Generating and testing ecological hypotheses at the pondscape with environmental DNA metabarcoding: a case study on a threatened amphibian

Lynsey R. Harper[1,2*], Lori Lawson Handley[1], Christoph Hahn[1,3], Neil Boonham[4,5], Helen C. Rees[6], Erin Lewis[4], Ian P. Adams[4], Peter Brotherton[7], Susanna Phillips[7] and Bernd Hänfling[1]

[1] Department of Biological and Marine Sciences, University of Hull, Hull, HU6 7RX, UK

[2] Illinois Natural History Survey, Prairie Research Institute, University of Illinois at Urbana-Champaign, Champaign, Illinois, USA

[3] Institute of Zoology, University of Graz, Graz, Styria, Austria

[4] Fera Science Ltd (Fera), Sand Hutton, York, YO14 1LZ, UK

[5] Newcastle University, Newcastle upon Tyne, NE1 7RU, UK

[6] ADAS, School of Veterinary Medicine and Science, The University of Nottingham, Sutton Bonington Campus, Leicestershire, LE12 5RD, UK

[7] Natural England, Peterborough, PE1 1NG, UK

***Corresponding author: Lynsey R. Harper**

Illinois Natural History Survey, Prairie Research Institute, University of Illinois at Urbana-Champaign, Champaign, Illinois, USA

E-mail: lynsey.harper2@gmail.com

Running title: Hypothesis testing with eDNA metabarcoding

Abstract

Environmental DNA (eDNA) metabarcoding is revolutionising biodiversity monitoring, but has unrealised potential for ecological hypothesis generation and testing. Here, we validate this potential in a large-scale analysis of vertebrate community data generated by eDNA metabarcoding of 532 UK ponds. We test biotic associations between the threatened great crested newt (*Triturus cristatus*) and other vertebrates as well as abiotic factors influencing *T. cristatus* detection at the pondscape. Furthermore, we test the status of *T. cristatus* as an umbrella species for pond conservation by assessing whether vertebrate species richness is greater in ponds with *T. cristatus* and higher *T. cristatus* Habitat Suitability Index (HSI) scores. *T. cristatus* detection was positively correlated with amphibian and waterfowl species richness. Specifically, *T. cristatus* was positively associated with smooth newt (*Lissotriton vulgaris*), common coot (*Fulica atra*), and common moorhen (*Gallinula chloropus*), but negatively associated with common toad (*Bufo bufo*). *T. cristatus* detection did not significantly decrease as fish species richness increased, but negative associations with common carp (*Cyprinus carpio*), three-spined stickleback (*Gasterosteus aculeatus*) and ninespine stickleback (*Pungitius pungitius*) were identified. *T. cristatus* detection was negatively correlated with mammal species richness, and *T. cristatus* was negatively associated with grey squirrel (*Sciurus carolinensis*). *T. cristatus* detection was negatively influenced by larger pond area, presence of inflow, and higher percentage of shading, but positively correlated with HSI score, supporting its application to *T. cristatus* survey. Vertebrate species richness was significantly higher in *T. cristatus* ponds and broadly increased as *T. cristatus* HSI scores increased. We reaffirm reported associations (e.g. *T. cristatus* preference for smaller ponds) but also provide novel insights, including a negative effect of pond inflow on *T. cristatus*. Our findings demonstrate the prospects of eDNA metabarcoding for ecological hypothesis generation and testing at landscape scale, and dramatic enhancement of freshwater conservation, management, monitoring and research.

Keywords: biodiversity assessment, environmental DNA (eDNA), hypothesis testing, metabarcoding, ponds, species associations, *Triturus cristatus*

52 ## 1. Introduction

53

54 Environmental DNA (eDNA) analysis offers ecologists exceptional power to detect organisms
55 within and across ecosystems. DNA released by organisms into their environment via
56 secretions, excretions, gametes, blood, or decomposition, can be sampled and analysed
57 using different approaches to reveal the distribution of single or multiple species (Rees et
58 al., 2014; Lawson Handley, 2015). eDNA analysis combined with high-throughput
59 sequencing (i.e. eDNA metabarcoding) can yield efficient, comprehensive assessments of
60 entire communities (Deiner et al., 2017), providing a step change in biodiversity monitoring
61 (Hering et al., 2018). eDNA metabarcoding has untapped potential to generate and test
62 ecological hypotheses by enabling biodiversity monitoring at landscape scale with minimal
63 impact to communities under investigation. This potential has already been demonstrated
64 with targeted eDNA analysis by Wilcox et al. (2018), where climate-mediated responses of
65 bull trout (*Salvelinus confluentus*) to biotic and abiotic factors were revealed using
66 quantitative PCR (qPCR) on crowd-sourced eDNA samples. Although eDNA metabarcoding
67 assessments of alpha and beta diversity along environmental gradients are increasing (e.g.
68 Hänfling et al., 2016; Olds et al., 2016; Kelly et al., 2016; Evans et al., 2017; Li et al., 2018a;
69 Nakagawa et al., 2018), this tool is less commonly used for ecological hypothesis testing,
70 such as the impact of environmental stressors (Li et al., 2018b; Macher et al., 2018).

71 Aquatic ecosystems are highly suited to eDNA studies as eDNA exists in multiple
72 states with rapid modes of transport and degradation, increasing detectability of
73 contemporary biodiversity (Rees et al., 2014; Barnes & Turner, 2015). Lentic systems
74 provide further opportunities for eDNA research, being discrete water bodies with variable
75 physicochemical properties that do not experience flow dynamics (Harper et al., 2019).

76 Ponds in particular have enormous biodiversity and experimental virtue that has not been
77 maximised in previous eDNA metabarcoding assessments of this habitat (Valentini et al.,
78 2016; Evans et al., 2017; Klymus et al., 2017; Ushio et al., 2017; Bálint et al., 2018). These
79 small and abundant water bodies span broad ecological gradients (De Meester et al., 2005)
80 and comprise pondscapes - a network of ponds and their surrounding terrestrial habitat (Hill
81 et al., 2018). Pondscapes contribute substantially to aquatic and non-aquatic biodiversity
82 across spatial scales, with ponds supporting many rare and protected species in fragmented
83 landscapes (De Meester et al., 2005; Biggs et al., 2016; Hill et al., 2018). Consequently,
84 ponds are model systems for experimental validation and examination of biogeographical
85 patterns (De Meester et al., 2005). Habitat complexity and tools required for different taxa
86 with associated bias (Evans et al., 2017) and cost (Valentini et al., 2016) once hindered
87 exhaustive sampling of pond biodiversity (Hill et al., 2018), but eDNA metabarcoding may
88 overcome these barriers (Harper et al., 2019).

89 In the UK, the threatened great crested newt (*Triturus cristatus*) is an umbrella
90 species for pond conservation. The extensive literature on *T. cristatus* ecology provides an
91 excellent opportunity to validate ecological patterns revealed by eDNA metabarcoding. Both
92 biotic (e.g. breeding substrate, prey, and predators) and abiotic (e.g. pond area, depth, and
93 temperature) factors are known to influence *T. cristatus* breeding success (Langton, Beckett
94 & Foster, 2001). The *T. cristatus* Habitat Suitability Index (HSI [Oldham et al., 2000; ARG-UK,
95 2010; O'Brien et al., 2017]) accounts for these factors using 10 suitability indices that are
96 scored and combined to calculate a decimal score between 0 and 1 (where 1 = excellent
97 habitat). Larvae are susceptible to fish and waterfowl predation (Edgar & Bird, 2006; Rannap
98 & Briggs, 2006; Skei et al., 2006; Hartel, Nemes & Oellerer, 2010), and adults reportedly
99 avoid ponds containing three-spined stickleback (*Gasterosteus aculeatus*) (McLee & Scaife,

100 1992), ninespine stickleback (*Pungitius pungitius*), crucian carp (*Carassius carassius*), and
101 common carp (*Carassius carpio*) (Rannap, Lõhmus & Briggs, 2009a, b). Conversely, *T.*
102 *cristatus* and smooth newt (*Lissotriton vulgaris*) prefer similar habitat and often co-occur
103 (Rannap & Briggs, 2006; Skei et al., 2006; Rannap et al., 2009a; Denoël et al., 2013; Cayuela
104 et al., 2018). *T. cristatus* individuals thrive in ponds with good water quality as indicated by
105 diverse macroinvertebrate communities (Oldham et al., 2000; Rannap et al., 2009a). Pond
106 networks encourage *T. cristatus* occupancy (Joly et al., 2001; Rannap et al., 2009a; Hartel et
107 al., 2010; Denoël et al., 2013), but larger pond area discourages presence (Joly et al., 2001).
108 Ponds with heavy shading (Vuorio, Heikkinen & Tikkanen, 2013) or dense macrophyte cover
109 (Rannap & Briggs, 2006; Skei et al., 2006; Hartel et al., 2010) are unlikely to support viable
110 populations. *T. cristatus* individuals also depend on terrestrial habitat, preferring open,
111 semi-rural pondscapes (Denoël et al., 2013) containing pasture, extensively grazed and
112 rough grassland, scrub, and coniferous and deciduous woodland (Oldham et al., 2000;
113 Rannap & Briggs, 2006; Rannap et al., 2009a; Gustafson, Malmgren & Mikusiński, 2011;
114 Vuorio et al., 2013).

