Arctic Shorebird Demographic Network Breeding Camp Protocol

Version 1 – May 2010

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Cover photo depicts three of the Network Focal Species including Dunlin, Semipalmated Sandpiper and Red Phalarope. 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 Demographic 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 near Nome, Cape Krusenstern, Barrow, the Ikpikpuk River, Prudhoe Bay, and the Canning River in Alaska, as well as at the Mackenzie River Delta, East Bay, and Churchill in Canada. Plans are underway to add additional sites in 2011, including two Alaska sites, Hooper Bay in the Yukon Delta National Wildlife Refuge, and Kanuti NWR; and one site in Canada at Coats Island.

Figure 1. Arctic Shorebird Demographic Network study sites across the North American Arctic for 2010.

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 Table 2.

Table 1. Network focal 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 Krusenstern	Barrow	Canning River	Ikpikpuk	Nome	Prudhoe Bay	Mackenzie Delta	East Bay	Churchill
SESA	1	1	1	1	1	1	2	2	2
DUNL	1	1	1	1	1	2	0	2	2
WESA	1	2	0	2	1	2	0	0	0
RNPH	2	1	1	1	1	1	2	0	2
REPH	2	1	1	1	1	1	1	1	0
PESA	2	2	2	2	2	2	1	0	0
SEPL	0	2	2	0	2	0	0	2	1
WHIM	2	0	0	0	0	0	1	0	2

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 point counts, 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 transects, 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)	Methodology	Metric (s)	Workforce required for minimum 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. Resight individuals multiple times within season. 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	Methodology	Metric(s)	Workforce required for minimum	Workforce required for intensive
Ecological monitoring					
1	**Predator index:** Document relative abundance of avian and mammalian predators	Point counts	Abundance estimates or minimum counts.	Min point counts at 10 locations -- 3 times/season (early, middle and late)	Min point counts at 10 locations /week
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, middle, 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 cover:** Monitor snow melt progression	Plot surveys	% snow cover at fixed interval	2 hrs/search area every other day, first 5 to 15 days of season, depending on snow melt	2 hrs/ plot every other day, first 5 to 15 days of season, depending on snow melt
1	**Food resources:** Document seasonal change in insect emergence and abundance. Terrestrial (wet and dry locations) and aquatic (surface sampling).	Modified pitfall traps, aquatic passive 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 of 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, precipitation, wind speed and 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 2- 3 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 diagonal 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								
Banding								
Food resources								
Predators								
Lemmings								
Snow								

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 Global Positioning System (GPS) unit. To ease future GIS applications, we insist that 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 meta database that accompanies each camp (see dataforms Network site establishment form).

Be sure you have the correct time adjusted on all GPS units (e.g., -9 hours from Greenwich time for Alaska, Canada times vary across sites, project leaders can go to this website: http://wwp.greenwichmeantime.com/time-zone/north-america/canada/time-zones.htm to obtain their location's Greenwich time zone.

ADULT SURVIVAL CAPTURE AND RESIGHT METHODS

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 4). 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 4. 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 or above 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

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.

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 can not do this with the solder gun, then use glue to seal them.**

Geolocators will also be attached to birds using a "logger flag" that is attached to the UPPER RIGHT leg with a shoehorn applicator and glued shut with Superglue (do not use soldering guns for fear of destroying the geolocator resistor with heat). Placing the geolocator in this position will ensure the light sensor is directed away from the bird. See more detailed instructions about attaching geolocators in the Network side-projects protocols.

Data recorded with each bird

Banding Datasheet (see page 76)
See banding sheet in the Data forms below:

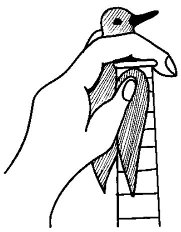
1. Bander (first name initial and last name e.g. loring for Lewis Oring)
2. Date (dd-mm-yy)
3. Time (24 hr)
4. Nest ID (unique code of spp and nest number)
5. Plot identification (unique plot code, alpha-numeric)
6. In or off plot (Was the bird captured on or off the plot)
7. How captured (bownet, mist 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 in Longmire (Y/N) Amount of blood (in micro liters), type of capillary tube (EDTA/Plain/Hep)
25. Blood for MethyHg (Y/N, % of tube)
26. AI swab (Y/N, sample #)
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. Instrument identification number (e.g. transmitter frequency or geolocator tag code)
34. If geolocator, record exact date and time applied to the bird (VERY IMPORTANT).
35. GPS location of nest

Morphological measurements

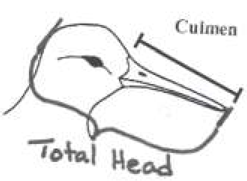
Figures from Prater et al 1977 or Gratto-Trevor 2004, Photos B. Lewis/USFWS

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.

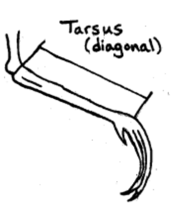


Exposed culmen: measured with calipers (to the nearest 0.1 mm) from the edge of feathering to the tip of the bill. Since bills of most shorebirds are very sensitive, hold bill lightly with the fingers, with the calipers resting on one's fingers and not the bird's bill. The calipers must remain **perpendicular** 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).



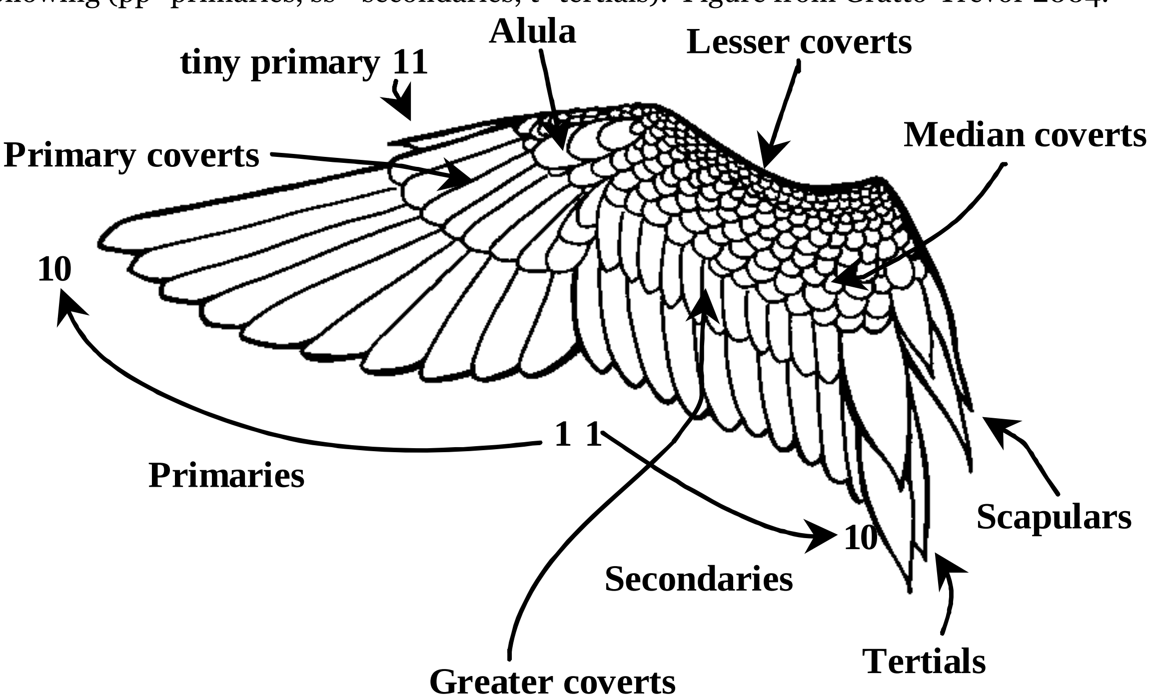
Mass: measured with a hanging Pesola scale or a digital scale (to nearest 0.1 g). If using a Pesola scale the scale needs to be held by the top ring or hook and allowed to dangle freely, while being protected from the wind. Place the bird in a weighed cloth bag, or plastic cone with the bill protruding 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.**

