



FLY TIMES

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Welcome to the latest issue of *Fly Times*! As usual, I thank everyone for sending in such interesting articles! I hope you all enjoy reading it as much as I enjoyed putting it together! Its being so late has allowed the issue to be larger than it would have been on time! But that said, the reason for this delay was that I was out of commission from mid-September through the beginning of December due to a motorcycle accident (not to go into the sordid details, but I was t-boned by someone who ran a stop sign). Please let me encourage all of you to consider contributing articles that may be of interest to the Diptera community for the next issue. *Fly Times* offers a great forum to report on your research activities and to make requests for taxa being studied, as well as to report interesting observations about flies, to discuss new and improved methods, to advertise opportunities for dipterists, to report on or announce meetings relevant to the community, etc., with all the associated digital images you wish to provide. This is also a great place to report on your interesting (and hopefully fruitful) collecting activities! Really anything fly-related is considered. And of course, thanks very much to Chris Borkent for again assembling the list of Diptera citations since the last *Fly Times*!

The electronic version of the *Fly Times* continues to be hosted on the North American Dipterists Society website at <http://www.nadsdiptera.org/News/FlyTimes/Flyhome.htm>. For this issue, I want to again thank all the contributors for sending me so many great articles! Feel free to share your opinions or provide ideas on how to improve the newsletter. Also note, the [Directory of North American Dipterists](#) is constantly being updated. Please check your current entry and send all corrections (or new entries) to [Jim O'Hara](#) – see the form for this on the last page.

Issue No. 56 of the *Fly Times* will appear next April. Please send your contributions by email to the editor at stephen.gaimari@cdfa.ca.gov. All contributors for the next *Fly Times* should aim for 10 April 2016 (maybe then I'll get an issue out on time!) – but don't worry – I'll send a reminder! And articles after 10 April are OK too!

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NEWS

Zurquí All Diptera Biodiversity Inventory (ZADBI)

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The past six months has seen important progress and change in our Costa Rican project. By the end of August, the last of our material, including specimens from Tapanti, Las Alturas and some remaining supplementary samples from Zurquí (collected with a variety of traps), was curated by our parataxonomists at INBio. These specimens were brought back to Los Angeles by Brian Brown and Estella Hernandez, who visited Costa Rica on Aug. 24-27 and they will be distributed to our collaborators in the next couple of weeks. This will see the last of the specimens collected by our project sent to our collaborators. Many of our colleagues have identified the bulk of material received earlier and our unofficial species count for Zurquí now stands at 2,529 species, which is pretty astounding for a site that measures only 150 X 260 meters! We expect, however, that number to rise significantly as further groups are interpreted. Brian and Estella also visited with Jorge Arturo Lizano, the owner of the property at Zurquí, again thanking him for so generously hosting our project.

So we are now in the home stretch of this effort, getting flies identified from the tremendous numbers of samples collected during the Zurquí All Diptera Biodiversity Inventory project. Over the coming months, we and our collaborators will finish identifying all the material at hand, using our web-based identification spreadsheets, giving us final numbers for each of the families and some estimates of beta level diversity. Our plan is to prepare a big splash publication this coming year providing an overview of the diversity of Diptera discovered by our project.

On a sad note, funding for the project ended in August, and we had to let our incredibly talented staff go at INBio. The National Museum of Costa Rica has now taken over responsibility for the collection previously held by INBio, but unfortunately has not been able to hire new personnel to further develop and care for it. Certainly it is a shame that the very talented and knowledgeable personnel at INBio haven't found employment that would allow them to continue to help interpret the biodiversity of Costa Rica. It is a tremendous loss for us all.

A brief update on the current status of the INBio Collection

Manuel Zumbado

As most of you probably know the INBio Collection was transferred to the Museo Nacional de Costa Rica (MNCR) on March 27, 2015.

The INBio Collection is now under control and managed by the Departamento de Historia Natural, Museo Nacional de Costa Rica. The collection is hosted in the same building, which now belongs to Agriculture. It is expected to remain there for years to come. The officials in charge of the collection are as follows: Directora (General Director): Sra. Rocío Fernández (dirección@museocostarica.go.cr). Jefe Departamento de Historia Natural (Head of Natural History Department): Cecilia Pineda (cpineda@museocostarica.go.cr). Encargado de Préstamos (Collection Manager): Armando Ruiz (aruiz@museocostarica.go.cr). Encargada de la Sede Santo Domingo (Museum Officer at Santo Domingo): Silvia Lobo (slobo@museocostarica.go.cr). Curador de Entomología (Insect Curator): German Vega (gvega@museocostarica.go.cr).

General correspondence must be directed to Mrs. Fernández. More specific correspondence and specially loan return or loan requests must be directed to Mrs. Pineda and Mr. Ruiz. The collection is open for visiting scientists. The working conditions, space, equipment, etc. are similar. Lending material at the time can be difficult o take long.

INBio is still providing Internet services to the collections building and staff. The Atta system is running and maintained by INBio. My contract with INBio ended up on November 23th, but I keep as an associated and I am in the process to become a Research Associate to Museo Nacional de Costa Rica.

Preliminary results of the Mitaraka expedition (French Guiana)

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In the October 2014 issue of Fly Times (Pollet *et al.* 2014) the Mitaraka expedition in southwestern French Guiana was introduced and everybody with interest in the dipteran fauna of this region was invited to collaborate. In short, the expedition took place between February 23 and March 26 by two subsequent teams of researchers and supporting staff. The first two authors carried out field work during a first 2.5 weeks period, and spent one more week in Cayenne to start sorting the collected samples. However, the idea of disseminating the dipteran fractions later on in the spring of 2015 was obviously too ambitious ... In the present contribution, we gladly present the current state of the art and some preliminary results.

Introduction

On February 23, Julien and I joined our primarily French colleagues on a flight from Paris to Cayenne, from where we flew the next morning to Maripasoula, a large village half way the country along the Maroni River that separates French Guiana from Suriname. Later on that day, a helicopter brought us in about 45 minutes to our final destination (Fig. 1), which was - without any exaggeration – “rather remote”. Next to the drop zone that had been created by cutting down rain forest and enabled us to land, we noticed upon arrival that the camp crew had done a terrific job by professionally putting up “carbets-bâches” for field laboratories, dormitories, a kitchen and an outdoors dining room. We slept in hammocks with according mosquito nets (Fig. 2). The latter were no superfluous luxury as it appeared that sand flies (Phlebotomidae) infested the place (as so many places in French Guiana). To get into one’s hammock at night without these undesired guests proved a true challenge. During and after the expedition and despite all precautions, a number of colleagues caught Leishmaniasis.

As was mentioned before (Pollet *et al.*, 2014), next to collecting Diptera, and Dolichopodidae in particular, I was also responsible for the processing of all Diptera collected during this expedition. More than 20 different sampling techniques were used, and within a perimeter of 1 km² no less than 401 different traps were installed, mainly to collect invertebrates (but all other biota were also inventorized). I mainly employed colored pan traps (Fig. 3) (with formaline solution and detergent) that were in operation between 9 and 12 days. However, my survey did not go as smooth as I had hoped for, due to the almost daily heavy rainfall in the afternoon. And after a few days, I was forced to gather the yields and refill the traps, this time with a salty water solution with detergent. Of course, only in those cases where I could relocate my “assiettes flottantes” (as my pan traps were so kindly called by my peers ...).

In a preparatory phase (January 2015), the camp crew cut four trails of 3.5 kms in four different directions (Fig. 5) along which the researchers could select their sampling sites. Different habitat types were found along these trails: steep forested hills with palm swamps (*bas fonds*) and rivers in between,



Figure 1. Helicopter on the airport of Maripasoula (French Guiana).



Figure 2. Field dormitory in the Mitaraka base camp.

and sparsely distributed smaller (*savanes roches*) and larger rounded rocky formations (*inselbergs*), some of which stood out hundreds meters above the surrounding lowland rain forest. Climbing the latter was very hard but the views on the top were truly rewarding (Fig. 4). Actually, these surprisingly large rocks present a totally different environment as compared to the rain forest, and experience more extreme weather conditions (hot and dry during the day, colder and damp during the night and often very windy).

1 Field work

Between February 24 and March 10, 2015 a total of 280 (70 white, 100 yellow, 110 blue) **pan traps** were in operation in 11 different sites (Fig. 6). The initial idea of investigating a hill top (*plateau*), slope (*pente*), and swamp habitat (*bas fond*) along each of the four trails was left very soon in the expedition as hardly any dolichopodid flies were observed in the two first habitat types. Moreover, installing and maintaining 12 widely separated sampling sites proved not feasible. All three habitats were thus investigated only along trail A and C, and 3 additional swamps and one river bank were selected for sampling near the base camp; the 11th site was situated in the forest bordering the drop zone (DZ). During servicing, in each site yields from all (5 or 10) traps of the same colour were pooled to one sample. Sampling with pan traps accounted for 3,161 trapping days (trapping day = a period of one day during which a trap is operational in the field) and yielded a total of 64 pan trap samples (Table 1).

Table 1. Summary of the pan trap survey in Mitaraka (French Guiana).
BPT: blue pan traps, WPT: white pan traps, YPT: yellow pan traps

sampling site cd *	habitat type	BPT	WPT	YPT
MIT-A-RBF1	palm swamp	10	10	10
MIT-A-RBF2	river bank	10	-	-
MIT-A-SL	slope	10	10	10
MIT-A-TOP	hill top	10	10	10
MIT-C-RBF1	palm swamp	10	10	10
MIT-C-RBF2	palm swamp	10		10
MIT-C-SL (MIT08)	slope	10	10	10
MIT-C-TOP (MIT07)	top hill	10	10	10
MIT-DZ1	top hill-slope	5	5	5
MIT-DZ2	top hill-slope	5	5	5
MIT-DZ-RBF1	river bank	10	-	10
MIT-DZ-RBF2	palm swamp	10	-	10
Total no. traps		110	70	100
Total no. samples		24	16	22

* A and C refer to trail codes, DZ = drop zone, TOP = hill top, SL = slope, RBF = river bank forest, MIT08 and MIT07 refer to DIADEMA-sites

Next to the pan traps, also a **sweep net** was used to collect Dolichopodidae and other Diptera, and sweeping was mainly conducted in sites where Dolichopodidae were visually observed and fairly abundant, such as palm swamps and river and creek banks but also in humid spots on savanes roches and inselbergs. A total of 101 sweep net samples were gathered.

Finally, Diptera were also collected and subsequently retrieved from a number of **other collecting devices** that were primarily used to collect Coleoptera.



Figure 3 (left). Coloured pan traps in a palm swamp at Mitaraka.

Figure 4 (right). View from the top of Borne 1, an inselberg in southwestern French Guiana.

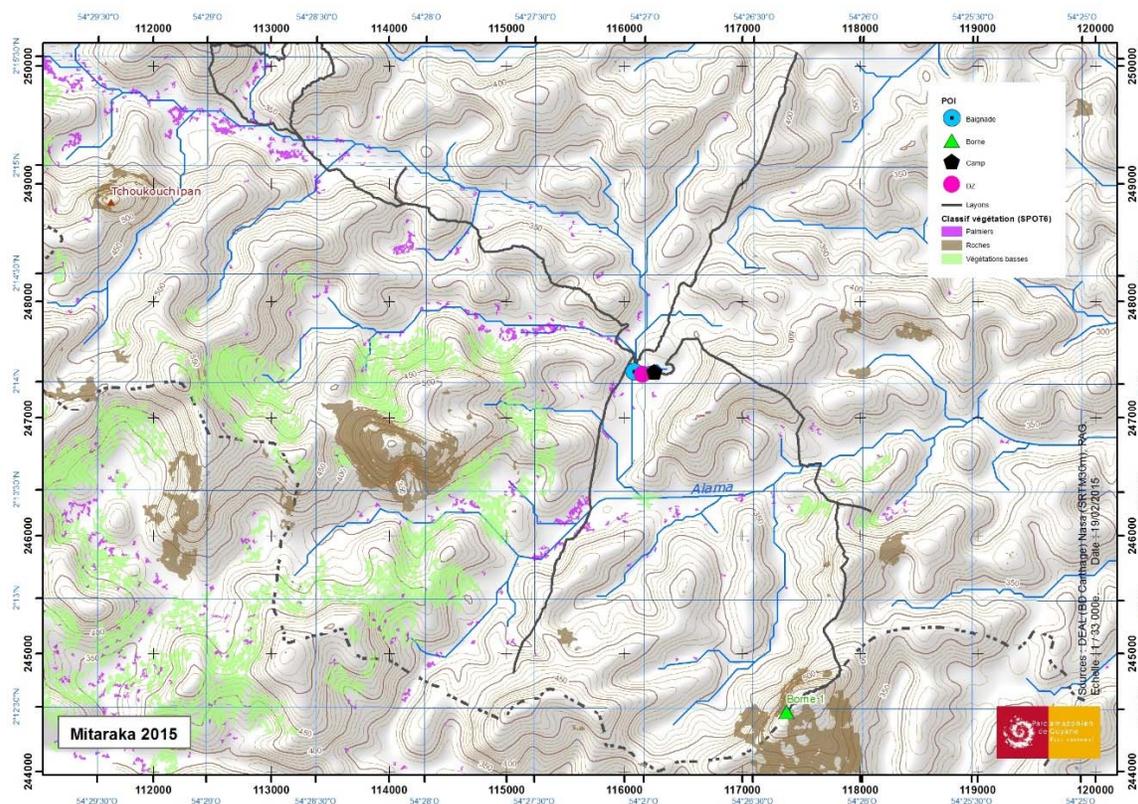


Figure 5. Site map with four trails indicated (map produced by Maël Dewynter).

