Practical guide for investigating breeding ecology of Kentish plover

Charadrius alexandrinus

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Photos by T. Székely, A. Kosztolányi & C. Küpper
Rationale
The Kentish/snowy plover *Charadrius alexandrinus* is a small cosmopolitan shorebird (body mass about 40-44 g). In the last few years we have developed a suite of methods to investigate its behaviour and ecology in the field. We thought this practical guide may be useful for students and researchers with an interest in small plovers. Some aspects of these methods may be relevant for other shorebirds and ground-nesting birds in general.

Our fundamental motivation in writing this guide is to show that the Kentish plover is an easy species to work with, if one is willing to pay attention to a few potential pitfalls. We hope that this guide will elicit further research. Please contact us if you have questions and comments, and let us know of any errors. Note that Kentish plovers have been studied in several countries and by a good range of researchers, and we don't claim that our methods work best.

Many Kentish plover populations are now declining. You need to be sensible about fieldwork, and carefully evaluate the costs and benefits of using a particular method. The last thing you want is to put an extra burden on plover populations - they have a hard time anyway to cope with predators, floods and threats humans are imposing upon them.


Essential breeding ecology
Good reviews of Kentish/snowy plover (KP henceforward) natural history can be found elsewhere (Cramp & Simmons 1983, Page *et al.* 1995, Amat 2003), and here we only focus on essential aspects. KPs are migratory in most parts of their range, although populations close to the equator are only partially migrant or resident. They breed on edges of saline lakes and lagoons, and inhabit salt-mashes and sand dunes. Their breeding season usually lasts for about 2-5 months; populations in the north tend to be single-brooded, whereas southern breeders may double (or triple) brood. Failed breeders often re-nest.

Adult males and females have dimorphic plumage. Males have incomplete black breast-bands, black eye-stripes and a black frontal head bar, whereas these areas are pale brown in females. Males also sport a cinnamon nape and crown, although there is a substantial plumage variation among breeding populations. In early breeding season the sexes are easy to distinguish, whereas the difference in plumage between sexes becomes blurred as the season progresses.

Kentish plovers lay their eggs in a small depression on the ground scraped by the male. The modal clutch size is three eggs, and the eggs hatch after 25-26 days of incubation. Both sexes incubate the eggs; females incubate mostly during the day, and males incubate mostly during the night. The parents lead the chicks away from the nest-scrape within a few hours of hatching. The parents attend, brood and defend their chicks for about 4-5 weeks, but they don't provide food for them. One parent (the male or the female) often deserts the brood, and re-nests with a new mate. Thus many KPs are socially monogamous, although both polygyny and polyandry occur in most populations that have been studied to date.

Searching for nests
*Equipment needed:* binoculars, spotting scope, mobile hide (see *Appendix 1*) or car.

There are three main methods of nest search:
(i) **On foot.** Potential breeding sites can be screened by walking and searching for nests. Empty nest
scrapes (with or without some nest materials) often indicate the presence of active males. On sand dunes, the plover footprints tend to concentrate around their nests. On salt marsh, it is worth looking for sites that are somehow more elevated from the rest, so that the nest is less prone to flooding. Objects that break monotony of the ground (e.g. debris, deep footprints, drift-wood) are often preferred locations. Eggs in fresh (or incomplete) nests tend to be fully exposed, and as incubation progresses nest materials gradually accumulate so that the eggs may be nearly completely covered.

(ii) Spotting incubating parents. The observer should sit on an elevated vantage point (such as a dyke or on a sand bank), or inside a car or mobile hide. Incubated plovers can be spotted by their distinctive white breast, or their contour against the background. Sitting plovers can be easily distinguished from incubating ones: the incubating parents appear to have bulging breasts, and when they run off the nest they often throw nest materials towards it. The behaviour of non-incubating plovers is more relaxed; they often preen and alternate between sitting and standing, and they don’t have the typical shaking moves of incubating parent when settle on the eggs.

When using a mobile hide (Appendix I) it is a good idea to move for a stretch of 40-50 m, and then stop and look around using both binoculars and spotting scopes. Make sure you screen the same spot from different positions – you may be surprised how many nests you miss by superficially looking around. It is worth checking the area close to the hide, because some plovers continue incubating until the hide is only a few meters from them. Make sure you screen your path very carefully to avoid pushing the hide over a nest.

Make notes about males that are advertising and defending a particular spot, and pairs that are profusely display, scrape and copulate. A few days later you may find their nest – although this is not guaranteed since many unmated males and pairs eventually nest elsewhere.

(iii) Watching parent(s) returning to the nest. Plovers can be very tolerant of observers inside a car or hide, but they are wary of observers on foot. The flushed parents may run back to the nest in a straight line, although cautious individuals may zigzag, or exhibit seemingly foraging movements whilst gradually approaching their nest. Carefully note the location where the plover disappeared from your sight: often this is a telltale of a nest.

Beginners often confuse false-brooding (a form of nest defence) from real incubation; the former is usually exhibited to observers on foot, and they can be displayed in unsuitable habitats (e.g. in shallow water or wet mud). If you spot a nest from a distance, you need to walk to the nest in a straight line whilst fixing your eyes on the suspected nest location. Many nests are well camouflaged, and a mistake of 1 m may mean you can’t see the eggs even though they are right in front of you. Watch your steps near nests and ignore the displaying parents: the last thing you want is crushing the eggs.