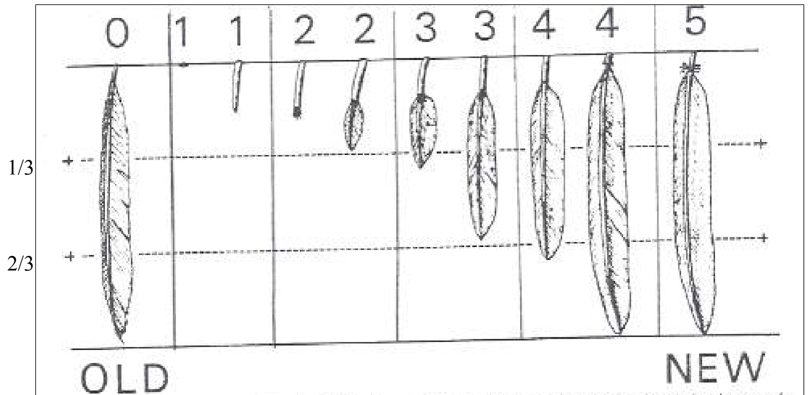
Feather tracts and individual feather names are standardized and abbreviations are used on datasheets. The below figure illustrates the individual names of each flight feather and their standard abbreviations are as following (pp=primaries, ss= secondaries, t=tertials). Figure from Gratto-Trevor 2004.



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 two focal species (e.g. DUNL, SESA).

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

Collection of sample materials from birds

This Network offers an unprecedented opportunity to collect samples from a large number of shorebirds over a large geographic area. These samples may fulfill existing Network Project demands and also be a valuable reservoir for future studies. Thus, we are recommending that all captured birds have a blood and feather sample collected. For all sample types, place the bird's USGS metal band number, species AND it's nest number on each sampling vial or envelope, using a permanent marker such as a Sharpie. Having these three items will allow accidental mistakes in the way samples were numbered to be resolved. Also, include at a minimum collection location, and collection date. You can also include sex and age if room on the vial allows. Materials for blood and feathers sample collection will be provided to all camps willing to participate.

Blood collection for genetic analysis or DNA only

Blood samples are collected from the basilic vein (also known as the brachial vein) under the wing of adult shorebirds (Figure 4). 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 run down the feather and be difficult to collect with the capillary tube. Then puncture the vein 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. 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.

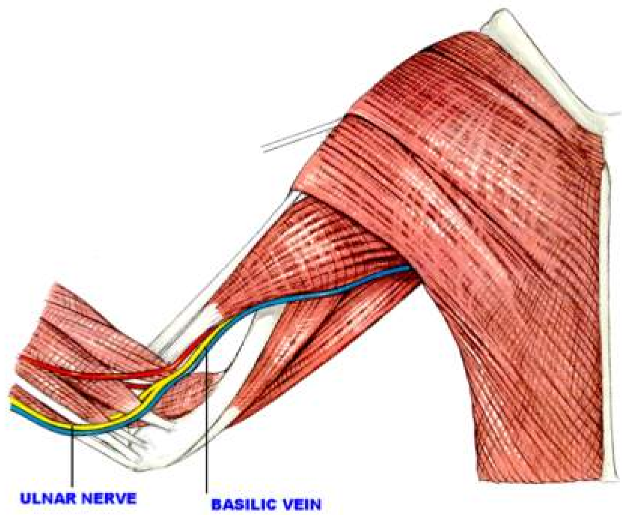


Figure 4. Drawing a blood sample using a heparanized capillary tube from a Dunlin. Illustration of basilic vein for blood withdrawal and the ulnar nerve. (Photo: B. Lewis/USFWS Figure: Evers 2008)

Note the size of the capillary tube. For basic genetic analysis (population structure, paternity), we would like to get 50 micro liters 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 Longmire buffer solution and stored at room temperature. Given the recent avian influenza issues, use a plastic transfer pipette to force air through the capillary tubes and remove the blood. If the blood clots inside the capillary tube (or you have only a very small amount of blood), break off the tube inside the buffer vial and leave it there. 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.

Note: Sharpie pens should not be used if ethanol is used as a preservative as any spillage of the ethanol will remove the sharpie markings. In this case, use special ethanol proof marking pens that will be provided.

Feather sample collection for stable isotope analysis

For all species (other than Dunlin 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).

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 Project 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 where it molted last year by collecting an old primary. 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.

W**estern Sandpiper only**: We will pull the entire inner most first primary feather from each side of the body, making a total of two feathers collected from each bird.

Photo documentation of all captured birds

In order to collect standardized age data and to verify color band codes we suggest that each individual adult captured have pictures taken of their full wing and legs once the bird has been banded. Ages will be assigned to birds in the field and will be verified by the pictures. At least 3 pictures should be taken, 1) data sheet that includes the band number, date bird was banded and color combo (Figure 5a), 2) picture of the color bands and alpha-numeric coded flag (Fig.5b) and 3) picture(s) of the bird's wing (Fig.5 c and d). For species that have longer wings (i.e. DUNL) two pictures of the wings should be taken. It is very important to take a clear photo of the **all inner wing coverts and flight feathers**. Including a dark background improves the standard of the images. These photos should be labeled by unique band number and species code and indicate what the photo is (e.g. 230155644dunl_data.jpg 230155644dunl_combo.jpg 230155644dunl_innerwing.jpg, 230155644dunl_outerwing.jpg).


Fig.5a


Fig.5b


Fig. 5c


Fig. 5d

Figures 5 a – d. Series of photos taken to document adult marking including color banding and ageing characteristics of Dunlin. Photos: R. Gates/USFWS

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 of 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 can not 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 3, 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.