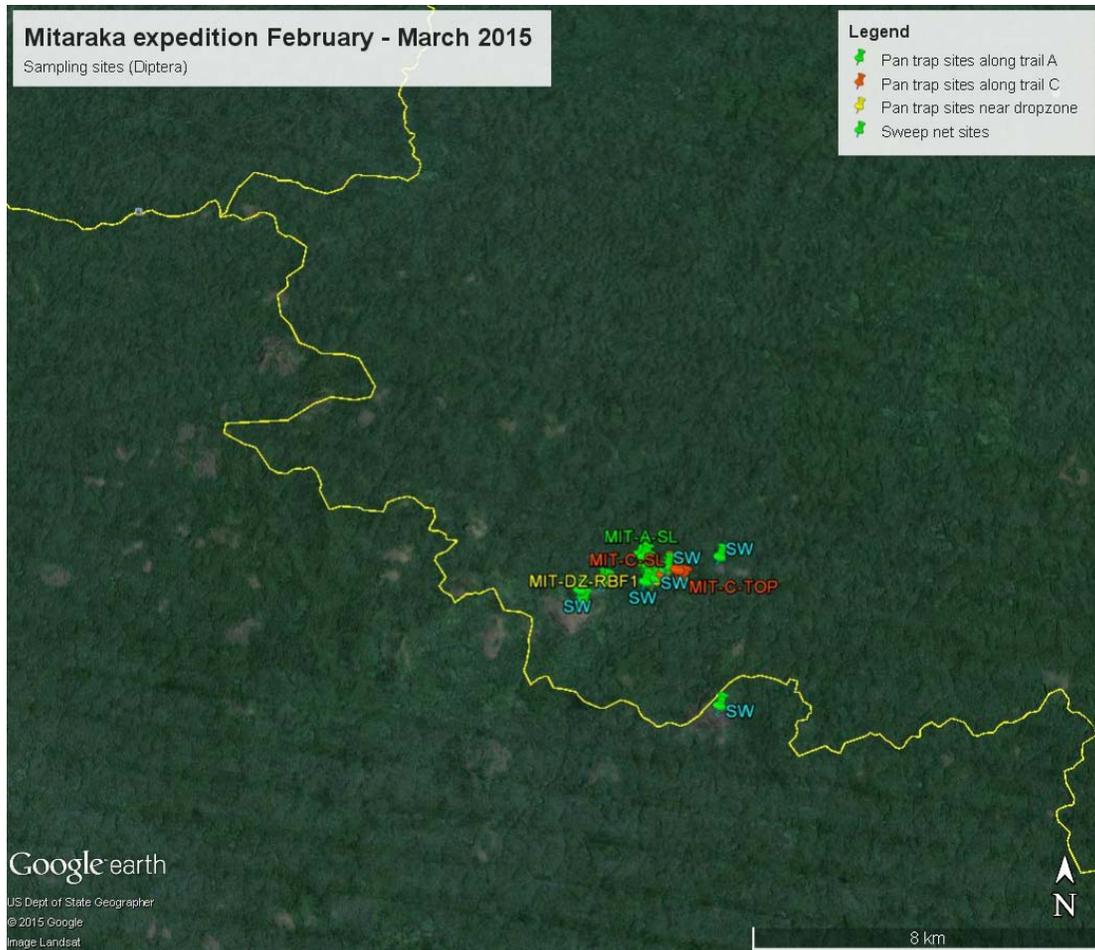


Figure 6. Pan trap and sweep net sampling sites within the Mitaraka area.

2 Sample processing

The bulk of the specimens was collected with traps and stored in alcohol solution. In addition, also some - mostly larger - fly specimens were preserved unmounted in insect envelopes and are currently stored in a freezer. The latter fraction has not been considered in the discussion below.

During March 11-18, at SEAG (Société Entomologique des Antilles et de Guyane (Cayenne, French Guiana); <http://insectafgseag.myspecies.info/fr>), Diptera and other invertebrates were retrieved from all SEAG samples collected during the first period of the expedition. The bulk of the samples (including sweep net and pan trap samples) were processed in the Belgian lab in June 2015. During this phase, the following 4 fractions were sorted and separately stored: Dolichopodidae, other Empidoidea, remaining Diptera and Coleoptera. The residue samples thus contained all other invertebrates. During September – November 2015, the “remaining Diptera” samples were split up further into family or superfamily subsamples for which I could engage a taxonomic expert.

Indeed, before I embarked upon this expedition, over thirty taxonomic experts (in other words: you!) already expressed their interest to study the samples and more recently, another four people joined the team (Table 2), raising the number of experts involved to 38! Unfortunately, Jorge Almeida (Portugal), Torsten Dikow (US), nor Michal Tkoc (Czech Republic) will receive samples as the following fly families remained absent from the samples: Acroceridae, Nemestrinidae, Mythicomyiidae, Mydidae, Platypezidae, Opetiidae, and Lonchopteridae.

Sample processing produced over 1,900 subsamples containing 28 fly families and superfamilies (Table 2), including the impressive Pantophtalmidae (Fig. 7). Other dipteran families were stored together in 216 samples. If anybody might be interested to study one of these families, just get in touch with me.

In August 2015, the Mitaraka site was visited once more by a smaller team of researchers and this survey produced another set of samples. I am currently working my way through a huge Malaise trap sample and upon completion, the dissemination of all samples will be started.

Table 2. List of separated dipteran families and superfamilies with number of subsamples and associated Diptera workers

Dipteran fraction	no. subsamples	coordinator (coord.)/ taxonomic expert
Agromyzidae	5	Stephanie Boucher (CANADA)
Asilidae	42	Rodrigo Marques Vieira (BRASIL)
Bibionidae	18	Woody Fitzgerald (US)
Bombyliidae	5	Carlos José Einicker Lamas (BRASIL)
Ceratopogonidae	99	Art Borkent (and Gustavo Spinelli, AR) (CANADA)
Chloropidae	112	Terry A. Wheeler (CANADA)
Dolichopodidae	175	Marc Pollet (BELGIUM: coord.), Justin Runyon (US: Enliniinae)
Drosophilidae	123	Gabriela Pirani (BRASIL)
Empidoidea	108	Christophe Daugeron (FRANCE: coord.), Brad Sinclair (CANADA), Josemir Teixeira Câmara (BRASIL), Adrian Plant (UK), Rosaly Ale-Rocha (BRASIL), Rafael Augusto Pinheiro de Freitas Silva (BRASIL), Patrick Grootaert (BELGIUM)
Lauxaniidae	122	Steve Gaimari (US)
Lygistorrhinidae	14	Vladimir Blagoderov (UK)
Micropezidae	92	? Steve Marshall (CANADA)
Muscoidea	140	Claudio José Barros de Carvalho (BRASIL: coord.), Terry Whitworth (US), Pierfilippo Cerretti (ITALY)
Mycetophilidae	108	Chris Borkent (US)
Pantophtalmidae	2	-
Phoridae	107	-
Pipunculidae	6	José Albertino Rafael (BRASIL)
Psychodidae	54	Gregory Curler (US)
Scatopsidae	11	Jean-Paul Haenni (SWITZERLAND)
Sciomyzidae	6	Jonas Mortelmans (BELGIUM)
Sepsidae	45	Vera Cristina Silva (BRASIL)
Sphaeroceridae	91	Steven Paiero (CANADA)
Stratiomyidae	55	Diego Aguilar Fachin (BRASIL)
Syringogastridae	12	José Albertino Rafael (BRASIL)
Syrphidae	39	Menno Reemer & John Smit (NETHERLANDS)
Tabanidae	42	Tiago Krolow (BRASIL)
Tephritoidea	69	John Smit (NETHERLANDS), Allen Norrbom (US)
Tipuloidea	85	Jorge L. Mederos López (SPAIN), Herman de Jong (NETHERLANDS)
miscellaneous Diptera *	216	-
Total no. subsamples	2003	

* Including a.o. Anisopodidae, Athericidae, Cecidomyiidae, Chironomidae, Culicidae, Ephydriidae, Neriidae, Rhagionidae, Sciaridae and Xylophagidae



Figure 7. Representative of the family Pantophtalmidae (picture: Rémy Pignoux).



Figure 8. *Cheiromyia pennaticornis* (Parent, 1931), a typical species of palm swamps in Mitaraka.

3 Identification

Collaborators are asked to provide IDs on the specimens in the samples within one year upon receipt of the samples. In the current case, this implies reporting by the end of December 2016.

Part of the dolichopodid samples has already been examined, including (the richest) sweep net samples and a representative selection of samples from other trapping techniques. With 2,197 specimens from 96 samples (or 55% of all samples) treated, a total of 157 different species could be established. The examination of the remaining traps will undoubtedly further increase this number, but will not double or triple it. The majority of the species is represented by singletons (n=74) or doubletons (n=22), and among the 22 species with over 10 specimens collected, 14 belong to the subfamily Dolichopodinae.

As observed in previous surveys, a high complementarity was apparent between the different collecting techniques, even those executed in the same sites. Collecting by sweepnet yielded 89 or nearly 57% of all species, 69 of which were only collected with this technique.

Three genera proved particularly speciose: *Chrysotus* (31 sp.), *Medetera* (25 sp.) and *Paraclius* (52 sp.). In particular, the diversity in the latter genus, *Paraclius*, is unprecedented and has not been observed in any other site in the Neotropics thus far. This genus seems to occupy similar habitat types as the related *Dolichopus* in the northern hemisphere which is often also quite species rich in marshy biotopes.

Four of the 157 species have been described (*Cheiromyia pennaticornis* (Parent, 1931) (Fig. 8); *Coeloglutus concavus* Aldrich, 1896; *Neotonnoiria maculipennis* (Van Duzee, 1929); *Neurigona brevitibia* Naglis, 2003), and it cannot be excluded that some of the other species might turn out described as well. However, the huge majority of the species will undoubtedly prove new to science. Moreover, four new yet undescribed genera have also been discovered with at least 8 new species.

References

Pollet, M., Pascal, O. & Touroult, J. (2014). Flies from French Guiana: a unique opportunity. Fly Times 53: 3-6.

Enzymatic clearing agents as an alternative approach to macerating Diptera specimens

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The clearing, or maceration, of fly abdominal structures to allow the study of taxonomically important features of male and female genitalia has long been a routine activity in dipterology research laboratories. The standard chemicals commonly used for clearing are strong bases (potassium hydroxide or sodium hydroxide) or lactic acid, but each of these approaches has significant drawbacks. Best results are obtained by heating, which increases the hazards associated with caustic chemicals. Potassium hydroxide (KOH), which we have routinely used for many years, has produced the best results in terms of clearing and flexibility, but must be carefully rinsed and neutralized with weak acids to prevent long term damage to the material. Many dipterists have found that specimens cleared in KOH continue to clear when stored in glycerin, even when rinsed and neutralized, and have thus switched to lactic acid in the interest of long-term archiving of genitalic preparations. Lactic acid, however, has disadvantages as well. Hot lactic acid creates corrosive and dangerous fumes, and specimens heated in lactic acid are prone to expansion and deformation. Cold lactic acid (in our opinion) does not give good results. With these issues in mind, we recalled an arachnologist mentioning (about thirty years ago!) that he had used contact lens cleaner (enzymatic protein removers) to clear spider pedipalps, so we bought some ordinary drug store contact lens cleaner to test its effectiveness for macerating fly genitalia. At the same time, we wondered if laboratory grade trypsin might yield similar results so we set out to assess the use of both sources of enzymatic cleaners for clearing fly abdomens. The results, summarized below, have resulted in the adoption of this approach as our preferred protocol for specimen preparation. We feel that the improvements in laboratory safety, increased quality of preparations, and expected long-term integrity of the macerated material are substantive and worth sharing with the community.

The foremost advantage is safety. Enzymatic cleaners are safe agents because they are not caustic, they do not require heating and thus do not produce fumes, and they do not create hazardous chemicals/wastes. Secondly, enzymatic cleaners only act to break down protein; they remove muscles and soft tissue without clearing sclerites and membranes. Lightly sclerotized cuticle, connective tissues, membranes, and ducts are preserved. Stored dissected specimens macerated in enzymatic cleaners will not indefinitely clear over time for the following reasons:

1. Enzymatic cleaners only target proteins, once all soft tissues are digested, the reaction will stop. Sclerotized structures will not be affected.
- 2.
3. Enzymes degrade and the potency declines over time, thus clearing will not continue after initial maceration.

We placed critical point dried and air dried entire abdomens and abdominal parts of Micropezidae and Sphaeroceridae directly into contact lens cleaner solution (mixed according to package directions) or trypsin (0.5%) EDTA solution in small plastic vials ("cryovials") of 1.5 to 2 ml for clearing. We obtained good results with clearing times from 12 to 16 hours at room temperature, or significantly shorter periods when the vials were placed in an ultrasonic cleaner (vibration seems to aid in removing soft tissues). Larger specimens were partially dissected to allow the solution to enter and contact

internal tissues. In some cases specimens were removed from the enzymatic cleaner solution, rinsed in distilled or deionized water to remove partially digested tissues and returned to the enzymatic solution for further digestion.

We found that enzymatic cleaners produced results equal or superior to macerations performed with lactic acid or potassium hydroxide. All sclerites were clearly visible; weakly sclerotized internal structures were well preserved, and female internal genitalic structures needed minimal to no staining. This technique also preserves the elasticity and rigidity of the membranes and sclerites, unlike lactic acid preparations where dissections are often brittle, or NaOH or KOH preparations where sclerites are softened and tergites often curl inwards.

Although enzymatic cleaners are considerably slower when compared to other agents, specimens can be left to clear overnight with no worries about damaged specimens. The main disadvantage of enzymatic cleaners is their rapid breakdown with use. Each contact lens cleaning tablet (or 2 ml vial of trypsin solution) can be used to clear only a few specimens before it must be discarded and replaced. This translates to a minor inconvenience and a cost of about 50 cents to clear a few specimens with a contact lens cleaning tablet, and about 30 cents with trypsin. Well worth it, in our opinion!

We would welcome comments from others as we continue to look for better ways to adapt this technique to our needs. The lens cleaning tablets we used were "Unizyme", bought in a box of 12 blister-packed tablets that still functioned after storage at room temperature for one year.

Potential impacts of a hurricane storm event on tree divot habitat for larval mosquitoes in central New Jersey: Hurricane Sandy case study

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Introduction

It is undeniable that as dynamic as the earth's climate is, natural ecosystems adapt as the existing climate conditions change over time (Reiter 2001; Kovats et al. 2001). However, climate changes that are rapid such as an increase in global temperature as well as the frequency and intensity of storm events solicits the question about how natural ecosystems will respond (Kovats et al. 2001; Githeko et al. 2000). Different climate change scenarios may alter the ecological balance and context for the biology and ecology of arthropod vectors as well as the hosts; thus, subsequently influence the risk of transmission (Githeko et al. 2000; Patz et al. 2000; Kovats et al. 2001). Because climate, vector ecology, social economics vary from one region to another due to scale issues, it is important to examine the extract case studies to demonstrate such regional differences (Githeko et al 2000).

Catastrophic weather events such as tornados and hurricanes can provide ideal conditions for significant increases in mosquito habitat and larval populations associated with subsequent disease outbreaks that follow these events (FEMA 2006). Hurricane Sandy or unofficially known as "Superstorm Sandy" was one of the most destructive and lethal hurricanes to hit landfall in the 2012 with winds spanning 1100 miles (Washington Post 2013). High winds and heavy rain during such intense storm events can create unique larval mosquito habitats as a result of tree blow down in heavily wooded areas (CDC 1993). Forest habitat in the path of intense storm events with heavy winds is at risk of generating temporary larval habitat for mosquitoes. Tree blow downs or as we term them, tree divots (Fig. 1) caused by the uprooting of trees provide stagnant pools with few predators. These conditions can allow large numbers of mosquito larvae to develop over a short period of time (Gaines 2015). As part of another study, we examined the tree divot habitat created post Hurricane Sandy within Teetertown Nature Preserve (TNP) in Port Murray, New Jersey. The objective of this article is to report baseline results of the physical attributes of tree divots, mosquito densities within these divots and spatial data designed to elucidate the potential abundance of tree divot habitat for mosquitoes within Sandy's path in New Jersey.

Methods

We surveyed a 15.4 acre area of wooded forest for tree divots caused by tree falls associated with Hurricane Sandy. From a subset (n=21) of that population, we monitored mosquito density, water volume and depth, pH, and temperature over the duration of the ephemeral divots. A 250 ml mosquito dipper was used to determine the presence or absence of larval mosquitos. Larval density was determined by taking five dip samples from each divot and pooling these samples for a total number of mosquitos per 1250 ml. Water volume was calculated by taking measurements of the length and width of the divot and then multiplying the area by the average of depth measurement from the divot. The pH and temperature were determined with hand-held portable probes (Etekcity Handheld pH Meter +/- 0.05 and Etekcity infrared thermometer +/- 1° F). The diameter of the tree was determined by measuring the trunk diameter at breast height (DBH) or 1.4 m from the base of the tree. For divots that were the result of more than one tree, the diameter of each tree was measured then averaged together to get a single measurement.



