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Methods in Ecology and Evolution

**THE WELFARE AND ETHICS OF RESEARCH INVOLVING WILD ANIMALS:
A PRIMER**

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17 **ABSTRACT**

18 1. Wild animals are used in scientific research in a wide variety of contexts both *in situ*
19 and *ex situ*. Guidelines for best practice, where they exist, are not always clearly linked
20 to animal welfare and may instead have their origins in practicality. This is complicated
21 by a lack of clarity about indicators of welfare for wild animals, and to what extent a
22 researcher should intervene in cases of compromised welfare.

23 2. This *Primer* highlights and discusses the broad topic of wild animal welfare and the
24 ethics of using wild animals in scientific research, both in the wild and in controlled
25 conditions. Throughout, we discuss issues associated with the capture, handling,
26 housing and experimental approaches for species occupying varied habitats, in both
27 vertebrates and invertebrates (principally insects, crustaceans and molluscs).

28 3. We highlight where data on the impacts of wild animal research are lacking and
29 provide suggestive guidance to help direct, prepare and mitigate potential welfare
30 issues, including the consideration of end-points and the ethical framework around
31 euthanasia.

32 4. We conclude with a series of recommendations for researchers to implement from the
33 design stage of any study that uses animals, right through to publication, and discuss
34 the role of journals in promoting better reporting of wild animal studies, ultimately to
35 the benefit of wild animal welfare.

36

37 Key words: capture-mark-recapture, animal ecology, ethics, 3Rs, 9Rs, animal welfare,
38 legislation

39

40 **1. INTRODUCTION**

41 Research involving wild animals covers a wide range of species using different techniques
42 and impacts individual animals, groups, up to the level of whole ecosystems (Sikes & Paul
43 2013). Fieldwork may often be conducted in less than ideal conditions—in poor weather,
44 non-sterile environments, areas exposed to climate extremes—and has the potential to
45 harm the study animals during capture and handling (Chinnadurai *et al.* 2016). Despite the
46 complexities of these situations, ensuring animal welfare should be a critical part of wild
47 animal study design.

48 In this paper, we use the World Organisation for Animal Health (OIE 2017)
49 definition of animal welfare, which states that welfare is, ‘*how an animal is coping with*
50 *the conditions in which it lives...Animal welfare refers to the state of the animal; the*
51 *treatment that an animal receives is covered by other terms such as animal care, animal*
52 *husbandry, and humane treatment.*’ Current ethical considerations surrounding the use of
53 wild animals in research are grounded principally in the 3Rs (reduce, refine, replace:
54 Russell, Burch & Hume 1959). The 3Rs were originally designed for laboratory animal
55 research, in which the animals are used as human models, and where the impact of
56 manipulations or procedures is limited to animals participating in the study (Russell *et al.*
57 1959; Lindsjö, Fahlman & Törnqvist 2016). There are specific issues in the wider
58 application of the 3Rs to wild animal research (Box 1), which led to new proposed
59 variations (9Rs: Curzer *et al.* 2013). Even so, a broad synthesis on working with wild
60 animals in research is lacking. In this paper, we outline the critical welfare-related
61 considerations associated with carrying out wild animal research. These include the
62 welfare implications of capturing, handling and housing; the welfare implications of
63 ecological manipulations and experimental approaches; the consideration of end-points for
64 the study: release, rehoming and euthanasia; and finally, the ethical considerations for

65 publishing research conducted on wild animals. It is not our goal to provide explicit
66 instructions but rather to provide a launch-point for discussions when planning
67 experiments, and encourage the researcher to consider both focal and non-focal animal
68 welfare when designing and implementing experiments. We provide a framework to aid
69 that goal.

70

71 **2. WELFARE CONSIDERATIONS IN CAPTURING, HANDLING AND** 72 **HOUSING OF WILD ANIMALS**

73 Any form of intervention on a wild animal will have some impact on that individual,
74 directly or indirectly. A standard ethical approach to the justification of research is to
75 balance research gains against the costs or harm to all involved, and attempt to minimise
76 the negative effects wherever possible (Graham & Prescott 2015; Brønstad *et al.* 2016). In
77 this section, we discuss some of the most common types of intervention in wild animal
78 studies.

79

80 **2.1 Capturing wild animals**

81 Capturing events are stressful for wild animals (Wilson & McMahon 2006). The impact on
82 the individual ranges from minor to severe; short to long-term; and may be physical,
83 physiological and/or psychological (see Table 1 in: Kukulová, Gazárková & Adamík
84 2013). The primary consideration of any field researcher must be to minimise these
85 impacts, both to the individual and population.

86 There are many ways to capture wild animals (see Schemnitz *et al.* 2009), but they
87 generally follow the same rules and techniques (Box 2). Selection of a context- and
88 species-appropriate method is of critical importance and should minimise the number of

89 injuries, mortalities and by-catch. Across studies (Table 1), it is clear that there is
90 considerable taxon-specificity in accepted welfare levels. For example, within vertebrate
91 research, avian studies report much lower injury and mortality rates than all other taxa
92 (Table 1). A key part of reducing any form of injury is continual review and refinement of
93 techniques. Sources of injury or mortality can be predicted by the technique chosen
94 (Vedhuizen *et al.* 2018), timing—e.g. cold or hot weather (Clewley *et al.* 2018; Read *et al.*
95 2018), or because the target animal has certain risk factors such as size, age, or species
96 (Schonfield *et al.* 2013; Clewley *et al.* 2018; Veldhuizen *et al.* 2018). These risks should
97 be appropriately identified before commencing (see suggested refinement below).

98 How can we improve capture techniques? There needs to be a universal maximum
99 level of acceptable injury and mortality. Rather than restricting methods of capture, such
100 thresholds would serve to identify problematic techniques that need urgent refinement.
101 Such rates should continue to be debated, but thresholds of <2% mortality are suggested
102 (Arnemo *et al.* 2007). Injury rates are harder to characterise since injuries could range
103 from minor (e.g. superficial abrasion) to serious (e.g. broken bone) (Iossa, Soulsbury &
104 Harris 2007). Studies have used injury scoring (e.g. mammals: Powell & Proulx 2003;
105 Iossa *et al.* 2007), but these typically focus on probability of survival and not pain or long--
106 term effects on fitness (Iossa *et al.* 2007). There is no accepted threshold for injury levels;
107 we suggest that: (a) researchers actively report whole body injury scores (e.g. Table 4 in
108 Iossa *et al.* 2007), and (b) the following maximum injury thresholds as acceptable for
109 capture techniques: <2% serious injuries, <5% moderate injuries, <10% mild injuries only.

110 A second way we can improve capture techniques is through more thorough risk
111 assessment processes identifying the potential consequences for both target species as well
112 as affected non-target species. This provides an opportunity to consider the entire
113 process—including handling and processing—and identify suitable areas for refinement.

114 Thirdly, there should be standard reporting in journal methods of injury and mortality
115 rates; such data would then available for future review, analyses and further refinement.

116 Regardless of method used, there is always the likelihood that non-target species
117 are caught. Selectivity of method is an important consideration in method choice, and
118 many non-target species may be at greater risk of injury and mortality than target species
119 (Iossa *et al.* 2007). Again, clear reporting of selectivity rates (% of total captures) and
120 injury rate of non-target species should be part of methods sections.

121 Finally, physical injury and pain are only one facet of the distress associated with
122 capture methods. Anxiety, stress and escape behaviour will also negatively impact animal
123 welfare (Marks *et al.* 2004). When prolonged, distress having deleterious effects on animal
124 health and subsequent survival (Moberg 1999). Trap type (Cattet *et al.* 2003) and
125 coverings (Bosson, Islam & Boonstra 2012) can impact capture stress levels.

