Arctic Shorebird Demographics Network
Breeding Camp Protocol

Version 2 – May 2011

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Cover photo depicts four of the Network Focal Species including Red Phalarope, Western Sandpiper, Dunlin, and Semipalmated Sandpiper. Photographs taken in Barrow, Alaska by N. Burell/USFWS.

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INTRODUCTION

Recent shorebird trend analyses indicate that many North America shorebirds are declining, but we do not know why (Morrison et al. 2006). The goal of the Arctic Shorebird Demographics Network (hereafter termed "Network") is to collaboratively conduct demographic studies on several shorebird focal species that will help determine factors limiting their population size. The Network will measure demographic rates such as adult apparent survival, annual productivity, population age-structure, etc. on the Arctic breeding grounds. Additionally, site-specific ecological and environmental variables (e.g. food resources, prey and predator abundance, weather, etc.) that influence demographic rates and are influenced by climate change and other anthropogenic forces will be measured and incorporated into the analyses. Finally, the Network will substantially increase our ability to address a wide variety of other science and conservation goals that can only be examined at a regional or global level (e.g. migratory connectivity studies that require marking individuals over a large area, the collection of tissue samples, analyses of contaminants, etc.).

Rationale for demographic approach

The existing large scale monitoring efforts developed under the Program for Regional and International Shorebird Monitoring (PRISM) are aimed at providing population size and trend estimates, along with collecting accompanying environmental data to assess habitat use and help infer range and distribution However, the current PRISM program does not provide information on the mechanisms behind declines (e.g., poor nesting success and survival, chick survival, or adult survival) and when (e.g., breeding, migration, non-breeding) shorebird populations are likely to be limited. Determining the stage of the annual cycle when shorebird populations are most negatively impacted will allow targeted conservation actions in the future to address population declines.

Network participation

The Network is open to participation by any collaborators who are actively conducting shorebird studies in the sub-Arctic and Arctic regions of the North American Arctic, and can implement the protocols designed by the group as a whole. Current participants span the entire Alaskan and Canadian Arctic (Figure 1), and include study sites (from west to east) near Nome, Cape Krusenstern, Barrow, the Ikpikpuk River, Prudhoe Bay, and the Canning and Colville Rivers in Alaska, as well as at the Mackenzie River Delta, East Bay, Churchill and Bylot Island in Canada.

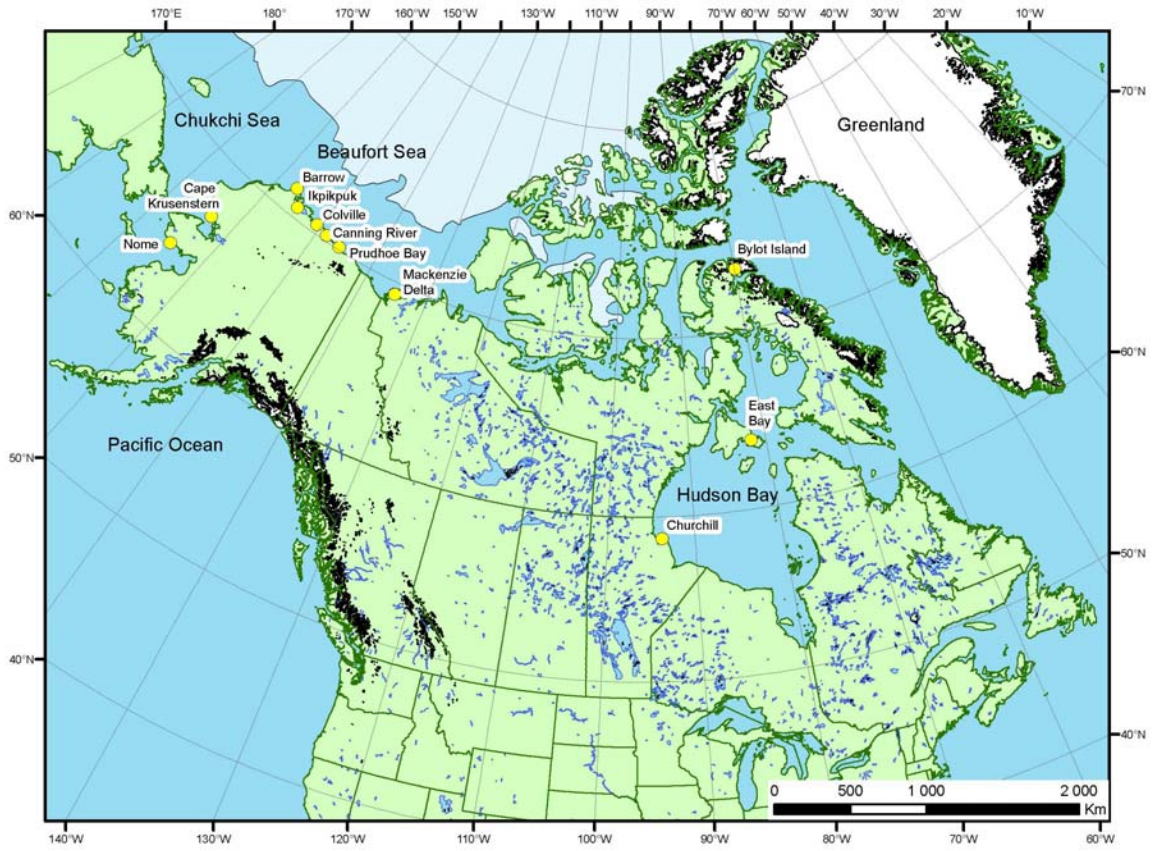


Figure 1. Arctic Shorebird Demographics Network study sites across the North American Arctic for 2011. Map prepared by J.F. Lamarre.

Network objectives

Our overall objective is to assess factors limiting population growth of shorebirds through an international, large scale collaborative effort that spans sites from Western Alaska to the Eastern Canadian Arctic and includes field work for up to 4 or 5 years/site. At each site, we will also collect information on environmental and ecological variables that may influence these factors. This approach will allow a large temporal and spatial perspective on factors limiting shorebirds populations in the Arctic. Our specific objectives include:

1) Collect demographic data (focused on adult survival but may include all or some of the following: breeding propensity, nest survival, hatchability, brood survival, mate and site fidelity, juvenile survival/recruitment, age at first breeding) on a select group of Arctic-breeding shorebirds (hereafter termed "focal species").

2) Document contemporary patterns of species presence and abundance of shorebirds, and when possible assess how species assemblages and abundance have changed historically.

3) Document breeding chronology and habitat use.

4) Collect information on the phenology and abundance of avian and mammalian predators of shorebirds and alternative prey such as lemmings.

5) Collect data on local weather and snow conditions, the timing of insect emergence, inter-annual patterns in habitat characteristics, and other variables that will help assess impacts of climate change on shorebird breeding ecology.

6) Maximize the biological capacity of the Network by participating in projects that span large geographic and temporal scales. This might include investigations of shorebird health, migratory connectivity, and ecotoxicology.

Focal species

A pre-field season assessment of Network site leaders indicated a variety of species were present at current Network sites (Table 1). Based on the number of nests likely to be found and the number of adults capable of being captured (ideally approximately 50 nests monitored and 30 birds captured and marked/site), we chose to focus our efforts on gathering demographic data on Semipalmated Sandpiper, Dunlin, Western Sandpiper, Red-necked Phalarope and Red Phalarope, designating them as first tier focal species. Pectoral Sandpiper, Whimbrel and Semipalmated Plover will be second tier focal species due to lower nesting densities or fewer sites with adequate numbers. We anticipate that project leaders at each field site may wish to study other species and other topics. Being part of the Network does not prohibit this, but only requires that efforts are made to collect all or portions of the data outlined in Tables 2 (a, b).

Table 1. Network focal species, avian target species and relative abundance of each species at each Network site. 1- represents common breeders (n=30 pairs/ year), 2 - represents low abundance (n<30 pairs/ year), or represents transient populations that have large annual variation in site fidelity and will be studied opportunistically.

	Alaska						Canada			
	Cape Krus	Barrow	Canning River	Ikpikpuk	Nome	Prudhoe Bay	Bylot	Mack Delta	East Bay	Churchill
SESA	1	1	1	1	1	1	0	2	2	2
DUNL	1	1	1	1	2	2	0	0	2	2
WESA	1	2	0	0	1	2	0	0	0	0
RNPH	2	1	1	2	1	2	0	2	0	2
REPH	0	1	1	2	1	2	2	1	1	0
PESA	2	2	2	2	2	2	2	1	0	0
AMGP	0	1	2	2	0	2	2	2	0	2
SEPL	0	2	2	0	2	0	0	0	2	1
WHIM	0	0	0	0	0	0	0	1	0	2
WRSA	0	0	0	0	0	0	2	0	2	0

General framework and Network monitoring strategies

The Network utilizes on-going projects and field camps and helps to support new sites that are willing to contribute to collecting some level of demographic data. The Network maintains flexibility by encouraging project leaders to choose the intensity of effort that is reasonable for each camp. Two priority levels have been established in order to standardize efforts. Priority level 1 includes minimum Network monitoring efforts, whereas priority level 2 methods are intensive methods to be accomplished if possible. Table 2a provides a summary of the difference between minimum and intensive efforts for assessing adult and nest survival. More plots or a larger nest search effort are needed for low density nesting areas to get adequate sample sizes.

Minimum nest search effort

This level of nest searching effort will focus on finding a sufficient number of nests necessary for adult survival estimates (n = 30). A loosely defined search area will be established the first year. Care should be taken in area selection, since the search area **must remain** consistent across years (Figure 2, outside white boundary). This area will be searched, focal species nests will be located and monitored to determine fate, and adults will be captured and marked for adult survivorship. The goal is to find 20 to 30 nests/focal species inside the study area to uniquely mark 30-50 adults/year. If you capture less than this, or if you only have 30 pairs of a particular focal species at your site, this is still okay – the uncertainty around your survival estimates will just be greater. This effort will yield adult and nest survivorship estimates for the focal species.

Intensive nest search effort

Intensive nest search effort will focus on establishing permanent nest survival plots where we attempt to find and monitor nests of all shorebirds and other avian species. Nest survival plots (Figure 2, white squares) will have standardized intensive nest searching methods and effort. Plots in size from 10 –ha to approximately 16 –ha depending on the nest density of the area. Plot size and shape may be variable across Network sites but each site's plot **must remain**

the same across years. In contrast to the minimum nest search effort, adult birds WILL NOT be captured within nest survival plot boundaries, but rather individuals will be captured outside or in close proximity to the boundaries of the intensive plots. This effort will yield adult and nest survival estimates for the focal species in addition to standardized species diversity and nest density of tundra breeding birds. We will also be able to compare nest survival where adults were and were not banded.

Figure 2. Example of a Network site layout that includes two intensive nest search plots (white squares), a greater search area for capturing adults off plot, and ecological monitoring components (e.g. predator surveys, food resources and lemming transects.)

For both the minimum and intensive nest searching approaches, we will gather ecological and environmental monitoring data on potential shorebird nest predators, alternative prey (lemming counts), terrestrial and aquatic food resources, weather, snow melt and surface water. Details of how each parameter is monitored are illustrated in Table 2b.

Table 2a. Priority level (1 = primary, to meet minimum goals; 2 = secondary, to meet intensive), Network objectives, methodology, metrics, and workforce required to accomplish objectives for shorebird demography studies.

Priority level	Objective (s)	Methods	Metric (s)	Workforce required for min. Network site	Workforce required for intensive Network site
Shorebird Adult Demography, Migratory Connectivity and Other Species-specific Ecological Traits					
1	Monitor individual annual and within-breeding season apparent adult survival of focal species	Capture and color banding adults and resighting	Adult survival	Mark at least 30 individuals within or off nest plot. Resight opportunistically within season. 1 Resight/Bander: 8-10 hrs/day for 4-6 weeks Adults banded on and off nest survival plots	Mark ≥ 30 – 50 individuals off nest searching plots. 2 Resight/Bander: 8 - 10 hrs/day for 4-6 weeks Adults banded only away from nest survival plots
2	Determine age structure of focal species populations for demographic modeling (applies to WESA, SESA, DUNL primarily)	Age determination of captured birds via plumage in the field or stable isotope techniques in the laboratory	Age at first breeding		
2	Collect appropriate specimens from individual birds or equip birds with instruments for Network projects and collaborative studies	Collection of feathers and blood. Equip birds with instruments, etc.	Disease or contaminant, migratory connectivity		
Shorebird breeding ecology					
1	Document nest survival	Nest fate, regular nest monitoring	Hatch success, nest survival	Only monitor nests (for survivorship) on those discovered to trap birds on.	Intensive and standardized nest searching effort with regular monitoring visits.
2	Document species assemblages and abundance	Standardized nest searching effort / unit area – must be consistent across years at a given site.	Identify species' presence/absence, nesting propensity, nest density	Depends on size of search area and site density of focal species.	Depends on # plots, 1 full time nest searchers/ 16 ha plot in high density areas ~ 8 hours/day for 4 weeks
2	Document breeding investment	Count and measure eggs	Clutch size, egg size		
2	Document breeding phenology	Nest age determination	Initiation date		

Table 2b. . Priority level (1 = primary, to meet minimum, 2 = secondary, to meet intensive), Network objectives, methodology, metrics and workforce required to accomplish objectives for ecological monitoring.

Priority level	Objective	Methods	Metric(s)	Workforce required for minimum	Workforce required for intensive
Ecological monitoring					
1	**Predator – lemming index:** Document relative abundance of avian and mammalian predators	Regular (daily or weekly) counts. Point counts for some sites	Abundance estimates or minimum counts.	Min seasonal survey -- 3 times/season (early, mid, and late)	Daily predator/lemming counts
1	**Lemming index:** Document relative and seasonal variation in abundance of lemmings	nest line transect, daily observations quantified by person-hour effort during "low" lemming years, line transects in "high" lemming years, live trapping at some sites	Index to over-winter abundance, seasonal variation in relative abundance in live animals	1 early season nest survey Seasonal (early, mid, late) abundance checklist counts, or weekly abundance transects on "high" lemming years	1 early season nest survey Weekly lemming checklist counts, or weekly abundance transects on "high" lemming years Live trapping
1	**Daily avian/mammal species list:** Document presence/absence of species; and large changes in number of individuals	Technicians enumerate observations throughout day	Daily check list with estimates of numbers seen, effort, and locations visited.	Daily 20 mins.	Daily 20 mins.
1	**Snow and surface cover:** Monitor snow melt progression	Plot surveys	% snow / water/ land cover at fixed interval	2 hrs/search area every other day, first 5 to 15 days of season, throughout season	2 hrs/ plot every other day, first 5 to 15 days of season, throughout season
1	**Food resources:** Document seasonal change in insect emergence and abundance: Terrestrial (wet and dry locations) and aquatic (surface sampling).	Modified pitfall traps, aquatic active sampling traps in tundra ponds	Bi-daily calculations of insect mass, abundance, species richness.	1 -2 hours/day every other day for sample collection and numeration during peak emergence, less frequent early and later	1 -2 hours/day every other day for sample collection and numeration throughout field season
1	**Weather:** Document within and inter-annual variability in weather conditions	Establish and monitor automated weather stations	Daily Min/Max temp, precip., wind speed/ direction	2 hr installation and retrieval, weekly download of data	2 hr installation and retrieval, weekly download of data

Personnel considerations and seasonal work responsibilities

The minimum goals of the Network can be accomplished with a 3- 4 person crew that arrives shortly before the snow has begun to melt and departs approximately 1 to 2 weeks after peak hatch. However, a crew size of 4 to 5 is more suited to accomplish all of the objectives. Ideally, two people work for the first month of the season as primary nest searchers before switching to helping two other people who resight banded birds from prior years, band new birds, assist with rope dragging and help with Network side projects. All staff work to collect the environmental variables. Table 3 illustrates an abbreviated work schedule for the season.

Start dates are dependent on annual variation in snow melt and approximate time when the tundra becomes snow-free. If possible, camps should be established shortly before or when snow melt is just beginning (typically a 4-7 day period). Field season lengths vary by site and at a minimum are 6 weeks (this includes 1 week of set up, 3 weeks of incubation and 2 weeks of hatch). 7 weeks is better to capture the variation in initiation and hatching dates and a maximum of 10 weeks is needed if brood-survival or post-breeding studies are conducted.

Table 3: Seasonal work schedule for minimum and intensive efforts. Gray indicates both minimum and intensive effort sites will conduct surveys and black lines indicate intensive effort sites only.

	Week 1	Week 2	Week 3	Week 4	Week 5	Week 6	Week 7	Week 8
Plot setup								
Nest searching								
Nest monitoring								
Resighting								
Banding								
Food resources								
Predators								
Lemmings								
Snow / Surface								

Geographic information datum and coordinate requirements

Geographic information must be collected for locations (plot locations, weather station, grid stakes within plots, transects lines, predator count locations, etc.) and boundaries of the study areas using a GPS unit. To ease future GIS applications, we insist all data be collected (or converted prior to data submission) in **latitude/longitude decimal degrees** (e.g., -145.78675N degrees, 56.3643W degrees) and the **WGS 84 (or NAD 83)** datum. These data should be saved in the geographic meta-database that accompanies each camp (see dataforms Network site establishment form).

Adjust time on all GPS units (e.g., -9 hours from Greenwich time for Alaska). Refer to: http://wwp.greenwichmeantime.com/time-zone/north-america/canada/time-zones.htm to obtain Greenwich time zone for your camp.

ADULT RESIGHT METHODS

Objectives:

1. Estimate annual survival of individually color-marked focal species at each Network site
2. Obtain between season resighting rates of individually color-marked focal species at each Network site

To estimate survival and resighting rates, we need to record the presence of as many birds marked in previous years as possible. The primary way we do this is to recapture them at nest sites, or resight them either near their nest or when found opportunistically as people conduct work around the study area. Obtaining survival estimates is one of the primary goals of the Network and subsequently a fair amount of effort needs to be directed towards this task.

Recapture or Resighting at Nests:

Many times it will be unknown if an adult attending a nest is marked already. It could be that one adult or possibly two adults are already marked. Some individuals may become quite skittish and not stay around the nest site as you approach and thus you never know they are marked until you capture them (e.g. phalaropes). Other individuals will stay very near the nest, especially later in incubation when they become protective of the nest itself. For the latter individuals, it is possible to read the color band combinations of the adults as opposed to recapturing them. This can be particularly helpful if you are having trouble recapturing a bird. Not recapturing the bird however prevents you from obtaining valuable data on exact metal and band colors, body size measurements, molt, and samples (blood, fecal material, feathers, etc.). For 2011, a focused effort will be made to recapture Dunlin equipped with geolocators (see this protocol for removal of geolocators). It is best to recapture birds if time allows.

Opportunistic resightings (away from nest sites):

Whenever an observer sees a color-marked individual, an attempt should be made to record and verify the unique color combination of the bird. These opportunistic resightings will be used to confirm annual survival when you are unable to recapture individuals on the nest. Try to resight birds as much as you can, try to get at least 2 visual 100% confident resights and more if the resighting is not 100% confident. If birds have been captured they do not need to resighted repeated afterwards. Opportune times to resight color-marked adults include during initial visits to plots when few birds are nesting yet and you are carrying out other tasks (snow / surface cover, lemming nest survey and other regular surveys). You can also resight birds during regular nest searching or banding should a marked bird present itself. Good times and places to search for marked birds are during early break-up when birds are concentrated near the first open water areas to feed and there are few nests to find. Another good time is after most birds have lost their nests but are still present on the study area. They will frequently feed in the study area for several days and are quite approachable. Observers should also record band combinations of birds seen off plot when traveling to and from camp or during other activities.

Some species return to breed near their previous breeding sites, and knowledge of past breeding locations for marked birds may therefore help to focus search efforts. Similarly, a list of birds recently marked or resighted in recent years will help to determine whether additional resighting effort is needed for certain species. These summaries of birds that are expected to be present in the plots could serve as a useful resource for workers in the field, and should be

prepared ahead of the field season. Similarly, it is useful for workers to keep track of the location and color schemes of birds banded in the current season in their field notebooks, so that undue effort is not expended to read bands that were applied in the current year. The latter can be reduced if a standardized method is made to band birds in each year (i.e., a form of cohort banding where perhaps the green flag and site color band code is always placed in part of the bird's legs in a given year).

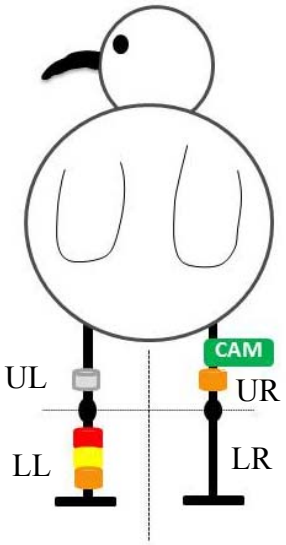
If you do resight a marked bird at a nest, be sure to enter its color band combination in the following places:

- For camps using ASDN nest and banding books:
    - Record color combination in the nest databook to indicate that you have identified the parent(s) of this nest
- For camps that do not use these forms notes can be taken in field notebooks
    - Transfer all resights at the end of each day from field note books to the *encounter data form*. It is probably best to put one person in charge of the encounter data form to record resighting info from the field crew at the end of each day.

Suggestions for how to resight a shorebird during the breeding season:

Shorebirds can be difficult to resight during the breeding season because their legs are concealed by vegetation or terrain. Consequently, we rely on both engraved flags (at least for focal species) and unique color combinations. The engraved flags are easy to read if the bird is in the open and the flag is oriented right but difficult to read in vegetation. The unique color band codes are easier to read in the vegetation. Below we describe how to collect resighting data and provide a form for recording these data. These forms can be put into the banding and nesting record books or used by themselves. We encourage people to write on the forms as opposed to a Rite-in-the-Rain notebooks because the form prompts people to record all the data required for easy editing and data entry later.

The most efficient way to gather resighting data is to establish a short hand system for taking notes in the field that allows you to record data as you gather it. For example, you can make quadrats in your field notebook in order to keep track of the colors you have figured out. Each quadrant represents different parts of the legs and breaks the color combination into 4 quadrants (Figure 3). Record colors from top to bottom, from the **bird's left** to the **bird's right**. Color codes are listed in Table 4. The following graphics show the standard shorebird short hand for recording marked shorebirds via resighting, with examples of the color-band combinations from the pictured bird.



Key to short hand codes for recording colorband combos:
UL = Upper left - *left tibia tarsus*
LL = lower left - *left tarsus*
UR = upper right - *right tibia tarsus*
LR = lower right - *right tarsus*
\- = no bands were present
?? = unknown color or bands
/ = separates tibia tarsus from tarsus were present
: = represent switching of leg from left to right

Example of short hand notation:
_UL_/_LL_:_UR_/_LR_
This bird's colors can be noted as such:
m /r,y,o :gfe, CAM, o/-

Figure 3. Example of ASDN color marked bird as seen from behind.

Table 4. List of color band marker types and standard color codes for unique Darvic colors.

Code	Marker type	Code	Marker type
m	metal band	r	red
y	yellow	w	white
bk	black	gy	gray
dg	dark green	gf	green flag
db	dark blue	bkf	black flag
lg	light green	wf	white flag
lb	light blue	wfe,xxx	white flag engraved
o	orange	gfe,xxx	green flag engraved

<u>Data to record for each bird resighted:</u>
Date: dd-mm-yyyy
Time: 24-hr
Observer: initials
Species: 4-letter BBL/CWS code
Plot or study area: code specific to each site
Reference plot stake/landmark: code specific to each site
GPS Location (lat/long): NAD83 Decimal degrees
Color-band combo upper left and lower left: _____ / _____
Color-band combo upper right and lower right: _____ / _____
Engraved flag code (if observed):
Observer confidence scale : **3 2 1** (select one)

3) All bands observed and recorded. Confident that location and colors of bands are correct. Confident in engraved flag code (if present). Observed bird and checked notebook several times to make sure written down correctly.

2) Most or all bands observed and recorded; if all bands recorded, exact location/order not certain, or colors not certain (i.e. red (r) or orange (o); read part of alpha-numeric flag code (if present); recorded in notes of all known/unknown parts of leg.

1) Locations of several color bands observed, although not all bands seen; exact location of some bands unknown; recorded in notes of all known/unknown parts of leg.

Notes: (please record all that apply)

Paired with color combo:

Behavior: territorial, singing, other

General location

Notes of colors seen or not seen very well (i.e.: "confidence "3" on all but UL. Could see 1 yellow, but couldn't see what was above it…" or "confident of band combo but R and L legs may be switched")

How to record resighting data:

The *encounter data form* is for recording opportunistic observations of marked shorebirds at or away from the nest. DO NOT use it for recording captures of birds. These will be in the banding database and be recorded as a recapture.

Encounter database and verification of resighting records:

Each sighting needs to be verified by the banding records to confirm that this color-band combination has been used on an individual of that particular species either this year or in a prior year. At the end of the day resighting records should be verified by confirming the color combo in the master banding database (accumulation of all banding data from that site in one file). Care should be taken to retrieve all records from field personnel if they don't enter the data on a standard sheet in the banding or nest record databooks (the recommended approach). See readme file in electronic database for more specific instructions on verifying records.

Adult survival capture

Objective

We would like to capture and uniquely mark 30 (but more is better – 50 to 100 is ideal) individuals of each focal species per year and make a concerted effort to resight individuals between breeding seasons. Our efforts to mark birds on the breeding grounds will also create opportunities for resighting individuals during migration and the non-breeding season.

Unique marking scheme

All birds captured for Network projects will be banded with a unique CWS/USGS BBL metal band and each site will have a unique site code associated with the flag (Table 5). Three of the focal species (e.g. SESA, DUNL, PESA) will be marked with both engraved alpha-numeric flags in addition to unique color combinations. Red and Red-necked Phalaropes will have unique color combos in addition to site specific flag legs but due to the low probability of resighting during the non-breeding season, phalaropes will not have engraved alpha-numeric flags but will have blank flags. Other species captured will be marked according to the project leaders' discretion. The Network will help organize the engraved alpha-numeric codes but will not organize the unique color bands for individual birds. Each project leader is responsible for coordinating color band codes for species in their study area and for using site-specific codes that

are coordinated through the Pan American Shorebird Program (PASP) currently supported by the Canadian Bird Banding Office (bbo_cws@ec.gc.ca).

Table 5. Unique site-specific codes for Network Sites and collaborators.

Site	**Site-specific flag colors**
Canadian sites:	
East Bay/ Coats Island	White flag over red
Mackenzie Delta	White flag over orange
Churchill	White flag over dark blue
Bylot Island	White flag over dark green
Alaskan sites:	
Barrow	Dark green flag over or above red (yellow in prior years)
Cape Krusenstern	Dark green flag over orange
Nome	Dark green flag over or above dark blue
Canning	Dark green flag over or above dark green
Ikpikpuk/Prudhoe Bay	Dark green flag over or above light blue
Colville	Dark green flag over white

How to apply metal and color bands to a bird

Metal bands are applied with special banding pliers (not needle nose pliers). To remove metal bands from the wire string, use a band spreader or if your banding pliers has a split pin built into the side, insert split pin into center of band and open handles evenly. Try to open band evenly. Place open band in proper sized hole on pliers, and then slide around leg where specified for your site's color marking scheme and close gently. Bands should be placed on birds in sequence if possible to make reporting data to the Bird Banding labs easier.

Color bands for smaller species (size 1B to 3) are usually 'butt-end' bands, similar to metal bands, while those for species size 3A and larger are usually 'wrap-a-round' bands. Butt-end bands are applied with a thin metal 'shoehorn' applicator: a smaller size applicator is used for bands up to 1A, and larger size applicators for size 2 and larger bands. The band is placed on the applicator with the opening in the band towards the depression in the shoehorn, and the band is slid up the applicator until the band is sufficiently open to fit on the leg. The applicator is laid against the leg, and the band is slid off the small end of the applicator onto the bird's leg. It is important to stretch these bands no more than is necessary to put them on the leg, and to ensure that the color band is completely closed on the leg. It may be necessary to click the edges of the band under each other with one's fingers to ensure that the band is completely closed. Wrap-a-round bands are twirled carefully onto the bird's leg, ensuring that the leg is not injured and the bands are not opened more than necessary. Again, these bands may be tightened with the fingers after they are on the bird. Ensure that the bands rotate freely around the leg, but are not so loose that they can pass over the 'knee' joint or 'ankle'. Wrap-a-round and butt-end bands should be permanently sealed with a battery-operated soldering gun. Only solder on the flat edge of the band, not the top or bottom areas near the bird's leg.

Suggestions for avoiding problems with color banding shorebirds:

- Do not put three color bands above the joint since the top color will get lost in feathers
- Do not put two bands of the same color next to each other.

- Do not put a dark green band next to a dark blue band, etc.

