ARTHROPOD MONITORING: SEASONAL VARIATION IN THE ABUNDANCE OF SURFACE ACTIVE, SUB-SURFACE AND LOW FLYING ARTHROPODS

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PURPOSE

Record the seasonal variation in the abundance and activity of surface active, sub-surface and low flying arthropods during the whole summer to 1) determine resource availability for shorebirds and 2) investigate the composition of the arthropod community.

Three different sampling procedures will be conducted. Sampling via modified pitfall traps (procedure 1) is the number one priority. When pitfall sampling is not logistically possible, observational data may be taken instead (procedure 2). An additional procedure to sample sub-surface larvae will be tested on Bylot Island in 2008.

TIME PERIOD

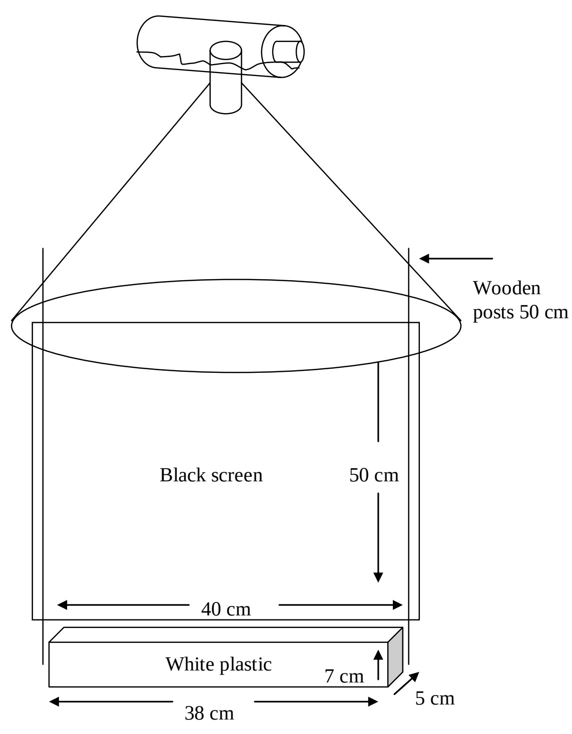
Timing of arthropod sampling will be determined by shorebird nesting phenology and permafrost conditions. Ideally, arthropod sampling should start just prior to the arrival of shorebirds on the nesting area and continue into the shorebird brood rearing period. Logistically, snow cover and permafrost conditions will determine when pitfalls can first be deployed. On Bylot Island, arthropod sampling begins approximately the first week of June and continues until the first or second week of August.

PROCEDURE 1 – MODIFIED PITFALL TRAPS

Modified Malaise/pitfall traps are used to capture surface active and low-flying arthropods. The method has been successfully used and tested on Bylot Island. Two transects of 5 traps each (20 m between each trap) should ideally be deployed in each of the two dominant shorebird foraging habitats that are used during the brood rearing period. This is generally either dry upland or low wetland habitat. Within each habitat type, ensure that the microhabitat surrounding each trap is relatively homogenous to reduce within site variation. Traps should be emptied every 48h (approximately) throughout the season. Due to the frequent sampling, it is better to set up the traps relatively close the camp.

The trap is composed of a white plastic pitfall trap above which is set a vertical mesh screen and a bottle trap to increase the capture of flying insects (see *Trap construction* section below; Fig. 1). The traps must be installed so that the perimeter of the pitfall trap is flush or 1cm below the top layer of vegetation and the mesh screen is perpendicular to the prevailing winds in the area. The top bottle trap and bottom pitfall trap should be filled with at least 1cm glycerine or soapy water (ideally odour free detergent like Tween 20, though a drop of regular dishwashing soap may be used instead). When sampling, the contents of both the top bottle trap and the bottom pitfall trap can be pooled, emptied in a sieve onto a piece of cloth and rinsed with water. The cloth and the arthropods on it can then be transferred and stored in a whirlpak with 70% alcohol. For proper sample preservation, the volume of ethanol in the whirlpak should be at least 4 times the volume of the arthropod sample or at least 2 cm at the bottom of the whirlpack for small samples. Using a lead pencil, note the date and trap number on a piece of paper and place the paper in the whirlpak with the sample. Samples may also be identified by writing on the whirlpak with permanent markers, however, 70% ethanol will erase permanent marker, thus do not rely on this method alone. Make sure that the whirlpaks are well inflated in order to decrease the crushing of samples. Store the samples upright until later identification and enumeration.

Common problem: Disturbance of the ground around the trap during sample collection.
Solution: Carefully replace the vegetation and soil around the trap after manipulation so as to avoid any gap between the ground surface and the edge of the trap.



Modified Pitfall Trap Specificiations

Modified Pitfall Trap in Field

Transect of 5 Modified Pitfall Traps

Figure 1. Modified pitfall trap specifications and field pictures (Note that the bottle trap at the top is missing on the traps shown on the field pictures).