115 We assessed vertebrate communities at the pondscape using a dataset produced by
116 eDNA metabarcoding for over 500 ponds with comprehensive environmental metadata. We
117 validated eDNA metabarcoding as a tool for ecological hypothesis generation and testing,
118 and compared its outputs to previous results produced by established methods. Specifically,
119 we identified and tested biotic (community presence-absence data) and abiotic
120 (environmental metadata on ponds and surrounding terrestrial habitat) factors influencing
121 *T. cristatus* detection at the pondscape - an impractical task by conventional means.
122 Furthermore, we tested the applicability of the HSI to predict eDNA-based *T. cristatus*
123 detection. Finally, we assessed the umbrella species status of *T. cristatus* by investigating

124 whether *T. cristatus* detection and the *T. cristatus* HSI score can predict vertebrate species
125 richness of ponds.
126
127
128 ## 2. Materials and methods
129
130 **2.1 eDNA samples**
131
132 Samples were previously collected from 508 ponds as part of Natural England's Great
133 Crested Newt Evidence Enhancement Programme and from 24 privately surveyed ponds
134 using established methodology (Biggs et al., 2015), detailed in Supporting Information:
135 Appendix 1. Briefly, 20 x 30 mL water samples were collected from each pond and pooled.
136 Six 15 mL subsamples were taken from the pooled sample and each added to 33.5 mL
137 absolute ethanol and 1.5 mL sodium acetate 3 M (pH 5.2). Subsamples were pooled during
138 DNA extraction to produce one eDNA sample per pond. Targeted qPCR detected *T. cristatus*
139 in 265 (49.81%) ponds (Harper et al., 2018).
140 Environmental metadata were collected for 504 of 532 ponds (Fig. S1) by
141 environmental consultants contracted for Natural England's Great Crested Newt Evidence
142 Enhancement Programme. Metadata included: maximum depth of ponds; pond
143 circumference; pond width; pond length; pond area; pond density (i.e. number of ponds per
144 km^2); terrestrial overhang; shading; macrophyte cover; HSI score (Oldham et al., 2000); HSI
145 band (categorical classification of HSI score [ARG-UK, 2010]); pond permanence; water
146 quality; pond substrate; presence of inflow or outflow; presence of pollution; presence of
147 other amphibians, fish and waterfowl; woodland; rough grass; scrub/hedge; ruderals; other

148 terrestrial habitat (i.e. good quality terrestrial habitat that did not conform to
149 aforementioned habitat types); and overall terrestrial habitat quality (see Table S1).
150
151 **2.2. eDNA metabarcoding**
152
153 We repurposed the taxonomically assigned sequence reads from Harper et al. (2018) that
154 were produced using eDNA metabarcoding of pond water to compare qPCR and eDNA
155 metabarcoding for *T. cristatus* detection. Here, we provide a summary of their eDNA
156 metabarcoding workflow (see Supporting Information: Appendix 1 for details).
157 A custom, phylogenetically curated reference database of mitochondrial 12S
158 ribosomal RNA (rRNA) sequences for UK fish species was previously constructed for eDNA
159 metabarcoding of lake fish communities (Hänfling et al., 2016). Harper et al. (2018)
160 constructed additional reference databases for UK amphibians, reptiles, birds, and
161 mammals. Reference sequences available for species varied across vertebrate groups:
162 amphibians 100.00% (*N* = 21), reptiles 90.00% (*N* = 20), mammals 83.93% (*N* = 112), and
163 birds 55.88% (*N* = 621). Table S2 lists species without database representation, i.e. no
164 records for any species in a genus. Sanger sequences were obtained from tissue of *T.*
165 *cristatus*, *L. vulgaris*, Alpine newt (*Ichthyosaura alpestris*), common toad (*Bufo bufo*), and
166 common frog (*Rana temporaria*) to supplement the amphibian database. The complete
167 reference databases compiled in GenBank format were deposited in a GitHub repository
168 and permanently archived (https://doi.org/10.5281/zenodo.1188710) by Harper et al.
169 (2018). Reference databases were combined for *in silico* validation of published 12S rRNA
170 primers 12S-V5-F (5'-ACTGGGATTAGATACCCC-3') and 12S-V5-R (5'-
171 TAGAACAGGCTCCTCTAG-3') (Riaz et al., 2011) using ecoPCR software (Ficetola et al., 2010).

172 Set parameters allowed a 50-250 bp fragment and three mismatches between each primer
173 and reference sequence. Primers were validated *in vitro* for UK fish by Hänfling et al. (2016)
174 and by Harper et al. (2018) for six UK amphibian species (Fig. S2).

175 eDNA samples were amplified in triplicate with the aforementioned primers, where
176 PCR positive controls (six per PCR plate; *n* = 114) were cichlid (*Rhamphochromis esox*) DNA
177 (0.284 ng/µL) and PCR negative controls (six per PCR plate; *n* = 114) were sterile molecular
178 grade water (Fisher Scientific UK Ltd, UK). PCR products were individually purified using
179 E.Z.N.A® Cycle Pure V-Spin Clean-Up Kits (Omega Bio-tek, GA, USA) following the
180 manufacturer's protocol. Purified product was pooled according to sample for the second
181 PCR which bound Multiplex Identification tags to the purified products. PCR products were
182 individually purified using magnetic bead clean-up and quantified with a Quant-IT™
183 PicoGreen™ dsDNA Assay (Invitrogen, UK). Samples were normalised, pooled, and libraries
184 quantified using a Qubit™ dsDNA HS Assay (Invitrogen, UK). Libraries were sequenced on an
185 Illumina® MiSeq using 2 x 300 bp V3 chemistry (Illumina, Inc, CA, USA) and raw sequence
186 reads processed using metaBEAT (metaBarcoding and Environmental Analysis Tool) v0.97.7
187 (https://github.com/HullUni-bioinformatics/metaBEAT).

188 After quality filtering (phred score Q30) and trimming (length filter of 90-110 bp)
189 using Trimmomatic v0.32 (Bolger, Lohse, & Usadel, 2014), merging (10 bp overlap minimum
190 and 10% mismatch maximum) using FLASH v1.2.11 (Magoč & Salzberg, 2011), chimera
191 detection using the uchime algorithm (Edgar et al., 2011) in vsearch v1.1 (Rognes et al.,
192 2016), and clustering (97% identity with 3 sequences minimum per cluster) with vsearch
193 v1.1 (Rognes et al., 2016), non-redundant query sequences were compared against the
194 reference database using BLAST (Zhang et al., 2000). Putative taxonomic identity was
195 assigned using a lowest common ancestor (LCA) approach based on the top 10% BLAST

196 matches for any query matching with at least 98% identity to a reference sequence across
197 more than 80% of its length. Unassigned sequences were subjected to a separate BLAST
198 against the complete NCBI nucleotide (nt) database at 98% identity to determine the source
199 via LCA as described above. The bioinformatic analysis was archived
200 (https://doi.org/10.5281/zenodo.1188710) by Harper et al. (2018) for reproducibility.
201
202 **2.3 Data analysis**
203
204 *2.3.1 Dataset refinement*
205
206 Analyses were performed in R v.3.4.3 (R Core Team, 2017). Data and R scripts have been
207 deposited in a dedicated GitHub repository for this study, which has been permanently
208 archived (https://doi.org/10.5281/zenodo.3365703). Assignments from different databases
209 were merged, and spurious assignments (i.e. non-UK species, invertebrates and bacteria)
210 removed from the dataset. The family Cichlidae was reassigned to *Rhamphochromis esox*.
211 The green-winged teal (*Anas carolinenisis*) was reassigned to *Anas* (dabbling ducks) because
212 this species is a rare migrant and available reference sequences were identical across our
213 metabarcode to those for mallard (*Anas platyrhynchos*) and Eurasian teal (*Anas crecca*),
214 which are widely distributed across the UK. Scottish wildcat (*Felis silvestris*) does not occur
215 at the sampling localities (Kent, Lincolnshire and Cheshire) and was therefore reassigned to
216 domestic cat (*Felis catus*). Wild boar (*Sus scrofa*) and grey wolf (*Canis lupus*) were reassigned
217 to domestic pig (*Sus scrofa domesticus*) and domestic dog (*Canis lupus familiaris*) given the
218 restricted distribution of *S. scrofa* and absence of *C. lupus* in the wild in the UK. The genus
219 *Strix* was reassigned to tawny owl (*Strix aluco*) as it is the only UK representative of this

genus. Where family and genera assignments containing a single UK representative had reads assigned to species, reads from all assignment levels were merged and manually assigned to that species.

