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Title: Drivers of macrofungal species composition in temperate forests, West Hungary: functional groups compared

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Footnote to the title: Environmental drivers of macrofungal species composition

Abstract

The most influential environmental drivers of macrofungal species composition were studied in managed, even-aged, mixed forests of Őrség National Park, Hungary. Functional groups of macrofungi were analyzed separately by non-metric multidimensional scaling and redundancy analysis exploring their relations to tree species composition, stand structure, soil/litter conditions, microclimate, landscape, and management history. There was some evidence that macrofungi are related to drivers that are relatively easy to measure. Wood-inhabiting fungal species composition is driven primarily by the species composition of living trees, while substratum properties and microclimate play minor roles. The terricolous saprotrophic community was determined principally by a litter pH gradient involving tree species composition and soil/litter properties. Microclimate had no concordant effect. No obvious underlying gradients were detected on ectomycorrhizal fungal species composition; however, tree size and litter pH had significant effects. For each group, no clear responses to landscape or management history were detected.

Key words: Biodiversity, Ectomycorrhizal fungi, Environmental variation, Fungal community gradients, Host specificity, Soil properties, Sporocarp sampling, Terricolous saprotrophic fungi, Wood-inhabiting fungi

Introduction

Forest-dwelling macrofungal assemblages have been classified into three main functional groups: wood-inhabiting (including wood saprotrophs and necrotrophic parasites), ectomycorrhizal (EcM) and terricolous saprotrophic communities (Winterhoff 1992). In a global perspective, an enormous volume of research has been reported on the responses of macrofungal community composition to environmental variation. Wood-inhabiting fungal communities are driven principally by the amount and diameter (Heilmann-Clausen & Christensen 2004; Sippola et al. 2005; Ódor et al. 2006; Lonsdale et al. 2008), decay stage (Heilmann-Clausen & Christensen 2003b; Siller 2004; Heilmann-Clausen et al. 2014), age (Heilmann-Clausen 2001), species identity (Sippola et al. 2005; K€uffer et al. 2008), complexity (Heilmann-Clausen & Christensen 2003a), and spatio-temporal availability (Siitonen 2001; Bäassler et al. 2010; Halme et al. 2013) of dead wood. The microclimatic variation and pH within the wood (Boddy 1992, 2001; Salerni et al. 2002) or the interactions with other organisms (van der Wal et al. 2013) also have significant effects. EcM community composition is structured strongly by the N content (Toljander et al. 2006; Cox et al. 2010; Suz et al. 2014), pH (Baar & ter Braak 1996; Talbot et al. 2013) as well as temperature and moisture of soil (Claridge et al. 2000; Jones et al. 2003), species composition of host trees (Kernaghan et al. 2003; Smith & Read 2008; Morris et al. 2009), season (over the course of even a month) (Courty et al. 2008), fungal dispersal limitation among host trees (Peay et al. 2010), and timing of colonization and interspecific competition on the root surface (Kennedy et al. 2009; Kennedy 2010). In the same context, little is known about the determinants of terricolous saprotrophic communities, but the effects of litter quantity and pH (Tyler 1991; Ferris et al. 2000; Talbot et al. 2013), P content of the soil (Reverchon et al. 2010), tree species composition (O'Hanlon & Harrington 2012), and temperature (McMullan-Fisher et al. 2009) are documented to be highly important.

Many influential environmental drivers have been revealed, but are there drivers with consistent importance to macrofungal functional groups? When such drivers are sought, many difficulties are encountered. The relative importance of drivers varies across spatial scales (Claridge et al. 2000; Lilleskov & Parrent 2007; Büntgen et al. 2012) and along environmental gradients, such as elevation (Gómez-Hernández et al. 2012; Sundqvist et al. 2013) and rainfall (Lindblad 2001; Salerni et al. 2002). Also, the relative effects of drivers can be biased strongly by the edaphic heterogeneity of the studied habitats, and the factors (resources or environmental conditions) that are actually limiting in a habitat can have a disproportionately high influence on species composition (McMullan-Fisher 2008). In addition, community level responses are difficult to reveal, since great species diversity is found within fungal communities in which each species has slightly different environmental requirements (Boddy et al. 2008).

Based on the studies mentioned in the first paragraph, our knowledge of fungal community responses to environmental variation is biased by research history: (1) the majority of studies have been conducted in Northern or Western Europe or in North America, thus, large regions are still underrepresented; (2) the studies have rarely been focused on more than two functional groups (except e.g. Humphrey et al. 2000; Sato et al. 2012); (3) to obtain a clearer picture, many authors have used a limited pool of environmental factors and hence, several environmental impacts with probable significant effects remained unexplored on the sampling sites.

Given these complexities and research gaps, the present study has been designed in even-aged, managed forests with a restricted number of habitat types to try to reduce the effects of edaphic heterogeneity. By including several variables suggested by the literature, other factors that characterize the landscape and management history were also examined.

In accordance with the studies referenced in the first paragraph, it can be hypothesized

that: (1) substratum properties, tree species composition, and microclimate have the strongest effects on macrofungal species composition at a stand scale, and (2) the relative influence of these factors differs among wood-inhabiting, EcM, and terricolous saprotrophic communities. The aims of this study are to find the most important environmental factors that best explain the macrofungal species composition of wood-inhabiting, EcM and terricolous saprotrophic communities, and provide information on the environmental requirements of fungal species.

Materials and methods

Study area

This study has been carried out in Őrség National Park (ŐNP), West Hungary (46° 51'–55' North, 16° 06'–24' East (**Fig 1A**). In the ŐNP, the precipitation ranges between 700 and 800 mm yearly. Between 1901 and 2000, the mean minimum and maximum temperatures in winter were respectively –7.4 and 6.0 °C, while in summer 13.5 and 23.8 °C (measured in a nearby town, Szombathely, Hungarian Meteorological Service, OMSZ). The landscape is divided into hills and wide valleys at the elevation range of 250–350 m above sea level. The bedrock consists of alluvial gravel and clay. Nutrient-poor brown forest soils with pseudogley or lessivage (planosols or luvisols) are the most frequent soil types (Halász 2006; Dövényi 2010). The pH of the soil is acidic; it tends to range from 4.0 to 4.8 with a mean of 4.3 (Juhász et al. 2011).

Presently, forests cover 80% of the ŐNP region, which has an area of ca. 350 km^2 (Dövényi 2010). Stands are dominated by beech (*Fagus sylvatica* L.), sessile and pedunculate oak [*Quercus petraea* (Matuschka) Liebl. and *Q. robur* L.], hornbeam (*Carpinus betulus* L.), and Scots pine (*Pinus sylvestris* L.). Forests are sometimes monodominant, but more often form mixed stands with great compositional diversity. The most frequent non-dominant tree species are *Betula pendula* Roth, *Picea abies* (L.) Karst., *Populus tremula* L., *Castanea sativa*

Mill., *Prunus avium* L., *Tilia* spp., and *Acer* spp. (Tímár et al. 2002). ŐNP is characterized by the highest proportion of private forest stands in Hungary where the dominant tree species usually varies from stand to stand. Therefore, the ŐNP is a suitable region for studying the effects of tree species on macrofungal communities.

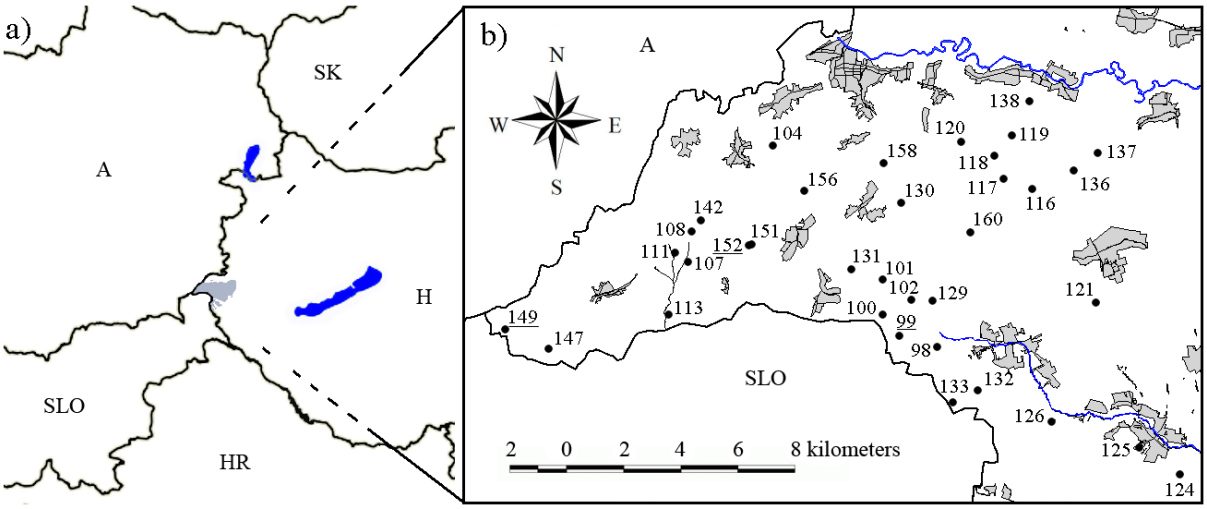


Fig 1 – Borders of West Hungary; Őrség National Park (440 km^2) is highlighted in grey (**a**). The geographical positions of the 35 sampling units are indicated by black dots (the underlined sampling units are moderately managed); built-up areas are shown by grey (**b**). A: Austria, H: Hungary, HR: Croatia, SK: Slovakia, SLO: Slovenia.

Between the 12th and 19th centuries, the landscape was characterized by a rotation cycle in land use: small areas of forests, meadows and arable lands were replaced by each other. Meanwhile all the pristine forests were cut. Leaf-litter was collected widely in the secondary stands and used as bedding for farm animals. A specific ridge planting system was applied on the arable lands to decrease the high levels of groundwater in the upper soil layers; plants were set onto the top of the ridges. The nutrient-poor arable lands had to be fallowed often for many years whilst they were frequently regrown by pine and spruce; slash and burn was used to return the regenerated forests to arable land uses. As a consequence of these activities, the region was characterized by much soil erosion, leaching and acidification. Due to that, the proportion of pioneer trees (*P. sylvestris* and *B. pendula*), acidofrequent herbs, bryophytes and lichens increased. Now, these traditional cultivation practices have ceased.

Currently, a spontaneous stem selection method in the private forests and a shelterwood management with a rotation period of 70–110 yr in the state forests are applied. As a result of this, an increasing proportion of deciduous trees and mesophytic herbs can be observed in the region (Gyöngyössy 2008).

Environmental data collection

Similar habitats without strong effects of edaphic heterogeneity (that would make the environmental data noisy) are required for finding the environmental factors that drive the species composition of forest macrofungi. Accordingly, forest stands were selected by a stratified random sampling based on the Hungarian Forestry Database (Hungarian Central Agricultural Office, Forestry Directorate, www.nebih.gov). The even-aged, 70–100 yr old, spatially independent stands (the minimum distance is 500 m between them) chosen were located in relatively flat areas and not influenced directly by surface waters. These stands were grouped based on the most frequent tree species. Thirty-five stands were selected randomly from these groups representing a gradient along the characteristic tree species combinations of the region. A 40 m × 40 m plot was assigned in each selected stand. Geographical positions of plots are shown in **Fig 1B**; GPS coordinates are available in Siller et al. (2013). The plots were scattered in a 160 km^2 area. In the middle of each plot, a 30 m × 30 m sampling unit was assigned for macrofungal surveys. Sampling units were divided into thirty-six 5m × 5m quadrats arranged systematically.

Environmental data that are easy to measure on the sites were used as potential explanatory variables to explain the species composition of macrofungal communities. Fifty-two variables representing tree species composition, stand structure, soil and litter conditions, microclimate, landscape structure, and management history were measured (**Table 1**).

Table 1 – The potential environmental variables influencing the species composition of macrofungal communities

Environmental variable	Unit	Mean (range)	Transformation
TREE SPECIES COMPOSITION			
Species richness of trees	number of species/1600 m^2	5.63 (2–10)	ln
Shannon diversity of tree species	–	0.847 (0.097–1.802)	ln
Relative volume of beech	%	27.9 (0.0–94.4)	ln
Relative volume of hornbeam	%	3.9 (0.0–21.8)	ln
Relative volume of oaks	%	36.4 (1.1–98.0)	ln
Relative volume of Scots pine	%	26.2 (0.0–76.9)	ln
Relative volume of non-dominant trees	%	0.02 (0.00–0.17)	ln
STAND STRUCTURE			
Density of trees (Diameter at Breast Height, DBH > 5 cm)	stems/ha	593.39 (217.75–1392.75)	–
Density of large (DBH > 50 cm) trees	stems/ha	17.14 (0.00–56.25)	ln
Density of shrubs and saplings (DBH = 0–5 cm)	stems/ha	952.14 (0.00–4706.25)	ln
Basal area of trees	m^2/ha	32.87 (21.49–42.26)	–
Mean DBH of trees	cm	26.65 (13.70–40.75)	–
Coefficient of variation of DBH of trees (DBH > 5 cm)	–	0.480 (0.172–0.983)	–
Volume of snags (d > 10 cm)	m^3/ha	8.99 (0.90–65.02)	ln
Volume of logs (d > 10 cm)	m^3/ha	10.51 (0.17–59.48)	ln
Total volume of logs and snags (d > 10 cm)	m^3/ha	19.50 (1.93–73.37)	ln
Relative volume of logs (d > 10 cm) in decay stages 3–6	%	54.86 (8.25–98.61)	–
Total cover of FWD and CWD	m^2/ha	261.57 (79.44–729.99)	ln
Cover of understory vegetation	m^2/ha	740.80 (19.19–4829.30)	ln
Cover of bryophytes	m^2/ha	247.37 (16.57–2201.59)	ln
SOIL AND LITTER			
Cover of soil	m^2/ha	146.75 (8.56–472.22)	–
Cover of litter	m^2/ha	9367 (7815–9834)	–
pH of litter	–	5.29 (4.86–5.68)	–
pH of soil *	–	4.33 (3.96–4.84)	–
Dry litter mass	g/900 cm^2	147.66 (105.41–243.08)	–
Mass proportion of deciduous litter	%	14.71 (2.54–32.80)	–
Mass proportion of decayed litter	%	67.71 (51.58–84.16)	–
Hydrolytic acidity of soil (y1) *	–	30.21 (20.68–45.22)	–
Exchangeable acidity of soil (y2) *	–	15.27 (3.94–30.47)	–
Fine texture (clay and silt) proportion of soil *	%	51.95 (27.60–68.60)	–
Carbon (C) content of litter	%	65.69 (42.87–78.09)	–
Carbon content of soil *	%	6.45 (3.30–11.54)	–
Nitrogen (N) content of litter	%	1.28 (0.83–1.84)	–
Nitrogen content of soil *	%	0.22 (0.11–0.34)	–
Phosphorus (P) content of soil *	mg P_2O_5/100 g	4.29 (1.96–9.35)	–
Potassium (K) content of soil *	mg K_2O/100 g	7.74 (4.00–13.10)	–
MICROCLIMATE			
Mean daily air temperature difference	°C	–0.10 (–0.93–0.73)	–
Daily air temperature range difference	°C	0.94 (–0.42–2.49)	–
Mean daily air humidity difference	%	0.84 (–1.83–3.32)	–
Daily air humidity range difference	%	1.89 (–2.27–6.58)	–
Mean relative diffuse light	%	2.93 (0.62–10.36)	ln
Coefficient of variation of relative diffuse light	%	0.51 (0.12–1.23)	ln
LANDSCAPE (radius = 300 m)			
Proportion of cutting areas	%	5.73 (0.00–23.03)	ln
Proportion of forests	%	89.80 (56.92–100.00)	–
Proportion of open patches (settlements, meadows, arable lands)	%	4.72 (0.00–45.25)	–
Shannon diversity of landscape elements	–	1.114 (0.108–1.858)	–
MANAGEMENT HISTORY			
Historical proportion of forests **	%	76.58 (24.03–100.00)	–
Historical proportion of meadows **	%	7.26 (0.00–40.73)	–
Historical proportion of arable lands **	%	16.16 (0.00–61.27)	–
Locality of forests in 1853	binary	0.800 (0–1)	–
Locality of arable lands in 1853	binary	0.171 (0–1)	–

* soil layer: 0–10 cm, ** radius = 300 m

Tree species composition was expressed based on the relative volume of tree species by merging all taxa within the same genus, e.g. oaks (*Q. cerris*, *Q. petraea*, *Q. robur*) and limes (*Tilia cordata*, *T. platyphyllos*). Volume of tree individuals was computed by species specific equations using the height and diameter of trees at breast height (DBH) (Sopp & Kolozs 2000). Shannon diversity of tree species was calculated based on relative tree volumes and using natural logarithm (Shannon & Weaver 1949).

Regarding stand structure, each tree within the 40 m × 40 m plots and larger than 5 cm DBH was mapped; tree species identity, DBH and height were recorded. Coarse woody debris (CWD) longer than 50 cm and thicker than 10 cm, and snags (including stumps) thicker than 5 cm were measured and mapped; volumes were computed by assuming that they were cylinders. Decay stage of CWD was determined according to Ódor & van Hees (2004). Projected onto the soil surface, the relative area covered by woody debris [fine (FWD) and coarse units together], litter, bare soil, bryophytes, and understory vegetation (including herbs and seedlings shorter than 50 cm) were estimated visually in the 5 m × 5 m quadrats; and their results were transformed into m^2 ha^{-1}. Shrub density was measured by counting each arboreal individual (including regenerating trees) thinner than 5 cm DBH and taller than 50 cm.

Soil and litter conditions were measured within the sampling units by sampling five points arranged systematically. Litter was collected from 30 cm × 30 cm areas. Soil cubes of 15 cm × 15 cm were sampled from the vertical layer of 0–10 cm. Soil and litter pH were measured potentiometrically by a pH meter in the supernatant suspension of the sample. Determination of hydrolytic (y1) and exchangeable (y2) acidity were carried out by titration with NaOH; soil samples were extracted by 1 mol dm^{-3} $Ca(CH_3COO)_2$ and 1 mol dm^{-3} KCl solutions, respectively (Bellér 1997). The organic C and total N content of soil and litter were measured according to ISO (1995, 1998) applying dry combustion elementary analysis by Elementar vario EL III CNS equipment. The P and K contents of the soil were extracted by an

ammonium lactate/acetic acid solution based on Bellér (1997).

Air humidity and temperature measurements were conducted in the center of each sampling unit at 1.3 m height using Voltcraft DL-120 TH data loggers. For both measurements, dissimilarity values were calculated between the measured values of two nearby reference sites and the measured values of the studied sampling units. Measurements were synchronized in time and lasted for 24 h by setting 5 min recording frequency. By repeating the same procedure, eight measurements were carried out in different months of the vegetation periods between 2009 and 2011, and the results were averaged. Relative diffuse light was measured by LAI-2000 Plant Canopy Analyzer in the center of each sampling unit at 1.3 m height and always at dusk (Tinya et al. 2009).

The proportion of landcover types (forests, permanently open patches and cutting areas) was calculated inside a circle of 300 m radius surrounding each plot. Measurements were carried out using aerial photographs and topographic maps. Stands older than 20 yr were considered to be forests; younger ones were defined as cutting areas. Landscape diversity was expressed by the Shannon diversity index based on the relative cover of landscape elements (Shannon & Weaver 1949).

Management history was demonstrated based on the map made by the Habsburg Empire in 1853 during the Second Military Survey (Arcanum 2006). According to this map, the same landscape variables were computed that were used for characterizing the recent landscape. Historical land use types of sampling units were fixed as binary variables.

Fungal data

Because of the large total area (31 500 m^2) of sampling units, sporocarp surveys were conducted to characterize the macrofungal species composition. Macrofungal surveys sampled basidiomycetes (excluding most of the resupinate non-poroid taxa) and ascomycetes

that develop sporocarps visible to the naked eye (larger than 2 mm). Sporocarps were sampled three times in each sampling unit: in Aug. 2009, May 2010 and during Sep.–Nov. 2010. The precipitation in 2010 was far above average, resulting in high sporocarp production in the region. Thus, the duration of the third survey was relatively long: 48 d between 19 Sep. and 5 Nov. (early Nov. is generally the end of the main fruiting period in Hungary). Dried specimens were deposited in the Hungarian Natural History Museum, Department of Botany (BP), Budapest.

To obtain presence-absence data for macrofungi, the species identity of taxa was recorded in each quadrat of each sampling unit in each sampling period. Accordingly, the total number of times a species was found in a quadrat in a sampling unit was a calculated abundance measure (a local frequency value) for each collected fungus. The maximum value of the local frequency of a species is $36 \times 3 = 108$, based on the 36 quadrats in a sampling unit and the three sampling periods. Therefore, the community data form a multivariate matrix of fungal species and sampling units where species performance was expressed by local frequency values. The total number of sampling units occupied by a fungus was also calculated (**Supplementary Table 1**).