Figure 1. Example of a tree divot caused by Hurricane Sandy (Photo Credit: Greg Vaccarino)

The estimated tree divot density was obtained by surveying a 250m X 250m area for the presence of divots. These data were then compared to aerial imagery (NJDEP ground accuracy +/- 4 ft) prior to Hurricane Sandy and pre-existing tree divots were removed from the sample. The pre-existing divots were removed from the sample to obtain a clearer estimate of the potential impacts of a single Hurricane Sandy sized storm event. Data for land use/ land change (2012) was obtained from New Jersey Department of Environmental Protection, Office of Information Resources Management, Bureau of Geographic Information Systems (NJDEP 2015). The minimum size map unit was one acre for the differentiation of land use type. Features mapped from digital imagery had a ground accuracy of +/- 4 ft. The percentage of the total land for each management area that was forested was determined by using the sum of all forest types divided by the total area of the management area. The total heavily forested area for each management area was determined by summarizing all forest types with a crown cover greater than 50%. The density of tree divots from our sample site was then used to extrapolate the potential number of divots for all the heavily forested area within each watershed management area. Geospatial analysis was done using ArcGIS (ESRI Software, Redlands CA).

Results

Aedes stimulans (Walker) larvae were the only mosquito species collected in the tree divots throughout the duration of the study. The mean mosquito density increased over the duration of the

study from 9 - 144 mosquitoes per liter (April 3 - May 8, 2015) (Fig. 2). The mean water volume and depth decreased over the duration of the study while the temperature and pH increased (Fig. 3A-D). All of the tree divots studied dried and could no longer support aquatic mosquito larvae by May 12, 2015. The average maximum volume of the tree divots observed in this study was 322 L (SE = 2.99). A total of 41 Hurricane Sandy induced tree divots were present in the 15.4 acre sample area (Fig. 4). We found our survey site at Teetertown Preserve to have a density of 2.65 tree divots/acre. In the divots we surveyed, we found no relationship between the DBH of the tree and the size of the tree divot (Fig. 5). The mean pit area was 1.3 m² (SE = 0.28). The mean pit volume was 0.322 m³ (SE = 0.13). The mean DBH was 0.57 m (SE = 0.02).

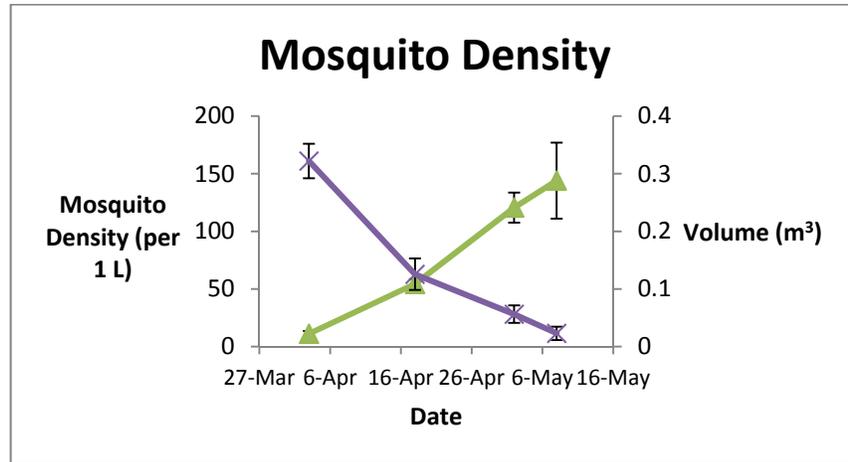


Figure 2. Comparison of mean mosquito density and mean water volume for tree divots collected March-May 2015, Teetertown Nature Preserve, Port Murray, NJ. Error bars represent SEM.

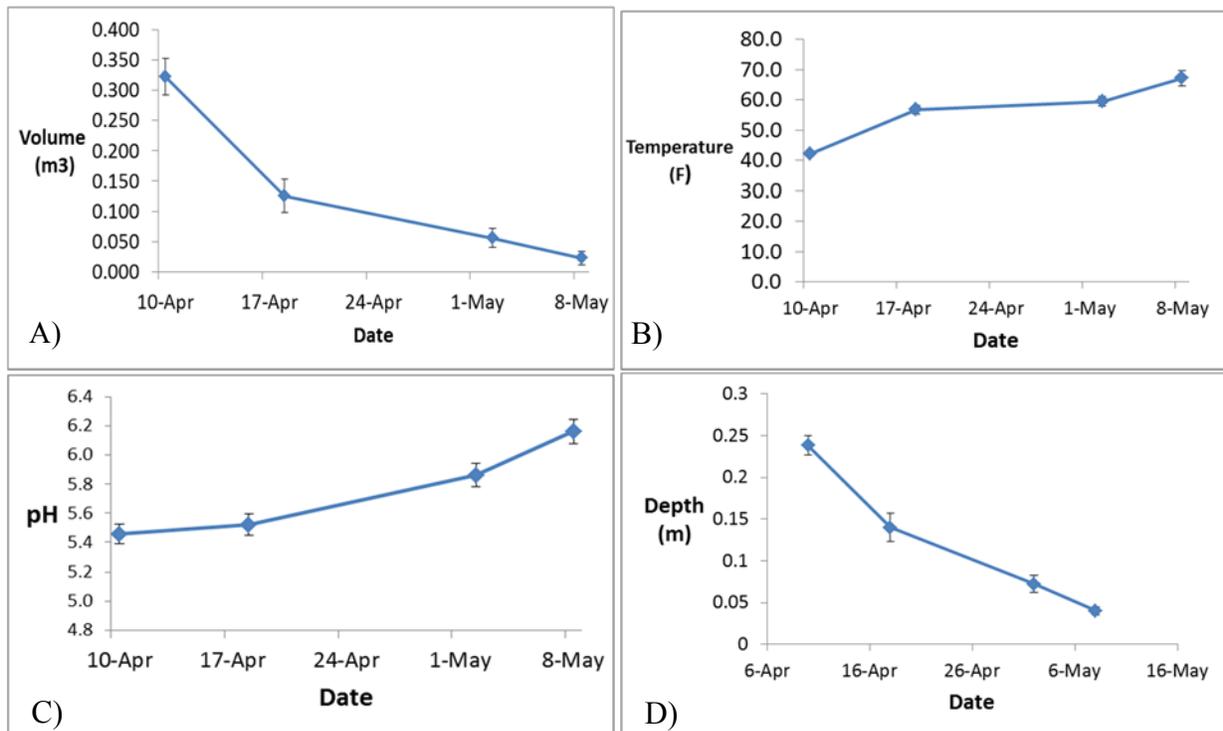


Figure 3. Mean physical/chemical attributes for a subset of 21 tree divots. All measurements were plotted by date. Error bars represent SEM.

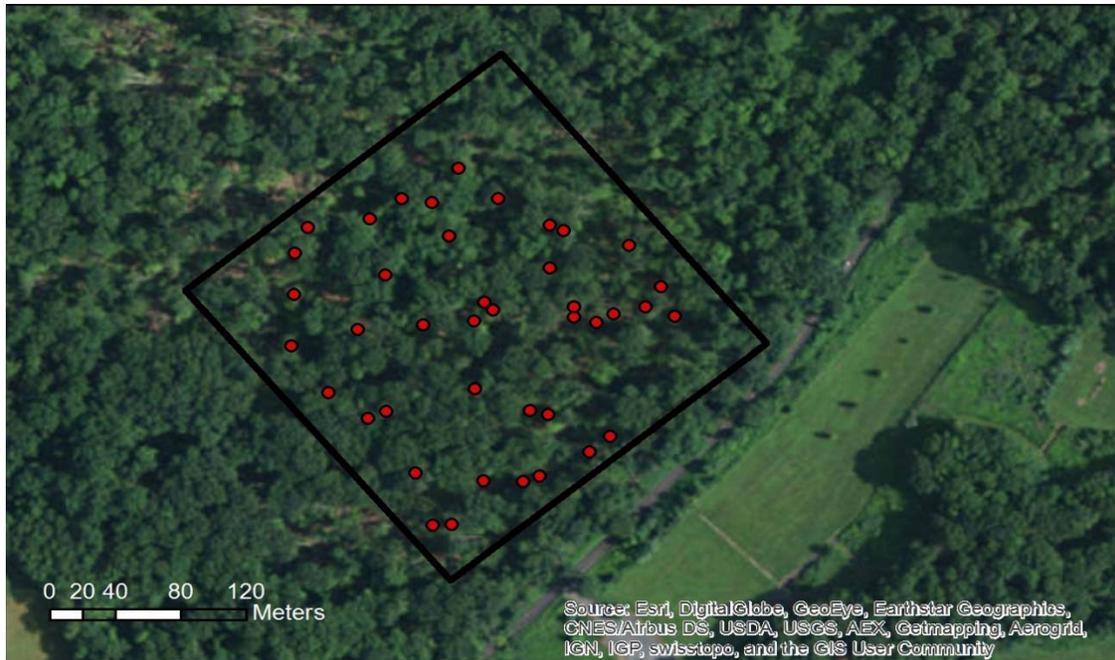


Figure 4. Location of 41 Hurricane Sandy induced tree divots within survey site at TNP. Sample area was 250m X 250m. Tree divots are represented by red dots.

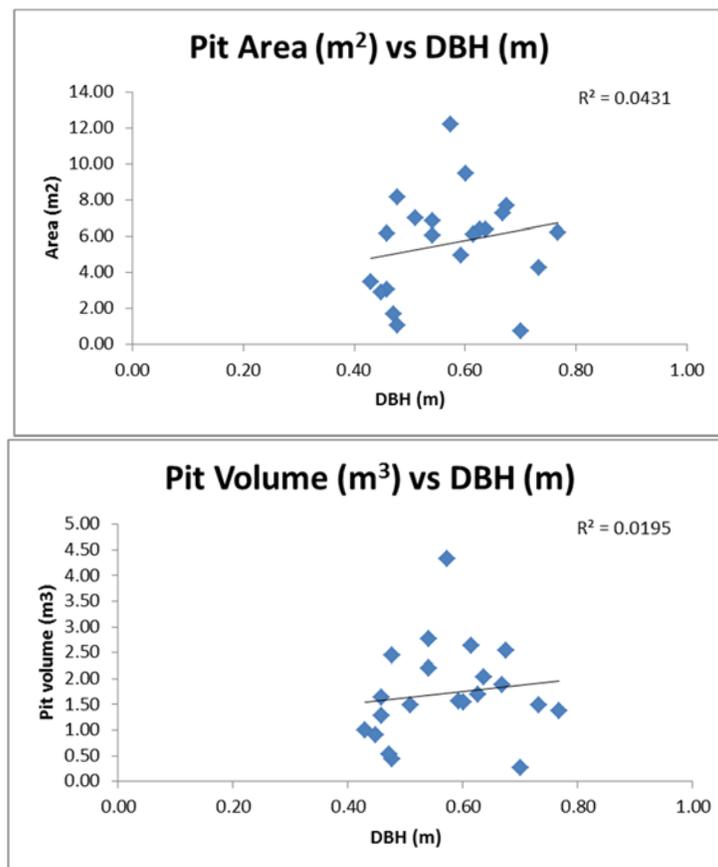


Figure 5. Pit area (top) and pit volume (bottom) plotted against tree diameter at breast height (DBH).

The percent of each watershed management area that was forested and the potential increase of tree divot mosquito habitat are shown in Table 1 and Fig. 6. In general, watershed management areas with large amounts of high percentage of forested area (e.g., area # 01, 14, 15, and 17) have a greater potential for increased mosquito habitat than smaller and less forested management areas (area # 04, 05, 07 and 20) (Table 1).

Table 1. Potential tree divots by watershed management area.
WMA corresponds with location in Figure 6.

WMA	Name	Potential Divots
01	Upper Delaware	663021
02	Wallkill	169062
03	Pompton, Pequannock, Wanaque, Ramapo	229884
04	Lower Passaic and Saddle	32085
05	Hackensack, Hudson, and Pascack	28507
06	Upper Passaic, Whippany, and Rockaway	194807
07	Arthur Kill	20396
08	North and South Branch Raritan	277561
09	Lower Raritan, South River and Lawrence	92101
10	Millstone	102722
11	Central Delaware	123684
12	Monmouth	82746
13	Barnegat Bay	370830
14	Mullica	487827
15	Great Egg Harbor	374799
16	Cape May	70181
17	Maurice, Salem, and Cohansey	371679
18	Lower Delaware	86295
19	Rancocas	204720
20	Assiscunk, Crosswicks and Doctors	69190

Discussion

Most global climate models can simulate El Niño/Southern Oscillation (ENSO) phenomena, supporting the finding that ENSO responses to global warming may cause serious climate and ecological consequences (Latif and Keenlyside 2008). In North America, not only have average daily temperatures been increasing (Karl et al. 1995), however, recent studies indicate that extremes in the hydrologic cycle have been increasing with more frequent heavy precipitation events involving both rain and snow over the last 30 years (Karl et al. 1995; Groisman and Easterling 1994). Tree divots created by Superstorm Sandy were an abundant ephemeral aquatic system in the study area and were filled with snow or rain thus creating an increase in mosquito habitat. Surprisingly, only one species of mosquito was collected from tree divots. We suspect that the shorter than typical life-span of ephemeral pools in this preserve prohibited the hatching or survival of other mosquito taxa.

Aedes stimulans is a common inhabitant of vernal pools or ponds in New Jersey (Crans 2014). The growth and development of *Ae. stimulans* can be slow early in the season due to these life history parameters being highly correlated with water temperature and food limitation (Wallace and Walker 2008). We noticed that by May 8, all tree divots were dry and few to no pupae were observed prior to May 8, 2015. While we did not determine adult survival from tree divots, we believe it was very low. Due to the small area of tree divots and lower volumes compared to established vernal ponds, we suspect that the aquatic phase of tree divot habitat is shortened comparatively to vernal ponds which were much larger and longer-lived than divots (though not studied in this project) thus limiting the abundance of emerging adult *Ae. stimulans* that would enter the terrestrial ecosystem upon emergence.

We hypothesize that the vertical root mass, soil and accumulated organic matter in these divots may attract spring *Aedes* mosquitoes via certain cues that invite adult oviposition, however, due to the brief aquatic phase of these divots, they may possibly serving as sinks for overwintering eggs. The determining factor in this hypothesis is in what form the precipitation takes to fall.