Of the 114 PCR negative controls included, 50 produced no reads (Fig. S3). Reads generated for 64 of 114 PCR negative controls ranged from 0 to 49,227, and strength of each contaminant varied (mean = 0.021%, range = 0 - 100.0% of the total reads per PCR negative control). To minimise risk of false positives, we evaluated different sequence thresholds. These included the maximum sequence frequency of *R. esox* DNA in eDNA samples (100%), maximum sequence frequency of any DNA except *R. esox* in PCR positive controls (83.96%), and taxon-specific thresholds (maximum sequence frequency of each taxon in PCR positive controls). The different thresholds were applied to the eDNA samples and the results from each compared (Fig. S4). The taxon-specific thresholds (Table S3) retained the most biological information, thus these were selected for downstream analysis. Consequently, taxa were only classed as present at sites if their sequence frequency exceeded their threshold. After applying the taxon-specific thresholds, the PCR positive control (*R. esox*), human (*Homo sapiens*), and domestic species (Table S4) were removed from the dataset. Higher taxonomic assignments excluding the genus *Anas* were then removed, thus taxonomic assignments in the final dataset were predominantly of species resolution. The read count data were converted to a binary species matrix for downstream analyses using the package cooccur v1.3 (Griffith, Veech & Marsh, 2016) and Generalized Linear Mixed-effects Models (GLMMs), which were fit by maximum likelihood with the glmer function using the package lme4 v1.1-21 (Bates et al., 2015). County was included as a random effect in each GLMM to account for spatial dependencies between ponds (see Supporting Information: Appendix 1).

244

245 *2.3.2 Hypothesis generation: biotic factors potentially influencing T. cristatus detection*

246

247 We first mined the eDNA metabarcoding data and investigated the influence of vertebrate
248 group richness on *T. cristatus* detection. We used a binomial GLMM with the logit link
249 function that included species richness of other amphibians, fish, waterfowl, terrestrial
250 birds, and mammals as fixed effects (model 1, *N* = 532). We then performed a preliminary
251 analysis using the package cooccur v1.3 (Griffith, Veech & Marsh, 2016) to identify species
252 associations between *T. cristatus* and other vertebrates (*N* = 532). Hypotheses generated by
253 the eDNA metabarcoding data relating to factors influencing *T. cristatus* detection are
254 summarised in Table 1.

255

256 *2.3.3 Hypothesis testing: biotic and abiotic factors known to influence T. cristatus detection*

257

258 Identified associations from the cooccur analysis in conjunction with the existing *T. cristatus*
259 literature informed candidate biotic variables to be modelled against *T. cristatus* detection
260 (model 2, *n* = 504). The existing *T. cristatus* literature informed candidate abiotic variables to
261 be modelled against *T. cristatus* detection (model 3, *n* = 504). Hypotheses tested using the
262 eDNA metabarcoding data and associated environmental metadata to examine biotic and
263 biotic factors influencing *T. cristatus* detection are summarised in Table 2. Selection of a
264 suitable set of explanatory variables and modelling framework is fully described in
265 Supporting Information: Appendix 1. Briefly, candidate biotic and abiotic explanatory
266 variables were assessed for: collinearity using a Spearman's rank pairwise correlation matrix
267 and examination of variance inflation factors (Zuur et al., 2009), relative importance using a

classification tree within the package rpart v4.1-13 (Therneau, Atkinson & Ripley, 2014), and non-linearity using Generalized Additive Models (GAMs) (Zuur et al., 2009). We constructed separate binomial GLMMs with the logit link function for biotic (model 2) and abiotic (model 3) explanatory variables that passed these assessments.

2.3.4 Hypothesis testing: T. cristatus HSI and umbrella status

We modelled HSI score (fixed effect) separately to prevent HSI score masking variation caused by the individual biotic and abiotic variables it encompasses (model 4). Using a Poisson GLMM, we tested the umbrella species status of *T. cristatus* by modelling vertebrate species richness against *T. cristatus* detection and the *T. cristatus* HSI score (model 5).

2.3.5 Model selection and validation

For each GLMM, we employed an information-theoretic approach using Akaike Information Criterion (AIC) to determine the most parsimonious approximating model to make predictions (Akaike, 1973). Biotic and abiotic models considered respectively were nested thus the best models were chosen using stepwise backward deletion of terms based on Likelihood Ratio Tests (LRTs). Models 1, 4 and 5 were compared to null GLMMs. The final models were as follows:

Model 1: *T. cristatus* ~ (1|County) + amphibian richness + fish richness + waterfowl richness + terrestrial bird richness + mammal richness

Model 2: *T. cristatus* ~ (1|County) + *L. vulgaris* + *B. bufo* + *C. carpio* + *G. aculeatus* + *Gallinula chloropus* + Waterfowl

Model 3: *T. cristatus* ~ (1|County) + inflow + pond area + shading

Model 4: *T. cristatus* ~ (1|County) + HSI score

Model 5: Vertebrate species richness ~ (1|County) + *T. cristatus* + HSI score

Final models were tested for overdispersion using a custom function testing overdispersion of the Pearson residuals. Model fit was assessed using the Hosmer and Lemeshow Goodness of Fit Test within the package ResourceSelection v0.3-2 (Lele, Keim & Solymos, 2016), quantile-quantile plots, and partial residual plots (Zuur et al., 2009). Conditional R^2 was calculated using the r.squaredGLMM function in the package MuMIn v1.42.1 (Bartoń, 2018). The Benjamini-Hochberg correction was calculated for explanatory variable p-values from the cooccur and GLMM analyses using the p.adjust function in the package stats v3.4.3, and this alpha applied to the results to account for potential Type I error. The original p-values and the adjusted Benjamini-Hochberg p-values are reported for comparison in Tables 1 and 2. Model predictions were obtained using the predictSE function in the package AICcmodavg v2.1-1 (Mazerolle, 2016) and upper and lower 95% CIs were calculated from the standard error of the predictions. Results were plotted using the package ggplot2 v3.1.0 (Wickham, 2016).

313 ## 3. Results

314

315 ### 3.1 eDNA metabarcoding

316

317 Harper et al. (2018) processed a total of 532 eDNA samples and 228 PCR controls across two
318 sequencing runs. The runs generated raw sequence read counts of 36,236,862 and
319 32,900,914 respectively. After quality filtering, trimming, and merging of paired-end reads,
320 26,294,906 and 26,451,564 sequences remained. Following removal of chimeras and read
321 dereplication via clustering, the libraries contained 14,141,237 and 14,081,939 sequences
322 (average read counts of 36,826 and 36,671 per sample respectively), of which 13,126,148
323 and 13,113,143 sequences were taxonomically assigned. The final dataset used here
324 (assignments corrected, thresholds applied, and assignments removed) contained 53
325 vertebrate species (Table S5), including six amphibians, 14 fish, 17 birds, and 16 mammals
326 (Fig. 1).

327

328 ### 3.2 Pondscape biodiversity

329

330 All native amphibians were detected as well as the non-native marsh frog (*Pelophylax*
331 *ridibundus*). *T. cristatus* (*n* = 148), *L. vulgaris* (*n* = 151) and *R. temporaria* (*n* = 122) were
332 widespread, but *B. bufo* (*n* = 42), palmate newt (*Lissotriton helveticus* [*n* = 5]) and *P.*
333 *ridibundus* were uncommon (*n* = 1). The threatened European eel (*Anguilla anguilla* [*n* =
334 15]), European bullhead (*Cottus gobio* [*n* = 14]), and *C. carassius* (*n* = 2) were detected
335 alongside native fishes, such as pike (*Esox Lucius* [n = 17]) and roach (*Rutilus rutilus* [*n* = 71]),
336 but also introduced species, including *C. carpio* (*n* = 40), ruffe (*Gymnocephalus cernua* [*n* =

