette galls could only be found on *S. barclayi*, suggesting that the two willow species may be galled by distinct *Rabdophaga* species. Alternatively, it may be that a single species of *Rabdophaga* prefers *S. barclayi* over *S. sitchensis* where both hosts are available.

Morphologically, all galls of Alaskan *Rabdophaga* that we collected were more open than what is typical of *R. strobiloides*. In the key of Gagné (1989) they would have been identified as *R. salicisbrassicoides*, *R. salicisrhodoides*, or *Rabdophaga saliciscoryloides*, which form more open rosette galls. Most Alaskan rosette galls more closely resemble illustrations and photographs of European *R. rosaria* than any of these North American species. An exception is the unusual morphology of rosette galls on *S. sitchensis*, which often contain more than one midge larva and which often have new stems originating from leaf axils within the galls.

At present only a single sequence from this group is available from the Old World. It would be helpful to obtain DNA barcode sequences from more Palearctic rosette gall makers to clarify the relationships of these entities and

⁴see http://www.boldsystems.org/index.php/Taxbrowser_Taxonpage?taxid=355957 and doi:10.7299/X73J3D8D

North American *Rabdophaga* cf. *rosaria* and *Rabdophaga* cf. *rosaria*. It would also be desirable to obtain access to sequences of North American *R. salicisbrassicoides*, *R. salicisrhodoides*, and *Rabdophaga saliciscoryloides* for comparison.

Finally, we hope that our exploratory work using DNA barcoding is followed up by a more integrated approach including morphology of the gall midges and galls.

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How heating affects growth rate of Dubia roaches

by Fionn D. Pietsch

Introduction

I have always loved entomology. My favorite insects are cockroaches. I currently am raising two colonies of cockroaches: one colony of Madagascar Hissing cockroaches (*Gromphadorhina portentosa*), and the other of Dubia Roaches (*Blattella germanica*). Dubia roaches live in central and south America. Madagascar hissing cockroaches live in Madagascar. Both of these areas are in the tropics and have warm climates but the Madagascar Hissing cockroaches prefer cooler habitats relative to the Dubia roaches. The only cockroaches that live in Alaska are non-native species that live in warm buildings.

You may wonder why you usually only see insects in the warmer months of the year. You may think it is because there is less food and water in the winter, which may be partially true. However, the primary reason is because of heat. Heat is one of the most important things for insect growth. I formed a hypothesis that supplementary heating

would increase the growth rate of Dubia roaches compared to their growth rate at room temperature.

Methods

This spring, for the Interior Alaska Science Fair, I performed an experiment where I used Dubia Roaches. I had four groups which were in plastic container with air holes in them. Three of the groups sat on a 17 watt seedling heating mat (Hydrofarm Seedling Heat Mat) and the control group did not. The heated enclosures went from 23.9 °C to 26.1 °C. The unheated enclosure was 16.1 °C to 21.1 °C. All groups had the same amount of food (Nature Zones bites for Roaches) and water (Fluker's cricket quenchers calcium fortified). In each group there were about 16 juvenile cockroaches, weighing between 1.5–2 grams per group. All the groups had similar enclosures (newspaper bedding with egg crate hides).

At the beginning I weighed all of the groups. I weighed them weekly for three more weeks. At the end I calculated how much the groups grew in percentages.



Figure 1: Weights of cockroach groups over time.

Table 1: Temperature and weight measurements over time of cockroaches in four treatment groups: treatments groups A–C (16 individuals each) and the control group (18 individuals).