If you flush several plovers, it is a good practice to choose the most anxious plover (the one that does lots of head-bobbing and short abrupt runs), and then follow his/her movements for 10-15 minutes. Bear in mind that if a plover is very agitated, for instance it zigzags for 5-10 minutes in front of you or tries to lure you away; these often mean you are too close to their nest (or chicks). You need to carefully reverse and let the parents return to their nest (or brood).

*Carry out nest search sensibly, especially if the weather is very cold or hot:* by keeping the parents away from their nest, you may fatally expose the eggs. A good practice is to work swiftly and efficiently in a location, and move to a different location as soon as possible to let plovers resume their normal life. *It may take years to figure out and avoid ‘the elephant in a china shop’ effect, but the sooner you start realising your potential effects the better.*

3
Measuring and checking nests

Equipment needed: Nest notebook, GPS device, sliding calliper, small jar filled with fresh water.

Once a nest is located, you need to record the essential data (Appendix 2). Work efficiently and don't spend more than 5-10 min at the nest: this is NOT the place to celebrate, or discuss the latest gossips. Try to leave as few footprints as possible, and don't approach the nest if visual predators (e.g. rooks, shrikes) or humans are around. If you suspect that nocturnal predators (e.g. foxes, hedgehogs, jackals) may locate the nest using your scent, you should avoid approaching nests and handling eggs in late afternoons and in the evening.

We note the nest and egg number on the blunt end of egg using black permanent marker, e.g. 34/2. These numbers are helpful when you only find egg shell remains. People often worry about the harmful effect of marker solvent on embryonic development: we have not seen any evidence of this.

You need to measure egg length and breadth using a sliding calliper (Figure 1). Hold the egg horizontally in your palm, and then gently push the sliding calliper downwards and simultaneously pull apart the jaws of the calliper. Record the measurement when the egg squeezes through between the jaws for the first time. Do NOT force the calliper: the eggshell is thin, and can easily break.

![Figure 1. Process of measuring eggs (from left to right) as illustrated with a hen egg.](image1)

To get accurate measurements, you may repeat the process three times and take the median of readings. Note that nothing comes free: this will increase the amount of time you spend at the nest.

If a nest is found after egg-laying is completed, you may need to estimate egg-laying date by floating the eggs in lukewarm water. Use a small transparent jar for this purpose (honey and jam jars work best). Hold the egg firmly between your fingers whilst placing it in the jar: do NOT drop the egg (Figure 2). The jar should be wide enough to let you hold the egg firmly, and short enough to allow safe removal of egg with your fingers. Do NOT roll (or pour) the egg from the jar. It is a good idea to wipe the egg to remove water droplets, and float only one or two eggs of a clutch.

![Figure 2. Placing an egg into a floating jar.](image2)
We use two methods for estimating the number of incubated days (see also Fraga & Amat 1996).

(i) Noszály & Székely (1993)

Table 1. (a) Variation in floating position of the Kentish Plover egg during incubation. (b) Floatation stage of eggs in relation to the number of days incubated. Eggs were measured daily from the date of laying.

(Miklapusztá records, 1991–1992)

<table>
<thead>
<tr>
<th>Number of days incubated (mean ± SE)</th>
<th>A</th>
<th>AB</th>
<th>B</th>
<th>C</th>
<th>D</th>
<th>E</th>
<th>F</th>
</tr>
</thead>
<tbody>
<tr>
<td>Number of eggs used for calibration</td>
<td>5</td>
<td>7</td>
<td>7</td>
<td>6</td>
<td>3</td>
<td>3</td>
<td>3</td>
</tr>
</tbody>
</table>

These flotation stages correspond to the number of incubated days as calibrated in Southern Turkey (J Kis, unpublished data). Note that stage F can be anything between 10 days and 25-27 days:
Olivier Pineau (Tour du Valat Biological Station, France) has designed the following chart:

It is a good idea to write down a concise description of nest location in your Nest notebook (see below), the distances from landmarks (for instance, a small bush, grass patch or a peculiar piece of rubbish), and make a sketch. This may seem old fashioned given the accuracy of handhold GPS devices, although experience shows that the notes help visualising the spatial distribution of nests so that make fieldwork efficient. For remembering digits of coordinates may require photographic memory, whereas associating a nest with a particular landmark is easy: ‘S6 is 4 m south from the blue shoe’. If you decide to use UTM coordinates these will give you the distances in meters.

A plastic straw at a sufficient distance from the nest (e.g. 10 - 15 m) in a standard direction will speed up relocating the nest. Be sensible and remove straws if you suspect that people (or clever predators) may use them as clues. Collect the straws once all chicks have hatched or the nest has been predated. Straws are not foolproof either: grazing sheep and cattle often fancy chewing them.