Applying focal species alpha-numeric flags

Use shoehorn applicators to place the flag on the bird (**with engraved numeric code upright – very important since certain codes can be read in either direction – e.g., E6 or 9E**) or use one's nails to open the flag slightly. Open the flag as little as necessary, so that the flag is not stretched (otherwise, remove and reshape later). Flag tabs should be sealed along the edges with a battery or propane operated soldering pen (http://www.all-spec.com/products/BP860MP.html), Markely Solvent Cement, or Superglue (e.g. cyanoacrylate) if the gun malfunctions. Glue is applied to the tabs of very slightly opened flags with an object such as the tip of a small screwdriver. Pliers or close pins are used to hold the flag tabs closed for about 20 seconds until the glue is set. Then the pliers are carefully removed to prevent the flag from opening. **For alpha-numeric flags, be very careful not to damage the letters when soldering. If you cannot do this with the solder gun, then use glue to seal them.**

Data recorded with each bird

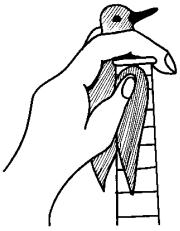
1. Bander (first name initial and last name e.g. loring for Lewis Oring)
2. Date (dd-mm-yy)
3. Time (24 hr)
4. Nest ID (unique code of spp and nest number)
5. Plot identification (unique plot code, alpha-numeric)
6. In or off plot (Was the bird captured on or off the plot)
7. How captured (bownet, mist net, walkin, standing decoy, other define)
8. Capture status (laying, incubation, brood, post-breeding, unknown)
9. Bandnumber (USGS/CWS 9 digit unique number)
10. Species (4-letter BBL species code)
11. Color combo: Upper Left /Lower Left**:** Upper Right /Lower Right . Place "/" between upper and lower part of the leg, ":" to separate legs, "," between color band colors or flags on one portion of the leg, and use gf to indicate green flag, wf to indicate white flag, o = orange, y = yellow, r = red, db = dark blue, bk = black, dg = dark green, lg = light green, lb = light blue, m = USGS or CWS metal band; if there is an engraved flag use the symbol "gfe" followed by the code [e.g., gfe,xxx].
12. Recapture (yes or no, and if yes, whether it was recaptured from prior year or this year)
13. Picture taken (yes or no, be sure to first take a picture of the banding sheet and then of the bird so it is possible to determine the identity of each bird)
14. Flight feather molt (score 0- 5)
15. Tail feather molt (score 0- 5)
16. Body molt (score 0, 2 – 5)
17. Exposed culmen (nearest 0.1 mm)
18. Total head (nearest 0.1 mm)
19. Diagonal tarsus (nearest 0.1 mm)
20. Flattened straightened wing (nearest 0.5 mm)
21. Bird Weight (nearest gram or nearest 0.1 if digital)
22. Bag Weight (nearest gram or nearest 0.1 if digital)
23. Fat (score 0 – 7)

24. Blood for avian malaria Plasma, RBC, Amount of blood (in micro liters), type of capillary tube (Plain/Hep)
25. Blood for genetics in Longmire (Y/N) Amount of blood (in micro liters), type of capillary tube (Plain)
26. Blood for RNAlater (Y/N) Amount of blood (in micro liters), type of capillary tube (Hep)
27. Feather sampled (Specify which feather was pulled and from where according to standard feather codes e.g. "brst, 10pL + 10pR= 10 secondary on the left and right sampled)
28. Sex (Male, Female, unknown)
29. Method of sex (culmen, morphology, plumage, brood patch, cloaca size, wing, overall size, egg in oviduct)
30. Age (chick, HY, SY, AHY, ASY)
31. Method of age (e.g. plumage, weight, recapture)
32. Release status: Band and Release, band and escape, Release unbanded, injured, band and release, mortality
33. If geolocator, record exact date and time removed from the bird (VERY IMPORTANT).
34. GPS location of nest

Morphological measurements

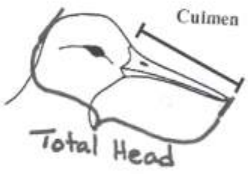
Figures from Prater et al 1977 or Gratto-Trevor 2004, Photos B. Lewis/USFWS

Wing length: maximum length with the wing **flattened and straightened**, measured with a wing ruler (to nearest mm) from the bend in the wing to the last primary. Be sure to hold the wing close to the body, not at a right angle to the body when measuring.

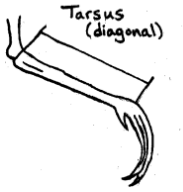


Exposed culmen: measured with calipers (to the nearest 0.1 mm) from the edge of feathering to the tip of the bill. Since bills of most shorebirds are very sensitive, hold bill lightly with the fingers, with the calipers resting on one's fingers and not the bird's bill. The calipers must remain **perpendicular** to the bill and not angled to measure a downturned bill. This is especially important for long-billed species such as Dunlin and Whimbrel.

Total Head: measured with calipers (to the nearest 0.1 mm) from tip of the culmen to the notch at the back of the head at a perpendicular angle.



Diagonal tarsus length: measured from the slight indentation below the 'knee' joint to the indentation above the longest toe (to the nearest 0.1 mm).



Mass: measured with a hanging Pesola scale or a digital scale (to nearest 0.1 g). If using a Pesola scale the scale needs to be held by the top ring or hook and allowed to dangle freely, while being protected from the wind. Place the bird in a weighed cloth bag, or plastic cone with the bill protruding from the bottom. The cone should be firmly attached to the teeth of the clip at the bottom of the scale. It is very easy to release shorebirds from plastic cones or cloth bags, by sliding them out into the palm of the hand until one can hold them in the banding grip. Mass should be the last measurement taken as it is the best opportunity for a bird to escape during handling.

Molt

Examining birds for body and flight feather molt can indicate age as well as provide information on timing and extent of molt, which is poorly known for most shorebirds. To describe **body molt** the bird is normally divided into five regions: head, neck, back, breast, and abdomen. Look for feathers emerging to detect molt, although be careful for cases where new feathers have already fully emerged. This is obvious when, for example, Red Phalaropes are no longer red but are gray. The extent of replaced body feather codes are as follows:

0: all old body feathers
2: a few new body feathers
3: about half body (30- 50%) replaced
4: most replaced (60 -90%)
5: all new (100%). **NOTE: there is no score of 1 here.**

Feather tracts and individual feather names are standardized and abbreviations are used on datasheets (see Figure 4).

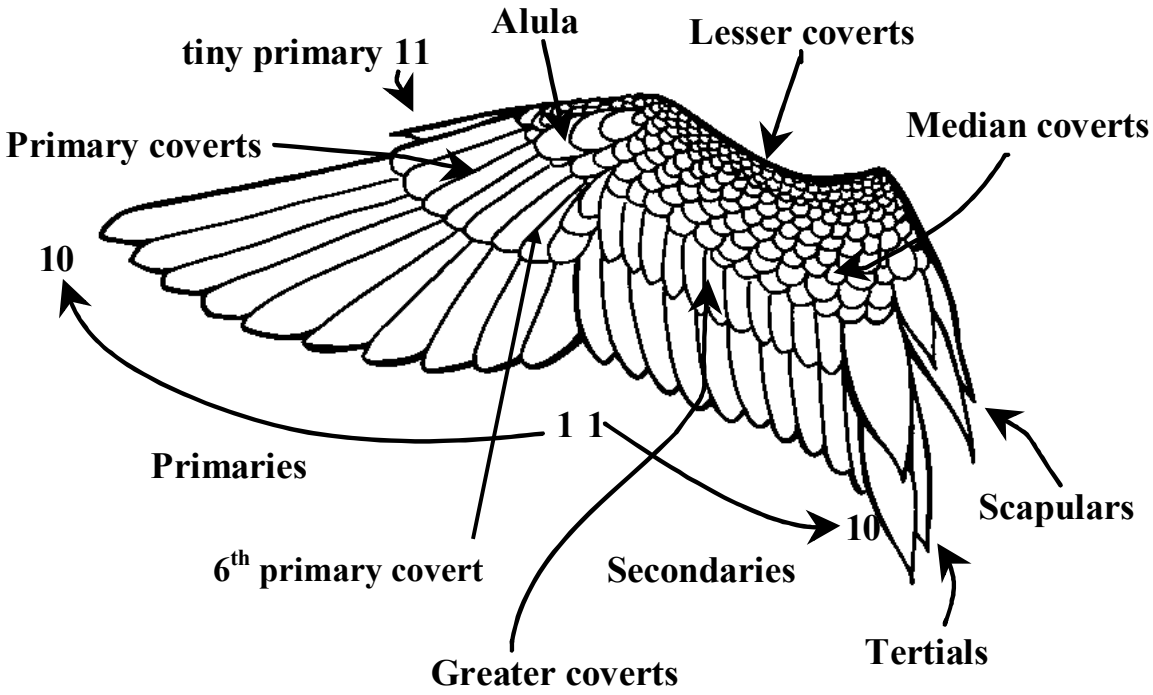
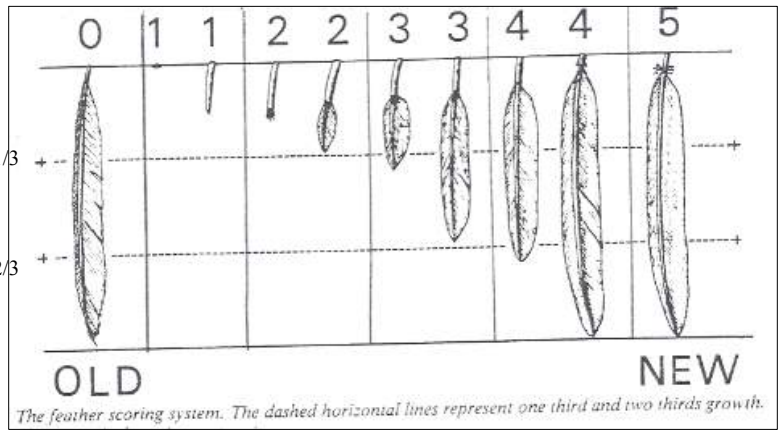


Figure 4. Specific names of each flight feather and their standard abbreviations are as following (pp=primaries, ss= secondaries, t=tertials). Figure from Gratto-Trevor 2004.

Flight feather molt scores are usually more complicated, with the condition of every primary, secondary, tertial and tail feather described (Figure from Ginn and Melville 1983):

Their condition is noted as follows:

0: old feather
1: feather missing or completely in pin
2: just emerging from sheath to one-third grown
3: one to two-thirds grown
4: more than two-thirds grown but still with waxy sheath at base
5: new feather fully developed and without waxy sheath



The feather scoring system. The dashed horizontal lines represent one third and two thirds growth.

Sex determination

For breeding shorebirds, sex can usually be differentiated by plumage, exposed culmen wing length, overall size or behavior. However, there is significant geographic variation prohibiting a standard morphological measure across the Network. A copy of the Pyle Guide Part II (2008) will be sent to sites as a reference guide that includes subspecies measures to determine how the focal species can be sexed. For species with no observable unique traits, sex can be determined with genetic techniques. Evidence of recent egg-laying (extended cloaca) or even a bulge indicative of an egg inside a bird can also be used. This is especially helpful for sexually monomorphic species. There is a location on the data form to record how the bird was sexed – please be sure to fill this in as it allows us to ascertain confidence in the field sexing technique.

Culmen length is commonly the most sexually dimorphic measurement in sandpipers, with female culmen length averaging longer than males. Other measurements may provide more information in other species. Measurements must be used carefully, however, especially when a study begins in a new area or an area where different populations of the same species overlap. For example, while the sex of >90% of Semipalmated Sandpipers can be accurately determined by measurements in a single breeding study site (Gratto-Trevor 1987), this approach is far less accurate when measuring birds in an area where eastern and western breeders mix during migration (e.g. Harrington and Taylor 1982). As well, the degree of overlap between sexes in measurements may vary from one breeding site to another.

Age determination

It is sometimes possible to determine the age of an individual by closely examining the condition of the flight feathers (primaries and secondaries) and the wing coverts (primary, secondary, median and lesser coverts). During the breeding season adult birds can be separated

into three groups. Second year (SY) can be identified by the presence of buffy edges on their innermost median coverts, and also by very worn primary feathers. After second years, (ASY) have white edged innermost median coverts and in general, will have less worn primaries and wing coverts. Birds that do not have these distinctive traits are aged as After Hatch Year (AHY). If you are uncertain about the age of adults, AHY is a conservative catch all. Most birds are usually considered AHY rather than SY/ASY, please be conservative in assigning ages to all individuals. Appendix I illustrates the specific criteria for determining age of three focal species (e.g. DUNL, SESA, WESA).

For *Calidris* sandpipers in general, most or all Second Year birds molt the most important outer primaries only, as well as inner secondaries. These birds may be identified as Second Year (between at least May through September) by the contrast between fresher outer primaries and more worn inner primaries. If all feathers had been molted the previous winter, outer primaries, which suffer the most wear, would be more worn than inner primaries. Note that the percentage of Second Years in these species with this Partial Post Juvenal Wing (PPW) molt can be variable among populations and years (e.g. Prater et al. 1977, Gratto and Morrison 1981, Nicoll and Kemp 1983). Individuals without the partial molt usually have not molted any primaries, but some undergo a complete molt.

Fat

Subcutaneous fat is a yellow or orange substance in appearance that is stored just under the skin. It is generally stored in three discrete areas with deposition occurring in the following order: (1) the hollow in the furculum (wishbone) just below the throat at the top of the breast muscles; (2) the hollow directly under the wing, essentially in the "wingpit"; and (3) the lower abdomen just anterior to the vent area. Holding the bird on its back, gently blow the feathers away from the upper breast to expose the furculum. Then check under the wing and on the abdomen by blowing the feathers out of the way. Fat scores are subjective and it requires looking at many birds to be consistent. Score fat as follows:

0) No fat in the furculum or anywhere on the body

1) A very small amount of fat in the furcular hollow (less than 5% filled) but not enough to cover the bottom of the furculum. No or just a trace of fat under the wing, on the abdomen, or anywhere else on the body.

2) The bottom of the furculum is completely covered but the furcular hollow is less than 1/3 filled. A small amount of fat may be present under the wing, on the abdomen, or both.

3) The furcular hollow is about half full (from 1/3 – 2/3 full). A covering pad of fat is definitely present under the wingpit and, usually, on the abdomen.

4) The furcular hollow is full (2/3 to level with the clavicles). A thick layer of fat also occurs under the wing and on the abdomen.

5) The furcular hollow is more than full; fat is bulging slightly above the furculum. The fat under the wing as well as that on the abdomen is also well mounded.

6) Fat is bulging greatly above the furculum. Large mounds of fat occur under the wings and on the abdomen.

7) The fat pads of the furculum, "wingpit," and abdomen are bulging to such an extent that they join. Nearly the entire ventral surface of the body is thus covered with fat, and fat even extends onto the neck and head. Such birds are nicknamed "butterballs."

Figure 5. Magnolia Warbler with a great deal of fat in the furcular hollow and abdomen. This individual is a 5 on the fat score scale.

Genetic and Disease Assessment

The Network offers an unprecedented opportunity to collect samples from a large number of shorebird species over a large geographic area. These samples may fulfill existing Network Project demands and also be a valuable reservoir for future studies. However, to make geographic and taxonomic comparisons, it is essential that staff from all Network sites collect samples first and foremost, and that these samples are collected in a consistent fashion. Table 6 at the end of this section provides a crude index of the number of samples we hope to obtain in 2011. While we hope to collect sufficient samples this summer, both genetic and disease samples may be collected over the next two years.

Objectives

In this study, we have four major objectives:

1) Collect and archive a large number of DNA, RNA, and fecal samples from a variety of shorebird species sampled over a large geographic area in the Arctic. We will capitalize on an unprecedented opportunity for geographically and taxonomically intensive sampling. ***This objective will be met if we do the items below.***

2) Assess shorebird blood samples for avian malaria, with a focus on several species that occur at multiple sites within the Network. Secondary objectives include:

a) *Determine if the prevalence of avian malaria varies depending on the technique used to assess a bird's exposure (i.e., PCR versus ELISA).* ***Must get large blood sample (200 ul), which can be separated into red blood cells and plasma, allowing researchers to determine if a bird has an active case of avian malaria (via the red blood cells and PCR test) or has antibodies that indicate past exposure (via the plasma and the ELIZA test).***

b) Determine if the prevalence and species of avian malaria in Arctic-breeding shorebirds vary with migration route and general wintering region of the world. ***Must get large blood sample from a variety of species – see Table 6.***

3) As a pilot project, assess shorebird fecal samples for the presence and quantity of fecal bacteria and opportunistic pathogens, with a focus on several species that occur at multiple sites within the Network. Secondary objectives include:

a) Describe the composition of bacterial communities using a combination of 16S rRNA bar codes and high throughput sequencing.

b) Determine if the presence and quantity of fecal bacteria and opportunistic pathogens in Arctic-breeding shorebirds vary with migration route, distance, and general wintering region of the world.

c) Determine if immune gene variability (in DNA) or expression (in RNA) varies with migration route, distance, general wintering region or disease exposure (malaria and gut bacterial community).

Must get whole blood, RNA and fecal samples from a variety of species – see Table 6.

4) Analyze light-level data from individual Dunlin and correlate their migration pathway and wintering areas with pathogen prevalence. Secondary objectives include:

a) Determine if the prevalence and species of avian malaria in Dunlin vary by where individuals migrate and winter within each subspecies.

b) Determine if the composition and quantity of fecal bacteria and opportunistic pathogens in Dunlin vary by where individuals migrate and winter within each subspecies?

c) Determine if immune gene variability (in DNA) or expression (in RNA) in Dunlin varies with migration route, distance, general wintering region or disease exposure.

Must get whole blood, RNA, plasma, and fecal sample from as many Dunlin as possible.

METHODS

Sample Collection

Fecal Samples

Samples will be taken as fast as possible after capture to minimize the effect of stress and to get a sample before the bird has voided the entire gut. Birds will be placed in containers until they poop. Birds usually poop within 5 to 10 min of capture. We envision a container with a top composed of a mesh material (or panty hose); the mesh material will be held on to the container with an elastic band and the mesh will be slit in the middle so a person could put their hand through the slit to grab the bird. We think it would be hard to have a normal lid because frequently birds escape when lids are open. Thus you need to be able to secure the bird before opening the lid. To get a sample that has not been contaminated from other birds, the bottom of the container should be lined with clean waxed paper (e.g., baking paper). We also recommend putting a chemical heat pack at the bottom of the container during cold days to keep birds warm. This will help you bleed the bird later. We also recommend using heat packs while banding birds. I wear a pair of fingerless gloves and put a heat pack in the palm of my hand that holds the bird. This keeps you and them warm while banding.

Once the bird has defecated and been processed (i.e., banded, bled, etc.), the entire fecal pellet is transferred into a sterile 2 ml Eppendorf tube (screw-top) containing 100% ethanol using a sterilized infant tongue depressor. Do not use sterilized tongue depressors more than once.

Pellets should be transferred to the 100% ethanol container within 30 minutes so if it is taking a long time to process a bird then you should likely do this first. Note that we tried using swabs to extract fecal material from the cloaca, but these did not yield enough DNA, hence a whole fecal pellet is needed. **If possible fecal samples should be kept cool or in a fridge – do not keep in freezer or cryoshipper.**

Materials provided by each field team:

1. Container and mesh lid (or something comparable) – to hold bird while waiting for it to defecate
2. Baking wax paper (1 per bird with large enough pieces to cover bottom of container) – to catch fecal material so that cross-contamination between birds is minimized
3. Chemical Heat packs – to put in container to keep bird warm on cold days (put under a bird bag or some other cloth within the container to avoid direct contact with the bird)

Materials provided by ASDN

1. 100% ethanol –0.3 liter (1 ml per bird) – to preserve fecal material. AK camps will be supplied this item. Canadian camps will need to purchase own supply (although I can help pay for this) since the material is hazardous and shipping across the US/Canada border would be difficult.
2. 2 ml Eppendorf tube with screw top (1 per bird) – hold feces and 100% ethanol
3. Sterilized tongue depressor (1 per bird) – to transfer fecal material to Eppendorf tube
4. Plastic pipette (5 per camp – can be used repeatedly) – for dispensing ethanol
5. alcohol-proof pens (4 per camp) – to label tubes containing samples
6. labels for Eppendorf tubes (1 per bird) – to place on tubes for writing sample information

Blood Samples – General Method

After the fecal sample has been collected, the bird should be removed from the container by reaching through the slitted mesh material, grabbing the bird, and then removing the elastic-held cover. The bird can then be transferred to your free hand. The bird should now be banded before being bled. Blood samples are collected from the basilic vein (also known as the brachial vein) under the wing of adult shorebirds (Figure 6). To collect blood, use a small amount of Vaseline or water on a swab to move feathers away from the vein. Make sure there are no loose feathers nearby else the blood may be soaked up by the feather and be difficult to collect with the capillary tube. Once the vein is obvious, puncture it with a sterile small (27.5) gauge needle by holding the needle at an angle, bevel side up, and slowly insert into the vein (much like a person takes blood from a human arm). After removing the needle, a small drop of blood should materialize. Draw the blood into a capillary tube (the type of tube depends on the procedure – see below). Hold the capillary tube in such a way that blood flows downward into the tube. In this way, gravity will help you draw the blood into the tube. You may need to pump the wing to get extra blood to flow out of the vessel. Normally the blood will quickly cease flowing, but if it does not, direct pressure on the wound will soon stop it, especially with feathers or cotton balls to aid in clotting. Injuries such as haematomas can occur if the vein area is repeatedly poked to increase blood flow, but normally the punctured area is not visible within a couple of days. It is possible to take blood from both wings if need be, but it typically does not help to continue poking one wing to get more blood. Note the size of the capillary tube – the ones we will provide typically hold 50 microliters of blood. Microliters is denoted by µl.

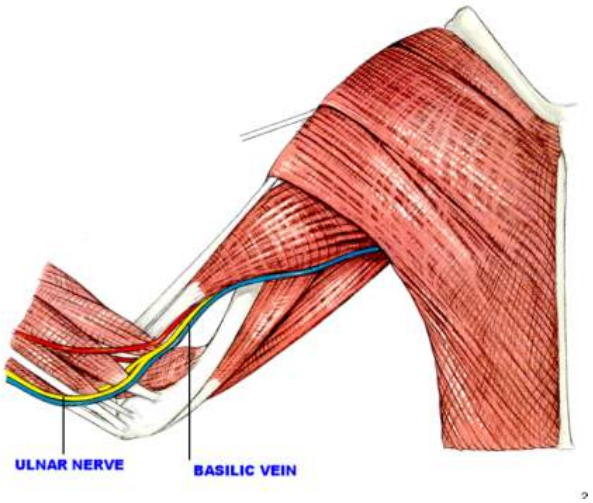


Figure 6. Drawing a blood sample using a capillary tube from a Dunlin. Illustration of basilic vein for blood withdrawal and the ulnar nerve. (Photo: B. Lewis/USFWS Figure: Evers 2008)

Given the recent avian influenza issues, use a plastic transfer pipette to force air through the capillary tubes and force the blood into an Eppendorf tube (see more details below about buffers, etc.). If the blood clots inside the capillary tube (or you have only a very small amount of blood), break off the tube inside the vial and leave it there.

Genetic Sample – sample collection

For most species (except WESA), we can obtain the genetic sample from the Red Blood Cells remaining after centrifuging blood for the avian malaria section described below. *For WESA use the protocol immediately below (except in cases where <200 ul of blood is collected, then follow this protocol for all species).*

For basic genetic analysis (population structure, paternity), we would like to get 50 µl for our analysis. This typically equates to a single capillary tube of blood. Samples for DNA analysis can then be blown into labeled 1.5 mL plastic Eppendorf tube (screw top preferred) with buffer solution and stored at room temperature. For genetic analysis, it is prudent to save whatever amount of blood you collect, no matter how small. However, **do not take more than the recommended amount of blood** as this can over whelm the buffer and keep the blood from being preserved. Remember you can now get DNA from the saliva of licking a postage stamp. Be sure to mix the buffer and blood by inverting the tube several times.

The type of capillary tube and buffer used frequently depends on the preference of the genetic laboratory.
WESA: Plain capillary tube and Queen's Lysis buffer
all other species: Plain capillary tube and Longmire Buffer

Disease Sample

1. Avian Malaria – sample collection ***avian malaria PLASMA samples can only be done where access to freezers are possible, however avian malaria/genetic DNA samples can be done anywhere***

To do this portion of the disease study, we need two types of samples. Both can be generated from a single collection of 200 µl of blood that will be centrifuged, providing 1) plasma sample and 2) DNA sample (i.e., the remaining Red Blood Cells). To gather 200 µl of blood will require **THREE full** capillary tubes of blood. Use **PLAIN** cap tubes for this purpose. The filled capillary tubes will be blown into an **empty** 1.5 ml Eppendorf tube and placed in a small flexible cooler bag with cold packs. After returning to camp (preferably within 4 to 6 hours), the tube will be centrifuged for 1 to 2 minutes at 1500 rpm or until the red blood cells form a pellet at the bottom of the tube. The plasma will then be transferred into a new 1.5 ml tube using a pipetteman (USE TIPS ONLY ONCE and THROW AWAY) and **frozen. Do not add anything to the plasma. It is okay to store in a regular freezer but is better if you can store at minus -20C (do as soon as possible)**. *The remaining red blood cells can then have 500* µl *Longmire buffer added to them for use in avian malaria PCR analysis and also genetic analysis (paternity and population genetic structure).* Be sure to flick the tubes so that the RBC pellet goes into solution. Red blood cells and whole blood preserved in Longmire can also be frozen.

For small species it will be difficult to get 200 µl of blood although this works if the vessel is hit well and you take blood from both wings. If you get less than 200 ul of blood (three full tubes), then save one tube of blood directly in a vial with Longmire Buffer or Queen's lysis buffer (dependent on species) and forego the centrifuge process. We can extract avian malaria DNA from whole blood sample, as well as get the bird's DNA from the whole blood sample. **A second tube of blood (if available) can be first transferred to an empty Eppendorf tube and then picked up with a heparin capillary tube and transferred to an Eppendorf tube containing RNA*later* (see below). IT IS ESSENTIAL THAT THE RNALATER SAMPLE BE COLLECTED WITH A HEPARIN CAPILLARY TUBE. Throw away any remaining blood – do not overwhelm either of these buffers with too much blood.**

Immune gene variability (in DNA) or expression (in RNA) – ***RNA samples can only be done where access to freezers are possible***

For this sample collect one 50 µl **HEPARIN** capillary tube of blood and add it to a 1.5 ml Eppendorf tube with ca. **1.3 ml** of RNA*later* (no more than one tube of anti-coagulated blood, otherwise the buffer will be overwhelmed). Mix thoroughly by inverting the tube several times. Blood collected with a PLAIN capillary tube will not work – the blood must be collected with a HEPARIN capillary tube. Once a sample is mixed with RNA*later*, it can be stored **up to 3 days at ambient temperature and up to 3 weeks at 4◦C (outside door of a refrigerator), but should be stored at –20°C or less for the long-term**. Storing RNA*later* treated samples for longer periods at warmer temperatures will result in a gradual decrease in RNA yield and quality. See guidelines below for more details on how to store RNAlater samples.

Materials provided by each field team:

1. Vaseline or water – to clear feathers from basilic vein
2. Power supply to run centrifuge
3. refrigerator/freezer – to refreeze ice packs, store RNAlater samples in short term

4. Small flexible cooler bags
5. Refreezable ice packs (or some way to have items kept cold)
6. Cotton balls – to stop bleeding if necessary
7. Small Nalgene with screw cap (200 ml) – for holding used needles and blood waste

Materials provided by ASDN (or Simon Fraser University for sampling WESA)

1. Longmire Buffer – 0.3 liter (500 µl per sample for RBC, and 1 ml/sample if storing whole blood) – to preserve WHOLE blood or RED BLOOD CELLS for genetic analysis and avian malaria PCR analysis
2. Queen's Lysis Buffer – (1 Eppenderf tube pre-filled tube per bird) – provided to camps sampling WESA – from Simon Fraser University
2. RNAlater – 0.2 liter (1.3 ml per bird) – to preserve WHOLE blood for Immune gene variability or expression
3. 1.5 ml Eppendorf tube with screw tops (up to 3 per bird) – to store plasma and Red blood cells for avian malaria test, and whole blood sample for immune gene sample
4. Plastic pipettes
   a. transfer samples from capillary tube to Eppendorf tube (5 per camp) – will need to cut off one end of plastic pipette at the right location so it fits tightly over capillary tube, then cut off the large end so you can blow into this section. Thus you will be able to blow air into capillary tube via this small plastic pippete to transfer blood to Eppendorf tube
   b. transfer bulk samples (10 per camp) – for putting Longmire, RNAlater or other material into Eppendorf tubes
5. 27.5 gauge needle (1 per bird + 20 extra) – to poke vessel to draw blood
6. capillary tubes
   a. plain tubes (70 µl size, 3 per bird) – to draw whole blood for avian malaria sample
   b. heparin tubes (70 µl size, 1 per bird) – to draw whole blood for immune gene sample
7. alcohol-proof pens (4 per camp) – to label tubes containing samples
8. labels for Eppendorf tubes (3 per bird) – to place on tubes for writing sample information
9. Centrifuge – (if needed – 1 per camp) -- capable of spinning 1.5 ml tubes
10. Dry Nitrogen cryoshipper (1 per camp) – for storage of RNALater and plasma samples. I can supply filled cryoshippers to sites in Alaska, and send an empty cryoshipper to sites in Canada but the people there will need to fill them up with nitrogen. We typically fill them up with liquid nitrogen and let them set for 3 days, before pouring off extra liquid and shipping them as a dry nitrogen tank. Our shippers stay cold about 65 days.
11. Storage Boxes – (81 samples/box) – enough for storing Eppendorf tubes that do not need to be placed in cryoshipper
12. Pippetman (200 µll) – 1 per camp – for removing plasma from centrifuged whole blood sample, and moving to new Eppendorf tube
13. Disposable Pippetman tips (200 µl size) – 1 per bird – **use different tip for each bird** – used to remove plasma from centrifuged whole blood sample, and moving to new Eppendorf tube

Tissue Samples – when birds die accidentally or are collected

Tissues should be preserved in RNAlater (using a ratio of 1 part sample to 5 to 10 parts RNAlater) for the few cases where Dunlin are specifically targeted for collection (as part of the

geolocator study or a stable isotope study) or in case of accidental causalities of any species. Here, we will focus on spleen tissue because it is especially important to look at immune genes. The spleen should be taken out of the bird before the liver and stomach, because is really small compared with these organs and might be damaged when the other organs are removed (see pictures of the spleen below for identification). With dead birds, it is also possible to get blood from the heart chambers – here we recommend obtaining up to 500 µl of blood and storing it in 1.3 ml RNAlater. The blood can be drawn up with Heparin capillary tubes as described above. Be sure to sterilize any cutting instruments by dipping them in 100% ethanol and burning between each dissection. Once a sample is mixed with RNA*later*, it can be stored for **up to 3 days at ambient temperature and up to 3 weeks at 4◦C (outside door of a refrigerator), but should be stored at –20°C or less for the long-term**.