Species identification procedures are detailed in Siller et al. (2013). The identity and nomenclature of sampled taxa were determined by using monographs, books and papers. MycoBank (www.mycobank.org, accessed between 19 and 20 of Apr. 2013) and more rarely Knudsen & Vesterholt (2012) were used to verify up-to-date scientific names and authorities of fungal species.

The macrofungal taxa were classified into three main functional groups: terricolous saprotrophic fungi living on litter or any kind of buried plant debris in the uppermost 10 cm of the soil; wood-inhabiting fungi colonizing dead branches, twigs, logs or snags on the ground, and trunks or roots of living wood; and EcM fungi representing a well definable, standalone

group (Tedersoo et al. 2010).

Data analyses

Environmental variables that drove the species composition of wood-inhabiting, terricolous saprotrophic and EcM fungi were examined separately for each functional group by two different ordination methods: redundancy analysis (RDA) and non-metric multidimensional scaling (NMDS). Results of both methods were evaluated by looking for consistency in their environmental interpretations, but more focus was put on the NMDS results because these models had better explanatory powers.

RDA plots points of species and sampling units in a space defined by the environmental variables, and was used here to represent the best fit of species abundances to the environmental data. This method is a constrained ordination based on a model of linear species response to the underlying environmental gradient where the rare species (listed in **Supplementary Table 1** and found on less than four sampling units) were often dropped from the analysis (Legendre & Legendre 1998). RDA was chosen as a suitable direct gradient analysis after the detection of short gradient lengths (2–3 SD units) revealed by the detrended correspondence analyses of functional groups (Lepš & Šmilauer 2003). RDA models were built by the manual forward selection of explanatory variables and testing the effects of variables on community data by F-statistics applying Monte Carlo simulations with 999 permutations; significance of all canonical axes were tested similarly (ter Braak & Šmilauer 2002). Log-transformed local frequency values of taxa were used for RDAs.

By contrast, NMDS is an unconstrained ordination that avoids the assumption of linear relationships among variables and provides a valuable representation of the overall community structures without constricting the analysis to the frequent species only. In this regard, NMDS is a powerful tool, but it is not designed principally for finding the most

important environmental drivers of species composition. That is, the environmental interpretation of the NMDS results can be achieved by fitting vectors subsequently onto the NMDS solutions, which are reached independently from the environmental data (Oksanen 2013). In the present study, NMDS was carried out following McCune & Grace (2002) and Oksanen (2013). Regarding each functional group, a "local" NMDS model (Sibson 1972) was fitted where an independent monotonic regression was used for each sampling unit in contrast to the "global" NMDS model (Kruskal 1964), which was fitted from a global point of view on ranked dissimilarities. According to Prentice (1977), local NMDS can be more suitable for evaluating ecological gradients than the global NMDS model because it is sensitive to the local environment of each point in the ordination space supposing that the environment itself can change along a gradient. NMDS was run on Sørensen (Bray–Curtis) distances and it obtained a much stronger description of community structures compared to the other tested distance methods: "Jaccard", "Canberra" and "Euclidean". Random starting configurations (20 for each functional group) were used for finding the best stable solutions. The dimensionality of each studied community dataset was revealed based on the **Supplementary Figs 1–3E**. Kendall's rank correlation coefficients (τ) were calculated between the original distance matrices and the ordination distances, and they were plotted against the final stress values testing the dimensions between one and ten. Three dimensional solutions were chosen to be plotted in this study. NMDS stress was measured by Kruskal's stress formula 1 multiplied by 100 (Kruskal 1964). For representing goodness of fit, Shepard diagrams and the best-fit monotonic regressions of distances were plotted in **Supplementary Figs 1–3C** and **D**. The environmental variables fitted significantly ($p < 0.05$) onto the NMDS solutions were screened for strong ($|r| > 0.5$) collinearities and intercorrelated ones with a weaker relationship to the response variables were removed.

Before the analyses, a preparative procedure was completed for the environmental

variables: (1) their normality was checked and, if needed, ln-transformation was applied (**Table 1**), and (2) they were centred and standardized by standard deviation. It was supposed that our community data were biased by the third, 48 d sporocarp survey during which the vast majority of records were obtained and the field visit to some sampling units was extended to the end of (or beyond) the fruiting period of some species. Therefore, the days of this sampling period were numbered from 1 to 48 and a "sampling time" variable was created. Sampling time correlated often strongly with any of the ordination axes regarding each functional group (ranges of |r| and p-values: 0.624–0.800; 0.003–0.001). Geographical longitude and/or latitude coordinates of sampling units also had strong correlations with the response variables (|r| = 0.534–0.642; p = 0.013–0.002). Moreover, unexpectedly, these three variables (sampling time, latitude and longitude coordinates) and some of the studied environmental variables were also related (|r| = 0.402–0.493; p = 0.014–0.003). Thus, the amount of variation that can be attributed exclusively to the effects of sampling time and geographical coordinates was measured by applying partial regression analysis according to Legendre & Legendre (1998); the residuals of the partial regression models were used for further analyses. These corrected environmental variables were fitted onto the NMDS solutions, while the RDA models (with the ability to use corrected variables) were built by using the original environmental variables and entering the geographical coordinates and sampling time as covariates on each occasion.

R for Windows 3.0.1 (R Core Team 2013) and, if required, the R package "vegan" v.2.0-8 (Oksanen et al. 2013) was used for carrying out preliminary tests of environmental variables, correlations, partial regressions, and NMDS. The R package "Rcmdr" v.2.1-4 (Fox 2005) was applied for displaying spinning 3-D NMDS solutions. Canoco for Windows 4.5 (ter Braak & Šmilauer 2002) was applied for RDAs.

Results

Fungal diversity

687 macrofungal taxa were collected and identified (**Supplementary Table 1**). Taxa belonging to the phylum Basidiomycota (631 species, 167 genera) were more species rich than ascomycetous taxa (56 species, 29 genera). A total of 13396 records and 1556 specimens were obtained. The vast majority of records (11647 pieces, 87 %) were collected during the third field survey in autumn 2010, whereas the total number of records was 1313 (10 %) in Aug. 2009 and 436 (3 %) in May 2010. Macrofungal taxa were classified into eight functional groups (**Supplementary Table 1**). The three most species rich functional groups were studied, in which a few abundant and a large number of rare species were found (**Table 2**).

Table 2 – Species richness and proportions of functional groups.

	Wood-inhabiting fungi	Terricolous saprotrophic fungi	EcM fungi	Other fungi *	Totals
Number of obtained species (genera)	245 (118)	127 (47)	290 (34)	25 (11)	687 (196)
Proportion of functional groups (%)	36	18	42	4	100
Descriptive statistics of species richness [mean, (SD **, range)]	40.14 (13.33, 20–83)	18.31 (11.65, 0–47)	41.17 (17.13, 14–92)	2.74 (2.17, 0–7)	102.40 (35.12, 38–178)
The five most frequent taxa	*Exidia nigricans*, *Schizopora flavipora*, *Sc. paradoxa* s.l., *Stereum hirsutum*, *St. ochraceoflavum*	*Auriscalpium vulgare*, *Gymnopus peronatus*, *Leotia lubrica*, *Lycoperdon perlatum*, *Mycena pura*	*Clavulina coralloides*, *Laccaria amethystina*, *L. laccata*, *Lactarius subdulcis*, *Russula cyanoxantha*	–	–
The richest genera (number of taxa)	*Mycena* (14), *Pluteus* (11), *Crepidotus* (8), *Postia* (7)	*Mycena* (25), *Clitocybe* (8), *Gymnopus* (8), *Lyophyllum* (6)	*Cortinarius* (100), *Russula* (44), *Inocybe* (28), *Lactarius* (26)	–	–
The number of species found in one sampling unit	74 (30%)	35 (28%)	109 (38%)	11 (44%)	229 (33%)

* five functional groups involved, ** standard deviation

Tree species identity drove wood-inhabiting fungal community composition

Thirty-two taxa (out of 245) were found in 14 or more sampling units (**Fig 2A**; **Supplementary Fig 4**). The explanatory powers and statistical reliability of the six variables fitted onto the final NMDS solution are detailed in **Table 3**. In brief, concordant results were revealed by each NMDS run (including all tested distance methods and dimensionality) and RDA: tree species composition (the relative volumes of dominant tree species) had the strongest effect on wood-inhabiting community composition. In **Fig 2A**, axis 1 represented 17.2 % of the variation and was correlated highly with the relative volume of oaks and the hydrolytic acidity of the soil. Axis 2 (9.4 % of variation) showed a strong correlation with the relative volume of beech, while axis 3 (6.9 % of variation) was related to the species richness of trees, the relative volume of conifers, and the total cover of dead wood. Unexpectedly, dead wood properties had no significant effects in RDA. Both methods pointed out that (1) the high proportions of deciduous (mainly beech and oak) trees on the sampling units were preferred by the majority of fungal species, (2) the relative volumes of tree species defined a clear deciduous–coniferous gradient in the ordination diagrams, and (3) there was no significant effect of the surrounding landscape on the species composition of wood-inhabiting fungi. The effects of air temperature and the historical proportion of meadows were significant based on the RDA only.

Both methods revealed very similar environmental requirements for the most frequent fungal taxa. The following fungal species were strongly associated with beech stands: *Antrodiella fragrans*, *Biscogniauxia nummularia*, *Hypoxylon fragiforme*, *Mycetinis alliaceus*, *Polyporus varius*, *Postia subcaesia*, *Skeletocutis nivea*, *Trametes versicolor*, *Xylaria carpophila* and *X. hypoxylon*. *B. nummularia*, *T. versicolor*, and *X. hypoxylon* were also correlated with more neutral litter pH and trees with larger mean DBH. Wood-inhabiting fungi in oak-dominated stands with higher air temperature and higher soil hydrolytic acidity

were *Hymenochaete rubiginosa*, *Schizopora paradoxa* s.l., *Stereum ochraceoflavum* and *S. subtomentosum*. Common species in coniferous (mainly pine-dominated) stands with higher total cover of dead wood and lower air temperature were *Mycena epipterygia* and *Ramaria stricta* the relative volume of conifers in the NMDS plot and the relative volume of Scots pine in the RDA were highly correlated ($|r| = 0.964$, $p < 0.001$) indicating that the two variables have a very similar effect on wood-inhabiting fungi in the sampling units.

A pH gradient structured terricolous saprotrophic fungal communities

One hundred and twenty-seven taxa were found, out of which 12 species occupied more than 14 sampling units. **Fig 2B** shows the optimal positions of these species in the final NMDS solution, while **Supplementary Fig 5A** depicts the RDA plot. All of the 35 sampling units were examined by RDA, whereas NMDS was run omitting the four sampling units with zero or very low counts of terricolous saprotrophic fungi. Four variables were fitted significantly onto the NMDS solution; their explanatory powers and statistical reliability are shown in **Table 3**. Broadly speaking, both methods gave similar results. Terricolous saprotrophic community composition was driven principally by a definite litter pH gradient along two environmental variables: Scots pine proportion and the pH of litter. The same results were obtained from all NMDS runs where the other tested distance methods and different dimensionalities were applied. Here, NMDS axis 1 represented 35.4 % of the variation and was not correlated strongly with any of the environmental variables. Axis 2 (9.1 % of variation) was correlated highly with the relative volume of Scots pine, the pH of litter, and the density of large trees. The K content of the soil had high scores along axes 2 and 3. By contrast, RDA highlighted two other variables: the mean daily air temperature and the N content of the soil as being of great importance. In general, both of these factors were correlated negatively with the whole fungal community (**Supplementary Fig 5B**). No

significant relations were detected by either RDA or NMDS with respect to the historical forest management or the surrounding landscape.

Concerning the environmental requirements of the frequent species, both methods supported *Auriscalpium vulgare*, *Baeospora myosura*, and *Lycoperdon molle* to be common elements of pine-dominated stands with a low litter pH and a low density of large trees. The positions of other frequent taxa in the two ordination diagrams were rather unstable, but *M. sanguinolenta* and *Rhodocollybia butyracea* were always found to be unrelated to the pH gradient.

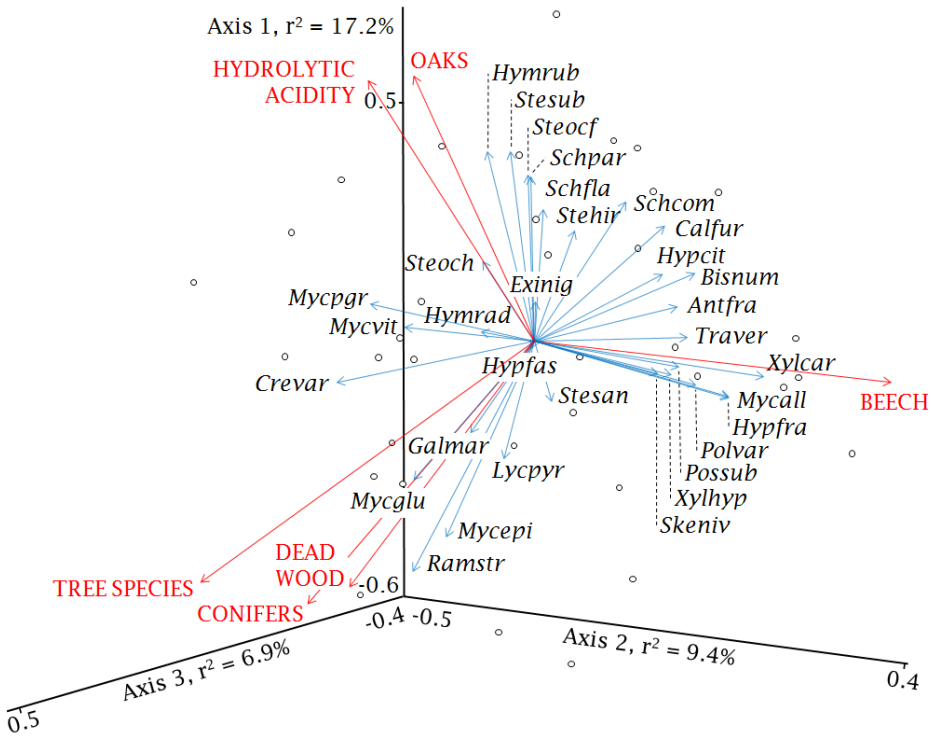
EcM fungi: no obvious gradients detected

Altogether 290 EcM taxa were identified. Thirty of them were frequent, and collected in more than 14 sampling units (**Fig 2C**) and (**Supplementary Fig 6**). Four variables were fitted significantly onto the final NMDS solution (for details of fit see **Table 3**). RDA revealed EcM fungi as a mainly host restricted functional group with the strongest effects being beech proportion and the mean DBH of trees, while NMDS detected the density of large trees (with the highest influence) and substratum related factors (the relative volume of decayed logs, litter pH, and soil P concentration) to be important drivers of EcM fungal species composition. Litter pH and tree size (mean DBH in RDA and large trees in NMDS) were important by both methods. When NMDS was run with the other tested dimensionality and distance methods, it returned concordant results. In **Fig 2C**, NMDS axis 1 explained 33.8 % of the variation and was not related strongly to any of the environmental variables. Axis 2 (15.6 % variation) correlated highly with the P content of the soil, while axis 3 (4.4 % variation) was related strongly to the relative volume of decayed logs, the density of large trees, and the pH of litter. Using RDA, three less important environmental factors were also significant: the proportion of forests in the landscape, the mean relative diffuse light, and the

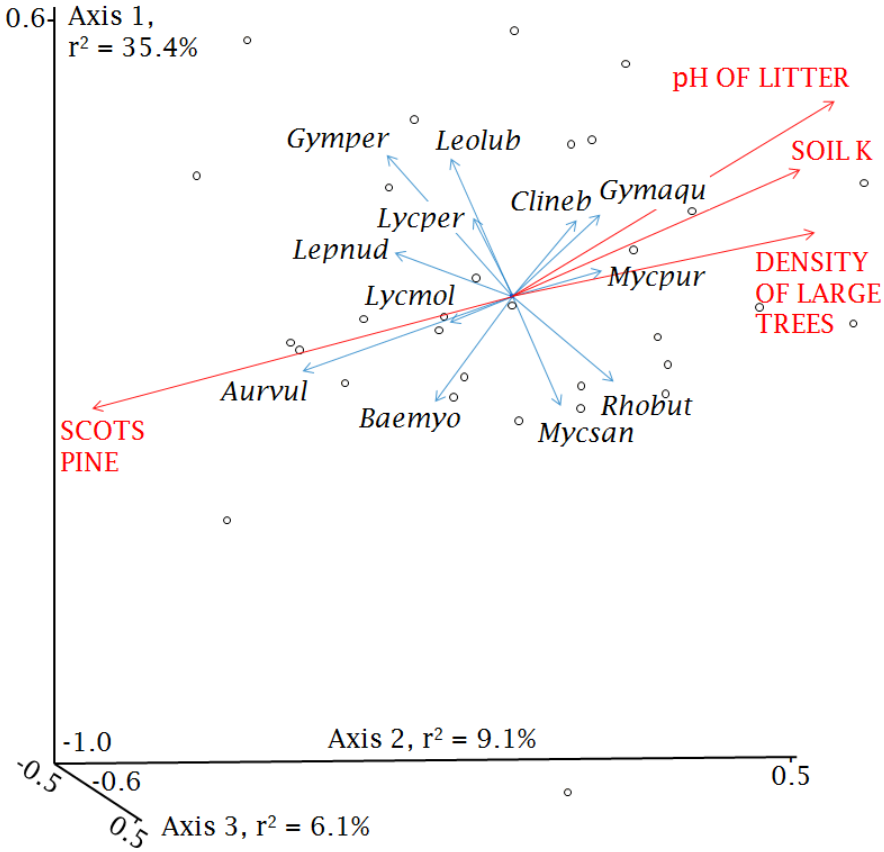
Shannon diversity of landscape elements. No obvious underlying gradients (supported by more than one fitted variable) were detected by either method.

Regarding fungal species, the relative volume of beech in the NMDS model, however, had no significant effect on the whole EcM community, but the optimal positions of beech-dominated sampling units were close to the species *Lactarius blennius*, *Pseudocraterellus undulatus*, *Russula emetica*, and *Tricholoma ustale* (data not shown). Except *P. undulatus*, these species also were associated with high relative volumes of decayed logs. RDA, more or less, underlined these results revealing three more species (*Inocybe petiginosa*, *L. subdulcis*, and *T. sulphureum*) as beech associated ones. Here, *I. petiginosa* and *P. undulatus* preferred closed canopy conditions. For both methods, the placements of *Amanita rubescens*, *Clavulina coralloides*, *L. quietus*, *R. heterophylla*, *R. nigricans*, and *R. undulata* in the ordination space were similar (they were close to each other), but RDA revealed them as characteristic taxa of open stands with more light and lower pH of litter, while NMDS emphasized strong relationships between these species and the high P content of the soil. In both models, mainly oakd-ominated stands were situated close to these taxa.