Nests may be checked at 4-5 days intervals to estimate egg survival. If possible, stay at a distance from the nest and don’t handle the eggs. Near the time of hatching (approximately after the 22nd day of incubation), it is a good idea to check nests daily and tap the eggs gently. To ring the downy chicks in the nest-scrape, you may need to check peeping eggs 2-3 times each day. Eggs may peep for 2-3 days before hatching, although some peculiar chicks may pop out without much peeping. Near hatching you may also notice minute cracks on the eggshell; these may be sensed by gently
turning the eggs between your fingertips. Tiny eggshell remains in the bottom of nest scrape let you identify successful nests for a few days after the chicks left the scrape. In contrast, chewed pieces of eggshell or egg remnants around the nest are telltale of predation: to figure out the culprits you need to use nest cameras.

We devote a full (or half) page for each nest in our Nest notebook, so that all data for a given nest are on a single page (Appendix 2). Bear in mind that incubation often speeds up toward the breeding season, so that nests laid late in the season may hatch faster than you expect. Small clutches (1 or 2 eggs) also have the tendency to hatch sooner than clutches of three.

**Trapping at the nest**

Equipment needed: traps, bird bag, binoculars and/or spotting scope.

Shorebird biologists use a variety of methods to trap plovers, including noose mats, mist-nets and funnel traps (see Conway & Smith 2000 for references). We found funnel traps by far the most reliable, safe and easy. Not all plovers can be caught; it is best to start with a simple method and get complicated ONLY if it is essential to trap a given individual. Bear in mind that the harder you push a parent the higher the chance it will abandon the nest.

(i) Funnel trap. The diameter of the trap is about 50 cm, and its height about 20-25cm (Figure 3). Use a local blacksmith to weld a frame from strong wire, and cover the frame with chicken wire of mesh size < 3cm in diameter. All sharp edges of the chicken wire should point outward to avoid injuring the trapped plover. It is good practise to check the traps weekly to make sure that all sharp edges remain outward. There is a slight preference for allowing the entrance width to vary (Figure 3b & c versus Figure 2a), to accommodate the needs of very cautious and crafty plovers that runs in-and-out the trap.

(a) ![funnel trap image](image-a)
(b) ![funnel trap image](image-b)
(c) ![funnel trap image](image-c)

*Figure 3. Funnel traps.*

1. Place the trap on the nest. You may position the nest in one of the 'corners' of the trap (Figure 3c), or in the middle (Figures 3 a & b). The former is safer in terms of trapping the parent, although it may take longer for the parent to enter the trap.
2. Hide at least 50m away from the nest to have a good view of the trap entrance. Watch out for humans, livestock herds and predators so they do not damage the trap, and/or the parents.
3. If a parent has entered the trap and sat on the eggs, you should run to remove it from the trap: quick actions will reduce stress and the chance of injury. When trying to grab the parent inside the trap you need to be careful to avoid damaging the eggs, or injuring the plover or yourself.

Females are usually easy to catch early in the morning and males just before dusk. Do NOT trap at extreme weather conditions such as raining, scorching heat or piercing cold. If it is essential to trap
during the heat of the day, you either (i) shade the eggs by placing a flat object on the top of the trap (dry cow dung just works fine), or (ii) replace the eggs with dummies. Traps should not be left on the nest for excessive periods - the definition of 'excessive' is up to you, but it is rarely sensible to go beyond 20-30 minutes. Instead of forcing your way through, it is better to repeat trapping 2-3 days later.

You should NOT trap at a nest that has been incubated for less than 4-5 days. Also, if both parents happen to enter the trap simultaneously, release one immediately to let him/her incubate the nest whilst you measure and ring the other.

(ii) Round trap. If the funnel trap fails, you can try the round trap (Figure 4) on a different day. It takes longer to set up than the funnel trap, and you will need more time to retrieve the fishing line. Also, bear in mind that you may need special permission to use this trap.

To make a trap you need a ring (approx 80 cm diameter) of wire (or iron) about 0.5 cm diameter. Attach a loose fishing net to the ring using a threat all around - the less visible the net the better. Avoid shiny materials. You will also need a stick (reed or bamboo, about 40 cm), 2-3 pegs to hold the circle firmly on the ground, and fishing line with a reel to hold about 100 m of fishing line.

Set up the trap about 15 cm from the nest so the ring is well above the nest. Attach the fishing line to the stick, and firmly hold the other end of the fishing line in your hide (or car). After the parent has resumed incubation, pull the fishing line with one quick and strong motion so that the ring falls to the ground. Make sure that the pegs are strong enough to keep the trap in its place, and the fishing net is loose enough that the parent will not be injured.

Figure 4. The round trap.

Ringing and measuring
Equipment needed: Metal rings, colour rings, ringing pliers, sliding callipers, wing ruler, spring balance, Capture notebook.

Ringing should follow the general protocols for a given country. Please stick to the rules. A useful reference is Redfern & Clark (2001). Before ringing plovers, especially freshly hatched chicks, you need to get advice from a trained ringer. As with nests, work efficiently during ringing; release the bird as soon as possible.

Appendix 4 summarises the main data we advise collecting from plovers. Kentish plovers usually live in saline environments, therefore rings made of non-corrosive material (e.g. steel) are preferred.
over aluminium ones. You may put the metal ring above the 'knee-joint' to reduce corrosion.

