





LIFE 13 BIO/UK/000315

LIFE Waders for Real

A.4 Production of protocols for Habitat, predator and wader monitoring.

Created: Andy Hoodless 14/08/14 Modified: Andy Hoodless 02/03/15 Modified: Lizzie Grayshon 11/02/2020

Abstract

Monitoring protocols for all field work conducted during the LIFE Waders for Real Project. Monitoring of breeding waders, predators, habitat and ecosystem services, detailed monitoring was undertaken to measure change in response to habitat management.

Project aims

This project seeks to reverse the decline of breeding waders in the Avon Valley, a river floodplain of high biodiversity interest, part of which is designated as an SPA. In 1982 the Avon Valley constituted one of the top eight lowland wet grassland sites in England for breeding waders. Since then four surveys at 6-7 year intervals have shown a dramatic decline in numbers of breeding waders, mirroring trends seen across Europe. Numbers of Northern lapwing *Vanellus vanellus* pairs have fallen from 208 in 1990 to 71 in 2010. Pairs of redshank *Tringa totanus* have dropped from 117 to 22 and common snipe *Gallinago gallinago* from 29 to one.

Monitoring of lapwing breeding success in the Avon Valley for the last eight years has shown that productivity was too low for maintenance of a stable breeding population, owing to high nest predation. To halt the decline of lapwing and redshank we urgently needed to intervene to improve breeding success, which in the longer term, should lead to increases in breeding density.

The objectives were, through a unique combination of habitat restoration and innovative targeted, seasonal exclusion of predators to:

- 1. Increase lapwing numbers in the Avon Valley, through the novel approach of creating strategic 'hotspots' of optimum habitat with reduced predation pressure.
- 2. Increase numbers of lapwing chicks fledged at 'hotspots' to the point where breeding densities become sufficient to enable lapwings to better fend off potential predators on their own.
- 3. Halt the decline of redshank in the Avon Valley by increasing productivity.
- 4. Create conditions to encourage snipe to return to breed.
- 5. Using a new approach called Planning for Real to deliver lasting conservation action.
- 6. Demonstrate how far habitat manipulation can be used to push the balance in favor of waders rather than predators. We will assess predator behavior in manipulated landscapes.
- 7. Demonstrate the most appropriate techniques for the efficient exclusion or reduction of predators and quantify any benefit or problems associated with predation control.
- 8. Quantify the costs of different techniques for increasing wader breeding success and the timescale over which this translates into higher wader numbers.
- 9. Monitor the effects of restoration for waders on other key elements of floodplain biodiversity, particularly the flora, invertebrates and wintering wildfowl.

Monitoring design

The aim of the monitoring outlined in the protocol is two-fold: to measure change at the four hotspot sites where the most intensive management for waders will take place and to compare habitat conditions and wader breeding success against four comparison sites.

Breeding wader surveys should be conducted on all suitable sites to give a baseline of breeding wader numbers.

Wader breeding success, predator monitoring and detailed vegetation monitoring should be undertaken on key hotspot sites, as well as wader breeding success and vegetation monitoring undertaken at comparison sites.

1. Wader pair surveys

During second and third weeks of April, walk all fields at all sites to determine the locations of lapwings, redshank and snipe. Walk to within 100 m of every point in all fields so that no waders are missed (although take care not to cause undue disturbance to nesting lapwings). Mark the positions of birds on 1:10,000 scale maps and try to determine sex for lapwings. Note paired birds and try to distinguish from feeding flocks. Keep a note of the time that each field is entered so that the time spent walking/watching each field can be extracted for use in analyses. Record the presence of all corvids (separating carrion crows, rooks, jackdaws and magpies), gulls, raptors, and ducks, geese, swans and herons. Distinguish birds flying over from those perched or feeding in the field (See Annex 1 Monitoring breeding waders field sheet).

In second and third weeks of May conduct an additional early morning survey of each site. Use the same methodology as above; walk to within 100 m of every point in all fields, fill out a survey sheet and mark the positions of all birds seen on a map.

On both surveys, fill out the 'Sward and land use' sheet. For each field note field type, number of livestock, % cover of standing water, rush, sedge, thistle/dock/nettle, and marsh marigold. Record vegetation height at 20 randomly selected point per field (Annex 3.1 Sward and land use field sheet).

1.1 Lapwing productivity surveys

Breeding productivity should be assessed from regular visits to fields with lapwing present, at approximately 10-day intervals. Surveys should commence in late March and span the lapwing breeding season. Do not visit occupied sites on wet or particularly cold days. These visits do not need to be made in the early morning. The surveys will yield information on site occupancy and overall productivity per site (Annex 1 Monitoring breeding waders field sheet).

On all visits to occupied fields a visit sheet should be completed, and the surveyor should:

- 1. Note time of arrival at the field. Record numbers and species of all corvids, gulls, raptors and herons on arrival. Distinguish birds on the field or on the field boundary from those flying over.
- 2. Make careful observations from several vantage points at the edge of the field to ensure that the whole of the field is scanned. Record the total number of adult lapwings (where possible, distinguish males from females), number of sitting adults, number of alarm-calling females and number of young. Where possible record the brood sizes and the age class of any chicks observed. Make sure to mark the positions of birds, nests and broods on large scale maps.

If it is not possible to make an accurate count of the number of birds by scanning from the field edge, the field should be walked until all areas can be seen. Once returned to a vantage point, watch carefully for birds walking back to nests.

- 3. Note any raptors flying over or stopping in the field during observations.
- 4. Record the time that you leave the field.

1.2 Nest finding

On each visit to a field, you should attempt to locate nests using the method detailed below. To minimize disturbance, nests should only be approached when first located or when clutches are

estimated to have failed or hatched. Care should be taken to avoid drawing the attention of potential egg predators to nest sites (Annex 1.2 Nest finding and monitoring field sheet).

All nest monitoring is done under Natural England Nest disturbance permit, under the Wildlife and Countryside Act 1981, assigned to each individual researcher.