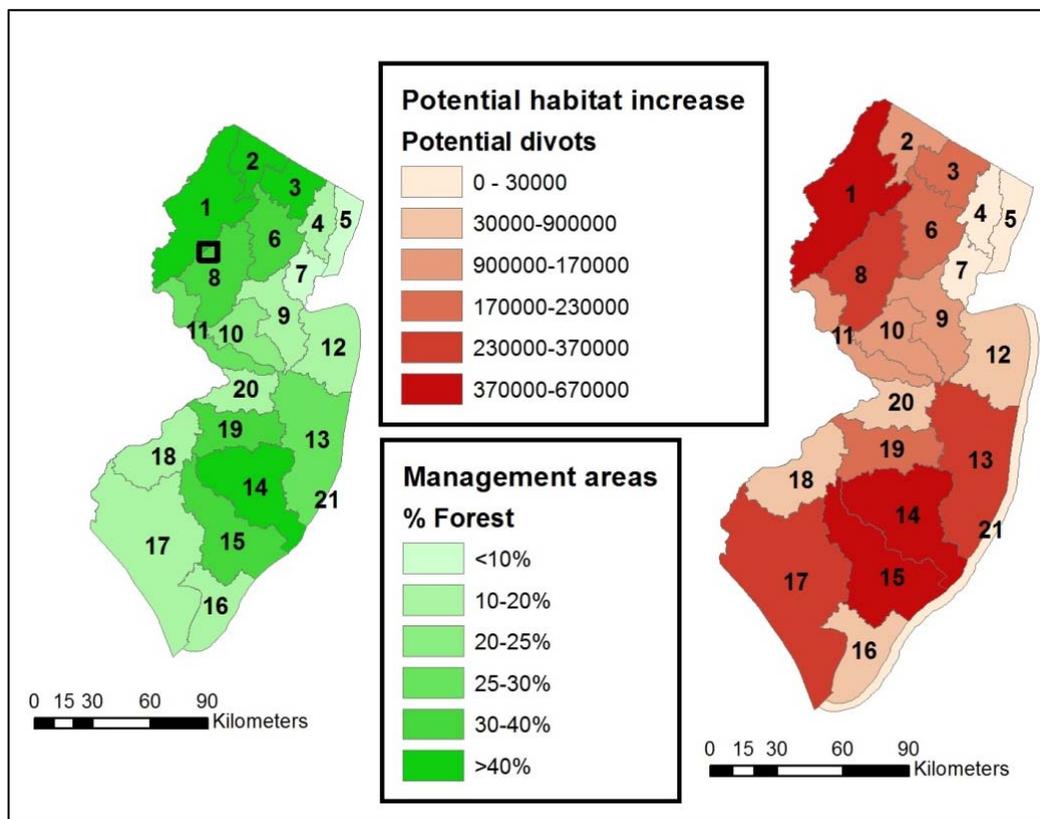


Figure 6. Forest as a percentage of total land for each watershed management area in New Jersey and the potential mosquito tree divot habitat increase. Location of Teetertown Nature Preserve is denoted by box on left map (sample site not to scale).

Forecasters predicted that an El Niño year would develop by March and evidence suggests that it has (Tollefson 2014). These predictions suggest that major storm activity may not occur in the Atlantic Ocean potentially not increasing tree divot formation. However, if precipitation amounts do increase, the question with regards to an increased risk to mosquito-borne diseases may be a function of the timing, rate of change of temperature and moisture regimes as predicted with tick-borne diseases (Glass 1993). While our study has elucidated which regions in New Jersey may be a high risk in terms of tree blow downs from future superstorms and hurricanes, we recommend additional research be conducted to determine if mosquito vector life history parameters such as adult survival can be influenced by changing climatic conditions such as those associated with ENSO events.

Acknowledgements

We would like to acknowledge logistical support from HCVCP personnel Matt Silva and Greg Vaccarino. MU students, Ryan Walker and Frank Herr provided valuable field support. The Hunterdon County Vector Control Program Black Fly Surveillance Grant # 6032305715 and the Millersville University Neimeyer-Hodgson Student Research Grant funded this project.

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Decline of the honey bee!

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Everyone has heard about the decline of the honey bee, but I was surprised to see that it is now so bad that people, like the following page of the World Wildlife Fund Christmas catalog show, are now using images of *Eristalis tenax* for *Apis mellifera*!



HONEYBEES AND FARMS
Fixing the Global Food System

Honeybees have fallen on hard times. During the past decade, loss of critical habitat, pathogens, exposure to pesticides and colony collapse disorder have caused a dramatic decline in commercially managed bees as well as some of their wild relatives. Today, honeybee colony populations have dropped to their lowest numbers in 50 years.

Ninety percent of the world's food supply is sourced from about 100 crop species and 71 of those crops – especially fruits and vegetables – depend on pollination from bees. Bee activity across apple orchards, pumpkin patches, alfalfa farms and others generate \$15 billion a year.

These tiny winged creatures play a major role in the world's agricultural system. There is so much more at risk than simply wildflowers and honey.

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Confirmatory bias and photographing the difference

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Zimmie (Elwood Zimmerman) introduced his monumental study of the *Insects of Hawaii* (Zimmerman, 1948) writing that the many images provided were less than he wanted but provision was expensive. The value of a picture ‘that tells a thousand words’ was true then, and remains so now. Thanks to major improvements in photographic equipment such that even non-specialists can take excellent images, combined with the growth of electronic communication leading to much reduced costs of dissemination means expensive paper printing is nearing elimination. We have even reached the stage in which, under exceptional circumstances, quality photographic evidence alone could be used in place of a physical holotype specimen in descriptive taxonomy (Marshall & Evenhuis 2015).

However some undoubted benefits of excellent photographs must be balanced by some more equivocal issues. Most taxonomists will be well aware of the potential (and sometimes actual) deluge of images from those seeking identifications –both from colleagues and those known previously as ‘the general public’ but now called more politely ‘citizen scientists’. We think we have a case that bears telling as there are several of these issues involved. Amongst the many images coming into our email inboxes earlier this year was a series of images (and a video available) of swarming insects and the flies believed responsible. The images and interesting details of swarm observations came from Roger Farrow, a retired CSIRO ecological entomologist and his colleague naturalist Martin Butterfield on whose rural properties, 30 km apart to the south of Canberra, the swarming was occurring. From the outset a chloropid, *Thaumatomyia*, was suspected, not least because the similar swarms of *T. notata* (Meigen) were well documented in the northern hemisphere (e.g. by Narchuk, 2000) were known to Farrow. Obtaining specimens for verification of identity was not so easy – sweeping under the active swarms revealed mostly the common ephydrid fly, *Hydrellia tritici* Coquillet, with a few other unidentified acalypterates and just a few specimens of yellow and black *Thaumatomyia*-like chloropids. *Thaumatomyia* is represented in Australia only by *T. subnotata* Malloch, described from a female holotype, and differentiated inadequately from the well known swarming *T. notata*. Comparison of Australian swarming chloropids (collected using a hand-crafted net extension) with the female holotype of *T. subnotata* from Mosman (New South Wales) held in the Australian Museum revealed considerable similarity, and thus the name was used in draft note prepared by Farrow and Butterfield. The authors presented some excellent observations on the timing, location and duration of the chloropid swarms (through much of Australian summer of 2016) at their respective properties, but also including what they termed stationary swarms (massed aggregations) observed on native vegetation in a wider area.

A persistent niggling worry was that if *T. subnotata* was an Australian analogue to *T. notata* in behaviour, as proposed, why was this the first record in Australia of such characteristic aggregations? The continent is by no means devoid of entomologists who would have been alert to the significance of such massed swarming. And the males lacked the ‘golden balls’ (subterminal abdominal vesicles) that might be expected for a *Thaumatomyia* related to *T. notata* (Kotrba, 2009). The answer came when an entomologist in the Australian National Insect Collection (CSIRO), Kim Pullen, alerted to our interest

in swarms, found a completely overlooked report in an early issue of the *Australian Entomological Magazine*, in which Barry Moore (1976) reported swarming of a *Chloromerus* species in Canberra. Identification was made by Don Colless as *C. nr striatifrons* (Becker). Specimens from Moore and from a range of subsequent routine surveys were present in ANIC. These historic specimens also were a good match to our swarmers – so how did the two species, not even congeners, differ? Were they perhaps synonyms? Ken Spencer's (1986) study of Australian Chloropinae revealed that generic differences could be found in the contour of the scutellum (rounder v flatter) and the degree of swelling of the hind femur (more v less). Such essentially subjective character state decisions are the bane of the inexpert, never-the-less side-by-side the flatter scutellum and the less swollen hind femur of the current swarmer conformed best to *Chloromerus striatifrons*. The 'characteristic' black /yellow pattern of the body is convergent, but a striking convergence.. A hasty 'global' replace and some tweaking of the near manuscript (Farrow *et al.*, 2015) averted taxonomic and taxonomists' embarrassment.

So what you might ask – is this not how such problems arise and are solved (or perhaps often ignored in ecological literature)? It is salutary to look at how we failed: mostly it can be laid at the door of 'confirmatory bias'. The ecologist's knowledge of *Thaumatomyia* swarming behaviour, and close morphological resemblance (including even by comparison with the type.) led us to a faulty identity. Everything (and there wasn't much) that conflicted, we let past. Searching Google Scholar with an incorrect term and the solitary, but critical, earlier reference does not appear. Only the encounter by a colleague, alerted by chance to the swarming behaviour, snapped us out of the 'confirmatory bias' just in time. What about the images? Now we know the subtle differences between *Chloromerus striatifrons* and *T. subnotata* we can see that no image (Fig. 1 C-D, F, G) shows the femoral shape and curvature of the scutellum adequately, no matter how much care was taken with expensive technology. Essentially the identity needed to be known already to take the appropriate images, as in the dissected and slide-mounted legs (Fig. 1H). An alternative is that we are inexperienced in Chloropidae taxonomy, and the subtleties of Ken Spencer's and his predecessors shape criteria are clear to others. We doubt this, given that the second author has been working with identification of orchid-pollinator chloropids, and at least one (un-named) expert in the family from outside Australia fell into the same 'confirmatory bias' as we did. Is this a "one-off"? We doubt it, but so few ecological / behavioural voucher specimens are deposited that we will never know the extent of other such identification problems.

Finally, this should be not the last word – although the swarming behaviour of *C. striatifrons* superficially resembles that of the well studied *T. notata*, but major differences remain. Swarming appears much rarer: only two swarming events (seasons) have been reported, nearly 40 years apart. Swarms, resembling dense smoke emanating from high points in the landscape, are unlikely to have occurred in south-east Australia without being reported (though plumes of bushfire smoke are all-too-common).

What is the natural habitat of the species – which is periodically identified only in small numbers from malaise traps and sweeping vegetation. Where do the larvae occur? Are these the mating / nuptial swarms of many "nematocerous" families, or is aggregation only when local populations are unusually large? That such aggregations occur in two apparently distinct genera suggests that this might be an apotypic behavior pattern in these taxa (and possibly other Chloropinae) expressed only under certain environmental conditions.

Adults have been seen aggregating around introduced pine trees (*Pinus radiata*) but also in native vegetation. With only two events, it is not possible to relate to climatic events, though we note that winter 2014 was a wet one against historic records. Will such high densities last season lead to a recurrence this year? Watch this space.

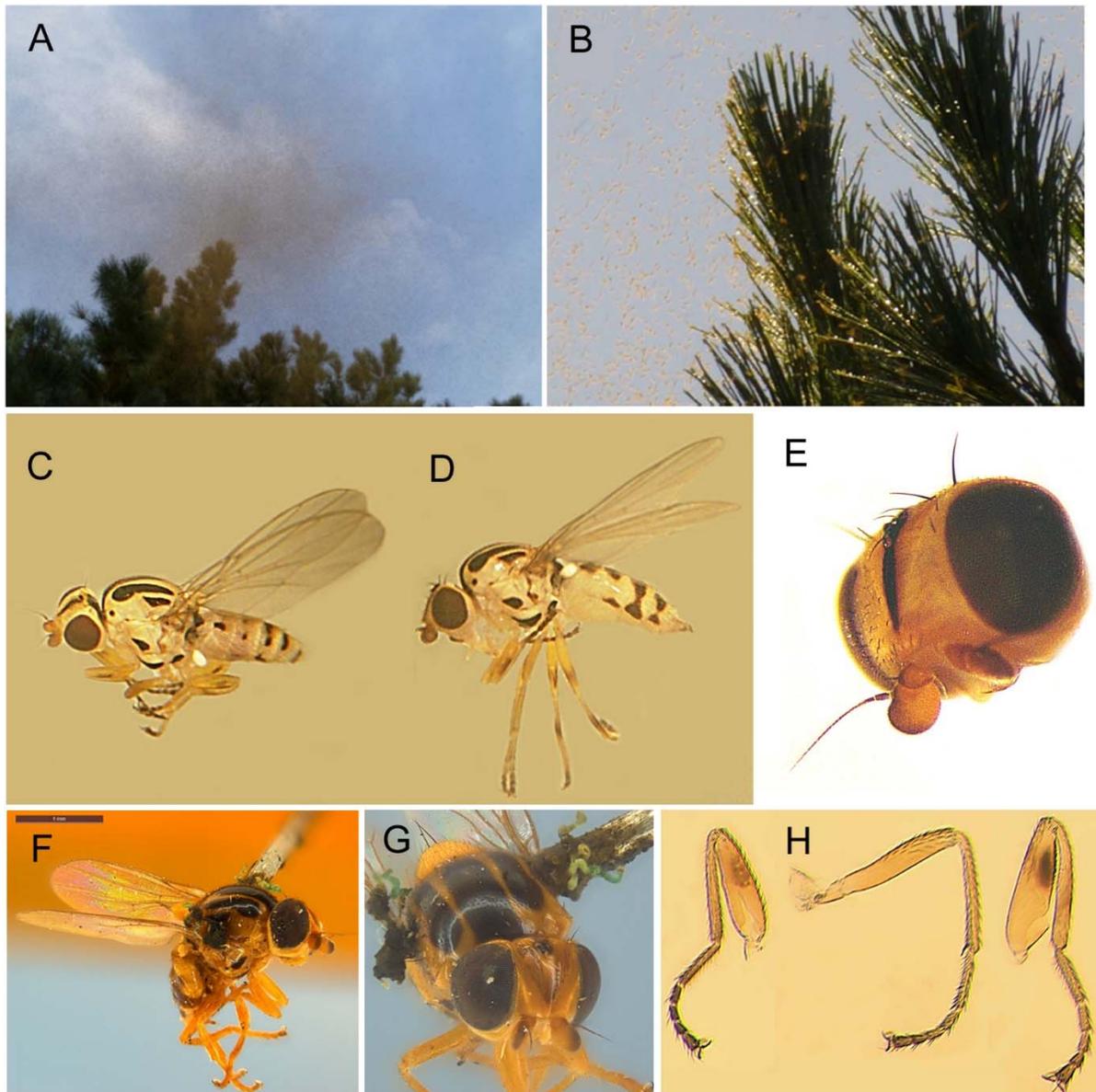


Figure 1. A. Aerial swarm, *C. striatipennis*; B. 'Stationary' swarm, *C. striatipennis*; C. Habitus, male *C. striatipennis*; D. Habitus, female, *C. striatipennis*; E. Anterior head, *C. striatipennis*; F. Habitus, Holotype ♀ *Thaumatomyia subnotata*; G. Anterior head, Holotype ♀ *T. subnotata*; H. Legs (anterior to posterior, right to left) of *C. striatipennis*.