Darvic colour rings are resistant to sunlight and environment, and we prefer to use overlapping colour rings (as opposed to split rings): White, Yellow, Orange, Red, Green, light green and dark Blue are easily distinguishable. Avoid black, dark brown and light blue. Countries may have their own colour ringing scheme, although overall, we do not recommend using more than four rings on a given plover (Figure 5, three colour rings and one metal ring). Leg flags are spreading among shorebird biologists: the pros and cons of leg flags need to be established. The safest ring position we found was one ring each above and below the 'knee-joint' on each leg. Chicks younger than three weeks should only be fitted with a metal ring and one colour ring at most. Note that colour rings may attract the attention of predators and bright colours such as Orange and Yellow are best avoided for chicks.

Figure 5. Schematic illustration of a coding system. The sketch shows a plover from behind.

You may start a series with a given ring in Colour_1 position. Then choose Colour_2 and Colour_3 using the following chart, and cross the already used combinations:

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<table>
<thead>
<tr>
<th>Colour_2</th>
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<tbody>
<tr>
<td>B G g O R W Y</td>
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<tr>
<td>B G g O R W Y</td>
</tr>
<tr>
<td>B G g O R W Y</td>
</tr>
<tr>
<td>B G g O R W Y</td>
</tr>
<tr>
<td>W Y X</td>
</tr>
</tbody>
</table>
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The scheme can be noted as XXX|XX.XX where X indicates a colour (or Metal) ring, the full stop marks the position of 'knee-joint' and vertical line divides the left and right leg. Thus the readout is left above . left below | right above . right below. For example, if Colour_1 is Blue, then the above plover is coded as B.M | R.W

This scheme may give 7 x 7 x 7 = 343 individuals. By rotating the location of Metal ring you can ring up to 1372 individuals.

Appendix 5. provides a guide how to take body measurements.
Blood and tissue sampling

Equipment needed: sterile needle (size 25G or 23G), glass capillary, tubes with buffer or ethanol, small piece of tissue paper.

Blood sampling: Blood sampling is one of those things you need to see in person. We do our best to describe the methods here and provide pictures for the important steps, but this may not replace the demonstration by an experienced person. Before you think about practical issues, make sure that you have all necessary permissions: blood/tissue sampling permit, if you work in a foreign country then you will need export and import permits. Bear in mind that it can take several months for the authorities to issue these permits, so submit the applications well in advance of the field season. With the emergence of avian influenza, there will be regulations that need to be heeded, on top of the usual legislative, health and safety issues.

From adults take the samples from their brachial (wing) vein. First, open the left wing of the plover whilst holding it on its back (if you're left-handed, this may be the right wing). Then push apart the inside wing covers to make the area clearly visible around the wing vein. Gently wetting the inside wing covers helps to clear the area around the vein. This vein can be spotted as it crosses over a wing-bone. Second, hold a sterile needle flat on the wing, and with a single move puncture the vein; do NOT insert the needle into the vein itself. The rule is to pierce ONLY if you clearly see the vein; do NOT pierce by trial & error: this could easily injure (or kill) the bird. Suck the drops of blood into a capillary - usually a couple of droplets come out. Press the tissue paper on the wound, hold for about 20 s and fold back the wing to the body with the tissue on the wound. Bleeding usually stops within a few seconds. However, check before releasing the bird that the bleeding has stopped.

![Figure 6. Blood sampling of adult plovers. Upper row: stretching the wing (left) and locating the brachial vein (middle), puncturing the vein with a 25G needle (right). Lower row: Filling the capillary with 25 µl of blood (left), stopping the bleeding by pressing a tissue to the wound (middle,) and after 20 s folding the wing with the tissue on the wound to the body (right).](image)

From chicks take blood samples from their leg vein. If you look carefully at the leg of a chick, you will see the vein goes along the inside of the tarsus at the 'knee-joint'. Carefully puncture the vein and collect about 25µl (1-2 droplets, a third of a 75 µl capillary) of blood. Make sure that the needle only pierces the vein and does NOT penetrate the bone or the muscles. Press a small piece of tissue paper on the wound to stop bleeding. Do not wipe off the blood with the tissue as it removes already coagulated blood from the wound and bleeding starts again. Empty the content of the
capillary into an Eppendorf tube that contains Queen's lysis buffer (QLB, Seutin et al. 1991), or a rubber sealed tube that contains pure ethanol. This is most easily done by either gently blowing the blood out of the capillary or more hygienically, using a pipette aid.

Figure 7. Blood sampling from chicks.

There are several ways to store the blood for genetic analyses; we recommend using QLB or ethanol. The DNA is preserved in both cases for many years, and it can be extracted using automated methods. Ethanol is cheap and can be obtained in most pharmacies. However, ethanol is inflammable and airline carriers might refuse to transport your samples stored in ethanol. Ethanol also evaporates easily, which means you have to make sure that the tube is tightly closed with the rubber sealed cap. QLB needs to be prepared in a lab before the field season (protocol after Yezerinac see Appendix 9), and aliquoted into 1.5 ml Eppendorf tubes. It’s not inflammable, does not need to be refrigerated in the field and is easier to handle in the lab and therefore our preferred buffer. If you use QLB make sure that you do not exceed the ratio of 25 µl of blood per 1ml of buffer. Otherwise the DNA will not be protected and may start degrading over time.