- 1. Scan for sitting birds. Females undertake most of the incubation, but males do incubate when the female is off the nest feeding. Typically, you will see a male stood alert within 100m of the sitting female. Females are quiet and secretive when nesting: watch for birds that appear to be agitated and creeping back to nests.
- 2. Once you have spotted a sitting bird, pick a point (distinctive tree, bush, house, etc.) on the horizon as a sighting line. Then judge the distance across the field and look for any distinctive features close to the nest. Once you are confident that you have located the nest and have a clear direction, walk to the nests (if there are two of you, the other person should stay at the vantage point to guide on a mobile phone or with walkie-talkies). Be aware that your perspective will change as you walk to the nests and keeping an accurate visual mark on the location the bird gets up from is essential. If there is more than one nest in a field, the first can be temporarily marked with a stick a few feet away while the next is located. Be aware that there may be other nests that you have not seen, so watch where you are walking, especially as you approach the location where you believe the nest to be!
- 3. When you find a nest (clutch size is typically 4, sometimes 3 or late in the season 2), use a GPS to record its location and note the date and time. Measure the length and breadth of each egg with calipers and record the mass. Take extreme care to avoid breaking any eggs, especially if your hands are cold. Replace each egg in the orientation found. Insert a temperature logger into the middle of the nest between the eggs. For speed, if all the eggs are very similar size and shape, measuring and weighing just two of the clutch may suffice, particularly if you have several clutches to measure.
- 4. Record vegetation height (to nearest cm) on the edge of the nest cup and at 4 points at 1 m around the nest. Estimate the % bare ground in a 1 m square quadrat around the nest. Take a photo of the nest from directly above, leaving a 50 cm border around the nest so we can estimate vegetation types later. No more than about 5 mins should be spent at the nest.
- 5. Try to minimize disturbance to soil/vegetation around nests. Do not disturb birds off nests on very cold days or if it is drizzling/raining. When leaving the nest, continue to walk past it and loop back to your vantage point rather than walking back on the exact route by which you approached the nest.
- 6. When you get back to the vantage point make a sketch map with notes on the viewing point, sighting line, field boundary features and landmarks close to the nest so that it can be relocated easily from the vantage point on the next visit.
- 7. Do not spend more than 10 mins looking for a nest, especially if it is cold and/or wet. If you walk out and are unable to find it, return to your vantage point or another vantage point and watch the bird walk back to the nest. Let her sit for 10 mins, note any further features distinguishing the nest site and then try again. If you are convinced there is a nest but you cannot find it, try putting a stick or stone where you think the nest is and then retire. Use this marker to gauge where the nest actually is.
- 8. Be aware that the male makes 3 or 4 scrapes within an area of a few metres and the female chooses one in which to lay. Hence if you find an empty scrape with no nest lining, you may be close to the actual nest.

- 9. Egg measures and weights can be used to estimate the stage of incubation of a clutch because eggs lose mass during incubation. This is useful because you then know approximately when a clutch is due to hatch. A list should be kept of when clutches are due to hatch, as you will need to visit soon after hatching to mark chicks.
- 10. First clutches tend to be fairly synchronous in late March/early April, but as clutches fail birds will start re-nesting. They typically take about 8 days to re-nest after losing a clutch. Birds may re-nest twice in a season, i.e. have up to 3 nesting attempts. Hence you are likely to have birds nesting into June.





Left: Typical clutch of 4 lapwing eggs in a grass field. Note base colour can vary from cream, as here, to green. Right: Clutch of redshank eggs – note concealed in a tussock and brown rather than black blotches on the eggs.

1.3 Nest monitoring

Temperature loggers will provide data on time of nest failure or hatching, avoiding the need to make frequent, repeat visits to nests to obtain survival data. As far as possible, you should aim to go back to nests within a day or two of the estimated hatch date. However, as you are likely to be returning to sites for other reasons (to look for more nests, to follow chicks, etc.), all occasions on which the nest is monitored should be entered on the nest data sheet (see Annex 1.2 Nest finding and monitoring field sheet). This makes it easier to identify the timing of nest outcomes when either interpreting logger data or when the logger data is somehow compromised.

1. On arrival at a site scan for the nest at a distance by using binoculars or a telescope to check for the presence of an incubating adult. If an adult is still sitting, the nest is obviously still active (although check that the bird is not brooding small chicks). If there is no bird sitting, approach the

nest site and check for signs of predation or hatching (see 3. and 4.). Consider taking photos of any remains that you find.

- 2. Retrieve the temperature logger, note the time that it is removed from the nest and download the data later the same day. If the temperature logger has been cannot be located in the nest cup, carefully search up to a 10 m radius around the nest.
- 3. At some predated nests it is possible to identify the nest predator from signs left at the nest. Eggs predated by corvids have a hole punched in the side. Those taken by foxes or badgers tend to be crushed to small pieces. Those taken by small mustelids tend to have tooth marks in the shell. Badgers will completely destroy the nest, foxes and corvids lift the eggs cleanly, leaving the nest lining intact. If in doubt, record details and, if possible, take photos of the nest and any egg remains. It is only possible to identify the nest predator with confidence in a small percentage of cases, so note exactly what you can see. Corvids and mammals often remove the eggs leaving no trace of shell near the nest, but mammals may leave footprints or scats.
- 4. Successful nests always have small eggshell fragments, c.1-2 mm in size, in the nest lining (the adults remove the eggshells, but these chippings are from where the chicks break out of the shell). Always search the nest lining carefully.
- 5. Nests may be hatching at the point of checking, if this is the case any dry chicks can by ringed and tagged (see 1.5 Monitoring lapwing brood survival).





Two examples of lapwing eggs predated by corvids. Left: one eggshell left beside a nest. Right: two eggshells carried about 50 m from the nest.

- 5. When a nesting attempt has finished, record the 5 vegetation height measures as before, % cover of bare ground and the composition of vegetation within a 1-m quadrat over the nest (% cover of grass, sedge, rush, dicot and herb). At the same time record these measures at a paired random point within the same field 100 m from the nest. To do this take a random compass direction, pace 100 m and then take the measurements. Mark the random location on a map.
- 6. Be alert to signs of predation of adult lapwings by scanning around predated nests. Note any carcasses and the likely predator.



Remains of a predated adult lapwing. Note the primary feather shafts have been chewed through and the carcass removed indicating predation by a fox. Raptors pluck the feathers, leaving the wing feather shafts intact and tend to leave the carcass upside down with the sternum notched.

1.4 Use of nest protectors

Deploy nest protectors on half the nests monitored at hotspots sites.

- 1. Locate nests as early as possible, ideally when eggs are still being laid.
- 2. As soon as the clutch is complete, place a cage about 5-10 m from the nest.
- 3. A day later move it 2 m from nest.
- 4. Two days later, put it over the nest.
- 5. Watch the female and ensure that she returns to the nest within 40 mins. Most birds accept the cages within 20 mins. If it is difficult to see the female returning to the nest site then when putting the cage over the nest turn one egg around the wrong way. This will tell you if the nest has been visited again, as the female will correct the egg's direction.
- 6. About 1 in 8 birds will not tolerate the cages, so do not persist with it if a bird will not accept the cage after 40 mins.
- 7. DO NOT USE WITH REDSHANKS, AS THEY TAKE OFF VERTICALLY FROM THE NEST.

1.5 Monitoring lapwing brood survival

During visits from late April to July, careful observation of adult behavior should be made to determine whether or not each pair of lapwings has managed to hatch a clutch of eggs (Annex 1.5 Chick finding and monitoring field sheet).

1.5.1 Chick observations

You will not locate every nest; hence it is important to establish as a minimum for each pair of breeding lapwings whether they hatch a clutch of eggs and whether they fledge a brood.