Additional Guidelines for using RNA*later* **when preserving tissues**

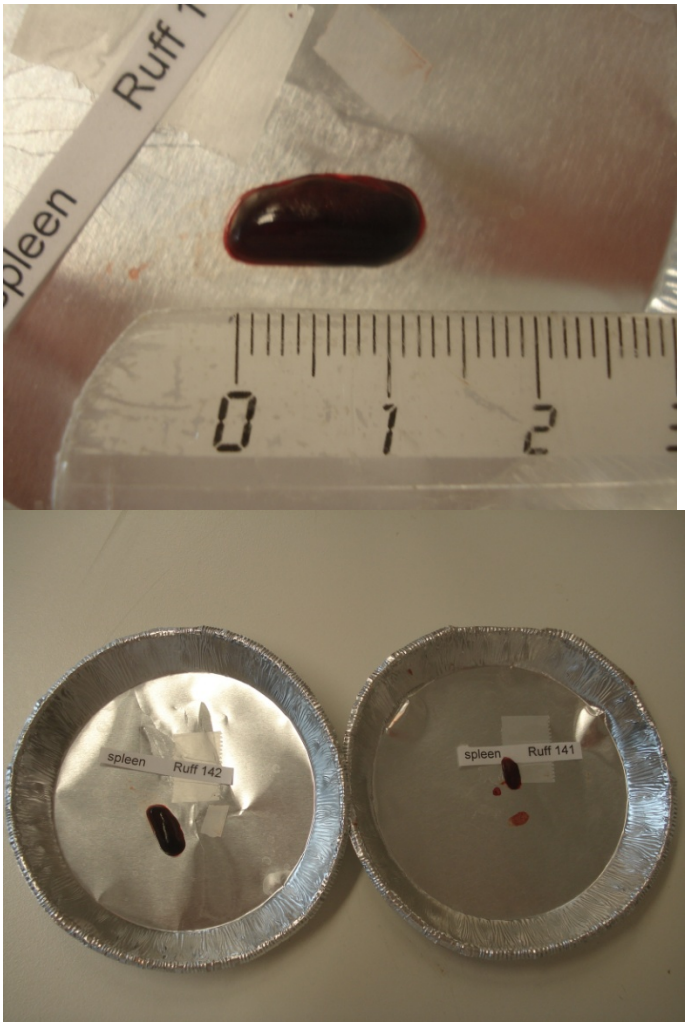
• Use RNA*later* Solution with fresh tissue only; do not freeze tissues before immersion in RNA*later* Solution.
• Before immersion in RNA*later* Solution, cut large tissue samples to ≤0.5 cm (5 mm) in any single dimension.
• Place the fresh tissue in 5–10 volumes of RNA*later* Solution.
• Most samples in RNA*later* Solution can be stored at room temp for 1 week without compromising RNA quality, up to 1 month at 4°C (fridge door), or at –20°C or –80°C indefinitely.
•***Important:*** Do not freeze samples in RNA*later* Solution immediately; store at 4 °C overnight (outside door on a standard fridge is fine) to allow the solution to thoroughly penetrate the tissue, remove supernatant (top layer of liquid above the tissue), then move to –20°C or –80°C for long-term storage.

Materials provided by each field team:

1. Cutting instrument for doing dissection (razor blades or from dissecting kit)

Materials provided by ASDN

1. 1.5 ml Eppendorf tubes (2 per bird – estimate 10 birds/camp max) – for storing spleen, and whole blood sample
2. Heparin Capillary tubes – take from stock supply mentioned above
3. Dry Nitrogen cryoshipper (1 per camp) – for storage of RNALater



Spleen pictures from Ruff

Labeling of sampling tubes

This is perhaps the most important section of the whole document. If we cannot read labels placed on tubes the samples will be destroyed and not used. The writing on the tubes must be legible and complete. Use a special ethanol proof marking pen to label tubes – do not use a sharpie as the preservative will destroy this.

For every sample include the following information on the vial (on labels provided if possible):

1) The individual bird's metal band number (absolutely necessary), species – put this on the side AND top of the vial.
2) 4-letter species code AND it's nest number
3) Collection location (use 4-letter code for your camp)
4) Collection date.

Having these items will allow accidental mistakes in the way samples were numbered to be resolved. This labeling approach is required because all samples will be returned to the USFWS office and we need a way of identifying samples from each other within and among camps.

Sample Collection Priority and choose your own adventure sampling chart, prepared by Brooke Hill. See Figure 7

1. Fecal Sample
2. Avian Malaria
3. RNA*Later* sample
4. DNA genetic sample

Don't forget to label all tubes with:
metal bandnumber,
species
nest number
collection location
date

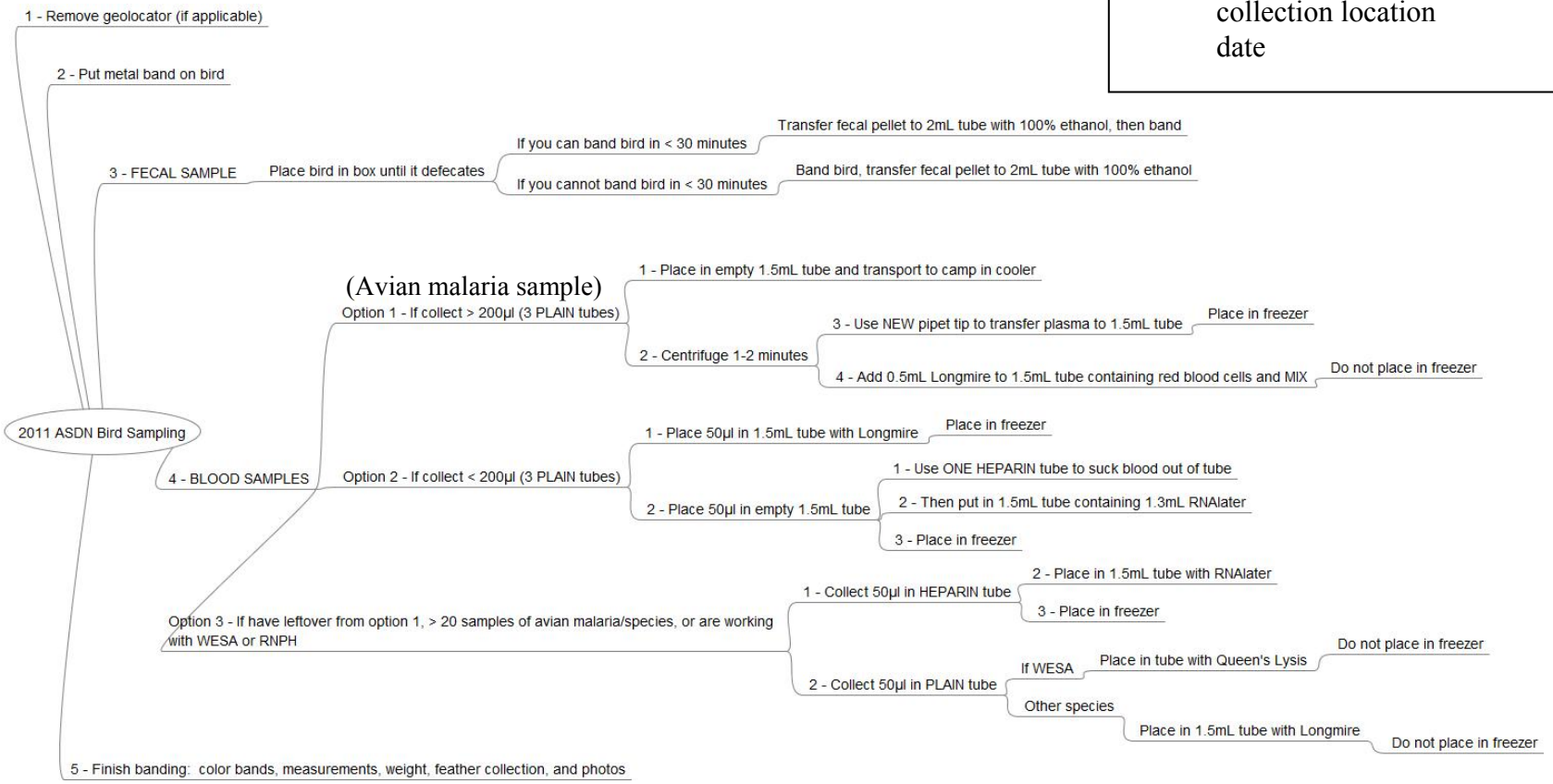


Figure 7. Suggested procedures for collecting avian health and/or genetic samples for ASDN.

Remember that if you CANNOT get 200 µl of blood (3 tubes) from a bird, then we WILL NOT centrifuge samples and collect plasma. We will however store 1 tube in Longmire buffer for DNA and avian malaria (PCR) analysis OR in Queen's lysis buffer (for WESA) and if you have sufficient blood, a second tube will be stored in RNA*Later*. Throw any remaining blood away. Too much blood in either RNA*Later* or Longmire buffer will overwhelm the buffer and not preserve it correctly.

Also, once you have 20 avian malaria samples of a given species, you can change the focus to be just RNA*Later* and DNA genetic samples (i.e., 50 µl of blood for each). We do not have funds to analyze more than 20 avian malaria samples per species anyway.

All of our capillary tubes (PLAIN and Heparin) hold 70 µl of blood. Three of them will be close to the 200 µl required for avian malaria. A little less than a full tube is needed for the DNA and RNALater samples.

Barrow Camp:

You are the only camp collecting blood from Dunlin for Stable Isotope questions. Try to get the fecal sample, as well as 20 blood samples needed for avian malaria if possible. However, Andy's project is priority. If you can get 200 µl of blood on slides to dry for Andy's study, and have remaining blood coming out of the bird, then preserve 50 µl of blood in Longmire buffer and 50 µl of blood in RNA*later*. For very good bleeders, you might be able to get 200 µl for Andy's study, and 200 µl for the Avian malaria (whole blood in empty Eppendorf tube and then centrifuge). Do not take more than 400 µl from any bird.

Feather sample collection for stable isotope analysis

For all species (other than Dunlin, Semipalmated and Western Sandpiper): We will pull the entire 10th secondary feather from each side of the body, making a total of two feathers collected from each bird. These feathers should reflect the stable isotope signatures of the wintering grounds where the birds molt and replace their flight feathers. The two feathers together should allow sufficient material for D, O, C, N, and S isotopes. By removing the inner most secondary, this should minimize the flight impact on the bird because the tertial feathers cover up this area. Feathers should be pulled in a symmetrical fashion (i.e., 10th secondary on the left AND the right – not just on one side) and efforts should be made to support the wing of the bird (by holding the bird's wing) before pulling the feather. This is especially important for sampling primary coverts (for SESA).

Place all feathers for a given bird in a brown manila envelope and fill out the information on the label. We will provide pre-labeled envelopes to each Network location to ensure data are recorded correctly. The label will include at a minimum the

collector, species, band number, date and location (both general and latitude/longitude data if away from main sampling sites – for example, indicate that it was captured at Prudhoe Bay and then provide lat, long data). Keep the manila envelopes in a zip lock bag to prevent from getting wet.

Dunlin only: Since most Dunlin molt their flight feathers during nesting or shortly thereafter on the breeding grounds, we can learn about the stable isotope signature of where the bird is currently breeding and molting (i.e. your study site) by collecting a new primary that was recently grown, and by collecting an old primary where it molted last year. This will provide information on isotopic signature of the local area, which can then be used to contrast other signatures from other breeding populations and to allow breeding local assignments from birds sampled away from the breeding grounds. Of course, it will only be possible to collect a new feather if the bird is in the process of molting. Collection of these feathers will not hurt the bird because the old feather will fall out shortly, and the new feather will be regrown if the entire feather is pulled. It is important, however, to label and store old and new primaries separately.

Only pull a new feather if it is completely grown (do not pull a feather that is growing). We recommend pulling the **1st primary on the left and right side** if present. If these feathers have already been molted, then pull the 2nd primary on each side. If these are gone, then pull the 3rd primary on each side. Continue this way until you pull the 6th primary on each side. Frequently if you are able to pull the old 6th primaries, you can also pull the 1st new primary on each side too.

Semipalmated Sandpiper only: We will pull the entire 6th primary covert (6pc), making a total of two feathers collected from each bird.

Western Sandpiper only: We will pull the entire inner most 1st primary feather from each side of the body, making a total of two feathers collected from each bird.

Table 6. Potential number of shorebird genetic and disease samples to be obtained from ASDN sites in 2011 (based on captures in prior years primarily). [1]

Species	Cold Bay	Colville River Delta	Yukon Delta	Nome - Brett	Nome - Jim	Cape Krus.	Barrow	Ikpik-puk	Can-ning River	Mac Delta	Bylot Island	East Bay	Churchill	Total Samples
AMGP					10		15				15			40
SESA		20		30		30	50	50	50	10				240
WESA[2]				30		30	10							70
PESA		20					30		30					80
DUNL														
arcticola		10					50	35	20					115
pacifica	30		30			30								90
hudsonia													30	30
REKN					20									20
REPH		10					50		10			30		100
RNPH[2]		20		15			5		15	5			5	65
RUTU		20										20		40
SEPL												15	30	45
WRSA											20	30		50
Grand Total	30	100	30	85	30	90	210	85	125	15	35	95	65	985

1 The protocol says 20 samples are needed per species per site. Given that any individual is unlikely to provide all three types of samples (plasma, red blood cells, and RNA*later*), we have listed larger numbers of individuals for a few sites where prior estimates suggest this is possible. This should allow a camp leader to collect samples of all three types more easily.

2 Note that RNPH, WESA genetic samples are needed separately for Simon Fraser University students. For these two species, we request a separate genetic sample be obtained (i.e., do not use Red blood cells that are remaining after centrifuging whole blood). These samples also need to be stored in Queen's lysis buffer that will be provided by Simon Fraser University. In a pinch, Longmire buffer may be used.

Retrieval and return of geolocators

Rick Lanctot, Shorebird Coordinator, USFWS, Alaska
Stephen Yezerinac, Biology Department, Mount Allison University, Canada

As you know, this season we need to recapture Dunlin and retrieve the geolocaters that were deployed last year. **The success of these studies will be proportional to the number of geolocators retrieved**. We need to get data from many birds in each population to accurately assess migration routes, stopover sites and winter segregation of populations. So we'd like to encourage everyone to make extra effort to get the geolocator birds this season.

1. Remove the entire band with the attached geolocator We suggest buying cuticle scissors or some other fine-tipped, sharp scissors to use to snip the rings around the bird's leg. Of course, it's critical to snip the rings without injuring the leg or damaging the geolocator.

2. Label the geolocator flag with the bird's Band Number. Use the same fine-tipped, permanent marker used for the blood vials and write the Band Number directly on the flap of the geolocator band. **It is critical that the geolocator be identified with the Band Number of the bird that carried it**; the geolocator will no longer have a readable unique ID and unless we can link the instrument to last year's records via the Band Number the data will be useless.

3. Place each geolocator in its own coin envelope or plastic baggy. It's possible that some geolocators may be loosely held to the flag, so it is critical that each geolocator be kept separate in its own coin envelope or plastic baggy.

4. Label the envelope or baggy with the following:

Species	_____	
Band Number	_____	
Date	_____	
24 hr Time of Retrieval	_____	
General Location (e.g. Barrow)	_____	
GPS location (in NAD83)	_____	(in correct time zone)

5. Store the loggers in the refrigerator or cool place (NOT freezer) to preserve battery life. The logger batteries will die sometime this Spring or Summer. While the data *may* be downloadable after the battery expires, this is a much more complicated process and has an accompanying loss of accuracy.

6. Mail the loggers back for data download ASAP. If you have field crew leaving before the end of the season, send out the first batch of loggers with them. Before mailing please check the following:

- ☐ Please check that you have filled out the data labels completely. The Band Number, Date, time and location are critical information for interpreting the data from the geolocator.

☐ Pack the geolocators so they are not loose and so that they have some cushioning to prevent damage in transit.

☐ Please email <Richard_Lanctot@fws.gov> or <syezerinac@mta.ca> to let us know to expect the shipment.

☐ If mailing internationally, be sure to attach the letter to Custom's officer (attached below) to the outside of the package.

☐ If mailing from USA, or to arrive before June 26, send to

Rick Lanctot
Alaska Shorebird Coordinator
U.S. Fish and Wildlife Service
Migratory Bird Management Division
East Tudor Road, MS 201
Anchorage, Alaska 99503
Phone: 907-786-3609

☐ If mailing from Canada and to arrive after June 26, send to

Stephen Yezerinac
Biology Department
Mount Allison University
63B York St.
Sackville, NB, E4L 1G7
Canada
Phone: 506-364-2260

If you need financial assistance shipping the geolocator, contact <Richard_Lanctot@fws.gov> for a FedEx account number to use. Thanks so much for your assistance!

NEST SURVIVAL METHODS

Objective

There are two approaches for locating and monitoring nests that are available to project leaders – please note clearly in your meta-database which approach you took. The Minimum Nest Search protocol includes searching a general area for nests, and then capturing adults at those nests and monitoring those nests for survival. The Intensive Nest Search protocol includes 1) designated intensive plots where nests are located in a standardized way but no adults are captured, and 2) a larger search area, typically surrounding these intensive plots, where nests are located and monitored, and adults are captured. The latter design will allow us to document nest density and species diversity on the plot, and also test whether banding of adults decreases nest survival. Study sites wishing to pursue the Minimum Nest Search protocol will have more flexibility in their nest searching efforts but will record the same information at nest sites and will visit nests at the same interval for monitoring nest survival.

Note: Much of the information below is specifically geared toward the intensive plot but many of the methods can be employed in the Minimum Nest Search protocol and on the larger search area portion of the Intensive Nest Search protocol.

Intensive Plot Protocol

We will rely on a combination of intensive area search and rope-drag techniques to document birds breeding on intensive plots. Area searchers and rope-drag crews will record their data on individual plot maps each day, and then at the end of the season, data will be combined across all workers to create a final nest and probable nest map (see more details below). The goal is to get the most accurate description of the species that nest on each plot (including nests found and probable nests) by documenting the presence of territorial birds (whose nests are not found) and finding nests during the field season. A second goal is to document nest survival. This will be accomplished by monitoring each nest on a regular basis until the nest hatches or fails. Attention to detail, communication with your co-workers and accurate and timely recording of data is imperative to the success of each Network site. To be able to accurately compare how a site varies across years, and how sites vary among one another, **it is essential that standardized methods be used at each site.**

Please ask questions as they arise and share what you learn.

Intensive Plot Size and Shape

Plots should be located within a given study site in habitats that will likely produce the highest densities of the focal species. If possible, place the plots randomly within these high quality habitats (i.e., stratified random placement). If these random plots fall in poor nesting habitat, then move the plot to a location that does have good numbers of birds (i.e., it will no longer be randomly placed). How plots are ultimately located will affect the extent to which your data can be extrapolated – thus be sure to indicate how this was done in the meta-database that accompanies the study site. Plot size should be at least 10 ha in size but can be much larger in low density areas. Plots can be irregularly shaped (following contours of natural landscape features) or more geometrical in shape. Square plots are preferred as they have less edge per surface area and thus are less likely to have birds establishing territories along the borders of the plot. If density allows, we recommend establishing 16-ha (400-m x 400-m) plots, and marking the boundaries and interior portions with survey stakes placed at 50-m intervals, thereby subdividing the plot into 64 50-m x 50-m grid (**Appendix A**). The 400 m^2 plots size was established to be consistent with Arctic PRISM intensive plots. The stakes will be labeled from A1 to A9 (west to east) and A1 to I1 (north to south). Researchers may alter the size or shape of the nest survival plots to conform to obvious geographic boundaries or other study objectives if necessary. The grid stakes are useful for ensuring complete coverage of the plot during nest searching, in addition to acting as landmarks for recording and relocating nest locations. All study plots must be marked with stakes that will be adequate to maintain and relocate the plot for 5 years. Sites where establishing a grid system is not helpful should at a minimum establish an outer plot boundary and midpoints (e.g. A1, A5, A9- west to east northern boundary; I1, I5, I9 - west to east southern boundary, E1 and L9 - north-south midpoints (**Appendix A**). Stakes should be labeled in a systematic fashion and can be labeled with identifying numbers and letters; the GPS location should also be collected and saved on the plot description data sheet and entered into the meta-database. We recommend that stakes be inserted in the frozen tundra by first making a hole with a pointed metal frost spike (*US Customary*: 18" x 1.5" x 3/4" or *Metric*: 45cm x 3.8 cm x 2 cm) and mallet (*US Customary*: 3 lb or *Metric*: 6.5 kg.). A wooden surveyor's stake (*US Customary:* 36" x 1.5" x 3/8" or *Metric*: 90 cm x 3.8 cm x 1 cm) can then be tapped into the hole. The stakes can be pushed into the tundra as the thaw depth increases

during the summer and pushed in as deep as possible at the end of the season to last the winter. You may want to set up a minimum number of stakes when the ground is frozen, and complete the stake installation at the end of the first breeding season when the ground is thawed. We also recommend painting the top of the stakes with a light color house paint and then painting large letters and numbers (observable from at least 50 meters) with house paint (do not use markers – the letters wear off in the wind and must be remarked each year). Stakes can also be oriented in different directions (i.e., flat side of stake facing north and then east to maximize your ability to read the stake letter/numbers from different areas of the plot). Ideally, you should be able to read a stake from any location within the 50 x 50 m cell within the plot.

Nest Searching Methods

General Techniques

Although our primary focus is to find the nests of all shorebirds, it is important to document the presence of other nesting species, especially predators and other waterbirds. We do not plan to search for or monitor Lapland Longspur nests as part of the Network although individual project leaders may choose to do so for their own objectives. Nests of all shorebirds, avian predators and waterbirds should be marked on plot maps (see datasheet-daily plot spot maps.doc) and receive a nest number and filled out plot form. We do not plan to monitor nest survival of avian predators.

Nest searching can be conducted in all types of weather, except perhaps in extreme cold (~25° F or -4 °C) and rain which might cause egg cooling when incubating birds are disturbed. Nest finding is least efficient on windy days. We don't recommend nest searching in winds >60km/h to avoid disturbing the birds. On cold days, birds appear to flush closer to the observer, which increases the detectability of nests for species that typically flush at great distances (e.g. AMGP).

During the first visit to a plot, it is useful to walk through the plot finding as many nests as possible and noting the location of these nests and other territorial birds that likely have nests on a daily plot map. Since nests found in this way are typically the easiest to find, this is referred to as 'high-grading'. By the end of the first full-day equivalent, all territories will have been plotted and nest searchers will have a good estimate of the number of birds present (although more pairs may move into an area as the snow melts). On subsequent visits, the goal is to find nests for each territory holder on the plot, including those pairs on the edges of the plot; sort out the number of territory owners, especially in cases where there are numerous unmarked pairs of the same species nesting near each other; and then search intensively for territories and nests that were not part of the first estimate. It is particularly important to find the nests of birds holding territories near the edges of plots so they can be conclusively ruled in or out of the plot. This information will be useful for estimating an accurate plot nest density that can be compared across years and study sites. In this process, nests found off plots will be useful for capturing adults to estimate adult survival. It is not a waste of time to spend hours looking for a nest that is eventually found off the plot.

It is advised that territorial birds thought to have probable nests be revisited on subsequent days, and that all portions of the plots are visited regularly (i.e., do not focus solely on the area nearest your approach location). Prior experience indicates it is better to visit each plot daily than wait 3-4 days between visits; many nests can be depredated during this interval. One way to do this is to have two intensive plots be located relatively close to each other (i.e.,

have intensive plots be "paired") and then have one observer visit one plot in the morning and the second plot in the afternoon. The next day a different observer can visit these plots but in the opposite order. Because observers vary in how good they can locate nests, all observers should rotate between all plots. Search as much of the plot on each day as possible. If a nest search on a given plot cannot be completed in one visit, indicate where you stopped on your daily nest searching map, so that 2nd nest searcher can focus on the area that was not searched.

It is common to find nests of other species within 1-2 m of a known nest, and Semipalmated Sandpipers sometimes nest within 1 m of a conspecific. Similarly, it may sometimes be necessary to flush a bird from a known nest to determine whether a nearby bird is associated with the known nest or is a member of a separate pair. It is also important to search for shorebird nests near the nests of charismatic species such as Tundra Swans, geese, and loons; the increased risk of predation to these species is an unfortunate necessity of obtaining an accurate estimate of shorebird density on a nest survival plot.

Intensive-area searches

The intensive area search method involves a single person (e.g. nest searcher) who walks throughout the plot, using the presence and behavior of birds to determine the location of territories and nests. Because each nest searcher has different capabilities, we will have nest searchers rotate between plots to ensure consistency amongst plots in the number of nests found. To ensure complete coverage of each plot, it is recommended that nest searchers cover the plot by walking between stakes in a "W" pattern. This will reduce observer bias and increase the probability of all nests being found. However, nest searchers should not focus too hard on performing their "W" walk at the expense of missing birds that are flushing from nests in front of them. Unique detailed maps of each plot should be created during plot set up, noting any landforms (e.g. lakes and creeks), notable features or human debris. This grid structure can be copied onto daily search maps and notes can be gathered daily to create a plot master map at the end of the season. The territorial birds or nests discovered should be recorded on these maps. These maps should be shared among the nest searchers so that information on potential nest sites are shared daily and subsequent visits to a plot can be planned to enhance finding nests.

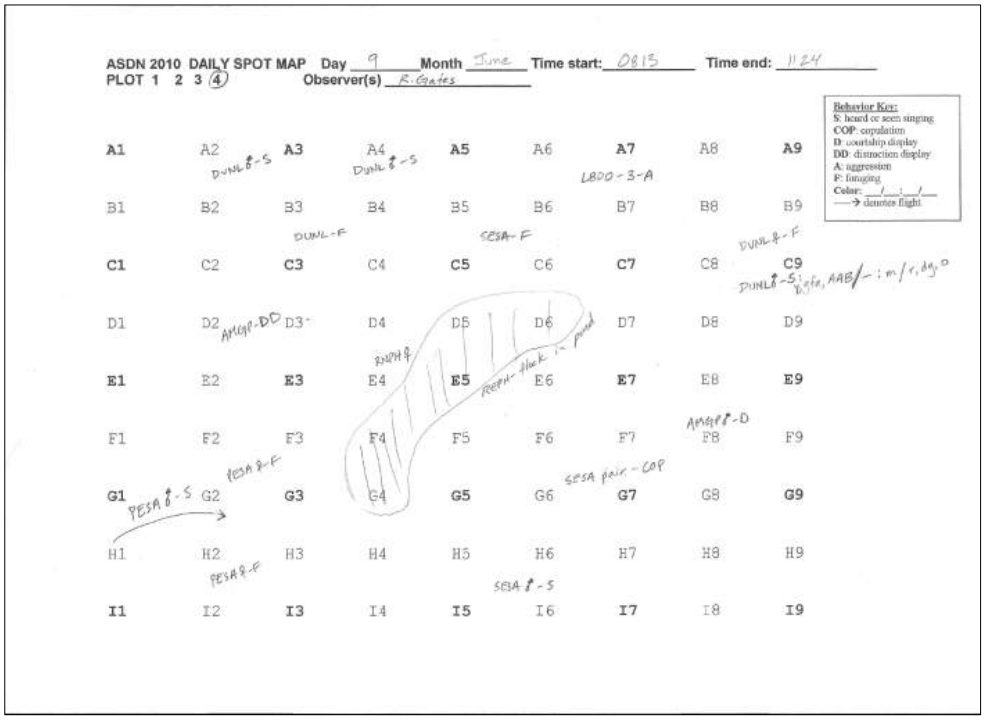
Plotting nests and territories during area searches

The following approach is one way to maximize communication among nest searchers as they visit the various intensive plots to look for nests. Each camp may use a different technique as long as their nest searchers can reliably document both nests and territories on the intensive plots at the end of the field season. Each plot will have a **nest plot book** that has a plot map that illustrates known nests. Area search and rope-drag crews will record nests in this book, but also record the location of territorial pairs on a separate map for each day they visit the plot (see Figure 8, top panel; and **Appendix A**). These maps will be shared among crew members to maximize the chance of finding nests before they are depredated. In the windy and misty arctic environment, we find it best to place maps in a standard sheet protector, anchor it to a clip board with rubber bands, and write on the sheet protector with an ultra-fine tipped Sharpie (alcohol will remove ink for re-use of sheet protector). **After each day in the field, transfer new nests into the nest plot book for their respective plot.** Also make a field copy of the permanent map and use it within a sheet protector for the next visit to the plot. Update both the field and camp copies after each visit. We use only black Sharpie on the sheet protector because other colors tend to rub or wash off easily.

The term "territory" is used in a broad sense to mean the area of primary use. For example, the area of use of a Semipalmated Sandpiper pair that is staying close to a nest may be drawn as a very small circle. The area of use of a Black-bellied Plover pair whose nest has not been found and who travels widely on and off the plot, may be indicated by a series of arrows denoting movements rather than by a well-bounded circle.