126 In contrast to vertebrates, invertebrates have received little attention in terms of
127 efficacy and mortality rates of capture techniques, with no comparative studies available.
128 Evidence from commercial fishing of crustaceans suggests injury and mortality rates can
129 be high during capture (Table 1). For insects, mortality is often an expected outcome of
130 sampling, unless the aim is the mark and recapture of individuals, live experimentation, or
131 husbandry in the laboratory. Mortality is not always necessary for sampling and many
132 techniques exist that minimise mortality and allow safe release of captured insects –
133 methods are often designed for convenience of sampling, rather than a specific purpose.
134 Drinkwater, Robinson and Hart (2019) provide important insights into the shifting public
135 opinion and laws to protect invertebrate welfare during scientific studies. Their
136 recommendations very much align with the principles of the 3Rs: to use appropriate power
137 analyses; reduce by-catch by refining trapping methods and retain by-catch for further
138 studies; and minimise suffering (Drinkwater *et al.*, 2019).

139

140 **2.2 Handling wild animals**

141 Handling wild animals should be avoided whenever but, if necessary, should be minimal.

142 Total processing time from capture to release should be minimised: faster total processing
143 time can reduce stress, injury and mortality (Langkilde & Shine 2006; Ponjoan *et al.* 2008;
144 Deguchi, Suryan & Ozaki 2014). During the interval between capture and release, many
145 species benefit from being kept in the dark, either completely or at least by covering the
146 eyes (e.g. Mantor, Krause & Hart 2014).

147

148 **2.3 Physical sampling**

149 The welfare implications of specific procedures used during handling have received little
150 attention, despite the importance of handling methods being recognised in laboratory
151 settings (Cloutier *et al.* 2015, Gouveia & Hurst 2017). A handful of studies have compared
152 broad outcomes, such as survival between groups undergoing different procedures
153 (Douglass *et al.* 2000; Wimsatt *et al.* 2005). However few studies have compared the stress
154 of specific procedures during handling: for example the stress of microchipping versus toe-
155 clipping in lizards (Langkilde & Shine 2006); or the additive stress of blood sampling that
156 after capture in snakes (Bonnet, Billy & Lakušić 2020). For most species and handling
157 procedures, the extent that procedures themselves cause additive stress and the duration
158 over which they compromise welfare is unclear. This component of wild animal studies
159 needs to be addressed.

160 The impact repeated exposure to procedures have on an animal, cumulatively, over
161 their lifetime is less clear. Existing evidence indicates repeated captures have either no
162 effect (Rode *et al.* 2014), or deleterious effects (Cattet *et al.* 2008; Sharpe *et al.* 2009). This
163 depends on the species, methods, and parameters measured. Research into cumulative

164 impacts of repeated procedures has also received little attention and again, needs urgent
165 research attention.

166

167 **2.3.1 Anaesthesia and surgery**

168 Anaesthesia can be used during the capture and/or handling process. Field wildlife
169 anaesthesia can improve safety for both researchers and animals, and is often necessary for
170 both invasive (e.g. surgical, blood collection) and non-invasive (e.g. morphometric,
171 collaring) research. The use of anaesthesia in wild animals is challenging as there are little
172 information available on procedures, difficult environmental conditions, and mixed welfare
173 outcomes (reviewed by Chinnadurai *et al.* 2016). Anaesthesia comes with its own
174 increased risk of mortality, even with well-established protocols (0.2-2.2% mortality:
175 Arnemo *et al.* 2006; 9% mortality (Chirife & Millan 2014). It requires a high level of
176 training and skill and may engage specific national legislation or regulation. It is
177 particularly challenging in smaller animals as there are smaller margins of error with
178 dosage. In particular, continuous monitoring of stress levels and degree of unconsciousness
179 is essential, in order to avoid over or under-dosing record-keeping of anaesthetic events
180 (Chinnadurai *et al.* 2016). Whilst most widely used in vertebrates, anaesthesia can also be
181 used for invertebrates (see Lewbart *et al.* 2012), some of which are suitable for field use
182 (e.g. Venarsky & Wilhelm 2006; Loru *et al.* 2010). However, in most scenarios anaesthesia
183 is unnecessary and in general has been poorly studied in invertebrates.

184 Anaesthesia can reduce stress during handling (e.g. Mentaberre *et al.* 2010), but can
185 also lead to behavioural changes post-anaesthesia (e.g. fish: Caudill *et al.* 2014; nest
186 abandonment in birds: Machin & Caulkett 2000). Handling without anaesthesia can
187 potentially return animals to their social groups more quickly and allow release without
188 danger of predation. When anaesthesia is used and recovery is slower, trapped animals

189 may need food, water, help to maintain thermoregulation, and other resources, as well as
190 protection from predation, conspecifics or weather until they can be returned to the wild.
191 Given the level of complexity involved in the use of anaesthesia and post-anaesthetic care,
192 it is essential that researchers and veterinarians evaluate all aspects of the protocol, prior to
193 commencing work, in an effort to minimize animal risk. All available options should be
194 considered before researchers choose to use anaesthesia.

195 Regardless of species, any form of surgery is significant and alternatives should be
196 considered. This is especially true when carrying out surgery in the field, given the
197 additional challenges of administering anaesthesia, maintaining aseptic techniques, and
198 potentially introducing antibiotics to wild animals and the environment (Mulcahy 2013;
199 Fiorello *et al.* 2016). Guidance on the considerations for field surgery are detailed in
200 Chinnadurai *et al.* (2016) and Fiorello *et al.* (2016), including the provision of analgesia.

201

202 **2.3.2 Blood and haemolymph sampling**

203 Blood sampling is invasive and should be justified in any study protocol. Many of the key
204 considerations in blood sampling are species- and study-specific. For vertebrates, these
205 include site of blood sampling (e.g. caudal, brachial, facial or pinnal veins), blood volume,
206 and the temporal pattern of sampling. In particular, no more than 10% of blood volume
207 should be taken at once, equating to approximately 1% body mass, or if sampled multiple
208 times, no more than 1% blood volume every 24 hours (Diehl *et al.* 2001). Little
209 consideration has been given to sampling from invertebrates. The small size of many
210 invertebrates makes it difficult to take haemolymph samples, and often small volumes
211 must be collected. With the exception of cephalopods, sampling of haemolymph from
212 invertebrates operates with little guidance. Cephalopods lack superficial blood vessels
213 making blood sampling difficult (Fiorito *et al.* 2015); additionally, their haemolymph is

214 pale blue (oxygenated) or colourless (deoxygenated), meaning haemorrhage can be
215 difficult to detect (Fiorito *et al.* 2015). For other invertebrates, it is recommended that a
216 minimum volume for analysis is taken if the animal is to be released or live afterwards.
217 Techniques for microsampling small invertebrates exist (e.g. Piyankarage, Featherstone &
218 Shippy 2012). The presence of an open haemocoel simplifies sampling, however, the
219 hydrostatic skeleton of many insects means that the haemolymph can be under pressure
220 and too large a puncture can result in excessive bleeding (SCC personal observation). To
221 ensure the insect survives the procedure, it is critical the cuticle is punctured at a shallow
222 angle to avoid piercing the gut. Moderate volumes of haemolymph (2-50ul) can be
223 sampled without adverse effects on survival by using a narrow gauge needle for larger
224 insects (e.g. >0.15g), or a pulled glass capillary tube for smaller insects. If large or whole
225 body volumes must be taken, researchers must consider welfare and plan for potential
226 euthanasia.