Females are typically silent and elusive when nesting, but vocal when they have chicks. Their alarm-calling behavior when they have chicks is distinctive and can be used to estimate the period for which broods survive. By carefully noting the combination of pair location, alarm-calling behavior and any chick observations on each visit, it is possible to estimate brood survival. Ideally, broods should not be disturbed more than once a week.

For each pair of lapwings and redshank on each visit, determine adult behaviour and alarm calling. On each visit record whether or not you think the pair has chicks. Aim to visit each field with lapwings and assess the status of each pair/brood once every 7 days. Note the start and end time of observations at every field and record all corvids, gulls, raptors and herons in the field or passing over.

For any broods seen, attempt to count the number of chicks and estimate their age to the nearest week. Chicks fledge at around 35 days but can fly short distances soon after 20 days.



An example of a camouflaged day-old chick.



An example of 2 camouflaged 25 - 30 day old chicks.

1.5.2 Catching, ringing and radio-tagging chicks

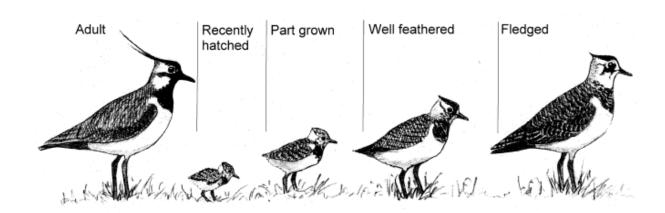
If chicks are not found at point of hatching in the nest cup, they can be caught using a similar method as nest finding. After careful observation approach the area where chicks have been seen, move quickly to get to the area then approach with caution as chicks can be well camouflaged. Where possible work in pairs, with one at a vantage point guiding the other in over radios.

Catching, ringing and tagging of chicks is done under BTO license, held by each researcher with specific endorsements to colour ring and radio-tag chicks.

Chicks can be ringed with a BTO metal ring from the day they hatch. Chicks can be colour-ringed from 20 days of age. Re-sighting of colour ringed chicks will provide data on chick survival rate and fidelity to breeding sites of fledged chicks in subsequent years. Where possible, please read and note colour ring combinations when seen.

Take biometrics from all chicks captured on all occasions – bill and tarsus length (to 0.1 mm), weight to 0.1g. For chicks older than 20 days, measure wing length and note the extent of primary growth (third, two-thirds, fully grown).

A sample of lapwing chicks are radio-tagged to obtain more accurate data on brood movements and on fate of chicks that perish. One chick is randomly selected from each of 30 broods and is radio-tagged shortly after hatching with a 0.4g, 30 day life, transmitter. Chicks are radio-tracked every 3 days by triangulation from field edges and will only be approached closely where death is suspected; if tags indicate a static signal, or no movement, or a very large movement since the previous day. Fixes should be recorded on 1:10,000 scale maps with the time of the fix, enabling average distances travelled and time spent in different habitats to be calculated subsequently using a GIS (See Annex 1.5.1 Chick radio-tracking triangulation sheet).



Guide to ageing lapwing chicks. Recently hatched, part grown, well feathered and fledged equate roughly to less than 1 week, 2 weeks old, 3 weeks old and 28-35 days respectively.





Lapwing chicks within a few hours of hatching. Note the white 'egg-tooth' which is retained for c.24 hours. Note size of foot in relation to body.



Lapwing chick c.7 days old. Again, note size of foot relative to body – body larger relative to foot now. White stripe on nape can be helpful when trying to locate chicks.





Two lapwing young c.28 days, wings fully feathered, crest starting to form.



Recently hatched redshank chicks. Note dark heads and striped bodies.

1.6 Chick diet and foraging habitat

1.6.1. Diet

Collect faecal samples when handling chicks. All the faeces from one brood can go in the same sample tube. Label with site, brood number/chick ring numbers and date. Add 70% ethanol to the sample tube, to preserve the sample, as soon as you return to the office. Re-sample broods at different ages where possible, provided the same brood is not disturbed again within 7 days of the previous capture (see Annex 1.5 Chick finding and monitoring sheet).

The feacal samples can be used to investigate diet in relation to chick age and type of foraging habitat. The faecal samples can be examined under a microscope to identify any invertebrate fragments present. Where possible the fragments can be classified to order or family level and overall numbers and weight of prey ingested can be calculated.

1.6.2 Foraging habitat

Data on foraging habitat is recorded as part chick monitoring (see Annex 1.5 Chick finding and monitoring sheet). Data should be collected on foraging habitat by taking measurements at the point where chicks are caught/observed foraging. Take 5 vegetation height measurements and 5 penetrability measurements. Assess % cover of bare ground at 3 separate 1 m quadrats around the foraging area. Assess the % cover of other vegetation (grass, sedge, rush, dicot, herb and weed) in a 3m quadrat over the foraging area.

At the same time record these measures at a paired random point within the same field 100 m from the foraging area. To do this take a random compass direction, pace 100 m and then take the measurements, use a hand-held GPS to record the location of the random point.

1.7 Colour ring sightings

Lapwing chicks are fitted with BTO metal rings when first located and then colour rings, to allow individual identification in the field, once they are c.20 days old. This will provide data on chick survival rate and fidelity to breeding sites of fledged chicks in subsequent years. Any adult lapwing caught are also colour ringed. Each bird colour ringed is fitted with a unique colour combination so that when the bird is observed in the field it can be individually identified.

Any sightings of colour ringed lapwing chicks or adults are recorded (see Annex 1.7 Lapwing adult colour ring sighting form, chick sightings recorded on chick form see Annex 1.5 Chick finding and monitoring sheet).

1.8 Catching and tagging lapwing adults

A sample of lapwing adults are caught in order to fit Pathtrack nanoFix-GEO-RF tags. These are remote download GPS tags and weight c.4.5g. The tags are solar charged and can deliver a minimum of 1-2 accurate locations a week throughout the year. They should function for 3-4 years. They will provide unbiased data on dispersal of first-year birds and selection of breeding sites between years, a factor that might prove important when making decisions on priorities for species conservation at a landscape scale.

Nest trapping, ringing and tagging is done under a BTO licence, with special endorsements required for nest trapping, colour ringing and tagging held by each researcher (=Annex 1.8 Lapwing adult capture field sheet).

Adult lapwing can be caught using spring traps on the nests, this is only attempted on nests which have 7 days or less to the predicted hatch date. The eggs are removed from the nest, safely stored to minimise chilling, and replaced with fake eggs. The spring trap is then placed over nest and pegged into the ground. The netting is camouflaged with vegetation and the trap is left for up to 30 minutes while being viewed from a nearby location. Once the adult is caught it is removed straight away, the eggs are replaced and the bird is ringed, tagged and processed away from the nest location, before release. If the adult does not approach the nest within 30 minutes of the trap being set the trap is removed and the eggs are replaced.