Identification and enumeration of samples

With the exception of spiders, collembolans and larvae, samples should be classified to family. Several families are also separated into size classes (Table 1). Once classified into families and size classes, all individuals should be counted. The length of all larvae, spiders, and spider egg sacs (Fig. 2) should be measured to the nearest 0,5mm and recorded. Equations to estimate biomass for each size class have been created based upon data collected on Bylot Island in 2005 (*equations to follow*). Samples should be refilled with ethanol if necessary every 5 months.

Figure 2. Wolf spider (Lycosidae) with egg sac.

Table 1. Size classes required for specific families.

Size Class 1	**Size Class 2**	**Size Class 3**
Anthomyiidae: 0-6mm	Anthomyiidae: 6-10mm	
Chironomidae: 0-4mm	Chironomidae: 4-10mm	
Muscidae: 0-5mm	Muscidae: 5-12mm	
Scathophagidae: 0-5mm	Scathophagidae: 5-10mm	
Syrphidae: 0-8mm	Syrphidae: 8-15mm	
Tachinidae: 0-6mm	Tachinidae: 6-10mm	
Tipulidae: 0-9mm	Tipulidae: 9-14mm	Tipulidae: 14-25 mm

PERSONNEL

Building traps can take up to 2 hours/trap. Installation of the traps in the field should take less than 10 hours (approximately 1 hour/trap). After installation, the sampling of traps is not time consuming. Sampling one transect of traps (5 traps) should take 1 person approximately 30 minutes every two days. If whirlpaks are prepared ahead of time and supplies (rinsing water, refill soapy water and ethanol) are left within proximity of the transect, sampling may occur en route from other projects.

For samples collected in 10 modified pitfall traps over the entire summer on Bylot Island, the identification and enumeration conducted at the University required about 4 to 5 weeks of work for 1 full-time technician experienced in arthropod identification

MATERIAL

- Sieve (coffee or flour strainer)
- Four extra Nalgene bottles (one to store water on site for rinsing samples and one to store soapy water on site to refill pitfall traps)
- 500 whirlpacks 4 oz
- 500 pieces of cloth (cheese cloth type)
- 70% ethanol - approximately 10 L (1 oz per sample × 10 traps × 30 days = 300 oz = 8.87 L)
- Pencil and paper
- Permanent marker
- Odour free detergent (eg. Tween 20)
- 10 Modified Pitfall Traps (instructions and detailed material list below)

MODIFIED PITFALL TRAP CONSTRUCTION

The pitfall trap consists of a white plastic pitfall trap (38 cm × 7 cm × 5 cm) and a vertical mesh screen (40 cm × 50 cm), which is topped with a white plastic funnel leading into a bottle trap at the top (Fig.'s 1, 3).

- 10 pitfalls traps

The catch tray for the pitfall trap is a white plastic 15x3x2 inch (38 cm × 7 cm × 5 cm) Rubbermaid drawer organizer. These can be purchased at most hardware stores or online at: http://www.rubbermaid.com/rubbermaid/product/product.jhtml;jsessionid=MVUAEYJ5KDJTACQHUB2CJ0QKA4QGIJCK?prodId=HPProd100147

- 40 wooden posts

The wooden posts holding the screen are cylindrical 1/2" diameter posts, approximately 50cm long and can be found in the paint section of most hardware stores. Two holes must be drilled into one end of the post approximately 5 cm from the extremity so that the post can be attached to the plastic cone via metal wire

- 2 rolls of 10 m × 1 m of Black screen (1 roll makes 8 nets)
- Heavy duty black thread

The mesh net or black screen is general black mosquito nylon screen that is used for screen doors. Black thread is needed to attach the screen to the posts (note that we tried glue at first but this was not successful). The screen surface between the posts is 50 cm high and 40 cm large.

- 5 ABS plastic sheets 4 × 8 (see Fig. 3)

The plastic funnel was made from ABS plastic sheets that were cut into a conical shape (approximately 40cm diameter at base and 5 cm diameter at the top) and glued with ABS glue. Plastic ABS sheets may be ordered from local hardware stores or we can provide a retailer in Rimouski.

- 10 – 500 ml nalgene wide mouth bottles

Nalgene bottles can be ordered from http://vwrlabshop.com/laboratory-bottles-high-density-polyethylene-wide-mouth-nalgene/p/0006939/ in the VWR catalog number 16125-082, the NNi number of the product is 2104-0016.