227

228 **2.3.3 Marking and tagging**

229 Animals can be marked using external marks—colouring, tattooing, branding or appendage
230 clipping (reviewed by Silvy, Lopez & Peterson 2005); external tags or devices—
231 radiotransmitters, leg rings, ear tags, collars, harnesses; or internal tags or markers—PIT
232 tags, chemical markers. The relative merit of each technique varies based on the species
233 and the study purpose (Figure 1 & Box 3).

234 Marking, even with small physical marks (such as leg rings or nail varnish), can
235 have negative effects on an individual's health and behaviour (Table 2). Marks made by
236 ear, toe, exoskeleton or fin clipping, skin punches, or permanent marks such as tattooing
237 and branding are considered controversial (Murray & Fuller 2000; Hagler & Jackson
238 2001). Ethically, the question remains whether these types of marking methods should be

239 permitted and contradictory findings regarding their impacts only muddy the water. For
240 example, when compared with other techniques, toe clipping has been reported as both
241 more (Narayan *et al.* 2011) and less stressful than PIT tagging (Langkilde & Shine 2006;
242 Guimaraes *et al.* 2014). Exoskeleton—or sometimes leg or wing—clipping in
243 invertebrates is only applicable to a handful of species (Hagler & Jackson 2001), but may
244 also impact reproduction (e.g. Hall *et al.* 2015). In many cases, alternative methods of
245 marking are available (visible and UV-visible tattooing: Petit *et al.* 2012; McGregor &
246 Jones 2016), and studies need to make compelling justification for using more invasive
247 methods of marking, including a specific cost-benefit analysis.

248 Some forms of identification are relatively lightweight (e.g. British Trust for
249 Ornithology, AA bird ring = 0.04g), but devices such as geolocators, radiotransmitters and
250 GPS transmitters are considerably heavier. Evidence suggests that behaviour and fitness
251 can be impacted by device weight (Bodey *et al.* 2017) and researchers follow a rule of
252 thumb that devices should weigh no more than 3-5% of an animal's body mass. These
253 thresholds are somewhat arbitrary (Gessaman & Nagy 1988) and based on limited data.
254 For example, the 3% rule appears to be extrapolated from studies of albatross and petrel
255 device load and behaviour (Phillips, Xavier & Croxall 2003). Although there are studies
256 demonstrating negative effects of devices at or greater than 5% of body mass, this has also
257 been shown to be the case with devices less than 3% of body mass (Table 2; Bodey *et al.*
258 2017). Exceeding the 5% and 3% thresholds in vertebrate studies is more commonplace for
259 specific groups, for example bats (O'Mara, Wikelski & Dechmann 2014) and chelonia
260 (Fordham *et al.* 2006).

261 Threshold rules are often not considered invertebrates, with insect biologists
262 weighing anything from 2 to 100% of the insect's body mass (Kissling, Pattermore &
263 Hagen 2014). Few studies have examined the impacts on insect welfare, particularly

264 regarding the energetic costs of carrying such loads and impacts on social behaviour and
265 survival (12% studies quantified impact: Batsleer *et al.* 2020). Tagged individuals are often
266 the largest in the population and have better inherent survival (Le Gouar *et al.* 2015), but
267 further research is needed to fill the knowledge gap and inform best practice (Batsleer *et al.*
268 2020). Additionally, for all species, it is important to consider the standard fluctuations in
269 body mass that individuals may experience even within relatively short timescales (e.g.
270 Blackburn *et al.* 2016). Despite technological advancement leading to ever-smaller
271 devices, this has not decreased the percentage device weight being carried but instead,
272 devices are being deployed on smaller species (Portugal & White 2018). Researchers must
273 minimise the weight of the transmitter, rather than to maximise the load carried.

274 In addition to the weight of any biologging device, researchers must consider the
275 mode of attachment to the animal's body. Broadly, there are two main methods: internal
276 implantation or external attachment. The effects of such attachments have been previously
277 reviewed in birds (see Barron *et al.* 2010; Costantini & Moller, 2013) and marine
278 mammals (Walker *et al.* 2012). Wide ranging effects of device attachment have been
279 reported, from seemingly no response, to negative impacts on behaviour, health,
280 reproduction and survival (key examples given in Table 2). Long term behavioural and
281 physiological measures outside of the focus of a given study are often not recorded and as
282 such, the true impact of devices is likely unknown. The choice and placement of
283 biologging devices needs careful consideration for the ecology, lifestyle, morphology and
284 physiology of the study species (Casper 2009). The impacts should be considered
285 beforehand (Todd Jones *et al.* 2013) and reported as standard in subsequent publications,
286 including, metrics of impacts (Wilson *et al.* 2019).

287 Before deciding on a device and attachment, consideration of data recovery is
288 required. Some devices capture, store, and send data remotely, whereas others use timed or

289 biodegradable drop-offs, thereby removing the need for a second capture event and
290 additional stress. Remote drop-off and download technology are not always feasible as
291 they can add significant weight to devices (Thomas, Holland & Minot 2012). Additionally,
292 using biodegradable material or weak links may limit long-term device attachment and
293 function—for example, the collection of physiological data may not allow remote
294 downloads or drop-offs. Though not always possible, attempts should be made to detach or
295 remove devices. Where devices are left on long-term post-study, this should be accounted
296 for in the cost-benefit analysis.

297

298 **2.3.4 Capturing and killing**

299 Field researchers may be faced with the choice whether animals need to be killed as part of
300 the study design. For some studies, the collection of samples by killing is almost routine
301 (e.g. collecting voucher specimens for museums: Russo *et al.* 2017; sampling for many
302 invertebrates: Hohbein & Conway 2018). At the opposite extreme, there is considerable
303 debate centred on whether it is ethical to ever kill an animal (Hayward *et al.* 2019). A
304 number of journals have published guidance on this issue—there will be scenarios where
305 killing of wild animals is justifiable, but that that justification needs to be provided and
306 prior exploration of alternatives evidenced (Vucetich & Nelson 2007; Costello *et al.* 2016;
307 ASAB 2020; Table 3), and reported in the ensuing publication. Journals editors and
308 reviewers ultimately play a key role in shaping this by rejecting studies that do not
309 adequately justify their choice, or where suitable available alternatives have not been used.
310 Where researchers hide their methods deliberately this should be viewed as research
311 misconduct.

312

313 **2.3.5 Holding and keeping wild animals in captivity**

314 Animals taken from the wild should only be held in captivity where completely necessary
315 and, if the aim is not form a captive population, for a duration that allows their safe release.
316 The process of bringing animals into captivity, e.g. transportation (Box 4), exposes
317 individuals to multiple stressors that can lead to significant initial stress and extended
318 changes to the stress-coping mechanisms that can allow adjustment to captivity (Adams *et*
319 *al.* 2011; Angelier *et al.* 2016). Researchers should not underestimate the difficulty of
320 designing sets of captive conditions for different species (Schmidt 2010; Box 5). There are
321 arguments for keeping the housing, diet and social conditions ecologically relevant
322 (Beaulieu 2016), however, using standard conditions allows greater reproducibility
323 between studies (Griffith *et al.* 2017). Where some studies include holding animals
324 temporarily in captivity (<24 hours; (Quinn *et al.* 2009) to ~60 days: (Mellish *et al.* 2006),
325 even short periods of confinement may impact an individual's physiology and behaviour
326 post-release (Cooper 2011). For invertebrates, it is possible to hold and breed many species
327 in captivity in large numbers. When obtaining breeding stocks, it is advisable to do so from
328 established captive colonies where these exist (Harvey-Clark 2011).