A



B


0.6
Axis 1,
r² = 35.4%
pH OF LITTER
SOIL K
DENSITY
OF LARGE
TREES
SCOTS
PINE
Gymper
Leolub
Clineb
Gymaqu
Lycper
Lepnud
Mycpur
Lycmol
Aurvul
Baemyo
Rhobut
Mycsan
-1.0
Axis 2, r² = 9.1%
-0.5
-0.6
0.5
0.5
Axis 3, r² = 6.1%

C

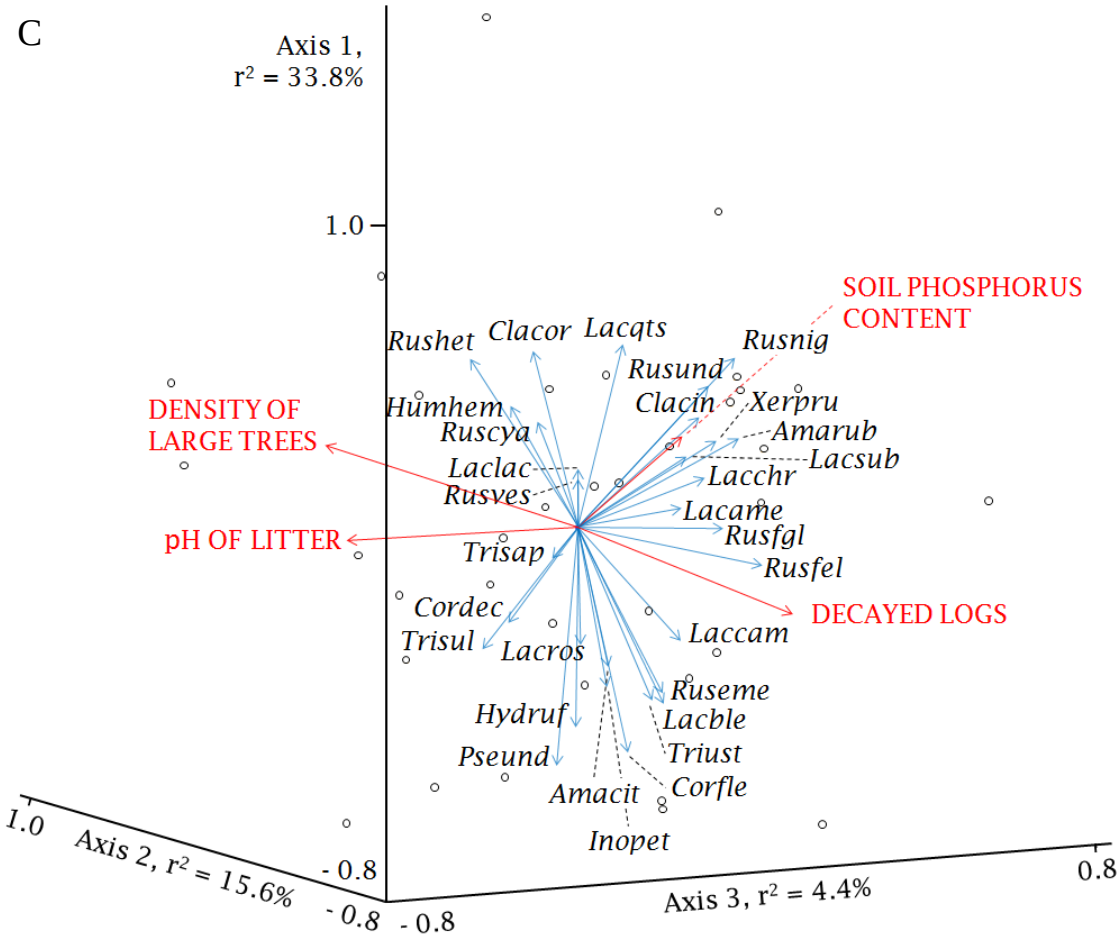

Axis 1,
r² = 33.8%
1.0
SOIL PHOSPHORUS
CONTENT
DENSITY OF
LARGE TREES
pH OF LITTER
DECAYED LOGS
Rushet
Clacor
Lacqts
Rusnig
Rusund
Clacin
Xerpru
Humhem
Ruscya
Amarub
Lacsub
Laclac
Lacchr
Rusves
Lacame
Rusfgl
Trisap
Rusfel
Cordec
Trisul
Lacros
Laccam
Ruseme
Hydruf
Lacble
Pseund
Triust
Amacit
Corfle
Inopet
1.0
Axis 2, r² = 15.6%
- 0.8
- 0.8
- 0.8
Axis 3, r² = 4.4%
0.8

Fig 2 – Local NMDS on wood-inhabiting (**A**), terricolous saprotrophic (**B**) and EcM (**C**) fungal species (black italics) representing the significantly ($p < 0.05$) fitted environmental variables (red capitals) and a tri-plot of sampling units (black circles). The most frequent taxa are displayed; the optimal positions of all recorded species are shown in **Supplementary Figs 1–3(A)** and (**B**) See **Table 3** for the explanatory powers and statistical reliability of environmental variables and **Supplementary Table 1** for abbreviations of species. Three dimensional diagrams were plotted; spinning diagrams provide real 3-D views in **Supplementary Figs 7–9**. NMDS was run on Bray–Curtis distances. The final stress values, following Kruskal (1964), are multiplied by 100 and were 15.282, 14.331 and 12.186, respectively.

Table 3 – Explanatory powers of the variables fitted significantly onto the NMDS results of functional groups (**Fig 2A–C**). The r^2-values are the squared correlation coefficients of the linear regression models built by using the NMDS results as response variables and including each of the environmental variables separately. P-values are based on 999 random permutations of NMDS data.

Environmental variable	r^2	p-value
WOOD-INHABITING FUNGI		
Tree species (species richness of trees)	0.3792	0.003
Oaks (relative volume of oaks)	0.3309	0.006
Hydrolytic acidity (hydrolytic acidity of the soil)	0.3235	0.010
Conifers (relative volume of coniferous trees)	0.3172	0.009
Beech (relative volume of beech)	0.2552	0.030
Dead wood (total cover of FWD and CWD)	0.2442	0.035
TERRICOLOUS SAPROTROPHIC FUNGI		
Scots pine (relative volume of Scots pine)	0.4395	0.002
pH of litter	0.3782	0.005
Soil K (potassium content of the soil)	0.3064	0.026
Density of large (DBH > 50 cm) trees	0.2751	0.043
EcM FUNGI		
Density of large (DBH > 50 cm) trees	0.3651	0.002
Decayed logs (relative volume of logs in decay stages 3–6)	0.2984	0.010
pH of litter	0.2329	0.039
Phosphorus content of the soil	0.2193	0.056

Discussion

Fungal diversity and drivers of frequent taxa

In this work, 687 macrofungal species were recorded in total during only three sporocarp surveys and by studying a restricted number of habitat types. Altogether 30 taxa were obtained with clear concordant responses to the environment based on both the NMDS and RDA diagrams. In general, these results agreed with the findings of other studies, shown in **Table 4**.

Table 4 – Macrofungal taxa with concordant responses to the environment according to both the NMDS and RDA models. Studies (from Central Europe) examining the environmental requirements of fungal species within the European temperate forests are listed. Factors in Column 2 are detailed in **Table 3**; the direction of their effect (increasing ↑ or decreasing ↓ units) is depicted.

Macrofungal taxa	Influential environmental factors revealed		Reference
	in present study	in other studies	
	WOOD-INHABITING FUNGI		
Hypoxylon fragiforme	beech↑	beech↑	Kacprzyk et al., 2014
Mycetinis alliaceus	beech↑	beech↑	Heilmann-Clausen, 2005
Polyporus varius	beech↑	beech↑	Ciortan, 2009
Skeletocutis nivea	beech↑	beech↑	Fischer and Wagner, 1999
Xylaria carpophila	beech↑	beech (cupule litter)↑	Whalley, 1985
Antrodiella fragrans	beech↑	deciduous trees↑	Miettinen et al., 2006
Postia subcaesia	beech↑	deciduous trees↑	Siller, 2004; Szabó, 2012
Biscogniauxia nummularia	beech↑, litter pH↑, mean DBH↑	beech↑	Lakatos and Molnár, 2009
Xylaria hypoxylon	beech↑, litter pH↑, mean DBH↑	beech↑	Heilmann-Clausen, 2005
Trametes versicolor	beech↑, litter pH↑, mean DBH↑	deciduous trees↑, conifers↓	Ryvarden and Gilbertson, 1994
Hymenochaete rubiginosa	oaks↑, air temperature↑, hydrolytic acidity↑	oaks↑, beech↓, hornbeam↓	Papp, 2013
Schizopora paradoxa s.l.	oaks↑, air temperature↑, hydrolytic acidity↑	oaks↑, deciduous trees↑	Bernicchia et al., 2007b, 2008
Stereum subtomentosum	oaks↑, air temperature↑, hydrolytic acidity↑	oaks↑, deciduous trees↑	Bernicchia et al., 2008
Stereum ochraceoflavum	oaks↑, hydrolytic acidity↑	oaks↑	Bernicchia and Gorjón, 2010
Mycena epipterygia	pine↑, dead wood↑, air temperature↓	conifers↑	Krieglsteiner, 2001
Ramaria stricta	pine↑, dead wood↑, tree species↑, air temperature↓	mixed (deciduous–coniferous) stands↑	Breitenbach and Kränzlin, 1986; Krieglsteiner, 2000
	TERRICOLOUS SAPROTROPHIC FUNGI		
Auriscalpium vulgare	pine↑, litter pH↓, density of large trees↓	pine (cones)↑	Bernicchia et al., 2007a
Baeospora myosura	pine↑, litter pH↓, density of large trees↓	conifers↑	Krieglsteiner, 2001
Lycoperdon molle	pine↑, litter pH↓, density of large trees↓	mixed stands↑, open areas↑, soil pH↑	Rimóczi et al., 2011
	EcM FUNGI		
Lactarius blennius	beech↑, decayed logs↑	beech↑	Tyler, 1992; Galli, 2006; Lang et al., 2011
Tricholoma ustale	beech↑, decayed logs↑	beech↑, deciduous trees↑, soil pH↓	Bohus, 1973; Buée et al., 2011
Russula emetica	beech↑, decayed logs↑	beech↑, soil pH↓	Bohus, 1973
Pseudocraterellus undulatus	beech↑, light↓	hornbeam↑, oaks↑, N deposition↑	Tyler, 1992; Suz et al., 2014
Inocybe petiginosa	beech↑, light↓	oaks↑, soil pH↓	Szemere, 1955; Babos, 1989
Lactarius subdulcis	beech↑, soil P↑, litter pH↑	beech↑, soil pH↓	Galli, 2006; Buée et al., 2011
Tricholoma sulphureum	beech↑, soil P↓	beech↑, deciduous trees↑	Christensen and Heilmann-Clausen, 2013
Russula undulata	soil P↑, light↑, litter pH↓	oaks↑, hornbeam↑, soil pH↓	Bohus, 1973
Lactarius quietus	soil P↑, light↑, litter pH↓	oaks↑, soil N↑, soil pH↓	Galli, 2006; Suz et al., 2014
Amanita rubescens	soil P↑, light↑, litter pH↓	soil P, N↑, mixed (deciduous–coniferous) stands↑	Pál-Fám, 2001; Buée et al., 2011
Russula nigricans	soil P↑, light↑, litter pH↓	soil pH↓, mixed (deciduous–coniferous) stands↑	Bohus and Babos, 1967; Pál-Fám, 2001

Tree species composition

It was shown that the species composition of trees has the highest relevance to wood-inhabiting fungal species composition at a scale of forest stands (**Fig 2A**). Comparative studies in Europe have also identified tree species composition to be a major determinant of wood-inhabiting fungal species composition (e.g. Humphrey et al. 2000; Sippola et al. 2005; O'Hanlon & Harrington 2012). In the present study, a clear distinction between coniferous and deciduous tree species was found, which has been confirmed also by other studies (Küffer et al. 2008; Buée et al. 2011). In the present study, more fungal species were found to be related to deciduous trees; however, it is worth mentioning that the proportions of the total volumes of deciduous (65 %) and coniferous trees (28 %) were biased in our sampling units. The number of fungal species in oak, beech and conifer-dominated stands was similar with respect to the total species pool of wood-inhabiting fungi (**Supplementary Fig 1**). The relatively strong effect of tree species on the community composition of wood-inhabiting fungi could be due to the great compositional diversity of tree species in the region. It is known that wood-inhabiting fungi are mainly substratum restricted, as they live within the wood, and species are often selective for certain tree taxa (Boddy & Heilmann-Clausen 2008). It was also underlined by other studies that tree species identity has a marked impact on wood-inhabiting fungal species composition across various spatial scales: indirectly at the centimeter scale via the species specific variation of the chemical environment within the wood, along pH (Schmidt 2006) and compositional differences of compounds (Boddy 1992, 2001; Renvall 1995), and directly at a stand scale (e.g. Heilmann- Clausen et al. 2005; Sippola et al. 2005; McMullan-Fisher et al. 2009) and at a continental scale along the distribution of major forest types (Heilmann-Clausen & Boddy 2008).

In the present study, terricolous saprotrophic fungal species composition was found to be shaped by tree species composition (**Fig 2B**). Previous studies have also confirmed this

finding by pointing out a positive response to tree species diversity at the stand scale (McMullan-Fisher et al. 2009; O'Hanlon & Harrington 2012) or even a negative one (Ferris et al. 2000). Terricolous saprotrophic fungi are thought to be mainly a substratum restricted functional group (Gebauer & Taylor 1999; Boddy et al. 2008) and tree species composition may affect them via the fundamental impacts of tree species on litter quality and quantity.

Regarding the EcM fungi, a contrasting response was revealed to tree species composition: NMDS found no significant effects, but RDA highlighted the proportion of beech to have the strongest importance on EcM species composition (**Supplementary Fig 6**). However, many previous studies (e.g. Såstad 1995; Ferris et al. 2000; Kernaghan et al. 2003; Morris et al. 2009) have supported the idea that EcM species composition is determined principally by the species composition of their host trees at a stand scale, but many other studies came to contradictory conclusions highlighting soil properties (e.g. Talbot et al. 2013; Suz et al. 2014) or other biotic factors (e.g. Kennedy 2010; Peay et al. 2010) as being major determinants. The picture is not clear, because there is usually a striking contrast between the great diversity of EcM fungal communities and the relatively species-poor stands of host trees in temperate forests (Tedersoo et al. 2014). A large number of EcM fungal species can be found on the root surface even of the same tree individual or root tip (Bahram et al. 2011), and until this complexity is better understood at finer scales, results suggesting changes in EcM species composition at a stand scale are a matter of debate (Erland & Taylor 2003).

Stand structure

In the present study, the total cover of FWD and CWD had a significant effect on wood-inhabiting fungal species composition (**Fig 2A**), but CWD volume alone was not important. Many studies (reviewed in Lonsdale et al. 2008) detected the quantity of dead wood to have the highest influence on wood-inhabiting fungi. However, the influence of

FWD and CWD on wood-inhabiting fungi cannot be separated in this study, but a considerable impact of FWD was revealed. Here, CWD was selective for only a very low proportion (12 %) of fungal taxa (details in **Supplementary Table 1**). This is probably because out of the total CWD volume on sites, oak and conifer logs in decay stages 2–3 amounted to 44 % which is mainly heartwood and hence, the most species-poor CWD type (Boddy & Heilmann-Clausen 2008). Comparative studies in Europe (e.g. Küffer et al. 2008; Abrego & Salcedo 2011) have also suggested that a large proportion of wood-inhabiting fungi can be harboured on FWD in managed forests.

The density of large trees (in NMDS, **Fig 2B** and **C**) and the mean DBH of trees (in RDA, **Supplementary Figs 4** and **6**) were significant in structuring the species composition of each functional group. However, these two variables were moderately correlated ($r = 0.381$, $p = 0.024$), but both of them may have the same effect on fungal communities influencing them via the presence of large trees in the forest stands. Only the EcM community was shaped considerably by both of these factors, but such a result, based on sporocarp data, is impossible to interpret adequately. However, large trees can serve as "hubs" in the common mycorrhizal network belowground (reviewed in Simard et al. 2012), or stands in different successional phases (with different tree sizes) can harbour distinctive EcM communities (Smith et al. 2002; Twieg et al. 2007) that can both influence sporocarp occurrences.

The relative volume of decayed logs was revealed to have a significant effect on EcM community composition (**Fig 2C**). The majority of EcM fungi evolved from humus and wood saprotrophic ancestors (Tedersoo et al. 2010), therefore many EcM fungi still have some ability to decompose wood in later decay stages. A similar EcM community response was revealed by Walker et al. (2012) to CWD volume in their clear-cut forest system, emphasizing that dead wood provides a balanced environment for fungi with respect to microclimate and available nutrients. By contrast, it was hypothesized by Baldrian (2009) that the

lignocellulose-decomposing enzymes of EcM fungi may support only escape from a dying root.

Soil and litter conditions

The pH of litter determined the species composition of terricolous saprotrophic fungi and had a considerable effect on EcM fungi (**Fig 2B** and **C**). Similar influences of soil pH have already been published on terricolous saprotrophic (Ferris et al. 2000; Talbot et al. 2013) and EcM community composition (Baar & ter Braak 1996). It was shown in the present study that the underlying litter pH gradient, with an effect on determining terricolous saprotrophic community composition, was related to Scots pine proportion highlighting that the tree species composition has a strong impact on litter pH, and Scots pine has a more acidic litter compared to that of deciduous trees (Augusto et al. 2003).

The weak, but significant effects of soil N, P and K contents on terricolous (EcM and terricolous saprotrophic) communities (**Fig 2B** and **C**, **Supplementary Fig 5A**) cannot be explained without mentioning their relatively strong collinearity (|r| = 0.4–0.5) compared to their relations to the ordination axes (|r| = 0.3–0.5). However, similar relationships were detected by Baar & ter Braak (1996) and Toljander et al. (2006) among N, P and K contents, who suggested that K likely plays a minor role compared to N and P (nutrients) in the occurrence of fungi. Soil K content has been suggested to be important in osmoregulation and sporocarp formation (Tyler 1982). Soil P content was reported by Conn & Dighton (2000) and Morris et al. (2009) to have important consequences for EcM community development, but in our study, only a marginal significance of soil P was detected. There was a general negative impact of soil N content on terricolous saprotrophic fungi in the ŐNP (**Supplementary Fig 5B**), and in other European countries, there have been concordant (e.g. Buée et al. 2011) and contradictory (e.g. Tarvainen et al. 2003) results. However, numerous N fertilization

experiments have been conducted (e.g. Tarvainen et al. 2003) on terricolous macrofungi, but in most cases the fruiting of EcM communities was negatively affected.

Microclimate

Supported by RDA only, wood-inhabiting and terricolous saprotrophic communities were structured by air temperature (**Supplementary Figs 4** and **5**). Regarding wood-inhabiting fungi, this result is in agreement with those of Boddy (1992, 2001) within wood, Renvall (1995) at a stand scale, and Heilmann-Clausen et al. (2014) at a continental scale. The general effect of air temperature on wood-inhabiting fungi is too difficult to interpret in our study. In contrast, air temperature had a clear negative effect on the majority of terricolous saprotrophic fungal species. According to Berg & McClaugherty (2014), the optimal temperature is vital for the right activity of cellulo- and ligninolytic enzymes of this functional group. In accordance, their optimal temperatures in the studied region may be in rather closed stands with shaded litter layers.

Other factors

Revealed by RDA only, management history and landscape characteristics were demonstrated with low and moderate effects on wood-inhabiting and EcM communities, respectively (**Supplementary Figs 4** and **6**). The negative effects of forest management on wood-inhabiting fungi has been widely studied (e.g. Lindner et al. 2006), but such a clear community response was not detected here. Only the EcM community was influenced by landscape characteristics indicating that this group is affected significantly also at larger (r = 300 m) scales compared to the other studied functional groups.

Limitations of data

Our community data is biased by all the disadvantages of using sporocarp incidences to estimate macrofungal abundance [see Tóth & Barta (2010) for a review]. The biggest weakness is the short duration (2 yr) of our field visits that can only provide an underestimate of fungal species richness in the sampling units. It has been shown that additional species can also be found after 21 yr of surveys (Straatsma et al. 2001). Another potential source of error is the variation among years in the fruiting of species (Fernández-Toirán et al. 2006), which also was observed in this study. Given these limitations, the current results must therefore be viewed with caution.

Conclusions

It is hypothesized that substratum properties, tree species composition and microclimate, in that order, are the most influential drivers of fungal species composition in the studied region, and their relative influences differ among functional groups. Wood-inhabiting fungal species composition was driven primarily by the species composition of living trees, while substratum properties and microclimate had minor relevance. The terricolous saprotrophic community was determined principally by a litter pH gradient involving tree species proportions and soil/litter properties. Microclimate had no concordant effect. The EcM fungal species composition was not structured by obvious ecological gradients supported simultaneously by more than one environmental variable, but litter pH and tree size had significant effects. The lack of detected gradients suggests that the most important drivers of EcM fungi remained unmeasured. Regarding each functional group, no clear responses to management history or to the surrounding landscape were found. However, it was confirmed that macrofungal communities are related significantly to environmental drivers that are relatively easy to measure at a stand scale. To gain further insight into

standscale drivers of fungal species composition, sporocarp surveys should be combined with DNA sequence based sampling methods in a below-ground study.

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Supplementary Material

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Supplementary Table 1. List of 687 macrofungi taxa collected and identified in this work. "**Code**" was generated by using the first three letters of the genus and species name of taxa. Occasionally, other letters were applied to avoid making redundant abbreviations. Six-letter codes are shown for analyzed taxa only. The column "**Analyzed by NMDS?**" shows the status of macrofungi taxa in the NMDS models: the species indicated by "yes, plotted" (the most abundant ones) are represented in the NMDS diagrams of the printed paper (the remaining taxa highlighted by "yes" are plotted also, but in Supplementary Fig 1–3); the only "omitted" species, *Entoloma jahnii*, was excluded from NMDS because it was collected exclusively in one of those four omitted (extremely species poor) sampling units that had outlying points in the NMDS of terricolous saprotrophic fungi; "skipped" taxa were excluded due to their currently indefinite trophic status or belonging to a functional group with too low species number for statistical computing. Data are sorted primarily by "**Functional group**" then by the "**Number of occupied sampling units**". Taxa found in less than four sampling units were excluded from the RDAs. Underlined wood-inhabiting taxa were selective for (d > 20 cm) CWD.

This list and the description of sampling units are more detailed in:
Siller, I., Kutszegi, G., Takács, K., Varga, T., Merényi, Zs., Turcsányi, G., Ódor, P., Dima, B., 2013. Sixty-one macrofungi species new to Hungary in Őrség National Park. Mycosphere 4, 871–924.