Probable nests are defined as a location from which an individual flushes using a broken-wing, rodent-run or other distraction display but the nest was not located. The location must be precise enough for the observer to be certain that the actual nest is on the plot. No other definitions of probable nests are acceptable. Probable nests should be mapped on the daily plot map (not Nest Book) and mentioned to the subsequent person visiting the plot so they can look for the nest on the proceeding day. Probable nests ARE NOT given a nest number and will not have a nest form filled out until found.

At the end of the field season, time should be reserved so that nest searchers can make a final map for each plot depicting the nest sites and territories of birds whose nests were not found (Figure 3, bottom panel). The data sheets from the field visits can be useful here as it allows nest searchers to collate everyone's data together. For example, if a pair of plovers was consistently found in one part of the plot but a nest was never located, it is likely a pair initiated a nest but that nest failed before discovery.



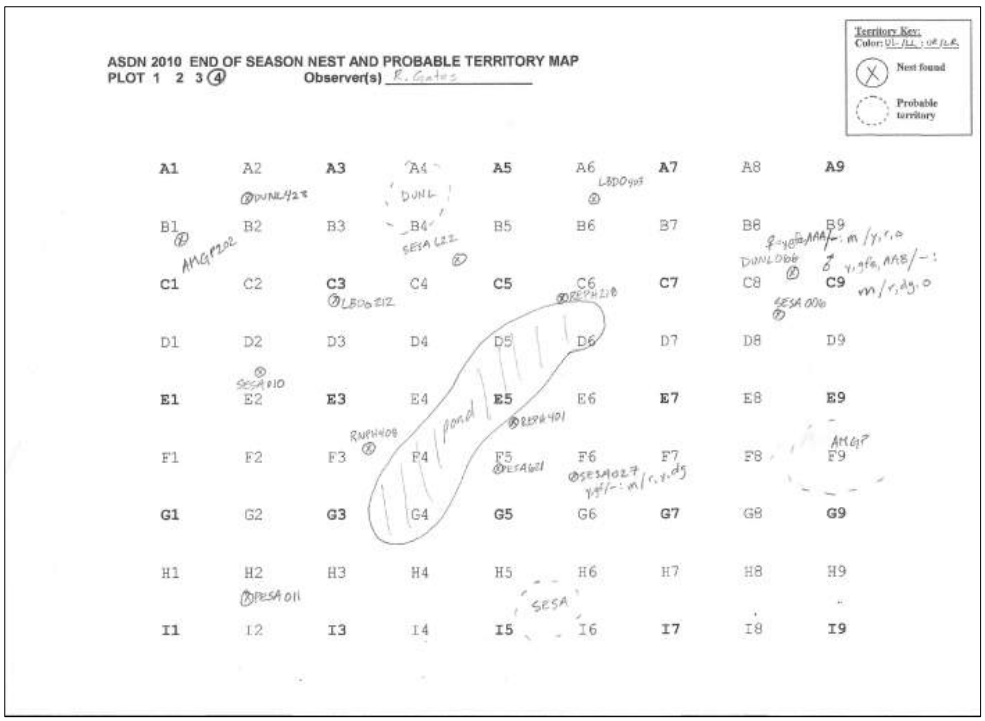


Figure 8: Example of a Daily Spot Map for taking behavioral notes while conducting intensive nest searches (top panel) and the End of the Season Nest and Probable Territory Map (bottom panel).

Rope-dragging

Rope-dragging is a commonly used tool for locating the nests of ground-nesting birds. In this study, 2 person teams will rope-drag one to two times during mid to late-incubation on the intensive plots, and if time allows on the larger search areas. Rope-dragging should begin approximately 14-16 days after the first clutches are complete. Rope-drags on each plot are scheduled 4-6 days apart, but the decision to rope drag a second time will depend on the species present in your area (i.e., do you have a lot of secretive species that only flush off a nest when nearly stepped on), the success in nest finding using the area search method, and the time available to do it.

Because dragging a simple rope across terrain with even mild topographic or vegetation height heterogeneity can be problematic (e.g., ropes gets caught on obstructions, freeing ropes from obstructions can result in some terrain not actually dragged, etc.), rope drags were designed to have hanging "plastic strips or dropper lines" placed every meter along the rope (see Figure 3). These strips will contact the substrate, while the rope itself is suspended slightly off the ground by the rope-dragging crew. The drag consists of a main line of 1/4 - 3/8" rope, with dropper lines (1/4" rope) tied to it perpendicularly at 1.5m intervals (Figure 9). Improvised handles (and/or waist belts) should be tied to the rope so that observers can maintain tension on it and keep it from snagging on irregularities in the ground. In cases where the ground is very flat, a longer rope without dropper lines may be used. The time it takes to rope drag a plot varies dramatically with terrain and bird densities, and some plots may take much longer; it is advisable to begin rope-dragging in the morning when possible. Remember the goal is to find nests, not complete the rope-dragging of a plot. If you are finding nests, you are being successful.

Although rope-dragging will occasionally cause a bird to flush from its nests directly under the rope, some birds will flush well ahead of the rope and the approaching crew. If a quick inspection of the flushing area fails to locate the nest, the crew should continue past the nest a sufficient distance to allow the bird to return (this will vary by species, weather, individual, etc.), then stop and monitor the bird's behavior until it returns to the nest. If the rope-dragging crew fails to find the nest of individuals or pairs that are obviously "nesty", and the crew is confident that the nest is on the plot, they should record it as a probable nest and estimate the probable nest location as accurately as possible. The rope drag team can revisit this spot later in the day, or at the very least, other nest searchers can go back to this spot on a subsequent day.

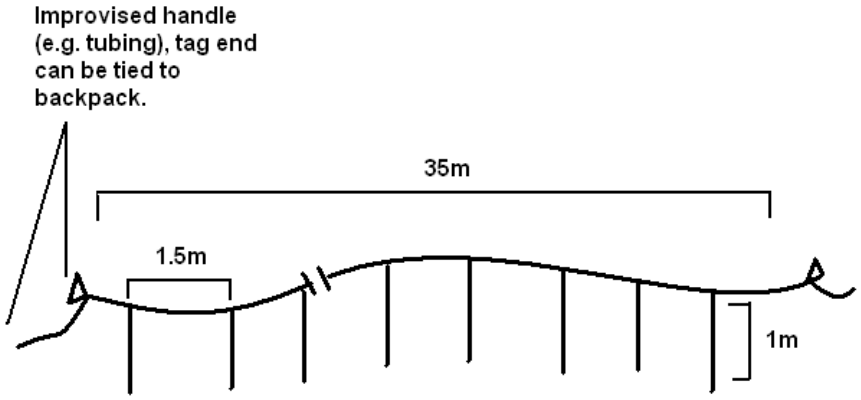


Figure 9. Construction of rope drag for intensive shorebird monitoring plots (Figure: Smith et al 2008)

Special Techniques for Finding Nests

While the area search and rope drag are general approaches to finding nests, these approaches can be enhanced by employing special techniques and using our general knowledge of the nest habitat preferences and general behavior of each species. Combinations of several nest-finding strategies are necessary to find all nests on a plot. Below are suggestions for becoming proficient at different techniques.

Systematic searches

During the beginning of the field season when snow covers most areas of the tundra, one-egg nests can sometimes be found by simply walking to all the available open spots and looking for nests (i.e., do not rely on bird behavior but simply search the tundra with your eyes). Occasionally 1 or 2 egg nests can be found this way, and these nests are generally the first laid nests of the year. Systematic searching can also be used later in the season when you have reduced the probable nest to a small area; the nest searcher can spend time thoroughly covering the area to find the nest. Nest searchers must be careful when using this technique to avoid stepping on the nest you are trying to find. Looking closely at each place you plan to step before taking a step will insure that you do not step on the nest you are trying to find.

Behavioral clues

When watching adults, nests may be found by following incubating birds back to the nest or by pinpointing the location from which a bird flushes. The following is a summary of behavioral clues:

- Pick a territory or pair whose nest has not been found (look at daily plot maps), walk the known area of use until you find a nest or detect a bird which can be followed back to a nest.
- Any suspicious bird, or bird that does not appear to be associated with a known nest, is worthy of an extended observation. In dimorphic species in which only one sex incubates, like Pectoral Sandpipers where only the female incubates, it is easier to decide which birds are worth watching (i.e., follow the incubating sex, although the non-incubating sex may display near an incubating bird). In monomorphic species (e.g. Semipalmated Sandpipers) or those with dual incubation (e.g. Dunlin), it is more difficult to decide if a bird should be watched or not. Incubating birds sometimes appear disheveled and breast-preening is a classic tell-tale sign of a recently incubating bird. Birds that are incubating have various displacement behaviors that indicated that they are stressed and nervous and would like to return to the nest. When you happen upon a bird that is preening, nervous or foraging really fast, it is likely that they are on an incubation break or you flushed them off a nest, back off the bird and allow it to return to the nest. In general, males will return to nests quicker than females, if you can determine the sex based on plumage, vocalizations and behavior this can help you determine whether a bird should be followed.
- Walk preferred nesting habitats of key species (e.g., pond edges or marshes for phalaropes; dryas benches for American Golden- or Black-bellied Plovers). Fourth, watch for birds that are frantically feeding. They may be on an incubation break and will likely return to the nest shortly – this applies especially to species where only one sex incubates.
- Nests of some species are more easily found by observing individuals at a distance before they are disturbed; this works well for Bar-tailed Godwits, American Golden- and Black-bellied Plovers. In these cases, try to take advantage of a higher elevated site where you can see the bird from a long distance. Using a spotting scope can also help (e.g., the black and white stripe pattern of an AMGP is very distinctive from a distance). Do not get too close else the bird will not return to the nest.
- Use your binoculars creatively, from a birds perspective they see a tall predator like thing staring at them with huge eyes (e.g. your binoculars). Sometimes you can get down low, making the bird feel less watched and point your binoculars away from where you are looking, and then watch the bird return to the nest with unaided eyes. Keep your eyes on the incubating bird; bring your binoculars to your eyes to see exactly where the bird is. Remember to look for landmarks in the area to help you find the nest as you approach it from afar…keep your eye on the nest. Don't allow yourself to get distracted by other birds in the area and the bird as it tries to distract you away from the nest.

Appendix B has a species-specific guide to shorebird behavior that helps nest searchers interpret the behaviors they are observing and streamline their time to find nests. Shorebirds have diverse mating systems, incubation strategies and suites of sex-specific behaviors and vocalizations. Taking the time to understand the differences in species and sexes will save time in finding nests.

Nest Monitoring

Once nests are located, institute a 5-day visitation schedule so that we can obtain accurate estimates of nest survival. **Do not skip nest checks**, a delay in scheduled nest check reduces the precision of our daily nest survival estimates. Curiosity leads observers to visit nests more often than necessary, however this likely has a negative impact on the nest's success. Nests should be

visited 4 days prior to expected hatch (even if it doesn't fall on the 5th day of visiting). If there are no signs of starring, then the nest can be visited 2 days later and checked for hatch again. Continue this process until you see the first signs of chicks trying to exit the egg. Starred eggs typically hatch 2 days later (but can hatch the next day), and pipped eggs may hatch later that same day or the next day (Figure 4). Continue to visit nests daily at this point until you have documented whether all four chicks have hatched. For nests where eggs do not hatch, collect the egg and determine whether an embryo was developing and died, or whether no embryo was present. These data on egg hatching should be recorded on the nest record form too (frequently overlooked!!!!).

We recommend a 5 day visitation rate for all nests found, whether on the intensive plots or larger general search area.

Figure 10. A Dunlin egg that has stars surrounding a "hole-pipped". Photo: D. Taylor/USFWS

Data recorded at each nest

The following data should be recorded at all nests when discovered (see Nest Record data form):

1. Nest identification number (Nest ID#):
2. Observer(s): (first name initial and last name e.g. loring, for Lewis Oring)
3. Plot and subunit if appropriate (e.g. unique subunit id, quadrat number or northwest stake)
4. Date and time of nest discovery (use military time)
5. Record GPS location of nest
6. Nest within plot boundaries (Yes or No, may need to measure if near border)
7. Estimated hatch/fledge date: calculated from age data
8. Species: (4-letter AOU code)
    a. 4 letter species code + number of nest found
9. Method of discovery (rope drag, area search, bander, other, explain observer first initial and last name)
10. Color band combos of pair, specific sex if known
11. Nest site map: a simple map of the nest site; include nearby distinguishing physical features (e.g., ponds, polygon rims, etc.).
12. Number of eggs/nestlings on discovery day

13. Flotation data: see **Appendix C** for specifics
14. Measure egg length and width to the nearest 0.01mm with dial calipers
15. Office: use estimation of important nest dates such as initiation date, start of incubation, and estimated hatch date

Nest monitoring data to record:

16. Date (dd-month)
17. Time (military time)
18. Observer initials
19. Nest stage (laying [L], incubation [I], hatch [H], brood [B], predation [P], fail [F], abandon [A])
20. Nest contents seen (Y/N)
21. Number of eggs[E] and/or number of chicks [C] if contents seen, otherwise N/A
22. Pip [P]/ star[star symbols with #of stars]/ crack [C]- dash for none observed
23. Done this visit: Flag, float, measure eggs =FFM, Nest check=N "with a check symbol" Hatch check H "with a check symbol" B=Band
24. Next visit date

After nest fate has occurred, record:

25. Percent nest concealment: use ocular tube and estimate to the nearest 10%
26. Dominant vegetation (e, b, u or m) and landform type. **Appendix G** has categories appropriate for the Western Alaska and the Arctic coastal plain. For other parts of the Arctic use the most reputable guide that lists dominant vegetation and landform.
27. Nest Fate determination (see procedures below):
    a. Date nest fate was determined (day and month)
    b. Was the nest scrape: Intact/scattered/Flattened and widened
    c. Fox urine smell (Yes or No), Fox scat present at nest site (Yes or No)
    d. Were egg fragments present (Yes or No), if yes describe number and location
    e. Were egg shells present (Yes or No), if yes, describe number and location
    f. Membranes: Attached/Separate/None
    g. Weather induced: (Y/N)
    h. Caribou trampling (Y/N)
    i. Adult present eliciting brooding behavior (Y/N)
    j. Was the brood seen (Y/N), if yes # of young, age, and distance from nest
    k. Nest fate (hatch/fledge, fail, unknown, undetermined)
    l. For nests that fail, indicate how: predation, weather, trampling, human cause, other, etc.)
    m. Write detailed notes on justification of nest fate determination (**be meticulous!**)

Marking nests

Marking nests in a standard way allows all co-workers to find nests more quickly and minimizes disturbance and the potential attraction of predators. Markers (and marker placement) should minimize olfactory cues for nest predators. We recommend marking nests with two Popsicle sticks and one pin flag. All nests should have one popsicle stick placed 1 meter north of the nest, a second popsicle 5 meters north of the nest and a green or blue (no red, orange or yellow) colored flag should be placed 10 meters to the north of the nest (see Fig.4). North can be

determined by using the grid stakes (e.g., walking from I1 to A1 would be in a north direction) assuming the plot was laid out so A1 was in the northwest corner, or by using a compass. The flag and Popsicle sticks must create a direct line to the nest (i.e., so you can walk south from the flag and find the nest). A medium-point Sharpie can be used to write the year the nest was found, species, and nest number on the Popsicle closest to the nest and on the flag itself (e.g. 10SESA401). The popsicle stick with the nest ID will be placed in the ground at the edge of the nest once the nest is completed for a year. Leaving these markers allows us to document reuse of nest cups. Nest locations MUST be plotted on a map in the nest booklet at the time they are found (see item 11 on nest record form). Use landmarks, number of paces and cardinal directions to guide the observer from one landmark to another, then to the nest marker and then to the nest. Keep the distance between landmarks short, especially if landmarks are small, and use a compass (don't guess) to get directions from nearby stakes to the nest site. A crude drawing indicating the landmarks (e.g. ponds, creeks, hummocks), paces and direction between landmarks, the nest marker, and the nest is very useful.

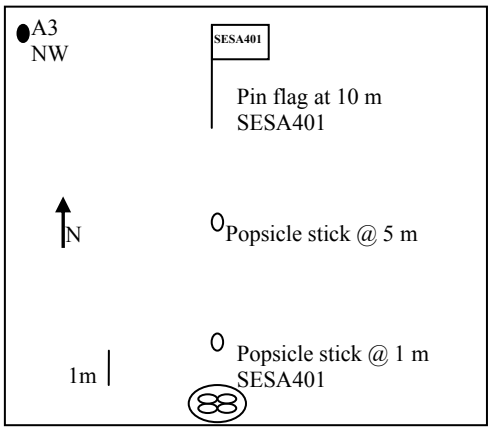


Figure 11: Schematic of marking nest to relocate for nest monitoring

Numbering nests

Each observer numbers their nests consecutively regardless of species or plot. The most common error is to use a number more than once. Use a numbering system for identifying each nest that avoids such errors. It is advisable for each observer to keep a list in their field notebook of nest number, species, plot, and day on which it was found. On maps and in notes, refer to nests by species and number (e.g., SESA401). Field supervisors should assign each observer a series of numbers for use throughout the field season.

Recording location of nests on maps

Record the location of each nest in using a GPS unit and record the location on the nest record data form. Be sure the GPS location is recorded on the nest form prior to leaving the general location of the nest. Do not plan on copying GPS locations from your GPS unit to the nest form at night – this is frequently forgotten and the data are lost and the nest needs to be visited again to retrieve these data again.

Nest initiation date determination

Nest initiation date should be determined for every nest. We define nest age as the period from the date the first egg is laid (nest initiation) until the estimated day of nest fate occurrence. Nest initiation day is defined as the day when the first egg was laid in the nest. We define the

start of nest incubation as the day when the fourth egg is laid in the nest. This date is also commonly known as the clutch completion date. This day is defined as "day 0" when using nest incubation periods to determine estimated hatch date (i.e., the day when the 4th egg was laid is day 0, the next day is the 1st day of incubation, etc.). A reliable estimate of nest age is important for two main reasons: 1. It assists in accurately assessing nest fate by allowing a calculation of the estimated hatch date and, 2. It provides a way to correct estimates of density for non-detection associated with early nest loss.

Use the following procedures to determine nest initiation and hatch dates:

1. For nests found in the laying stage (incomplete clutch) count 1 day backward for each egg laid to estimate initiation date. You can forward calculate (using the estimated incubation/nestling stages for the respective species – see **Appendix D**) to estimate the hatch date. Shorebird nests found with less than four eggs, the average shorebird clutch size, should be revisited the next day, and every subsequent day until clutch size stays the same. Daily visits to an incomplete clutch will determine if the female is still laying eggs versus partial nest predation. This is particularly critical late in the season when 3-egg nests may be more common. You cannot assume a 3-egg nest will have a fourth egg – this has to be verified. 3 egg nest found during late incubation should be floated upon discovery to determine nest initiation date.

2. Age of eggs for shorebirds nests found during incubation (i.e., with 4 eggs) should be estimated using the egg flotation technique. Egg floatation boxes will be distributed across the Network to standardize out efforts for nest initiation estimates. See **Appendix C** for complete instruction on egg flotation procedures. A simple float program and corresponding table (**Appendix J**) has been developed to aid in standardizing initiation dates based on egg floatation. Instructions can be found in the Excel program that is included with the Network electronic files.

3. If nests are found with star cracks or pip holes in the eggs, or hatchlings (i.e., via nest monitoring near the expected hatch date), the initiation date of the nest can be determined by subtracting the number of days of a typical incubation period for that species from the calculated hatch date (see **Appendix D**; unless this nest was found during laying in which case the laying information should be used). For our purposes, hatch date will be the day the first chick is found in the nest (even though chicks may continue hatching for an additional 1 or 2 days sometimes). If for some reason you do not get to visit a nest to see chicks, use the following rules: 1) star-cracked eggs, assume hatch day is in two days (for shorebirds, one day for waterfowl), 2) pipped eggs, assume they will hatch the next day, 3) wet chicks, assume they hatched 4 hours prior to your visit – if your visit is at 4:00 AM, then the chicks likely hatched the prior day. If at all possible, visit nests to confirm hatching of chicks.

Nest fate determination

Put as much effort as possible in determining the fate of each nest. If there are doubts, the nest fate should be recorded as "unknown". Nests with unknown fate can still be used in the

survival analysis (until the check prior to that where fate became unknown), but they contribute significantly less information than nests with known fate. Generally, **2 pieces of evidence** are needed to classify a definitive nest fate. If nest fate is determined, record supporting evidence on the data form and how confident you are in this assessment. Determination of nest fate will be most accurate when nests of known age are visited at more frequent intervals as expected hatching approaches. See **Appendix F** for fate determination definitions. See **Appendix E** for more information on finding eggshell remains and using them as evidence of nest fate for shorebirds.

Nesting habitat classification protocol

Basic habitat information should be collected at each nest that describes the macro and micro site conditions, or landform and vegetation type, respectively. **Nest concealment measures should be taken during mid-incubation for each nest that is still active**. **At the final visit to a nest (generally after hatches or when found depredated),** record the landform and vegetation types according to the habitat classification for your location (**Appendix G**).

1. Landform type: Record the dominant landform type within a 10 m diameter of where the nest occurs. Landform types for the North Slope of Alaska can be found in the Geobotanical Atlas of the Prudhoe Bay Region, Alaska (Walker et al. 1980). See **Appendix G** for landform descriptions. These landforms are large-scale, geophysical features that may contain a variety of vegetation types. For other areas, use the most reputable guide available.
2. Vegetation type: Record the dominant vegetation type within a 10 m diameter of where the nest occurs. See **Appendix G** for vegetation descriptions. For other areas, use the most reputable guide available.
3. Nest concealment: Estimate the percent of the nest (nearest 10%) obscured by vegetation when viewed from 1m directly above the nest. **To reduce bias, use an "ocular tube" (piece of PVC approx. 1.5" inside diameter x 4.5" tube length (James and Shugart 1970) a cardboard toilet roll can also be used.** When estimating concealment, view the nest from 1m above, looking through the tube with one eye while keeping the tube centered on the nest. For species that do not have vegetation that falls over the rim of the nest (e.g. waterfowl, plover, loon, gull, and jaeger nests), the nest concealment value would be 0%. This measurement is taken during mid-incubation.

Recommendations to reduce anthropogenic effects on predation rate

1. **Avoid leaving scent at the nest.** Common mistakes include the following: touching vegetation around the nest with hands (use a stick or nest marker if necessary), standing at the nest while making the nest card or marker (move away a few meters where you can still see the nest), or placing a backpack or notebook on the ground near the nest.

2. Conduct nest checks from a distance using binoculars if possible. Assume that the presence of an incubating adult indicates an active nest. However, if it is close to the hatch date, flush the bird and check the nest contents.

3. Avoid creating dead-end paths when checking nests. Approach the nest along one route and leave on another. This will make it more difficult for predators to locate nests by watching your activity or following your scent.

4. **Do not approach an active nest if predators are nearby or watching you.** Stop nest searching when predators are in the area. Do something else and return later.

5. Do not sit down or set down your pack or other belongings near a nest.

6. **Only touch nest contents if floating and measuring eggs and use surgical gloves if possible.**

7. Do not eat on study plots. Eat at least 50m outside of the plot boundary.

8. Cover unattended waterfowl nests with down, feathers, and vegetation to conceal them from avian predators.

9. Do not urinate on the plots. When urinating off plot, do so in water to diffuse scent.

10. Collect only necessary data at each nest site and leave as soon as possible.

ECOLOGICAL MONITORING

Objective

In establishing a long- term monitoring Network it is important to employ standard techniques for ecological monitoring components. Daily species lists, food resources collection, predator and alternative prey indices, site and weather conditions descriptions complement interpretation of avian monitoring studies.

Daily camp journal

See dataforms for daily camp journal and daily species list forms.
Adapted from Barrow Protocols and ArcticWOLVES (Smith et al. 2008).

Each camp will maintain a daily camp journal that details the activities at the field camp. This information can be hand written on the daily camp journal data form. Each day's entry should include of summary of the following daily information:

- Personnel on and off duties, including where they worked and what they were doing.
- Arrival/departure of personnel
- Denote ecological monitoring surveys, bug collection, snow surveys, etc.
- Summary of the day's weather
- Significant natural events
- Rare sightings
- Visitors (coming and going)

Vertebrate species list

Each camp will maintain a daily incidental observation of vertebrate species list including observer effort to understand the relative abundance of terrestrial vertebrates. This will be useful in comparing between study sites or between years. This protocol outlines a technique for providing a relative abundance estimate based on incidental observations.

The technique can be applied to virtually all vertebrate species, whether or not they are subjects of more accurate and labor-intensive population estimation protocols. There is value in obtaining a record of incidental observations for all of our focal prey and predator species, because these give an index of the relative levels of activity within the study area. However, the technique is most useful for giving us insights into the abundance and timing of less frequently encountered species, such as certain birds (waterfowl, ptarmigan), and mammals (ungulates, larger wide-ranging carnivores).

The relative abundance of species is recorded as the number of individuals seen per hour spent in the field per observer. We will group these data for all observers and over selected time periods to give a cumulative encounter rate index. Please take note of each observer's effort who contributes to the daily species list including number of hours observing and mode of transportation on the Daily Camp Journal. The likelihood of making observations varies with the observers' activities and mode of transport in the field. The observer should record whether they were on foot, using a motorized ground vehicle (boat, ATV, Snowmobile), or using aerial transport. If the observer(s) were doing very focused ground work (e.g., vegetation sampling, or

building exclosures) with little likelihood of making observations in the broader landscape, then that time should be subtracted from the total time elapsed for observations.

Encounter rates should be calculated for time spent in the field, away from base camp. Observations made right from the camp should be recorded separately, and merely as a record of species observed, rather than an encounter rate. This is because it is difficult to estimate the amount of time spent in such observations at camp when much of the observers' attention is mostly on camp infrastructure.

Records should be kept daily in field note books, including start and end times for the period of the day spent in the field doing activities that have a reasonable chance of allowing observations, and a list of species with number of individuals, sex and age if possible, and any comments about unusual sightings. These should be written on the daily species list data form and transferred to a digital data file if possible.

Supplies required for food resources monitoring:

Provided

Item	**Habitat type**	**Use(s):**
Triton soap	Terrestrial	To break surface tension in pitfall trap solution
One 4-inch fine mesh fish net (brine shrimp mesh)	Terrestrial	For filtering insect from the solution samples
Whirl- Paks	Both	5 for aquatic traps and 10 for terrestrial
All materials for terrestrial pit fall traps (e.g. pvc pipes and glue, window screen netting, zipties)	Terrestrial	Building terrestrial traps
One 5-inch fine mesh fish net (be sure to use the correct one!)	Aquatic	For collecting aquatic sweeps

Supplied by each camp leader

Item	**Use (s)**
1 ½ gallons (~6 liters) of full strength (>95%) isopropanol (e.g. rubbing alcohol)	Sample preservation
Propylene glycol (non-toxic anti-freeze)	To preserve DNA for future barcoding and inhibit the trap solution from freezing
20 pieces of rebar (1 ft long and ½ inch diameter)	Anchoring screen above pitfall trap
20 10-inch nails	Anchoring trap to ground
Duct tape (big roll should be plenty)	Binding objects of all types together

Objective

The distribution, phenology and abundance of invertebrate adults and larvae will be measured in terrestrial and aquatic habitats. We will identify and enumerate samples to acquire estimates of biomass, biodiversity, emergence and seasonal abundance. Trapping methods were designed to be comparable with on-going and historic shorebird food resources studies (e.g. ArcticWOLVES, MacLean 1969, MacLean and Pitelka 1971).

Terrestrial monitoring

Terrestrial food resources will be collected from mesic and dry terrestrial habitats using modified Malaise pitfall traps. Terrestrial sampling will commence when the habitats are mostly snow and ice free (and likely before aquatic traps). The terrestrial pitfall trap design is similar to those used by ArcticWOLVES and includes modifications from Bylot Island in 2009 (JF Lamarre and E. Bolduc pers. comm.). It is recommended that each site permanently establish **2 trap line transects of 5 pitfall traps each.** The traps should be spaced 15 meters apart and the lines be put somewhere convenient for frequent sampling. One trap line will be established in a dry habitat-type (e.g. centers of high-centered polygons, non-patterned ground, or generally dry areas), and one transect line in more mesic (wet) habitats (e.g. pond edges and rims of low-centered polygons or strangmoor).

Building and deploying terrestrial traps

A step by step method to make Bylot Arthropod Traps

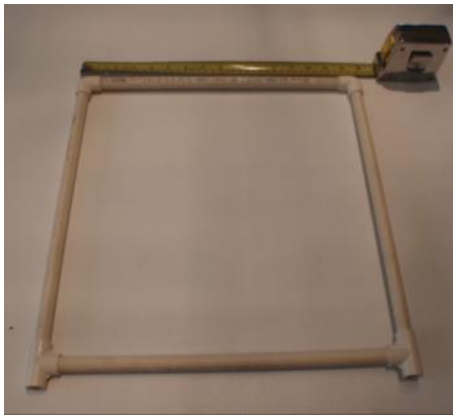
by Jean-François Lamarre and Elise Bolduc, based on previous experience with Modified Malaise traps.