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Collecting flies throughout Eastern Australia, October 2015

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This October our project in transcriptome 'fly-logeny' sent Keith Bayless, free of teaching obligations, to Australia in order to collect fresh material of a number of challenging families, and to work with Bryan Lessard, in the middle of a postdoc on Stratiomyidae and looking to catch every genus present in Australia. We were also on the lookout for any interesting Tabanidae for our continued collaborations on the group. The trip began immediately after the Australian Entomological Society in Cairns meeting where we both presented research talks.

The first stop for collecting was Daintree, slightly north of Cairns. We stayed at a new facility built by James Cook University. They had to give us a safety primer Powerpoint presentation for insurance reasons, which was an upbeat way to start the trip. While animals like crocodiles, venomous snakes (all snakes in Australia other than pythons are venomous), and spiders can kill you, fear should rest instead on the plants. They can't kill you but will make you suffer. Wait-a-while plants are full of sharp thorns and drape strong tendrils throughout the forest. If they catch you or your net you can't rip free, you have to remove the thorns one by one, preferably with forceps and not your soon-to-be bloodied fingers. Bryan and I went through about 4 net bags on this trip. Stinging trees (gympies) can cause serious burning pain that lasts years, and the sap from tar trees can permanently blind you if you break a branch while sweeping then touch your eyes. Great news for us, as sweeping is the primary collecting method for strats and acalyptrates! Daintree is a true tropical rainforest spot, but the rainy season was just beginning so we didn't find anything too useful. I visited several beaches but found no Xenasteiidae. I got stung on my palm by some kind of wasp which caused all of the muscles of my hand to lock up for a few minutes. Very strange. Welcome to Australia!

We then drove south and checked Hypipamee, a site for *Nemo* but it was raining all day. Finding *Nemo* was still a goal. We did find *Cyamops* near the type locality of *Plinthina beyoncae* (Lessard), the Beyonce fly, Davies Creek. Our hopes of finding new members of *Destiny's Child* was stymied by poor weather. We stayed in Innisfail and had an authentic Australian pub experience. 'Pub' in Australia means something very specific, which is weird for an American. It is a small bar upfront, then a dining area and generally a performance stage/dance floor/ slot machine/video poker area in the back, often a liquor store, and rooms to stay in. Any place that serves drinks is not a pub, it needs this elaborate floor plan. On the way south from Innisfail we stopped at several rainforest parks for an hour or so each. Cassowaries (giant deadly birds) may have some interest in Diptera as one walked up to Keith, oblivious, aspirating flies from his net. Paluma NP was the most productive of these parks, it is the southernmost park considered to be eastern tropical rain forest.

We stopped for two days at Eungella, one of the best Diptera collecting localities. The most famous site is Broken Creek, which also attracts tourists looking for platypuses. We didn't see any, but every splash or rustle was ascribed to a fleeing monotreme. This was the kind of locality where every sweep of a net brought in mostly flies, and a great diversity of them. We set up malaises, bait traps, and yellow pans. Here we found Teratomyzidae, Paraleucopidae, *Pentachaeta*, *Stylogaster*, *Asteia*,

Strongylophthalmyia, piles of pachygastrines, on and on, just a great place. Green metallic *Myioscaptia* was such a great visual and auditory mimic one had to fly into our car before we realized it wasn't a calliphorid. We stayed there another day then headed south and spent the night of Keith's birthday in Rockhampton. This is not recommended. Future expeditions, drive on by 'Rockie.' On the positive side, this is the southern limit for saltwater crocodiles, so now we can go swimming in the ocean! Just watch out for bull sharks, blue-ringed octopus, and irukandji box jellyfish.

We did spend a little time at the beach to keep looking for Xenasteiidae then went inland to higher mountains with an Araucaria forest at Bunya Mtns NP. Beautiful place but a bit cold. Full of stinging nettles which hit through jeans and hurt for a day. We collected *Diplogeomyza* (Heleomyzidae: Cnemospathidinae: Allophylopsini). Next was the Conondale range slightly north of Brisbane, looking for neurochaetids in *Alocasia* and helosciomyzids but failing to find any. The roads into the parks here had river crossings we deemed too treacherous, and it was rainy.



Figure. Clockwise from top left: David McAlpine at Bradley's Head Sydney Harbour, surveying for *Zalea*; the infamous stinging tree (*Dendrocnide moroides*); the less horrifying but more frustrating wait-a-while (*Calamus* sp.); Dan Bickel, Keith Bayless, David Yeates, and Bryan Lessard following in the footsteps of Charles Darwin and David McAlpine in the Blue Mountains, NSW (photo by John Martin); Keith doggedly and fruitlessly searching for *Xenasteia* in *Pandanus* phytotelmata until sunset at Sarina Beach, QLD.

Mt. Glorious in D'aguilar NP was next. We set up a malaise trap since this was only ~1 hour away from Brisbane we could pick it up on the way back. We also stopped for an adorable live echidna along the side of the road. This is where I found a great number of neurochaetid upside down flies in the *Alocasia* plants. Marshy areas preferred by *Alocasia* are also preferred by land leeches, so watch out. Further south at Mt. Tamborine, the stream was trashed by people from Brisbane using it as a swimming hole, but we went to a hilltop and found a number of tabanids, *Cryptochetum*, and *Borboroides* (Heleomyzidae: Cnemospathidinae: Borboroidini).

Our final two days of collecting in Queensland was at the famous Binna Burra Lodge in Lamington NP, on the border with New South Wales. The lodge was built in the early 1900s on a ridge next to two vast valleys. Lamington was one of the first preserved parks in the area so very old locality labels that say "Queensland: National Park" probably refer to Lamington. The accommodations were relatively cheap but clean and comfortable, the food was quite good. There were a lot of other people staying and visiting for hikes, so the nearby accessible trails were far from pristine. It rained in the afternoon of the second day which contributed to the light traps- crazy with activity. These lights just had flies on them, barely any moths or beetles. I was accompanied at the lights by several hungry pademelons (adorable tiny kangaroo things). We caught more Paraleucopidae and Cryptochetidae, dozens of Fergusoninidae, nearly every subfamily of strats, and found a *Cairnsimyia* (Rhinatorinae), one of the prettiest flies.

The Queensland leg over, we spent the morning visiting the Queensland Museum and talking with Chris Lambkin, then flew to Canberra. The first order of business was of course to set up some malaise traps in the Black Mountain park behind ANIC. On Sunday, we travelled to Sydney, accompanied by David Yeates met us there. Along the way we stopped at the type locality for some *Nemo* species, to try our luck again at "finding *Nemo*". They were lekking on smooth bark eucalyptus trees at the exact locality shared by David McAlpine. Pixar's original plot had more twists and drama.

Monday we met with David McAlpine in the morning, got lunch with Dan Bickel (it was a surprising relief to hear an American accent), then David McAlpine took Keith to a favorite locality in Sydney Harbour to look for shore flies, a park next to the zoo. The only people on the beach were 2 dipterists, 3 Asian teenage boys, and 4 suited up bodyguards - some kind of k-pop boy band? David found tons of *Zalea* on the intertidal rocks then dropped Keith off at the ferry. David McAlpine's deep knowledge of Diptera really fueled the whole trip. The ferry ride from the zoo to the museum went right by the Sydney opera house. Tourism! We then went for drinks with David Yeates and Dan Bickel.

Tuesday we all went up into the Blue Mountains west of Sydney to collect at David McAlpine's favorite spot- Mt. Wilson. He was feeling too frail and had some friends in town for lunch, so the expedition was me, Bryan, David Y., Dan, and John Martin. Mt. Wilson is a really great dense secluded rainforest where I nailed Helosciomyzidae and *Sciadocera*. We then had a late lunch at the furthest inland inn where Charles Darwin stayed during his trip to Australia.

The end of Keith's visit was really busy in terms of organizing specimens, and discussing ideas for analyses with Karen Meusemann and Lars Jermin. We took some time to collect in Black Mountain and check the malaise traps, where we found *Aphyssura* (Calliphoridae) and some more *Cryptochetum* and *Borboroides*. Saturday morning Pete Cranston agreed to drive Keith and Xuankun, a new ANIC PhD student under David and Bryan, 3 hours down to the coast to look for Australimyziidae and Xenasteiidae. We caught about 20 *Australimyza* and Pete bought some oysters right from the source so mission accomplished. David had a farewell/good riddance party at his place and I left Australia at 5 am. We found nearly every subfamily of Stratiomyidae and every Australian target family of Acalyptratae except for Xenasteiidae and Cypselosomatidae. It was truly a productive, worthwhile trip.

Here are the acalyptrate groups which are still missing transcriptome data. Any help with obtaining fresh material of these taxa would be deeply appreciated:

Camillidae
Celyphidae
Ctenostylidae
Cypselosomatidae
Gobryidae
Heleomyzidae: Heteromyzini/inae/idae
Huttoninidae
Marginidae
Megamerinidae
Nannodastiidae
Natalimyidae
Nothybidae
Phaeomyiidae
Xenasteiidae

Thanks to Karen Meusemann, Philippa Carr, Pete Cranston, Penny Gullan, and David Yeates for your hospitality. Thanks to Lori Lach (JCU Cairns), Chris Lambkin (QM), and Dan Bickel (AM) for giving us lab space and/or supplies. And particularly hearty thanks to Michelle Trautwein, David Yeates, and Brian Wiegmann for funding Keith's trip.

Robber flies in eastern New Mexico

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Well, at the risk of mixing metaphors, this old dog has learned a new trick, turned over a new leaf, and changed his spots!

I've been a student of "little brown beetles" since my M.Sc. thesis work back in the mid 1980s and have been very satisfied with the adoption of various tenebrionoid families as topics of taxonomic research. Since arriving in the Biology Department of Eastern New Mexico University in August, 2001, I have restricted my taxonomic endeavors to Coleoptera.

However, while out collecting beetles locally (in Roosevelt Co., New Mexico) in late May, 2014, I observed and collected something I'd never seen before: a mating pair of *Phyllomydas phyllocerus* mydid flies. I wasn't sure whether I'd just not noticed them previously, or if this was perhaps some kind of "freak" occurrence in eastern New Mexico. After identifying it as a mydas fly, and enquiring among colleagues who the "mydas fly person" was, I got in touch with Torsten Dikow at the Smithsonian, who was very interested in my discovery and provided much encouragement about all things asiloid. Well, the *Phyllomydas* turned out to be rather common last spring; in addition, we collected for the first time several other species, in *Mydas* and *Nemomydas*. Very cool.

One of the *P. phyllocerus* I observed was, at the moment of its collection by me, being fed upon by a rather large robber fly, later to be identified as a species of *Proctacanthus*. That was the beginning of the robber fly quest (aka obsession)! I soon discovered that unlike my Melandryidae and Tetratomidae, which require trees and/or fungi (both of which are few and far between out on the "llano estacado"), the diversity of robber flies was pretty high, within 10-20 miles driving distance from my office. Through continued communication with Torsten, Rob Cannings, Eric Fisher and other experts, I learned that despite these flies being large, loud and charismatic, there was much more remaining to learn about them, especially in an under-collected region such as eastern New Mexico. I was encouraged to continue collecting and be on the lookout for predator-prey associations (like the robber fly/mydas fly combo that started it all).

Through the rest of 2014 and so far up until late November, 2015, many specimens of Asilidae have been collected in various different habitats in eastern New Mexico (mainly Roosevelt, Curry, De Baca, Chaves, and Lee counties), including mammal burrows. I know that I'm not the first collector to have succumbed to the charm of the robber fly, but we have accumulated what I feel is/will be a very important regional collection of specimens; I'm eager to share/loan/exchange specimens to further other workers' studies of Nearctic taxa. Two genera from our efforts (*Heteropogon* and *Sintoria*) are presented in Figs 1–3.

Before the recent collection of the new material, the ENMU asilid collection comprised 4 drawers of specimens; most of these specimens were student-collected as part of the last 30 years or so of our General Entomology course. However, many of these older specimens were identified by John Wilcox, who I soon discovered, was one of the major players in robber fly taxonomy.



Figures 1–2. *Heteropogon ?cazieri* Wilcox, New Mexico: Quay County, Apache Canyon, 11 October, 2015. Photo credit – Lisa Reichert.



Figure 3. *Sintoria pappi* Wilcox, New Mexico: Quay County, Apache Canyon, 11 October, 2015. Photo credit – Lisa Reichert.

The recent collecting has yielded approximately 6,500 specimens (all of which have been mounted and labeled), with the following breakdown to genus:

<i>Ablautus</i>	307
<i>Atomosia</i> Macquart	74
<i>Atomosiella</i> Wilcox	26
<i>Backomyia</i> Wilcox & Martin	175
<i>Cerotainiops</i> Curran	60
? <i>Comantella</i> Curran	18
<i>Cophura</i> O.S.	70
<i>Diogmites</i> Loew	268
“ <i>Efferia</i> ”	2,331
<i>Eucyrtopogon</i> Curran	143
<i>Hadrokolos</i> Martin	3
<i>Heteropogon</i> Loew	218
<i>Hodophylax</i> James	57
<i>Holcocephala</i> Jaennicke	48
<i>Holopogon</i> Loew	38
<i>Laphystia</i> Loew	40
<i>Leptogaster</i> Meigen	72
<i>Machimus/Asilus</i>	494
<i>Mallophora</i> Macquart	170
<i>Megaphorus</i> Bigot	165
<i>Metapogon</i> Coquillet	6
<i>Microstylum</i> Macquart	110
<i>Ospriocerus</i> Loew	437
<i>Proctacanthella</i> Bromley	289
<i>Proctacanthus</i> Macquart	115
<i>Promachus</i> Loew	63
<i>Psilocurus</i> Loew	47
<i>Saropogon</i> Loew	215
<i>Scleropogon/Stenopogon</i>	68
<i>Sintoria</i> Hull	45
<i>Stichopogon</i> Loew	80
<i>Triorla</i> Parks	92
<i>Wilcoxia</i> James	224

Included in the above number are over 1,400 robber/prey associations; both the robber fly and prey item are mounted in the same pin. Some interesting patterns are beginning to emerge, though it appears that most taxa of robber flies are opportunistic feeders and will go after whatever is abundant and available on any particular day – one day it’s green treehoppers and then a few days later, it’s small syrphids...

The level of identification varies among the genera. My wife, Lisa Reichert, has become very adept at identifying robber flies, and has all of our material determined to at least genus. We have discovered that for some genera, species-level identification is virtually impossible, while others have been more recently revised and are in good shape. We would be very pleased to have this material studied by others; if you would like to arrange a loan or exchange (or if you would like to visit the collection for Asilidae or other families of flies!), please let me know – my contact information is given below.

In addition to this concerted effort to put eastern New Mexico on the asilid map (literally and figuratively!), we are revising the genus *Wilcoxia* (Fig. 4); this is a small genus with five described species and several undescribed known from collections (Eric Fisher, pers. comm). Individuals appear late in the season, and in fact, our association with *Wilcoxia* came as a result of collecting a long series of probably an undescribed species in Quay Co., NM in late October – early November of 2015. If you have material of this genus of small robbers, we would be pleased to borrow them for taxonomic study.



Figure 4. *Wilcoxia* sp., male. New Mexico: Quay County, Apache Canyon.

I look forward to hearing from anybody interested in this material, or other fly taxa from eastern New Mexico; if you are passing through this part of the world, let me know. This old beetle guy appreciates the encouragement that he has received from fly folks and looks forward to continued collaboration and relationships!