329

330 **3. WELFARE CONSIDERATIONS IN ECOLOGICAL MANIPULATIONS AND** 331 **EXPERIMENTAL APPROACHES**

332 There is widespread use of ecological and environmental manipulations on wild animals in
333 the field. These studies are undoubtedly important in disentangling complex processes, yet
334 few studies properly consider the resulting welfare impact (Cuthill 1991). There is real
335 diversity in the type and nature of experiments and manipulations carried out in the wild
336 (Table 4). Many of these studies directly aim to induce some sort of change that impacts
337 fitness, but it is important to consider longer term and lifelong impacts on individuals.
338 Where studies are likely to have foreseeable direct harm, it is important to consider the

339 balance of risk and reward (Emlen 1993) and utilize frameworks such as the 3Rs in study
340 design (Cuthill 2007) with evidence-based justification of samples sizes, e.g. power
341 analysis. Since manipulation studies can, and do, impact individual animals as part of their
342 aims, it is important that journals and referees interrogate the study's design thoroughly,
343 ensuring full justification of the method.

344 Researchers should also generally consider the unintended consequences of any work
345 in the field. Researchers may change the environment (see Fedigan 2010) either by direct
346 action or through the presence of the researcher, e.g. impacting predation rates (Isbell &
347 Young 1993). Similarly, studies that manipulate the environment can have ecosystem-wide
348 effects, such as changing species assemblages (Thompson 1982).

349

350 **4. THE WELFARE IMPLICATIONS OF THE COGNITIVE ABILITIES OF** 351 **THE STUDY SPECIES**

352 Our understanding of animal sentience, the ability of an animal to experience positive and
353 negative affective states (Duncan 2006), is inextricable to our perception of the cognitive
354 abilities of that particular species. Researchers must consider the cognition of their study
355 species and the implications of their research on the animal as a result of this.

356 Unfortunately, there are still vast gaps in our knowledge of cognition across the animal
357 kingdom and our general perception of a species' cognition is not necessarily reflective of
358 their actual cognitive abilities. Recent research has found remarkable cognitive abilities in
359 species that are traditionally considered unintelligent (e.g. Matsubara, Deeming &
360 Wilkinson, 2017). This presents a challenge to our knowledge of animal sentience.

361 Researchers should familiarise themselves with information regarding the cognitive
362 abilities of their study species and, where there is uncertainty around their cognitive

363 abilities, they should be treated as though they have the capacity for both positive and
364 negative affective state (Chan, 2011).

365

366 5. **END-POINTS: THE CONSIDERATION OF RELEASE, REHOMING AND** 367 **EUTHANASIA FOR WILD ANIMALS**

368 During work involving wild animals, researchers will be faced with a choice of how to
369 proceed at the end of any capture event or study. The available options are normally
370 limited to keeping the animal in captivity temporarily or indefinitely, releasing it back into
371 the wild, or euthanasia, depending on local or national regulations. We note that use of the
372 term euthanasia (as opposed to killing, which we have used more generally throughout the
373 paper) is reserved for those situations where killing is not only carried out humanely, but
374 also to the benefit of the animal (Broom 2007).

375

376 **5.1 Release of wild animals**

377 Where capture, handling, and processing durations are rapid, animals should—wherever
378 practically, legally and ecologically feasible—be released back at the site of capture when
379 they have fully recovered from procedures (Box 6). For animals held for long time periods,
380 their absence from the social group, territory, or home range can cause changes in status
381 with knock-on impacts for resource retention (Krebs 1982). If animals are released after
382 being held in captivity, as small a number as possible should be used, based upon sample
383 size calculations. In addition, if kept for extended periods in captivity, reintroduction is
384 needs to be carefully managed. Unless animals are bred specifically for release, i.e.
385 research surrounding reintroduction programmes for conservation or restocking of wild
386 populations, wild animals bred in captivity are generally unsuitable for release into the
387 wild.

388

389 **5.2 Injured or sick wild animals**

390 It is inevitable that researchers will encounter, or unintentionally cause, sickness or injury
391 to wild animals. When faced with a sick or injured wild animal there are three possible
392 courses of action: no intervention; treatment; or euthanasia (Kirkwood, Sainsbury &
393 Bennett 1994). From a purely welfare perspective, there are circumstances under which
394 each of these is justifiable. Treatment is justifiable if an animal is likely to recover without
395 treatment but its welfare will be improved by treatment (e.g. by reducing the time to
396 recovery), or if the animal is unlikely to recover without treatment and treatment—with
397 subsequent management and release—can be accomplished with relatively little stress to
398 the animal. Treatment can involve minor procedures such as cleaning wounds and
399 administering antibiotics (Elbroch *et al.* 2013) to minor stitching (Melton 1980). In most
400 countries, such treatment must be conducted by, or under the guidance of a veterinarian.
401 From the perspective of wildlife research, rapid *in situ* treatment is preferable. Choosing to
402 treat a wild animal is therefore an important part of contingency planning during the design
403 stage (Box 2).

404 In rare cases, injured wildlife may be brought into captivity for rehabilitation, but
405 this should only be considered in extreme cases. For most researchers, there is insufficient
406 capacity for the housing and treatment of wild animals for extended periods of time. If a
407 wild animal requires such a significant degree of rehabilitation, then dedicated
408 rehabilitation centres or euthanasia should be considered as the only options. If animals are
409 to be released from rehabilitation centres, careful consideration needs to be given to the
410 impact of release on host populations (Mullineaux 2014).

411

412

413 **5.3 Euthanasia**

414 Inevitably, there will be circumstances when wild animals will need to be euthanised. This
415 is performed when an animal's pain and/or distress is substantial and/or giving treatment is
416 not possible (Figure 2), or where post-study release is not feasible (e.g. many invertebrate
417 studies). Once the decision to euthanise has been made, it is the researcher's responsibility
418 to ensure that it is conducted in a way that minimises pain, distress, and time to clinical
419 death. In evaluating methods of euthanasia, researchers should consider the following key
420 factors: (1) their ability to induce loss of consciousness and death with minimal pain and
421 distress; (2) time required to induce loss of consciousness; (3) reliability of method; (4)
422 safety of personnel; (5) irreversibility of method; (6) compatibility with intended animal
423 use and purpose; (7) documented emotional effect on observers or operators; (8)
424 compatibility with subsequent evaluation, examination, or use of tissue; (9) drug
425 availability and human abuse potential; (10) compatibility with species, age, and health
426 status; (11) ability to maintain equipment in proper working order; (12) safety for predators
427 or scavengers should the animal's remains be consumed; (13) legal requirements; and (14)
428 environmental impacts of the method of disposal of the animal's remains (AVMA 2013).

429 Methods of euthanasia are exceptionally varied, and it is beyond the scope of this
430 review to cover them all (but see Leary *et al.* 2013). Preparation beforehand is critical,
431 especially knowing the identity and availability of the responsible person with the
432 appropriate level of training and experience. Species that are less commonly used should
433 have appropriate methods and guidance drawn up in advance of the work (e.g.
434 cephalopods: Andrews *et al.* 2013). There is continued debate about the use of certain
435 methods (e.g. for reptiles and amphibians: Lillywhite *et al.* 2017), so it is important to
436 check current, up-to-date guidance and periodically check for refinements in euthanasia
437 protocols. Appropriate methods for euthanasia of invertebrates, including cephalopods,

438 requires further study, but there is existing taxa-specific guidance available (see Murray
439 2006; Andrews *et al.* 2013).