EcM = ectomycorrhizal; **t. sapr.** = terricolous saprotrophic; **wood-inh.** = wood-inhabiting; **entomopath.** = entomopathogenic; **lign./t. sapr.** = lignicolous and/or terricolous saprotrophic; **t. sapr./myc.** = terricolous saprotrophic and/or mycorrhizal

Macrofungi taxa	Author(s)	Code	Functional group	Number of occupied sampling units	Analyzed by NMDS?
Laccaria amethystina	Cooke	*Lacame*	EcM	34	yes, plotted
Russula cyanoxantha	(Schaeff.) Fr.	*Ruscya*	EcM	34	yes, plotted
Clavulina coralloides	(L.) J. Schröt.	*Clacor*	EcM	28	yes, plotted
Laccaria laccata	(Scop.) Cooke	*Laclac*	EcM	27	yes, plotted
Lactarius subdulcis	(Pers.) Gray	*Lacsub*	EcM	27	yes, plotted
Xerocomus pruinatus	(Fr. & Hök) Quél.	*Xerpru*	EcM	26	yes, plotted
Lactarius blennius	(Fr.) Fr.	*Lacble*	EcM	25	yes, plotted
Cortinarius decipiens s.l.	(Pers.) Fr.	*Cordec*	EcM	24	yes, plotted
Russula emetica	(Schaeff.) Pers.	*Ruseme*	EcM	24	yes, plotted
Clavulina cinerea	(Bull.) J. Schröt.	*Clacin*	EcM	23	yes, plotted
Pseudocraterellus undulatus	(Pers.) Rauschert	*Pseund*	EcM	23	yes, plotted
Russula fellea	(Fr.) Fr.	*Rusfel*	EcM	23	yes, plotted
Inocybe petiginosa	(Fr.) Gillet	*Inopet*	EcM	22	yes, plotted
Tricholoma sulphureum	(Bull.) P. Kumm.	*Trisul*	EcM	21	yes, plotted
Russula fragilis	Fr.	*Rusfgl*	EcM	20	yes, plotted
Russula nigricans	Fr.	*Rusnig*	EcM	20	yes, plotted
Russula vesca	Fr.	*Rusves*	EcM	20	yes, plotted
Russula undulata	Velen.	*Rusund*	EcM	19	yes, plotted
Amanita rubescens	Pers.	*Amarub*	EcM	18	yes, plotted
Lactarius chrysorrheus	Fr.	*Lacchr*	EcM	18	yes, plotted
Lactarius camphoratus	(Bull.) Fr.	*Laccam*	EcM	17	yes, plotted
Lactarius quietus	(Fr.) Fr.	*Lacqts*	EcM	17	yes, plotted
Tricholoma saponaceum	(Fr.) P. Kumm.	*Trisap*	EcM	17	yes, plotted
Tricholoma ustale	(Fr.) P. Kumm.	*Triust*	EcM	17	yes, plotted
Humaria hemisphaerica	(F.H. Wigg.) Fuckel	*Humhem*	EcM	15	yes, plotted
Lactarius rostratus	Heilm.-Claus.	*Lacros*	EcM	15	yes, plotted
Amanita citrina	(Schaeff.) Pers.	*Amacit*	EcM	14	yes, plotted
Cortinarius flexipes var. *flexipes*	(Pers.) Fr.	*Corfle*	EcM	14	yes, plotted
Hydnum rufescens	Pers.	*Hydruf*	EcM	14	yes, plotted
Russula heterophylla	(Fr.) Fr.	*Rushet*	EcM	14	yes, plotted
Cantharellus cibarius	Fr.	*Cancib*	EcM	13	yes
Cortinarius sp.15		*Cor_15*	EcM	13	yes
Cortinarius casimiri	(Velen.) Huijsman	*Corcas*	EcM	13	yes
Cortinarius elatior	Fr.	*Corela*	EcM	13	yes
Lactarius vellereus	(Fr.) Fr.	*Lacvel*	EcM	13	yes

Russula acrifolia	Romagn.	*Rusacr*	EcM	13	yes
Russula grata	Britzelm.	*Rusgra*	EcM	13	yes
Clavulina rugosa	(Bull.) J. Schröt.	*Clarug*	EcM	12	yes
Hebeloma velutipes	Bruchet	*Hebvel*	EcM	12	yes
Russula ochroleuca	Pers.	*Rusoch*	EcM	12	yes
Cortinarius anthracinus	(Fr.) Sacc.	*Corant*	EcM	11	yes
Inocybe assimilata	Britzelm.	*Inoass*	EcM	11	yes
Lactarius serifluus	(DC.) Fr.	*Lacser*	EcM	11	yes
Inocybe geophylla	(Fr.) P. Kumm.	*Inogeo*	EcM	10	yes
Cortinarius cagei	Melot	*Corcag*	EcM	9	yes
Cortinarius flexipes var. *flabellus*	(Fr.) H. Lindstr. & Melot	*Corflf*	EcM	9	yes
Craterellus cornucopioides	(L.) Pers.	*Cracor*	EcM	9	yes
Hebeloma sordescens	Vesterh.	*Hebsor*	EcM	9	yes
Hydnum repandum	L.	*Hydrep*	EcM	9	yes
Lactarius quieticolor	Romagn.	*Lacqtc*	EcM	9	yes
Russula illota	Romagn.	*Rusill*	EcM	9	yes
Cortinarius trivialis s.l.	J.E. Lange	*Cortri*	EcM	8	yes
Lactarius aurantiacus	(Pers.) Gray	*Lacaur*	EcM	8	yes
Paxillus involutus	(Batsch) Fr.	*Paxinv*	EcM	8	yes
Russula sardonia	Fr.	*Russar*	EcM	8	yes
Tricholoma sciodes	(Pers.) C. Martín	*Trisci*	EcM	8	yes
Amanita phalloides	(Fr.) Link	*Amapha*	EcM	7	yes
Cortinarius diasemospermus var. *diasemospermus*	Lamoure	*Cordia*	EcM	7	yes
Cortinarius infractus s.l.	(Pers.) Fr.	*Corinf*	EcM	7	yes
Cortinarius rigidipes	M.M. Moser	*Corrig*	EcM	7	yes
Cortinarius torvus	(Fr.) Fr.	*Cortor*	EcM	7	yes
Lactarius acris	(Bolton) Gray	*Lacacr*	EcM	7	yes
Lactarius glaucescens	Crossl.	*Lacgla*	EcM	7	yes
Leccinum pseudoscabrum	(Kallenb.) Šutara	*Lecpse*	EcM	7	yes
Russula densifolia	Secr. ex Gillet	*Rusden*	EcM	7	yes
Scleroderma areolatum	Ehrenb.	*Sclare*	EcM	7	yes
Tricholoma album	(Schaeff.) P. Kumm.	*Trialb*	EcM	7	yes
Xerocomus badius	(Fr.) E.-J. Gilbert	*Xerbad*	EcM	7	yes
Amanita argentea	Huijsm.	*Amaarg*	EcM	6	yes
Amanita excelsa	(Fr.) Bertill.	*Amaexc*	EcM	6	yes
Cortinarius tabularis	(Fr.) Fr.	*Cortab*	EcM	6	yes
Craterellus lutescens	(Pers.) Fr.	*Cralut*	EcM	6	yes
Hygrophorus eburneus	(Bull.) Fr.	*Hygebu*	EcM	6	yes
Inocybe cincinnata	(Fr.) Quél.	*Inocin*	EcM	6	yes
Russula mairei	Singer	*Rusmai*	EcM	6	yes
Xerocomus subtomentosus	(L.) Quél.	*Xersub*	EcM	6	yes
Cortinarius subporphyropus	Pilát	*Corsbp*	EcM	5	yes
Cortinarius venetus	(Fr.) Fr.	*Corven*	EcM	5	yes
Hygrophorus poëtarum	R. Heim	*Hygpoe*	EcM	5	yes
Inocybe asterospora	Quél.	*Inoast*	EcM	5	yes
Inocybe fuscidula	Velen.	*Inofus*	EcM	5	yes
Inocybe lilacina	(Peck) Kauffman	*Inolil*	EcM	5	yes
Lactarius fuliginosus	(Fr.) Fr.	*Lacful*	EcM	5	yes
Lactarius pterosporus	Romagn.	*Lacpte*	EcM	5	yes
Lactarius uvidus	(Fr.) Fr.	*Lacuvi*	EcM	5	yes
Leccinum aurantiacum	(Bull.) Gray	*Lecaur*	EcM	5	yes
Russula amoenolens	Romagn.	*Rusamo*	EcM	5	yes
Russula caerulea	Fr.	*Ruscae*	EcM	5	yes
Russula raoultii	Quél.	*Rusrao*	EcM	5	yes
Boletus edulis	Bull.	*Boledu*	EcM	4	yes
Cortinarius sp.14		*Cor_14*	EcM	4	yes
Cortinarius emunctus	Fr.	*Coremu*	EcM	4	yes
Cortinarius largus	Fr.	*Corlgs*	EcM	4	yes
Cortinarius psammocephalus	(Bull.) Fr.	*Corpsa*	EcM	4	yes
Elaphomyces muricatus	Fr.	*Elamur*	EcM	4	yes
Russula aquosa	Leclair	*Rusaqu*	EcM	4	yes

Tricholoma portentosum	(Fr.) Quél.	*Tripor*	EcM	4	yes
Tylopilus felleus	(Bull.) P. Karst.	*Tylfel*	EcM	4	yes
Amanita fulva	(Fr.) Fr.	*Amaful*	EcM	3	yes
Amanita gemmata	(Fr.) Bertill.	*Amagem*	EcM	3	yes
Chroogomphus rutilus	(Schaeff.) O.K. Mill.	*Chrrut*	EcM	3	yes
Cortinarius sp.08		*Cor_08*	EcM	3	yes
Cortinarius acetosus	(Velen.) Melot	*Corace*	EcM	3	yes
Cortinarius acutus s.l.	(Pers.) Fr.	*Coracu*	EcM	3	yes
Cortinarius emollitoides	Bidaud, Moënne-Locc. & Reumaux	*Coremo*	EcM	3	yes
Cortinarius erubescens	M.M. Moser	*Coreru*	EcM	3	yes
Cortinarius hinnuleus s.l.	Fr.	*Corhin*	EcM	3	yes
Cortinarius luhmannii	Münzmay, Saar & B. Oertel	*Corluh*	EcM	3	yes
Cortinarius nolaneiformis	(Velenovský) Dima, Niskanen & Liimat.	*Cornol*	EcM	3	yes
Cortinarius olivaceofuscus	Kühner	*Coroli*	EcM	3	yes
Cortinarius talus	Fr.	*Cortal*	EcM	3	yes
Cortinarius violaceus	(L.) Gray	*Corvio*	EcM	3	yes
Craterellus tubaeformis	(Fr.) Quél.	*Cratub*	EcM	3	yes
Hebeloma cavipes	Huijsman	*Hebcav*	EcM	3	yes
Hebeloma crustuliniforme	(Bull.) Quél.	*Hebcru*	EcM	3	yes
Hygrophorus persoonii	Arnolds	*Hygper*	EcM	3	yes
Inocybe hirtella	Bres.	*Inohir*	EcM	3	yes
Inocybe praetervisa	Quél.	*Inopra*	EcM	3	yes
Inocybe sindonia	(Fr.) P. Karst.	*Inosin*	EcM	3	yes
Lactarius circellatus	Fr.	*Laccir*	EcM	3	yes
Lactarius necator	(Bull.) Pers.	*Lacnec*	EcM	3	yes
Russula chloroides	(Krombh.) Bres.	*Ruschl*	EcM	3	yes
Russula odorata	Romagn.	*Rusodo*	EcM	3	yes
Russula pectinatoides	Peck	*Ruspec*	EcM	3	yes
Russula puellula	Ebbesen, F.H. M?ller & Jul. Schäff.	*Ruspla*	EcM	3	yes
Russula sanguinea	(Bull.) Fr.	*Russan*	EcM	3	yes
Suillus bovinus	(L.) Roussel	*Suibov*	EcM	3	yes
Thelephora palmata	(Scop.) Fr.	*Thepal*	EcM	3	yes
Amanita muscaria	(L.) Lam.	*Amamus*	EcM	2	yes
Amanita vaginata	(Bull.) Lam.	*Amavag*	EcM	2	yes
Boletus reticulatus	Schaeff.	*Bolret*	EcM	2	yes
Cortinarius sp.07		*Cor_07*	EcM	2	yes
Cortinarius sp.22		*Cor_22*	EcM	2	yes
Cortinarius alboviolaceus	(Pers.) Fr.	*Coralv*	EcM	2	yes
Cortinarius balaustinus	Fr.	*Corbal*	EcM	2	yes
Cortinarius bolaris	(Pers.) Fr.	*Corbol*	EcM	2	yes
Cortinarius calochrous	(Pers.) Gray	*Corcal*	EcM	2	yes
Cortinarius cinnabarinus	Fr.	*Corcib*	EcM	2	yes
Cortinarius callisteus	(Fr.) Fr.	*Corcll*	EcM	2	yes
Cortinarius croceus	(Schaeff.) Gray	*Corcro*	EcM	2	yes
Cortinarius duracinus s.l.	Fr.	*Cordur*	EcM	2	yes
Cortinarius glaucopus	(Schaeff.) Gray	*Corgla*	EcM	2	yes
Cortinarius lepidopus	Cooke	*Corlep*	EcM	2	yes
Cortinarius melleopallens	(Fr.) Britzelm.	*Cormll*	EcM	2	yes
Cortinarius nemorensis s. *Saar*	(Fr.) J.E. Lange	*Cornem*	EcM	2	yes
Cortinarius orellanus	Fr.	*Corore*	EcM	2	yes
Cortinarius praestigiosus	(Fr.) M.M. Moser	*Corpra*	EcM	2	yes
Cortinarius renidens	Fr.	*Corren*	EcM	2	yes
Cortinarius safranopes	Rob. Henry	*Corsaf*	EcM	2	yes
Cortinarius veregregius	Rob. Henry	*Corver*	EcM	2	yes
Cortinarius vibratilis	(Fr.) Fr.	*Corvib*	EcM	2	yes
Hebeloma birrus	(Fr.) Sacc.	*Hebbir*	EcM	2	yes
Hebeloma hiemale	Bres.	*Hebhie*	EcM	2	yes
Hebeloma radicosum	(Bull.) Ricken	*Hebrad*	EcM	2	yes

Hygrophorus russula	(Schaeff.) Kauffman	*Hygrus*	EcM	2	yes
Inocybe calida	Velen.	*Inocal*	EcM	2	yes
Inocybe cervicolor	(Pers.) Quél.	*Inocer*	EcM	2	yes
Inocybe furfurea	Kühner	*Inofur*	EcM	2	yes
Inocybe jacobi	Kühner	*Inojac*	EcM	2	yes
Inocybe mixtilis	(Britzelm.) Sacc.	*Inomix*	EcM	2	yes
Inocybe nitidiuscula	(Britzelm.) Lapl.	*Inonit*	EcM	2	yes
Inocybe pseudoreducta	Stangl & Glowinski	*Inopse*	EcM	2	yes
Inocybe soluta	Velen.	*Inosol*	EcM	2	yes
Lactarius flexuosus	(Pers.) Gray	*Lacfle*	EcM	2	yes
Lactarius fluens	Boud.	*Lacflu*	EcM	2	yes
Lactarius ruginosus	Romagn.	*Lacrug*	EcM	2	yes
Lactarius torminosus	(Schaeff.) Pers.	*Lactor*	EcM	2	yes
Ramaria fennica var. *fennica* cf.	(P. Karst.) Ricken	*Ram_fe*	EcM	2	yes
Ramaria flavescens cf.	(Schaeff.) R.H. Petersen	*Ram_fl*	EcM	2	yes
Ramaria fennica var. *fumigata*	(Peck) Schild	*Ramfvf*	EcM	2	yes
Russula fragrantissima	Romagn.	*Rusfgs*	EcM	2	yes
Russula foetens	Pers.	*Rusfoe*	EcM	2	yes
Russula graveolens	Romell	*Rusgrv*	EcM	2	yes
Russula puellaris	Fr.	*Ruspls*	EcM	2	yes
Scleroderma citrinum	Pers.	*Sclcit*	EcM	2	yes
Sebacina incrustans	(Pers.) Tul. & C. Tul.	*Sebinc*	EcM	2	yes
Sistotrema confluens	Pers.	*Siscon*	EcM	2	yes
Suillus variegatus	(Sw.) Kuntze	*Suivar*	EcM	2	yes
Xerocomus cisalpinus	Simonini, H. Ladurner & Peintner	*Xercis*	EcM	2	yes
Xerocomus ferrugineus	(Schaeff.) Alessio	*Xerfer*	EcM	2	yes
Xerocomus porosporus	Imler	*Xerpor*	EcM	2	yes
Amanita eliae	Quél.	*Amaeli*	EcM	1	yes
Amanita franchetii	(Boud.) Fayod	*Amafra*	EcM	1	yes
Amanita porphyria	Alb. & Schwein.	*Amapor*	EcM	1	yes
Chalciporus piperatus	(Bull.) Bataille	*Chapip*	EcM	1	yes
Cortinarius sp.01		*Cor_01*	EcM	1	yes
Cortinarius sp.02		*Cor_02*	EcM	1	yes
Cortinarius sp.03		*Cor_03*	EcM	1	yes
Cortinarius sp.04		*Cor_04*	EcM	1	yes
Cortinarius sp.05		*Cor_05*	EcM	1	yes
Cortinarius sp.06		*Cor_06*	EcM	1	yes
Cortinarius sp.09		*Cor_09*	EcM	1	yes
Cortinarius sp.10		*Cor_10*	EcM	1	yes
Cortinarius sp.11		*Cor_11*	EcM	1	yes
Cortinarius sp.12		*Cor_12*	EcM	1	yes
Cortinarius sp.13		*Cor_13*	EcM	1	yes
Cortinarius sp.16		*Cor_16*	EcM	1	yes
Cortinarius sp.17		*Cor_17*	EcM	1	yes
Cortinarius sp.18		*Cor_18*	EcM	1	yes
Cortinarius sp.19		*Cor_19*	EcM	1	yes
Cortinarius sp.20		*Cor_20*	EcM	1	yes
Cortinarius sp.21		*Cor_21*	EcM	1	yes
Cortinarius albocyaneus	Fr.	*Coralc*	EcM	1	yes
Cortinarius anomalus	(Fr.) Fr.	*Corano*	EcM	1	yes
Cortinarius anserinus	(Velen.) Rob. Henry	*Corans*	EcM	1	yes
Cortinarius barbatus	(Batsch) Melot	*Corbar*	EcM	1	yes
Cortinarius bataillei	J. Favre	*Corbat*	EcM	1	yes
Cortinarius camphoratus	(Fr) Fr.	*Corcam*	EcM	1	yes
Cortinarius caperatus	(Pers.) Fr.	*Corcap*	EcM	1	yes
Cortinarius cinnamomeus	(L.) Gray	*Corcin*	EcM	1	yes
Cortinarius citrinus	(J.E. Lange) P.D. Orton	*Corcit*	EcM	1	yes
Cortinarius comptulus	M.M. Moser	*Corcom*	EcM	1	yes
Cortinarius croceocaeruleus	(Pers.) Fr.	*Corcrc*	EcM	1	yes
Cortinarius delibutus	Fr.	*Cordel*	EcM	1	yes
Cortinarius depressus	Fr.	*Cordep*	EcM	1	yes