2. Predator monitoring

2.1 Avian predator monitoring

From early April to early June, carry out 1 hour watches over each field where lapwings are present. When you first arrive, record every corvid, gull and heron seen within the field or perched at the edge of it. Use the standard BTO codes for each species as during the wader surveys. During the next 5 minutes tally all corvids, gulls or herons flying over the field and at the end of the 5 minutes scan the whole field and again record all corvids, gulls and herons within it or perched at the edge. Continue at 5 min. intervals. The aim is to estimate the maximum number of potential predators using the field per hour and the activity of predators over the field (See Annex 2.1.1 Avian predator monitoring field sheet).

In addition, record each raptor sighting separately. Record each occasion that lapwings chase off a potential avian predator and, for incubating lapwings, record when they come off the nest, for how long and the cause.

2.2 Avian predator monitoring - Revised protocol 2016

In the revised protocol a focal bird is added, either a bird incubating a nest or with a brood (See Annex 2.1.2 Avian predator monitoring revised field sheet).

In the revised protocol the survey period is extend to 1 hour 30 minutes survey, during these surveys a focal nest or brood of chicks is identified. As in the previous protocol (2.1), at 5 minute intervals tally the total numbers of corvids, gulls, herons and raptors perched in or on the boundary of the field.

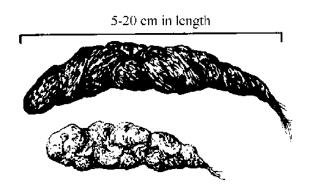
Record all avian predators (corvids, gulls, herons and raptors) flying across the field and the behavior of the focal adult lapwing to each predator, along with the total number of other lapwings that respond. Estimate the horizontal distance and height of the predator from the focal bird when the focal bird responds. If the focal bird does not respond record the horizontal distance and height at the point when the predator is closest to the focal bird. The duration of any chase of the predator by the focal bird should be recorded to the nearest second using a stopwatch.

2.3 Foxes and other mammals

Fox activity-abundance can be assessed by the four methods: scat counts, dawn and dusk watches spot-lamp searches and trail camera traps.

2.3.1 Scat counts

Approximately 2 km routes following boundary features and livestock tracks should be devised at each site. These should be walked three times, in the first week of April, May and June. All scat positions are marked on a map and all scats are covered or removed. Badger latrines should also be noted (see Annex 2.3.1 Fox scat field sheet).





Typical appearance of fox scats. They typically have one twisted end by can vary considerably in colour and consistency dependent on what the fox has been eating – they often contain beetle elytra and rabbit remains but can also contain site specific prey such as berries or fish remains.

2.3.2 Dawn and dusk high seat watches

These should be made from portable high seats at each site and last 90 mins, commencing either 30 mins before sunrise or 60 mins before sunset. Fixed vantage points will be selected, and arcs of view measured. There should be at least four watches per site from April to mid-June. During each watch any sightings of potential lapwing predators are recorded, if the predator is a fox additional information, such as gender or distinguishing features, is noted (See Annex 2.3.2 High seat count field sheet).

2.3.3 Trail cameras

Trail cameras should be used on hotspot sites to determine mammalian predator occupancy and abundance. 10 cameras are set on each hotspot site and switched between paired locations every two weeks. Consequently, a total of 20 locations are used at each site. Cameras should be set at access points to best pick up mammalian predators; gateways, bridges etc. When camera trap locations are switched the SD card in the cameras are changed. This means the photos on the cameras can be backed up and then reviewed for predator sightings every two weeks. This also means recent camera trap images of foxes and mink can be passed to local keepers to improve lethal predator control almost in real time (see Annex 2.4 Camera trap form).

2.3.4 Trail cameras on temporary electric fences

As part of non-lethal predator control temporary fences should be erected early spring, see *Guidance* note for farmers on electric fencing to protect wader nests and chicks. Fences should be checked and maintained during the breeding season twice a week, until taken down (see Annex 2.5 Electric fence checking sheet). Two trail camera traps should be deployed on each fence on opposite corner posts viewing the inside of the fences. These cameras can be used to record any predators that breach the fence.

3. Vegetation recording

3.1 Sward and land use

This information should be collected twice each year for each field (at the same time as the wader pair survey (see 1. Wader pair survey). Field type and number of livestock in each field are noted. The % cover of standing water, rush, sedge, thistle/dock/nettle and marsh marigold are recorded. Vegetation heights are recorded at 20 random points (Annex 3.1 Sward and land use field sheet).

3.2 Detailed vegetation quadrats

Vegetation in quadrats at fixed locations in hotspots and comparison sites should be surveyed to species level in June each year (see Annex 3.2 Vegetation Quadrat field sheets).

3.3 Vegetation records - penetrability

As well as the above surveys additional vegetation surveys should take place, in April, May and June in each field with waders in. A W-shaped transect is followed across each field in order to sample all parts of the field in a representative manner. At 30 points on the transect vegetation type (grass/rush/sedge/dicot/dock/thistle/nettle), vegetation height and soil penetrability (measured using a penetrometer) is recorded (see Annex 3.2 Vegetation record – penetrability field sheet).

4. Invertebrates Communities

4.1 Ditch Condition

4.1.1 Dragonfly and damselfly surveys

Dragonfly and damselfly surveys should be conducted along sections of the river and ditch systems on the four hotspot sites in spring/summer. In total30 transects are surveyed, six on the main river and 24 along ditches. One survey should be carried out at each transect site each month from June toSeptember. Species and numbers of dragonflies and damselflies are recorded along the transect (See Annex 4.1.1 Dragonfly and damselfly surveys, ditch environment and vegetation).

4.1.2 Ditch vegetation

Features of ditch vegetation are recorded alongside the dragonfly and damselfly community. (See Annex 4.1.1 Dragonfly and damselfly surveys, ditch environment and vegetation). In each zone the main vascular plant species present are recorded and their cover is estimated on the DAFOR scale (Dominant/ Abundant/ Frequent/ Occasional/ Rare). In each zone the % cover of bare ground, litter, grasses and bryophytes are also recorded. The % cover of open water in the ditch should be estimated for the whole transect area. In the August survey only ten vegetation height measures are taken in each zone (when vegetation height is at it's maximum). The % Cover of trees rooted inchannel or on the bankside should be estimated.