337 1]), and rainbow trout (*Oncorhynchus mykiss* [*n* = 3]). Some identified waterfowl were
338 ubiquitous, such as common moorhen (*Gallinula chloropus* [*n* = 211]), whereas others were
339 less common, e.g. grey heron (*Ardea cinerea* [*n* = 1]) and Eurasian oystercatcher
340 (*Haematopus ostralegus* [*n* = 1]). Terrestrial fauna were often detected in fewer than five
341 ponds (Figs. 1c, d). Buzzard (*Buteo buteo* [*n* = 4]), Eurasian jay (*Garrulus glandarius* [*n* = 7]),
342 dunnock (*Prunella modularis* [*n* = 4]), and starling (*Sturnus vulgaris* [*n* = 4]) were the most
343 frequently detected terrestrial birds. Introduced mammals (Mathews et al., 2018), such as
344 grey squirrel (*Sciurus carolinensis* [*n* = 57]) and Reeve's muntjac (*Muntiacus reevesi* [*n* = 3]),
345 outweighed native mammals. Nonetheless, we detected several mammals with Biodiversity
346 Actions Plans and/or of conservation concern (Mathews et al., 2018), including otter (*Lutra*
347 *lutra* [*n* = 1]), water vole (*Arvicola amphibius* [*n* = 16]), European polecat (*Mustela putorius*
348 [*n* = 1]), brown hare (*Lepus europaeus* [*n* = 1]) and water shrew (*Neomys fodiens* [*n* = 8]).
349 Notably, the invasive American mink (*Neovison vison*) was not detected despite widespread
350 UK distribution (Mathews et al., 2018). All species and their detection frequencies are listed
351 in Table S5.
352
353 **3.3 Hypothesis generation: biotic factors potentially influencing *T. cristatus* detection**
354
355 *T. cristatus* detection was positively influenced by amphibian and waterfowl species
356 richness, yet negatively influenced by mammal species richness. *T. cristatus* detection was
357 less probable as fish and terrestrial bird species richness increased, but these trends were
358 not significant (Fig. 2; GLMM: overdispersion $\theta = 0.995$, $\chi^2_{525} = 522.276$, $P = 0.525$; fit $\chi^2_8 =$
359 1.568, $P = 0.992$, $R^2 = 12.74\%$). With no correction for Type I error, *T. cristatus* had
360 significant positive associations with three species (Fig. S5), including *L. vulgaris*, common

361 coot (*Fulica atra*) and *G. chloropus*, and significant negative associations with six species
362 (Fig. S5), including *B. bufo*, *C. carpio*, *G. aculeatus*, *P. pungitius*, common pheasant
363 (*Phasianus colchicus*) and *S. carolinensis*. However, only *T. cristatus* associations with *L.*
364 *vulgaris* and *G. chloropus* were retained after applying the Benjamini-Hochberg correction.

365 *T. cristatus* detection was more likely in ponds with more amphibian species (Fig.
366 2a). *T. cristatus* was detected in 51.66% of ponds (*n* = 151) identified as containing *L.*
367 *vulgaris*, but in only 11.91% of ponds (*n* = 42) where *B. bufo* was detected (Fig. 1a).
368 Probability of *T. cristatus* detection was lower in ponds with more fish species, and *T.*
369 *cristatus* was not detected in ponds with more than four fish species (Fig. 2b). *T. cristatus*
370 was only detected in 15.00% (*n* = 40), 14.55% (*n* = 55) and 6.67% (*n* = 15) of ponds identified
371 as containing *C. carpio*, *G. aculeatus* and *P. pungitius* respectively (Fig. 1b). In contrast, *T.*
372 *cristatus* detection was more likely in ponds where more waterfowl species were detected
373 (Fig. 2c). *T. cristatus* was detected in 41.67% (*n* = 48) and 36.02% (*n* = 211) of ponds
374 identified as containing *F. atra* and *G. chloropus* respectively (Fig. 1c). *T. cristatus* detection
375 was negatively influenced by higher terrestrial bird species richness, but not significantly so
376 (Fig. 2d). However, *T. cristatus* was only detected in 12.00% (*n* = 25) ponds where *P.*
377 *colchicus* was detected (Fig. 1c). *T. cristatus* detection was less likely in ponds where more
378 mammal species were detected (Figs. 2e). Specifically, *T. cristatus* was only detected in
379 15.79% (*n* = 57) of ponds where *S. carolinensis* was detected (Fig. 1d).

380

381 **3.4 Hypothesis testing: biotic and abiotic factors known to influence *T. cristatus* detection**

382

383 Biotic variables put forward for model selection were eDNA-based detection/non-detection
384 of *L. vulgaris*, *B. bufo*, *C. carpio*, *G. aculeatus*, *P. pungitius*, *G. chloropus*, and *F. atra* as well

385 as presence/absence of other amphibians, fish, and waterfowl as recorded during HSI
386 assessment by environmental consultants. Only detection/non-detection of *L. vulgaris*, *B.*
387 *bufo*, *C. carpio*, *G. aculeatus*, and *G. chloropus* as well as waterfowl presence were retained
388 by model selection as explanatory variables for the biotic GLMM of *T. cristatus* detection
389 (Figs. 3a-f; GLMM: overdispersion θ = 1.013, χ^2_{495} = 495.000, P = 0.411; fit χ^2_8 = 4.437, P =
390 0.8157, R^2 = 27.77%). Abiotic variables put forward for model selection were max. depth,
391 pond density, presence of inflow, pond area, pond substrate, presence of outflow,
392 percentage of macrophyte cover, water quality, pond permanence, percentage of shading,
393 ruderals, scrub/hedge, and woodland. Only presence of inflow, pond area, and percentage
394 of shading were retained by model selection as explanatory variables for the abiotic GLMM
395 explaining *T. cristatus* detection (Figs. 3g-i; GLMM: overdispersion θ = 1.000, χ^2_{499} = 499.126,
396 P = 0.490; fit χ^2_8 = 4.898, P = 0.768, R^2 = 10.00%). Results are summarised and relationships
397 compared to those previously reported for *T. cristatus* in Table 2. Application of the
398 Benjamini-Hochberg correction for Type I error did not alter variable significance.
399
400 **3.5 Hypothesis testing: *T. cristatus* HSI and umbrella status**
401
402 HSI score positively correlated with *T. cristatus* detection probability (GLMM: overdispersion
403 θ = 0.995, χ^2_{501} = 498.372, P = 0.525; fit χ^2_8 = 5.395, P = 0.715, R^2 = 6.66%), where *T. cristatus*
404 detection was more likely in ponds with a higher HSI score (Fig. 3j; estimate ± standard error
405 = 3.094 ± 0.803, χ^2_1 = 16.020, P < 0.001). Vertebrate species richness was positively
406 associated with *T. cristatus* detection (GLMM: overdispersion θ = 1.112, χ^2_{500} = 555.800, P =
407 0.042; fit χ^2_8 = -11.91, P = 1.000, R^2 = 9.14%), with more species detected in ponds identified
408 as containing *T. cristatus* (Fig. 4a; estimate ± standard error = 0.408 ± 0.059, χ^2_1 = 46.265, P

409 < 0.001, adjusted *P* [Benjamini-Hochberg] < 0.001). However, vertebrate species richness
410 did not significantly increase with the *T. cristatus* HSI score (Fig. 4b; estimate ± standard
411 error = 0.228 ± 0.206, χ^2_1 = 1.227, *P* = 0.268, adjusted *P* [Benjamini-Hochberg] = 0.268).
412
413
414 **4. Discussion**
415
416 We have validated eDNA metabarcoding for ecological hypothesis generation and testing
417 using the community data produced by this tool in combination with environmental
418 metadata for ponds. We identified and tested biotic and abiotic factors influencing *T.*
419 *cristatus* detection, whether the HSI can be applied to *T. cristatus* eDNA survey, and
420 whether *T. cristatus* is truly an umbrella species for pond conservation. *T. cristatus*
421 detection was more likely in ponds where *L. vulgaris* and *G. chloropus* were detected, and
422 ponds where *B. bufo*, *C. carpio*, and *G. aculeatus* were not detected. *T. cristatus* detection
423 was also more likely in ponds where waterfowl presence was recorded during HSI
424 assessment. Ponds identified as containing *T. cristatus* were typically small, absent of inflow,
425 and not excessively shaded. The *T. cristatus* HSI correlated with *T. cristatus* detection, but
426 not vertebrate species richness. Nonetheless, more vertebrates were detected in ponds
427 where *T. cristatus* was detected thus presence of this amphibian may indicate good quality
428 habitat for other vertebrates. Our findings demonstrate the power of eDNA metabarcoding
429 to enhance freshwater monitoring and research by providing biodiversity data *en masse* at
430 low cost.
431

432 **4.1 Pondscape biodiversity**

433

434 eDNA metabarcoding detected six amphibian, 14 fish, 17 bird, and 16 mammal species
435 across 532 UK ponds. This diverse species inventory emphasises the importance of ponds as
436 habitat for aquatic taxa, but also as stepping stones for semi-aquatic and terrestrial taxa (De
437 Meester et al., 2005; Hill et al., 2018) through provision of drinking, foraging, dispersive, and
438 reproductive opportunities (Biggs et al., 2016; Klymus et al., 2017). Some species detections
439 may be the result of eDNA transport from water bodies in the surrounding area (Hänfling et
440 al., 2016) to ponds via inflow. However, this signifies the capacity of ponds to provide
441 natural samples of freshwater and terrestrial biodiversity in the wider catchment (Deiner et
442 al., 2017).