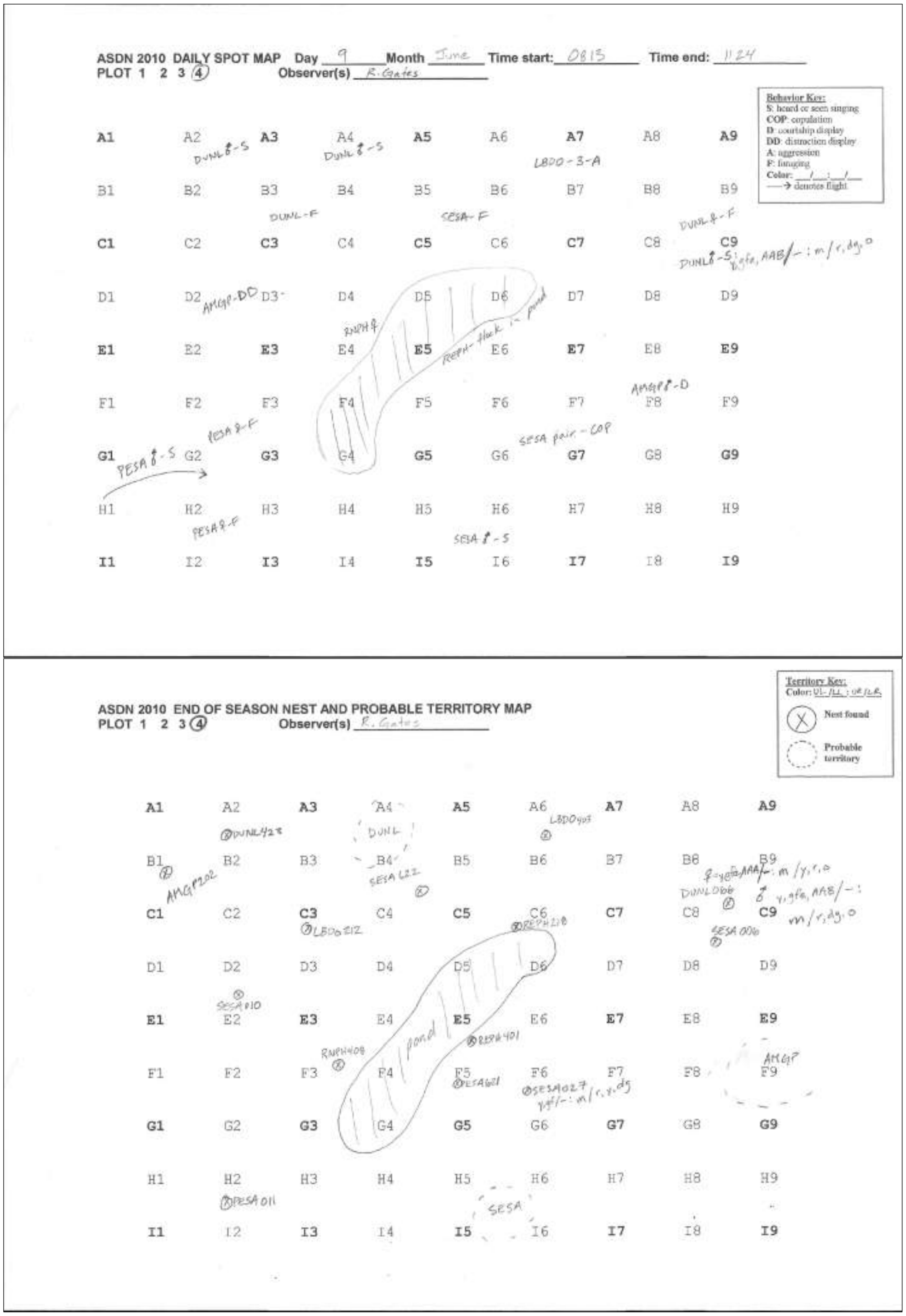


Figure 3: Example of a Daily Spot Map for taking behavioral notes while conducting intensive nest searches (top panel) and the End of the Season Nest and Probable Territory Map (bottom panel).

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 4). 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.

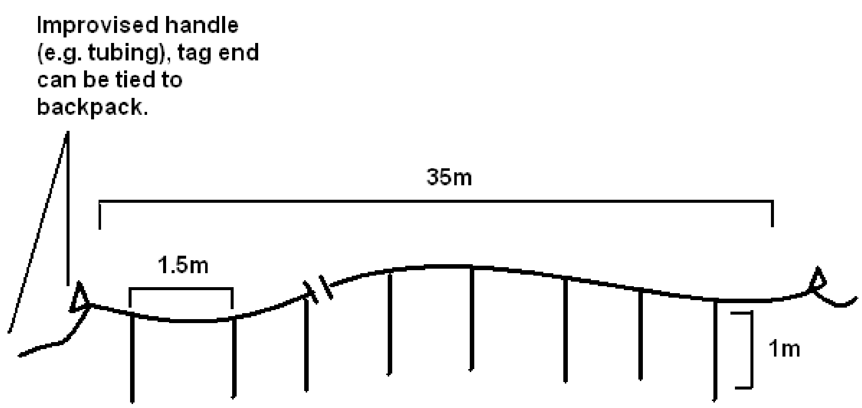


Figure 4. Construction of rope drag for intensive shorebird monitoring plots (Figure: Smith et al 2008)

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

Data recorded at each nest

Nest Record Forms: see page 88

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. Last 2 digits of year + 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. Office use estimation of important nest dates such as initiation date, start of incubation, and estimated hatch date

Nest monitoring data to record:

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

After nest fate has occurred, record:

24. Percent nest concealment: use ocular tube and estimate to the nearest 10%
25. 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.
26. Nest Fate determination:
    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 note 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.

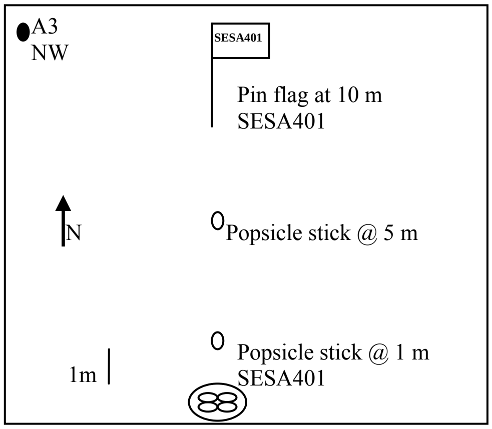


Figure 4: Schematic of marking nest to relocate for nest monitoring

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., 10SESA401). 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 can not 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 lines 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 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. <u>Landform type:</u> 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 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 geographically wide 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 page 75 for daily camp journal and vertebrate species list page 77.
Adapted from Barrow Protocols and ArcticWOLVES (Smith et al. 2008).

Daily camp journal

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.

Food Resources

Objective

The distribution, phenology and abundance of insect adults and larvae will be measured in terrestrial and aquatic habitats. We will identify and enumerate samples to acquire estimates of biomass, biodiversity, peak 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 food resources will be collected from mesic and dry terrestrial habitats using modified Malaise pitfall traps (Lamarre and Bolduc 2010). Terrestrial sampling will commence when the habitats are mostly snow and ice free. The terrestrial pitfall trap methodology is similar to those used by ArcticWOLVES and Bylot Island (Bety, J. pers. comm.). **Aquatic food resources** will be measured with surface associated activity traps placed at the surface of the water in local water bodies where shorebirds have been observed feeding. Sample collection and storage should take approximately 30 -45 minutes for one person in the field and 15 minutes in the base camp for each sampling period.

Terrestrial monitoring

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. frost boil tundra or dry lowland sites), and one transect line in more mesic (wet) habitats (e.g. pond edges and low-centered polygons).

Aquatic monitoring

It is recommended that each site identify **5 ponds with high bird use** to conduct aquatic invertebrate sampling. We will deploy 5 surface associated activity traps in a horizontal position to capture surface and shallow sub-surface aquatic invertebrates (Hanson et al 2000, Muscha et al. 2001).

Collection frequency and materials

Ten terrestrial traps (5 mesic and 5 dry) will be checked and emptied every **3 days** until the season is complete (e.g. end of hatch or brood –rearing). For a 6 week camp this will result in 14 sample collection days and generate 140 unique terrestrial samples. Aquatic traps will be collected every **3 days** and generate in 12 unique sample collection days. All samples will be collected in individually labeled jars in the field and stored in full strength isopropanol (e.g. isopropyl alcohol, rubbing alcohol) in the office. **Some sites will increase the frequency of collection to daily during peak emergence to better understand this phenomenon.** 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).

Collectors should use caution to avoid impacting the micro site (within 1 meter) where the terrestrial and aquatic traps are set while collecting the samples. Care should be given to avoid compacting the soil around the trap or disturbing the shore and near-shore habitats during the aquatic collection.

Data collection and sample storage

Samples will be stored separately for each trap (regardless of type) in whirl packs filled with preservative solution (full strength isopropanol). Do not combine across traps else you will loose your measure of variability. Each whirl pack should have a Rite-in-the-Rain label inside that indicates date, location, habitat type (terrestrial and aquatic), and if terrestrial whether it is a wet or dry site and the replicate number (e.g. Terre Mesic 1, Aquatic 1). Also record the observer who collected the sample and note any lost or damaged samples. This information should be written in pencil on Rite–in-the-Rain paper so the data are not lost once placed in preservative.

In 2010, we will not measure invertebrates in the field but rather ship them back to the USFWS in Anchorage for later enumeration. We plan to dry and weight the majority of samples to estimate biomass, and also ship a subsample of the samples to different laboratories so the insects can be identified and enumerated.