4.1.3 Ditch physical environment and water chemistry

Physical and chemical attributes of the ditch/river sections surveyed for dragonflies and damselflies are recorded mid-season (See 4.1.2 Annex Ditch condition field sheet). These attributes should be recorded at five sample sites which are located at c.25m intervals at each ditch/river section. At each sample site a water sample should be collected using either a hand-held bottle or one attached to a pole. Sample bottles are marked with a ditch identifier and date. Samples will be amalgamated for each transect location for analysis. In addition, at each sample site other physical and chemical attributes are recorded:

- Water and silt depth to the nearest 10 cm.
- Conductivity to the nearest 10 μ Scm-1 (measured on a sub-sample of water in the field using a portable meter).
- pH to the nearest 0.1 (measured on a sub-sample of water in the field using a portable meter).
- Turbidity on a five-point scale based on 1 = gin clear, 3 = base of ditch just visible at 50cm, 5 = visibility < 10cm.
- Water colour is measured by looking into the ditch at a white marker on the end of a ranging pole, rather than taking water out and looking through a bottle, but this is only possible if turbidity is low. Peatiness can be recorded on a five-point scale based on 1 = gin, 2 = lemon juice, 3 = white wine, 4 = medium black tea, 5 = black coffee.
- Bank slope (based on the angle from the bank top to the water's edge, irrespective of variations between the lower and upper sections) is noted.
- Profile under water (the angle of the bank from water level to about 20cm depth) is recorded. This is an indicator of the amount of shallow water at the margins.
- Width of water body (m)
- Width between bank tops (m)
- Freeboard (distance from water surface to bank top)

4.2 Aquatic Invertebrates

4.2.1 Aquatic Invertebrate field sampling

Aquatic invertebrates should be sampled along the same river and ditch transects as for ditch condition (see above and Annex 4.1.2 Ditch condition field sheet).

Timed sweep net samples should be taken from each ditch. Sampling should be done over a 3 minute period only along a 20m representative section, using the following method (taken from the Buglife guidance):

While standing at the water margin, the sample is taken by netting the ditch vegetation using short jabbing thrusts in dense emergents and raft-forming plants (Glyceria, Apium, etc), and occasional longer strokes into submerged plants (Elodea, Ceratophyllum, etc) in deeper water. The surveyor moves along the bank as the netting proceeds, selecting patches of vegetation that exhibit the greatest small-scale mosaic structure, since these patches seems to yield more specimens. Moving backwards appears to work better than moving forwards, because the pole can be kept angled in towards the feet and provide greater pulling power. Netting stops when the net begins to fill to the point that it becomes more difficult to push, and is usually about a quarter to a third full of plant material (about 2-3 litres by volume). This takes 1-3 minutes and can be fairly exhausting in dense vegetation. Where there is little vegetation, for example in newly cleaned ditches, a sample can be obtained from the few scraps that are left, where invertebrates tend to congregate. The bottom sediment is avoided since it clogs the net and contains almost no species that form part of the

analysis (although Libellula may be overlooked). If sediment is collected by accident, it is advisable to discard the sample and start again.

An initial sorting of invertebrates and vegetation/debris should be carried out on the bank, using a large white tray. A wash bottle containing water can be used to release smaller specimens from the net. Large specimens can be collected using a small tea strainer and placed directly into a sample tube while the smaller ones are left in the tray while the water is siphoned off until only the invertebrates are left. The invertebrates will then be transferred to a labelled sample bottle containing 70% ethanol to be preserved and taken back to the laboratory for identification. Large beetles will mangle soft animals in the sample unless they die quickly, so should be preserved separately.

4.2.2 Aquatic Invertebrate sampling lab analysis

Identify all invertebrates collected down to species level where possible back in the lab (See Annex 4.2.2 Ditch invertebrate – aquatic invertebrate lab field sheet)

Equipment used:

- Development tray
- 90% alcohol
- 30ml invert tubes
- Forceps different sizes
- Sieve (two different sizes, one for sieving the initial sample, one for removing vegetation when in development tray)
- Pot for waste
- Petri dishes
- Paintbrush
- Permanent marker



The samples collected from the field contain lots of vegetation debris which makes sorting through and identifying the invertebrates difficult. Therefore, another step can be added, separating the inverts from the vegetation.

- 1. The sample can be emptied into a sieve small enough that organisms of around >2mm wouldn't fall through it (This is because the purpose of this study is to analyse the inverts present as chick or lapwing food). The samples are then washed with tap water until the wastewater runs clear.
- 2. Then the sample is placed into an identification tray (a white photography development tray was used). Ensure there isn't too much sample in the tray, if necessary separate the sample into different lots.
- 3. Water is then poured over the sample into the tray to make sorting through the vegetation easier (1-2 L).
- 4. Any large pieces of vegetation are carefully removed, ensuring there are no invertebrates stuck to them. Be especially careful with clusters of vegetation, it could be a caddisfly larva.
- 5. Then the sample is sorted through, by moving the sample from one side of the tray to the other and back again, removing vegetation as you go.



- Caddisfly larvae casing made of snail shells

- 6. Each invertebrate found is then placed into a tube filled with 70% ethanol solution (one tube per sample), and labelled to match the sample being analysed.
- 7. Each tube should be filled no more than half way with invertebrates to ensure there was little dilution of the alcohol. Larger invertebrates were placed into their own tubes.
- 8. This was carried out for each sample.

Some samples are more difficult than others:

- Those which contained more sediment need to be washed for longer, or separated before washing.
- Those which contain pond weed are a lot harder to sample. For samples like this fill the tray with much more water (2-4 L), and then scrape the top layer of weeds off, and sort through them separately afterwards (As some of the invertebrates, especially crustaceans, float in water).



Caddisfly larvae casing made of vegetation

4.3 Terrestrial Invertebrates

To investigate effects of field condition and past management on soil invertebrate communities and the food resource they represent for birds the terrestrial invertebrate community should be sampled. Fields are paired and categorized as "Improved" or "Floristically Diverse" depending on the known history of management, and the floral diversity present in initial site assessments. All fields possess wet features in the form of scrapes, depressions, or ditches. Surveys are undertaken during May and June.

4.3.1 Pitfall Traps

Ground active invertebrates are surveyed using pitfall traps, constructed from plastic cups of 75 mm depth and 60 mm diameter. Scentless washing up liquid is diluted in a ratio of 0.2 Lto 2 L of tap water. Traps are filled with 40 mm of diluted washing up liquid and placed in the ground within two m of the soil sample location. For each field, four traps were positioned in a grid with two traps placed approximately 20 m apart on the edge of a wet feature present in the field, and the other two approximately 20 m away in the drier field area. A wire netting cage is secured to the ground over each pitfall trap using six inch metal tent pegs. The design is cost effective and reduces interference from birds and livestock, in addition to preventing vertebrates falling into the traps. Traps are left in position for 7 days. Upon collection, traps are sealed and stored in fridges and processed within 2 weeks (See Annex 4.3.1 Terrestrial invertebrate sampling sheet).

When trap contents are processed organisms are removed from the trapping liquid and stored in 70% ethanol until identification. Identification is carried out as per the soil samples (see 4.3.2). In addition, each individual invertebrates' body length is recorded to the nearest 0.1 mm.

4.3.2 Soil sampling

To sample soil invertebrates soil samples should be taken. Four soil blocks are taken per field within a 20 m square, two in wet areas and two in dry areas. The soil blocks are 185 mm x 185 mm x 100 mm and are taken using a bladed spade and a quick levering action to minimize escape of soil organisms. Samples will be sealed in plastic bags, stored in a fridge and processed within two weeks (See Annex 4.3.1 Terrestrial invertebrate sampling sheet).