Material needed per trap (40 cm X 40 cm)

- 2 90° elbow – for ½ inch CPVC tubing
- 2 T elbow – for ½ inch CPVC tubing
- 4 36 cm long ½' inch CPVC tubing
- 2 10 inch nails (to anchor down trap)
- 1 Rubbermaid drawer organiser (15' X 3' X 2'---L3-2917-RO-WHT)
  http://www.rubbermaid.com/Category/Pages/ProductDetail.aspx?Prod_ID=RP091340
  http://www.containerstore.com/shop/office/deskAccessories/drawerOrganizers?productId=10000149
- 1 Screen 45 cm X 92 cm(out of a roll of 92 cm wide)
- 2 rebar (1 ft ½ inch diameter)
- Rope (approx. 2mm wide, 5 m per trap)
- 4 small Zip ties (to anchor the screen to rebar)
- 1 PVC pipe glue (for securing the elbows)
- 1 Large sewing needle
- 1 spool of heavy gauge thread for sewing screen

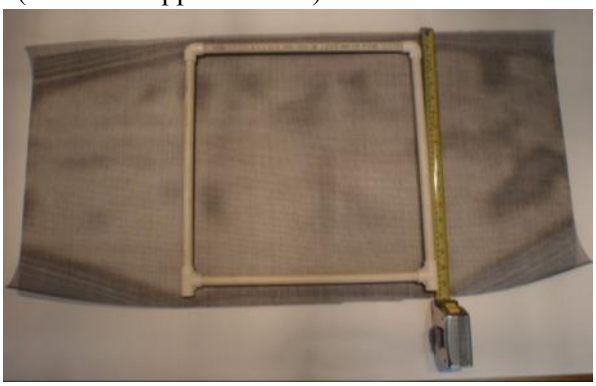
FRAME AND SCREEN

1. Cut the tubing in length of 36 cm.



2. Assemble with 90° and T elbow. Make sure elbows are straight. Use PVC glue if necessary (I didn't use any: connections were tight enough)

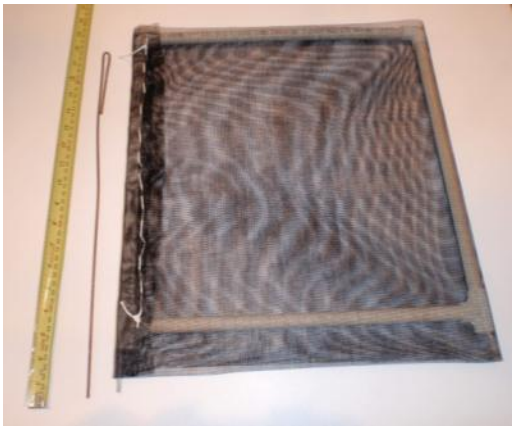
3. Cut a slice of 45 cm X approximately 92 cm (there is 92 cm rolls sold) of normal window screen (mesh size approx. 2 mm).



4. Use approx. 80 cm of small rope to sew both screen edges together to wrap the tubing frame with a big needle.

5. Secure each end with a few knots. Note that the screen is meant to extend below the bottom so that the inverts are guided into the solution in the pitfall container.

6. Attach 2 zipties on each side of the frame for later use anchoring the screen to the rebar. The rebar should be pounded in the ground just outside the pitfall. Picture shows wire but rebar is preferred.

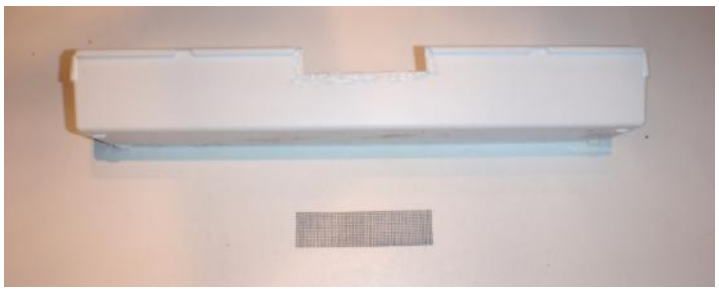


7. Tie a knot on the top tubing (3 turns around the tube and 2 flat square knots).

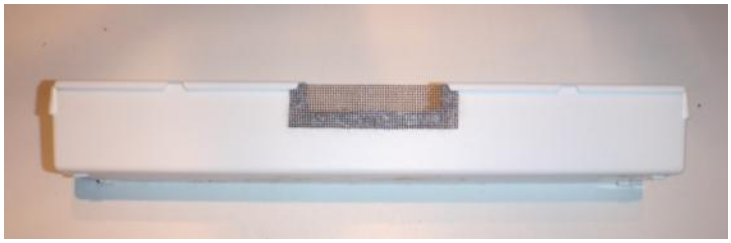
8. Tie both end of this rope together. This loop will be used to keep the frame vertical by providing easy location to secure ropes for anchoring trap.

PITFALL and overflow

9. Cut a hole into the side of the pitfall, approx 80 mm X 17 mm. Use a hacksaw to make vertical cuts and a knife to score the horizontal cut (then bend over and break off)



10. Cut a piece of screen approx. 10 cm X 2.5 cm, apply glue (Loctite 454 instant adhesive) on the outside of the container surrounding the edge of the hole and install the screen. Use plastic from the hole to push the screen in place and add glue to secure it. A catalyst may be used to make the glue set faster. The trap is now ready to be assembled outside in your favorite field site.



ASSEMBLING THE TRAP

11. Determine the direction of the dominant winds and configure placement of trap and screen to be **perpendicular** to the dominant wind.
12. Dig just the size of your pitfall container and avoid disturbing the surrounding area.
13. Install the pitfall container in the hole and replace soil so the pitfall is even with the ground.
14. While frame is in the ground pound one piece of rebar just outside each end of the pitfall container on the short ends of the rectangular pitfall container.
15. Install the frame. Use zip ties to secure frame to rebar, small holes in the netting for the zip ties, will be okay. Keep zip ties loose enough so frame can slide off rebar.
16. Use nails (1 or 2 on each side) and rope to keep the frame straight with the loop made on the top tubing of the frame. You may use simple knots to allow an easy removing of the frame to make it fast and easy to remove the container from the ground and drain its content.

PITFALL TRAP SOLUTION

To effectively capture and preserve the terrestrial arthropods a solution for the pitfall traps must be created. The solution will contain 20-30% propylene glycol (non-toxic antifreeze) and 70-80% water with a drop or two of a commercial-grade surfactant (e.g., TRITON X-100). Add this solution to the bottom of the pitfall trap to completely cover the bottom but does not pour out the overflow (about 1 inch or 2.5 cm deep). The solution should be changed periodically: weekly or when the solution is dirty. Pitfall trap solution should be disposed of by flushing down a toilet, putting down a gray water sump or at remote camps by dispersal.

Traps can be emptied by releasing the guy lines on the trap, lifting the screen off the rebar and placing it on ground. The pitfall container is accessible for removal and emptying of contents into Whirl-Pak (see Field collection procedures below)

Materials needed in the field for terrestrial sampling (see collection procedures below):

4 inch^2 or 12.5 cm^2 fine mesh dip net
Squirt bottle with water
Whirl-Paks (10 total per collection, 5 for mesic and 5 for dry)
Pre-labeled Rite-in-the Rain labels
Extra *fresh* water for wash bottle
Meter stick
Empty container for carrying used solution to camp
Sawed-off juice or milk jug for catching solution as it goes through net
Sponge

Pour contents (solution and captured insects) of pitfall container through the fine mesh fish net into an open container (e.g. big juice container) so solution can be returned to pit fall trap. Make sure the seam is on the outside so insects do not get stuck in the net's seam. The key is to first concentrate the collected material into the apex of the net by repeated dipping into a nearby wet area (but do not allow edge of net to go below surface since you may collect other material this way), or flushing with the squirt bottle. Then let excess water drain, or even blot the captured material from the outside. A moist sponge is great for this if you plan ahead, but you can use any absorbent material that is not already saturated (e.g. damp clothing, moss). Transfer insects from net into a standard 6-inch diameter Nalgene powder funnel fit snugly into the mouth of a wide-mouth, 4oz (125ml) LDPE poly bottle whose bottom has been cut out (see picture below). The cut off bottle serves as a funnel extension that can be inserted into the Whirl-Pak. This holds the Whirl-Pak open, and directs the sample into the bottom of the Whirl-Pak. Be sure not to leave material stuck to the inside of the bottle! One can hold the whole business in one hand, and use the other for the squirt bottle. Or if two hands are needed for the net and bottle, the unit can be propped up in the vegetation, in a pail or backpack, or held between the knees. It can be difficult to remove the net contents if the sides of the net bag stick together.

Aquatic monitoring

by Mac Butler – North Dakota State University

Aquatic food resources will be measured using sweep- net sampling techniques at all sites. This method entails collecting aquatic invertebrates at the water surface along the downwind edge of ponds. Selected sites (Nome, Canning River, East Bay) will also maintain surface associated activity traps (i.e., pop-bottles) as in 2010, placed just beneath the surface of the water in local water bodies where shorebirds have been observed feeding. It is recommended that each site return to the same 5 ponds investigated in 2010 to conduct aquatic invertebrate sampling unless these ponds proved not to be permanent. Ponds should have water present throughout the summer (i.e., permanent). It is advisable to look at remote imagery maps to look for ponds that appear very dark colored or use your personal experience at a site to locate permanent ponds. Permanent ponds are characterized by having rather sharp edges, edges lined with either sedges (*Carex* sp.) or pendant grass (*Arctophila sp*.), and mud bottoms in the middle.

During emergence of winged adults, all aquatic insect species become available to shorebirds that forage along pond margins. Collecting standardized sweep samples with a small dip net will permit monitoring of the seasonal timing of insect emergence. The bottle traps used in 2010 (and

retained at selected sites in 2011) collect invertebrates swimming or crawling in the top 10 cm of water, but detection rates of emerging insects are low with this method. Many arctic insects remain on the water surface to swarm and mate, while other species take flight and leave the pond. In either case, emergence failures and spent adults will accumulate along the leeward shore, where they decompose after several days. This flotsam contains the cast exoskeletons (exuviae) that are shed when aquatic insect larvae or pupae emerge to their adult form. By collecting a regular series of semi-quantitative samples of this washed-up material throughout the nesting season, one can document the seasonality of insect emergence. This material will reflect the whole insect community in a pond, including species from microhabitats that may not be sampled well by the activity traps. Arctic insects typically show highly synchronous emergence within a species, but both seasonal timing and the total span of community emergence can vary considerably.

Materials needed in the field for aquatic sampling (see collection procedures below):

Prepare ahead of time, you will need a standard set of Whirl-Paks or collection jars for each collection that can be used repeatedly throughout the season. Leave bulky supplies (net, funnel, bottle, etc.) within proximity of the transect, allowing sampling to occur en route from other projects.

- 5 inch^2 or 12.5 cm^2 fine mesh dip net for collecting sample
- Squirt bottle with water
- Whirl-Paks and sample pre-filled out Rite-in-the Rain labels. 5 per collection.
- Funnel with cut-off polybottle extension for sample transfer to Whirl-Pak.

Sweep net technique

Samples should be collected where there is windswept accumulation of material, so the exact sampling location may change between sampling dates. It's important to use the same sampling effort (i.e. five 1m sweeps) each time (e.g. across different days and ponds). Realize that less abundant species in one sample may be swamped by high numbers of another species, so a consistent effort is necessary for each species to have a similar detection probability. The most valuable data will come from counts of the pupal exuviae (cast skins), so be sure to follow through with consistent collection and sample processing, even if it appears that little or nothing is being caught.

Go to the downwind shore (or where wind has blown across the water surface for most of the past 24-hr (Figure 12). More material will accumulate during a period of time when there is more wind to produce greater waves. Look for white foam along the shore, or a line where the emergent vegetation may serve as a filter to trap drifting material. Use the 5-inch brine shrimp mesh dip net to skim the water surface along this line and parallel to the lake shore **for a 1-meter long "sweep"**. The net should gather materials floating <u>at the surface</u>, thus the bottom of the net can be about 2 inches (5 cm) below the surface, with the top half of the net above surface. Use a meter stick to estimate the distance of each sweep. Collect five (5) replicate 1m-long sweeps from the downwind edge of each pond; the <u>5 sweeps can be contiguous, but don't re-sweep the same 1m line.</u>

Deposit the material collected in all 5 replicate sweeps into a single Whirl-Pak. Any large pieces of vegetation can be picked out by hand and discarded, but should first be rinsed over the net with the squirt bottle to flush off clinging insects or exuviae. The key is to first concentrate

the collected material into the apex of the net by repeated dipping into the pond, or flushing with the squirt bottle. Then let excess water drain, or even blot from the outside. A moist sponge is great for this if you plan ahead, but you can use any absorbent material that is not already saturated (e.g. damp clothing, moss). Then the net bag can be inverted so the bulk of collected material falls, and/or can be readily flushed, into the container (or funnel). Use the funnel with polybottle extender to flush the material from the net into the bottom of the Whirl-Pak (Figure 13). This may take 3 hands: one to hold the container and funnel, and two to invert the net and rinse with the squirt bottle. One person may do this by holding the Whirl-Pak and funnel between feet or knees, thus freeing both hands. It is best to plan on doing this transfer in the field with fresh water (i.e. bring along a gallon jug of clean water so squirt bottle can be refilled) or pre-strained pond water, **being sure to decant and replace the water with alcohol after returning to camp**

At times you may notice little in the net, but under magnification there will likely be evidence of insect emergence, in the form of cast larval and pupal skins. These insect exuviae are often small and nearly transparent, but for some species they may exceed 10 mm in length and be grey, brown, or gold in color. At times of heavy emergence the insect bodies and pupal skins may accumulate in great quantity. Some sites may wish to collect samples daily during peak emergence.

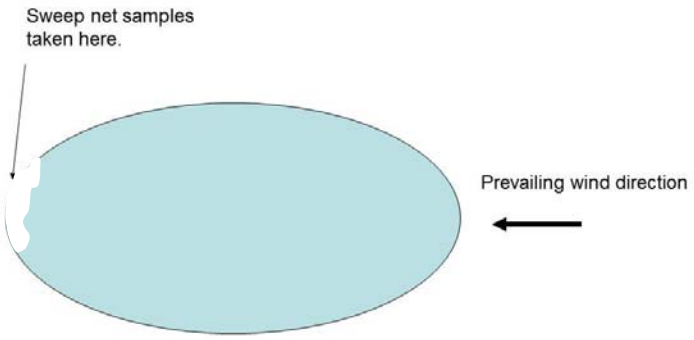


Figure 12. Schematic of location in pond where aquatic invertebrate emergence sweep net samples will be collected.

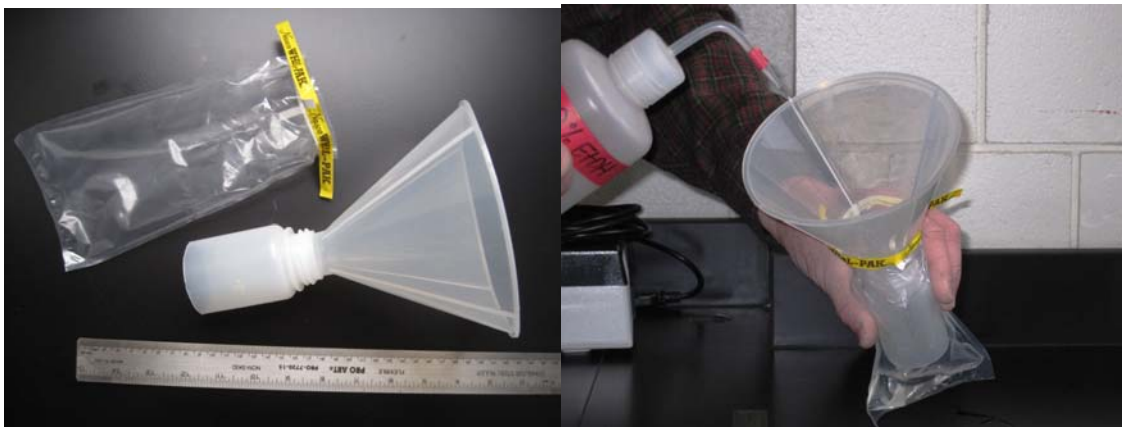


Figure 13. Powder funnel with polybottle extender (bottom removed), used for transferring aquatic samples into Whirl-Pak.

Building and deploying "pop-bottle activity trap" for monitoring aquatic invertebrate prey availability

Materials for trap construction:
2-liter carbonated beverage bottles (two per trap)
Closed-cell foam insulating board (for trap buoyancy and line float)
Duct tape
Silicone cement, most convenient in 150ml (5oz) tubes
Nylon cord, 1/8-inch diameter (for trap harness and anchor line)
Small aluminum carabineers or equivalent light-weight attachment clips (one per trap)
Zip-ties to secure floats to traps (>42cm single or combined length)

Tools:
Sharp cutting tool (single-edge razor blade, utility knife, or very sharp pocket knife)
Scissors
Heavy-duty hole-punch, drill with 3/16-inch bit, or awl
Felt-tipped marking pen
Ruler

Trap Assembly:
Rinse the pop bottles and save the caps (you'll need one cap for each assembled trap; keep some spares). Each trap requires two identical bottles. Cut the bottom off one bottle in each pair, right where the straight sides end and the curved base begins. Some bottle styles have a small ridge at this point. It's good to cut the first bottle as smooth and square as possible, so this one can be used as a guide in marking subsequent bottles. It may help to girdle the first bottle with masking tape, to be sure of your cutting line. A straight cut is best made by lightly scoring the plastic with a razor-sharp blade on the first pass, then cutting through on a second pass. The blade will follow the scored line, rather than tracking off. Alternatively, you can make an initial cut with a knife and use sharp scissors to follow the marked. The PET plastic used in these bottles is tough, but not easily repaired if cut wrong. The bottles are cheap (typically free), if you have access to trash or recycling.

Place the cut bottle over the neck of the second bottle, and push until the two are snug. Mark the second bottle with a felt-tip pen, then cut off the top end. This short top section of bottle #2 serves as the entry funnel for invertebrates that swim or crawl into the trap.

The two bottle sections are best glued together with silicone cement. Run a small bead of cement around the inside of the longer bottle section, no more than 1cm in from the cut edge. Stand the short funnel vertically, and carefully lower then longer bottle over the funnel. The cement should bridge between the trap's outer bottle and the entry funnel, so trapped inverts are less likely to get wedged in the tapered space.

Once the trap has dried (overnight) replace the lid and attach the harness. Pass a 70cm piece of 1/8" nylon line through holes (punched, drilled, or bored) on opposite sides of the entry funnel, 1cm from the edge. Use tabs of duct tape to reinforce the plastic. Tie a figure-8 knot on each end of the line where it passes outside the trap.

The trap should float just beneath the surface of the water, supported by a buoyant float attached to the top of the trap. Extruded polystyrene foam board ("blue/pink/yellow styrofoam") is easiest to work with, but expanded polystyrene (white bead board) will also work. In either case, the cut foam should be protected by a wrapping of duct tape. Float dimensions are: 2.5cm x 3cm x 15cm. The float can be glued or taped to the trap, but is most secure if also bound with a zip tie or string lashing. If long zip ties (~45cm) are not available, two or more shorter ties can be zipped together. NOTE: The trap in the attached pictures was photographed BEFORE the zip tie was added. If only duct tape is used to attach the float, the tape should go *all the way around* the bottle and *overlap itself*, or the float could come free in the water.

Traps should be connected to an anchored line with a light-weight clip such as a small aluminum carabineer, to allow quick release of the trap for emptying. A smaller float on the anchor line helps to retrieve the line to reattach the trap, and prevents the clip from weighing down the mouth of the trap. The length of the anchor line will depend on depth of the sampling location. Anchor lines need be only as long as this depth; the trap can drift on its harness downwind of the line float.

The intent is for these traps to collect invertebrates from the top ~10cm of the water column – the zone most available to foraging phalaropes. If possible, traps should be placed at the edge of any emergent vegetation, in water at least 12cm deep. In some pond types, this could be along the very edge of the pond shore; in other cases sedges may extend some meters out from shore. In yet other cases the entire pond area may be covered by *Arctophila* or other emergent plants – in which case the trap should be placed within the vegetation.

Trap Emptying Procedure:

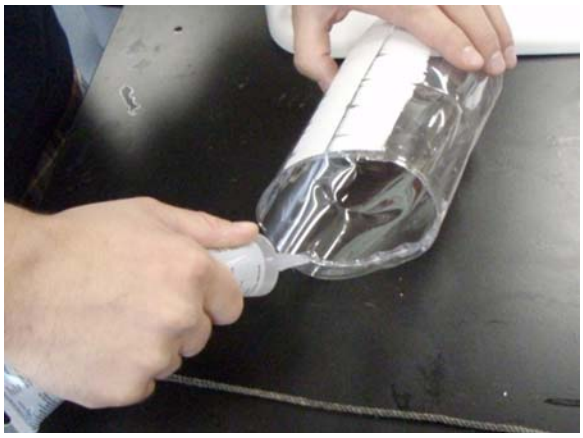
Materials needed: Fine-mesh sieve net, neoprene gloves, plastic containers for sample transfer (specimen cups with lids and labels), water-filled squirt bottle, spare bottle caps in case of loss, [isopropyl alcohol & Whirl Paks at base camp]

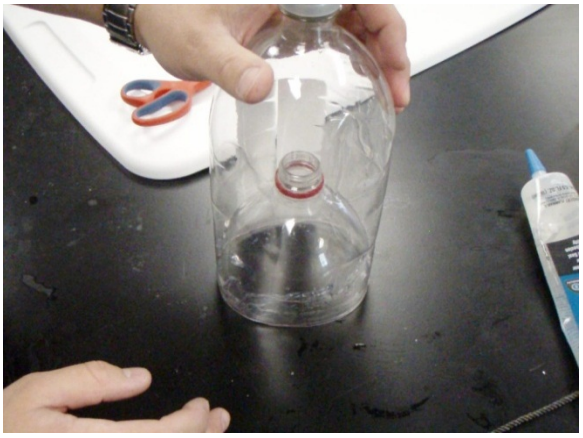
Make sure the bottle cap is in place before removing a trap from the water! Unclip the trap from the anchor line and lift by the harness. Snails and insect larvae will sometimes attach themselves to the sides of the trap. If there are any obvious animals in the funnel, but not inside the trap, try to include them in the sample. Animals on the *outside* of the trap are not part of the sample. While supporting the water-filled trap with one hand, pour as much of the trap contents as possible out the funnel end of the trap, and through the collecting net. Then turn the trap over,

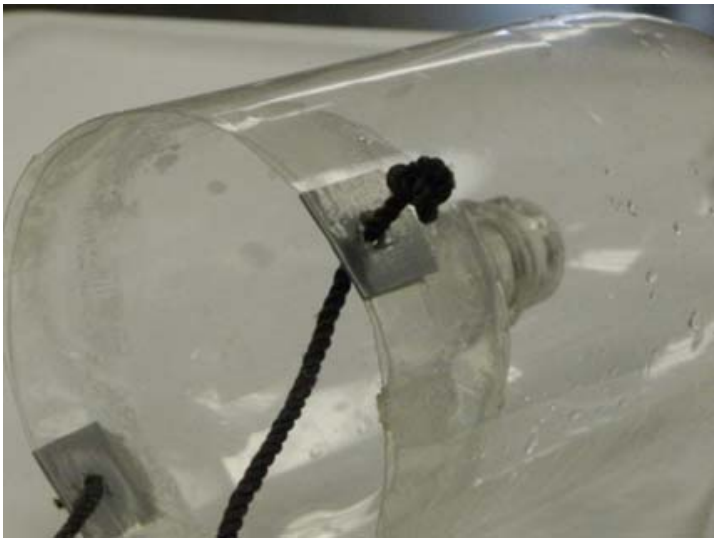
remove the bottle cap, and drain the remaining water through the neck. Look for any invertebrates still inside the trap, and dislodge them by swirling water in the trap, or by spraying water with the squirt bottle. Again rinse everything into the net. Replace the bottle cap, and set the trap aside.

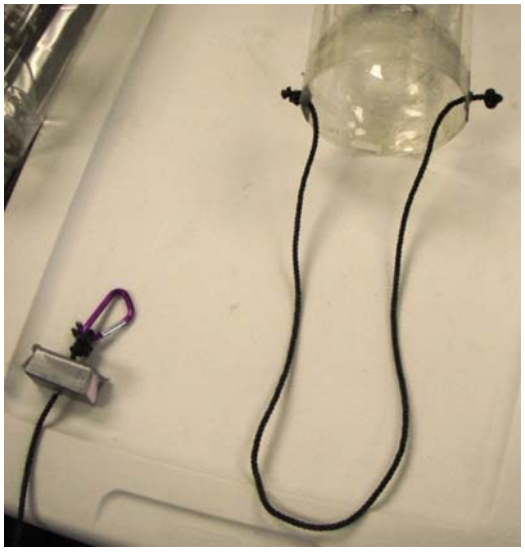
Transfer the contents of the net to a labeled collecting jar, using the squirt bottle. Material can be rinsed to the bottom of the net, and when the water has drained the net can be everted so the mass of material (invertebrates and any detritus or plant fragments that entered the trap) can be flushed into the jar. A Rite-in-Rain paper label with the date and pond/site identification written in pencil is best. If these labels are written ahead of time and put inside the collecting jars, they're less likely to be blown across the tundra and time in the field will be saved. The labels can be transferred to the Whirl Paks in camp when the samples are preserved. [Don't take alcohol into the field; using only water to collect the samples will conserve the isopropanol, which is difficult to resupply.]

Once the sample is securely in the collecting jar, redeploy the trap. The cap must be removed to allow water to completely fill the trap, so it will float level at the water surface. Then screw on the cap underwater. A pair of neoprene gloves will be appreciated! A few small air bubbles are OK, as long as the trap floats level beneath the surface.









Timing of trap installation, collection frequency,

All terrestrial (5 mesic and 5 dry) sweep net sampling and/or pop-bottle traps (5) should be established as soon the snow melts (for terrestrial) and ponds thaw (for aquatic). Typically traps will be installed in the following order: terrestrial dry, then, terrestrial mesic. Aquatic sweep samples should begin as soon as ponds are thawed; don't wait until you actually notice insects on the water surface. Do not wait until you can install all the traps to begin your collection. Start each as soon as possible. Relocate sites of traps from prior years and for the terrestrial pitfall traps chip out any water in the pitfall trap locations to expedite the thawing process. Once established, traps should be checked and emptied, and aquatic sweep samples collected, every **3 days** until the season is complete (e.g. end of shorebird chick hatch or brood – rearing). The sample collection of traps may be difficult for one person to accomplish if it is windy. Collecting five terrestrial traps should take 1 - 2 people, approximately 1 – 2 hours. Collecting aquatic samples should take about 1 hour for 1 person.

All samples will be collected with freshwater in individually labeled Whirl-Paks in the field, but then transferred and permanently stored in full strength isopropanol (e.g. isopropyl alcohol, rubbing alcohol) back at the office. Most materials (e.g. trap materials, trapping supplies and sample storage) will be provided by USFWS for camps (with the exception of the preservative and rebar), and should be on hand from 2010 field work.

Collectors should use caution to avoid impacting the micro site (within 1 meter) where the terrestrial traps are set while collecting the samples. Care should be given to avoid

compacting the soil around traps, or disturbing the shore and near-shore habitats during the aquatic collections.

Field collection procedures

Samples will be stored separately for each trap (regardless of type) in Whirl-Paks filled with preservative solution (full strength isopropanol). Do not combine samples from different traps. Each Whirl -Pak should have a cut (do not tear paper) Rite-in-the-Rain label inside that indicates the following information:

- collection date
- camp name
- sample type [for terrestrial (wet or dry) and for aquatic (sweep or pop-bottle)]
    - (e.g. Terre Mesic 1, Terre dry 1, Aquatic pop 1, Aquatic sweep 1).
- Collector's name

This information should be written in **pencil** (not pen) on Rite–in-the-Rain paper so the data are not lost once placed in preservative. DO NOT USE REGULAR PAPER AS IT WILL DESINTEGRATE IN ALCOHOL. Ink from Sharpie pens will also be lost in Alcohol. Cut the labels with scissors or a knife, because bugs can get caught in the rough edges of torn paper labels. The outside of the bags should also be labeled with an alcohol proof pen (see below).

Do not crush the specimens during transport back to camp.

Lab procedures (back at camp)

Within 12hr, decant as much water as possible from the Whirl-Pak, and **preserve the contents with full-strength isopropyl alcohol**. For proper sample preservation, the volume of isopropyl alcohol in the Whirl-Pak should be at least 4 times the volume of the arthropod sample or at least 2 cm at the bottom of the Whirl-Pak for small samples or about 10-20ml for most samples. Most air can be pressed out of the Whirl-Paks before tightly rolling the tops, but leave a small pillow of air/alcohol to minimize crushing of specimens. It's important to drain off most of the water first, so the isopropanol is not diluted. Use the net to recapture any insects when decanting the water, then back-flush any accidentally decanted material with a bit of alcohol. Realize that the collected materials and especially the insect pupae exuviae may be very small, clear, and difficult to notice among the detritus (but they will be in there!).

Sample storage

Record sample collection information on *food resources sample collection catalog* and enter into the database.

Besides the Rite-in-the Rain labels, samples should also be identified by writing on the outside of the Whirl-Pak with alcohol-proof permanent markers, beware that Sharpies are not alcohol proof, thus do not rely on this as the only way to mark the sample. Markers provided for blood collection will suffice here. Make sure that the Whirl-Paks are sufficiently inflated to decrease the crushing of samples when stored in a box. Store the samples upright until later identification and enumeration.

Pool all samples of one type in a larger plastic bag and label with Date of collection and contents and camp name (e.g. all terrestrial samples for one collection event). Double-bag the samples in a quart or gallon sized Ziplock bag to minimize leakage, store the entire collection of samples in secure plastic box at field station or camp.