Supplies required for invertebrate sampling:

<u>Provided</u>
Coffee filters
Triton soap
Fine mesh fish nets
Field storage containers (5 for aquatic traps and 10 for terrestrial)
Whirl Paks
All materials for terrestrial pit fall traps (e.g. pvc pipes and glue, window screen netting, zipties)

<u>Supplied by each camp leader</u>
5 gallons (18 liters) of propylene glycol (non-toxic anti-freeze)
1 ½ gallons (~6 liters) of full strength isopropanol (e.g. rubbing alcohol)
15 2 Liter soda pop bottles with the lids
Healthy does of enthusiasm for installing the traps and collecting the samples
20 pieces of rebar (1 ft long and ½ inch diameter)
20 10-inch nails for anchoring trap down
Duct tape (big roll should be plenty)
Scissors
Heavy duty-hole punch, drill with 3/16-inch bit or awl (from a pocket knife/tool)
Ruler (metric)

Building and deploying terrestrial traps

A step by step method to make Bylot Arthropod Traps (design to be further tested at Bylot in 2010) with slight modifications in how to anchor the screens for the ASDN

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

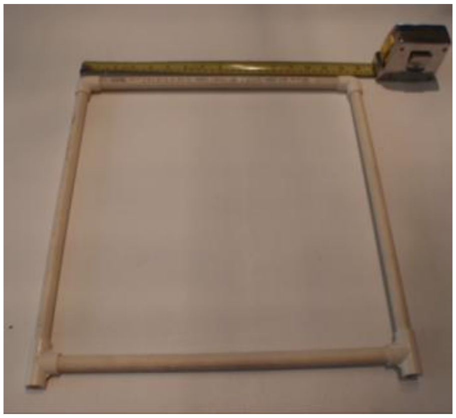
Material needed per trap (40 cm X 40 cm)

- 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)

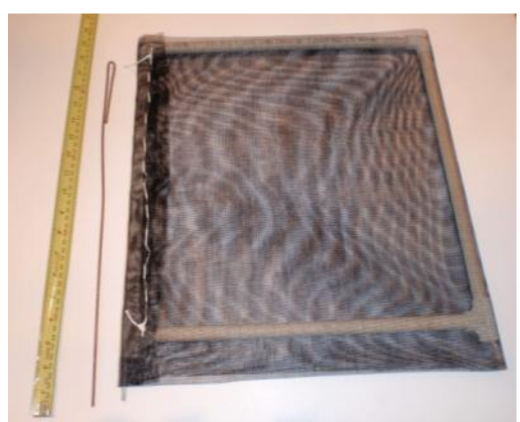


3. Cut a slice of 45 cm X approximately 92 cm (there is 92 cm rolls sold) of normal window screen (mesh size approx. 2 mm).

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

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

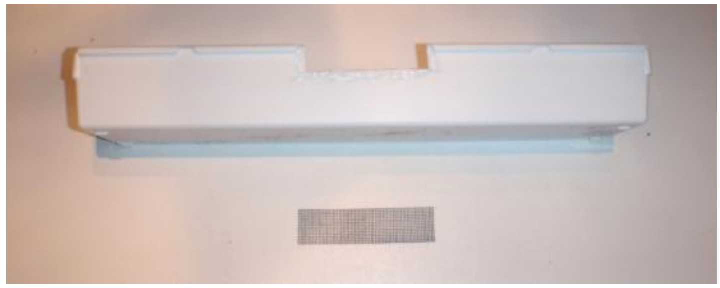
6. Tie rebar to vertical tubing with ziptie on each side to the frame should sit on top of the edges of the pitfall trap and the rebar should be pounded in the ground just outside the pitfall. Picture shows wire but rebar is preferred.
7. Tie a knot on the top tubing (3 turns around the tube and 2 flat knots).



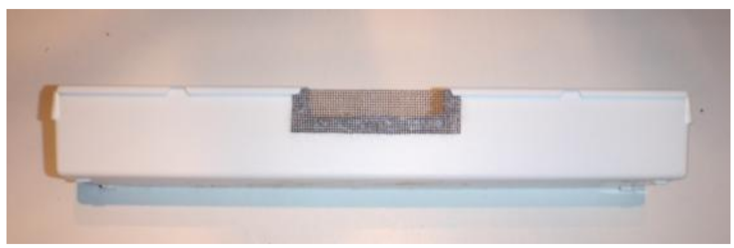
8. Tie both end of this rope together. This loop will be used to keep the frame vertical by providing easy location to secure ropes for anchoring trap.

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 (I'm using 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 the screen in place. A catalyst may be used to make the glue set faster.



11. The trap is now ready to be assembled outside in your favourite field site.

ASSEMBLING THE TRAP

12. Determine the direction of the dominant winds and configure placement of trap and screen to be <u>**perpendicular**</u> to the dominant wind.
13. Dig just the size of your pitfall container and avoid disturbing the surrounding area.
14. Install the pitfall container in the hole and replace soil so the pitfall is even with the ground.
15. 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.
16. 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 be slid off rebar.
17. 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
In order to 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 enough solution to the bottom of the pitfall trap to completely cover the bottom and be about 1 inch (2.5 cm) deep but does not pour out the overflow. 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, dispersing into a flowing water body or putting down a gray water sump.

Trap emptying and collection procedures

Release the guy lines on the trap and lift the screen and place on ground. Pour contents (solution and captured insects) of pitfall container through the fine mesh fish net into an open container so solution can be returned to pit fall trap. Transfer insects from mesh fish net to Melitta paper coffee filters and empty captured arthropods into a Melitta) paper coffee filter (https://shop.melitta.com/itemdy00.asp?T1=62+0122&Cat= size 2 or 4). Place the sample into the pre-labeled storage container and transport back to base camp. Take paper cone filters and gently rinse with plain water to remove trap solution. The paper cone filter and the arthropods on it can then be transferred and stored in a Whirlpak with full strength Isopropyl alcohol (e.g. isopropanol). For proper sample preservation, the volume of Isopropyl alcohol in the Whirlpak should be at least 4 times the volume of the arthropod sample or at least 2 cm at the bottom of the Whirlpack for small samples. Using a lead pencil, note the date and trap number on a piece of paper and place the paper in the Whirlpak with the sample. Samples may also be identified by writing on the outside of the Whirlpak 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. Make sure that the Whirlpaks are well inflated in order to decrease the crushing of samples and stored in a box. Store the samples upright until later identification and enumeration.

After installation, the sampling of traps is not time consuming. Sampling one transect of traps (5 traps) should take 1 person approximately 30 minutes every three days. Prepare a standard set of collection jars that can be used repeatedly throughout the season, ahead of time and leave supplies (refill trap solution) within proximity of the transect, allowing sampling to occur en route from other projects.

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

Methods for monitoring aquatic invertebrate prey in tundra ponds
by Mac Butler - NDSU malcolm.butler@ndsu.edu 701 231 7398

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.

Sampling pond insect emergence for seasonal phenology (OPTIONAL):

During emergence of winged adults, all aquatic insect species become available to shorebirds that forage along pond margins. Collecting a second type of sample will permit monitoring of the seasonal timing of insect emergence. The bottle traps may collect some pupae, but detection rates of emerging insects are unknown and may be low for many species. Many arctic insects remain on the water surface to swarm and mate, and in any case emergence failures and spent adults will accumulate along the leeward shore, where they decompose after several days. This flotsam will also contain the cast exoskeletons (exuviae) that are shed when aquatic insect larvae or pupae emerge to their adult form. By collecting a seasonal series of semi-quantitative samples of this washed-up material, one can document the seasonality of insect emergence. This material will reflect the whole insect community in a pond, including species from microhabitats that will not be sampled well by the activity traps. Arctic insects typically show highly emergence synchrony within a species, but both seasonal timing and the total span of community emergence can vary considerably – with consequences for avian predators.