Soil moisture content is measured by gravimetric method. Soil samples are sorted by hand, as a reliable way for surveying earthworms and diptera larva, samples are sorted twice. Organisms are stored in 70% ethanol and later identified.

4.3.3 Vegetation

Conduct a structured W walk between two pitfall locations in both the dry and wet areas of each field, take 10 survey points in the dry area and 10 survey points in the wet area. At each survey point, take a vegetation height measurement and assess a 1 m quadrat for % cover of bare ground and litter and % cover of positive and negative indicator species (See Annex 4.3.3 Terrestrial invertebrate – vegetation sampling field sheet).

5 Wildfowl and Wader Monitoring

Eurasian wigeon, teal, common snipe, black-tailed godwits all winter in the Avon Valley and it is expected that they could redistribute following habitat management and make greater use of hotspot sites. Monthly counts should be undertaken through December – February each winter (See Annex 5 Wildfowl and wader monitoring field sheet).

Enter and walk through each field (if possible fields are likely to be flooded) in a 'W' shape, as this allows most areas of the field to be covered. This procedure allows more cryptic species such as Snipe *Gallinago* gallinago to be detected, alongside other waterfowl species, and allows the river and riverbanks to be viewed easier. The number and location of each bird present is recorded on survey sheets using British Trust for Ornithology (BTO) bird codes. Survey sheet records are inputted into a database as soon as possible after surveying. . To reduce double counting of individuals where birds land and which direction they were flying is recorded. In the event of flooding, when fields are inaccessible, surveys are done from access points using a scope to get a full view of the fields. If the fields are completely inaccessible, the fields are noted as not surveyed.

Annex - Field Sheets

1. Monitoring breeding waders field sheet

LIFE Waders for Real

Waders for Real

Breeding Wader Survey 2015

Site number			Dat	e (visit)	
Site name			Sta	rt time	
Observer			Sto	p time	
18741					
Weather					
Clear		Calm		Dry	
Partially cloudy		Breeze		Rain/drizzle	
Overeast	_	Minch	_		

Time field started	Field	Wader species	Behaviour & notes	Corvids	Gulls	Raptors	Ducks, geese, swans, herons

Time field started	Field	Wader species	Behaviour & notes	Corvids	Gulls	Raptors	Ducks, geese, swans, herons

Species codes:

Lapwing	L.	Jay	J.		Black-headed Gull	BH
Redshank	RK	Magpie	MG		Lesser BB Gull	LB
Snipe	SN	Jackdaw	JD		Herring Gull	HG
Curlew	CU	Rook	RO		_	
Oystercatcher	OC	Carrion Crow	C.		Sparrowhawk	SH
Green Sandpiper	GE	Raven	RN		Buzzard	ΒZ
Ringed Plover	RP				Kestrel	K.
Little Ringed Plover	LP	Little Grebe		LG	Hobby	HΥ
_		Great Crested	d Grebe	GG	Peregrine	PE
Shelduck	SU	Mute Swan		MS	_	
Gadwall	GA	Greylag Goos	e	GJ		
Teal	T.	Canada Goos	e	CG		
Mallard	MA	Egyptian Goo	se	EG		

1.2 Nest finding and monitoring field sheet

LIFE Waders for Real Lapwing nest sheet

Nes	t Co	ode				Finder Initials					
Farm	Т					Nest Lo	ogger				
Field	\top					Numbe					
GPS	\top					Date &	time se	t			
Photo						Date &	time re	trie	ved		
Egg	mea	surements:									
	Dat	te Measured									
	Ler	igth (mm)		Widt	th (mm) Wei				eight (g)		
1											
2											
3											
4											
Veg	etati	on characterist	ics:	•			•				
		Nest at finding			Nest at fi	nish			Randon	n at finish	
Veg Heig	hts										
% veg typ Bare, grass, dicot, sedge/rush, weeds etc.											
No. of sp	g.										
Visit	t att	endance: Date	Time		No. Eggs/	/chicks	A duit a	nn /1	/es/no)	In electric fence?	
Nest four	nd	Date	Time		NO. Lggs/	CHICKS	Addit	טוו ()	/es/110)	in electric rence:	
2											
3											
4											
5											
6											
7											
atched/de kely preda		ed/predated?									
ntered hecked? Ir	nitia		est Hatch	ı dates		Nest chec	cks 🗆		Nest v	regetation	

1.5 Chick finding and monitoring field sheet

LIFE Waders fo Brood ID 🕵 WA-01		l Lapwi	ing Chic	ks Si	ghting	gs	and Ca	ptı	ure	Ringing/ Survey/s Radio-tra	ighting			
Date			Time						GP5	3				
Site			Field/plot	t		0	bserver/R	inge	r					
Total brood size						Ą	ge				Estimated/Known??			
Total adults						N	est ID					Т	IN NEST	,
TOTAL BROODS IN	FIELD					Ca	aught/see	n			In elec	ctric	fence?	
Ring	New/R	Bill	Tarsus		Weight		Wing	Fa	ecal	Colour ringe	d	- 1	Radio Tag	Buccal Swab?
				1				\vdash				\dagger		
								_						<u> </u>
Vegetation	Don	ninant ve	g	% b	are grou	ıno	d	Veg	getati	ion height	Pe	enet	trability	
Brood location				1										
				2										
Random location				3										
GPS														
				2										
				3										
Notes	•						<u>'</u>				1			
Entered? (Chick bio	metrics	- Bro	ood ch	heck [1	Brood fo	ragi	ing lo	cations	R	≀adi	o trackin	g 🗀
Checked? Initials					٦	1		_	_ [\neg \Box				- U

1.5.1 Chick radio-tracking triangulation field sheet

In electric fence??

	Locatior (site, fiel habitat)				
Point 4	GPS Bearing				
Pc					
Point 3	Bearing				
۵	SdD				
Point 2	Bearing				
۵	SdD				
Point 1	Bearing				
ď	SdS				
	Female seen? Alarm calling?				
	Tagged chick seen?				
	Radio signal?				
	No. of adults present Radio me Chick Freq. (Beh.)				
	Freq.				
	Chick				
	ae a				

Chick Tracking – Triangulation

1.7 Lapwing adult colour ring sighting form

LIFE Waders for Real Adult Lapwing Colour Ring Sighting

Date	Time	Observer	Left Leg
Location	Site and Field	GPS	
Flock?	Behaviour	% of flock viewed	
% Confidence	Age	Sex	
			N

1.8 Lapwing adult capture field sheet



Waders for Real Lapwing Capture Sheet Waders



Right Leg

Capture Details		Data Entered:	Data Checked:
Intials			
Site			Colour Marks
Field			
G.Ref			
Date			
Cap.Time			
Release Time	:	Nest Id	
Cap.Type (N or L)	Nest / Lamp	→	