443

444 **4.2 Biotic factors influencing *T. cristatus* detection**

445

446 *T. cristatus* detection was more likely in ponds with higher amphibian species richness -
447 particularly ponds identified as containing *L. vulgaris* but not *B. bufo*. *T. cristatus* and *L.*
448 *vulgaris* have similar habitat requirements and tend to breed in the same ponds (Skei et al.,
449 2006; Rannap et al., 2009a; Denoël et al., 2013; Cayuela et al., 2018), with >60% overlap
450 reported (Rannap & Briggs, 2006). However, *L. vulgaris* can inhabit a broader range of
451 habitat (Rannap & Briggs, 2006; Skei et al., 2006) than *T. cristatus*, which depends on larger,
452 deeper ponds with abundant macrophytes and no fish located in open, semi-rural
453 landscapes (Denoël et al., 2013). *B. bufo* can inhabit fish-containing ponds (Manenti &
454 Pennati, 2016) and *T. cristatus* may predate *B. bufo* eggs and larvae (Langton et al., 2001).
455 This may explain the negative association between *B. bufo* and *T. cristatus* as opposed to

456 the positively associated *T. cristatus* and *L. vulgaris*.

457 *T. cristatus* detection marginally decreased with higher fish species richness, and *T.*
458 *cristatus* was negatively associated with *C. carpio*, *G. aculeatus*, and *P. pungitius*. These
459 fishes are common in and typical of ponds. All *T. cristatus* life stages may be predated by
460 fishes (Langton et al., 2001) and negative effects of fish presence-absence on *T. cristatus*
461 occupancy, distribution, and abundance are repeatedly reported (Joly et al., 2001; Rannap &
462 Briggs, 2006; Skei et al., 2006; Denoël & Ficetola, 2008; Rannap et al., 2009a, b; Hartel et al.,
463 2010; Denoël et al., 2013). *G. aculeatus* predates *T. cristatus* eggs and larvae (McLee &
464 Scaife, 1992; Jarvis, 2010), and has non-consumptive effects on *T. cristatus* embryos (Jarvis,
465 2010). *T. cristatus* larvae were also found to alter their behaviour when exposed to
466 predatory *G. aculeatus* but not non-predatory *C. carassius* (Jarvis, 2012), another fish
467 characteristic of ponds.

468 In our study, we detected *T. cristatus* in 50% of ponds where *C. carassius* was
469 detected, but <20% of ponds where large and/or predatory fishes were detected, e.g. *C.*
470 *carpio*, *G. aculeatus*, *E. lucius*. Although we observed fewer detections from ponds for *C.*
471 *carassius* than *C. carpio*, *G. aculeatus* or *E. lucius*, previous research also indicates large
472 and/or predatory fish are more detrimental to *T. cristatus* occurrence (Skei et al., 2006;
473 Hartel et al., 2010; Chan, 2011). *C. carassius* does not hinder *T. cristatus* oviposition, larval
474 behaviour, or recruitment success (Chan, 2011; Jarvis, 2012), or pond invertebrate and
475 macrophyte diversity (Stefanoudis et al., 2017). In contrast, *C. carpio* foraging reduces
476 invertebrate density and macrophyte cover (Maceda-Veiga, López & Green, 2017), which
477 lowers *T. cristatus* reproductive and foraging success and heightens predator exposure
478 (Rannap & Briggs, 2006; Gustafson et al., 2006; Chan, 2011). *C. carassius* and *C. carpio* are
479 both included among fish species assumed to negatively impact *T. cristatus* and whose

480 presence-absence is assessed for the *T. cristatus* HSI (ARG-UK, 2010). However, given that *C.*
481 *carassius* does not directly predate *T. cristatus* or indirectly alter its behaviour, reproductive
482 success, or habitat, we advocate a systematic re-evaluation of problematic fish species for *T.*
483 *cristatus* conservation.

484 *T. cristatus* detection was positively associated with waterfowl species richness,
485 namely *F. atra* and *G. chloropus* detection. These waterfowl species share macrophytes and
486 macroinvertebrates as resources with amphibians, feeding on both directly (Perrow et al.,
487 1997; Paillisson & Marion, 2001; Wallau et al., 2010). *F. atra* and *G. chloropus* crop
488 emergent macrophytes to search for invertebrate prey (Paillisson & Marion, 2001; Wallau et
489 al., 2010), which may indirectly benefit *T. cristatus* foraging. Although *Fulica* spp. can also
490 pull up submerged vegetation and damage vegetation banks (Lauridsen, Jeppesen &
491 Andersen, 1993), diet is macrophyte-dominated in late summer and autumn (Perrow et al.,
492 1997) and unlikely to impact *T. cristatus* breeding in spring (Langton et al., 2001). The
493 positive association identified here between *T. cristatus* and these waterfowl most likely
494 reflects a shared preference for macrophyte-rich ponds.

495 *T. cristatus* detection was less frequent in ponds with higher mammal species
496 richness. Our preliminary cooccur analysis indicated *T. cristatus* had negative associations
497 with *P. colchicus* and *S. carolinensis,* but not when a correction for Type I error was applied.
498 *T. cristatus* associations with *F. atra*, *B. bufo*, *C. carpio*, *G. aculeatus*, and *P. pungitius* were
499 also non-significant when this correction was applied. These associations require further
500 investigation to determine whether they are real, and if so, whether they are direct and
501 reflect competition, predation or shared resources as discussed above, or indirect and
502 reflect land-use or effects of additional species.

503

504 **4.3 Abiotic factors influencing *T. cristatus* detection**

505

506 *T. cristatus* detection was less likely in large ponds with inflow present and a greater
507 percentage of shading. Effects of pond area may depend on the size range of ponds studied.
508 Although our results indicate *T. cristatus* prefers smaller ponds, pond area does not always
509 influence occupancy (Maletzky, Kyek & Goldschmid, 2007; Denoël & Ficetola, 2008;
510 Gustafson et al., 2011) and was deemed a poor predictor of reproductive success (Vuorio et
511 al., 2013). *T. cristatus* has been found to utilise small and large ponds (Rannap & Briggs,
512 2006; Skei et al., 2006); however, very small ponds (<124 m^2) may be unable to support all
513 life stages, and larger ponds may contain fish and experience eutrophication due to
514 agricultural or polluted run-off (Rannap & Briggs, 2006). Inflow to ponds may exacerbate
515 these problems by facilitating entry of agricultural or polluted run-off and connections to
516 streams and rivers containing large, predatory fish (Freshwater Habitats Trust, 2015). Our
517 results corroborate existing research where viable *T. cristatus* populations were unlikely in
518 ponds that were shaded (Vuorio et al., 2013) or had dense macrophyte cover (Rannap &
519 Briggs, 2006; Skei et al., 2006; Hartel et al., 2010).

520 In our study, most environmental metadata available were qualitative, preventing
521 detailed analyses on pond properties and terrestrial habitat in relation to *T. cristatus*
522 detection. Better understanding of *T. cristatus* detection in relation to species interactions
523 and habitat quality could be achieved with quantitative data on pond properties (e.g. water
524 chemistry), terrestrial habitat (e.g. type, density, distance to ponds), and aquatic and
525 terrestrial habitat usage by different vertebrate species. Furthermore, given the
526 metapopulation dynamics of *T. cristatus*, future research should investigate spatial drivers
527 (e.g. pond density and other indices of connectivity, land cover, climate variables, roads,

528 rivers, elevation) of *T. cristatus* detection using innovative modelling approaches, such as
529 individual-based models (Messager & Olden, 2018). However, acquiring this data to perform
530 these models is a phenomenal task for large numbers of ponds across a vast landscape
531 (Denoël & Ficetola, 2008).
532
533 **4.4 *T. cristatus* HSI and umbrella status**
534
535 We found the HSI can predict eDNA-based *T. cristatus* detection at the UK pondscape. This
536 contradicts conventional studies which deemed the index inappropriate for predicting *T.*
537 *cristatus* occupancy or survival probabilities (Unglaub et al., 2015). We detected more
538 vertebrates in ponds identified as containing *T. cristatus*, which may support its status as an
539 umbrella species for pond biodiversity and conservation (Gustafson et al., 2006). We also
540 observed a non-significant increase in vertebrate species richness with increasing *T.*
541 *cristatus* HSI score. An adapted HSI, designed to predict species richness, could help select
542 areas for management and enhancement of aquatic and terrestrial biodiversity. Until then,
543 presence of *T. cristatus* and its HSI may confer protection to broader biodiversity by
544 identifying optimal habitat for pond creation and restoration to encourage populations of
545 this threatened amphibian. The HSI is not without issue due to qualitative data used for
546 score calculation and subjective estimation of indices (Oldham et al., 2000; O'Brien et al.,
547 2017). For future application of this index in *T. cristatus* eDNA survey, we recommend
548 metabarcoding to quantify some qualitatively assessed indices (e.g. water quality via
549 macroinvertebrate diversity, fish and waterfowl presence) alongside *T. cristatus* detection.
550 Provided rigorous spatial and temporal sampling are undertaken, eDNA metabarcoding can
551 also generate site occupancy data to estimate relative species abundance (Valentini et al.,

552 2016; Hänfling et al., 2016; Lawson Handley et al., 2019; Li et al., 2019a).