440 Death must be confirmed before disposal of animal remains. A combination of criteria
441 is most reliable in confirming death. In mammals and birds these include a lack of central
442 pulse, breathing, corneal reflex and response to firm toe pinch, inability to hear respiratory
443 sounds and heartbeat through a stethoscope, greying of the mucous membranes, and *rigor*
444 *mortis*. None of these signs alone, except rigor mortis, confirms death. For other taxa,
445 death must be verified carefully using taxa-specific criteria (Andrews *et al.* 2013;
446 Lillywhite *et al.* 2017). Animal remains must be handled appropriately and in accordance
447 with local or national legislation. Regulations apply not only to the disposal of remains, but
448 also the management of chemical residues (e.g. medicines, euthanasia agents) that have the
449 potential to cause secondary poisoning.

450

451 6. KEY RECOMMENDATIONS TO RESEARCHERS AND PUBLISHERS

452 Throughout this paper, it has been clear that there needs to be greater emphasis on the
453 ethical standards of studies conducted on wild animals. Journals often require varying
454 amounts of details about the welfare precautions taken, state of the animals, and the
455 procedures undertaken with justification; many published papers have neglected to include
456 such key information (Field *et al.*, 2019). Journals must take a more active role in
457 protecting animal welfare as a ‘critical control point’ for publications.

458 To move forward, we have three key recommendations:

- 459 1. Any research proposal involving the use of animals—including invertebrates—should
460 embed the 3Rs (Box 2) or 9Rs (Curzer *et al.* 2013) firmly within the design phase of
461 the study and, where possible, include and report post-study or post-experimental
462 monitoring.

- 463 2. The research proposal should be subject to ethical review prior to study
464 commencement. The ethics committee, and reference number, should be identified in
465 the publication's methods or ethics section to allow reviewers and editors to query the
466 ethical review independently. Retrospective applications to an ethics committee
467 should be clearly identified as such within the manuscript and should only be
468 approved if replication of the work would result in significant further harm, and the
469 original work would have otherwise been approved using standardised approaches.
- 470 3. There needs to be standardised reporting of key information in methods and results for
471 all studies using wild animals. For some time, these have been used or advocated in
472 laboratory animal work (Kilkenny *et al.* 2010), a similar standard for wild animals is
473 critical (ARROW: Field *et al.* 2019). Within this, details of the impacts of experiments
474 should be included even if they are not part of the study, e.g. injury and mortality
475 rates. A key future aim should be to use the availability of data in publications to
476 inform future welfare guidance in areas that have currently little research or
477 information.

478

479 7. CONCLUSIONS

480 Wildlife research is an exceptionally broad subject that incorporates a wide variety of
481 study types on many different species and in wildly differing locations. In all areas of
482 research on wild animals, the concept of welfare remains the same. Consideration of
483 welfare should be paramount when studies are designed and conducted to safeguard the
484 welfare of the study animals and improve the quality of science. Whilst this paper is not
485 meant to be the definitive guide to wild animal welfare, it represents a condensed
486 information source that crystallises key areas of ethical and welfare concern and highlights
487 specific areas that need future study. We stress the need for clear reporting and minimum

488 requirements with regard to research practice (Bodey *et al.* 2017; Field *et al.* 2019). Clear
489 reporting in published articles will allow the research community to benefit from collective
490 information to enhance and refine research techniques for wild animals.

491

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495

496 **Dedication**

497 Since writing this paper, our colleague Professor Victoria Braithwaite has sadly passed
498 away. Victoria was an inspirational scientist and hugely influential in the field of animal
499 behaviour and welfare. The authors wish to acknowledge Victoria's contributions both to
500 this paper and to scientific thinking in this area. Thank you Victoria, you are very much
501 missed.

502

503 **Author contributions**

504 CDS, HG, LS drafted the main text, with all authors (LC, RE, AW, VB, SC, CDS, LS,
505 HG) contributing to sections and to revisions.

506

507 **8. REFERENCES**

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942

943 **FIGURE LEGENDS**

944 **Figure 1:** Decision tree for marking wild animals

945 **Figure 2:** End-point decision tree: the consideration of release, rehoming and euthanasia

946 for wild animals

947

Table 1: Examples of capture-related mortality and injury across different methods in vertebrates and invertebrates

Taxa	Method	% injury	% mortality	Reference
Birds	Mist netting	0.59%	0.23%	Spotswood <i>et al.</i> 2012
Birds	Canon-netting	0.42%	0.1%	O'Brien <i>et al.</i> 2016
Mammals	Longworth traps		<1%-10.4%	Jacob <i>et al.</i> 2002; Anthony <i>et al.</i> 2005; Jung 2016
Mammals	Sherman traps		10-93%	Schonfield <i>et al.</i> 2013
Mammals	Box trap	0-87%	0%	Iossa <i>et al.</i> 2007
Mammals	Leg hold snare	18-100%	0-3%	Iossa <i>et al.</i> 2007
Mammals	Leg-hold snare			Iossa <i>et al.</i> 2007
Mammals	Darting		0-20%	Haulton, Porter & Rudolph 2001
Mammals	Box trap		0-7.6%	Haulton, Porter & Rudolph 2001
Mammals	Clover trap		0.9-20.7%	Haulton, Porter & Rudolph 2001
Mammals	Canon net		4.6-10%	Haulton, Porter & Rudolph 2001
Fish	Electrofishing	0-50.3%		Culver & Chick 2015
Fish	Trammel net		44%	Chopin, Arimoto & Inoue 1996
Fish	Rod and line		3.4-4.3%	Chopin <i>et al.</i> 1996; Albin & Karpov 1998
Herptiles	Funnel trap		1.1-23.4%	Enge 2001; Jenkins, McGarigal & Gamble 2003
Herptiles	Pitfall trap		1.0-19.4%	Enge 2001; Jenkins, McGarigal & Gamble 2003
Crustacean	Trawl		1.2-21%	Blackburn & Schmidt 1988

Table 2. Examples of impacts of marking and tagging to the health and welfare of wild animals.

Taxa	Mark or device	Impact category	Details	Reference
Echinoidea	Fluorochrome markers	Survival; Health	Some markers resulted in a growth slowing in the month post-marking. Six-months post-marking there were no differences between controls and marked individuals in growth rate, survival, gonad production or jaw weight.	Ellers & Johnson 2009
Arthropoda	Nail varnish; queen bee marker	Survival; Behaviour	No impacts of marking on survival, but marked individuals showed reduced activity and increased hiding compared to controls.	Drahokoupilova & Tuf 2012
Gastropoda	Glued plastic marks; gouache paint; car body paint; nail varnish; corrective fluid	Reproduction; Survival	There were no effects of any of the marking treatments on life history traits or survival of the animals.	Henry & Jarne 2007
Fish	Surgically or gastrically implanted radio transmitter	Behaviour; Health	Devices weighing 2.3-5% of body mass. Gastrically implanted fish had slower growth, mouth abrasions caused by antennae and impaired feeding behaviour. Inflammation was present for 22% of fish that had surgery.	Adams <i>et al.</i> 1998
Mammals	GPS collar	Behaviour	Distances travelled and home range sizes were smaller when cats wore a collar weighing ~ 3% of body mass, compared to those weighing <1% or ~2%.	Coughlin & van Heezik 2014
Mammals	Radio collar	Social	Changes in dominance structure were not affected by collars weighing < 10% body mass, but voles lost dominance when their collar was > 10% body mass.	Berteaux <i>et al.</i> 1994