Bird Details

Bird Deta	alis				
Ring No		Crest Length (LF)		P10 End Shape	Blunt / Rounded
Species		Head & Breast Pattern		P10 End Shape	Broad / Narrow
Wing		P10 (folded) =		P10 Base Colour	Black / Inter / Brown
Bill to F.		Wing Tip (spread) =	P	P10 Spot Angle	90° / 45°
Tarsus		Upper Part Descri	ption	P10 Spot Colour	W/WB
Weight				P10 Spot Width	Both edges / Top only
Age				P10 Inner vein	No Line / Dark band
Sex				If Dark band	Wide / Narrow
				P10 patch extends	Yes / No
Commen	ts (e.g. p	lumage colouration)			

2.1.1 Avian predator monitoring field sheet

LIFE Waders for Real Avian predator monitoring

Site	Date
Field	Start time
Observer	Stop time

Time	Corvids	Gulls		Herons	Herons		
	Field	Flying	Field	Flying	Field	Flying	
0 mins							
+5							
+10							
+15				1			
+20				1			
+25							
+30							
+35							
+40							
+45							
+50							
+55							
+60							

Raptors

Species	Time	Notes

Lapwing chases

Time	Species	Time off nest	Time back on	Cause
	chased		nest	

2.1.2 Avian predator monitoring revised field sheet

LIFE Waders for Real - Avian predator monitoring

Site	Date
Field	Start time
Observer	Stop time

ime	Corvids	Gulls/Raptors	Herons	Notes	
) mins					
-5					
-10					
+15					
-20					
-25					
+30					
+35					
- 40					
+45					
÷50					
-55					
+60					
-65					
- 70					
-75					
+80					
-85					

Avian predator observation sheet

Site	Cloud	Clear/partly cloudy/ overcast	Total adults	
Date	Wind	Calm/ breeze/ windy	Total females on nests	
Observer	Temperature		Total females with chicks	

Predators							Lapwings Focal nes	t/brood (+ details):	
Time predator enters field	Predator species	Predator height (m)	Time chased (s)	Number attempts to take a chick	Attempt successful?	Time it leaves field	Maximum number chasing	Horizontal distance to predator (m)	Time focal bird away from nest/chicks (s)

2.3.1 Fox scat field sheet

Fox Scat Survey Record Sheet

Date: DD/MM/YYYY	Site:			Observer(:	s):			
Map Number:								
Start Time(24hr): HH:	MM	End Tir	me(24hr):	HH:MM				
GPS Start:		GPS End	i:					
Scat Code: DATE_GRID-REF_INITIAL_NO. SCATS	Envelope or Pot (E/P)	Time Collected (24hr)	Photo? (√/X)	Map point:	Location Notes:	Freshness? (1-5)		
e.g. DD-MM-YY_SU-XXXXX-YYYYYY_TP_01	E E	16:55	√ √	1	Corner of bridge	(1-3)		
	I		1	I	I	1		

2.3.2 High seat count field sheet

Somerley high seat watches 2019 Name: Date: Start: End: Sunset: Wind: Cloud cover: Temp:



tim e	predato r	type of activity e.g. hunting, walking	if fox, tagged ?	white collar mark?	collar numbe r, if visible	gender, if obvious	distinguishing features e.g. white tip on tail	age	safe to shoot ?	if not safe, why?
		11 1 1 1 1 1 1 1 1 1 1 1 1 1 1 1 1 1 1	tim predato activity e.g.	activity e.g. if fox, tim predato hunting, tagged	tim predato activity e.g. if fox, white tagged collar	activity e.g. if fox, white numbe tim predato hunting, tagged collar r, if	tim predato activity e.g. if fox, white numbe gender, tagged collar r, if if	tim predato activity e.g. if fox, white numbe gender, distinguishing tagged collar r, if if features e.g.	activity e.g. if fox, white numbe gender, distinguishing tim predato hunting, tagged collar r, if if features e.g.	tim predato hunting, tagged collar r, if if features e.g. shoot

Vegetation record 2019 (fox range fields)

Site name:
Date:
Recorder:
Veg types:
G = grass G (m) = mown grass R = rush S = sedge D = dicotyledon W = dock/thistle/nettle E = bare earth or mud

	Field		Field			Field		Field			Field		
	Veg ht	Cover	Veg type										
1			1,750			1,750			1,750			1,750	
2													
3													
4													
5													
6													
7													
8													
9													
10													
11													
12													
13													
14													
15													
16													
17													
18													
19													
20													

2.4 Camera trap forms



Camera Traps 2019 Field Sheet

General

<u></u>
- 10e

		GPS:	
Date:		Field No:	
Sellel ID.	Location	Site Name:	

	Photo Interval Time:	
	Number of Triggers Set:	
	SD Card No:	
Hardware	Camera Serial No:	

	Orientation (bearing):		Batteries Replaced? :
	Camera Angle (Straight/Down):		Photo of Board? :
Setup	Boundary Type:	Check	Internal Date and Time Set:

Vegetation type Uniform or Tussock
Average vegetation height: 0-5, 5-10, 10-20, 20-50, 50-100 cm
Livestock: Cows Horses Sheep Number of:

ife				Veg & livestoc					
Game & Wildlife				Comments					
Gam	2018			Internal D&T					
	Camera Trap Retrieval Form 2018	ä		Functional					
ea	Retr	Site Name:		In Situ					
<u> </u>	а Тгар	Sife	Set:	SD no.					
Waders for Real	Camera			Cam Serial					
****				Collection Time					
		Date:	Setter:	Location					

2.5 Electric fence checking sheet

	*	*	*	
*		,	1	*
*	1	休	e	*
*	-	U		*
	*		*	





Electric fence checking sheet

	DICCUITC IC.			,	
Checker Initial	Date	Time Sta	rt	Site	Field
		Time End	t		
Comments			Livestock: Number o	Horses Sheep Cows f:	

Voltage (mV)	At finding	After maintenance
Cable 2		
Cable 4		
Cable 6		
Cable 8		

Vegetat	ion measure	ements (cn	1)			

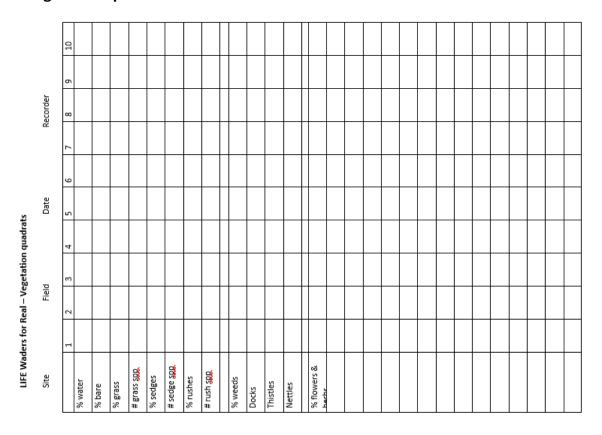
3.1 Sward and land use field sheet

LIFE Waders for Real 2015	Site no.	Site name	Observer
Sward and land use sheet	Date	Visit no.	