Group	Date	Temperature (°C)	Group Weight (g)
A	2/12/18	26.1	2.0
A	2/18/18	26.1	2.7
A	2/25/18	26.1	3.2
A	3/4/18	23.9	4.1
B	2/12/18	26.1	2.0
B	2/18/18	26.1	2.4
B	2/25/18	26.1	3.0
B	3/4/18	23.9	3.9
C	2/12/18	26.1	1.5
C	2/18/18	26.1	2.3
C	2/25/18	26.1	2.8
C	3/4/18	23.9	3.7
Control	2/12/18	21.1	2.0
Control	2/18/18	20.6	2.2
Control	2/25/18	18.9	2.3
Control	3/4/18	16.1	2.2

Results and Discussion

My hypothesis was correct. In just three weeks the heated groups grew 112.73% compared to the unheated group which only grew 10% (Figure 1, Table 1). That means that the heated groups grew over eleven times more than the unheated group.

My conclusion is that heat is one of the most important things to an insect's growth. This is why you usually see insects during the summer in Alaska, they have to hibernate or migrate to survive the winter. The closer you get to the equator the more insects you will see. To have a successful breeding colony of cockroaches you should keep

them warm, which I do by keeping them on heating pad. I am not worried about the Dubia roaches breeding in my house if they escape from their tank, because they do not grow well at room temperature.

You may be wondering why I chose Dubia roaches for the growth project over other insects. One reason is Dubia roaches are super hard to kill. They are easier to keep than other insects. Another reason is because they are one of the easiest insects to get. Finally, I picked them because they grow fast enough to see a difference while we did this experiment. Also, they make a good example insect, even for Alaskan insects.

Review of the eleventh annual meeting

by Adam Haberski¹



Figure 1: Members present at the end of the meeting. Back row, from left: Stephen Burr, Isaac Davis, Garret Dubois, Jason Moan, Michael Baldwin, Alexandria Wenninger and Mary Wyatt . Front row, from left: Derek Sikes, Matt Bowser, Kathryn Daly, Jacquelyn Schade, Roger Burnside, Renee Nowicki, Jessie Moan, Dan Bogan, Adam Haberski and Robin Andrews.

The eleventh annual meeting of the Alaska Entomological society was held at the Anchorage Cooperative Extension office on February 3rd, 2018. We are grateful to Jessie Moan for offering us this space.

Presentations

Jacquelyn Shade began with an update on the Alaska CAPS program. Matt Bowser then serenaded us with his musi-

cal presentation "Biomonitoring bugs by molecules: Slikok Creek." Matt not only sang his presentation but accompanied himself on guitar. Derek Sikes followed with an overview of the staphylinids of Alaska. Jason Moan then presented "Spruce beetle status in Southcentral Alaska," and Stephen Burr presented "Forest Health Conditions Report 2017."

We were fortunate to have five student presentations this year. Alexandria Wenninger presented her master's

¹University of Alaska Fairbanks, Fairbanks, Alaskaahaberski@gmail.com

thesis, “Predatory hymenopteran assemblages in boreal Alaska: associations with forest composition and post-fire succession.” I presented on my observations of pseudoscorpion in Denali National Park, Kathryn Daly presented on the morphological impact of climate warming on Alaskan butterflies, and Robin Andrews presented on soil microarthropod communities. Renee Nowicki gave the final presentation of the day, a “Key to common ants of Alaska.” Renee received the 2017 Kenelm W. Philip Entomological Research Award to produce a dichotomous key to Alaska’s ants and we were excited to hear of her progress. The number of excellent presentations presented a welcomed challenge to the student award committee, and they gave the Student Presentation Award to Alexandria Wenninger. Congratulations, Alex!

Business items—highlights

- The Student Presentation Award has been increased from \$50 to \$100.
- There will be increased cross-reference between our website, Facebook page, and listserv, to ensure content consistently reaches all of our members.
- DOI’s were successfully implemented in our newsletter for all articles related to Arctos specimens, and the newsletter is now open to non-arthropod invertebrate content.
- Election results: Adam Haberski (president), Robin Andrews (vice president), Renee Nowicki (secretary), and Roger Burnside (treasurer).

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



Nelima paessleri, Soldotna, August 14, 2017. Image CC BY Matt Bowser (<https://www.inaturalist.org/photos/12965403>).