We will not measure invertebrates in the field but rather ship them back to the USFWS in Anchorage for later enumeration. Samples can either be shipped directly to the entomology lab (this applies particularly to Canadian samples) or sent to the USFWS office in Anchorage. Coordinate with River Gates (hrivergates@gmail.com) prior to shipping samples.

Predator and lemming indices

These data will be used to create an index of potential predators and lemmings at each study site, which will be incorporated into analyses as covariates when investigating adult and nest survival rates of species. In 2010, a point count method was used to document predator numbers (see ASDN protocol V1) and Network collaborators requested a protocol change to address the low encounter/detection rate at some sites. Accordingly, an index approach was developed to cover a wider geographic area within each study area and allow predator abundances to be recorded throughout the day. Some camps may wish to continue doing point counts along with this new version (see methods at bottom) so that there long-term continuity is preserved. Thus, we have retained the methods for the point-count surveys in the version 2 protocols as well. We also added a lemming abundance count to this same protocol (i.e., lemmings and predators will be counted concurrently), although our winter nest count, live count (when extremely high densities of lemmings occur), and live-trapping for lemmings will also take place (but at only Nome, Barrow and East Bay).

Counts will be conducted daily or at least once per week depending on predator and lemming levels (high levels = less frequently) by at least one or multiple observers. Designated "predator-lemming counter (s)" will record observations of all predators and lemmings throughout the day regardless of their primary activity (e.g. banding, nest searching and/or environmental monitoring). When people are working as a team they will make efforts to not-double count predators/lemming but observations for the group can be summed. Please indicate on the dataform that the count represents a team effort and indicate number of observers in team.

Live counts will be conducted by the "designated counter" by keeping track of the number of predator and lemmings observed during the day's activity within the study area. Predator and lemming counts will only occur when people are within the study area and should not include observations gathered during transportation to and from the study area nor incidental observations while at camp (e.g. these observation should be recorded on the daily species list).

If the size of the study area is large enough to require multiple observers to count during the same time period, observers will count independently and make efforts to minimize double-counting. To make comparisons across Network sites, it is essential to not double count predators or lemmings (observers should not overlap in the geographic area surveyed), to only count individual predators or lemmings once, and to keep track of the time spent tabulating animals (either as a group or independent observer).

Daily Predator and Lemming Count data recorded

1. Observer name(s) (first initial and full last name)
2. Location: Plot ID/section of study area. For camps without plots, please define study areas surveyed using a map. This could be done at the beginning of the season to represent "sections of the study area" visited over and over. But we want to know rough boundaries as opposed to names that may not mean anything to anyone else. This is important so we have an approximate area surveyed during a given day.
3. Team effort (yes/no)
4. Number of observers
5. Date – dd-mm-yr
6. Time of arrival on plot/search area
7. Time of departure from plot/search area
8. Species observed
9. At end of the day (or when leaving a study plot/area), record the number of individuals for each species observed during your time within the study area. Indicate weather your number is an estimate or an exact number. This will make it easier to combine data across multiple observers later. See **Appendix H** for list of potential predator and lemmings, their scientific name and corresponding codes on the data card.
10. Notes
    a. Interactions among predators or with other species
    b. Observation of nest or adult predation on a prey item, or the detection of a predator nest.
    c. other
11. Observation of nesting/denning predators should be recorded – if a breeding location is found be sure to collect a GPS location, note the species, record nest/den contents (# of eggs or young) and how many adults. Since it will be hard for anyone to know if these data have been recorded yet, we encourage you to record this information during each survey. This will allow us to also document how long a nest/den is active.

FOR THOSE OF YOU NOT DOING SURVEYS ON A GIVEN DAY, PLEASE BE SURE TO RECORD ALL PREDATORS ON THE DAILY SPECIES LIST AT THE END OF THE DAY.

Lemming surveys

We will employ three ways to keep track of lemmings. The first will be a winter nest count that is done at the beginning of the spring season and indexes lemming abundance during the preceding winter. The second will be a series of live counts done either by directed personnel (see predator and lemming indices instructions) or by all people reported lemming sightings during daily species list. These focused lemming counts will be done daily throughout the summer. In the rare years and locations with lots of lemmings, a more directed lemming count will be done (see live transect count). The third method involves live trapping lemmings. This will only be done at a limited number of camp sites.

Winter Nest Counts

(adapted from Krebs et al 2008) See Winter Nest Count in data forms.

Natural history

Lemming abundance over the previous winter is relatively easy to measure indirectly by a survey for winter nests. Lemmings build winter nests of grasses and sedges under the snow and use them to keep warm. They appear to us like a ball of cut grass, (Figure 14) about 12 cm (5 inches) in diameter. Since they are abandoned in spring and not reused, they can be counted and picked up without harming the animals.

Figure 12. Lemming winter nest found during early summer snow surveys.

Both the brown lemming and the collared lemming build winter nests, as do voles like *Microtus* (tundra vole) and *Clethrionomys* (red-backed vole) in tundra habitats. It may be possible to tell what species constructed the nest from small amounts of hair left in with the grass, but this is relatively difficult and time consuming.

Methods

In most cases we would simply record the nest and not know what species constructed it. Count only fresh winter nests. Nests that are one year old are usually completely flattened and the grass has a grey color rather than a tan color. **All nests found should be ripped apart to avoid re-counting them the next year.** You may find gigantic winter nests 30 cm or more in diameter and lined with fur. These are weasel (ermine) nests. Weasels hunt lemmings and voles under the snow and convert lemming nests to their own use. Often you will find lemming stomachs left behind in weasel nests. We record weasel nests separate from lemming nests, since it gives a rough indication of the amount of weasel predation over the past winter.

Winter lemming nest surveys are best done as soon as possible after snow melt (early in summer), since high winds can blow the nests around after the snow melts. At high arctic sites where vegetative growth is low, these searches can happen later in the summer. They cannot be done with great confidence in dense willow habitats or in tussock tundra where the winter nests may often be invisible under the tussocks and willows.

To obtain a density of winter nests, we will employ the line transect method as follows:

- The observer walks a straight line searching visually for lemming nests. Upon sighting a nest, he or she records the perpendicular distance of the nest from the line of travel. The data set consists of these perpendicular distances and the total length of survey line the observer walks. This information will be later used to estimate density using the Program DISTANCE.
- This approach has several key assumptions, as illustrated in Figure 9. First, this method assumes that all winter nests exactly on the line of travel are detected. Second, as the distance from a nest to the line travel increases, the likelihood of detecting it decreases.

Another key point, if you detect nests you were not aware of when pacing out towards a nest (to get perpendicular distance or to destroy a nest), you should not include it in your data. Finally, the perpendicular distance to each nest seen is measured, no matter how far it is from the line of travel.

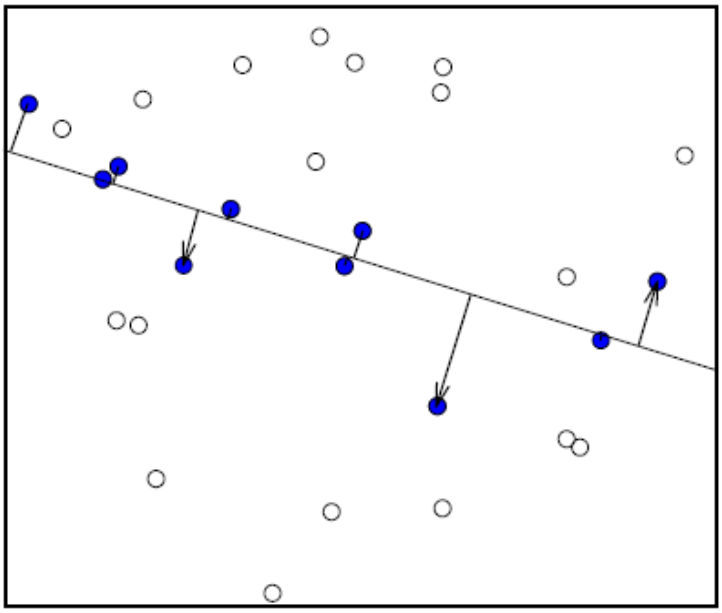


Figure 15. Schematic of winter nest count transect circles represent winter nests, and the line marks the survey line walked by the observer. Solid circles represent winter nests seen by the observer, and the lines mark the perpendicular distances measured to the center of the winter nest. Figure from Krebs et al 2008.

Line transect sampling should be done until at least 40 nests are seen and their perpendicular distances measured. In general, Krebs recommends about one day of walking effort per site will be sufficient to generate an estimate of the number of winter nests per hectare. The larger the sample size the more precise the estimate will be. The distance traveled can be determined from a GPS, or alternatively by following grid lines within your plot and then summing up the distance from grid stake to grid stake. Record start and stop points for your line transect on the camp GPS coordinates page. On the tundra it is clearly impossible to walk a straight line, but this should not matter as long as one does not double back to cover the same ground.

Data to record on Lemming Winter Nest Count

1. Site (Network site name)
2. Date (dd-mm-year)
3. Start time (24 -hr)
4. End time (24-hr)
5. Observer (e.g. first initial and last name)
6. Approximate distance (km): Estimate distance walk for transect count based on GPS data or grid stakes.
7. Start location: (Unique plot stake id or GPS location)
8. End location: (Unique plot stake id or GPS location)
9. Nest number (Used to help keep perpendicular distances unique and track sample size)
10. Perpendicular distance to nest (measure in decimeters)

Live lemming counts

(1) Daily prey counts (do in low to mid lemming years) – see "Predator and Lemming Indices instructions

These data will be used to create an index of lemmings at each study site to examine how these factors affect adult and nest survival. In 2010, observers recorded lemmings anecdotally and tallied up the number of lemmings observed by all people on the daily species list. An effort was made to have special "live lemming days" but this proved unsatisfactory since people either forgot to focus more on lemmings on these days or lemmings were so rare these days seemed meaningless (i.e., data from these days were no better than other days). To standardize the location (i.e., on bird study areas only) and amount of time spent counting, we developed an index approach to cover a wider geographic area within each study area and record predator and lemming abundance throughout the day. We advise camps continue to tally lemmings on the daily species list as in the past but these more focused survey protocols should be better.

(2) Live transect counts (do only on high lemming years! e.g., 100 lemmings seen per day per person)

In the rare years where lemmings are everywhere and it is impractical to count them during regular duties, lemmings will be indexed by conducted focused lemming transects that includes distance traveled and time in the field. Here, the observer should focus entirely on lemmings and not be doing other things. It will likely be sufficient for one person to spend two hours doing such counts, and should travel at least 2 kilometers. These counts should also be done weekly (intensive effort) or three times per year (early, mid and late for less intensive effort).

(2a) Data to record on lemming live transect counts

(see page 83)

1. Site (Network site name)
2. Date (dd-mm-year)
3. Start time (24 -hr)
4. End time (24-hr)
5. Observer (e.g. first initial and last name)
6. Start location: (Unique plot stake id or GPS location)
7. End location: (Unique plot stake id or GPS location)
8. Transect ID (Unique transect identification code that corresponds to meta database
9. Approximate distance (km): Estimate distance walk for transect count based on GPS data or grid stakes.
10. Species (4 letter species code, see **Appendix H**)
11. Total number of individuals observed / species.

LIVE TRAPPING

Use live trapping on trapping grids to obtain density estimates of each rodent species present. Information on sex, age and breeding condition can be collected if desired. Live trapping is considered the most accurate approach for obtaining a true estimate of the number of lemmings that occur in an area, and will be used at a limited number of sites (most likely Nome, Barrow, East Bay, and Bylot Island) to correct our cruder indices.

Consistency in Approach between Camps

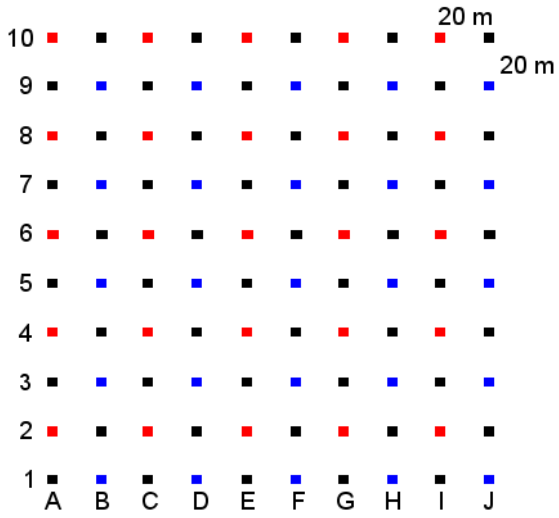
There are a number of constraints that will make following the protocol below difficult for all camps. Among these constraints are trap type. Longworth traps are recommended due to the fact that they have box on the end that can be filled with food and bedding to minimize lemming mortality due to exposure. We recognize however that these traps are expensive and that several camps may need to use the cheaper and potentially more accessible Sherman traps. Another constraint might be the topography of the study site. Although we recommend that square trapping grid below, traps set along a transect might be preferable in areas with lots of wetlands. Also some researchers may prefer using a "chevron" trapping grid (L-shaped). Despite these differences, our hope is that each camp can generate a density estimate that is both reliable and comparable. At present I do not know if there are any differences in how effective Sherman and Longworth traps are in capturing animals but given we are not in a position to effect change, we will need to live with this.

Time Period

Live trapping should be done twice every year, once soon after spring melt, and once before the snow sticks in late summer. This is the best way to infer stage in a cycle, because populations can change dramatically during the summer, and mid-summer trapping alone does not give a picture of population trend. Given the time constraints of most ASDN camps, we anticipate only a single round of live-trapping to be conducted (shortly after snow melt) but encourage more if possible.

Trapping grid setup

Use a 100 m long measuring tape and a compass to measure out a 10x10 trapping grid with 20 meter spacing (3.24 ha), or a grid of larger square shape, such as a 16x 16 grid with 20 m spacing (9 ha). This can be done with 2 people but 3 people are most efficient. Mark each grid point with a wooden stake (wooden surveyor's lathe, 60 cm lengths) labeled with the row and column. Place a Longworth (recommended) or Sherman trap at a suitable site within 5 m of every second grid station for a total of 50 or more depending on the size of the grid (or every grid station if the population density is high and the large majority of traps are occupied each check). Stagger the trap placement so they are evenly spaced over the area (black squares represent traps in the schematic below). It is useful to color code the rows to help with orientation while in the middle of the grid. One way to do this is to temporarily place red and blue surveyor flags next to each station. These can be picked up at the end of trapping session (or laid flat on the ground) to minimize wind damage to the flags.



Setting traps

Oats – Place a small handful of whole oats in the box of the Longworth traps or at the rear portion of the Sherman traps (careful not to get under treadle)

Bedding - Line the box of the Longworth traps or the back of the Sherman traps (again careful not to get under treadle) with upholstery cotton or wool fleece for bedding. Use just enough cotton or fleece to create a warm nest. If too much cotton is stuffed in the box, the lemmings may view it as a blockage as opposed to nesting material.

Apple – Place a chunk of apple in the box in front of the bedding. We get 32 squares from a large apple. Cut in squares rather than slices to minimize surface area and desiccation.

Trap assembly –

Longworth traps: Unlock the locking mechanism of the Longworth tunnel and check that the door falls properly when the treadle is depressed. Adjust the mechanism if necessary and when all is in good working order, set the trap and connect the tunnel with the box. Make sure that the apple is not too close to the treadle.

Sherman traps (3”x3.5”x9” folding aluminum): unfold flattened trap so it becomes rectangle in shape. Adjust treadle holder so the door will snap shut easily. When setting traps in field, be sure food and cotton are not too close to entrance so as to prohibit door from closing.

Trap placement – Place traps as close to the stake marking the location as possible (maximum 5 meters radius from stake). Search the area carefully for fresh sign such as an active burrow or runway. If you find a good runway, place the trap right on the runway. If there is a burrow, place the trap in front of the entrance but do not block the entrance. If there is no sign, place the trap under cover if possible. Make sure the trap is flat and stable and make sure that the door hasn’t fallen shut or the apple rolled under the treadle while placing the trap. Place a board or cover over the live trap to shelter it from too much sun or rain.

Checking traps

A trapping session should last 2 days (48 hours) and all traps should be checked every 4-6 hours. (If you do not want to check traps at night (we prefer not to), it is acceptable to set the traps early in the morning, check every 4-6 hours and then lock the traps open at the last evening trap check. Traps will have to be re-opened the next morning.) You should have at least 12 trap checks in a 2 day trapping period. Do not trap more than 2 days in a row or "trap-happy" individuals will be caught too often and may die in the trap. At the last trap check the doors should be locked open and the traps should be left in 2 pieces so that there is no chance of an animal being caught by mistake. <<< differs from approach proposed by Eider team >>>

Place a few cm of oats in the bottom of your trapping bucket and walk past every trap. If you find a trap with the door closed, gently open the trap in the bucket and tip the animal out onto the oats. The oats provide a soft landing surface for them. At this point you can check species and get your tagging equipment prepared. Scoop the animal up with a gloved hand (see footnote 1 below). Use the other un-gloved hand to get a good grasp of the tail. Be careful not to squeeze the animal and keep the eyes covered to minimize squirming. Check carefully for an eartag and record the number if it's already tagged. If a new tag is required and the species is *Dicrostonyx* you will need 2 people to tag properly. The person handling the lemming can use both hands to hold it so that its right ear is exposed but eyes covered. The person tagging can use the blunt tweezers to gently extend the ear flap with one hand and tag with the other hand. Make sure the tag point has pierced through the ear flap and tag hole and folded over properly (check each tag's alignment before placing in the pliers). Be careful to not catch too much skin in the tag (increases chance of infection) but place the tag far enough in so that it doesn't easily rip out. The ears of brown lemmings and voles are big enough that they can be successfully tagged without the use of tweezers. Record the tag number.

Check the sex, reproductive condition and weight of the animal. Record the data and release the animal. Reset the trap with fresh bait and dry bedding.

If the animal caught already has a tag and was caught earlier in the same trapping period, you do not need to check the sex and weigh the animal. Simply record the tag number, location, and which check number it is then release the animal.

Equipment needed

- 100m measuring tape and compass for setting up trapping grids
- 20 litre bucket with minimum 40 cm high sides for holding animals
- Traps and trap boards
- Whole oats
- Apple (preferably Granny Smith)
- Upholstery cotton or wool fleece for bedding
- Ear tags & tagging pliers (or injectable transponder tags and readers if you have lots of money)

- Blunt tweezers for holding tiny *Dicrostonyx* ears when tagging
- 100 g Pesola scales for weighing adults; 30 g scale for juveniles; 300 g scale for some pregnant females.
- Light drawstring bag for holding the animal while weighing
- Trapping gloves
- Bandaids

Data analysis

Data should be entered into a database and checked immediately for errors. Density estimates can be computed using Program Capture or Program MARK, or by the simpler Petersen or Schnabel estimators.

Footnote 1: another method for handling is to place a clear plastic bag around the trap. Hold the bag shut and work through the bag to break the trap open and release the animal into the bag. Scruff the animal through the bag so that you can open the bag and remove the trap without letting the animal escape.

Weather

Daily weather conditions will be measured at each site using a locally established field camp weather station (HOBO U30 datalogger) or by retrieving data from established weather stations (e.g. Barrow, Prudhoe Bay, Churchill sites). The field camp weather stations will gather hourly measures of air temperature, relative humidity, wind speed and direction. Precipitation will be measured manually at each site with a rain/snow gauge. Weather stations should be checked daily to ensure they are functioning and data should be downloaded **weekly** using the data back up data shuttles. Be sure to download the data off the shuttles and view this on the computer to be sure data are being recorded correctly. In the event that the field station weather station is not recording it is important to collect daily minimum weather conditions (see below for procedures).

Automated weather stations specifications

Measurement and location of sensor:	**Instrument specification:**
Air temperature and relative humidity (2.5 m above ground)	Smart Sensor Temp/RH (Onset model S-THB-M002)
Wind speed and direction (3 m above ground)	Smart Sensor Wind speed and direction (Onset S-WCA-M003)

Precipitation (see below) (½ meter above ground)	A separate rain gauge is employed for this. It is not part of the automated station but needs to be mounted at correct height.

Daily precipitation – recorded manually (from Cadieux and Gauthier 2008)

We want the data to represent the daily accumulation of precipitations, it is recorded only once at the end of **every day** between 9PM and 10PM. If it does not rain, you can put zero without checking it. This can be adjusted depending on the latitude of each study sites. Precipitation is recorded in **millimeters**.

Precipitations include rain, drizzle, freezing rain, freezing drizzle, hail and snow. All these types of precipitations, *except snow*, can be measured using a standard rain gauge (pluviometer) installed at 1.5 meters above the ground. It is important to make sure that it is installed **vertically** and that nothing will obstruct the arrival of the rain within a radius of a few meters from the funnel. Hence, care must be taken to locate the rain gauge above the highest point of the structure where it is installed (e.g. roof of a low building or a tent).

Snowfalls are not recorded using a pluviometer. Instead, it should be estimated by measuring the accumulation of snow (in cm) at a few locations on the ground using a ruler. The average of these measures can then be transformed into rain accumulation using the "ten-to-one" rule (1 cm of snow = 1 mm of rain). See data forms for manual weather recording.

Manual weather recording
(in the event the automatic weather recorder goes down)

Daily air temperature Minimum and maximum air temperatures are recorded twice daily (12 hrs apart) at consistent time period.

Daily wind patterns A description of the daily wind including speed and direction (e.g. N, W, S, E, NW, NE etc.) should be noted in the daily journal. Please include pattern of wind throughout the day, a range of wind speed for the day and the predominant direction.

Snow and surface water

Snow cover is believed to be a key determinant of timing of breeding for shorebirds (e.g., Meltofte et al., Smith et al. 2010). Our intention is to monitor snowmelt with sufficient resolution to determine the date of 50% melt, as well as variation in rates of melt among years. Throughout the season we will collect frequent visual measures of relative surface cover (e.g. snow [includes slush], water and land) to quantify the seasonal changes in hydrology. All sites will collect at least 10 replicate samples of relative cover at permanent locations (Please record GPS locations of each survey location in the meta-database) preferably **every other day** during the beginning of the season when the snow melts quickly, and **weekly after the snow has melted until the field season is completed.**

For sites with established plots, relative surface cover will be quantified by randomly or systematically selecting **at least ten 50- m^2 quadrats** within the study area and visually estimating absolute snow (which includes ice and slush), water and land (bare or vegetated ground) cover within the quadrat **to the nearest 5 to 10%, totaling to 100% for each station** (for plots, you might reference each station with the northwest stake number).

For sites without established plots, relative surface cover will be quantified by establishing 10 survey stations and estimating snow cover in a **ten 50-m^2 quadrat** surrounding the observation point. In order to increase the accuracy and consistency of this method, we ask that observers pace 50 meters in at least 2 directions and pin flag the corners and the center. Efforts should be made to have surface cover sampling areas be spread throughout the study area as much as possible to give a representative sample of snow melt for the area. Camp leaders can reduce observer bias by training all observers in estimating cover at the beginning of each season or designating one observer to conduct all snow surveys. Crew leaders can have multiple observers estimate cover and discuss ways to calibrate between observers. Cover estimates are more accurate if observers subdivide area into smaller units and estimate cover for each subunit and sum for the total estimate.

For example, the NW quadrat (Figure 10) contains 25% land, NE quadrat contains 5% land, 10% water and 10% snow, the SW quadrat contains 15 % land, 10 % water, and the SE quadrat contains 15% land, 5% water and 5% snow for a summed total of 25 % water, 60 % land and 15 % snow.

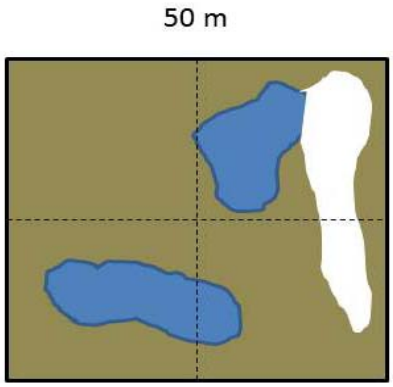


Figure 16. 50 m^2 quadrat for estimating relative surface cover. Surface cover values for each category illustrated here are as follows: 25 % water, 60 % land and 15 % snow.

Pond hydrology: temperature monitoring, water level monitoring, and habitat measurements

This supplement describes methodology for collecting data to complement the aquatic invertebrate emergence phenology studies. Please don't hesitate to contact Daniel Rinella (University of Alaska Anchorage; rinella@uaa.alaska.edu; 907-748-2154) if you have any questions regarding these activities.

Activity	Frequency
A. Temperature monitoring	Install at beginning of season and remove at end of season
B. Water level monitoring	Each time invertebrate sampling is conducted
C. Habitat measurements	When ice is melted in the pond & when you can

A. Temperature monitoring

- Equipment list provided by **ASDN**: 5 Hobo temperature loggers, plastic cable ties

- Equipment list provided by **camp**: 5 rebar (4- ft) stakes, hammer for driving stakes, chest waders for deploying loggers.

1. Deployment: For each pond, record temperature logger number and other requested information on the *Pond water level and temperature monitoring datasheet*. Note that each pond will have its own datasheet.

   Deploy one temperature logger in each of the 5 ponds at the beginning of the field season. Temperature loggers should be deployed in a location with relatively deep water so they do not go dry when water levels fall. Attach the temperature logger directly to the rebar stake using two cable ties, making sure the sensor end (i.e., the end with the hole) is pointing down. Sink the stake into the pond bottom so that the temperature logger is just above the pond bottom. Check periodically to ensure that temperature logger remains near the pond bottom; adjust the depth if necessary. If water levels fall enough that any temperature logger is in danger or going dry, move it to a deeper location.

2. Retrieval: pull the rebar stake from the pond, cut the cable ties holding the temperature logger, and return all the temperature loggers to USFWS Regional office

B. Water level monitoring

- Equipment list provided by **ASDN**: short piece of wire, nylon string, line level, meter stick or tape measure,
- Equipment list provided by **camp**: 5 rebar (4-ft) stakes, hammer for driving stakes, pliers for twisting wire

1. Prior to the first water level measurement, drive the rebar securely into the pond shoreline. It is preferable to place it along the shoreline where there is a sharp edge to limit the wading distance required as lake levels drop over the summer. Once the rebar is in place, make a permanent mark on the rebar near the ground by wrapping a piece of wire around the stake and twisting it in place with pliers. The wire will serve as an elevation benchmark for the duration of the study, so leave the rebar in place. Record GPS coordinates of the benchmark on the *Pond water level and temperature monitoring datasheet*.
2. The vertical distance between the benchmark (i.e., the wire) and the pond surface will be measured each time aquatic invertebrate sampling is conducted. To do this, tie a string to the rebar at the benchmark (i.e., the wire), unspool the string until it extends over the water surface, and hang the line level on the string. Raise or lower the pond end of the string until the bubble is centered within the line level. You may need to clear some vegetation to have a clear path for the string.
3. Once the line is level, measure the vertical distance between the line and the water surface (see Figure 11). Record this distance (along with the date) and any comments on the *Pond water level and temperature datasheet.* These repeated measurements will allow us to track changes in water level throughout the sampling season.

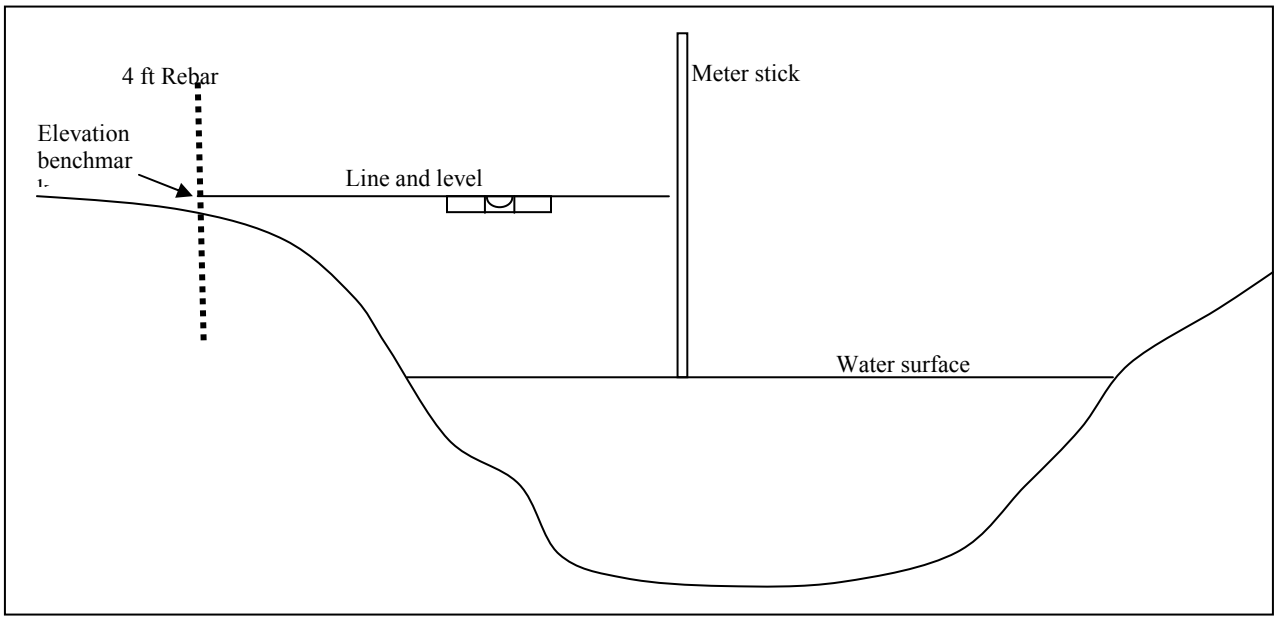


Figure 17. Schematic showing method for measuring vertical distance between elevation benchmark and pond water level.