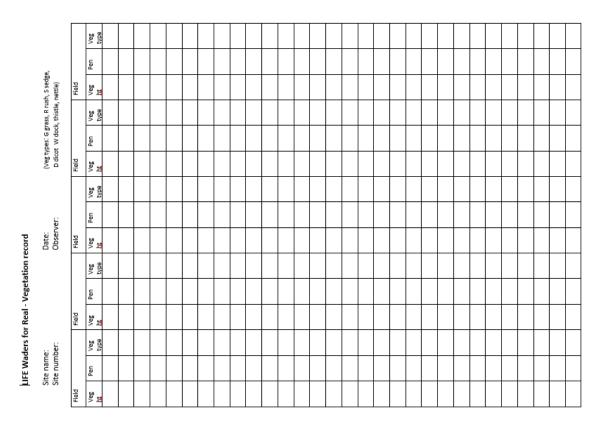
Field	Vegetati	on type			Vege	tation h	eight (d	m)					Land use	Livestock	(Surface water %	Dampness
	Juncus	Carex	Thistle, docks	Flower plants	0-5	6-10	11- 20	21- 30	31- 40	41- 50	51- 60	>60		Cattle	Sheep	Horses		

Vegetation type: % per field Vegetation height: 20 measures per field Land use: PP permanent pasture, RPP rough permanent pasture, H hay, S silage, A arable Livestock: numbers of each, count cows and calves separately Dampness: % of field F flooded, DA damp, DY dry

3.2 Vegetation quadrat field sheet



3.3 Vegetation record penetrability field sheet



4.1.1 Dragonfly and damselfly, ditch environment and vegetation field sheet

Ctant Time		(Ditch/River)			+	
Start Hille	Sta	Cloud Cover				
Finish lime	Ter	Temperature Water Level				+
	Wa	Water Speed				
	Ditt	Ditch Width Grazed				+
	Bar	Bank Vegetation				
	Hei	Height				
						 -
		Fmergent	†	1		1
	Neg.	Vegetation				
		Height				
	Agi	Aguatics				
	Nes	Vegetation	_			
	Tei	Height				
-						
		_				-

4.1.2 Ditch condition field sheet

Ditch recording sheet

Conductivity

Chemistry –
Temperature

a) Environmental variables

Site: Ditch Ref:							
Date:			Recorder:				
20m Ditch section GPS start: GPS finish							
Ditch features/sample	1	2	3	4	5		
Water width							
Bank top width							
Freeboard							
Water depth							
Silt depth							
Bank slope a/b							
Slope under water a/b							
Turbidity							
Water colour							
Flow m/s							
Water sample taken							
Invertebrate sample taken							
Chemistry – pH							
Chemistry –							

4.2.2 Ditch aquatic invertebrate lab field sheet







Ditch Sample Lab Sheet

Site	Ditch number	Sample number
Date collected	Processor	Date processed

Species/Genus/Family	Number present	Taxonomic level

4.3.1 Terrestrial invertebrate sampling sheet

LIFE Waders for Real – Invert sampling location record						
Site name:	Date:					
ield number:	Location (wet/dry	GPS	Soil sample?			
	etc)					

4.3.3 Terrestrial invertebrate vegetation sampling field sheet

LIFE Waders for Real – Invert sampling Vegetation record

Site name: Date: Site number: Observer:

Field	d Field			Field			Field				
Veg ht	Bare %	Litter %									
	70		100		70		,,,	70		,,,	,,,

		-
POSITIVE INDICATORS	Indicator status	Score
Meadowsweet (Filipendula ulmaria)	SSSI	
Water mint (Mentha aquatica)	SSSI	
Small blue-green (C.flacca, C.nigra, C.panicea)	SSSI	
Cuckooflower (Cardamine pratensis)	SSSI	
Lesser water parsnip (Berula erecta)	SSSI	
Water avens (Geum rivale)	SSSI	
Marsh/Fen bedstraws (Galium palustre, G.	SSSI	
uliginosum))		
Greater Bird's-foot-trefoil (Lotus	SSSI	
pendunculatus)		
Ragged-robin (Lychnis flos-cuculi)	SSSI	
Marsh-marigold (Caltha palustris)	SSSI	
Meadow thistle (Cirium dissectum)	SSSI	
Hemp-agrimony (Eupatorium cannabinum)	SSSI	
Marsh valerian (Valeriana dioica)	SSSI	
Devil's-bit scabious (succisca pratensis)	SSSI	
Common meadow-rue (Thalictrum flavum)	SSSI	
Amphibious bistort (Persicaria amphibia)	SSSI	
Lesser spearwort (Ranunculus flammala)	SSSI	
Knapweed (Centaurea nigra)	SSSI	
Hairy hawkbit Leontodon hispidus)	Medium-high quality	
	MG/M grassland	
Creeping Jenny (Lysimachia nummularia)	Medium-high quality	
01 1 1 1 10 11 11 11 11 11	MG/M grassland	
Other slender sedges (C.disticha, C.hirta, other	Medium-high quality	
Carex spp)) Booklime (V.beccabunga)	MG/M grassland Medium-high quality	
bookiime (v.beccabunga)	MG/M grassland	
Jointed rushes (Juncus acutiflorus, J.	Medium-high quality	
articulatus)	MG/M grassland	
Spiked rush (Eleocharis palustris)	Medium-high quality	
pince restriction perestris,	MG/M grassland	
Forget me not (Myosotis spp)	Medium-high quality	
,,,,,,,,,,,,,,,,,,,,,,,,,,,,,,,,,,,,,,,	MG/M grassland	
Divided-leaved Umbellifer spp (eg Silaum	Medium-high quality	
silaus, Pimpinella saxifraga)	MG/M grassland	
Meadow vetchling (Lathyrus pratensis)	Medium-high quality	
	MG/M grassland	
Yellow-flag iris (Iris pseudacorus)	Medium-high quality	
	MG/M grassland	
NEGATIVE INDICATORS		
Broad-leaved dock (Rumex obtuifolius)		
Curled dock (Rumex crispus)		
Common nettle (Urtica dioica)		
Ragwort (Senecio jacobaea)		
Horsetail (Equisetus spp)		
Creeping thisle (Cirsium arvense)		
Spear thistle (Cirsium vulgare)		
Tussocky Juncus spp. J inflexus, J. effusus, J.		
conglomeratus		
Large grasses (>10mm wide)		
Large sedges (>5mm wide)		

5. Wildfowl and wader monitoring field sheet

LIFE Waders for Real Wintering Wildfowl Survey 2017/2018



Site number		Date	e (visit)		
Site name			Start time		
Observer			Stop time		
Weather					
Clear	Calm		Dry		
Partially cloudy	Breeze		Rain/drizzle		
Overcast	Windy				

Time field started	Field	Wader species	Duck species	Geese species	Gulls	Raptors	Other