Birds	Transmitter in a back harness	Behaviour; Health; Physiology	Transmitters weighing either 2.5% or 5% of the bird's body mass slowed down flight times to a similar extent on 90 and 320km journeys. Pigeons produced 85-100% more CO ₂ on the longer journey with a transmitter than with no equipment attached.	Gessaman & Nagy 1988
Mammals	Toe clipping	Survival	Males lived 2.1 weeks less than non-clipped controls. No effects on female survival.	Pavone & Boonstra 1985
Mammals	Toe clipping	Health; Survival	No infection caused by toe clipping, no growth impacts and no effects on survival in captivity or the wild.	Fisher & Bloomberg 2009
Mammals	Toe clipping	Behaviour; Health; Survival	No impact of toe clipping on body weight or survival. Newly clipped animals travelled further, but may be due to handling effects.	Borremans <i>et al.</i> 2015
Herptiles	Toe clipping	Survival	Toe clipping decreased the return rate of animals as a function of the number of toes removed	McCarthy & Parris 2004
Birds	Ringling	Survival	Decreased life expectancy (28% shorter) for individuals without conspicuous rings than for those with inconspicuous rings.	Tinbergen <i>et al.</i> 2014
Birds	Flipper bands	Survival	Banded penguins had lower breeding probability and lower chick production. Survival rate of banded chicks after 2–3 years was significantly reduced.	Gauthier–Clerc <i>et al.</i> (2004)
Birds	Geolocator in backpack-style harnesses	Aerodynamics	Increased drag for backpack-style harnesses, compared with no harness. Drag was higher when the device was between the wings than when on the rump.	Bowlin <i>et al.</i> 2010
Birds	Geolocator attached to leg	Reproduction	Reduced return rates; reduced nesting success; increased partial clutch failure for three out of 23 taxa studied.	Weiser <i>et al.</i> 2016

			Mounting perpendicular to the leg increased negative effects on nesting, compared with parallel to the leg. No impact for 20 of the taxa studied.	
Birds	Implantation of intracoelomic devices	Reproduction	Three years post-implantation, 16% lower yearly survival than non-implanted group. Only three eggs were found from two implanted birds and all three were deformed.	Hooijmeijer <i>et al.</i> 2014
Fish	Implanted interperitoneal acoustic transmitter	Behaviour and physical health	Short term effects (first five days post-tagging) on behaviour, though not seen long-term. Incisions for implantation were well-healed and clean upon recapture.	Gardner <i>et al.</i> 2015
Herptiles	Multiple electronic tags attached to shell	Behaviour; hydrodynamics	Tags had negligible impacts on adult drag (< 5% additional drag), but increased drag significantly (> 100%) for juvenile turtles. Potential negative impact on an individual's ability to conduct standard behavioural repertoire	Todd Jones <i>et al.</i> 2013
Herptiles	Implantation of intracoelomic devices	Health	Inflammation in 66% of tested snakes and bacterial infection in 33%.	Lentini <i>et al.</i> 2011
Mammals	GPS collar	Behaviour	Negative impact on feeding behaviour, with heavier collars reducing the animals' rate of travel by > 50% when in the foraging patch and drinking area.	Brooks <i>et al.</i> 2008
Mammals	Implanted intraperitoneal radio-transmitter	Health	Mortality caused by severe constipation in two animals (the device compressed the colon) and dystocia in another.	Lechenne <i>et al.</i> 2012

1

2 Table 3: Key considerations for choosing to capture and kill animals for scientific research.

3Rs	Theme	Priority	Considerations
Replacement	Research Question	1	Does the research question require animals to be captured and killed? Can alternatives be used – with non-animals or live animals?
Refinement	Techniques	2	Can different research techniques be used? Cost should not be used as justification for killing animals, compared to other, non-lethal techniques.
Refinement	Source	3	Can existing samples or sources of dead animals be used? Can sample collection avoid collecting new animals?
Reduction	Sample size	4	Can minimal sample sizes be used? If large numbers are needed, then these need to be clearly justifiable with a power analysis.
Refinement	Method	5	The most humane, selective method must be used to kill animals.

3

4 Table 4: Examples of different manipulation type experiments and direct and long-term effects
 5 on individuals

Manipulation type	Direct Effect	Long term effect	Reference
Vaccination study	Increasing immune response	Reduced survival	Soulsbury <i>et al.</i> 2018
Increased egg production	Reduced breeding female condition Reduced chick production Smaller chick size		Monaghan <i>et al.</i> 1998
Breeding female removal	Infanticide		Emlen <i>et al.</i> 1989
Hormone increase	Increased breeding attempt Sexual ornament size increase	Reduced survival Reduced sexual ornament size	Siitari <i>et al.</i> 2007
Playback of predator calls	Reduced incubation behaviour		Ibanez-Alamo & Soler 2012
Playback of predator calls	Reduced clutch size		Egger <i>et al.</i> 2006
Reduced female plumage brightness	Reduced offspring quality		Berzin & Dawson 2018
Induced tail loss in lizards	Reduced survival		Fox & McCoy 2000
Food supplementation	Altered egg composition		Siitari <i>et al.</i> 2014

6

7

8

9 **BOX 1: 3Rs CHALLENGES FOR WILD ANIMAL RESEARCH**

10 **Reduction:** A key aim of the 3Rs is to minimise the number of animals used. It is challenging to
11 translate Reduction into practice in wild animal research for several reasons: (i) genetic variation
12 is generally greater in wild animals, meaning they respond more heterogeneously to a given set
13 of conditions. This increased variation often necessitates larger sample sizes than captive
14 populations; (ii) the environmental variation of animals is considerably greater than in controlled
15 laboratory conditions, meaning larger sample sizes are required; (iii) in wild-based studies,
16 animals will be lost due to natural mortality or other random events. Conducting pre-study power
17 analysis is therefore especially important (Steidl, Hayes & Schaubert 1997).

18

19 **Replacement:** In laboratory-based research, 98% of all animals used are rodents (UK Home
20 Office 2014). The 3Rs principles promote the use of the lowest sentient forms where possible.
21 In biomedical research, the typical targets are to move towards more *in vitro* and *in silico*
22 research. This is possible because the research focus is a physiological, genetic or other
23 biochemical response within the animal. In wild animal research, Replacement is often not
24 possible as the study focus is often at the level of individual animals, and their interactions
25 within the wider ecosystem. There are scenarios where a species considered less sentient or less
26 protected could be used to test hypotheses (Lane & MacDonald, 2010; Sneddon, Halsey & Bury
27 2017); in practise such scenarios are likely to be rare, or difficult to generalise with confidence
28 without confirmation at the higher/more protected level.

29

30 **Refinement:** A greater diversity of non-invasive methods has been devised in wild studies,
31 compared to lab-based studies. One driver of this is the need to return animals to the wild as
32 quickly as possible or because techniques may harm the species or population. Approaches such
33 as DNA analysis from the collection of hair or faeces have been well established. There is still a
34 need to collaborate with other disciplines to improve and refine techniques (Cattet 2013). These
35 include greater use of remote methods of monitoring such as camera trapping (Burton *et al.*
36 2015) or passive acoustic monitoring (Gibb *et al.* 2019), and advances in analytical methods (e.g.
37 machine learning: Tabak *et al.* 2019). Though, there must be awareness that these may still have
38 a negative effect (e.g. drones: Bennitt *et al.* 2019).