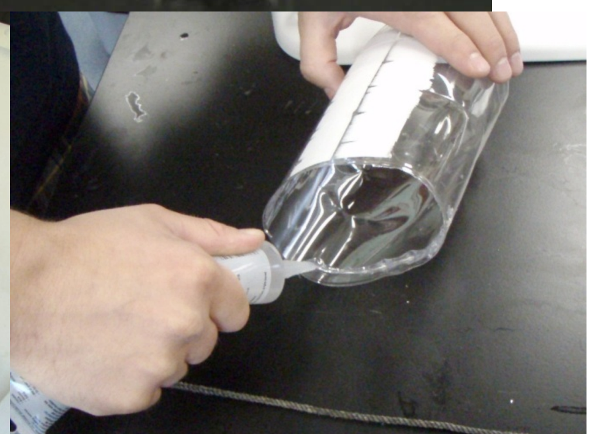
Sampling procedure for insect emergence:

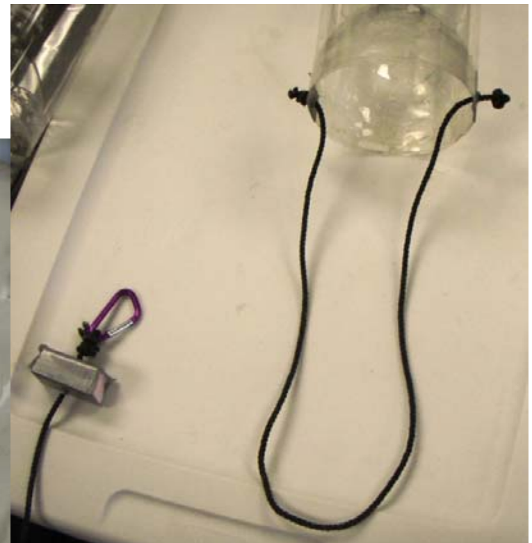
Materials needed: fine mesh dip net, collecting jar with label, squirt bottle with water, [isopropyl alcohol & Whirl Paks at base camp]

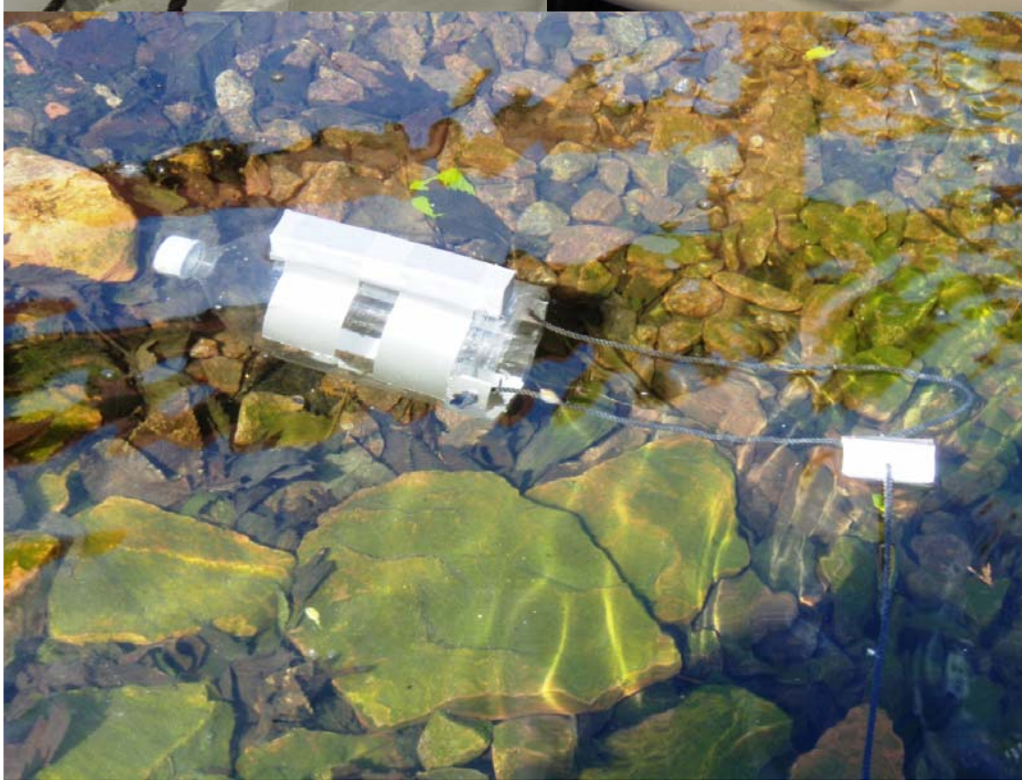
Samples should be collected at least every 3 days, every other day will provide even better resolution of emergence timing. These samples can be collected simultaneously with clearing of the bottle traps.

Go to the downwind shore (or where wind has blown across the water surface for most of the past 24hr). The grater the fetch (distance across the water), the more material will have accumulated. Look for white foam along the shore, or a line where the emergence vegetation may serve as a filter to trap drifting material. Use the dip net to skim the water surface along this line for a series of one-meter intervals. It's important to use the same sampling effort each time (different days and ponds), so the samples can be compared across time and among ponds. At times you may notice little in the net, but under magnification there will likely be evidence of insect emergence. At times of heavy emergence the insect bodies may accumulate in great quantity. 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. Generally, pooling five (5) 1m-long sweeps from the downwind edge of a pond is a good standard. If a longer or shorter total length is sampled, note such on the sample label.

As for the trap samples, the net contents must be rinsed into the collecting jar with water. 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. Within 12hr, decant as much water as possible, transfer the collected material to a Whirl Pak, and preserve with full-strength isopropyl alcohol. Only enough alcohol to thoroughly wet the collected material need be used, perhaps 10-20ml for most samples. Most air should be pressed out of the Whirl Paks before tightly rolling the tops. Double-bag the samples in a second Whirl Pak or a ziplock bag to minimize leakage, and pool all samples of one type in a larger plastic bag or secure box.







Predator point counts

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).

a) 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.
b) To identify predators during point counts use binoculars (8 x 42 or 10 x 40).
c) 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 **<u>the initial sighting</u>**.
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.

<u>Additional guidelines</u>:

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 Count Data recorded

(see page 91)

1. Observer name (first initial and full last name)
2. Plot ID
3. Date – dd-mm-yr
4. Time of arrival on plot/search area

5. Time of departure from plot/search area
6. Record start and end time for each timed census count at each survey point; record GPS location of these predator point counts.
7. Write species (including number of individuals) detected within 300m of the survey point. See **Appendix H** for list of potential predators and their codes
8. Record perpendicular distance (meter) to predator.
9. 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)
10. 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.

Lemming surveys

We will employ two 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 live count. Live counts will be conducted primarily by having people keep track of the number of lemmings observed during typical work days (see checklist count below). These focused lemming observation days can be done weekly or three times a summer. In the rare years and locations with lots of lemmings, a more directed lemming count will be done (see live transect count).

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 8) 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.

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.

Figure 8. Lemming winter nest found during early summer snow surveys.

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:

a) 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.

b) 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, he perpendicular distance to each nest seen is measured, no matter how far it is from the line of travel.

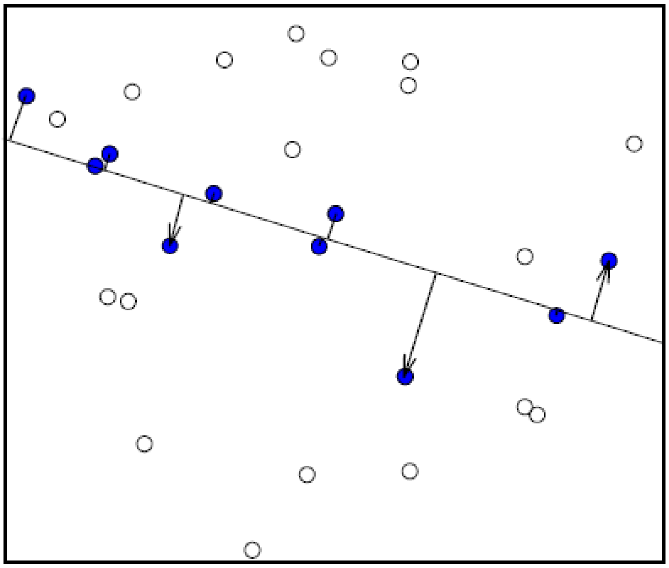


Figure 9. Schematic of winter nest count transect circles represent winter nests, and the line marks the survey line walked by the observer. Solid circles represent winter nests seen by the observer, and the lines mark the perpendicular distances measured to the center of the winter nest. Figure from 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

(see page 89)

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) Checklist counts (do in low to mid lemming years) – see page 77

As part of the Daily Checklist, people will take note of lemming numbers. Because people will be pre-occupied doing many routine tasks during the day, these counts are subject to a fair bit of error. To help correct this, camps will select particular days (either once a week or three times a year) during which personnel will pay particular attention to lemmings. On the given "live lemming day", field personnel will be told to make a conscientious effort to keep track of all lemmings (even more so than they typically do for the checklist). At the end of the day, lemmings will be recorded on the checklist but noted as a "lemming day". This method will be done in most cases. There is no official data form for this – it will go on the Daily Checklist but you must indicate when a focused effort was made on lemmings on the Daily Checklist and the Camp Journal.