Hurray for *Triogma exsculpta*!

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On a beautiful, cool morning, the 20th of May 2015, I netted a prize – a male *Triogma exsculpta* Osten Sacken, a fly in the family Cylindrotomidae (previously a subfamily of Tipulidae). It was resting on a blade of grass with the wings outstretched. This was a species that I had long suspected to be extirpated from eastern North America where it had been recorded, so it was a real thrill to verify my find under the microscope a couple of hours later. Back I went that afternoon to the same spot, but despite diligent searching, I saw no sign of this species. In the following days the weather turned dreary and much colder, close to freezing, so I abandoned hopes of finding more specimens.

The picture (Fig. 1) does not do the fly justice. The distinctive cylindrotomine venation can be seen; the radius merges with R₂ and R₃ so that only two branches of radius extend to the wing margin. Dusky wings are quite characteristic as are the rugose (bumpy) thorax and aspects of the male genitalia. Unfortunately the latter two characters are not visible here.



Figure 1. *Triogma exsculpta* O.S.

I was collecting in a hardwood swamp along a side road in Pigeon River Country State Forest, Cheboygan County, Michigan (45°17'36" N 84°25'43" W). The swamp forest (Fig. 2) had *Typha* mainly in the undergrowth with some grassy patches and lots of Gold Thread (*Coptis groenlandica*) nearby in bloom. The area was dominated by *Acer rubrum* with some *Betula*, *Picea*, *Populus* and *Thuja* and much dead *Fraxinus* covered with mosses. The larvae of *Triogma exsculpta* have been collected from aquatic mosses such as *Fontinalis antipyretica* (Brodo 1967). I was a guest on a lichenological field trip based at the University of Michigan Biological Field Station on Douglas Lake.



Figure 2. Hardwood swamp, Cheboygan Co., MI.

Every spring for the past 50+ years, I have been on the lookout for this species and a few others that I think are no longer in northeastern North America. When I monographed the cylindrotomines in 1967, as a Master's thesis project, all my work was based on specimens collected by others, and *Triogma exsculpta* had last been collected decades previously. It was described by Osten Sacken from a single female collected somewhere in Pennsylvania in 1865. Since then it has been recorded from Connecticut, Massachusetts, Michigan, New Hampshire, New Jersey, New York, Wisconsin, and in Canada from Gatineau County, Quebec. The latter locality is in what we call the Ottawa District so it should have been possible for me to find if it still exists here. It is an early spring species that is rare but can be locally abundant.

Foolishly, I did not record the year nor the collector for the specimens cited in my monograph, being simply interested in the seasonal distribution. This was long before rumbles about climate change were heard.

Reference

Brodo, F. 1967. A review of the subfamily Cylindrotomidae in North America (Diptera: Tipulidae). Univ. Kans. Sci. Bull. Vol. XLVII, pp. 71-115.

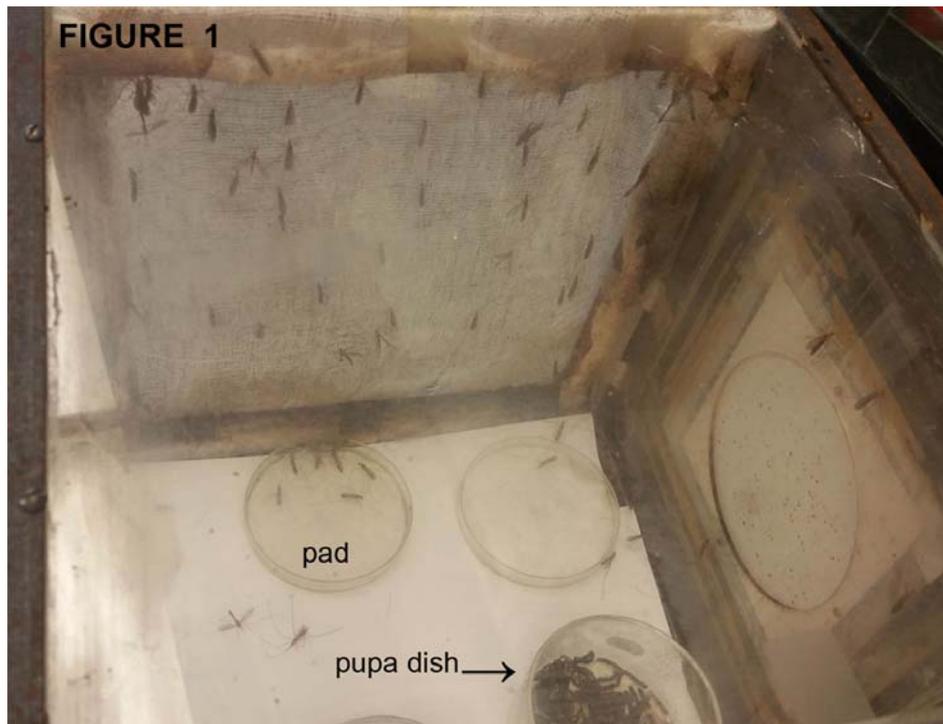
Refugee crisis: Soon to be homeless 53-year-old crane-fly colony looking for a new home(s)

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I am trying to find potential parents for a colony of crane flies, *Nephrotoma suturalis* (Loew), that I started rearing in 1961 as I worked toward my Ph.D. thesis in cell biology. I still work on their cell biology, but anticipate that they will need new homes in a few years. We study cell-division in their spermatocytes: they have large cells, small numbers of chromosomes, are convenient to rear (i.e., cheap and easy), and have a convenient (for us) meiotic cycle in that the meiotic divisions occur during one-day in the life of the animal (about the 7th day after the final larval instar).

Easy to rear. We put moist paper mache (papier-mâché) pads, in a 90mm plastic Petri dish bottom, into a cage of adult flies (Fig. 1), who then oblige us by laying eggs in the pads, sometimes so many eggs that often we see a black 'mass' instead of individual eggs (Fig. 2). The eggs hatch after about 6 days – sometimes we 'relax' and film the larvae coming out of their eggs (Figs 3-4). After the larvae hatch we add powdered dried nettle leaves on top of the pads; the larvae chomp away on both the paper and the nettle leaves (Figs 5, 6). We transfer them to clean paper pads in larger Petri dishes when they finish eating the paper and nettles in the first pads and/or when the dishes have too much excrement. We dissect 4th instar larvae, but most larvae are not killed and eventually become pupae. We remove pupae from the culture dishes, place them on moist pads in a crystallising dish, put the dish-plus-pupae in the cage, and the cycle starts again: adults emerge from the pupae, the adults mate (Fig. 7), and Bob's your uncle, more eggs. Egg-to-egg takes of the order of 6 weeks at room temperature – faster at higher temperatures, but the animals die above 30°C. The methods we use are described in (perhaps too much) detail in Forer (1982).



Cheap to rear. We rear lots and lots of animals. Because we want to be able to dissect animals every day, we have a large number of 150mm plastic Petri dishes of larvae of all ages, maybe 70-80 Petri dishes, and we have lots of adults in cages, maybe 40-50 at peak times. Caring for them takes me (or grad students who care for them) about 2 hours per week (an hour a day, twice a week); care consists of removing egg dishes from the cages and replacing them with fresh paper pads, removing pupae from Petri dishes and putting them in the cage, and sorting out the dishes for which the larvae need clean 'digs' (i.e., fresh paper mache). An undergrad 'work-study' student works two days per week, making the paper pads and taking the larvae from the old Petri dishes into dishes with clean paper; this takes about 8-10 hours per week for all our dishes. It would take fewer hours for smaller numbers of animals. The expenses we incur, then, are for 'work study' student, toilet paper (from which we make paper mache), and 'cut and sifted' dried nettle leaves. The only other expense is for $MgSO_4$ which we buy in bulk, as bulk Epsom salts. (My purchasing department went crazy when I first arrived at York University and ordered cases of toilet paper and bulk Epsom salts.) The salts are for removing the larvae from the 'used' paper mache and debris: we use an old ecology trick of putting the contents of the Petri dishes into a concentrated salt solution. When the concentration of $MgSO_4$ is high enough the larvae float and the debris sinks. We then pick up the larvae on the surface of the liquid with strainers that we get from the dollar store (or, converting to Canuck bucks, the \$1.35 store). So, relatively cheap to rear.



We have reared various crane fly species this way (some of which are listed in Forer (1982)). The ones we have had trouble with are those with diapausing eggs, and I bet that if I was patient enough to wait for diapause to break, or knew how to break diapause, I could rear them too.

Having a lab stock allows us to study meiosis in live cells. A lab stock could be used for non-cell biology purposes as well, including for studying their biology, as illustrated in some of the pictures (taken from movies we have made); for looking for variations in features used for identification, even for looking for mutants; and for using them as 'canaries-in-the-cage' indicators of pollution: we

Figure 3

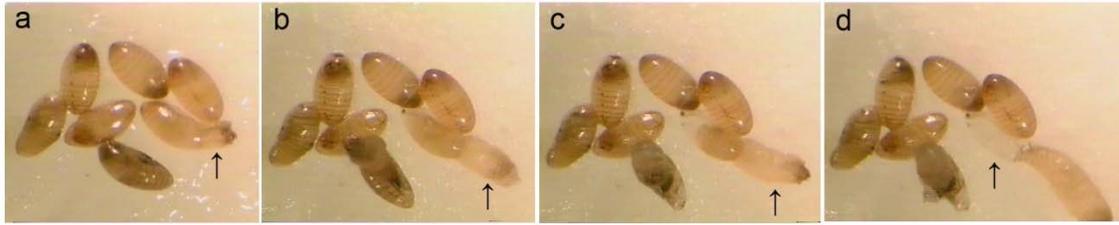


Figure 4



Figure 5



Figure 6



Figures 3 and 4: larvae emerging from eggs; Chlorox removed the black chorion and made the eggs in Figure 3 transparent. [The arrows point to a larva emerging, and then the empty egg shell (3d).]

Figure 5: a newly hatched larva chowing down on a piece of 'powdered' nettle leaves.

Figure 6: a first instar larva with full stomach.

Figure 7

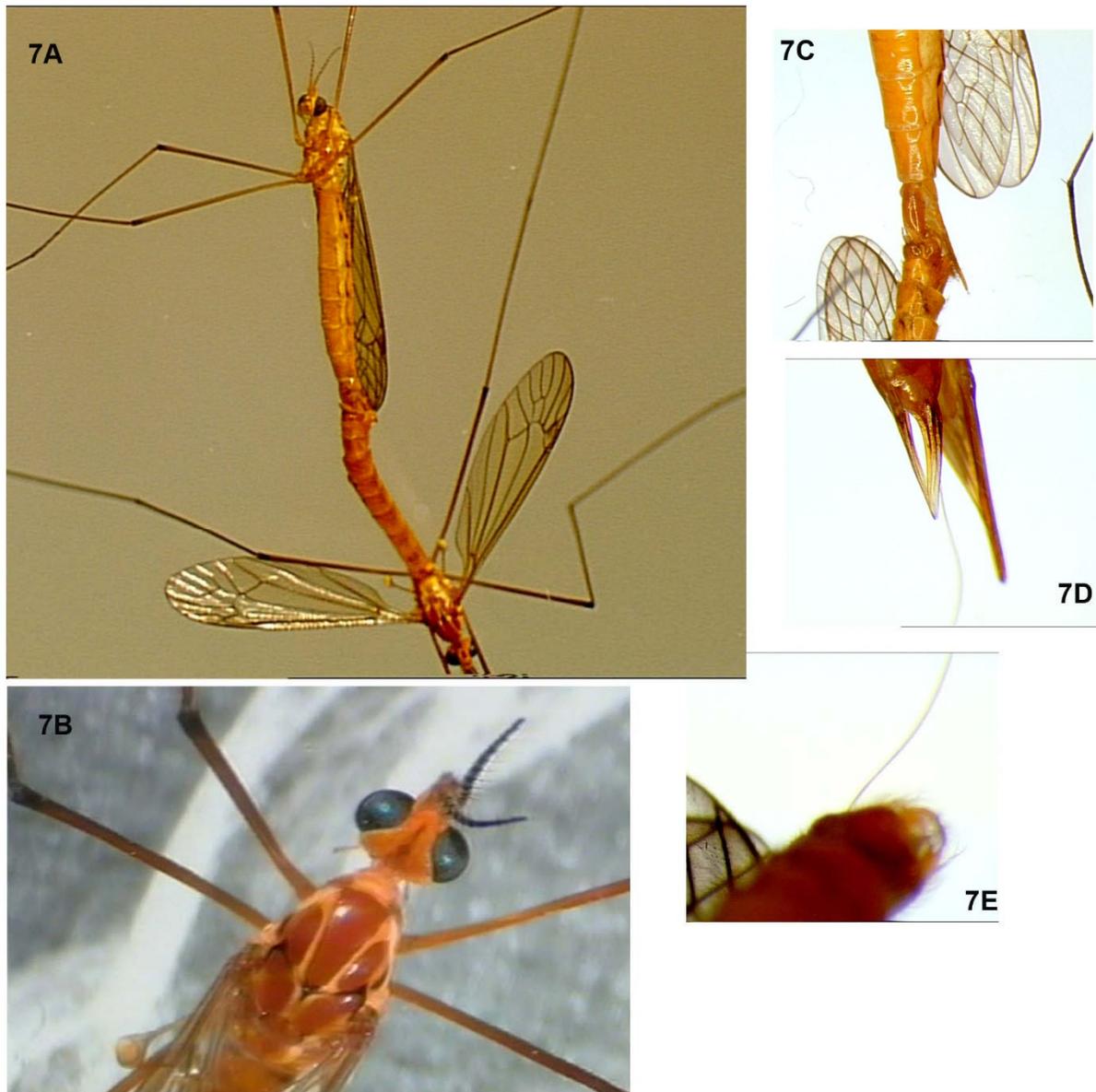


Figure 7. Live adults, **A:** *in copulo*; **B:** an onlooker; **C:** a side view during copulation; and **D** and **E**, the thin thread of sperm that often extends between male and female when they separate after copulating.

discovered that (to the detriment of our research activities) there are deleterious effects on meiosis (and larval growth and pupal eclosion) when the student cafeterias in distant buildings are sprayed with pesticide against cockroaches, when painters paint in our hallways, and when my lab is subjected to any of a number of ‘normal’ pollutants in the air, such as those arising from spraying grass with herbicides or putting asphalt on neighbouring streets (Forer & Swedak 1991). The animals generally survive, but we are out of business for a week or two after the incident and sometimes the numbers are decimated, depending on the strength of the pollution attack. For self defence (of my research) we keep more animals than we need.

Any takers, before I try *Kijiji* or *Craig's list*? No cumbersome adoption procedures will be required, nor immigration procedures since the stock arose from eggs laid in North Carolina. As a bonus I can throw in rearing records I kept over some years.

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- Forer, A. & J. Swedak, J. 1991. Practical experiences in using biological systems to monitor indoor air pollutants, pp. 129-158 in Baird, J.C., B. Berglund, & W.T. Jackson, *Indoor air quality for people and plants*, Swedish Council for Building Research, Stockholm.