39

40 **BOX 2: WELFARE CONSIDERATIONS FOR CAPTURING AND HANDLING WILD**
41 **ANIMALS**

- 42 1. **Capture methods:** Capture techniques should be as selective as possible to minimise the risk
43 of capturing non-target species. They should be species-appropriate to minimise injury and
44 mortality during capture and reduce welfare impacts. For example, considering whether the
45 study species' would benefit from being held in darkness prior to handling.
- 46 2. **Appropriate checking:** Capture devices should be checked frequently, at appropriate
47 intervals for the target species.
- 48 3. **Location:** Even if the capture technique itself has little welfare impact, undertaking capture
49 in an inappropriate location places the user and animals at risk. This includes placing traps
50 on slopes or near water. Being aware of potential predators is also important. Trapping
51 individuals near breeding sites may lead to offspring abandonment.
- 52 4. **Seasonal timing:** Some species are sensitive to disturbance during key parts of their life
53 cycle. This includes keeping animals away from dependent young for long periods.
- 54 5. **Time of day:** Animal's circadian activities should be considered. Nocturnal animals should
55 not be released during daytime, and individuals should have enough time to forage after
56 release.
- 57 6. **Weather:** Researchers should avoid capturing animals when weather conditions may lead to
58 hyper- or hypothermia. If necessary, regular monitoring of capture sites and provision of
59 bedding should be considered. Researchers should avoid using capture sites with high sun
60 exposed for parts of the day.
- 61 7. **How many times:** Capture events should be minimised, but where captures are necessary,
62 researchers should take care to avoid repeated capture of the same individual. This may
63 mean moving capture locations, or cessation of capturing for set time periods. If capture is
64 for removal of tags/devices, consider whether self-removing tags/devices can be used.
- 65 8. **Contingency planning:** Before trapping begins, researchers must have management plans in
66 place for animals that are injured or killed during capture. Plans should include evaluating
67 injuries, determining when euthanasia is appropriate, and ensuring that persons who will
68 conduct this are trained and licensed.
- 69 9. **How many animals?** A clear maximum number of animals caught at any one time must be
70 considered and numbers should be based upon power analyses. This ensures researchers can
71 safely process animals in as short a time as possible to minimise capture and handling time.

72 **10. Minimise the number of procedures:** The cumulative impacts of procedures (even minor
73 procedures) on study animals is a poorly understood area for most laboratory species, and
74 unknown for wild species. Reducing the number of procedures an individual is subjected to
75 has the benefit of reducing direct handling time.

76

77 ***BOX 3: KEY QUESTIONS WHEN MARKING/TAGGING WILD ANIMALS***

- 78 1. If using natural marks, will data collection interfere with the species biology?
- 79 2. How long does the mark or tag need to last to complete the study; and how durable is the
80 proposed marking method?
- 81 3. Will the proposed marking/tagging method interfere with other studies?
- 82 4. Will the marks/tag promote public concern about the study; and will the marks/tag have to
83 be removed after study completion?
- 84 5. Have the appropriate approvals (animal welfare and state and/or federal permits) to
85 mark/tag animals been obtained?
- 86 6. Will the mark have any direct or indirect effect on survival or behaviour? Can alternative
87 methods be used or mitigated e.g. reducing size of mark?

88

89 **BOX 4: NC3Rs BEST PRACTICE FOR WILD VERTEBRATE TRANSPORT GUIDELINES**

90 Some wild animals will undergo transportation from the field to a captive housing location.
91 Although longer distances need additional planning and care, it is important to note that any
92 transport can be a significant stressor that may impact animal welfare and study research
93 outcomes. The primary objective should be to move the animals in a manner that does not
94 jeopardise their well-being and ensures their safe arrival at their destination in good health, with
95 minimal distress. Many aspects of the transport process need to be considered, including: the
96 route and journey plan; container design; vehicle design; the competence and attitude of drivers
97 and others involved in the transportation; travel duration; the nature of food and water supplies;
98 arrangements for acclimatisation after transport.

99 Critical appraisal and refinement of the logistical aspects of transport is essential if animal
100 welfare is to be safeguarded during journeys. Guidance is available from a working group of the
101 UK Laboratory Animal Science Association (LASA) (Swallow *et al.* 2005) and the US Institute
102 for Laboratory Animal Research (ILAR) guidelines for the humane transportation of research
103 animals (National Research Council 2006).

104 It is important that all relevant legislation on animal transport is followed - designating a person
105 in each establishment with responsibilities on understanding and implementing transport
106 legislation will help to ensure compliance.

107 Within Europe, Council Regulation (EC) No. 1/2005 on the protection of animals during
108 transport and related operations determines minimum standards for the welfare of animals during
109 transport. The Regulation applies to the transport of all live vertebrate animals for the purposes
110 of economic activity, i.e. a business or trade. It is implemented in England by The Welfare of
111 Animals (Transport) (England) Order 2006 and by parallel legislation in Scotland, Wales and
112 Northern Ireland. Defra has published an overview of the requirements of the Regulation.
113 European Convention for the Protection of Animals during International Transport (Revised)
114 (2006) also applies to the movement of live animals within the EU. The transport of live animals
115 by air is governed by the Live Animals Regulations of the International Air Transport
116 Association (IATA). CITES permits must be obtained for all movements (import and export) of
117 CITES listed species (e.g. non-human primates) between countries signed up to the Convention.

118

119 ***BOX 5: MINIMUM CHECKLIST OF WELFARE CONSIDERATIONS FOR HOUSING***
120 ***WILD ANIMALS***

121 If your study design requires wild animals to be housed in captivity, the following checklist
122 should be completed alongside ethical approval documentation.

123 **Housing arrangements**

- 124 How do the housing arrangements meet the daily needs of your study species?
- 125 housing type
- 126 space allowance per individual
- 127 temperature
- 128 humidity
- 129 lighting
- 130 noise levels
- 131 food and water access
- 132 social conditions
- 133 Have the housing conditions been checked by a suitable expert (e.g. veterinarian)?
- 134 How do the proposed cleaning regimes for the housing meet the needs of your study species
135 and help to prevent the spread of infection?
- 136 cleaning schedule
- 137 cleaning products to be used
- 138 protocol for moving animals during cleaning
- 139 Has the proposed cleaning regimes checked and approved by a relevant expert (e.g. a
140 veterinarian)?
- 141 Will individual animals be checked for infections prior to entering housing?
- 142 What is the protocol for housing infected animals?
- 143 What biosecurity procedures are in place upon entry and exit of the housing area?

144

145 ***BOX 6: WELFARE CONSIDERATIONS FOR RELEASE OF WILD ANIMALS***

- 146 1. Check legislation regarding release of wild animals. Is it legal?
- 147 2. Are animals healthy enough to be released, including having recovered fully from any
- 148 procedures or anaesthesia?
- 149 3. Release the animal as soon as it is feasible to do so, with attention paid to:
- 150 a. conspecifics and dependent young
- 151 b. time of day
- 152 c. likely harm to animal
- 153 4. Release site should be as close to capture site as is safe for the animal.
- 154 5. Confirm that:
- 155 a) it is legal to release the animals
- 156 b) that the animal's state of health allows it to be released or re-homed;
- 157 c) that the animal poses no danger to public health, animal health or to the
- 158 environment;
- 159 d) that there is an adequate scheme in place for ensuring the socialisation of the
- 160 animal upon being released or re-homed where appropriate;
- 161 e) that appropriate measures have been taken to safeguard the animal's welfare
- 162 when released or re-homed.