The sample tubes should be carefully labelled: ring number, date, location and sex/age (if known). Samples can be stored at room temperatures for several months, although a refrigerator is preferred. Ethanol samples are best stored at –20 ºC – +4 ºC for long term. Don’t store them below these temperatures, because the ethanol will freeze and the DNA might be damaged.

Tissue sampling: Tissue sampling is particularly useful for dead plovers or embryos. We usually take a tiny piece of leg muscle and preserve it in ethanol. A few mm³ tissues are enough. DNA in dead bodies degrades fast particularly in warm environments due to the work of enzymes such as DNAses. The best is to make sure that (some) DNA is preserved is by cutting the tissue with a sterile blade into small pieces so that the cells get smashed, and get in contact with ethanol which inhibits the enzyme activity. The storage conditions are the same as for blood.

Trapping with chicks

Equipment needed: traps, tea-sieve, bird bag, binoculars and/or spotting scope.

It is possible to trap parents with chicks up to about 2 weeks of age. First, you need to catch ALL chicks in a brood, and carefully place the chicks under a tea-sieve (or strainer) large enough for all chicks. The sieve needs to be fixed on the ground with 2-3 pegs. Second, place a trap (funnel or round) over the sieve as you would do with the nest (Figure 3e). You may cover the bottom 10-15 cm around the trap with mud (or plant leaves) from the outside to block the direct view of parents of their chick; leave the funnel entrance open, however. This tends to entice parents to enter the trap.

If you’re lucky enough to trap both parents, then you should keep the chicks in a warm place to
avoid overcooling, and release them at the same time as the parents. If one parent was caught and the other parent is around the family, you may release the chick(s) whilst measuring the caught parent, because the other parent will take over brood care. You should release ALL chicks at the same time in the location where you caught them.

Checking broods

Equipment needed: binoculars, spotting scope, notebook, GPS device, mobile hide or car

Chicks are precocial so that they often wander over kilometres from the nest. Therefore, it is challenging to establish whether the chicks have fledged or died. We recommend revisiting marked families every 2-4 days, and recording the number and sex of attending parents, and the number of chicks (Appendix 6).

If you capture a brood that has not been marked before, you can estimate the age of these chicks by using a formula (Székely & Cuthill 1999). Since tarsus grows approximately linearly until the age of 25 days (as opposed to body mass that initially drops, and then accelerates), linear estimates appear to be acceptable: \[ \text{AGE (in days)} = 2.520 \times \text{TARSUS (in mm)} - 48.341. \]

It is a good idea to spend at least 15 minutes with each family to count all chicks and to establish whether both parents are still attending the brood. Brood desertion may not be permanent, so you need a few visits to make certain of desertion by one (or the other) parent. Marking chicks with different colour rings can help to establish which chicks are still alive. Brood mixing does occur in the KP, thus it makes hard to separate mortality from adoption.

Broods can be efficiently checked and recaptured at night using a powerful spotlight. We also caught parents using the tea-sieve method at night. Most chicks, even those beyond 20 days, are brooded at night so that they are easy to count. In addition, capturing chicks at night appears less distressing than during the day. An extra bonus of captures at night is that next day the parents are not bothered about you, whereas chasing chicks during daytime may force the parents to move with their chicks to a different brood-rearing site. You need to work carefully at night: you must know your study site and the whereabouts of the plovers to avoid getting stuck, or squashing nests and families. Night time fieldwork has implications on safety: your best interest is to check this out.

Resightings

Equipment needed: binoculars, spotting scope, GPS device, notebook, mobile hide or car.

To build up a dataset on the movements of individuals, it is a good practice to note the location of colour-ringed plovers (Appendix 7). Every time you spot a colour-ringed individual, we recommend noting basic information as well, for instance on behaviour and possible pair-bonds.

A very useful summary file is ‘BirdRef’ (Appendix 8) that includes the identity (e.g. ring number) of all family members. In addition to captures and recaptures, you may also use unambiguous resightings to establish the identity of a family member. BirdRef can be linked to different files (e.g. nests, captures, behaviour), so this file is often the backbone of statistical analyses.

Notebooks

It is good practice to have two A5-sized notebooks for fieldwork (one for nest data i.e. Nest notebook, and one for captures i.e. Capture notebook), and a one-page-a-day diary. In the latter you may also note brood observations and resightings. In the diary, it is a good idea to record the major activities relevant to a given day (e.g. 5 May: 5.50 start fieldwork; 5.55 trapping at B1/2 nest, 6.15
measuring & ringing female at B1/2....). These notes can be essential when you need to clarify the circumstances of an experiment, or reconstruct the details of an important observation.

*********************************** Disclaimer ***********************************

Please note we will not take responsibility for any consequences of the use (or misuse) of this guide. You need to check the regulations and legislations in your country and where fieldwork is carried out. We did not deal with many essential conceptual and practical issues for successful fieldwork (e. g. research hypotheses, experimental design, experimental protocols, logistics): to overcome these hurdles you need to be innovative. Good luck!

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REFERENCES
Using a mobile hide in wader research

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We describe a mobile hide (or blind) that we designed for making observations of breeding plovers. We recommend it as being particularly suitable and flexible for studying waders in open habitats.