(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).

In 2011, one or more sites will conduct live or snap trap lines to validate the live transects counts. Protocols for this will be developed later.

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

(see page 90)

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.

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 (see page 82 and 83; 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 surveys

(see page 92 for data form)

During the initial plot establishment/maintenance visits and subsequent monitoring visits, snow cover should be evaluated every other day until 90% or more is melted. A visual absolute snow cover estimate (to the nearest 5%) is given for an established area within the search area or a permanent intensive nest survival plot.

All sites should collect 20 replicate samples of snow cover at permanent locations every other day during the beginning of the season until the snow has melted. For sites establishing plots, snow cover will be quantified by randomly selecting twenty 50- m square quadrats within the nest survival plot and visually estimating absolute snow cover within the quadrat (with the northwest stake as the reference stake). For sites not establishing nest survival plots, snow cover will be quantified by establishing 20 snow survey locations and estimating snow cover in a 50-m radius surrounding the observation point. Please record GPS locations of each snow survey site in the meta-database. Surveys can be done during nest search and plot set up activities.

Surface water

(see data form on page 86)

We have talked with several people about establishing a remote method for measuring surface water via a collaboration within the Arctic Landscape Conservation Cooperative in Alaska. For this year, we propose a very simple method to record changes in surface water. We will work on developing finer resolution methods to estimate surface water in 2011. For this year, we advise each camp to locate 3 unique sites within each of four habitat types: the troughs of high-centered polygons, the centers of low-centered polygons, small ponds or waterbodies, and non-polygonized areas. If your site does not have one or more of these habitat types, then sample a representative habitat type that has water (but describe it).

It is important that you mark the locations where you measure water depth so you can go back to this site within and across years. Perhaps pound rebar in the ground to do this. Be sure to record the GPS location of all sample areas.

Water depth should be recorded at each site by placing a metric ruler through the water column until it rests on the surface. Do not push the ruler into the sediment but only lightly place it on the ground. Water measurements should be taken every week as water levels change quickly throughout the summer. Typically water levels will be high at the beginning of the year and then become dry. The exception could be during those years with large amounts of rainfall.

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 and then lands and calls.	Quick to moderate

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

The egg flotation method has been used to age the eggs of many bird species including shorebird biologist who have developed float methods generally getting accuracy estimates within ± 2-4 days of actual hatch dates.

<u>Materials</u>

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

<u>Methods</u>

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.

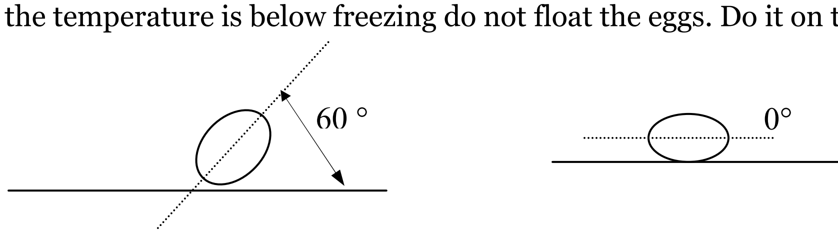
Carefully place eggs back in the nest. Remember to record the date you floated the egg.

<u>Important</u>

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

* Birds typically lay one egg / day. ** Nestling stage = 8-11 days
Note: The information in Appendix E 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.

<u>Egg shell top and bottoms</u>: 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.
<u>Eggshell fragment</u>: 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.
<u>Eggshell pieces</u>: 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.

<u>For finding eggshell tops and bottoms</u>:
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.

<u>For finding pip fragments</u>
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 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.

<u>Abbreviated list of Vegetation Units</u>

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 nest predators in the Arctic

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. <u>We do not record Sabine's Gulls and Arctic Terns as potential nest predators</u> because they are only believed to very rarely depredate nests.

Appendix I. Dunlin ageing guide

The information presented below is a reference for trying to age Dunlin. It is not complete and more information is available from the Pyle Guide. For this year, we advise using this reference and more importantly, **advise all people to take pictures of their captured birds so that age can be assessed post-capture in a consistent manner**.

Suggested 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.

Criteria	***Age***	
	SY-yearling	**ASY-adult >1 year old**
Inner median coverts	All heavily worn, with slight buffy edges (Fig. 1a)	Few distinct buff covs, fresh (Fig. 2)
Primary coverts (1st 2 covs)	Convex shaped white edge	Distinctive droplet or step
Flight feather wear	Lots of wear	Moderate wear
Flight feather shape	Thin and pointed at tip	Broad and rounded at tip

Figure 1a: A second year (SY) *arcticola* Dunlin. Note the heavily worn inner median coverts with a few innermost retained buffy edges. Figure 1b: Hatch year chick, ~14 days old to show fresh buffy inner median coverts. Photos by J. Choi/USFWS.

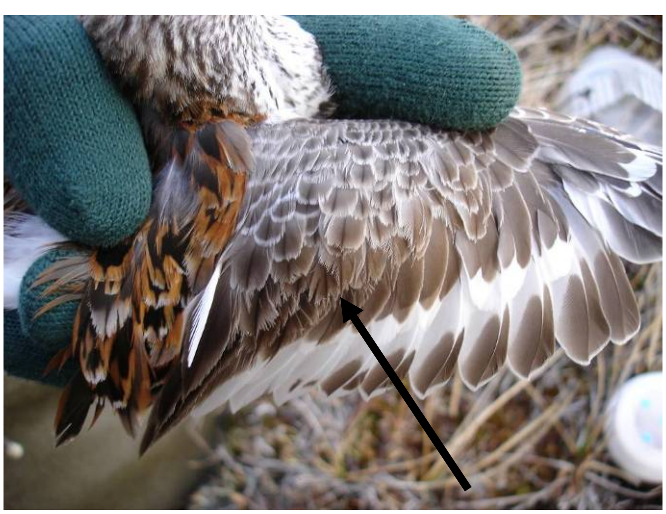


Figure 2. Known after second year (ASY) bird (by recapture). Note several inner median covert feather generations. Broad white tips to newly replaced inner median coverts

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.

<u>% of incubation complete</u> = 0.59 (from “other shorebird” table) x 21 (mean incubation length for BTGO) = 12.4 days old
So, the eggs will hatch in approximately (21 – 12.4) = 8.6 days.

DATA FORMS

Field Camp Meta database Information

Project PI name(s): Contact info (address, email and phone):	**Field Crew Leader (s):** Contact info (address, email and phone):

Seasonal staff or volunteers (first name, middle initial and last name) **and dates at camp**

Site name:	**Year of study initiation:**	**Type of study** (circle one): minimum intensive
Total field crew number:	**Field season start date:**	**Field season end date:**

Human activity in the vicinity description: (Check all that apply):

Vicinity (within 50 km) of permanent human settlement? ☐

Area affected by industrial development (e.g. oil and gas extraction, mining)? ☐

Established field station? ☐

Summer only field camp ☐

Other significant features of site (please specify):

Focal species methods checklist (Check all that apply):

	Adult band	Nest Survival	Side Project (describe)
SESA	☐	☐	☐
DUNL	☐	☐	☐
WESA	☐	☐	☐
REPH	☐	☐	☐
RNPH	☐	☐	☐
WHIM	☐	☐	☐
PESA	☐	☐	☐
______	☐	☐	☐
______	☐	☐	☐
______	☐	☐	☐
______	☐	☐	☐

Methods employed at site: Check all that apply and briefly describe.