Cortinarius diasemospermus var. *leptospermus*	H. Lindstr.	*Cordil*	EcM	1	yes
Cortinarius flexipes var. *inolens*	H. Lindstr.	*Corfli*	EcM	1	yes
Cortinarius fulvescens s.l.	Fr.	*Corful*	EcM	1	yes
Cortinarius herpeticus	Fr.	*Corher*	EcM	1	yes
Cortinarius lebretonii	Quél.	*Corleb*	EcM	1	yes
Cortinarius obtusus	(Fr.) Fr.	*Corobt*	EcM	1	yes
Cortinarius raphanoides	(Pers.) Fr.	*Corrap*	EcM	1	yes
Cortinarius subbalaustinus	Rob. Henry	*Corsbb*	EcM	1	yes
Cortinarius subpurpurascens	(Batsch) Fr.	*Corsbu*	EcM	1	yes
Cortinarius scaurotraganoides	Rob. Henry ex Rob. Henry	*Corsca*	EcM	1	yes
Cortinarius semisanguineus	(Fr.) Gillet	*Corsem*	EcM	1	yes
Cortinarius turgidus	Fr.	*Cortur*	EcM	1	yes
Cortinarius uraceonemoralis	Niskanen, Liimat., Dima, Kytöv., Bojantchev & H. Lindstr.	*Corura*	EcM	1	yes
Cortinarius urbicus	(Fr.) Fr.	*Corurb*	EcM	1	yes
Cortinarius valgus	Fr.	*Corval*	EcM	1	yes
Cortinarius variecolor	(Pers.) Fr.	*Corvar*	EcM	1	yes
Cortinarius vulpinus	(Velen.) Rob. Henry	*Corvul*	EcM	1	yes
Cortinarius xanthocephalus	P.D. Orton	*Corxcp*	EcM	1	yes
Cortinarius xanthophyllus	(Cooke) Rob. Henry	*Corxph*	EcM	1	yes
Gomphidius roseus	(Fr.) Fr.	*Gomros*	EcM	1	yes
Hebeloma candidipes	Bruchet	*Hebcan*	EcM	1	yes
Hebeloma sacchariolens	Quél.	*Hebsac*	EcM	1	yes
Hydnum sp.		*Hyd_sp*	EcM	1	yes
Hygrophorus agathosmus	(Fr.) Fr.	*Hygaga*	EcM	1	yes
Hygrophorus lindtneri	M.M. Moser	*Hyglin*	EcM	1	yes
Hygrophorus penarioides	Jacobsson & E. Larss.	*Hygpen*	EcM	1	yes
Hygrophorus unicolor	Gröger	*Hyguni*	EcM	1	yes
Inocybe amblyospora cf.	Kühner	*Ino_am*	EcM	1	yes
Inocybe auricoma cf.	(Batsch) J.E. Lange	*Ino_au*	EcM	1	yes
Inocybe castanea	Peck	*Inocas*	EcM	1	yes
Inocybe flocculosa	Sacc.	*Inoflo*	EcM	1	yes
Inocybe grammata	Quél. & Le Bret.	*Inogra*	EcM	1	yes
Inocybe leiocephala	D.E. Stuntz	*Inolei*	EcM	1	yes
Inocybe microspora	J.E. Lange	*Inomic*	EcM	1	yes
Inocybe putilla	Bres.	*Inoput*	EcM	1	yes
Inocybe rimosa	(Bull.) P. Kumm.	*Inorim*	EcM	1	yes
Inocybe splendens	R. Heim	*Inospl*	EcM	1	yes
Lactarius bertillonii	(Neuhoff ex Z. Schaef.) Bon	*Lacber*	EcM	1	yes
Laccaria bicolor	(Maire) P.D. Orton	*Lacbic*	EcM	1	yes
Lactarius deterrimus	Gröger	*Lacdet*	EcM	1	yes
Lactarius glyciosmus	(Fr.) Fr.	*Lacgly*	EcM	1	yes
Lactarius pallidus	Pers.	*Lacpal*	EcM	1	yes
Lactarius vietus	(Fr.) Fr.	*Lacvie*	EcM	1	yes
Leccinum cyaneobasileucum	Lannoy & Estad?s	*Leccya*	EcM	1	yes
Leucocortinarius bulbiger	(Alb. & Schwein.) Singer	*Leubul*	EcM	1	yes
Phellodon melaleucus	(Sw. ex Fr.) P. Karst.	*Phemel*	EcM	1	yes
Ramaria fagetorum cf.	Maas Geest. ex Schild	*Ram_fa*	EcM	1	yes
Ramaria formosa	(Pers.) Quél.	*Ramfor*	EcM	1	yes
Rhizopogon roseolus	(Corda) Th. Fr.	*Rhiros*	EcM	1	yes
Russula aeruginea	Lindblad	*Rusaer*	EcM	1	yes
Russula amarissima	Romagn. & E.-J. Gilbert	*Rusama*	EcM	1	yes
Russula clavipes	Velen.	*Ruscla*	EcM	1	yes
Russula cremeoavellanea	Singer	*Ruscre*	EcM	1	yes
Russula farinipes	Romell	*Rusfar*	EcM	1	yes
Russula grisea	Fr.	*Rusgri*	EcM	1	yes
Russula lutensis	Romagn. & Le Gal	*Ruslut*	EcM	1	yes
Russula minutula	Velen.	*Rusmin*	EcM	1	yes

Russula nitida	(Pers.) Fr.	*Rusnit*	EcM	1	yes
Russula pseudointegra	Arnould & Goris	*Ruspse*	EcM	1	yes
Russula queletii	Fr.	*Rusque*	EcM	1	yes
Russula rhodella	E.-J. Gilbert	*Rusrho*	EcM	1	yes
Russula solaris	Ferd. & Winge	*Russol*	EcM	1	yes
Russula tinctipes	J. Blum ex Bon	*Rustin*	EcM	1	yes
Russula torulosa	Bres.	*Rustor*	EcM	1	yes
Russula virescens	(Schaeff.) Fr.	*Rusvir*	EcM	1	yes
Scleroderma cepa	Pers.	*Sclcep*	EcM	1	yes
Strobilomyces strobilaceus	(Scop.) Berk.	*Strstr*	EcM	1	yes
Suillus luteus	(L.) Roussel	*Suilut*	EcM	1	yes
Thelephora terrestris	Ehrh.	*Theter*	EcM	1	yes
Tricholoma batschii	Gulden	*Tribat*	EcM	1	yes
Tricholoma scalpturatum	(Fr.) Quél.	*Trisca*	EcM	1	yes
Tricholoma stiparophyllum	(N. Lund) P. Karst.	*Tristi*	EcM	1	yes
Xerocomus chrysonema	A.E. Hills & A.F.S. Taylor	*Xerchr*	EcM	1	yes
Xerocomus parasiticus	(Bull.) Quél.	*Xerpar*	EcM	1	yes
Xerocomus ripariellus	Redeuilh	*Xerrip*	EcM	1	yes
Auriscalpium vulgare	Gray	*Aurvul*	t. sapr.	25	yes, plotted
Lycoperdon perlatum	Pers.	*Lycper*	t. sapr.	24	yes, plotted
Mycena pura	(Pers.) P. Kumm.	*Mycpur*	t. sapr.	22	yes, plotted
Gymnopus peronatus	(Bolton) Antonín, Halling & Noordel.	*Gymper*	t. sapr.	21	yes, plotted
Leotia lubrica	(Scop.) Pers.	*Leolub*	t. sapr.	20	yes, plotted
Mycena sanguinolenta	(Alb. & Schwein.) P. Kumm.	*Mycsan*	t. sapr.	18	yes, plotted
Gymnopus aquosus	(Bull.) Antonín & Noordel.	*Gymaqu*	t. sapr.	17	yes, plotted
Rhodocollybia butyracea	(Bull.) Lennox	*Rhobut*	t. sapr.	16	yes, plotted
Clitocybe nebularis	(Batsch) P. Kumm.	*Clineb*	t. sapr.	15	yes, plotted
Baeospora myosura	(Fr.) Singer	*Baemyo*	t. sapr.	14	yes, plotted
Lepista nuda	(Bull.) Cooke	*Lepnud*	t. sapr.	14	yes, plotted
Lycoperdon molle	Pers.	*Lycmol*	t. sapr.	14	yes, plotted
Mycena galopus var. *galopus*	(Pers.) P. Kumm.	*Mycglp*	t. sapr.	13	yes
Lepiota clypeolaria	(Bull.) P. Kumm.	*Lepcly*	t. sapr.	12	yes
Mycena aurantiomarginata	(Fr.) Quél.	*Mycaur*	t. sapr.	12	yes
Mycena flavescens	Velen.	*Mycflv*	t. sapr.	12	yes
Mycena rosea	Gramberg	*Mycrsa*	t. sapr.	11	yes
Clitocybe ditopa	(Fr.) Gillet	*Clidit*	t. sapr.	10	yes
Gymnopus erythropus	(Pers.) Antonín, Halling & Noordel.	*Gymery*	t. sapr.	10	yes
Mycena zephirus	(Fr.) P. Kumm.	*Myczep*	t. sapr.	10	yes
Lepista flaccida	(Sowerby) Pat.	*Lepfla*	t. sapr.	9	yes
Clitocybe candicans	(Pers.) P. Kumm.	*Clican*	t. sapr.	8	yes
Clitocybe phyllophila	(Pers.) P. Kumm.	*Cliphy*	t. sapr.	8	yes
Gymnopus androsaceus	(L.) J.L. Mata & R.H. Petersen	*Gymand*	t. sapr.	8	yes
Infundibulicybe gibba	(Pers.) Harmaja	*Infgib*	t. sapr.	8	yes
Lepiota castanea	Quél.	*Lepcas*	t. sapr.	8	yes
Marasmius bulliardii	Quél.	*Marbul*	t. sapr.	8	yes
Atheniella flavoalba	(Fr.) Redhead, Moncalvo, Vilgalys, Desjardin, B.A. Perry	*Athfla*	t. sapr.	7	yes
Lycoperdon nigrescens	Pers.	*Lycnig*	t. sapr.	7	yes
Lyophyllum platypum	Kühner	*Lyopla*	t. sapr.	7	yes
Strobilurus tenacellus	(Pers.) Singer	*Strten*	t. sapr.	7	yes
Collybia cirrata	(Schumach.) Quél.	*Colcir*	t. sapr.	6	yes
Conocybe tetrasporoides	Hauskn.	*Contet*	t. sapr.	6	yes
Hygrophoropsis aurantiaca	(Wulfen) Maire	*Hygaur*	t. sapr.	6	yes
Macrotyphula juncea	(Alb. & Schwein.) Berthier	*Macjun*	t. sapr.	6	yes

Macrolepiota procera	(Scop.) Singer	*Macpro*	t. sapr.	6	yes
Mycena amicta	(Fr.) Quél.	*Mycami*	t. sapr.	6	yes
Mycena galopus var. *leucogala*	(Cooke) J.E. Lange	*Mycgll*	t. sapr.	6	yes
Collybia tuberosa	(Bull.) P. Kumm.	*Coltub*	t. sapr.	5	yes
Entoloma juncinum	(Kühner & Romagn.) Noordel.	*Entjun*	t. sapr.	5	yes
Gymnopus confluens	(Pers.) Antonín, Halling & Noordel.	*Gymcon*	t. sapr.	5	yes
Gymnopus dryophilus	(Bull.) Murrill	*Gymdry*	t. sapr.	5	yes
Lycoperdon excipuliforme	(Scop.) Pers.	*Lycexc*	t. sapr.	5	yes
Mycena rosella	(Fr.) P. Kumm.	*Mycrla*	t. sapr.	5	yes
Mycena stylobates	(Pers.) P. Kumm.	*Mycsty*	t. sapr.	5	yes
Roridomyces roridus	(Scop.) Rexer	*Rorror*	t. sapr.	5	yes
Tubaria minutalis	Romagn.	*Tubmin*	t. sapr.	5	yes
Clitocybe metachroa	(Fr.) P. Kumm.	*Climet*	t. sapr.	4	yes
Clitocybe phaeophthalma	(Pers.) Kuyper	*Clipha*	t. sapr.	4	yes
Collybia cookei	(Bres.) J.D. Arnold	*Colcoo*	t. sapr.	4	yes
Conocybe moseri	Watling	*Conmos*	t. sapr.	4	yes
Lyophyllum mephiticum	(Fr.) Singer	*Lyomep*	t. sapr.	4	yes
Mycena capillaris	(Schumach.) P. Kumm.	*Myccap*	t. sapr.	4	yes
Naucoria bohemica	Velen.	*Nauboh*	t. sapr.	4	yes
Ramaria flaccida	(Fr.) Bourdot	*Ramfla*	t. sapr.	4	yes
Agaricus essettei	Bon	*Agaess*	t. sapr.	3	yes
Chlorophyllum olivieri	(Barla) Vellinga	*Chloli*	t. sapr.	3	yes
Clitocybe odora	(Bull.) P. Kumm.	*Cliodo*	t. sapr.	3	yes
Clitopilus prunulus	(Scop.) P. Kumm.	*Clipru*	t. sapr.	3	yes
Cystoderma amianthinum	(Scop.) Fayod	*Cysami*	t. sapr.	3	yes
Gymnopus quercophilus	(Pouzar) Antonín & Noordel.	*Gymque*	t. sapr.	3	yes
Helvella elastica	Bull.	*Helela*	t. sapr.	3	yes
Lepiota cristata	(Bolton) P. Kumm.	*Lepcri*	t. sapr.	3	yes
Lepiota ignivolvata	Bousset & Joss. ex Joss.	*Lepign*	t. sapr.	3	yes
Marasmius cohaerens	(Pers.) Cooke & Quél.	*Marcoh*	t. sapr.	3	yes
Mycena filopes	(Bull.) P. Kumm.	*Mycfil*	t. sapr.	3	yes
Mycena metata	(Fr.) P. Kumm.	*Mycmet*	t. sapr.	3	yes
Phallus impudicus	L.	*Phaimp*	t. sapr.	3	yes
Pholiotina brunnea	(Watling) Singer	*Phobru*	t. sapr.	3	yes
Ripartites tricholoma	(Alb. & Schwein.) P. Karst.	*Riptri*	t. sapr.	3	yes
Strobilurus stephanocystis	(Kühner & Romagn. ex Hora) Singer	*Strste*	t. sapr.	3	yes
Tubaria conspersa	(Pers.) Fayod	*Tubcon*	t. sapr.	3	yes
Tubaria furfuracea	(Pers.) Gillet	*Tubfur*	t. sapr.	3	yes
Agaricus semotus	Fr.	*Agasem*	t. sapr.	2	yes
Ampulloclitocybe clavipes	(Pers.) Redhead, Lutzoni, Moncalvo & Vilgalys	*Ampcla*	t. sapr.	2	yes
Anthina flammea	(Jungh.) Fr.	*Antfla*	t. sapr.	2	yes
Clitocybe fragrans	(With.) P. Kumm.	*Clifra*	t. sapr.	2	yes
Conocybe enderlei var. *enderlei*	Hauskn.	*Conend*	t. sapr.	2	yes
Cystolepiota seminuda	(Lasch) Bon	*Cyssem*	t. sapr.	2	yes
Entoloma hebes	(Romagn.) Trimbach	*Entheb*	t. sapr.	2	yes
Gymnopus ocior	(Pers.) Antonín & Noordel.	*Gymoci*	t. sapr.	2	yes
Helvella lacunosa	Afzel.	*Hellac*	t. sapr.	2	yes
Helvella macropus	(Pers.) Gray	*Helmac*	t. sapr.	2	yes
Lyophyllum leucophaeatum	(P. Karst.) P. Karst.	*Lyoleu*	t. sapr.	2	yes
Mycena abramsii cf.	(Murrill) Murrill	*Myc_ab*	t. sapr.	2	yes
Mycena fagetorum cf.	(Fr.) Gillet	*Myc_fa*	t. sapr.	2	yes
Mycena clavicularis	(Fr.) Gillet	*Myccla*	t. sapr.	2	yes
Mycena diosma	Krieglst. & Schwöbel	*Mycdio*	t. sapr.	2	yes
Mycena rubromarginata	(Fr.) P. Kumm.	*Mycrub*	t. sapr.	2	yes
Peziza saniosa	Schrad.	*Pezsan*	t. sapr.	2	yes

Rhodocybe gemina	(Paulet) Kuyper & Noordel.	*Rhogem*	t. sapr.	2	yes
Strobilurus esculentus	(Wulfen) Singer	*Stresc*	t. sapr.	2	yes
Agaricus sylvaticus	Schaeff.	*Agasyl*	t. sapr.	1	yes
Agrocybe vervacti	(Fr.) Singer	*Agrver*	t. sapr.	1	yes
Ciboria amentacea cf.	(Balb.) Fuckel	*Cib_am*	t. sapr.	1	yes
Clavariadelphus pistillaris	(L.) Donk	*Clapis*	t. sapr.	1	yes
Conocybe macrocephala cf.	Kühner & Watling	*Con_ma*	t. sapr.	1	yes
Conocybe ochrostriata var. *ochrostriata*	Hauskn.	*Conovo*	t. sapr.	1	yes
Coprinopsis jonesii	(Peck) Redhead, Vilgalys & Moncalvo	*Copjon*	t. sapr.	1	yes
Cystodermella cinnabarina	(Alb. & Schwein.) Harmaja	*Cyscin*	t. sapr.	1	yes
Entoloma conferendum var. *pusillum*	(Velen.) Noordel.	*Entcon*	t. sapr.	1	yes
Entoloma jahnii	Wölfel & Winterh.	–	t. sapr.	1	no, omitted
Lepiota boudieri	Bres.	*Lepbou*	t. sapr.	1	yes
Lepista glaucocana	(Bres.) Singer	*Lepgla*	t. sapr.	1	yes
Lycoperdon lividum	Pers.	*Lycliv*	t. sapr.	1	yes
Lyophyllum baeospermum	Romagn.	*Lyobae*	t. sapr.	1	yes
Lyophyllum boudieri	Kühner & Romagn.	*Lyobou*	t. sapr.	1	yes
Lyophyllum rancidum	(Fr.) Singer	*Lyoran*	t. sapr.	1	yes
Macrocystidia cucumis	(Pers.) Joss.	*Maccuc*	t. sapr.	1	yes
Macrolepiota mastoidea	(Fr.) Singer	*Macmas*	t. sapr.	1	yes
Marasmius epiphyllus	(Pers.) Fr.	*Marepi*	t. sapr.	1	yes
Marasmius setosus	(Sowerby) Noordel.	*Marset*	t. sapr.	1	yes
Marasmius wynneae	Berk. & Broome	*Marwyn*	t. sapr.	1	yes
Mycena rebaudengi cf.	Robich	*Myc_re*	t. sapr.	1	yes
Mycena cinerella	(P. Karst.) P. Karst.	*Myccin*	t. sapr.	1	yes
Mycena pelianthina	(Fr.) Quél.	*Mycpel*	t. sapr.	1	yes
Mycena polyadelpha	(Lasch) Kühner	*Mycpla*	t. sapr.	1	yes
Mycena rhenana	Maas Geest. & Winterh.	*Mycrhe*	t. sapr.	1	yes
Mycena vulgaris	(Pers.) P. Kumm.	*Mycvul*	t. sapr.	1	yes
Mycetinis scorodonius	(Fr.) A. Wilson & Desjardin	*Mycsco*	t. sapr.	1	yes
Peziza arvernensis cf.	Roze & Boud.	*Pez_ar*	t. sapr.	1	yes
Peziza badia	Pers.	*Pezbad*	t. sapr.	1	yes
Peziza phyllogena	Cooke	*Pezphy*	t. sapr.	1	yes
Peziza succosa	Berk.	*Pezsuc*	t. sapr.	1	yes
Ramaria eumorpha	(P. Karst.) Corner	*Rameum*	t. sapr.	1	yes
Stropharia cyanea	(Bull.) Tuom.	*Strcya*	t. sapr.	1	yes
Tarzetta cupularis	(L.) Svrček	*Tarcup*	t. sapr.	1	yes
Stereum hirsutum	(Willd.) Pers.	*Stehir*	wood-inh.	35	yes, plotted
Exidia nigricans	(With.) P. Roberts	*Exinig*	wood-inh.	31	yes, plotted
Schizopora paradoxa s.l.	(Schrad.) Donk	*Schpar*	wood-inh.	31	yes, plotted
Schizopora flavipora	(Berk. & M.A. Curtis ex Cooke) Ryvarden	*Schfla*	wood-inh.	29	yes, plotted
Stereum ochraceoflavum	(Schwein.) Sacc.	*Steocf*	wood-inh.	29	yes, plotted
Steccherinum ochraceum	(Pers.) Gray	*Steoch*	wood-inh.	26	yes, plotted
Mycena vitilis	(Fr.) Quél.	*Mycvit*	wood-inh.	25	yes, plotted
Xylaria hypoxylon	(L.) Grev.	*Xylhyp*	wood-inh.	25	yes, plotted
Hymenochaete rubiginosa	(Dicks.) Lév.	*Hymrub*	wood-inh.	24	yes, plotted
Antrodiella fragrans	(A. David & Tortič) A. David & Tortič	*Antfra*	wood-inh.	21	yes, plotted
Skeletocutis nivea	(Jungh.) Jean Keller	*Skeniv*	wood-inh.	21	yes, plotted
Hymenopellis radicata	(Relhan) R.H. Petersen	*Hymrad*	wood-inh.	19	yes, plotted
Mycetinis alliaceus	(Jacq.) Earle ex A.W. Wilson & Desjardin	*Mycall*	wood-inh.	19	yes, plotted
Biscogniauxia nummularia	(Bull.) Kuntze	*Bisnum*	wood-inh.	18	yes, plotted
Mycena polygramma	(Bull.) Gray	*Mycpgr*	wood-inh.	18	yes, plotted
Stereum subtomentosum	Pouzar	*Stesub*	wood-inh.	18	yes, plotted