INTRODUCTION

Researchers often investigate the behaviour and ecology of waders using hides (or blinds). Ideally, these should be mobile since waders (or their nests) may be scattered, and observations may be conducted in several locations. For instance, the parents may leave their territory to feed, or the precocial chicks may move to distant areas. For these reasons, research workers often use motor vehicles such as cars, particularly all-terrain vehicles. However, these can be expensive; moreover their chassis can easily overheat and the noise of vehicles may disturb the birds. An additional source of disturbance may arise if researchers get out the vehicle, for instance to catch waders or to check their nests. To overcome the limitations of motorised vehicles, we developed an inexpensive and convenient mobile hide for our studies of Kentish Plovers Charadrius alexandrinus in Southern Turkey. For details on the study site and methods see Szekely & Cuthill (2000), Kosztolányi et al. (2003) and Lendvai et al. (2004).

We focus on using the mobile hide in breeding ecology and behaviour, although it might be worth considering applications in studying migratory shorebirds at stopover sites or coastal wintering areas. This hide is probably best suited to studies of waders in open terrain where the substrate is fairly hard.

THE MOBILE HIDE

The mobile hide had three main components: the frame and accessories, the wheels and the cover. First, the frame was made of 20 × 20 mm square-profiled iron (Fig. 1a). The frame ended in two forks at the front (see Fig. 1). The lateral branches of the forks were made of 500 × 20 × 5 mm iron (L × W × H). Two small horizontal plates at the rear stopped the frame from sinking in the mud. The observer sat on a wood bench of 1050 × 250 × 50 mm (L × W × H) that was put across the bottom bars. We also fixed a basket of 300 × 170 × 250 mm (L × W × H) on the frontal and lateral middle bars, and 2–4 hooks on the top bars to hold bags, traps, tripods and binoculars. The frame, the basket and the hooks were all painted with rustproof metal paint.

Second, one bicycle wheel of 635 × 38 mm diameter with standard road tires was screwed to each fork so that the frame stood in an upright position (Fig. 1). The observer moved the hide by gripping the bottom bars (either lateral or rear), and pushing the hide forward or backward. Before the hide was moved, the bench was pushed forward to let the observer walk.

Third, a hessian (or burlap) cover was made to cover the frame, and attached to it with straps (Fig. 1b). On each side of the hessian cover, there were two window slits; the top slits were used when the observer stood or walked, and the bottom ones were used when the observer sat down. When the slits were not in use, they were covered by roll-down hessian flaps from the inside and strapped to the cover. Three corners of the cover were not fully sewn down to the bottom: one slit of 1700 mm in the rear was left open for the observer to enter the hide, and two slits of 900 mm were left free for the wheels. The total weight of the hide was about 20 kg.

USING THE MOBILE HIDE

Mobile hides constructed according to this design were indispensable to our fieldwork. They were relatively inexpensive (total cost: about 200 €), and made locally using simple materials. They were very effective, and caused less disturbance (and probably less stress) than motor vehicles.

Four hides were spread over the study site so that they were stationed about one km apart. We only used one car to reach the hides, and to relocate them to different parts of the study site as necessary. We used the hides for the following tasks:

First, we searched for Kentish Plover nests and checked nests from the hide (Fig. 2). When an incubating plover was spotted we carefully approached its nest, and measured the eggs whilst we stayed inside the hide. Care was taken to avoid trampling the eggs. Plovers were tolerant of the approaching hide; for instance a female only left her nest when the hide was less than one metre from her. After the hide was pushed away from the vicinity of nests, the parents quickly resumed incubation.

Second, we trapped parents at their nest using funnel traps. The trap was put above the nest, and after the parent went inside the trap, the hide was slowly pushed over the trap and the parent was gently removed from it. Captured plovers were measured and ringed in the hide away from the nest. This procedure was less disruptive than standard nest-trapping when the researcher walks (or runs) to remove the trapped wader.
Third, we carried out various behavioural observations from the hides (Szekely & Cuthill 2000, Kosztolányi et al. 2003). Even secretive behaviours, such as brood attendance and courtships, were easy to record, and the plowers appeared to behave naturally. A telescope was mounted on a middle bar using a window-mount (Fig. 2b), and it remained there while the hide was being moved. The mobile hide was particularly handy when the plower(s) under observation waded across mudflats, shallow shores, ditches or thick vegetation, since the observer was able to follow the bird monitoring its behaviour without disruption using the hide. Note that particularly strong winds may make the hide unstable and difficult to manage and manoeuvre.

All things considered, we strongly recommend this mobile hide for wader researchers, since it is more economic and effective than motor vehicles, and causes less damage to the habitat. Unlike hides or blinds that are commercially available and designed for hunters, fishermen or photographers, our hides were easy to move around. Moreover, the hessian cover both shades the observer from direct sunlight, and also allows a breeze to blow through the hide and reduce discomfort in hot weather. If the hide is used in locations where the weather is cold and/or wet, we recommend using a waterproof canvas cover instead of hessian.

ACKNOWLEDGEMENTS
Ö. Karacabak (Turkish Ministry of Forestry and National Parks), and Drs S. Berberoglu, M. Özdemir, T. Yılmaz (Çukurova University, Adana) gave us logistic help. We acknowledge the comments of Drs Brett Sandercock and Humphrey Sitters on a previous version of the manuscript.