553

554 **4.5 Limitations of repurposed eDNA samples for metabarcoding applications**

555

556 This study was based on samples that were repurposed from targeted eDNA surveys for *T.*
557 *cristatus*. eDNA sampling, capture, and extraction was conducted in accordance with Biggs
558 et al. (2015), whose methods were chosen based on the eDNA literature at that time. Six 15
559 ml water samples were taken from a homogenised sample (600 ml). Ethanol precipitation
560 was used for eDNA capture, followed by DNA extraction on the combined lysate from the six
561 subsamples. Consequently, ponds were represented by a single eDNA sample, for which 12
562 qPCR replicates were performed (Biggs et al., 2015). However, eDNA metabarcoding may
563 require an entirely different workflow to enhance species detection (Harper et al., 2018;
564 Harper et al., 2019).

565 Independent biological replicates as opposed to pseudoreplicates from a single
566 water sample are key to account for spatial heterogeneity of eDNA from different species
567 (Hänfling et al., 2016; Bálint et al., 2018; Lawson Handley et al., 2019; Li et al., 2019b).
568 Larger volumes should be filtered instead of ethanol precipitation on small volumes to
569 maximise eDNA capture (Harper et al., 2019). Samples should be extracted individually to
570 maintain biological replication followed by independent technical replicates for PCR and
571 sequencing. These levels of replication are required for hierarchical occupancy modelling to
572 identify false positives and species detection probabilities as well as minimise false
573 negatives (Ficetola et al., 2015; Bálint et al., 2018; Dorazio & Erickson, 2018).

574 We performed three PCR replicates for each sample that were pooled prior to
575 sequencing. We included a large number of PCR controls and applied sequence thresholds

576 to control for any false positives in our dataset arising from potential laboratory
577 contamination. Pooling of technical replicates allowed us to screen vertebrate communities
578 from over 500 ponds, but prevented occupancy modelling to verify species detection.
579 Samples collected for targeted eDNA analysis can vary widely in terms of sampling strategy,
580 capture, extraction and storage protocols. Ultimately, these protocols may limit the data
581 generated by eDNA metabarcoding and the analyses that can be performed. This is an
582 important consideration for any future research where eDNA samples are repurposed to
583 address ecological questions.
584
585 **4.6 Prospects of eDNA metabarcoding for freshwater conservation, management, and**
586 **research**
587
588 We have demonstrated the effectiveness of eDNA metabarcoding for landscape-scale
589 biodiversity monitoring as well as ecological hypothesis generation and testing. We
590 combined metabarcoding with environmental metadata to identify new and revisit old
591 hypotheses relating to biotic and abiotic factors that influence a threatened amphibian at
592 the UK pondscape. Our findings will guide *T. cristatus* conservation in the face of increasing
593 land-use and habitat fragmentation - a poignant issue as protective legislation for this
594 species in the UK is changing. Whilst conservation of threatened species and their habitat
595 should be a priority, the bigger picture should not be ignored. eDNA metabarcoding could
596 enhance our understanding of freshwater networks, particularly pondscapes, to enable
597 more effective monitoring, protection, and management of aquatic and terrestrial
598 biodiversity. We are only now beginning to realise and explore these opportunities.
599

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Data Availability

The taxonomically assigned sequence reads used in this study were generated by Harper et al. (2018). The raw sequence reads were archived on the NCBI Sequence Read Archive (Bioproject: PRJNA417951; SRA accessions: SRR6285413 - SRR6285678). The bioinformatics analysis was deposited in a GitHub repository and permanently archived (https://doi.org/10.5281/zenodo.1188710). R scripts and corresponding data for this study have been deposited in a separate GitHub repository, which has been permanently archived (https://doi.org/10.5281/zenodo.2634427).

Author Contributions

B.H, L.R.H, L.L.H and N.B conceived and designed the study. H.C.R and N.B contributed samples for processing. L.R.H performed laboratory work and analysed the data. I.P.A and E.L offered advice on and supervised sequencing. C.H assisted with bioinformatics analysis. P.B and S.P contributed datasets for analysis. L.R.H wrote the manuscript, which all authors revised.

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896 **Table 1.** Summary of hypothesis generation using eDNA metabarcoding data, specifically
897 biotic factors that may influence *T. cristatus* detection probability. Direction of observed
898 effects on eDNA-based *T. cristatus* detection identified by the GLMM assessing species
899 richness in each vertebrate group (*N* = 532) and the preliminary cooccur analysis (*N* = 532)
900 are given. Negative and positive effects are listed as - and + respectively. Test statistic is for
901 LRT used and significant P-values (<0.05) are in bold.

Variable	**Analysis**							
	Cooccur			**GLMM**				
	Effect	***P***	**Adjusted *P* (Benjamini-Hochberg)**	**DF**	**Effect size (SE)**	**χ^2**	***P***	**Adjusted *P* (Benjamini-Hochberg)**
Amphibians								
Species richness				1	0.526 (0.140)	14.195	**<0.001**	**<0.001**
L. vulgaris	+	**<0.001**	**<0.001**	1	1.616 (0.232)			
B. bufo	-	**0.010**	0.489	1	-1.793 (0.643)			
Fish								
Species richness				1	-0.163 (0.124)	1.798	0.180	0.225
G. aculeatus	-	**0.012**	0.489					
C. carpio	-	**0.040**	0.714					
P. pungitius	-	**0.049**	0.714					
C. carassius								
Waterfowl								
Species richness				1	0.491 (0.134)	13.530	**<0.001**	**<0.001**
G. chloropus	+	**0.001**	0.012					
F. atra	+	**0.022**	0.237					
Terrestrial birds								
Species richness				1	-0.044 (0.302)	0.021	0.884	0.884

Variable	Analysis							
	Cooccur			GLMM				
	Effect	*P*	Adjusted *P* (Benjamini-Hochberg)	DF	Effect size (SE)	χ^2	*P*	Adjusted *P* (Benjamini-Hochberg)
P. colchicus	-	**0.050**	0.714					
Terrestrial mammals								
Species richness				1	-0.515 (0.188)	8.501	**0.004**	**0.006**
S. carolinensis	-	**0.020**	0.685					

902 **Table 2.** Summary of hypothesis testing using eDNA metabarcoding data and associated
903 environmental metadata, specifically biotic and abiotic factors known to influence *T.*
904 *cristatus* detection probability. Reported effects on *T. cristatus* detection in the literature
905 and hypothesised effects on eDNA-based *T. cristatus* detection are given for each variable.
906 Any variables not explicitly reported in the literature that were identified by the preliminary
907 cooccur analysis are listed as UNK. Direction of observed effects on eDNA-based *T. cristatus*
908 detection identified by each analysis (biotic GLMM, *n* = 504; abiotic GLMM, *n* = 504) are
909 given. Negative and positive effects are listed as - and + respectively. For categorical
910 variables with more than one level, effect size and standard error (SE) are only given for
911 levels reported in the model summary. Test statistic is for LRT used and significant P-values
912 (<0.05) are in bold. Variables included for model selection but not retained in the final
913 models are listed as NR.
914

Variable	Effect reported	Hypothesised effect	GLMM				
			DF	Effect size (SE)	χ^2	*P*	Adjusted *P* (Benjamini-Hochberg)
Amphibians							
L. vulgaris	+	+	1	1.616 (0.232)	50.024	**<0.001**	**<0.001**
B. bufo	UNK		1	-1.793 (0.643)	11.175	**<0.001**	**0.002**
Fish							
G. aculeatus	-	-	1	-1.048 (0.453)	6.300	**0.012**	**0.014**
C. carpio	-	-	1	-1.457 (0.586)	7.854	**0.005**	**0.008**
P. pungitius	-	-	NR				
C. carassius	-	-					
Waterfowl							
G. chloropus	UNK		1	0.893 (0.228)	15.703	**<0.001**	**<0.001**
F. atra	UNK		NR				
Waterfowl presence	-	-	2		6.350	**0.042**	**0.042**
Minor				0.531 (0.234)			
Major				0.808 (0.495)			
Pond area	-/+	-	1	-0.0003 (0.0002)	4.134	**0.042**	**0.042**