Crepidotus variabilis	(Pers.) P. Kumm.	*Crevar*	wood-inh.	17	yes, plotted
Galerina marginata	(Batsch) Kühner	*Galmar*	wood-inh.	16	yes, plotted
Hypocrea citrina	(Pers.) Fr.	*Hypcit*	wood-inh.	16	yes, plotted
Lycoperdon pyriforme	Willd.	*Lycpyr*	wood-inh.	16	yes, plotted
Mycena epipterygia	(Scop.) Gray	*Mycepi*	wood-inh.	16	yes, plotted
Ramaria stricta	(Pers.) Quél.	*Ramstr*	wood-inh.	16	yes, plotted
Stereum sanguinolentum	(Alb. & Schwein.) Fr.	*Stesan*	wood-inh.	16	yes, plotted
Trametes versicolor	(L.) Pilát	*Traver*	wood-inh.	16	yes, plotted
Hypholoma fasciculare	(Huds.) P. Kumm.	*Hypfas*	wood-inh.	15	yes, plotted
Postia subcaesia	(A. David) Jülich	*Possub*	wood-inh.	15	yes, plotted
Schizophyllum commune	Fr.	*Schcom*	wood-inh.	15	yes, plotted
Xylaria carpophila	(Pers.) Fr.	*Xylcar*	wood-inh.	15	yes, plotted
Calocera furcata	(Fr.) Fr.	*Calfur*	wood-inh.	14	yes, plotted
Hypoxylon fragiforme	(Pers.) J. Kickx f.	*Hypfra*	wood-inh.	14	yes, plotted
Mycena galericulata	(Scop.) Gray	*Mycglu*	wood-inh.	14	yes, plotted
Polyporus varius	(Pers.) Fr.	*Polvar*	wood-inh.	14	yes, plotted
Cyathus striatus	(Huds.) Willd.	*Cyastr*	wood-inh.	13	yes
Pluteus cervinus	(Schaeff.) P. Kumm.	*Plucer*	wood-inh.	13	yes
Postia stiptica	(Pers.) Jülich	*Possti*	wood-inh.	13	yes
Trametes hirsuta	(Wulfen) Lloyd	*Trahir*	wood-inh.	13	yes
Clitocybula platyphylla	(Pers.) Malençon & Bertault	*Clipla*	wood-inh.	12	yes
Diatrype stigma	(Hoffm.) Fr.	*Diasti*	wood-inh.	12	yes
Fuscoporia contigua	(Pers.) G. Cunn.	*Fuscon*	wood-inh.	12	yes
Panellus stipticus	(Bull.) P. Karst.	*Pansti*	wood-inh.	12	yes
Aleurodiscus disciformis	(DC.) Pat.	*Aledis*	wood-inh.	11	yes
Armillaria lutea	Gillet	*Armlut*	wood-inh.	11	yes
Exidia glandulosa	(Bull.) Fr.	*Exigla*	wood-inh.	11	yes
Junghuhnia nitida	(Pers.) Ryvarden	*Junnit*	wood-inh.	11	yes
Mycena maculata	P. Karst.	*Mycmac*	wood-inh.	11	yes
Phlebia rufa	(Pers.) M.P. Christ.	*Phlruf*	wood-inh.	11	yes
Psathyrella pygmaea	(Bull.) Singer	*Psapyg*	wood-inh.	10	yes
Heterobasidion annosum	(Fr.) Bref.	*Hetann*	wood-inh.	9	yes
Laxitextum bicolor	(Pers.) Lentz	*Laxbic*	wood-inh.	9	yes
Mycena haematopus	(Pers.) P. Kumm.	*Mychae*	wood-inh.	9	yes
Plicaturopsis crispa	(Pers.) D.A. Reid	*Plicri*	wood-inh.	9	yes
Pseudohydnum gelatinosum	(Scop.) P. Karst.	*Psegel*	wood-inh.	9	yes
Antrodia albida	(Fr.) Donk	*Antalb*	wood-inh.	8	yes
Antrodia malicola	(Berk. & M.A. Curtis) Donk	*Antmal*	wood-inh.	8	yes
Auricularia auricula-judae	(Bull.) Quél.	*Auraur*	wood-inh.	8	yes
Crepidotus cesatii	(Rabenh.) Sacc.	*Creces*	wood-inh.	8	yes
Galerina pruinatipes	A.H. Sm.	*Galpru*	wood-inh.	8	yes
Gymnopilus penetrans	(Fr.) Murrill	*Gympen*	wood-inh.	8	yes
Hypholoma lateritium	(Schaeff.) P. Kumm.	*Hyplat*	wood-inh.	8	yes
Pholiota lenta	(Pers.) Singer	*Pholen*	wood-inh.	8	yes
Polyporus alveolaris	(DC.) Bondartsev & Singer	*Polalv*	wood-inh.	8	yes
Simocybe centunculus	(Fr.) Singer	*Simcen*	wood-inh.	8	yes
Steccherinum fimbriatum	(Pers.) J. Erikss.	*Stefim*	wood-inh.	8	yes
Xylaria polymorpha	(Pers.) Grev.	*Xylpol*	wood-inh.	8	yes
Antrodiella faginea	Vampola & Pouzar	*Antfag*	wood-inh.	7	yes
Byssomerulius corium	(Pers.) Parmasto	*Byscor*	wood-inh.	7	yes
Calocera viscosa	(Pers.) Fr.	*Calvis*	wood-inh.	7	yes
Mycena arcangeliana	Bres.	*Mycarc*	wood-inh.	7	yes
Psathyrella piluliformis	(Bull.) P.D. Orton	*Psapil*	wood-inh.	7	yes
Bjerkandera adusta	(Willd.) P. Karst.	*Bjeadu*	wood-inh.	6	yes
Ceriporiopsis mucida	(Pers.) Gilb. & Ryvarden	*Cermuc*	wood-inh.	6	yes
Daedaleopsis confragosa	(Bolton) J. Schröt.	*Daecon*	wood-inh.	6	yes
Diatrype disciformis	(Hoffm.) Fr.	*Diadis*	wood-inh.	6	yes
Marasmiellus ramealis	(Bull.) Singer	*Marram*	wood-inh.	6	yes
Mycena inclinata	(Fr.) Quél.	*Mycinc*	wood-inh.	6	yes

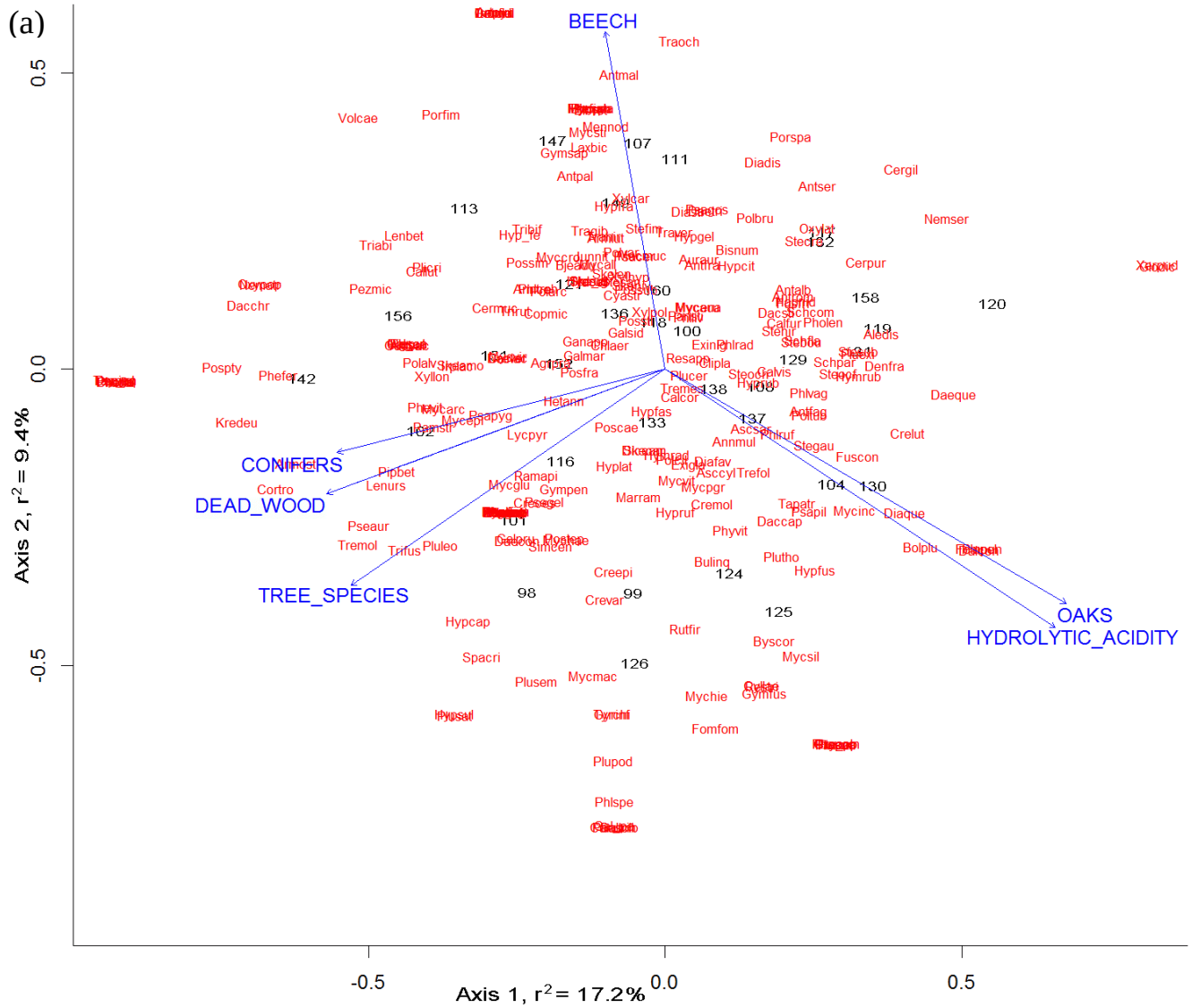
Phellinus viticola	(Schwein.) Donk	*Phevit*	wood-inh.	6	yes
Polyporus ciliatus	Fr.	*Polcil*	wood-inh.	6	yes
Trichaptum abietinum	(Dicks.) Ryvarden	*Triabi*	wood-inh.	6	yes
Xylaria longipes	Nitschke	*Xyllon*	wood-inh.	6	yes
Antrodiella pallescens	(Pilát) Niemelä & Miettinen	*Antpal*	wood-inh.	5	yes
Calocera cornea	(Batsch) Fr.	*Calcor*	wood-inh.	5	yes
Hapalopilus nidulans	(Fr.) P. Karst.	*Hapnid*	wood-inh.	5	yes
Mucidula mucida	(Schrad.) Pat.	*Mucmuc*	wood-inh.	5	yes
Phlebia radiata	Fr.	*Phlrad*	wood-inh.	5	yes
Piptoporus betulinus	(Bull.) P. Karst.	*Pipbet*	wood-inh.	5	yes
Pluteus semibulbosus	(Lasch) Quél.	*Plusem*	wood-inh.	5	yes
Postia caesia	(Schrad.) P. Karst.	*Poscae*	wood-inh.	5	yes
Resupinatus applicatus	(Batsch) Gray	*Resapp*	wood-inh.	5	yes
Trichaptum biforme	(Fr.) Ryvarden	*Tribif*	wood-inh.	5	yes
Annulohypoxylon multiforme	(Fr.) Y.-M. Ju, J.D. Rogers & H.-M. Hsieh	*Annmul*	wood-inh.	4	yes
Ascocoryne cylichnium	(Tul.) Korf	*Asccyl*	wood-inh.	4	yes
Ascocoryne sarcoides	(Jacq.) J.W. Groves & D.E. Wilson	*Ascsar*	wood-inh.	4	yes
Chlorociboria aeruginascens	(Nyl.) Kanouse ex C.S. Ramamurthi, Korf & L.R. Batra	*Chlaer*	wood-inh.	4	yes
Coprinellus micaceus	(Bull.) Vilgalys, Hopple & Jacq. Johnson	*Copmic*	wood-inh.	4	yes
Crepidotus mollis	(Schaeff.) Staude	*Cremol*	wood-inh.	4	yes
Dacrymyces capitatus	Schwein.	*Daccap*	wood-inh.	4	yes
Diatrypella favacea	(Fr.) Ces. & De Not.	*Diafav*	wood-inh.	4	yes
Gymnopilus sapineus	(Fr.) Murrill	*Gymsap*	wood-inh.	4	yes
Hypoxylon rubiginosum	(Pers.) Fr.	*Hyprub*	wood-inh.	4	yes
Hypocrea rufa	(Pers.) Fr.	*Hypruf*	wood-inh.	4	yes
Lentinellus ursinus	(Fr.) Kühner	*Lenurs*	wood-inh.	4	yes
Mensularia nodulosa	(Fr.) T. Wagner & M. Fisch.	*Mennod*	wood-inh.	4	yes
Nemania serpens	(Pers.) Gray	*Nemser*	wood-inh.	4	yes
Peziza micropus	Pers.	*Pezmic*	wood-inh.	4	yes
Polyporus brumalis	(Pers.) Fr.	*Polbru*	wood-inh.	4	yes
Porotheleum fimbriatum	(Pers.) Fr.	*Porfim*	wood-inh.	4	yes
Postia tephroleuca	(Fr.) Jülich	*Postep*	wood-inh.	4	yes
Rutstroemia firma	(Pers.) P. Karst.	*Rutfir*	wood-inh.	4	yes
Sparassis crispa	(Wulfen) Fr.	*Spacri*	wood-inh.	4	yes
Steccherinum bourdotii	Saliba & A. David	*Stebou*	wood-inh.	4	yes
Stereum gausapatum	(Fr.) Fr.	*Stegau*	wood-inh.	4	yes
Tremella foliacea	Pers.	*Trefol*	wood-inh.	4	yes
Tremella mesenterica	Retz.	*Tremes*	wood-inh.	4	yes
Agrocybe praecox	(Pers.) Fayod	*Agrpra*	wood-inh.	3	yes
Annulohypoxylon cohaerens	(Pers.) Y.-M. Ju, J.D. Rogers & H.-M. Hsieh	*Anncoh*	wood-inh.	3	yes
Antrodiella romellii	(Donk) Niemelä	*Antrom*	wood-inh.	3	yes
Bulgaria inquinans	(Pers.) Fr.	*Bulinq*	wood-inh.	3	yes
Ceriporia purpurea	(Fr.) Donk	*Cerpur*	wood-inh.	3	yes
Dacrymyces stillatus	Nees	*Dacsti*	wood-inh.	3	yes
Daedalea quercina	(L.) Pers.	*Daeque*	wood-inh.	3	yes
Fuscoporia ferruginosa	(Schrad.) Murrill	*Fusfrr*	wood-inh.	3	yes
Galerina sideroides	(Bull.) Kühner	*Galsid*	wood-inh.	3	yes
Hypoxylon fuscum	(Pers.) Fr.	*Hypfus*	wood-inh.	3	yes
Hypocrea gelatinosa	(Tode) Fr.	*Hypgel*	wood-inh.	3	yes
Lenzites betulina	(L.) Fr.	*Lenbet*	wood-inh.	3	yes
Mycena stipata	Maas Geest. & Schwöbel	*Mycsti*	wood-inh.	3	yes
Oxyporus latemarginatus	(Durieu & Mont.) Donk	*Oxylat*	wood-inh.	3	yes
Phlebia livida	(Pers.) Bres.	*Phlliv*	wood-inh.	3	yes
Phlebia tremellosa	(Schrad.) Nakasone &	*Phltre*	wood-inh.	3	yes

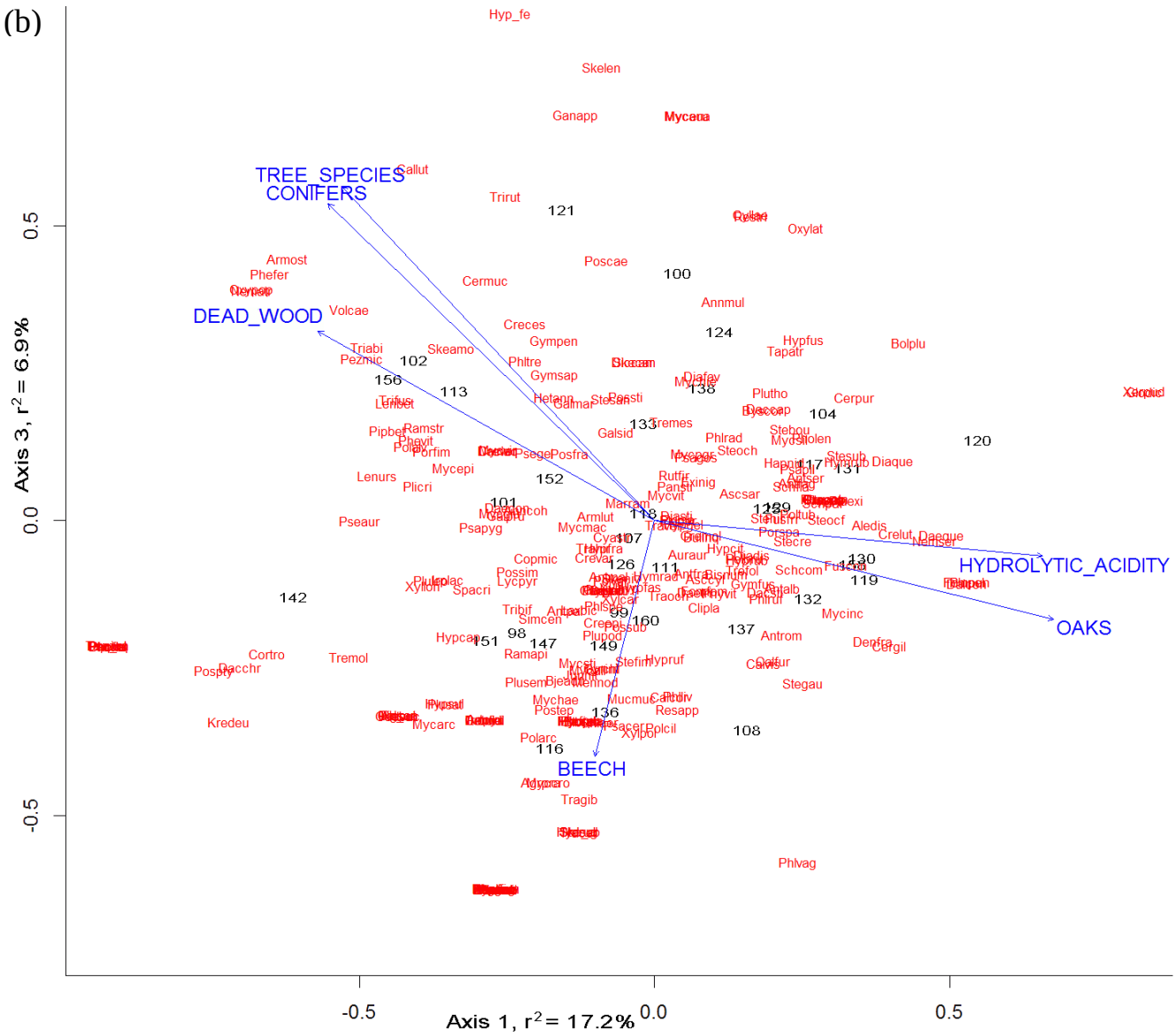
	Burds.				
Polyporus arcularius	(Batsch) Fr.	*Polarc*	wood-inh.	3	yes
Polyporus tuberaster	(Jacq. ex Pers.) Fr.	*Poltub*	wood-inh.	3	yes
Postia fragilis	(Fr.) Jülich	*Posfra*	wood-inh.	3	yes
Postia simanii	(Pilát ex Pilát) Jülich	*Possim*	wood-inh.	3	yes
Ramaria apiculata	(Fr.) Donk	*Ramapi*	wood-inh.	3	yes
Skeletocutis amorpha	(Fr.) Kotl. & Pouzar	*Skeamo*	wood-inh.	3	yes
Tricholomopsis rutilans	(Schaeff.) Singer	*Trirut*	wood-inh.	3	yes
Antrodiella serpula	(P. Karst.) Spirin & Niemelä	*Antser*	wood-inh.	2	yes
Callistosporium luteo-olivaceum	(Berk. & M.A. Curtis) Singer	*Callut*	wood-inh.	2	yes
Coriolopsis trogii	(Berk.) Domański	*Cortro*	wood-inh.	2	yes
Crepidotus epibryus	(Fr.) Quél.	*Creepi*	wood-inh.	2	yes
Crepidotus luteolus	Sacc.	*Crelut*	wood-inh.	2	yes
Dacrymyces chrysospermus	Berk. & M.A. Curtis	*Dacchr*	wood-inh.	2	yes
Daedaleopsis tricolor	(Bull.) Bond. & Sing.	*Daetri*	wood-inh.	2	yes
Dentipellis fragilis	(Pers.) Donk	*Denfra*	wood-inh.	2	yes
Diatrypella quercina	(Pers.) Cooke	*Diaque*	wood-inh.	2	yes
Fomes fomentarius	(L.) Fr.	*Fomfom*	wood-inh.	2	yes
Ganoderma applanatum	(Pers.) Pat.	*Ganapp*	wood-inh.	2	yes
Gymnopus fusipes	(Bull.) Gray	*Gymfus*	wood-inh.	2	yes
Hypholoma capnoides	(Fr.) P. Kumm.	*Hypcap*	wood-inh.	2	yes
Irpex lacteus	(Fr.) Fr.	*Irplac*	wood-inh.	2	yes
Kretzschmaria deusta	(Hoffm.) P.M.D. Martin	*Kredeu*	wood-inh.	2	yes
Mycena crocata	(Schrad.) P. Kumm.	*Myccro*	wood-inh.	2	yes
Mycena hiemalis	(Osbeck) Quél.	*Mychie*	wood-inh.	2	yes
Mycena silvae-nigrae	Maas Geest. & Schwöbel	*Mycsil*	wood-inh.	2	yes
Phellinidium ferrugineofuscum	(P. Karst.) Fiasson & Niemelä	*Phefer*	wood-inh.	2	yes
Phloeomana speirea	(Fr.) Redhead	*Phlspe*	wood-inh.	2	yes
Physisporinus vitreus	(Pers.) P. Karst.	*Phyvit*	wood-inh.	2	yes
Pluteus leoninus	(Schaeff.) P. Kumm.	*Pluleo*	wood-inh.	2	yes
Pluteus podospileus	Sacc. & Cub.	*Plupod*	wood-inh.	2	yes
Pluteus thomsonii	(Berk. & Broome) Dennis	*Plutho*	wood-inh.	2	yes
Porostereum spadiceum	(Pers.) Hjortstam & Ryvarden	*Porspa*	wood-inh.	2	yes
Postia ptychogaster	(F. Ludw.) Westerh.	*Pospty*	wood-inh.	2	yes
Psathyrella cernua	(Vahl) G. Hirsch	*Psacer*	wood-inh.	2	yes
Psathyrella gossypina	(Bull.) A. Pearson & Dennis	*Psagos*	wood-inh.	2	yes
Pseudomerulius aureus	(Fr.) Jülich	*Pseaur*	wood-inh.	2	yes
Skeletocutis lenis	(P. Karst.) Niemelä	*Skelen*	wood-inh.	2	yes
Steccherinum cremeoalbum	Hjortstam	*Stecre*	wood-inh.	2	yes
Tapinella atrotomentosa	(Batsch) Šutara	*Tapatr*	wood-inh.	2	yes
Trametes gibbosa	(Pers.) Fr.	*Tragib*	wood-inh.	2	yes
Trechispora mollusca	(Pers.) Liberta	*Tremol*	wood-inh.	2	yes
Trichaptum fuscoviolaceum	(Ehrenb.) Ryvarden	*Trifus*	wood-inh.	2	yes
Agrocybe firma	(Peck) Singer	*Agrfir*	wood-inh.	1	yes
Antrodia vaillantii	(DC.) Ryvarden	*Antvai*	wood-inh.	1	yes
Armillaria mellea	(Vahl) P. Kumm.	*Armmel*	wood-inh.	1	yes
Armillaria ostoyae	(Romagn.) Herink	*Armost*	wood-inh.	1	yes
Artomyces pyxidatus	(Pers.) Jülich	*Artpyx*	wood-inh.	1	yes
Ascotremella faginea	(Peck) Seaver	*Ascfag*	wood-inh.	1	yes
Bjerkandera fumosa	(Pers.) P. Karst.	*Bjefum*	wood-inh.	1	yes
Bolbitius reticulatus	(Pers.) Ricken	*Blbret*	wood-inh.	1	yes
Bolbitius pluteoides	M.M. Moser	*Bolplu*	wood-inh.	1	yes
Cantharellula umbonata	(J.F. Gmel.) Singer	*Canumb*	wood-inh.	1	yes
Ceriporiopsis gilvescens	(Bres.) Dom.	*Cergil*	wood-inh.	1	yes
Cerrena unicolor	(Bull.) Murrill	*Ceruni*	wood-inh.	1	yes
Crepidotus applanatus	(Pers.) P. Kumm.	*Creapp*	wood-inh.	1	yes
Crepidotus calolepis	(Fr.) P. Karst.	*Crecal*	wood-inh.	1	yes