Adult survival banding ☐

Nest survival effort ☐Intensive nest survival plot ☐Minimum nest survival search area

Snow survey ☐ 50 m circle ☐ 50m quadrat

Lemming index ☐ Winter nest ☐ Live lemming transect ☐ Trapping

Predator surveys: ☐ Weekly ☐ Seasonal

Food resources: ☐ Terrestrial ☐ Aquatic ☐ 3-day all season ☐ 3 day with daily peak emergence
Other (please explain):

Weather: ☐ Field camp station ☐ Manual recording ☐ Retrieve from other station
Other (please describe):

General timing of surveys conducted: List range of dates when surveys were conducted and frequency of occurrence (e.g. interval).

	Start date (dd-mm-yr)	End date (dd-mm-yr)	Interval (i.e. seasonal, weekly, bi-daily)
Adult survival banding			
Nest survival effort			
Snow surveys			
Predator surveys			
Food resources			
Terrestrial			
Aquatic			
Weather monitoring			
Lemming surveys			
Other:			

Site weather conditions:
Snow present on arrival in flat area? **YES** **NO**
Date of estimated 50% snow cover on flat areas (dd-mm-yr): ________________________
Date of ice break up on major rivers (dd-mm-yr): ________________________
Date of final loss of snow cover on flat areas(dd-mm-yr): ________________________
Has the site been visited in previous years? Please describe site history

Field Camp Meta database: Study Area Map

Create a study area map illustrating geographic locations for the study area. If at all possible, obtain a Google Earth image of the area and add the location of the items listed above. Be sure to add labels to identify all items including the location of the following:

1. Intensive plots
2. General search area
3. Snow cover survey sites
4. Winter lemming nest transect
5. Weather station
6. Predator count stations
7. Terrestrial invertebrate trap line (mesic and dry)
8. Aquatic activity traps in tundra ponds
9. Camp

Field Camp Meta database: Geographic Information:

Geographic information must be collected for each Network component using a Global Positioning System (GPS) unit. **To ease future GIS applications, we insist that all data be submitted in latitude/longitude decimal degrees (e.g., -145.78675 degrees) and the WGS 84 (or NAD 83) datum.** Geographic information collected in other formats must be converted to the above mentioned coordinate system and datum before the data is submitted.

Component	**Latitude**	**Longitude**
Field Camp		
Field camp (housing)		
Weather station		
Ecological Monitoring		
Insect dry transect start:		
Insect dry transect stop:		
Insect wet transect start:		
Insect wet transect stop:		
Aquatic insect trap #1		
Aquatic insect trap #2		
Aquatic insect trap #3		
Aquatic insect trap #4		
Aquatic insect trap #5		
Lemming Winter nest transect start:		
Lemming Winter nest transect stop:		
Lemming live transect count start:		
Lemming live transect count stop:		
Predator point count stat #1, plot 1		
Predator point count stat #2, plot 1		
Predator point count stat #1, plot 2		
Predator point count stat #2, plot 2		
Adult and nest survival plots		
Nest plot ID ___northwest corner		
Nest plot ID ___northwest corner		
Nest plot ID ___northwest corner		
Nest plot ID ___northwest corner		
Nest plot ID ___northwest corner		
Nest plot ID ___northwest corner		
Other reference stake locations:		
e.g. Plot 1 stake # A1		

Network Site Daily Camp Journal

Camp______________ Year ________

Date:__________________________
Personnel on and off duty (where they worked and what they were doing):
Arrival/departure of personnel , visitors:
What ecological monitoring was done? What nest searching or banding was done?
Weather (e.g., sunny, cloudy, rainy, wind speed and direction, and general terms like beautiful and shitty)
Significant natural events and rare sightings
Personnel Incidental Observation Effort Name (1st initial and full last name) Hours Mode of transport Location surveyed

Banding data form

Bander	DD-MO-YY	Time 24hr	Book	Nest
NestID	PlotID	ON / OFF Plot		

Capture Method: Bownet / Walkin / Mistnet / Standing Decoy / Other define

Capture Status: Pre-laying or Laying / Incubation / Brood / Post-breeding / Unknown

Band Number	Species Code	Color Combo UL	LL	UR	LR

Recapture from prior year: Y N   Picture: Y N
Recapture from this year: Y N

Flight Feather Molt (0-5)

Secondaries inner←middle										Primaries middle→outer										Tail (right side) middle→outer					
10	9	8	7	6	5	4	3	2	1	1	2	3	4	5	6	7	8	9	10	1	2	3	4	5	6

Body Feather Molt (0, 2-5)	Head	Neck	Back	Breast	Abdomen

Morphometrics

Culmen 0.1 mm	Total Head 0.1 mm	Diagonal Tarsus 0.1 mm	Wing 1 mm	Weight g	Bag Weight g	Fat 0-7
.	.	.				

Collections

Blood in Longmire	Blood for MethyHg	AI Swab	Feathers
Y / N Capillary: EDTA / Plain / Hep % of tube:	Y / N % of tube:	Y / N Sample # CP only /OP only/both	Y / N List e.g. 2pL , 2pR, breast

Sex: M F Unknown
How Sexed: Culmen / Plumage / Brood Patch / Cloacal Size / Wing / Overall Size / Egg Laying

Age: SY / ASY / AHY / Hatch Year Local /Unknown / Chick Can't Fly
How Aged: Plumage (notes required for SY & ASY) / Weight / Recapture

Release Status: Band&Release / Band&Escape / Release Unbanded / Injured,Band&Release / Mortality

Notes geolocator Tag #: ___________________ time put on bird: _____________________

GPS Location: latitude: _____________________longitude:___________________________

Daily species list data form

Network Site ___________________ Year ________________ Month _____________________________ Page 1

Species	1	2	3	4	5	6	7	8	9	10	11	12	13	14	15	16	17	18	19	20	21	22	23	24	25	26	27	28	29	30	31	Nest	Hatch	Fledge
Pacific Golden-Plover																																		
American Golden-Plover																																		
Black-bellied Plover																																		
Ringed Plover																																		
Semipalmated Plover																																		
Eurasian Dotterel																																		
Hudsonian Godwit																																		
Bar-tailed Godwit																																		
Eskimo Curlew																																		
Whimbrel																																		
Bristle-thighed Curlew																																		
Lesser Yellowlegs																																		
Greater Yellowlegs																																		
Spotted Sandpiper																																		
Wandering Tattler																																		
Ruddy Turnstone																																		
Black Turnstone																																		
Red-necked Phalarope																																		
Red Phalarope																																		
Wilson Snipe																																		
Short-billed Dowitcher																																		
Long-billed Dowitcher																																		
Surfbird																																		
Red Knot																																		
Sanderling																																		
Semipalmated Sandpiper																																		
Western Sandpiper																																		
Red-necked Stint																																		
Least Sandpiper																																		
White-rumped Sandpiper																																		

Species	1	2	3	4	5	6	7	8	9	10	11	12	13	14	15	16	17	18	19	20	21	22	23	24	25	26	27	28	29	30	31	Nest	Hatch	Fledge
Baird's Sandpiper																																		
Pectoral Sandpiper																																		
Purple Sandpiper																																		
Rock Sandpiper																																		
Dunlin																																		
Curlew Sandpiper																																		
Stilt Sandpiper																																		
Buff-breasted Sandpiper																																		
Trumpeter Swan																																		
Whistling Swan																																		
Greater White-fronted Goose																																		
Snow Goose																																		
Ross's Goose																																		
Emperor Goose																																		
Canada Goose																																		
Brant																																		
American Wigeon																																		
Green-winged Teal																																		
Mallard																																		
Pintail																																		
Northern Shoveler																																		
Greater Scaup																																		
Lesser Scaup																																		
Common Eider																																		
King Eider																																		
Spectacled Eider																																		
Steller's Eider																																		
Harlequin Duck																																		
Long-tailed Duck																																		
Black Scoter																																		
Surf Scoter																																		
White-winged Scoter																																		
Barrow's Goldeneye																																		
Red-breasted Merganser																																		