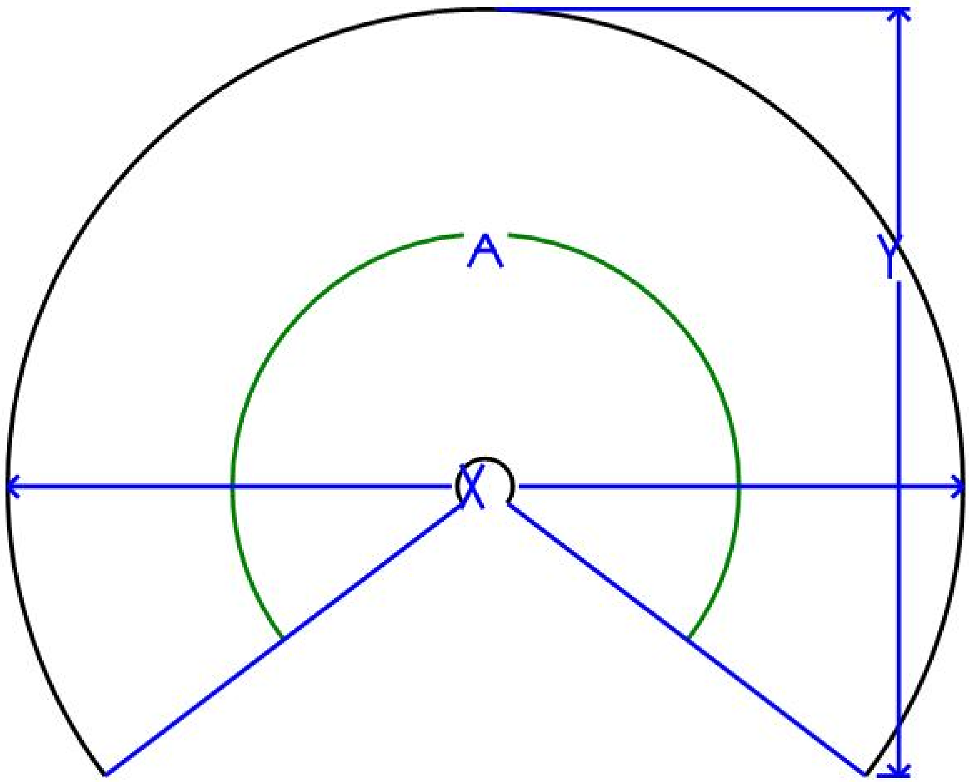
- 10 – 5 cm diameter (5cm long) plastic vaccum connector pipes
- 20 gauge metal wire
- Glue (plumbers goop)

A 500ml nalgene wide mouth bottle, with a circular hole cut in the side was used as the catch bottle for the top of the trap. 5 cm diameter (5 cm long) hard plastic piping (found in the plumbing section of a hardware store) was placed in the hole of the nalgene and glued in place allowing liquid to stay in the nalgene while it was attached to the funnel. Metal wire (20 gauge) was also wrapped around and glued onto the nalgene bottle on either end and then hooked onto the plastic funnel to hold the bottle in place.

Common problem: Leaking of the top bottle.
Solution: Bottles designed for our purpose can be made out of plexiglass. Contact us in Rimouski for more information.

- Rope
- 40 (10 inch) nails or posts

The wooden posts will need to be secured to the ground using guy wires (simple nylon ropes) and nails or posts hammered into the ground.



Cone Dimensions	
Outside Dia	85.0000
Inside Dia	5.0000
Height	40.0000
Cone Angle	45.0000

Flat Pattern Dimensions	
Large Radius	60.1041
Small Radius	3.5355
Angle A	254.5584
Width X	120.2082
Height Y	96.5091
Cord	95.6488
Full or Half Cone	Full

Figure 3. Cone construction details (measures other than angles are in centimetres). Calculations tailored to the size of the plastic sheets can be made with the ConeCalc website. http://www.i-logic.com/conecalc.htm

PROCEDURE 2 – ARTHROPOD PHENOLOGY - DIRECT OBSERVATIONS

PURPOSE

Pitfall trapping is the most efficient method by which to document the phenology and abundance of arthropods. However, some species such as adult butterflies are not easily captured in pitfall traps, even when they are modified to catch flying insects. Adding observational data will provide an additional level of accuracy in estimates of arthropod phenology. Direct observations can be used alone to provide an index of seasonal phenology where logistical difficulties prohibit pitfall sampling.

TIME PERIOD

Observations should be conducted throughout the entire field season.

PROCEDURE

The presence or absence of bumblebees, moths and butterflies (Fig. 4), biting mosquitos and craneflies (Tipulidae) must be recorded every two days. Craneflies are easily distinguishable from biting mosquitos (see Fig. 5). If possible, a qualitative description of abundance should also be recorded for each group. Abundance categories will be; low, medium, high and very high density. Observations can be taken incidentally while conducting other field activities.

Figure 4. Common moths and butterflies.

Figure 5. Tipulidae (Crane Fly)

PERSONNEL

One person should be in charge of making sure that the absence/presence data is collected every two days throughout the season.

MATERIAL

- Field notebook and pencil

LITERATURE

Holmes, R. T. 1966. Feeding ecology of the red-backed sandpiper (*Calidris alpina*) in arctic Alaska. Ecology **47**:32-45.