Variable	Effect reported	Hypothesised effect	GLMM				
			DF	Effect size (SE)	χ^2	*P*	Adjusted *P* (Benjamini-Hochberg)
Pond density	+	+			NR		
Pond depth	+	+			NR		
Pond substrate	+	+			NR		
Pond permanence	+	+			NR		
Water quality	+	+			NR		
Inflow Present	-	-	1	-0.820 (0.251)	11.437	**<0.001**	**0.001**
Outflow	UNK				NR		
Macrophyte cover	-/+	-			NR		
Shading	-/+	-	1	-0.010 (0.003)	12.266	**<0.001**	**0.001**
Woodland	+	+			NR		
Scrub/hedge	+	+			NR		
Ruderals	UNK				NR		

915

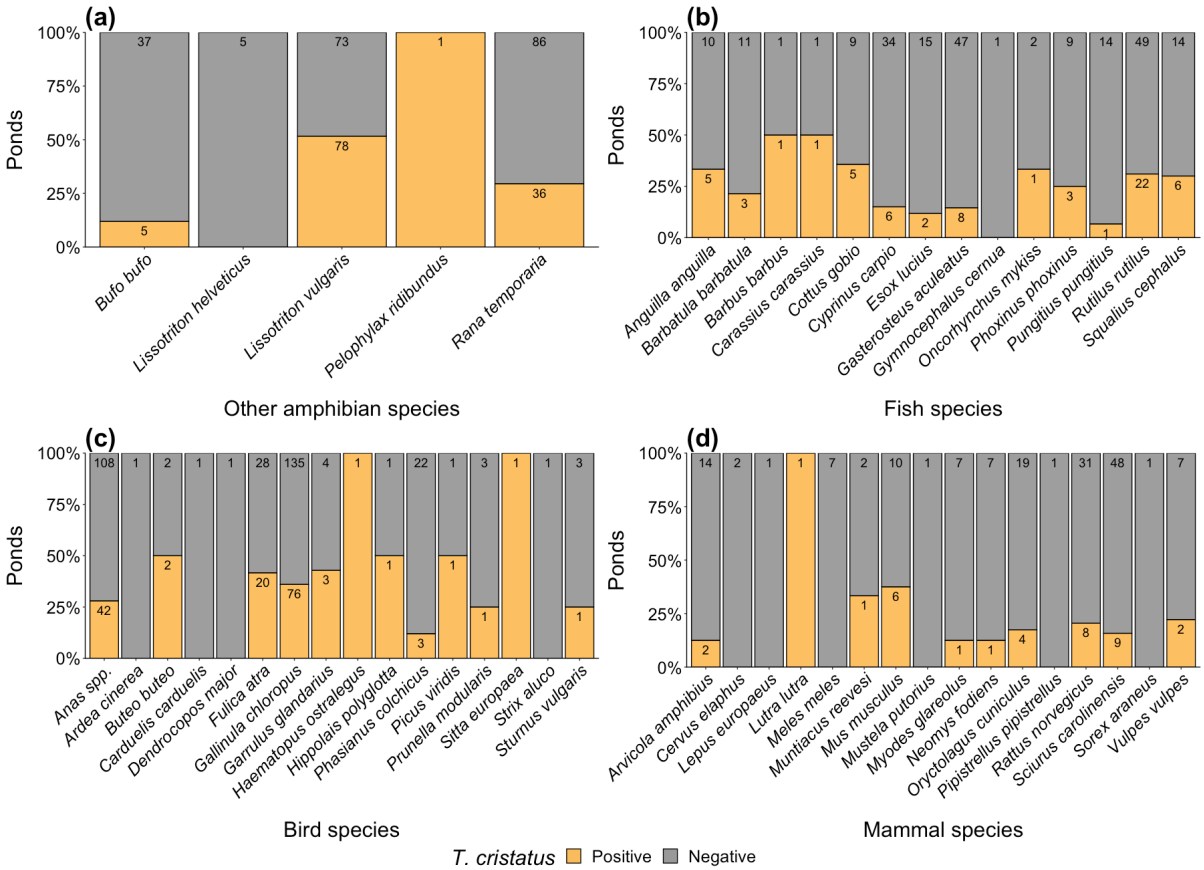

Figure 1. eDNA metabarcoding detection of *T. cristatus* in relation to other vertebrate species (*N* = 532 ponds): **(a)** other amphibians, **(b)** fish, **(c)** birds, and **(d)** mammals. Numbers on each bar are the number of ponds with (orange) and without (grey) *T. cristatus* in which a vertebrate species was detected.

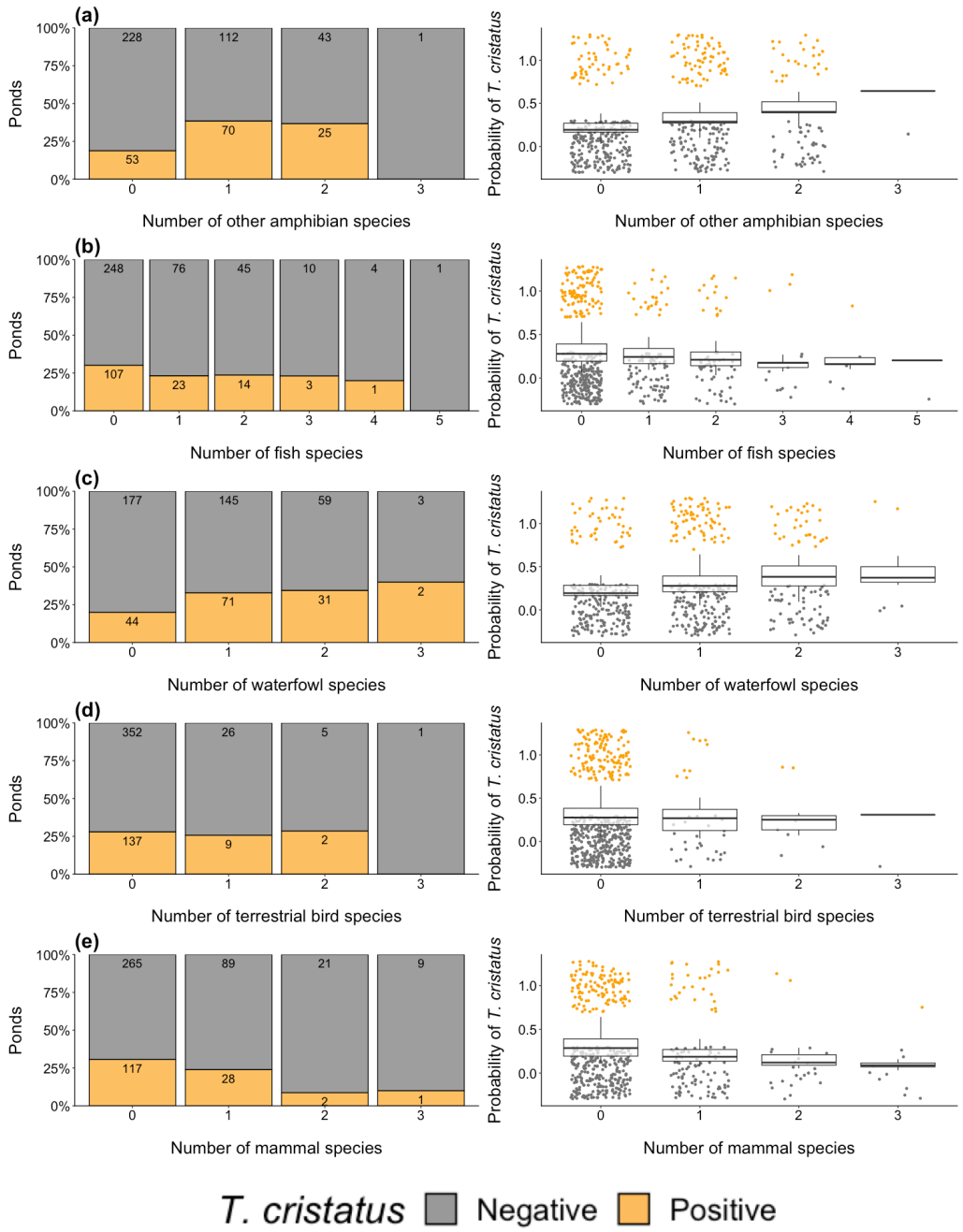

Figure 2. *T. cristatus* detection (orange) and non-detection (grey) in relation to species richness of different vertebrate groups (*N* = 532 ponds): **(a)** other amphibians, **(b)** fish, **(c)** waterfowl, **(d)** terrestrial birds, and **(e)** mammals. Observed proportion of ponds with and without *T. cristatus* (left) is plotted alongside predicted probability of *T. cristatus* detection (right). Numbers on barplots of observed detection are the number of ponds for each category. In plots showing predicted *T. cristatus* detection, the observed data is shown as points (jittered around 0 and 1 to clarify variation in point density) and boxes are the model predictions.