REFERENCES
**Appendix 2.** Nest records. First, you need to note all data for a given nest on a separate page (or half-page) in your Nest notebook. These will include nest location, egg measurements, dates of nest checks and nest fate. Second, you may type these data in a spreadsheet that looks like this:

<table>
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<tr>
<th>Year</th>
<th>Site</th>
<th>Nest ID</th>
<th>Latitude</th>
<th>Longitude</th>
<th>Found date</th>
<th>Laying date</th>
<th>End date</th>
<th>Fate</th>
<th>No. chicks</th>
<th>Clutch size</th>
<th>L1</th>
<th>B1</th>
<th>S1</th>
<th>L2</th>
<th>B2</th>
<th>S2</th>
<th>L3</th>
<th>B3</th>
<th>S3</th>
<th>Observer</th>
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</tbody>
</table>

YEAR
SITE – it is a good practice to divide the study site in small units that are refereed as 'sites'
NEST ID – give consecutive numbers to each nest for a given site
LATITUDE – GPS coordinate; use UTM coordinate system, if possible
LONGITUDE – GPS coordinate
FOUND DATE – the date the nest was found
LAYING DATE – the date of the laying of the last egg (either known or estimated using floatation stages)
ENDDATE – last date when the nest was checked
FATE – hatched/failed for one reason or the other
NO. CHICKS – number of chicks that hatched
CLUTCH SIZE - maximum number of eggs
EGG LENGTH (L1, L2 & L3) – length of each egg as measured using sliding calliper
EGG BREATHE (B1, B2 & B3) – breath of each egg as measured using sliding calliper
EGG FLOATATION (S1, S2 & S3) – floatation stage of each egg, if laying date is unknown (separately for each egg)
OBSERVER
COMMENTS
Appendix 3. Nest summary. It is often helpful to make a summary table on the last few pages of Nest notebook. This allows you to get updated how many nests you found, when the expected hatching dates are, and how many of nests failed or hatched.

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<th>Hatching date</th>
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<th>Comments</th>
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<td>2</td>
<td>412</td>
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<td>4</td>
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Site: Sand dune

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<th>22nd day of incubation</th>
<th>Hatching date</th>
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### Appendix 4. Capture notebook. You may use this table for both your field notes and your spreadsheet file.

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<th>Weight</th>
<th>Wing length</th>
<th>Tarsus length</th>
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</table>

RING – metal ring number  
YEAR  
SITE  
NEST ID – same as in Nest file (you may use negative numbers for broods that were found after hatching)  
SEX – M, F, J, for adult male, adult female and chick, respectively  
DATE – date of capture  
TIME – time of capture  
WEIGHT – body mass (g)  
WING LENGTH – measured by stretching the right wing (only adults and chicks older than 3 weeks, mm)  
TARSUS LENGTH – the length of right tarsus (mm)  
CODE – colour ring code in the form XX.XX | XX.XX  
OBSERVER - ringer  
COMMENTS – optional extra information about the bird

Make sure you do not duplicate the same information in different files. For instance, if nest coordinates are registered in the Nest file, there is no point including nest coordinates in the Capture file.
Appendix 5. Measurements of adults and chicks

In case of wings and tarsus we usually take the measurements of both right and left limb. This can give you information about the asymmetry of the birds, but serves also as a control to check your measurements.

Tarsus

Equipment needed: Vernier or dial calliper, notebook

We use the following technique according to Redfern and Clark (2001).

Figure A5.1. The minimum tarsus method. Image from Redfern and Clarke (2001).

There is always some variation in tarsus measurements between people. To keep this variation at a minimum it is important to keep bird and leg in a standardised way and know exactly what you want to measure.

We measure the minimum tarsus of plovers. If you do it for the first time, take a moment of time to feel for the tarsus bone with your finger tip, before applying the calliper. Then hold the bird firmly in your hand, with the head between index and middle finger in the so called ringer’s grip. The leg you want to measure should be aligned parallel to the main axis of the body. Tarsus and tibia should be in an angle of 90° as shown on the picture. The leg and three sides of the calliper (ruler and the two brackets) should form a rectangle.

Fix the tarsus using your thumb and your ring finger. Open the calliper and press the bracket until the tarsus is firmly between the adjustable and firm bracket. Check the position of the bone, if it still as shown in Figure A5.1 you are fine, if not readjust the leg. Then press the adjustable bracket gently until you feel some resistance by the end of the bone. Note the measurement to the nearest 1/10 mm. Repeat the process twice more and take the mean of your measurements.

Measuring the tarsus needs some practice. The point of resistance is usually the source of variation between measurements. This might lead to differences of up to 1 mm between people. At a total length of 25-30 mm of adults this is quite a lot. Remember that the tarsus length will not change in full grown adults, so the variation is entirely due to measurement variance. If you recapture adult birds later in the season, compare your measurements with the previous ones. An accuracy of 0.2-0.3 mm is reasonable. For chicks measurement errors can be an even bigger problem, particularly if you want to measure growth rates.