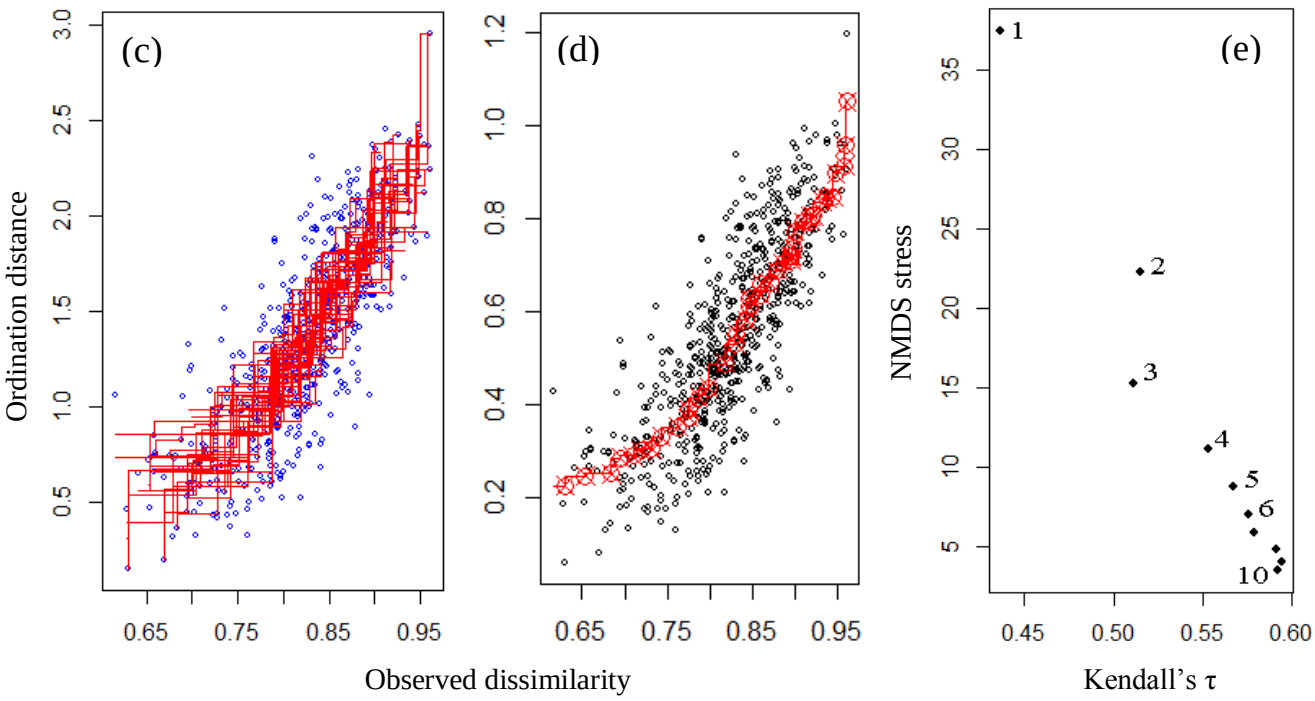
Crepidotus versutus	(Peck) Sacc.	*Crever*	wood-inh.	1	yes
Cylindrobasidium laeve	(Pers.) Chamuris	*Cyllae*	wood-inh.	1	yes
Dacrymyces lacrymalis	(Pers.) Sommerf.	*Daclac*	wood-inh.	1	yes
Daldinia concentrica	(Bolton) Ces. & De Not.	*Dalcon*	wood-inh.	1	yes
Datronia mollis	(Sommerf.) Donk	*Datmol*	wood-inh.	1	yes
Deconica inquilina	(Fr.) Romagn.	*Decinq*	wood-inh.	1	yes
Dichomitus campestris	(Quél.) Dom. & Orlicz	*Diccam*	wood-inh.	1	yes
Flammulaster carpophilus	(Fr.) Earle	*Flacar*	wood-inh.	1	yes
Flammulaster limulatus var. *lituus*	Vellinga	*Flalim*	wood-inh.	1	yes
Fomitiporia punctata	(P. Karst.) Murrill	*Fompun*	wood-inh.	1	yes
Fomitiporia robusta	(P. Karst.) Fiasson & Niemelä	*Fomrob*	wood-inh.	1	yes
Galerina camerina cf.	(Fr.) Kühner	*Gal_ca*	wood-inh.	1	yes
Galerina pallida cf.	(Pilát) E. Horak & M.M. Moser	*Gal_pa*	wood-inh.	1	yes
Galerina triscopa	(Fr.) Kühner	*Galtri*	wood-inh.	1	yes
Gloeoporus dichrous	(Fr.) Bres.	*Glodic*	wood-inh.	1	yes
Guepiniopsis buccina	(Pers.) L.L. Kenn.	*Guebuc*	wood-inh.	1	yes
Gyromitra infula	(Schaeff.) Quél.	*Gyrinf*	wood-inh.	1	yes
Hydropus subalpinus	(Höhn.) Singer	*Hydsub*	wood-inh.	1	yes
Hypoxylon ferrugineum cf.	G.H. Otth	*Hyp_fe*	wood-inh.	1	yes
Hypoxylon howeanum	Peck	*Hyphow*	wood-inh.	1	yes
Hypocrea sulphurea	(Schwein.) Sacc.	*Hypsul*	wood-inh.	1	yes
Inonotus nidus-pici	Pilát	*Inonid*	wood-inh.	1	yes
Lentinellus cochleatus	(Pers.) P. Karst.	*Lencoc*	wood-inh.	1	yes
Lentinellus flabelliformis	(Bolton) S. Ito	*Lenfla*	wood-inh.	1	yes
Mycoacia aurea	(Fr.) J. Erikss. & Ryvarden	*Mycaua*	wood-inh.	1	yes
Mycena erubescens	Höhn.	*Myceru*	wood-inh.	1	yes
Mycoacia uda	(Fr.) Donk	*Mycuda*	wood-inh.	1	yes
Mycena viridimarginata	P. Karst.	*Mycvir*	wood-inh.	1	yes
Nemania atropurpurea	(Fr.) Pouzar	*Nematr*	wood-inh.	1	yes
Oxyporus obducens cf.	(Pers.) Donk	*Oxy_ob*	wood-inh.	1	yes
Oxyporus populinus	(Schumach.) Donk	*Oxypop*	wood-inh.	1	yes
Phaeomarasmius erinaceus	(Fr.) Scherff. ex Romagn.	*Phaeri*	wood-inh.	1	yes
Phaeolus schweinitzii	(Fr.) Pat.	*Phasch*	wood-inh.	1	yes
Phellinus pomaceus	(Pers.) Maire	*Phepom*	wood-inh.	1	yes
Phellinus tremulae	(Bondartsev) Bondartsev & P.N. Borisov	*Phetre*	wood-inh.	1	yes
Phlebiella vaga	(Fr.) P. Karst.	*Phlvag*	wood-inh.	1	yes
Pholiota flammans	(Batsch) P. Kumm.	*Phofla*	wood-inh.	1	yes
Pholiota gummosa	(Lasch) Singer	*Phogum*	wood-inh.	1	yes
Pholiota jahnii	Tjall.-Beuk. & Bas	*Phojah*	wood-inh.	1	yes
Pholiota spumosa	(Fr.) Singer	*Phospu*	wood-inh.	1	yes
Pleurotus pulmonarius	(Fr.) Quél.	*Plepul*	wood-inh.	1	yes
Pluteus exiguus	(Pat.) Sacc.	*Pluexi*	wood-inh.	1	yes
Pluteus nanus	(Pers.) P. Kumm.	*Plunan*	wood-inh.	1	yes
Pluteus pellitus	(Pers.) P. Kumm.	*Plupel*	wood-inh.	1	yes
Pluteus romellii	(Britzelm.) Sacc.	*Plurom*	wood-inh.	1	yes
Pluteus salicinus	(Pers.) P. Kumm.	*Plusal*	wood-inh.	1	yes
Pluteus satur	Kühner & Romagn.	*Plusat*	wood-inh.	1	yes
Psathyrella olympiana cf.	A.H. Sm.	*Psa_ol*	wood-inh.	1	yes
Resupinatus trichotis	(Pers.) Singer	*Restri*	wood-inh.	1	yes
Rigidoporus sanguinolentus	(Alb. & Schwein.) Donk	*Rigsan*	wood-inh.	1	yes
Skeletocutis alutacea cf.	(J. Lowe) Jean Keller	*Ske_al*	wood-inh.	1	yes
Skeletocutis carneogrisea	A. David	*Skecar*	wood-inh.	1	yes
Stereum rugosum	Pers.	*Sterug*	wood-inh.	1	yes
Tapinella panuoides	(Fr.) E.-J. Gilbert	*Tappan*	wood-inh.	1	yes
Trametopsis cervina	(Schwein.) Tomšovský	*Tracer*	wood-inh.	1	yes
Trametes ochracea	(Pers.) Gilb. & Ryvarden	*Traoch*	wood-inh.	1	yes
Trametes suaveolens	(L.) Fr.	*Trasua*	wood-inh.	1	yes
Tyromyces chioneus	(Fr.) P. Karst.	*Tyrchi*	wood-inh.	1	yes

Volvariella caesiotincta	P.D. Orton	*Volcae*	wood-inh.	1	yes
Xerula pudens	(Pers.) Singer	*Xerpud*	wood-inh.	1	yes
Rickenella fibula	(Bull.) Raithelh.	–	bryophilous	8	no, skipped
Rickenella swartzii	(Fr.) Kuyper	–	bryophilous	4	no, skipped
Cordyceps larvicola	Quél.	–	entomopath.	1	no, skipped
Marasmius rotula	(Scop.) Fr.	–	lign./t. sapr.	6	no, skipped
Marasmius torquescens	Quél.	–	lign./t. sapr.	5	no, skipped
Mycena leptocephala	(Pers.) Gillet	–	lign./t. sapr.	4	no, skipped
Psathyrella lutensis	(Romagn.) Bon	–	lign./t. sapr.	3	no, skipped
Pholiota scamba	(Fr.) M.M. Moser	–	lign./t. sapr.	1	no, skipped
Psathyrella cortinarioides	P.D. Orton	–	lign./t. sapr.	1	no, skipped
Psathyrella fagetophila	Örstadius & Enderle	–	lign./t. sapr.	1	no, skipped
Psathyrella microrrhiza	(Lasch) Konrad & Maubl.	–	lign./t. sapr.	1	no, skipped
Psathyrella prona	(Fr.) Gillet	–	lign./t. sapr.	1	no, skipped
Psathyrella spadiceogrisea	(Schaeff.) Maire	–	lign./t. sapr.	1	no, skipped
Asterophora lycoperdoides	(Bull.) Ditmar	–	mycotrophic	7	no, skipped
Tremella encephala	Pers.	–	mycotrophic	6	no, skipped
Elaphocordyceps ophioglossoides	(Ehrh.) G.H. Sung, J.M. Sung & Spatafora	–	mycotrophic	3	no, skipped
Tremella globispora	D.A. Reid	–	mycotrophic	1	no, skipped
Entoloma rhodopolium	(Fr.) P. Kumm.	–	t. sapr./myc.	15	no, skipped
Otidea onotica	(Pers.) Fuckel	–	t. sapr./myc.	11	no, skipped
Otidea alutacea	(Pers.) Massee	–	t. sapr./myc.	5	no, skipped
Entoloma politum	(Pers.) Donk	–	t. sapr./myc.	4	no, skipped
Otidea bufonia	(Pers.) Boud.	–	t. sapr./myc.	4	no, skipped
Otidea fuckelii	M. Carbone & Van Vooren	–	t. sapr./myc.	1	no, skipped
Otidea grandis	(Pers.) Arnould	–	t. sapr./myc.	1	no, skipped
Otidea propinquata cf.	(P. Karst.) Harmaja	–	t. sapr./myc.	1	no, skipped

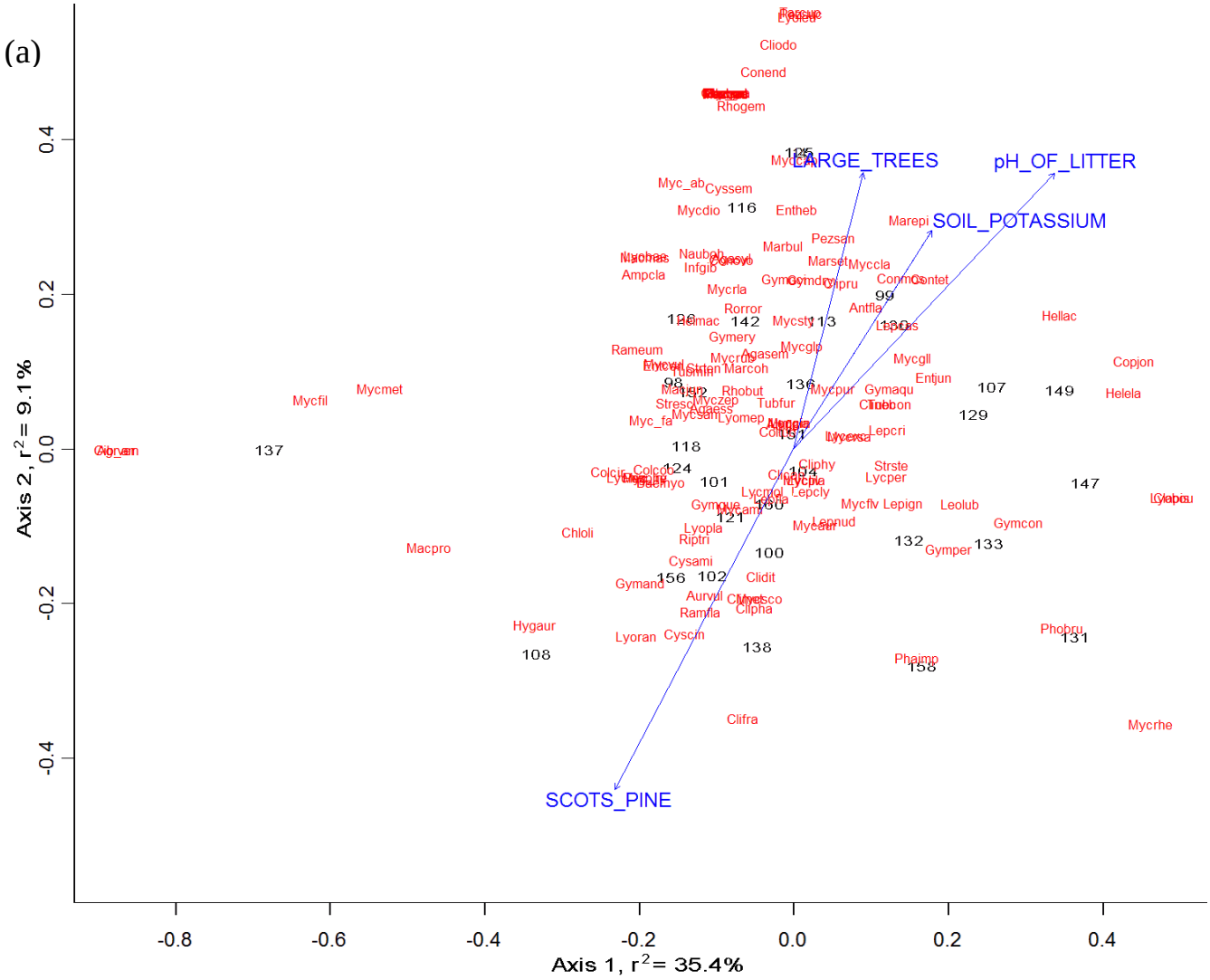
Supplementary Fig 1. NMDS results of wood-inhabiting macrofungi representing the optimal positions of all the 245 collected species (red letters; see Supplementary Table 1 for abbreviations), the significantly ($p < 0.05$; based on 999 replications) fitted environmental variables (blue capitals), and a tri-plot of 35 (all) sampling units (black numbers). By seeking a stable NMDS result with a low final stress, a 3-D solution was chosen: in Fig (**a**) axis 1 of the ordination is plotted against axis 2; while in Fig (**b**) the first and the third axes of the same run are shown. As a determination of goodness of fit, a Shepard diagram is drawn in Fig (**c**) where ordination distances are plotted against the observed dissimilarities and 20 fits are shown as monotonic step lines representing each run after which the best NMDS solution was reached (non-metric fit: $r^2 = 0.977$; linear fit: $r^2 = 0.748$). Random starting configurations were used for finding the best solution. Fig (**d**) displays the same diagram, but with the best-fit monotonic regression of distances. The red line denotes hypothetical distances that would be in the perfect rank-order with the dissimilarities (scatter about this line defines the NMDS stress). Fig (**e**) was used to select the optimal dimensionality where the Kendall's rank correlation coefficients (τ), indicating that how good the original distance matrix was recovered by the ordination distances, were plotted against the final NMDS stress values. Ten dimensions (black numbers) were tested in total. To the 3-D solution chosen, $\tau = 0.5114$ was related. The 4-D final solution with a better τ and lower stress was avoided because of the decreased interpretability of the results. Distance method applied: Bray–Curtis; final stress: 15.282 using Kruskal's stress formula 1 multiplied by 100 (Kruskal, 1964).