HISTORICAL DIPTEROLOGY

Dates of the Diptera in Volume 7 of the *Zoología* of Gay's “*Historia Física y Política de Chile*”

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Claudio Gay's magnum opus, the “*Historia Física y Política de Chile*” took almost 30 years to complete (1844–1871) and comprises 30 volumes (published in 87 *entregas* [parts]). Gay, a French-born naturalist, travelled to Chile in the late 1820s and again in the early 1840s to collect specimens of plants and animals, record the history, culture, geography, and geology of the country, and to eventually make more than 3,000 sketches of plants, animals, landscapes, and cultural activities, to be turned into plates for his 2-volume atlas that accompanied the 28 text volumes. He then went back to Paris and assembled a team of specialists for the various parts, gathered translators to turn the French manuscripts into Spanish, and employed various engravers, lithographers, and printers to put text on paper and illustrations on plates.

The dating of the entire work has never been attempted, mainly because in the contemporary European publisher's recording journals and/or society proceedings there are virtually no records of this work as each part or volume was published. Before this work, Chile was little-known to Europeans, as was most of the rest of South America. But this publication was not intended to be sold to Europeans. It was printed in Spanish and shipped to Chile as each *entrega* was printed. Gay had early on entered into an agreement with the Chilean government to subsidize the work and he obtained an initial 400 subscribers (all in Chile) promising them a 20-volume work to be completed in a few years.

Johnson (1941) gave an excellent overview of the dating of the *entrega* of the eight botanical volumes based on wrappers and contents that existed in the Gray Herbarium at Harvard and a copy at the Natural History Museum in London. His lack of finding dates for all the *entrega* was punctuated with the hope of future workers finding possible correspondence of Gay that might lead to better dating.

Since Johnson's research, a number of works have been published regarding the life and works of Claudio Gay (e.g., Cruz & Stuardo Ortiz, 1962; Stuardo Ortiz, 1953, 1973; Rojas & Camousseigt, 2010). Much of the background information here draws from those studies. Some of those works include the correspondence that Johnson had hoped existed. I have researched the correspondence of Gay, the Chilean Charges d'Affaires Francisco Javier Rosales (who administered transport of the *entregas* from Paris to Santiago), and others in Chile, Paris, and Geneva, and have as a result found accurate dates of publication for almost all of the *entregas* comprising this work, many of which are different than previously published (those previous dates were based primarily on the year printed on the title page of each volume). The dating of Diptera in volume 7 of the “*Zoología*” series is discussed here. A more full accounting of dates for the entire work is in preparation to be published elsewhere.

Gay's method of publication for almost all of the 28 quarto-sized text volumes was to issue to subscribers each volume in 4 parts, each part consisting of 8 *feuilles* [signatures] of text (ca. 128 pages). This publishing of volumes in 4 parts changed only during times of personal hardship and for

expediency when funding to complete the series was threatened in 1852 by Rosales to be cut off (Rosales was only interested in the history volumes and did not like the scientific volumes). Each *entrega* dealt with pages for more than one volume (i.e., for *Historia*, *Zoología*, *Botánica*, *atlas*, etc.). Receipts by various libraries, information in Johnson (1941), and the correspondence of Gay and Rosales allowed association of *entregas* and volumes. Toward the end of the work (after Gay returned from Spain in 1850) each *entrega* contained pages dealing only with one volume.

The Diptera chapter was given to Charles Émile Blanchard to complete. However, Blanchard was also tasked with a number of other insect chapters in this series and was involved at around the same time with a number of large projects including a catalog of beetles in the Paris Museum and his *Histoire des insectes* (Blanchard, 1845). The Diptera specimens Gay collected were deposited in the Paris Muséum upon his return there in September 1842. Since Blanchard did not get to them right away, Macquart ended up describing many of them in his *Diptera exotica* series in the mid-1840s. Blanchard indeed cited Macquart's names and descriptions in his work on the Chilean Diptera; however, not all of Gay's Chilean specimens were described by Macquart: three genera and 91 species of Diptera were described by Blanchard as new in the *Zoología* of Gay's *Historia* (see Table 2 for a full list of new species).

All Diptera except one (*Opomyza marginipennis* Blanchard on page 475 of volume 8) were published in volume 7 of the *Zoología* (Fig. 1). This seventh volume has always been dated as 1852, based on the year printed on the title page: "MDCCCLII". However, research conducted in this study shows that volume 7 was published in parts from 1852 to 1854, with the true date of publication for Diptera being 1854 and not 1852.

Four *entrega* make up volume 7 of the *Zoología* where the Diptera are found on pages 327–468 (which fall into the last two of the four parts of this volume). Johnson (1941) gave evidence that the title pages for the *Botánica* volumes were printed along with the first *entrega* making up each volume. This appears to be case with all the volumes in the work including volume 7 of the *Zoología*, in which the title page and first *entrega* were printed and issued together in 1852. The printer, Maulde et Renou, are listed on the reverse of the half-title page as being at Fosses St.-Germain-l'Auxerrois 14 (Fig. 2), which they were, but only in late 1852 and 1853 (Anonymous, 1853). However, the colophon on the last page of the volume gives the same printer's address as Calle [= Rue] de Rivoli, 114 (Fig. 3). They were not at this address until the beginning of 1854 (Anonymous, 1854; Maulde & Renou, 1954), which means that the last *entrega* of the volume containing the colophon could not have been published until early 1854 (see Table 1 for dates of all the parts comprising volume 7).

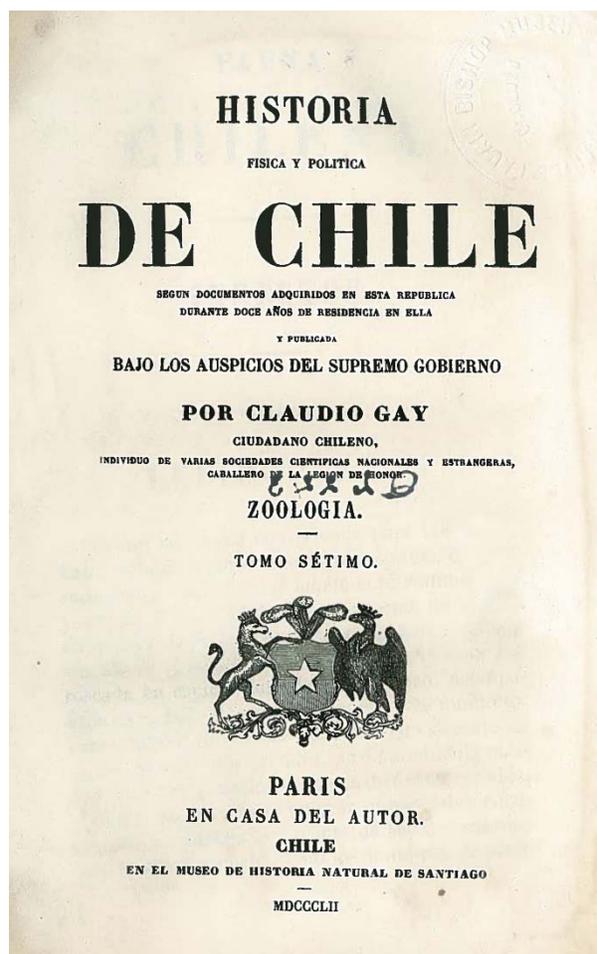


Figure 1. Title page of volume 7 of the *Zoología*.

Table 1. Dates of publication of the parts making up volume 7 of the *Zoologia*

Part	Pages*	Date of publication	Source	Notes
1	1-224	27 November 1852	Stuardo (1973: 350)	
2	225-256	20 December 1852	Stuardo (1973: 350)	
3	257-400	26 April 1854	Stuardo (1973: 355)	
4	401-471	26 April 1854	Stuardo (1973: 355)	printer not at colophon address until early 1854

*pages of each part are tentative and were determined by obvious breaks in the copies examined (e.g., different colored paper through aging, different margins, etc.) as well as the trends in numbers of *feuilles* [with 16 pages consistently comprising a *feuille* throughout the duration of the project] making up parts in other parts of this series.

The correspondence of Rosales in Stuardo (1973) gives chronological updates of the number of *entregas* and volumes that were issued at the time a letter was written (e.g., “*Botánica 9 entregas*”; “*Zoología 3 entregas*”, etc.). The initial 21 *entregas* for the entire work were given unique numbers, but the numbering stopped shortly before Gay took a long break from the project in December 1849 and visited Spain (under doctor’s orders because the stress of the project was making him ill). Why the numbering was never re-initiated when Gay returned in late 1850 is unknown but it could have been because he remained despondent over the tardy contributors (Blanchard being high among them), delayed publication of parts, and lack of support coming from his Paris liaison, Rosales. Numbering any further *entregas* was possibly unimportant to him since he was losing subscribers and was just trying to get the work finished as soon as he could. Despite the lack of unique numbers for each *entrega*, by keeping a running tally of the newly published *entregas* as they were updated in the correspondence, it allowed me to discover the parts per volume and the dates of their publication (via the dates of the correspondence). This evidence in the correspondence shows that there were four parts that comprised volume 7 and that the last two parts were not issued until late April 1854. Thus, all the new taxa of Diptera in this work previously dated as 1852 should be changed to 1854.



Figure 2. Detail of the reverse of the half-title page giving the printer and address where they were located in 1852 and 1853.

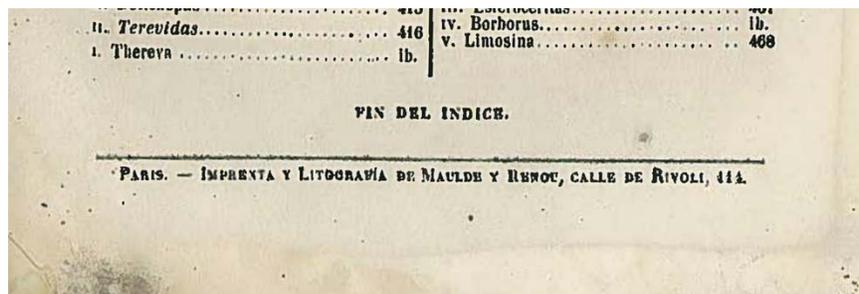


Figure 3 Detail of the colophon giving the printer and address where they had moved to in early 1854.

Table 2. Alphabetical List of Newly Described Diptera in Blanchard in the *Zoología*.

Taxon	Year/page	Family
Acanthocnemis	1854: 355	Bibionidae
Acanthocnemis hyalipennis	1854: 358	Bibionidae
Acanthocnemis immaculipennis	1854: 358	Bibionidae
Acanthocnemis obscurus	1854: 357	Bibionidae
Acanthocnemis pallens	1854: 357	Bibionidae
Acanthocnemis rubricollis	1854: 356	Bibionidae
Acinia bella	1854: 460	Tephritidae
Acinia cinerea	1854: 461	Tephritidae
Acinia delicatella	1854: 459	Tephritidae
Acinia impluviata	1854: 461	Tephritidae
Acinia marmorata	1854: 460	Tephritidae
Acinia modesta	1854: 458	Tephritidae
Acinia nymphula	1854: 462	Tephritidae
Acinia plagiata	1854: 458	Tephritidae
Acinia simplex	1854: 459	Tephritidae
Anopheles annuliventris	1854: 334	Culicidae
Anopheles variegatus	1854: 333	Culicidae
Anthrax hyalinipennis	1854: 382	Bombyliidae
Anthrax rufiventris	1854: 382	Bombyliidae
Anthrax vicinus	1854: 381	Bombyliidae
Beris maculipennis	1854: 400	Stratiomyidae
Borborus femoralis	1854: 467	Sphaeroceridae
Cecidomyia flavida	1854: 350	Cecidomyiidae
Cephalocera albocinctus	1854: 361	Mydidae
Chironomus articuliferus	1854: 336	Chironomidae
Chironomus maculipennis	1854: 335	Chironomidae
Chironomus obscurellus	1854: 336	Chironomidae
Chironomus pallidulus	1854: 335	Chironomidae
Chironomus tessellatus	1854: 336	Chironomidae
Culex annuliferus	1854: 333	Culicidae
Dilophus maculipennis	1854: 354	Bibionidae
Dilophus nigripes	1854: 354	Bibionidae
Dilophus rufipes	1854: 355	Bibionidae
Dilophus testaceipes	1854: 355	Bibionidae
Ensina obscurella	1854: 463	Tephritidae
Erioptera obscurata	1854: 343	Limoniidae
Erioptera uniformis	1854: 343	Limoniidae
Eristalis elegans	1854: 406	Syrphidae
Gonypes fuscipennis	1854: 371	Asilidae
Helomyza elongata	1854: 451	Heleomyzidae
Helomyza pallidiceps	1854: 451	Heleomyzidae
Helomyza vitticollis	1854: 452	Clusiidae
Hilarempis cotoxantha	1854: 372	Empididae
Hirmoneura flaviventris	1854: 384	Nemestrinidae
Laphria rufiventris	1854: 365	Asilidae
Leptogaster fuscipennis	1854: 371	Asilidae
Lestremia nigra	1854: 349	Cecidomyiidae
Limnobia elquiensis	1854: 341	Limoniidae
Limnobia linecollis	1854: 341	Limoniidae
Limnobia ornatipennis	1854: 342	Limoniidae
Limnobia stictica	1854: 342	Limoniidae
Limnobia stigmatica	1854: 341	Limoniidae
Limnophila chilensis	1854: 339	Limoniidae

Table 2. (continued). (note, * published in volume 8).