C. Habitat measurements

- Equipment list provided by **camp**: meter stick (or longer graduated stick) for measuring depth, camera, GPS, chest waders for person conducting measurements
- Procedure for each pond: (a) record a GPS track of the shoreline perimeter to calculate pond's surface area, (b) record depth measurements and substrate type at a systematic array of 15 points distributed across the pond, (c) take a series (6 – 8) of photos.
    1. Walk the shoreline with a handheld GPS and have it calculate the pond's surface area. Record the surface area measurement on the *Pond habitat datasheet*. The procedure for measuring areas varies by GPS make and model, so check your owner's manual. If you have a Garmin etrex GPS, go to the main menu, select *Tracks*, press the menu button again, select *Area Calculation*, select *Start*, walk the pond's perimeter, then select *Stop*. The etrex will then calculate the area.
    2. For the depth and substrate measurements, visually locate three equally spaced transects that will run the length of the lake and take measurements at five points roughly equally spaced along each transect. Thus, measurements will be taken at 15 points in a grid pattern equally distributed across the lake (see Figure 12). Record the approximate distance between each of the three transects and the approximate distance between each of the 5 sampling points on each transect on the datasheet. At each of the points, lower a meter stick (or make a longer graduated stick if necessary) to the bottom and record the depth and type of bottom substrate on the *Pond habitat datasheet*. The various substrate categories are given on the datasheet; be sure to record the two-letter code. If pond is too deep or soft to safely wade, then note that on the datasheet and skip the depth and substrate measurements. Also, record the predominant color of the bottom substrate on the datasheet.
    3. Take 6 – 8 photos of the pond from various angles and distances. Include a few photos that show the crew taking habitat measurements, measuring the pond water level, and collecting invertebrate samples. Include any other interesting photos from the pond. Store all the photos from a given pond in a folder bearing the camp code and pond number.

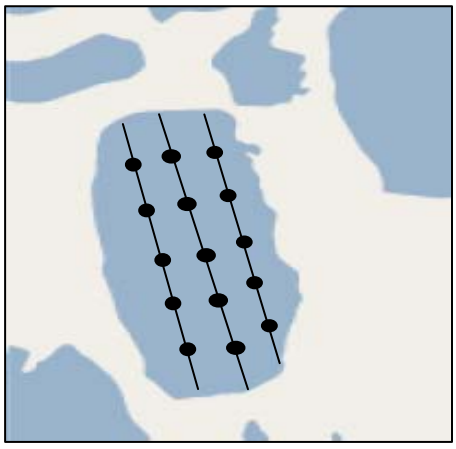


Figure 18. Schematic showing layout of 5 sampling points on each of three equally spaced transects.

D. Pond notes.

Please record notes regarding each pond on the *Pond notes datasheet*. Include comments on water level fluctuations, periods of heavy aquatic insect emergence, bird activity in and around the pond, or anything else of interest. Please include dates of observations and feel free to attach additional sheets as necessary.

APPENDICES

Appendix A. How to set up and label intensive nest search plots.

Plots consist of 1 meter tall wooden stakes placed in the ground every 50 meters. The top of the stakes are painted white (or a neutral color) and then painted with the alpha-numeric numeric code. Be sure the alpha-numeric numeric code is sufficiently large to see from at least 50 meters. This will require putting the letters and numbers on top of each other as depicted below. Letters and numbers written adjacent to each other (left to right) will be too small to read. Periphery stakes can be placed during the early part of the year when the ground is frozen and stake placement is difficult, and then the remaining stakes can be installed later in the year. Use GPS units to locate stake positions.

A 1	A 2	**A** 3	A 4	**A** 5	A 6	**A** 7	A 8	**A** 9
B 1	B 2	**B** 3	B 4	**B** 5	B 6	**B** 7	B 8	**B** 9
C 1	C 2	**C** 3	C 4	**C** 5	C 6	**C** 7	C 8	**C** 9
D 1	D 2	**D** 3	D 4	**D** 5	D 6	**D** 7	D 8	**D** 9
E 1	E 2	**E** 3	E 4	**E** 5	E 6	**E** 7	E 8	**E** 9
F 1	F 2	**F** 3	F 4	**F** 5	F 6	**F** 7	F 8	**F** 9
G 1	G 2	**G** 3	G 4	**G** 5	G 6	**G** 7	G 8	**G** 9
H 1	H 2	**H** 3	H 4	**H** 5	H 6	**H** 7	H 8	**H** 9
I 1	I 2	**I** 3	I 4	**I** 5	I 6	**I** 7	I 8	**I** 9

Appendix B. Species-specific nest searching tips

Species	Mating system	Adult who Incubates	Nest location	Flushing distance	Flushing and Other Unique Behaviors	Return time
Single adult incubation						
REPH	Polyandry – focus on males	Male	Wet	Close	Adult leaves area and monitors nest from a great distance; upon returning the male will typically fly from one little pond to the next until eventually reaching its nesting pond	moderate
RNPH	Polyandry – focus on males	Male	Wet	Close	Adult leaves area and monitors nest from a great distance	moderate
PESA	Polygynous – focus on females	Female	Wet	Moderate	Often does rodent run and alarm calls ; male can be watched – will sometimes fly and "boom" over female who may be either feeding or on a nest	moderate
BBSA	Lekking – single male display indicates nest	Female	Moderate	Close	May do rodent run and alarm call; single male occasionally displays near nest site; frantic feeding female likely on break -- can be followed to find nest.	quickly
WRSA	Monogamous to Polygynous	Both or female	Moderate			
Bi-parental incubation						
DUNL	Monogamous – key on male display	Both	Moderate	Moderate	Both adults do rodent runs, but females more likely. Frequently flies off nest low	Quick to moderate

					and then lands and calls.	
SESA	Monogamous – male display	Both	Mod to dry	Close	Both adults do rodent runs, but females more likely.	quickly
LBDO	Monogamous - pairs	Both	Mod to wet	Very Close	Tight sitter, does not flush easily; very secretive upon return to nest.	Slow
STSA	Monogamous – male display	Both	Mod to wet	Close	Tight sitter, does not flush easily	moderate
AMGP	Monogamous – look for pairs	Both	Dry	Far	Will stand up or leave nest when observer far away (>100 m)	Quickly if observer is out of sight
BASA	Monogamous	Both	Dry to mod	Close	Rodent run and alarm calls	quickly
WESA	Monogamous	Both	Mod to wet	Close	Rodent run and alarm calls	quickly

Appendix C. Egg flotation - A method to determine egg age

Materials

1. **Float container** (e.g. 3 inch plexiglass cube) with compass angles written on side
2. **Water** from the nearest natural water body

Methods

If a shorebird nest contains a full clutch of eggs (typically 4) when it is discovered, float at least 2 eggs. If the 2 eggs differ in angle significantly float the 3rd egg (and 4th if necessary).

1. Place eggs on the bottom of the jar before releasing to prevent egg damage from dropping and to ensure they are not held by surface tension. Float each egg separately but keep track of the ones you've already floated.
2. With a protractor, measure the angle between the bottom of the cup and the center axis of the egg to the nearest 5° for each egg. Record the angle on the Nest discovery and fate data sheet.
3. If the egg floats at the surface, using a clear ruler, record the # of millimeters above the water surface that is exposed to the air and also use the protractor to measure the angle of the egg in the water column. Keep in mind the egg may float at the surface but not break the surface. Record these measurements while you are viewing the floating egg at eye level.

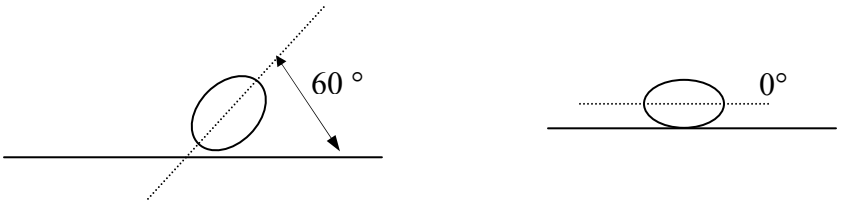
Carefully place eggs back in the nest. Remember to record the date you floated the egg.

Important

Float shorebird eggs **once** during the course of incubation unless you have issues with the original float data. **Float eggs on the discovery visit** because the nest may be depredated on your next visit and age estimates tend to be more accurate when eggs are floated early in incubation.

Use the float program in the Electronic Appendix (float_program.xls) to estimate the age of the eggs when calculating the estimated hatch date in the office. The program uses regression equations specific to species or use Float Tables in **Appendix J,** that were originally published in Liebezeit et al. (2007)

If the temperature is below freezing do not float the eggs. Do it on the next visit.



Appendix D. Nest stage periods for common breeding birds

Species	Laying (days)*	Incubation (days)
Shorebirds		
Black-bellied Plover	4	23-27
American Golden-Plover	4	26-27
Pacific Golden-Plover	4	25
Semipalmated Plover	4	24
Greater Yellowlegs	4	23
Lesser Yellowlegs	4	22 - 23
Whimbrel	4	27-28
Bristle-thighed Curlew	4	26
Hudsonian Godwit	4	22 - 23
Marbled Godwit	4	24 -26
Bar-tailed Godwit	4	20-21
Ruddy Turnstone	4	22-24
Black Turnstone	4	22 -24
Surfbird	4	22 – 24
Red Knot	4	21 -23
Sanderling	4	23 -27
Semipalmated Sandpiper	4	20-22
Western Sandpiper	4	20-22
Least Sandpiper	4	19 - 23
White-rumped Sandpiper	4	21-22
Baird's Sandpiper	4	21
Pectoral Sandpiper	4	21-23
Rock Sandpiper	4	20 - 21
Dunlin	4	21-22
Stilt Sandpiper	4	20
Buff-breasted Sandpiper	4	23-25
Long-billed Dowitcher	4	21-22
Short-billed Dowitcher	4	21 -22
Red Phalarope	4	18-20
Red-necked Phalarope	4	19-21
Water birds		
Pacific Loon	2	23-25
Red-throated Loon	2	24-26
Yellow-billed Loon	2	27-29
King Eider	4-6	22-24
Common Eider	3-5	24-26
Long-tailed Duck	5-7	26
Northern Pintail	7-9	22-24
Tundra Swan	3-4	31-32
Brant	3-5	22-25
Greater White-fronted Goose	4-5	25
Snow Goose	2-6	23-24
Cackling Canada Goose	4-7	25-30
Passerines		
Lapland Longspur**	4-6	11-13
Other birds		
Willow Ptarmigan	4-14	21-23
Rock Ptarmigan	6-13	20-26
Pomarine Jaeger	2 -3	26 - 28
Parasitic Jaeger	2	25-28
Long-tailed Jaeger	2	23-25

* Birds typically lay one egg / day. ** Nestling stage = 8-11 days

Note: The information in this Appendix was obtained from various sources in the literature (mostly BNA accounts and the Birder's Handbook – Ehrlich et al. 1988). Nest stage lengths may vary somewhat across sites.

Appendix E. Using eggshell remains to determine nest fate in shorebirds

The presence of pip fragments and eggshell "tops" and "bottoms" at Piping Plover, Snowy Plover, and Killdeer nests typically indicate a successful hatch (Mabee 1997). This evidence is probably reliable for assessing the fate of other precisian species. The following is a brief description of a methodology for finding eggshell remains and assessing fate of the nests they are associated with.

Egg shell top and bottoms: those parts of an eggshell that exhibit a nearly equidistant length from the center of the top or bottom eggshell to the broken edge of the shell. Many hatching chicks pip through an eggshell at a fairly uniform level around the top of the egg and produce well-defined tops and bottoms.
Eggshell fragment: pieces that range from ~ 1 to 5 mm. Fragments within this size range are expected in successful nests because chicks break through the eggshell and produce small pipping fragments when hatching.
Eggshell pieces: any piece larger than 5 mm in length. Fragments > 5 mm may be found in both successful and depredated nests and are less reliable in classifying nest fate.

For finding eggshell tops and bottoms:
When a nest is no longer active and you are assessing its fate, search within a **5m radius** of the nest for eggshell tops and bottoms. Most shorebirds remove eggshells from the nest immediately after the chicks hatch. They often deposit the eggshells not far from the nest. Remember to check to see if the membrane adheres to or easily pulls away from the eggshell "tops" and "bottoms". This may be difficult to determine for the smaller shorebird species.

For finding pip fragments
With the tip of a mechanical pencil or tongue depressor carefully pull away the top layers of the nest. Continue all the way down to the soil. Pip fragments often sink into the nest lining.

1. Some birds may re-use a nest scrape from a previous year and may contain pip fragments from a previously successful nest. Only count pip fragments that look new. New fragments typically are bi-colored (i.e. different colors on the 2 sides – mottled on outside of egg, white on inside).
2. If you do not find fragments after reaching the bottom of the nest, place the nest contents on a sheet of white paper (or on the center-fold of your rite-in-the-rain field notebook). Carefully go through the nest contents as you place it back in the bowl.
3. Inexperienced researchers may collect the nest, place it in a plastic bag or Tupperware and bring it back to the office for further scrutiny.

- Be careful not to confuse egg fragments with pieces of lichen
- Sometimes you will just find eggshell membrane pieces (and no actual shell fragments)

Phalaropes and dowitchers often leave a large number of eggshell fragments in the nest, often pip fragments are > 5mm for these species.

Appendix F. Nest fate determination and causes of nest failure

SUCCESSFUL

At least one egg hatched.

This is confirmed if chicks are located in the nest. If no chicks are in the nest, then at least one of the two conditions must be met:

1) Hatchlings are observed within 50m of the nest within 2 days of the expected hatch date
2) The nest contained fragments of pipped eggs (typically 1-5 mm in size) and/or eggshell tops/bottoms are observed within 5m of the nest AND it is within 4 days of the estimated hatch date based on:
   - Starring or pipping of eggs on previous visit.
   - Dating from known laying dates or laying dates estimated from float tables.

The following lines of evidence suggesting a successful nest are considered much less reliable. They should be recorded as supporting evidence but not used as one of the two conclusive lines of evidence:

- Parent bird(s) defending nest territory
- Presence of infertile eggs remaining in an inactive nest

FAILURE

Assume a nest has failed if the nest contents are gone prior to 4 days before the calculated hatch/fledge date OR if it is within 4 days of the calculated hatch date and there is not adequate evidence to classify as successful (see above). Only classify a nest as "failure" if you are sure it failed but cannot determine the cause of failure (See below for causes of failure)

UNKNOWN

Put a nest's fate as unknown If you cannot determine the fate of the nest or there is conflicting evidence of hatching, fledging or failure (e.g., shorebird eggs gone > 4 days before hatch date yet egg fragments are found). Nests within 4 days of calculated hatch/fledge dates that do not have adequate evidence to classify as successful and do not have evidence of cause of failure (i.e., signs of predation, weather, trampling, etc.).

UNDETERMINED

Nest occupied but monitoring discontinued (e.g. nest never actively monitored or the nest was still active when field work was concluded).

CAUSES of FAILURE (If there is evidence for cause of failure, classify as follows:

PREDATION

Assume a nest has been depredated if predation was observed or there is at least one line of evidence strongly indicating predation. The following evidence is suggestive of a predation event and should be recorded if observed:

Eggs are missing from nest > 4 days prior to the estimated hatch/fledge date
Attached membranes with blood or yolk
Destroyed nest; egg fragments (larger than 5mm for shorebirds) outside of nest
Remains of adult or chicks within 10 m of the nest
Fox urine or scat at empty nest is confirmatory evidence but must occur with other signs above to indicate predation.

ABANDONMENT	
If eggs are present in a nest longer than the expected hatch/fledge date (>7d) and/or no attendant adults are present on repeated visits (eggs are typically cold or wet). Causes of abandonment may include: Infertile eggs or dead embryos: when an attendant adult is observed incubating eggs long after (at least 7d) the calculated hatch date. Adult mortality: a strong possibility when the nest is abandoned for no apparent reason in the late incubation or nestling stage. Abandoned prior to egg-laying because of nest site disturbance If a nest is suspected of abandonment, reorient eggs (e.g. all large ends toward the center) so you can reconfirm abandonment on the next visit. If the adult is still present, the eggs should be reoriented so that the large ends are now facing out.	
WEATHER A nest that is active up until a severe storm occurs. The nest may be inundated with water or covered with snow. Be sure to check these nests multiple times after a weather event because they often are quite resilient and survive.	
TRAMPLING A nest (and its contents) that is crushed by caribou or other large animal (e.g. Musk oxen, caribou). Supporting evidence would include animal presence in the area, tracks, droppings, and the presence of smashed eggs with contents still present.	
OBSERVER Any nest failure directly attributable to research work. Examples include: 1. The presence of researchers cue predator(s) to nest location (only use this if you witness predation due to your visit – do not assume a nest was depredated due to your visit if you find it empty on the next visit). 2. A nest that is inadvertently destroyed or damaged by a researcher.	

Appendix G. Habitat classification schemes for Network sites:
Western Alaska (Nome and Cape Krusenstern sites)

Adapted from Petterson 1991.

Terrestrial habitat near Krusenstern Lagoon was stratified according to structural components for the purpose of examining shorebird nesting ecology and determining shorebird nesting densities. Habitats were classified as follows:

Mixed Sedge-Dwarf Ericaceous Shrub Tundra (DEST) - Mesic lowland tundra composed of 25-35 percent dwarf (<10 cm.) and low shrub (<20 7 cm.), 30-35 percent non-tussock forming sedges, 10-20 percent tussock- forming sedges, 10-15 percent moss, 5-10 percent lichen, and 10 percent non-sedge graminoid on some plots. These islands and peninsulas are elevated about 1 meter above the slough channels, lagoons, brackish lakes, unvegetated mudflats, and margins of wet sedge meadow and other wet graminoid types that fringe them.

Mixed Shrub~Sedge Tussock Tundra (MST) - Upland tundra with at least 25% shrub cover and co-dominated by tussock-forming sedges. Similar to tussock tundra but with a greater shrub component. Structural composition is 25 percent Salix sp., 30 percent *Eriophorum vaginatum*, 15 percent *Carex* sp., 15 percent *Dryas* sp., and 15 percent moss and lichen. Frost heave and frost boil account for the hummocky character of this terrain.

Willow-Sedge Shrub Tundra (WST) - Upland tundra structurally similar to mixed shrub-sedge tussock tundra, but tufted grasses and sedges more important than tussocks. The shrub component is 25-30%Salix sp. commonly occurring in stringers along frost heaves. Tufted grasses and sedges compose 45 percent. *Eriophorum vaginatum* composes 25 percent and moss compose 5 % willow stringers and hummocks, oriented in long rows in the direction of the slope are a prominent feature in this habitat and are probably indicative of subsurface drainage patterns. Small triangular-shaped thaw puddles line occur in pockets along the willow stringers. Aerial photographs exhibit characteristics of fluvial action perhaps originating with solifluction lobes high on adjacent Dryas slopes. Soil creep retards the development of tussocks in this habitat.

Dryas-Dwarf Shrub Tundra (DT) - *Dryas* sp. is dominant but sparse (10-50 percent) on stony well-drained soils at windswept alpine sites. Graminoids and herbaceous perennials (e.g. Anemone,
Carex, Pedicularis, Poa, Potentilla) are common but not co-dominant. Solifluction and wind erosion are the primary landscape processes in this habitat.

Tussock Tundra (TT) - Dominated by *Eriophorum vaginatum* (60percent), a tussock-forming cottongrass, and *Carex bigelowii* (10percent). Low birch and ericaceous shrubs constitute up to 25 percent of the cover, growing up between tussocks but usually lower than the tops of the sedges. *Ledum palustre* comprises 20 percent while lichens and mosses make up 10 percent of cover.

Wet Sedge Meadow Tundra (WSM) - Dominated by *Carex aguatilis* and *Eriophorum angustifolium* in standing water with occasional hummocks of moss, graminoid and dwarf woody vegetation. These wetlands associated hydrologically with lake lakes. Frost wedging is an important process in this habitat.

Arctic Coastal Plain (Barrow, Ikpikpuk, Prudhoe Bay, Canning, Colville and Mackenzie Delta sites)

From Walker et al. 1980. Geobotanical Atlas of the Prudhoe Bay Region, Alaska

Abbreviated list of Landforms Units
HCP > 0.5m: High-centered polygons, Center-Trough Relief >0.5m
HCP < 0.5m: HCP, C/T Relief ≤ 0.5m
LCP > 0.5m: Low-centered Polygons, Rim-Center Relief >0.5m
LCP < 0.5m: LCP, R/C Relief ≤ 0.5 m
Mixed HCP/LCP: Mixed H & LCP
Frost boil: Frost boil Tundra
Strangmoor: Strangmoor and/or Disjunct Polygon Rims
Hummocky: Hummocky Terrain
Reticulate hummocks: Reticulate-Patterned Ground
Non-patterned: Non-patterned Ground
Alluvial: Alluvial Floodplain
Pingo: Pingo
Unveg Dune: Unvegetated dunes
Veg Dune: Vegetated dunes
Upland Bluff: Upland Bluff

HCP > 0.5m: High-centered polygons, Center-Trough Relief >0.5m
High-centered polygons in the Prudhoe Bay region occur most commonly in a narrow band extending only a few tens of meters inland along streams and the shorelines of former thaw lakes. They are the product of thermokarst and/or thermal erosion in the troughs of former low-centered polygons. These processes become active when drainage of the thaw lakes or change in stream gradient permits better surface and subsurface drainage, resulting in melting of the ice and subsequent deepening of the troughs. The over-deepened (greater than 0.5 m deep and commonly 1.0 m or more) troughs permit slumping of the rim elements and a gradual topographic reversal of the polygon center. This is accompanied by a reduction in surface area of the center. HCP > 0.5m commonly has no other units included with it, although in some circumstances Mixed HCP/LCP (mixed high- and low-centered polygons) may be associated.

HCP < 0.5m: High-centered Polygons, Center-Trough Relief < 0.5m
Certain upland areas, or broadly convex interfluves, have large-diameter (5 to 10 m) polygons whose centers are slightly convex or raised with respect to the adjoining contraction crack or trough. Although the difference in height between the center and the trough may reach 0.5 m it is commonly on the order of 10 to 20 cm and sometimes much less. The central portions of these polygons may be patterned with small (25-50 cm) polygons suggestive of desiccation. The unit may include Reticulate in areas where the desiccation cracks (polygons) are the dominant landform.

LCP > 0.5m: Low-centered Polygons, Rim-Center Relief >0.5m

Low-centered polygons predominate in the unit. In plan the landform consists of polygonal cells with diameters ranging between 5 and 12 m. Each polygon is composed of three elements. The central portion, circular or weakly polygonal in shape and commonly 8 to 10 m in diameter, is surrounded by a rim 0.5 m or slightly more high and up to 1.0 m wide. Centers may contain up to 10 cm of standing water early in the summer but commonly become only moist as the thaw season progresses. The rim of one polygon is separated from that of the adjacent one by a trough whose depth ranges to 50 cm below the rim crest. The troughs mark the position of contraction cracks and ice wedges that extend to depths of 3 to 5 m. Associated landform units that in aggregate compose less than 20% of LCP > 0.5m include LCP < 0.5m, Mixed HCP/LCP, and Frost boil.

LCP < 0.5m: Low-centered Polygons, Rim-Center Relief < 0.5 m
In this extensive unit the polygons tend to be more orthogonal than those of LCP > 0.5m and relief contrast is commonly less than 0.5 m. Basin areas of these polygons are quite wet, with water at or near the surface throughout the thaw period. Landform elements commonly associated with LCP < 0.5m are Stangmoor, LCP > 0.5 m.

Mixed HCP/LCP: Mixed High- and Low-centered Polygons
This landform unit contains high-centered polygons similar to those of HCP > 0.5m and low-centered polygons undergoing topographic reversal and conversion to high-centered polygons. The unit is restricted in area and represents incomplete topographic adjustment to recently decreased base level, for example the drainage of a thaw lake or the relatively recent head ward extension of a tributary drainage.

Frost boil: Frost-Boil Tundra
Frost boil tundra reaches its maximum development in the Prudhoe Bay region along the Putuligayuk River. The landform consists of two elements; the frost boils proper and the vegetated areas between them. The boils consist of active, frost-susceptible mineral materials exposed at the surface or apparently inactive beneath a thin organic mat. The center spacing of individual boils is on the order of 2.5 m; however, areas with much closer spacing are common. Ordinarily the other landform units do not occur within Frostboil, although Reticulate may border it adjacent to the Putuligayuk River.

Strangmoor: Strangmoor and/or Disjunct Polygon Rims
This very wet landscape unit consists of string bogs (strangmoor) in which the hummock ridges (strangs) are less than 0.5 m high and are commonly discontinuous. In extreme cases they are merely an aligned series of hummocks. In some instances the strangs appear oriented normal to the hydrologic gradient and thus serve as a clue to the direction of surface and subsurface water movement. Commonly, however, they grade to low, discontinuous rims of poorly defined, large diameter polygonal cells. The landscape unit is a young terrain feature. The principal associated landscape units are Non-patterned ground and LCP < 0.5m.

Hummock: Hummocky Terrain

This unit is common on slopes greater than 6% on the sides of Pingoes and along stream bluffs. It consists of hummocks whose surface areas range between 25 and 50 cm and which extend to 20 cm or more above the adjacent inter-hummock areas. The unit commonly grades into Unit 9 as slope and angle decreases at the top of the bluff or slope. Thus the hummocks may represent the Reticulate-patterned ground landform rounded and accentuated by erosion, partly thermal and partly related to runoff from the snow banks, which form in these areas.

Reticulate: Reticulate-Patterned Ground
The reticulate landform occurs on the uplands immediately adjacent to active drainage ways and on low linear interfluves or hydrostatic forms underlain by sandy-textured mineral materials. The pattern is an intricate arrangement of slightly convex, small diameter polygons (less than 1.0 m), commonly with a hummocky micro relief (less than 15 cm). As topographic slope steepens toward an adjacent drainage the reticulate landform grades in to the large hummocks of Hummocky Terrain. Away from the drainage and marginal to the wetter tundra elements, especially along the Putuligayuk River, Reticulate-Patterned may include small amounts of Frost boil and HCP < 0.5m.

Non-patterned: Non-patterned Ground
Areas designated as non-patterned ground occur within the basins of recently drained thaw lakes and surrounding shallow, active thaw lakes. Such areas are wet, commonly with standing water throughout the thaw period. They are considered to represent some of the youngest areas in the landscape. Randomly distributed hummocks or short non-aligned hummock ridges, a few tens of centimeters in height, may characterize the surface in some localities. Low relief, low-centered polygons of LCP < 0.5m may compose up to 20% of this unit.

Alluvial: Alluvial Floodplain
This unit contains the river floodplains. Micro topographic expression is commonly lacking or consists of undulating scour pits and abandoned stream channels and bars or the beds of intermittently flowing streams.

Pingo: Pingo
Pingoes are probably the most distinctive and least extensive of the landform units recognized at Prudhoe Bay. In the area covered by this atlas the features are conical to slightly elliptical in form, with basal diameters of several tens to several hundreds of meters. They extend up to 15 m above the surrounding tundra. Their summits may be cracked or may have a central depression due to collapse as the ice core melts. Although the upper portions of the steep side slopes may be severely wind-eroded, the lower portions display the hummock forms of Hummocky Terrain. Pingoes are common features in drained lake basins.

Unveg Dune: Unvegetated (Sand) Dunes
Although they do not appear on the main mapped area in this atlas, sand dunes form a unique landform element in the area just west of the delta of the Sagavanirktok River. Dunes consist of sand ridges 1 to 2 m high extending leeward from stabilized or partly

stabilized coppice-like dunes or dune remnants. Sandy areas between ridges are mostly devoid of vegetation and commonly moist. In some areas polygon terrain similar to LCP < 0.5m can be seen underlying areas recently or thinly covered by the sands. .Note: these are active sand dunes too unstable for vegetation establishment with <30% vegetation cover.

Upland: Upland Bluff
As described from the Canning Delta site; these areas consisted of ridges or low-sloping bluffs that extended 1-2 m above the surrounding tundra. Sites were typically well-drained and consisted mainly of vegetation types (from Walker et al. 1980). U3 (moist *Eriophorum vaginatum, Dryas integrifolia, Tomenthypnum nitens, Thamnolia vermicularis* graminoid meadow) and U4 (moist *Carex aquatilis, Dryas integrifolia, Tomenthypnum nitens, Salix arctica* graminoid meadow).

Veg Dunes: Vegetated Dunes
Inactive and stabilized dunes with ≥30% vegetation cover. Generally dominated by dwarf and upland shrubs such as *Dryas* or *Cassiope* with associated forbs and low-growing *Salix*. Dwarf scrub tundra on upland ridges, stabilized sand dunes and river terraces dominated by *Dryas integrifolia* or *Cassiope tetragona*. Upland *Dryas* sites typically are dry and sandy with deep thaw depths (>1.0 m), common associated species include *Salix glauca, S. reticulata, Arctostaphylos alpina, Arctagrostis latifolia, Thamnolia vermicularis,* and *Cetraria cuculata*. Riverine *Dryas* sites occur on well-drained, sandy river terraces, co-dominant species often include *Equisetum variegatum* and *Salix reticulata,* with *S. lanata richardsonii, Arctostaphylos rubra*, *Oxytropis deflexa, Tomentypnum nitens,* and *Thamnolia vermicularis* as associated species. *Cassiope tetragona* is found on slightly moister sites such as banks of thaw basins, riverbanks, and banks of older, well-stabilized dunes. On intermediate soils *Dryas integrifolia* may be co-dominant. Species found in association with *Cassiope* include *S. phlebophylla, Salix reticulata*, *Vaccinium vitis-idaea*, *Carex bigelowii*, *Hierochloe alpina,* and *Arctagrostis latifolia*. Cryptogams present include crustose lichens, *Hylocomium splendens, Dicranum* sp., *Tomentypnum nitens*, and *Rhytidium rugosum*. All sites have a wide variety of forbs.