As usual be considerate with the bird, you don’t want to break it’s leg. Also don’t stress the plover to long by practising exhaustively to get the exact measurement. After some practise each measurement should not take more than two minutes.
Wing

Equipment needed: Wing ruler, notebook

Figure A5.2. Wing measurement using the maximum chord method. Image taken from Redfern and Clarke (2001).

We use the maximum chord method as shown in Redfern and Clarke (2001). As with tarsus the position of the wing and bird are important. Hold the bird as shown in Figure A5.2. Open the wing, straighten the primary feathers and carefully slide in the ruler as shown. Fold the wing back that it is as close to its natural resting position as possible. Again, the position of the wing in regards to the body is essential. Using your right index finger straighten the alula. Make sure that the alula falls into a line that the curvature of the wing is reduced (Figure A5.2). Take the measurement of the second primary, that is the longest feather if not worn or replaced during moult. Make sure that the primaries are not broken or heavily worn. If they are make a note. Again, if you measure the wing length for the first time repeat the procedure to get an idea about your measurement error.

Weight

Equipment needed: Pesola spring balance, cone (different size for chicks and adults), notebook

Figure A5.3. Cone (in red) for measuring weight using spring pesolas.

Weight measurement is important because it can tell a great deal about the conditions of the birds. Females can sometimes carry eggs which you can feel by gently pressing their belly. (If you trap on the nest they shouldn’t because you should not catch them before clutch completion!)

We use pesola spring balances to measure plovers, but it also possible to use electronic balances. Adult Kentish plovers have a body weight of 35 – 45 g, so a 50 g pesola spring is appropriate. A useful device to hold the bird during measuring with a pesola is a plastic cone that can be hung to the clip of the balance. Calibrate the spring before measuring the first plovers. When you are read gently force the bird into the cone with the feet stretched out towards the tail and the head pointing
Towards the narrow end of the cone (as shown in Figure A5.3). If you position the bird as shown, it won’t be able to struggle. Never hold the balance by the tube, but always at the hook or loop and make sure that cone and bird hang freely. For chicks you will need a smaller cone. Note the weight. Regularly clean the cones after a couple of days measuring, since the birds often defaecate in this situation.

Fat

![Fat scores. The fat distribution is shown in black. Image taken from Redfern and Clarke (2001)](image)

Fat provides an indicator about the condition of birds. To measure it, hold the bird with its feetsing pointing towards you. With your free hand slightly extend its neck, that the furcula is visible. The furcula is located at the beginning at throat, indicated by the black triangle in Figure A5.4.1. Blow the feathers at the furcula aside. You need good light conditions to see the fat, especially if there is no or very little fat. Note the deposit according to the figure. Breeding birds usually have fat scores between zero and four.

Bill

Equipment needed: Calliper, notebook

This is an optional measurement that you may want to take. There are several measures that can be taken including bill depth and length. We usually measure bill length, which can in some waders identify sex or race. We measure it from the tip to the start of the feathers using a calliper to the nearest 1/10 mm.
### Appendix 6. Variables recorded for brood encounters.

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<th>Year</th>
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<th>Time</th>
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<th>Chicks</th>
<th>Latitude</th>
<th>Longitude</th>
<th>Habitat</th>
<th>Observer</th>
<th>Notes</th>
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**YEAR**
SITE – see nest file
BROOD ID – brood identifier; negative signs indicate that the brood hatched from a nest we did not find
DATE – date of re-sighting
TIME – time of re-sighting
PARENT – number and sex of parents (4 – both parents, 3 – only male, 2 – only female)
CHICKS – number of chicks
LATITUDE – GPS coordinates; UTM coordinates are often more useful than other types
LONGITUDE – GPS coordinates; UTM
HABITAT - sensible description of habitat
OBSERVER
COMMENTs – notes & additional details
Appendix 7. Resightings of colour-marked plovers.

<table>
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<tr>
<th>Year</th>
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YEAR
SITE
LATITUDE – GPS coordinates
LONGITUDE – GPS coordinates
DATE – date of re-sighting
TIME – time of re-sighting
CODE – colour ring combination
SEX – sex of observed plover
OBSERVER
COMMENTS – notes about behaviour
**Appendix 8.** BirdRef: a useful summary of family members. Note that only those families are included that have at least one marked member. Negative nest numbers refer to broods that hatched from unknown nests.

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Appendix 9. Protocol to prepare Queens Lysis Buffer (after Dr Stephen Yezerinac, Queens University Kingston, Canada)

Recipe for 1X strength

0.01 M Tris-Cl
0.01 M NaCl
0.01 M EDTA
1% (w/v) n-lauroylsarcosine (or n-lauroylsarcosine sodium salt which dissolves better)

adjust to pH=7.5 using NaOH

DO NOT AUTOCLAVE AFTER ADDITION OF n-lauroylsarcosine, WHICH BUBBLES-UP.

For example, to make 500 ml, enough for 500 blood samples: add to 400 ml sterile distilled water, 0.61 grams Tris-Cl, 0.3 grams NaCl, 1.7 grams EDTA, 5 grams n-lauroylsarcosine; stir until dissolved, then adjust pH to 7.5 using Sodiumhydroxide and bring volume up to 500 ml by adding sterile distilled water. Dispense into 1 ml aliquots and use.