Taxon	Year/page	Family
Limosina consanguinea	1854: 468	Sphaeroceridae
Mycetophila obscuripennis	1854: 346	Mycetophilidae
Mycetophila ornatipennis	1854: 344	Mycetophilidae
Mycetophila punctipennis	1854: 345	Mycetophilidae
Mycetophila viticollis	1854: 345	Mycetophilidae
Mycomya chilensis	1854: 347	Mycetophilidae
Mydas rubrocincta	1854: 362	Mydidae
Opomyza marginipennis*	1854: 475	Opomyzidae
Ortalidina	1854: 455	Chamaemyiidae
Ortalidina cellularis	1854: 455	Chamaemyiidae
Ortalis decorata	1854: 453	Tephritidae
Ortalis picta	1854: 454	Uliidiidae
Ortalis striolata	1854: 454	Tephritidae
Panops nigratarsis	1854: 375	Acroceridae
Panops ocelliger	1854: 376	Acroceridae
Panops rufovestitus	1854: 376	Acroceridae
Psychoda fimbriatissima	1854: 351	Psychodidae
Psychoda hyalinata	1854: 351	Psychodidae
Psychoda notata	1854: 351	Psychodidae
Sapromyza delicatula	1854: 447	Lauxaniidae
Sapromyza lineatocollis	1854: 447	Lauxaniidae
Sapromyza longipennis	1854: 445	Lauxaniidae
Sapromyza nigriventris	1854: 446	Lauxaniidae
Sapromyza pallens	1854: 445	Lauxaniidae
Sapromyza parvula	1854: 447	Lauxaniidae
Scatopse carolina	1854: 359	Scatopsidae
Scatopse parvula	1854: 359	Scatopsidae
Sciara fuliginosa	1854: 348	Sciariidae
Sciara infuscatipennis	1854: 348	Sciariidae
Sciara pallipes	1854: 348	Sciariidae
Sciomyza fulvescens	1854: 448	Sciomyzidae
Sciomyza nigripes	1854: 449	Lauxaniidae
Sciophila obsoleta	1854: 347	Mycetophilidae
Simulium fulvescens	1854: 353	Simuliidae
Syrphus macquarti	1854: 411	Syrphidae
Syrphus similis	1854: 410	Syrphidae
Tabanus lasiophthalma	1854: 394	Tabanidae
Tabanus rubrifrons	1854: 396	Tabanidae
Trichoceromyza	1854: 449	Unplaced Acalyptratae
Trichoceromyza livida	1854: 449	Unplaced Acalyptratae
Trichoceromyza lutescens	1854: 449	Unplaced Acalyptratae

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MEETING NEWS



2015 Field Meeting of the North American Dipterists' Society: Red River Gorge, Kentucky, USA, 7-11 June 2015

Gregory A. Dahlem

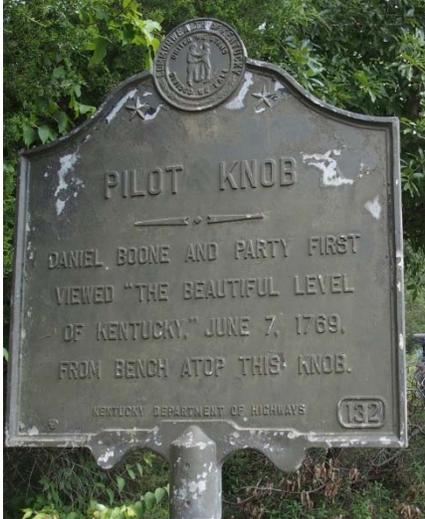
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The 2015 NADS field meeting took place in the Appalachian foothills of eastern Kentucky. The Red River Gorge Geological Area and adjoining Natural Bridge State Park are especially beautiful in spring and I think that everyone attending found something spectacular that they will remember from their visit.

The organizing team, made up of Greg Dahlem, John Stireman, and Ron DeBry stayed at the central meeting cabin named “Living on the Edge”. This was aptly named given the large rock cliff with an impressive drop off in the backyard (see top photo for view off the deck). Several other participants, including Jim O’Hara, Brad Sinclair, and Evan Wong joined us in this large cabin during the meetings.





On Monday, about half our group spent some time collecting at Pilot Knob State Nature Preserve (less than an hour's drive down the road). I think just about everyone took a trip out to this little park during the meetings. Beautiful place with a gorgeous view from the top (above), a small spring that yielded some interesting flies along the trail (right), and a creek bed at the bottom (below).





We all congregated at the main cabin on Monday night for cheesy snacks and a trip down the Kentucky Bourbon Trail. After some drinks and tall tales, we finished up the evening with the:

NADS PRESENTATIONS– 2015

1. **Andrew Fasbender**. "The phylogeny of *Ptychoptera* (Diptera: Ptychopteridae)" Rhithron Associates, Inc.; Missoula, MT; afasbender@rhithron.com
2. **Bradley Sinclair**. "Nearctic balloon flies: resolving the diversity of *Empis* (*Enoplemis*) (Diptera: Empididae)". Canadian National Collection of Insects and Ottawa Plant Laboratory – Entomology; Canadian Food Inspection Agency, Ottawa, Ontario; bradley.sinclair@inspection.gc.ca
3. **Z.L. "Kai" Burington**. "Knife flies and sword flies (Tachinidae: Blondeliini: Eucelatoria)". Department of Biological Sciences, Wright State University, OH; keroplatus@gmail.com
4. **Jessica Gillung** and Shaun Winterton. "New remarkable Acroceridae Baltic amber fossils (Diptera: Acroceridae)". Department of Entomology, University of California, Davis, CA; jpgillung@ucdavis.edu
5. **Sonja Scheffer**. "Biodiversity of Agromyzidae: An inordinate fondness for Utah?? (Diptera: Agromyzidae)". Systematic Entomology Lab, USDA, Beltsville, MD; Sonja.Scheffer@ARS.USDA.GOV
6. **Isai Madriz**. "Tanyderidae, advances and breakthroughs on the family treatment". Department of Entomology, Iowa State University, Ames, IA; rimadriz@iastate.edu
7. **Kevin Moulton**. "DNA fingerprinting reveals cryptic species of dixid and net-winged midges" (Diptera: Dixidae, Blephariceridae)". Department of Entomology and Plant Pathology, University of Tennessee, Knoxville, TN; jmoulton@utk.edu
8. **Robert Pivar**. "Molecular systematics of World Thaumaleidae and Nearctic *Androprosopa* (Diptera: Thaumaleidae)". Department of Entomology and Plant Pathology, University of Tennessee, Knoxville, TN; robertjpivar@gmail.com
9. **Evan Wong**. "DNA-based species delimitation in the agriculturally important genus, *Ravinia* (Diptera: Sarcophagidae)". Department of Biological Sciences, University of Cincinnati, Cincinnati, OH; wonges@mail.uc.edu
10. **Greg Dahlem**. "Everybody Poops – natural history observations on the genus *Ravinia* (Diptera: Sarcophagidae)". Department of Biological Sciences, Northern Kentucky University, Highland Heights, KY; dahlem@nku.edu





Tuesday and Wednesday everyone headed out in small groups to a variety of locations in Red River Gorge, Natural Bridge State Park and Pilot Knob. I think that everyone found some habitat that they were hunting for and specimens in their special interest. Some investigated the rock arches found in this area (Natural Bridge above), others went to lowland creeks, or to huge rock cliff faces, or to highland ridgelines with spectacular views. A variety of wildlife beyond Diptera came out to see what we were up to. Bill Grogan showed off his awesome snake handling skills. The bot fly that Greg Dahlem caught ended up laying eggs in its peanut butter jar before passing away peacefully.





We all got together for the conference dinner and group photo on Wednesday night. A Kentucky Derby dinner with Kentucky Fried Chicken, green beans and ham, corn on the cob, and much, much more. We paired our down-home dinner with wine from Kentucky's own Verona Vineyards, finishing up with Derby Pie.

The night sky was pretty spectacular, with no moon and a nice view of the Milky Way. Several people pulled out their night sky apps on their phones to investigate the multitude of stars. Whippoorwills were calling and the lightning bugs were flashing. It was great to see everyone and I look forward to the next meeting (in Montana?).



For more photos from the meeting, see:

<https://www.flickr.com/photos/yonkstireman/sets/72157654938556571>

<https://www.flickr.com/photos/134327931@N07/sets/72157654688177850/>

<https://www.flickr.com/photos/132302132@N02/albums/72157654857744636>

ICD8 Newsflash

Congratulations to Frank Menzel, Marion Kotrba and Netta Dorchin, the organizing team of the 8th International Congress of Dipterology, who received the 1st place prize in the Potsdam Congress Competition, in the category “Best Single Event”. It is a great distinction to get this prize amongst 28 competing events. Altogether three prizes were given, one for the best single event, one for the best repeating (annual) congress (and the winner was “Satellite and Dwarf Galaxies in the Local Group”) and one for the most innovative and unusual congress (and the winner was “Conspiracy Theories in the Contemporary European Crisis”). The jury judged the events by the quality of the organization of the event, the scientific impact, the international make up, as well as the supporting program (excursions, etc) and public relations outreach. And the flies came out on top of the 8th (!) International Ikebana Conference and the 150th Anniversary meeting of the International Association of Geodesy.

<http://www.wis-potsdam.de/de/nominierungen>



**XXV International Congress of Entomology,
Orlando, Florida, USA 25-30 September 2016,
<http://ice2016orlando.org>**

Torsten Dikow

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The *Entomological Society of America* will be the host of the *XXV International Congress of Entomology* next year. Thomas Pape and Torsten Dikow have organized a symposium entitled, "Diptera systematics: deciphering evolutionary relationships with diverse and novel data." Speakers will include Dalton Amorim, Eliana Buenaventura, Pierfilippo Cerretti, Torsten Dikow, David Grimaldi, Bryan Lessard, Rudolf Meier, Thomas Pape, Jeff Skevington, Katharina Schneeberg, Seungwan Shin, John Stireman, Mauren Turcatel, and David Yeates and will cover the Diptera Tree of Life from A–Z employing morphological and molecular data as well as other relevant topics.

The *North American Dipterists Society* will also have its annual meeting during the congress, as we always do during *ESA* meetings, and we hope that all attending dipterists will participate. Torsten Dikow will organize that meeting and invites dipterists to contact him with presentation suggestions (DikowT@si.edu).

Please consider attending the *International Congress of Entomology* and in particular the Diptera systematics symposium and NADS meeting.

OPPORTUNITIES

Opening for an Entomologist in Florida!

Paul E. Skelley

Entomology Section, Florida State Collection of Arthropods, Division of Plant Industry,
Florida Department of Agriculture and Consumer Services, Gainesville, Florida, USA;
Paul.Skelley@FreshFromFlorida.com

At long last our curator position has been opened. The closing date is listed as December 25, but I have requested it be extended to December 31. If interested please don't procrastinate. If you have been discussing it with others or students, please forward.

Position number 42001235, Biological Scientist IV

Posted in the State employment system at <https://peoplefirst.myflorida.com/peoplefirst>

Direct link to posting: [Apply for 42001235](#)

Biological Scientist IV, position number 42001235, assigned to Entomology and headquartered in Gainesville/Alachua County. The announcement will close Friday, December 25, 2015, and is open to all qualified candidates. If questions, please contact Dr. Paul Skelley at (352) 395-4678 or by e-mail to Paul.Skelley@freshfromflorida.com

The Florida Department of Agriculture and Consumer Services has an open position for a Ph.D. level curator at the Florida State Collection of Arthropods (FSCA). The arthropod taxonomic expertise of the candidate is open, but they will be assigned identification of some regulatory taxa. The groups available at the moment are Thysanoptera and non-insect arthropods (but not Acari). Curation will focus on those groups, the wet collections, as well as the candidate's area of expertise.

Equally important are other collection related administrative tasks, which may include working on collections improvement grants, liaison with the Florida Museum of Natural History and University of Florida as we build a larger partnership, managing the FSCA website, curation of wet collections and bulk samples, maintaining acquisition records, handling 3-177 declarations, maintaining our CITES registration, managing varied databases, etc.

The salary listed is in the mid to upper 40s, but the candidates qualifications may allow us to justify more.

GOOD LUCK to all.

Opening at the Walter Reed Biosystematics Unit

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Walter Reed Biosystematics Unit (WRBU) Culicidae researcher Dr. Pollie Rueda will be retiring at the end of 2015. I just learned that WRBU (<http://wrbu.org>) will most likely fill his position as Research Entomologist for medically important Diptera again. Qualifications:

- Ph.D.
- background in mosquito or sand fly biosystematics (or tick systematics)
- preferably U.S. citizenship

Dr. Rueda informed me that interested applicants should send their CV/resume to the WRBU chief entomologist MAJ Jeff Clark (jeffrey.w.clark1@us.army.mil).

WRBU is based at the Museum Support Center of the National Museum of Natural History, Smithsonian Institution and is in charge of all medically important Diptera (and a few others) of the USNM, namely Psychodidae, Ceratopogonidae, Chaoboridae, Corethrellidae, Culicidae, Dixidae, Simuliidae, Tabanidae, and Hippoboscidae.

DIPTERA ARE AMAZING!

As usual, we've got some great pics to display that Diptera ARE amazing! Thanks to the folks who submitted the pics! The first photo was submitted by Marjolaine Giroux and photographed by Maxim Larrivée (Space for life, Insectarium, Montréal, Québec, Canada), we clearly see the fly, a *Laphria* sp., keeping the sting of the prey (*Bombus* sp.) away from itself, with its legs.



This next set of photographs were made by Jim Hogue (California State University, Northridge, California, USA). This is a great selection of gorgeous flies of the gorge, from the NADS Field Meeting, 2015, in Red River Gorge, Kentucky.



Elephantomyia westwoodi Osten Sacken, male



Gonomyia subcinerea Osten Sacken, male



Chrysops sp., female



Dialysis rufithorax (Say), female



Tricyphona inconstans (Osten Sacken), male



Rainieria antennaepes (Say), female

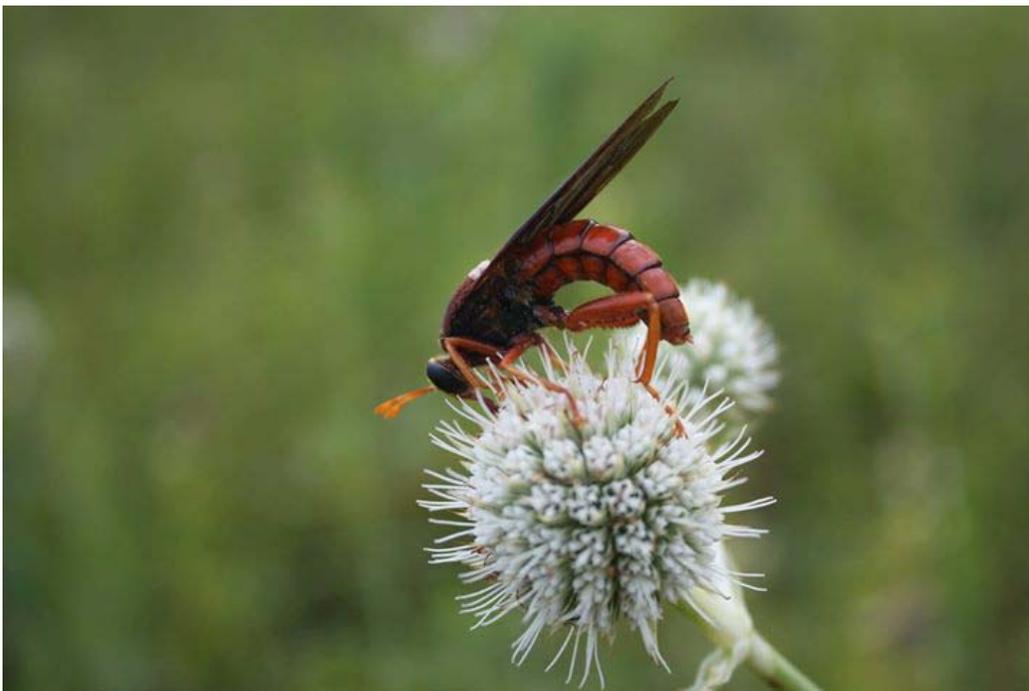


Spilomyia fusca Loew, male



Tabanus sp., female

And finally, this great photo of a male *Dalmannia* sp., by Bob Parks (top), and an unidentified Mydidae visiting a flower, submitted via Steve Hamilton and Steve Murphree (bottom).



BOOKS AND PUBLICATIONS

Here's the latest in the wonderful world of Diptera literature, including larval movement patterns, a variety of pollination studies, Buffalo wallow breeding biters, scorpion parasites and odonate food! Hope you enjoy!

As usual if we have not included a paper that you think should have been here please feel free to pass it along to Chris (chris.borkent@gmail.com) and we will include it in the next issue. Unfortunately the online resources do not always catch everything and are a couple of months behind. We also apologize for the missing diacritics in some author's names, unfortunately this is a product of searching in Zoological Record and Web of Science, where they are removed.

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