Abbreviated list of Vegetation Units

Generally B = DRIEST U = MOISTER M = WET E = EMERGENT

Codes	**Description**
B1	driest, most exposed: sides of pingos, centers of high-centered polygons
B2	less exposed to wind than B1; otherwise similar
B3	tops of frost boils
B14	dry, early-thawing snowbanks with hummocky terrain
U1	polygons rims and aligned strangmoor in acidic tundra; LICHEN
U2	well-drained upland sites; tussocks < 20 cm & dense sedge cover; LICHEN
U3	well-drained upland sites with slightly high-centered polygons; LICHEN

U4	moister upland sites, centers of low polygons or poly rims; NO LICHEN
U6	well-drained snowbanks with *Cassiope tetragona*
U7	late-thawing snowbanks with *Salix* present
U8	stream banks or lake margins with *Salix* and *Carex* present
U9	upland stream banks swept by spring flood
U10	pingo tops, bird mounds, animal dens – graminoid meadow
M1	wet micro sites in acidic tundra with aligned strangmoor; NO LICHEN/*Salix*
M2	wet polygon center and troughs, lake margins; NO LICHEN
M4	very wet polygon centers, drained lakes, lake margins; NO LICHEN
M5	moist stream banks; *Carex* and *Salix* present
E1	very wet: water to about 30 cm; *Carex* present
E2	very wet: water to about 100 cm: *Arctophila* present

More complete descriptions of these vegetation units are available in Walker et al. (1980) but the traits listed above focus on the most relevant features of each unit.

East Bay Habitat types

TABLE 1. Features of the habitats of East Bay, Southampton Island, Nunavut, Canada.

Habitat type	Distinguishing features
Intertidal zone	Intertidal or within splash range of fall storms Dead or dormant moss (organic crust) occurs Bare substrate dominant, living moss and graminoids sparse and patchy
Moss carpet	Pond edges in coastal areas Living moss covers substrate Sparse to moderate abundance of grasses and sedges Numerous herbs, but patchy and sparse
Scrub willow	Drier areas in central and northern portions of plot (0.5–1 km inland) *Salix* spp. abundant Herbs, grasses, sedges, and lichens common Substrate of bare soil and small rocks
Dry heath	Drier areas >1 km inland Ericaceous shrubs dominant; dense cover of mountain avens (*Dryas integrifolia*) Willows and lichens abundant Herbs moderate in richness and abundance Substrate variable: soil, rock, and gravel Relief varies from flat to extremely hummocked
Sedge meadow	Moist areas and pond edges inland Moss covers substrate, few rocks present Sedges and grasses tall (>50 mm) and dense Herbs abundant and diverse Relief varies from flat to highly hummocked
Gravel ridge	Bare gravel dominant Flora sparse and depauperate Visibly raised from surrounding areas Colonized sparsely by mountain avens at low edges

Churchill Habitat types

To be determined

Appendix H. Potential adult and egg predators in the Arctic and lemmings.

Potential nest predator*	**Species code**
AVIAN	
Glaucous Gull *(Larus hyperboreus)*	GLGU
Pomarine Jaeger *(Stercorarius pomarinus)*	POJA
Parasitic Jaeger *(Stercorarius parasiticus)*	PAJA
Long-tailed Jaeger *(Stercorarius longicaudus)*	LTJA
Common Raven *(Corvus corax)*	CORA
Ruddy Turnstone *(Arenaria interpres)*	RUTU
Sandhill Crane *(Grus canadensis)*	SACR
Golden Eagle *(Aquila chrysaetos)*	GOEA
Snowy Owl *(Nyctea scandiaca)*	SNOW
Peregrine Falcon (*Falco peregrinus*)	PEFA
Northern Harrier (*Circus cyaneus*)	NOHA
Rough-legged Hawk (*Buteo lagopus*)	RLHA
MAMMALIAN	
Arctic fox (*Alopex lagopus*)	ARFO
Red fox (*Vulpes vulpes*)	REFO
Brown (Grizzly) bear *(Ursus arctos)*	BRBE
Wolverine (*Gulo gulo*)	WOLV
Polar bear *(Ursus maritimus)*	POBE
Short-tailed weasel *(Mustela erminea)*	STWE
Least weasel *(Mustela nivalis)*	LEWE
Arctic ground squirrel (*Spermophilus parryii*)	AGSQ
Brown lemming (*Lemmus trinucronatus*)	BRLE
Collared lemming (*Dicrostonyx groenlandicus*)	COLE

* Species that have been observed or suspected of depredating nesting birds, eggs, or young at tundra-nesting bird nests. We do not record Sabine's Gulls and Arctic Terns as potential nest predators because they are only believed to very rarely depredate nests.

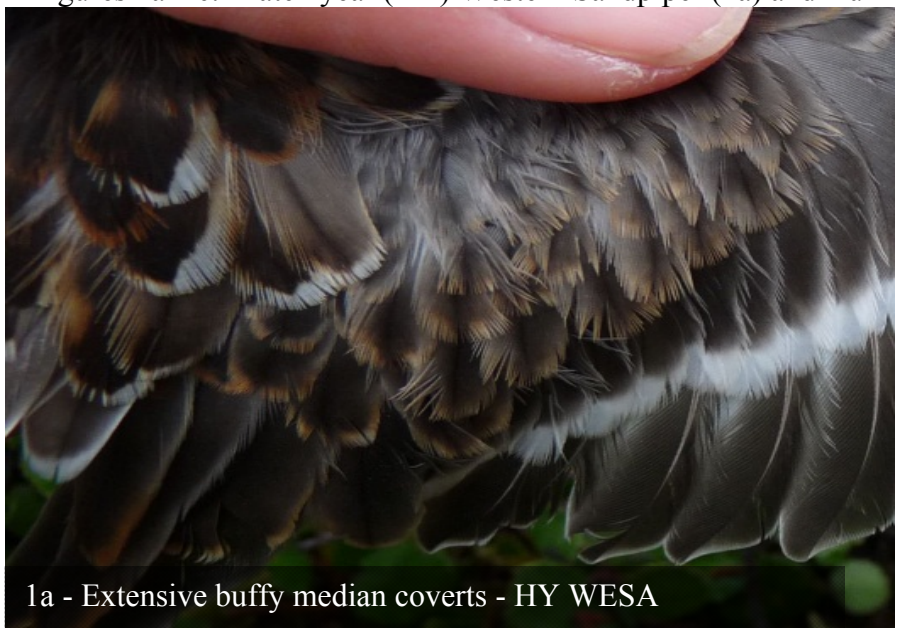
Appendix I. Focal species ageing guide by species

The information presented below is a reference guide for ageing Calidris sandpipers. For more extensive resources about molt and age classification see list of additional resources at the end of this guide. We advise using this reference, in addition to training with someone who has extensive banding experience and ageing. Please take pictures of **captured bird wings according to instructions in banding section. This creates a permanent record of the banded bird and will be used to verify and assess age in a consistent manner**. This guide was developed by Samantha Franks and River Gates.

Plumage criteria that can be used to age Calidris sandpipers: Semipalmated and Western Sandpipers and Dunlin.

	Age		
Plumage criteria	**HY-juvenile**	**SY-yearling**	**ASY-adult >1 year old**
Inner median coverts	Extensive buffy or reddish edging on all coverts, dark centers (WESA: Fig 1a, DUNL: Fig 1b). Calidris HY plumages are all very similar in appearance.	All heavily worn, a few with slight buffy or reddish edges (SESA: Fig 2a, WESA: 3a & b, DUNL: 4a). Inner coverts may have darker centers with less distinction in color between shaft and feather center	Slightly worn, No distinct buffy or reddish-edged coverts apparent, coverts are white-edged and (SESA: Fig 5a, WESA: Fig 5c, DUNL: Fig 6c) Inner coverts may have lighter centers with more distinction in color between shaft and feather center
Primary and greater coverts	Fresh white tips, smooth edges	Extremely worn white tips, frayed edges	Moderate wear of white tips, more smoothly edged
Flight feather wear	Very fresh, dark grey feathers with no wear at tips and little fraying along edges (WESA: Fig 1c)	Lots of wear at tips of outer primaries, frayed primary edges (SESA: Fig 2b, WESA: Fig 3c)	Minimal to moderate wear at tips of outer primaries, more smoothly edged primaries (SESA: Fig 5b, WESA: Fig 5d)
Primary molt	n/a	May exhibit partial primary molt, where outer primaries (~p6-10) have been replaced (SESA: Fig 2c)	n/a

Figures 1a – c. Hatch year (HY) Western Sandpiper (1a) and Dunlin both ~14 days old (1b). Photos by S. Franks and J. Choi



1a - Extensive buffy median coverts - HY WESA

1b - Extensive buffy median coverts – HY DUNL

1c - Fresh primary tips – HY WESA

Figure 2a-c. A second year (SY) Semipalmated Sandpiper.

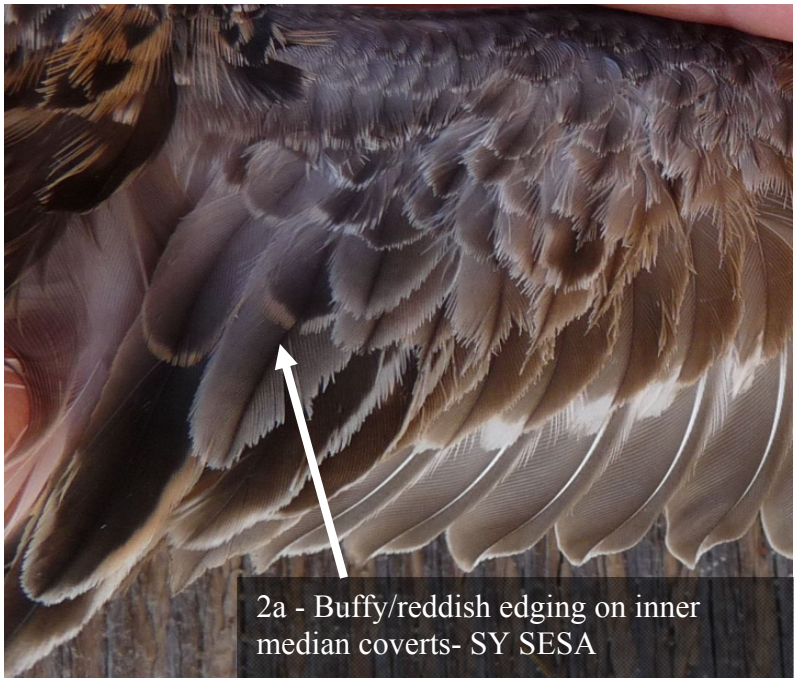
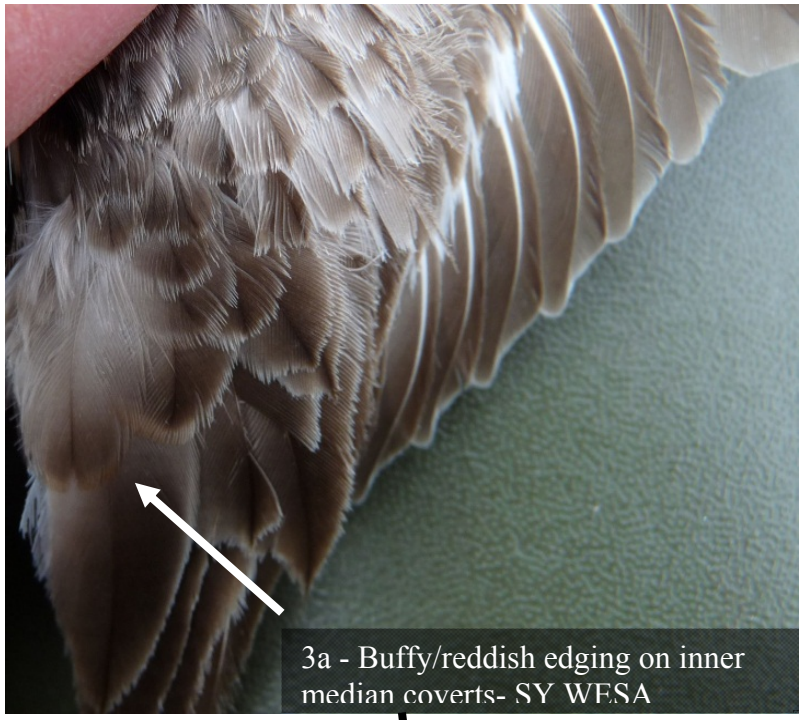


Figure 3a-b. A second year (SY) Western Sandpiper.



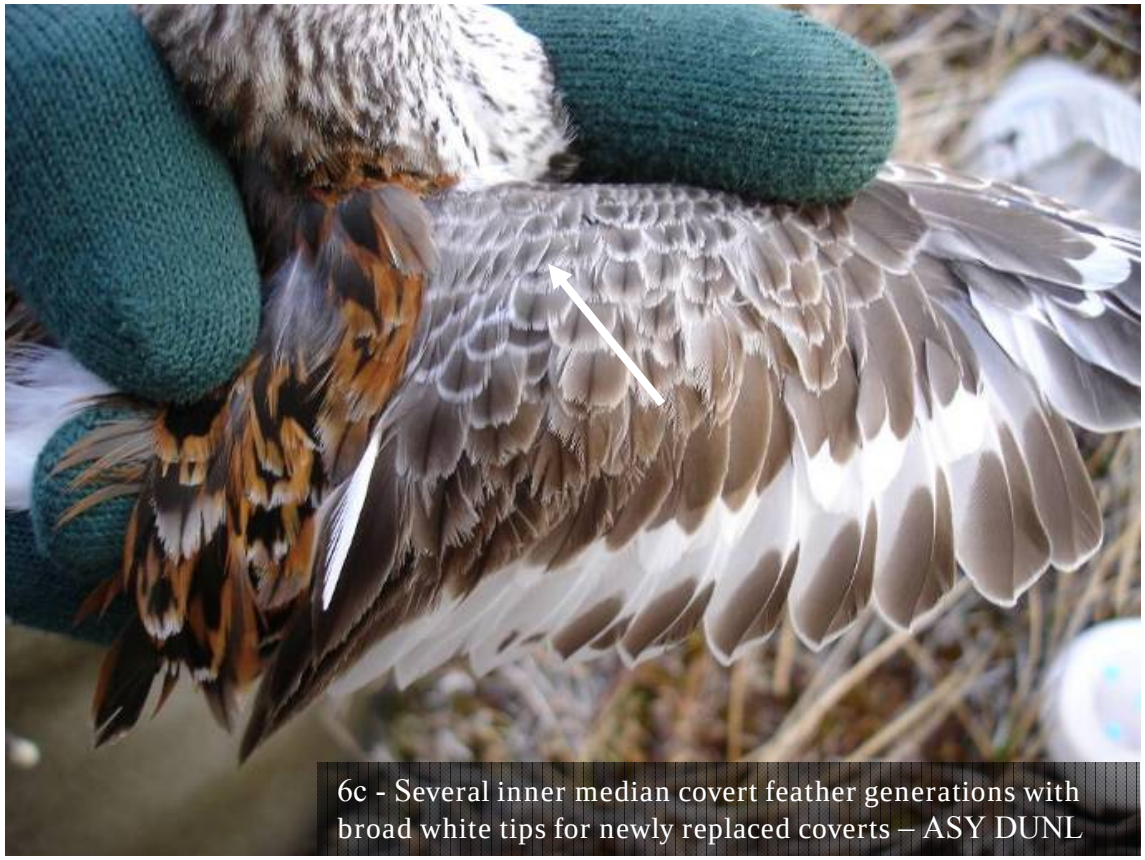
3a - Buffy/reddish edging on inner median coverts- SY WESA

3b - Buffy/reddish edging on inner median coverts- SY WESA

3c - Extremely worn primary tips- SY WESA

Figure 4a: A second year (SY) Dunlin. Photos by J. Choi/USFWS.

Figure 5a-b. An after second year (ASY) Semipalmated Sandpiper, and c – d an after second year (ASY) Western Sandpiper.



6c - Several inner median covert feather generations with broad white tips for newly replaced coverts – ASY DUNL

Additional resources:

Pyle, P. 2008. Identification Guide to North American Birds. Part II. Slate Creek Press, Point Reyes Station, CA
Prater, A.J., Marchant, J.H. and J. Vuorinen 1977. Guide to identification and ageing of Holarctic waders.
Gratto-Trevor, C. L. 2004. The North American Bander's Manual for banding shorebirds. NABC

Appendix J. Sinking and floating egg float tables for select shorebird species

SINKING EGGS DTH = "days to hatch"									
AMGP		BBPL		BBSA		DUNL		LBDO	
Angle	**DTH**	**Angle**	**DTH**	**Angle**	**DTH**	**Angle**	**DTH**	**Angle**	**DTH**
21	25.8	21	25.3	21	27.8	21	21.3	21	22.4
25	23.4	25	23.1	25	24.2	25	19.8	25	20.0
30	22.2	30	22.1	30	22.6	30	19.1	30	18.9
35	21.5	35	21.5	35	21.5	35	18.6	35	18.2
40	20.9	40	21.0	40	20.7	40	18.3	40	17.6
45	20.4	45	20.5	45	20.0	45	18.0	45	17.2
50	20.0	50	20.1	50	19.3	50	17.7	50	16.7
55	19.6	55	19.8	55	18.7	55	17.5	55	16.3
60	19.1	60	19.4	60	18.1	60	17.2	60	15.9
65	18.7	65	19.0	65	17.5	65	16.9	65	15.5
70	18.2	70	18.6	70	16.8	70	16.6	70	15.0
75	17.6	75	18.1	75	16.0	75	16.3	75	14.4
80	16.9	80	17.4	80	14.9	80	15.8	80	13.7
85	15.8	85	16.4	85	13.2	85	15.1	85	12.6
89	13.3	89	14.2	89	9.7	89	13.6	89	10.2
PESA		RNPH		REPH		RUTU		SESA	
Angle	**DTH**	**Angle**	**DTH**	**Angle**	**DTH**	**Angle**	**DTH**	**Angle**	**DTH**
21	20.9	21	21.2	21	18.1	21	21.5	21	20.2
25	19.6	25	19.4	25	17.0	25	19.4	25	18.6
30	18.9	30	18.5	30	16.5	30	18.4	30	17.9
35	18.5	35	18.0	35	16.1	35	17.8	35	17.5
40	18.2	40	17.6	40	15.9	40	17.3	40	17.1
45	17.9	45	17.2	45	15.6	45	16.9	45	16.8
50	17.7	50	16.9	50	15.4	50	16.5	50	16.5
55	17.5	55	16.6	55	15.2	55	16.2	55	16.2
60	17.2	60	16.3	60	15.0	60	15.8	60	16.0
65	17.0	65	16.0	65	14.8	65	15.4	65	15.7
70	16.7	70	15.6	70	14.6	70	15.0	70	15.4
75	16.4	75	15.2	75	14.3	75	14.6	75	15.0
80	16.0	80	14.7	80	14.0	80	13.9	80	14.6
85	15.4	85	13.8	85	13.5	85	13.0	85	13.9
89	14.0	89	12.0	89	12.3	89	10.9	89	12.3

STSA		Other shorebirds	
Angle	DTH	Angle	% of incubation complete
21	17.6	21	0.016
25	16.9	25	0.075
30	16.5	30	0.118
35	16.3	35	0.145
40	16.2	40	0.166
45	16.0	45	0.184
50	15.9	50	0.200
55	15.8	55	0.216
60	15.7	60	0.232
65	15.5	65	0.248
70	15.4	70	0.266
75	15.2	75	0.287
80	15.0	80	0.314
85	14.7	85	0.356
89	14.0	89	0.448

FLOATING EGGS DTH = "days to hatch"

AMGP			BBPL			BBSA			DUNL		
Angle	height	DTH	Angle	height	DTH	Angle	height	DTH	Angle	height	DTH
90	0	14.0	90	0	15.8	90	0	15.9	90	0	12.8
90	1	12.6	90	1	12.8	90	1	13.7	90	1	11.2
90	2	11.2	90	2	9.9	90	2	11.5	90	2	9.5
90	3	9.8	90	3	7.0	90	3	9.4	90	3	7.9
90	4	8.4	90	4	4.0	90	4	7.2	90	4	6.3
90	5	7.1	90	5	1.1	90	5	5.0	90	5	4.7
90	6	5.7	90	6	1.9	90	6	2.8	90	6	3.1
90	7	4.3				90	7	0.6	90	7	1.5
90	8	2.9	80	0	16.0				90	8	0.1
			80	1	13.1	80	0	15.1			
80	0	13.6	80	2	10.1	80	1	12.9	80	0	11.7
80	1	12.2	80	3	7.2	80	2	10.7	80	1	10.1
80	2	10.8	80	4	4.2	80	3	8.5	80	2	8.5
80	3	9.4	80	5	1.3	80	4	6.3	80	3	6.9
80	4	8.0	80	6	1.6	80	5	4.1	80	4	5.3

80	5	6.6				80	6	1.9	80	5	3.7
80	6	5.2	70	0	16.2	80	7	0.3	80	6	2.1
80	7	3.9	70	1	13.3				80	7	0.5
80	8	2.5	70	2	10.3	70	0	14.2			
80	9	1.1	70	3	7.4	70	1	12.0	70	0	10.7
80	10	0.3	70	4	4.5	70	2	9.8	70	1	9.1
			70	5	1.5	70	3	7.7	70	2	7.5
70	0	13.2	70	6	1.4	70	4	5.5	70	3	5.9
70	1	11.8				70	5	3.3	70	4	4.3
70	2	10.4				70	6	1.1	70	5	2.7
70	3	9.0							70	6	1.1
70	4	7.6							70	7	0.6
70	5	6.2									
70	6	4.8									
70	7	3.4									
70	8	2.1									
70	9	0.7									

LBDO			PESA			RNPH			REPH		
Angle	height	DTH	Angle	height	DTH	Angle	height	DTH	Angle	height	DTH
90	0	15.4	90	0	13.0	90	0	11.6	90	0	11.1
90	1	14.4	90	1	11.8	90	1	9.1	90	1	9.8
90	2	13.4	90	2	10.5	90	2	6.6	90	2	8.6
90	3	12.5	90	3	9.3	90	3	4.0	90	3	7.3
90	4	11.5	90	4	8.1	90	4	1.5	90	4	6.1
90	5	10.5	90	5	6.8				90	5	4.8
						80	0	11.7			
80	0	12.0	80	0	12.4	80	1	9.2	80	0	10.1
80	1	11.0	80	1	11.1	80	2	6.7	80	1	8.9
80	2	10.0	80	2	9.9	80	3	4.2	80	2	7.6
80	3	9.0	80	3	8.7	80	4	1.7	80	3	6.4
80	4	8.0	80	4	7.4	80	5	0.8	80	4	5.1
80	5	7.0	80	5	6.2				80	5	3.8
80	6	6.0	80	6	5.0	70	0	11.9	80	6	2.6
80	7	5.0	80	7	3.7	70	1	9.4	80	7	1.3
80	8	4.0	80	8	2.5	70	2	6.9	80	8	0.1
80	9	3.0	80	9	1.3	70	3	4.4			
80	10	2.0	80	10	0.0	70	4	1.9	70	0	9.2
						70	5	0.7	70	1	7.9
70	0	8.6	70	0	11.7				70	2	6.7
70	1	7.6	70	1	10.5				70	3	5.4
70	2	6.6	70	2	9.3				70	4	4.2
70	3	5.6	70	3	8.0				70	5	2.9

70	4	4.6	70	4	6.8				70	6	1.6
70	5	3.6	70	5	5.6				70	7	0.4
70	6	2.6	70	6	4.3				70	8	0.9
70	7	1.6	70	7	3.1						
70	8	0.6	70	8	1.9						

FLOATING EGGS DTH = "days to hatch"

RUTU			SESA			STSA			Other Shorebird*		
Angle	height	DTH	Angle	height	DTH	Angle	height	DTH	Angle	height	% of incubation complete
90	0	13.4	90	0	11.5	90	0	11.2	90	0	0.42
90	1	10.0	90	1	10.2	90	1	10.4	90	1	0.48
90	2	6.7	90	2	8.9	90	2	9.5	90	2	0.55
90	3	3.3	90	3	7.5	90	3	8.7	90	3	0.62
90	4	0.0	90	4	6.2	90	4	7.8	90	4	0.68
			90	5	4.9	90	5	7.0	90	5	0.75
80	0	13.8	90	6	3.5				90	6	0.82
80	1	10.4	90	7	2.2	80	0	10.5	90	7	0.89
80	2	7.0	90	8	0.9	80	1	9.7	90	8	0.95
80	3	3.7				80	2	8.8			
80	4	0.3	80	0	10.8	80	3	8.0	80	0	0.46
			80	1	9.5	80	4	7.1	80	1	0.53
70	0	14.1	80	2	8.1	80	5	6.3	80	2	0.59
70	1	10.7	80	3	6.8	80	6	5.4	80	3	0.66
70	2	7.4	80	4	5.5	80	7	4.6	80	4	0.73
70	3	4.0	80	5	4.1	80	8	3.7	80	5	0.79
70	4	0.6	80	6	2.8	80	9	2.9	80	6	0.86
			80	7	1.5	80	10	2.0	80	7	0.93
			80	8	0.1				80	8	0.99
						70	0	9.8			
			70	0	10.0	70	1	9.0	70	0	0.50
			70	1	8.7	70	2	8.1	70	1	0.57
			70	2	7.4	70	3	7.3	70	2	0.63
			70	3	6.1	70	4	6.4	70	3	0.70
			70	4	4.7	70	5	5.6	70	4	0.77
			70	5	3.4	70	6	4.7	70	5	0.84
			70	6	2.1	70	7	3.8	70	6	0.90
			70	7	0.7	70	8	3.0	70	7	0.97
						70	9	2.1			
						70	10	1.3			

* To calculate the "% of incubation complete" for species for which we do not have species-specific float tables, use the "other shorebird" float table.

For example: You discover a Bar-tailed Godwit nest and float the eggs. The eggs are floating at the water surface at an angle of 80° and the egg is exposed 2 mm above the water line.

% of incubation complete = 0.59 (from "other shorebird" table) x 21 (mean incubation length for BTGO) = 12.4 days old. The eggs will hatch in approximately (21 – 12.4) = 8.6 days.

Appendix K. Predator point counts 2010

These data will be used to index the abundance and activity level of potential predators at each study site and will be used in analyses to determine the relationship between predator numbers, and adult and nest survivorship. We will conduct predator surveys **once a week for intensive sites or a minimum of three times per season (early, mid and late) for minimum sites** from the beginning of nesting to the end of hatch (or through brood-rearing for situations where brood survival is being monitored). The following protocol will be used during each survey day (adapted from Leibiezit 2009).

- Conduct **a 10-minute "point count**" at a minimum of **10** different locations within the study area. If you have plots, you can conduct 10-minute counts within each plot (spaced at least 200m apart). The goal is to collect enough observations of dominant predators to be able to use the DISTANCE program (ideally 50 observations of each species across the entire season). Record GPS locations for these points on the meta database for the camp.
- To identify predators during point counts use binoculars (8 x 42 or 10 x 40).
- Timed surveys will be conducted **weekly (intensive approach) or 3 times (less intensive)** during the season: 1st: during early nesting (5 June to 20 June), 2nd: mid-incubation: (21 June to 5 July), 3rd: late-nesting (6 July to 25 July).

2. Wait at least **10** minutes (longer if possible) between individual point counts and conduct the consecutive counts at a stake at least **200m** from the previous one.
3. During the "point count" the observer should scan the surrounding terrain for any visual or auditory detection of potential nest predators and record any predators seen within a **300**m radius of the point count stake you are at.
4. Record the predator **species** and the **distance** to the predator upon **the initial sighting**.
5. Estimate the distance to the predator within the **nearest 5-10m**. If the predator is within 50-100m of you, try to estimate its distance to the **nearest meter**.
6. To estimate distances use rangefinders, pace the distance on foot, or use adjacent centerline plot markers as a guide (they are 50m from each other). You will not be able to get a distance on a moving predator with the rangefinder, instead, obtain a distance on a patch of tundra that is below a flying bird or where a moving fox was seen.

Additional guidelines:

1. Do **not** perform predator counts during rope drag visits or at times when more than one observer is on a study plot.
2. Take effort not to re-count the same individual predators.
3. Do not wait at a point for a "settling period" before starting a count. Start the count right away.
4. Fill out a nest record form for all avian and mammalian predator nests within the study area, especially Snowy Owl and Pomarine Jaegers. Note contents of nest (e.g., number of eggs or chicks) and adult behavior.

Predator Point Count Data recorded
(see ASDN V1 protocol)

12. Observer name (first initial and full last name)

13. Plot ID
14. Date – dd-mm-yr
15. Time of arrival on plot/search area
16. Time of departure from plot/search area
17. Record start and end time for each timed census count at each survey point; record GPS location of these predator point counts.
18. Write species (including number of individuals) detected within 300m of the survey point. See **Appendix H** for list of potential predators and their codes
19. Record perpendicular distance (meter) to predator.
20. Comment on any important observations:
    a. Hunting behavior observed
    b. Interactions among predators or with other species
    c. Observation of nest predation
    d. Possibility you already recorded this same predator during this survey.
    e. Plumage variations that may aid in identifying individuals (e.g. dark-morph versus light-morph jaegers)
21. Observation of nesting predators should be recorded – if a new nest is detected be sure to fill out a nest card so that the presence of this nest (and its GPS location) is present for later predator abundance estimates.

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