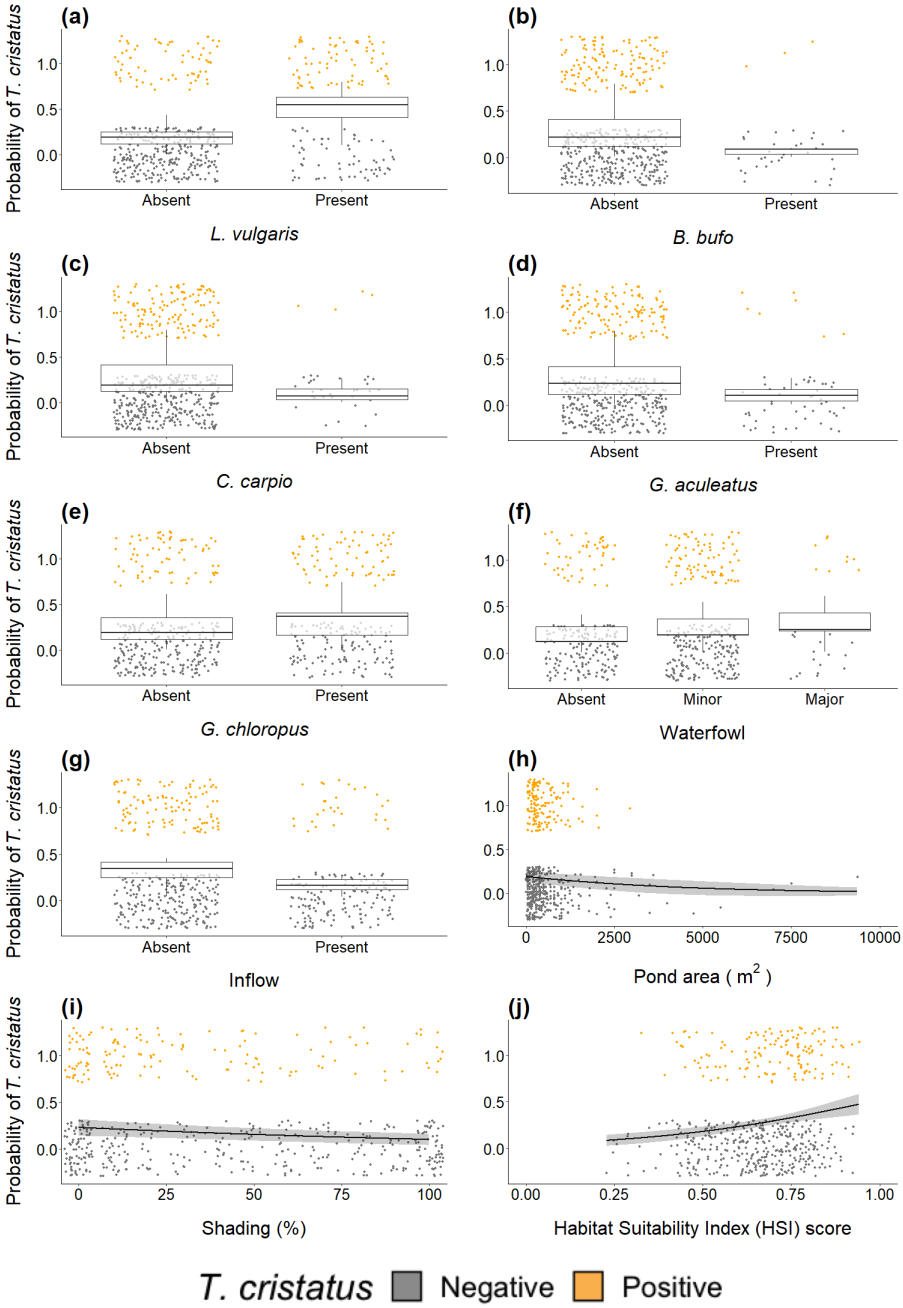

Figure 3. Biotic and abiotic factors influencing *T. cristatus* detection (*n* = 504 ponds): **(a)** *L. vulgaris* detection, **(b)** *B. bufo* detection, **(c)** *C. carpio* detection, **(d)** *G. aculeatus* detection, **(e)** *G. chloropus* detection, **(f)** extent of waterfowl presence, **(g)** presence of inflow, **(h)** pond area, **(i)** percentage of shading, and **(j)** HSI score. The 95% CIs, as calculated using the predicted *T. cristatus* probability values and standard error for these predictions, are given for each relationship. The observed *T. cristatus* detection (orange) and non-detection (grey) data are displayed as points (jittered around 0 and 1 to clarify variation in point density) against the predicted relationships (boxes/lines).

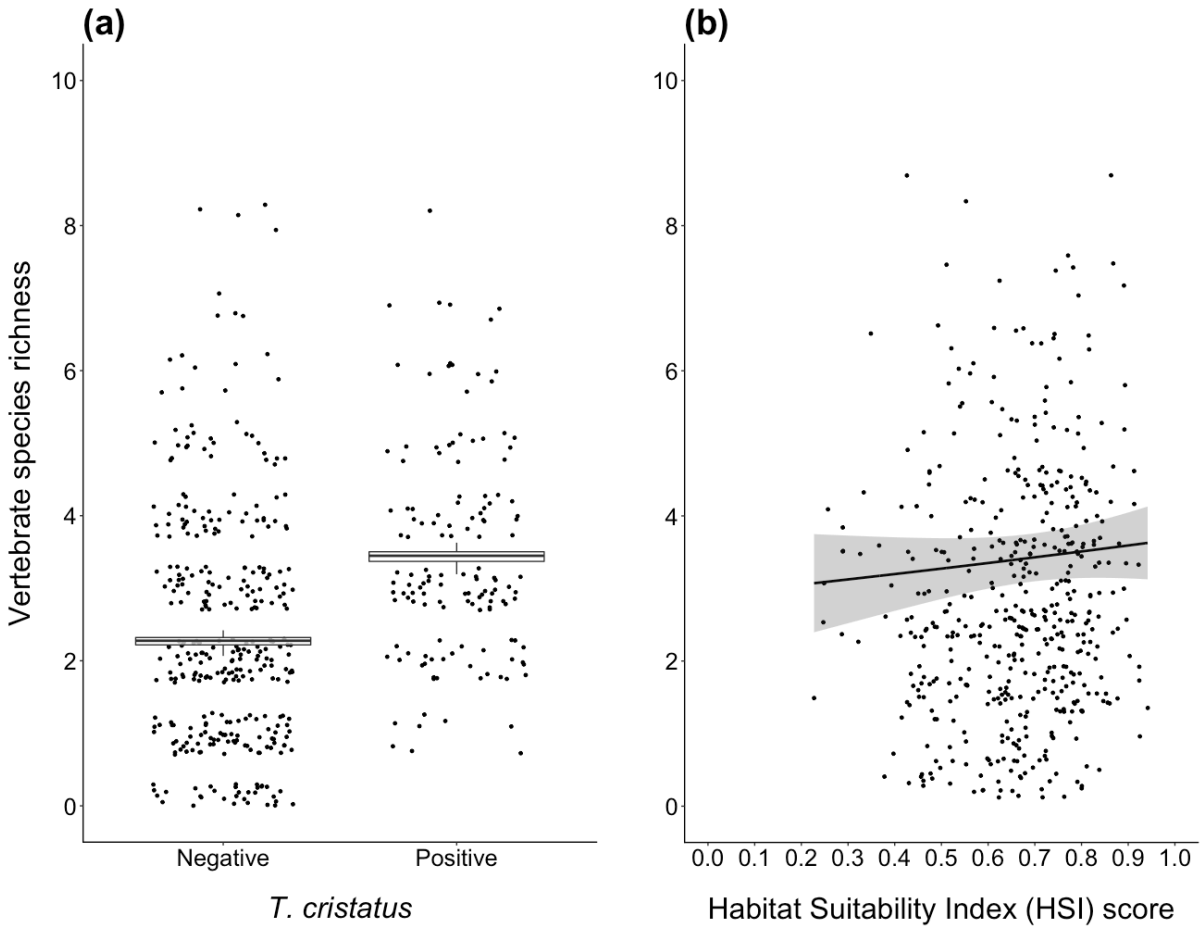

Figure 4. Vertebrate species richness in ponds (*n* = 504) in relation to: **(a)** *T. cristatus* detection and **(b)** the *T. cristatus* HSI score. The 95% CIs, as calculated using the predicted species richness values and standard error for these predictions, are given for each relationship. The observed data are displayed as points (jittered to clarify variation in point density) against the predicted relationships (boxes/lines).