163

164

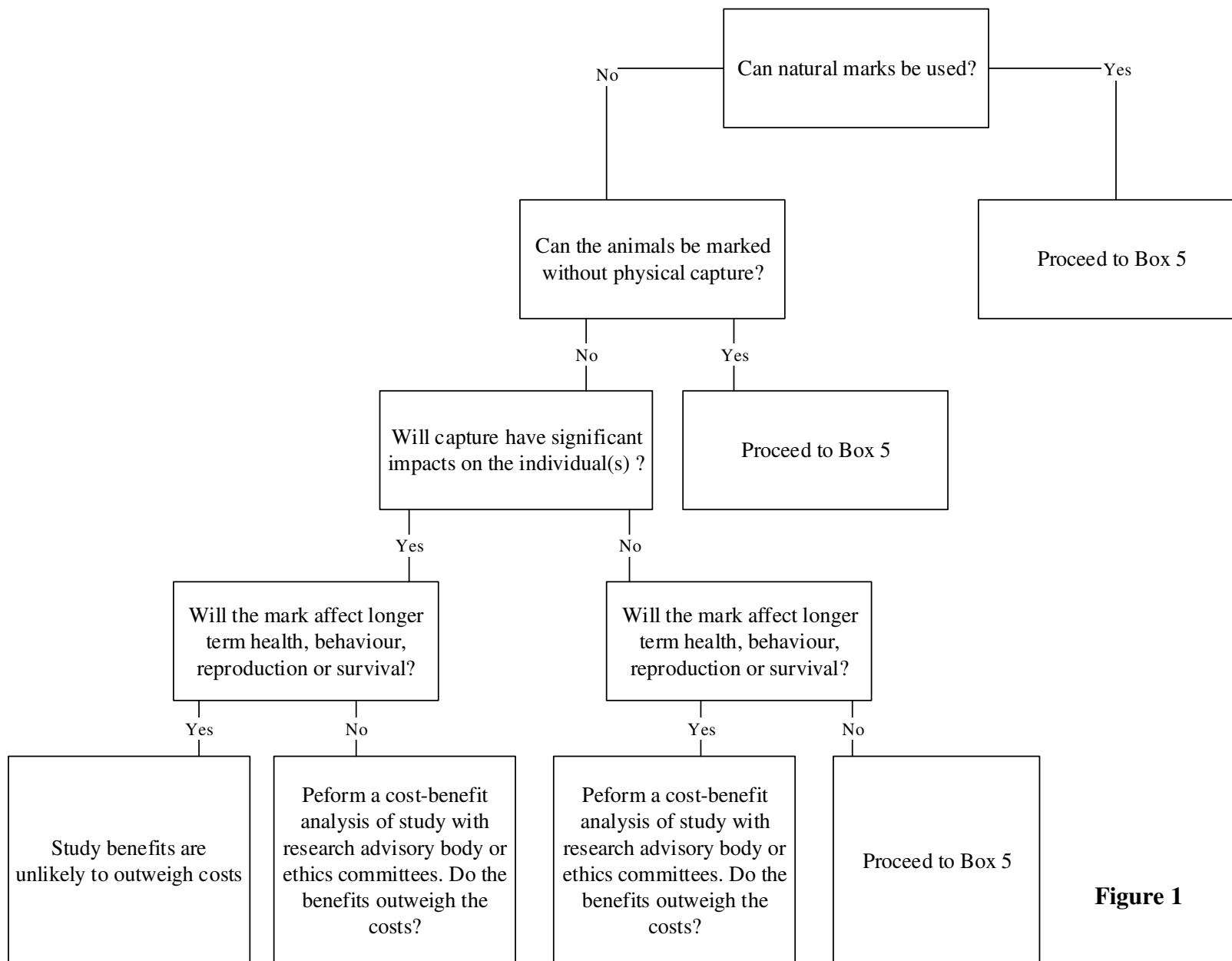


Figure 1

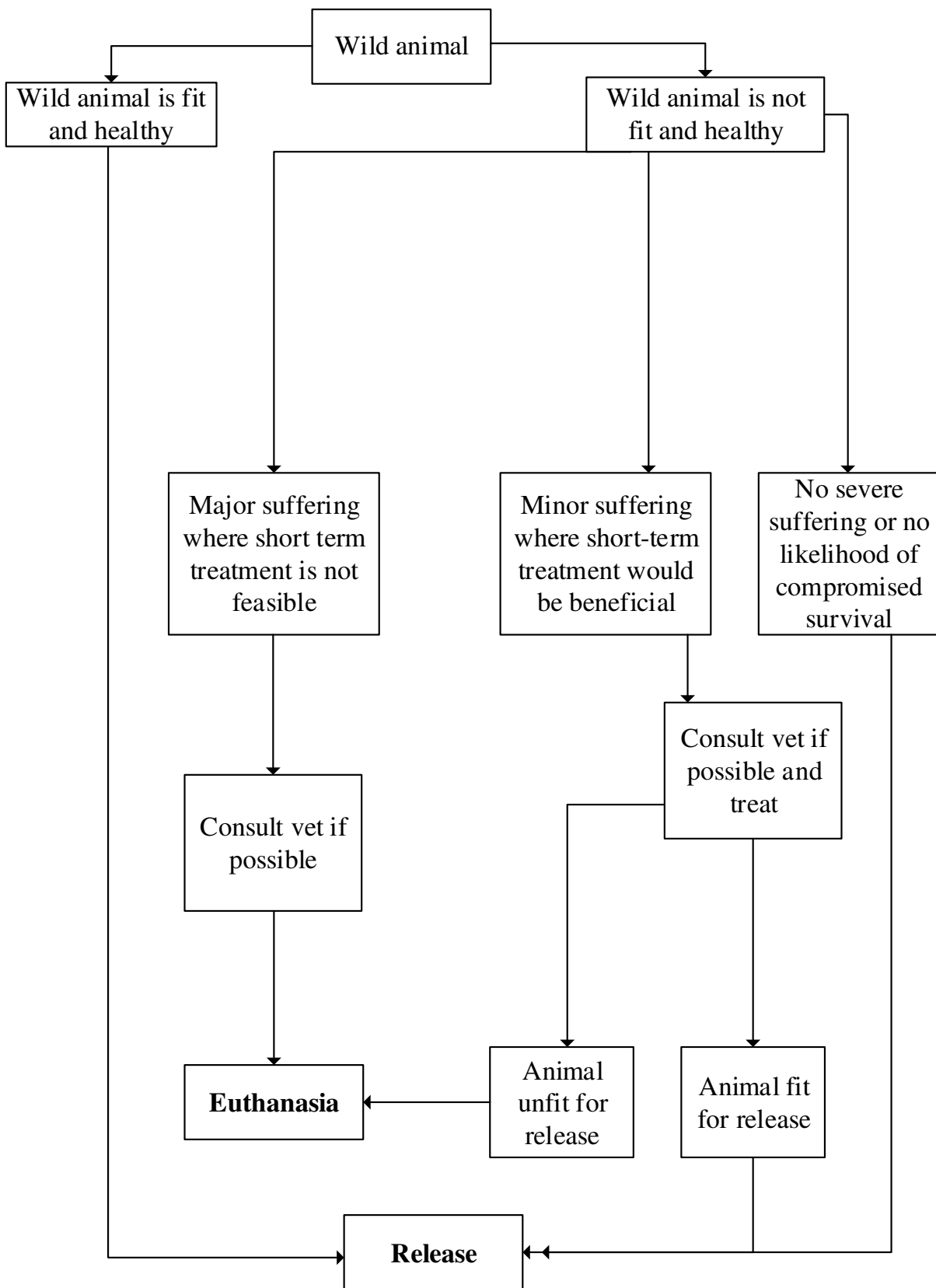


Figure 2