Species	1	2	3	4	5	6	7	8	9	10	11	12	13	14	15	16	17	18	19	20	21	22	23	24	25	26	27	28	29	30	31	Nest	Hatch	Fledge
Common Merganser																																		
Red-throated Loon																																		
Pacific Loon																																		
Common Loon																																		
Yellow-billed Loon																																		
Red-necked Grebe																																		
Horned Grebe																																		
Northern Fulmar																																		
Double-crested Cormorant																																		
Pelagic Cormorant																																		
Red-faced Cormorant																																		
Osprey																																		
Bald Eagle																																		
Northern Harrier																																		
Rough-legged Hawk																																		
Golden Eagle																																		
Merlin																																		
Gyrfalcon																																		
Peregrine Falcon																																		
Willow Ptarmigan																																		
Rock Ptarmigan																																		
Sandhill Crane																																		
Pomarine Jaeger																																		
Parasitic Jaeger																																		
Long-tailed Jaeger																																		
Ivory Gull																																		
Mew Gull																																		
Herring Gull																																		
Thayer's Gull																																		
Great Black-backed Gull																																		
Glaucous-winged Gull																																		
Glaucous Gull																																		
Bonaparte's Gull																																		
American Tree Sparrow																																		

Species	1	2	3	4	5	6	7	8	9	10	11	12	13	14	15	16	17	18	19	20	21	22	23	24	25	26	27	28	29	30	31	Nest	Hatch	Fledge
Orange-crowned Warbler																																		
Yellow Warbler																																		
Blackpoll Warbler																																		
Northern Waterthrush																																		
Wilson's Warbler																																		
Rusty Blackbird																																		
Common Redpoll																																		
Hoary Redpoll																																		
Grey-crowned Rosy Finch																																		
Pine Grosbeak																																		
Gray Jay																																		
Black-billed Magpie																																		
Common Raven																																		
Other (give names):																																		

Species	1	2	3	4	5	6	7	8	9	10	11	12	13	14	15	16	17	18	19	20	21	22	23	24	25	26	27	28	29	30	31	Nest	Hatch	Fledge
Terrestrial Mammals																																		
Caribou																																		
Arctic Fox																																		
Red Fox																																		
Arctic Ground Squirrel																																		
Short-tailed Weasel																																		
Greenland Collared Lemming																																		
Brown Lemming																																		
Tundra Vole																																		
Polar Bear																																		
Brown Bear																																		
Marine Mammals																																		
Ringed Seal																																		
Bearded Seal																																		
Bowhead Whale																																		
Beluga																																		
Walrus																																		
Grey Whale																																		

Weather data forms-manual recording: Daily precipitation

Site: _______________

Date	**Precip. (mm)**	**Notes:**

Weather Data forms: Manual snow fall measures data sheet

Site: ______________________ Year: _________________

	Replicate measures of snow depth (cm)									
Date (dd-mm)	**1**	**2**	**3**	**4**	**5**	**6**	**7**	**8**	**9**	**10**

Food Resources Sample Collection catalog

Year_______ Field site:____________

Terrestrial- mesic Date installed:_____________

Date	**Collector**	**Total samples collected**	**Notes (e.g. number of samples lost or damaged)**

Terrestrial- dry Date installed:_____________

Date	**Collector**	**Total samples collected**	**Notes (e.g. number of samples lost or damaged)**

Aquatic Date installed:_____________

Date	Collector	Total samples collected	Notes (e.g. number of samples lost or damaged)

Daily spot map **ASDN Date: ________________ Time start:____________ Time end: __________Site ________________**

Observer(s) _______________________________________ PLOT ID __________

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

Behavior Key:
S: heard or seen singing
COP: copulation
D: courtship display
DD: distraction display
A: aggression
F: foraging
Color: / : /

Master nest/ territories map *ASDN* Year: _________ Site: _________________ PLOT ID:____________________

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

Territory Key:
Color: ___/___:___/___

Nest found

Probable territory

Book *Page*

Nest record sheet

Nest ID	Observer
NW Stake (if stakes used)	Date Found
GPS N	Plot # / Locale
GPS W	Located WITHIN or OUTSIDE Plot
Est. Hatch Date	Found by SEARCHER / ROPE / BANDER
Bands (UL/LL:UR/LR sex)	Bands (UL/LL:UR/LR sex)

Nest Site Map / Description

Nest Visits including discovery

Day & Month	Time 24 hr	Obs. Initials	Stage	Nest Seen (Y/N)	# Eggs/ Chicks	Pip Star Crack	Done this visit	Next visit date	To do next visit

Nest ID: **Book** ___________ **Page** _________

Eggs

		1	2	3	4		
	length (mm)						
	width (mm)						
Date 1	float angle					Est. Age	Initiation Date
	surfaced (Y/N)					days	Incubation Date
	# mm above						

Nest Fate Dav & Month:

Nest: INTACT / SCATTERED / FLATTENED & WIDENED

Fox: URINE / SCAT / NO

Egg Frags found: YES NO

Egg Bits in nest: YES NO

Membranes: ATTACHED / SEPARATE / NONE

Membranes /shell with attached: BLOOD / YOLK / NONE

Weather Induced: YES NO

Caribou trampling: YES NO

Adult broody: YES NO

Brood seen: YES NO

# of young _________

distance from nest (m) _________

Final Nest Fate: HATCH / FAILED /ABANDON / UNKNOWN / UNDETERMINED

Cause of failure: WEATHER / PREDATION / TRAMPLING / OBSERVER / OTHER

____

Complete after Fate

Dominant Landform

____

Dominant Vegetation

____

Concealment %

____

Lemming winter nest count transect data form

Network Site: ________________Date (dd/mm/yy):_______________
Start time:_________ End time:_________________
Obs:________ distance surveyed (km): _______________
Start loc. (Plot and grid stake or Lat/Long): ___________________________________
End loc. (Plot and grid stake or Lat/Long): ___________________________________

Nest number	Perpendicular Distance to nest (m)	Nest number	Perpendicular Distance to nest(m)
1		36	
2		37	
3		38	
4		39	
5		40	
6		41	
7		42	
8		43	
9		44	
10		45	
11		46	
12		47	
13		48	
14		49	
15		50	
16		51	
17		52	
18		53	
19		54	
20		55	
21		56	
22		57	
23		58	
24		59	
25		60	
26		61	
27		62	
28		63	
29		64	
30		65	
31		66	
32		67	
33		68	
34		69	
35		70	

Lemming live transect count data form

Network Site: ______________________ Year ___________
Transect ID _______________________
Start loc. (Plot and grid stake or Lat/Long): ___________________________
End loc. (Plot and grid stake or Lat/Long): ___________________________

(Each line represents a single count on a transect established for the Network site on a single day; thus there is one sheet for entire season. If there are more than one transects at a Network site, then you will need multiple sheets.)

Date	**Observer**	**Start time (24-hr)**	**End time (24-hr)**	**Species / Count**	**Species / Count**

Predator monitoring data sheet

Observer: _______ Study site: _____________ Plot: _____ Date (dd/mm/yy) ________

Species	**Distance (meters)**	**Number of individuals**	**Comments**
COUNT #___	**Start time:**	**End time:**	**Survey location:**
COUNT #___	**Start time:**	**End time:**	**Survey location:**
Species	**Distance (meters)**	**Number of individuals**	**Comments**

Predator nest location (Species and lat/long):_____________________________________
Predator nest location (Species and lat/long):_____________________________________

Snow survey form

Site: ______________Observer: ______ Plot: ______________ Date (dd/mm/yr): ____________

Location ID	Snow Cover (to nearest 5%)	Notes
e.g. B2	25	

Surface Water

Site: _____________Observer: _______________ Date (dd/mm/yr): ___________

Habitat Type	Location ID	Water Depth (cm)	Notes

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