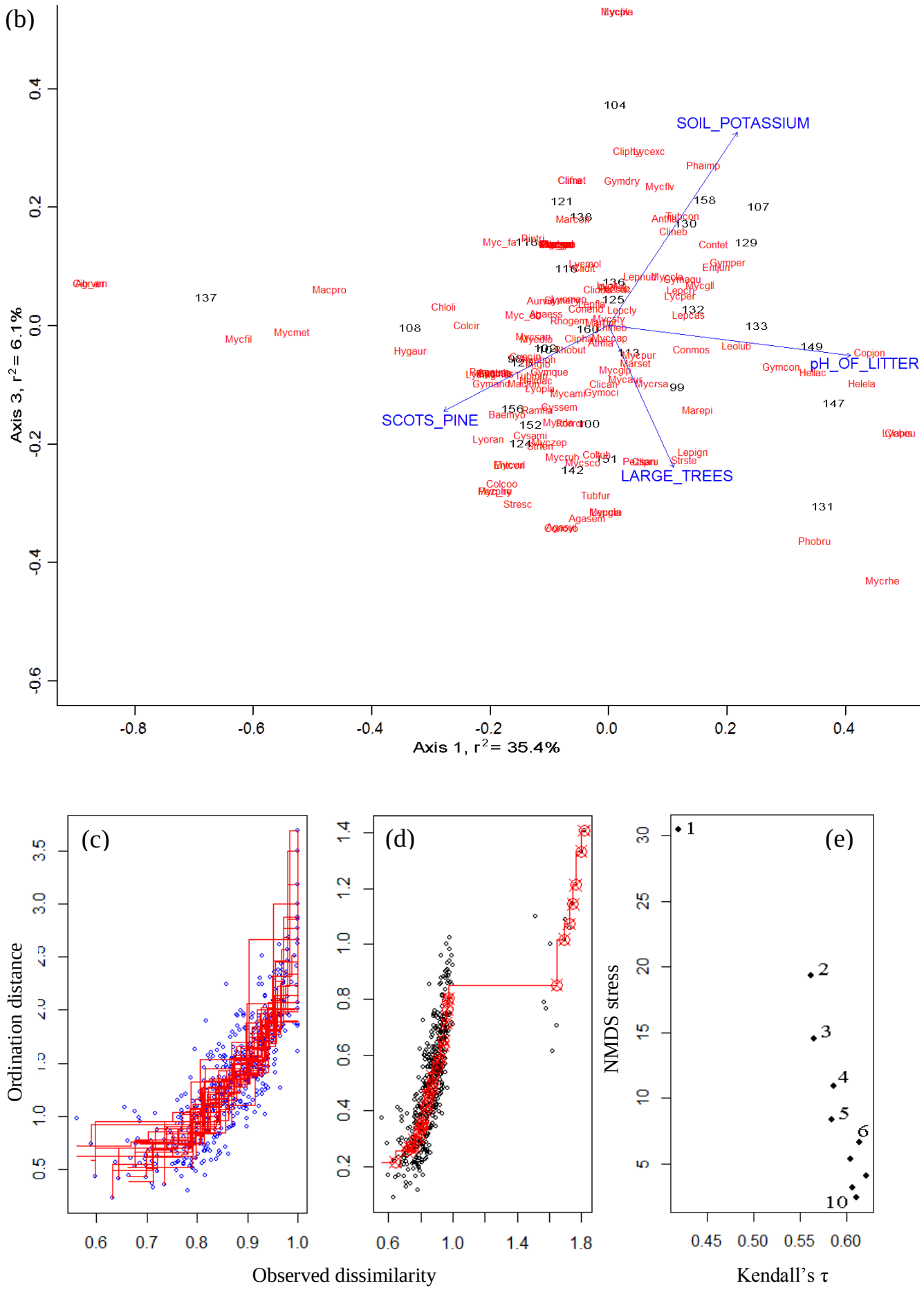



(b)
TREE_SPECIES
CONIFERS
DEAD_WOOD
HYDROLYTIC_ACIDITY
OAKS
BEECH
Axis 3, r² = 6.9%
Axis 1, r² = 17.2%
0.5
0.0
-0.5

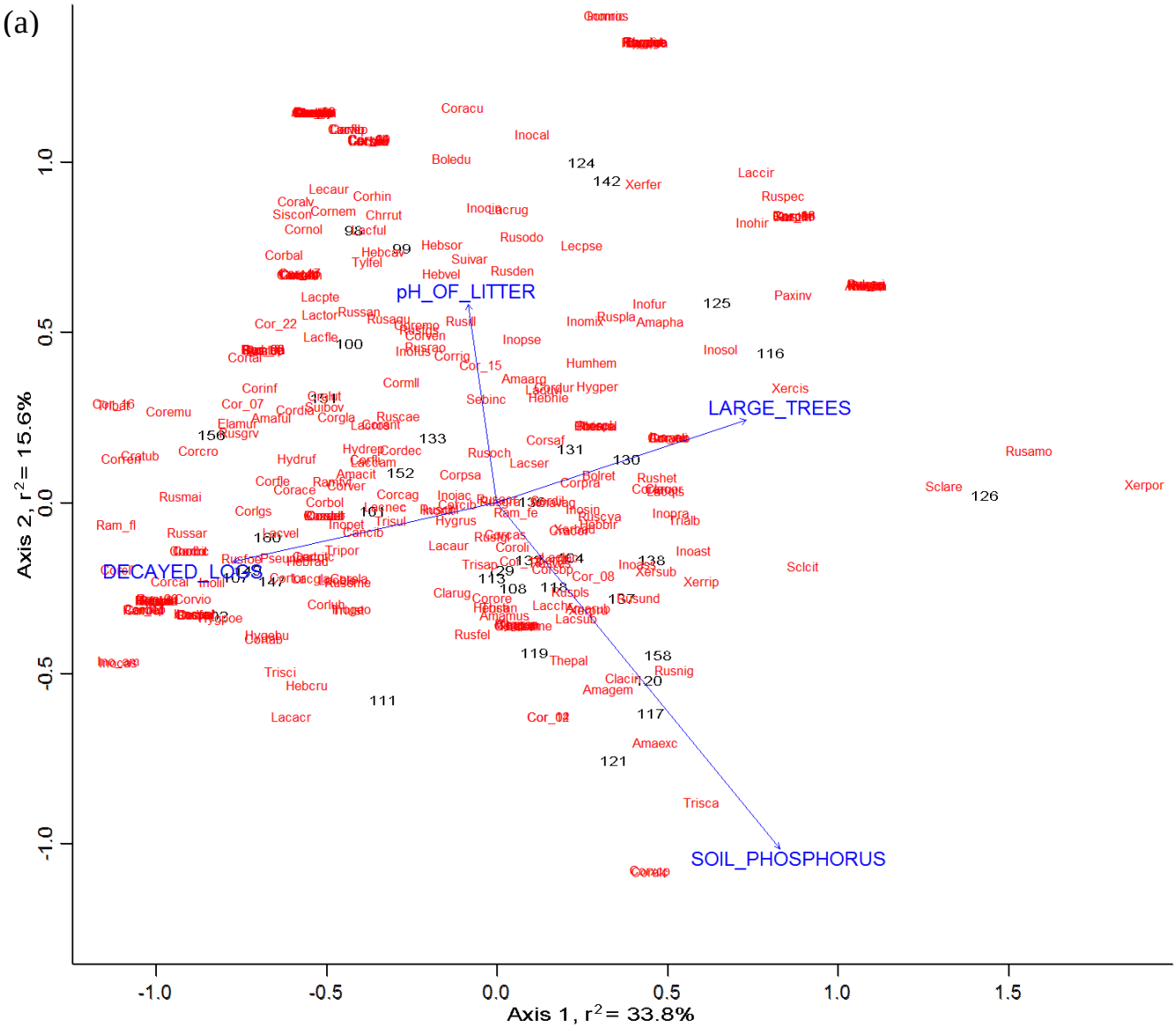

(c)
(d)
(e)
Ordination distance
Observed dissimilarity
NMDS stress
Kendall's τ

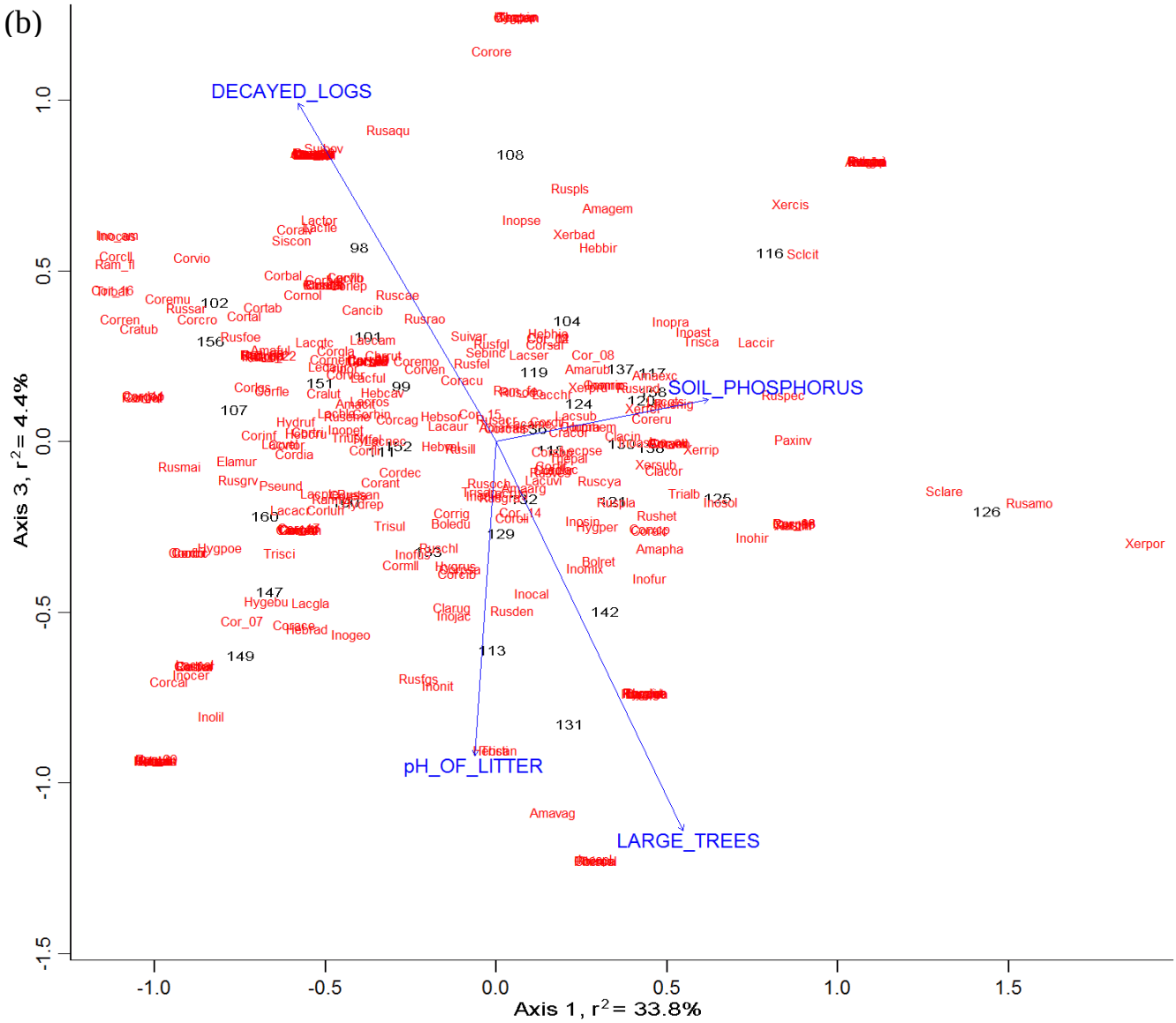
Supplementary Fig 2. NMDS results of terricolous saprotrophic macrofungi showing the optimal positions of 126 collected species (red letters; labels are coded as per Supplementary Table 1), the significantly ($p < 0.05$) fitted environmental variables (based on 999 replications, blue capitals), and a tri-plot of 31 sampling units (black numbers). Four sampling units (namely 111, 117, 119 and 120) with zero or very low species counts were excluded as these can have a disproportionate effect on the results. For finding a stable NMDS result with a low final stress, a 3-D solution was carried out: in Fig (**a**) the first two NMDS axes; while in Fig (**b**) the first and the third axis of the same run are plotted. In Fig (**c**), goodness of fit was mapped by the Shepard diagram of the NMDS result where ordination distances are plotted against the observed dissimilarities and 20 fits are shown as monotonic step lines representing each run after which the best NMDS solution was reached (non-metric fit: $r^2 = 0.979$; linear fit: $r^2 = 0.82$). Random starting configurations were used for finding the best solution. Fig (**d**) displays the same diagram, but with the best-fit monotonic regression of distances. The red line denotes hypothetical distances that would be in the perfect rank-order with the dissimilarities (scatter about this line defines the NMDS stress). A large step is demonstrated in the diagram because despite the fact that four sites had already been exluded, some sites (especially plot 137) did have a number of no shared or relatively rare species. To handle these tied dissimilarity values adequately, the function "step-across dissimilarities" were used in the package "vegan" for improving the NMDS results. Fig (**e**) was used to select the optimal dimensionality where the Kendall's rank correlation coefficients (τ), indicating that how good the original distance matrix was recovered by the ordination distances, were plotted against the final NMDS stress values. Ten dimensions (black numbers) were tested in total. To the 3-D solution chosen, $\tau = 0.5864$ was related. Distance method applied: Bray–Curtis; final stress: 14.331 using Kruskal's stress formula 1 multiplied by 100 (Kruskal, 1964).

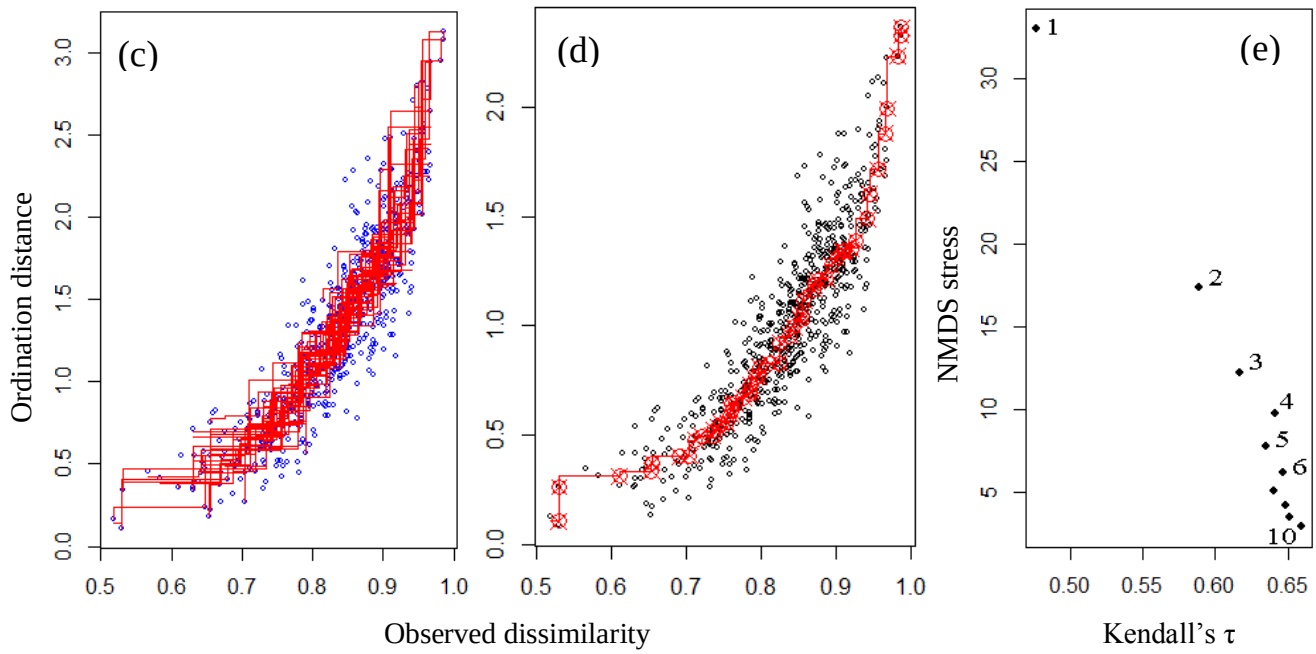



(b)
SOIL_POTASSIUM
pH_OF_LITTER
SCOTS_PINE
LARGE_TREES
Axis 1, r2= 35.4%
Axis 3, r2= 6.1%
(c)
Ordination distance
Observed dissimilarity
(d)
(e)
NMDS stress
Kendall's τ

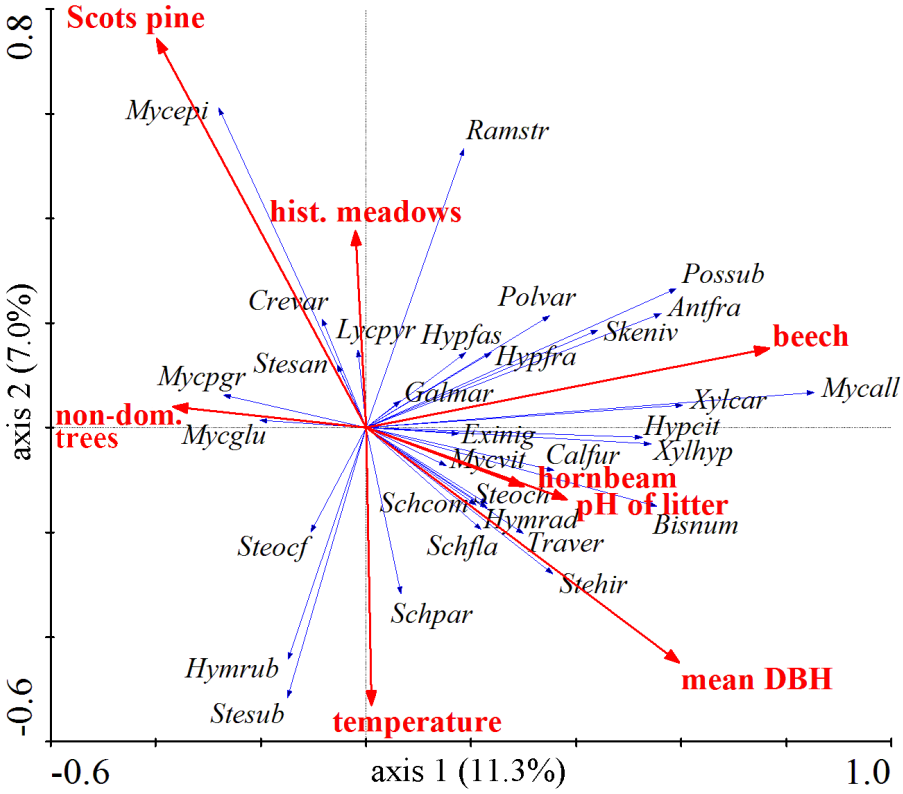
Supplementary Fig 3. NMDS results of ectomycorrhizal macrofungi showing the optimal positions of all the 290 collected species (red letters; legend in Supplementary Table 1), the significantly ($p < 0.05$) fitted environmental variables (based on 999 replications and depicted by blue arrows), and a tri-plot of 35 (all) sampling units (black numbers). NMDS reached a stable solution with the lowest final stress by adding a third dimension. In Fig (**a**), NMDS axis 1 against axis 2, while in Fig (**b**) axis 1 against axis 3 are plotted. For determining goodness of fit, a Shepard diagram was displayed in Fig (**c**) where ordination distances are plotted against the observed dissimilarities with 20 fits as monotonic step lines representing each run after which the best NMDS solution was reached (non-metric fit: $r^2 = 0.985$; linear fit: $r^2 = 0.846$). Random starting configurations were used for finding the best solution. Fig (**d**) reports the same diagram, but with the best-fit monotonic regression of distances. The red line denotes hypothetical distances that would be in the perfect rank-order with the dissimilarities (scatter about this line defines the NMDS stress). Fig (**e**) was used to select the optimal dimensionality where the Kendall's rank correlation coefficients (τ), indicating that how good the original distance matrix was recovered by the ordination distances, were plotted against the final NMDS stress values. Ten dimensions (black numbers) were tested in total. To the 3-D solution chosen, $\tau = 0.6165$ was related. Distance method used: Bray–Curtis; final stress: 12.186 applying Kruskal's stress formula 1 multiplied by 100 (Kruskal, 1964).




(b)
DECAYED_LOGS
SOIL_PHOSPHORUS
pH_OF_LITTER
LARGE_TREES
Axis 3, r2 = 4.4%
Axis 1, r2 = 33.8%


(c)
(d)
(e)
Ordination distance
Observed dissimilarity
NMDS stress
Kendall's τ

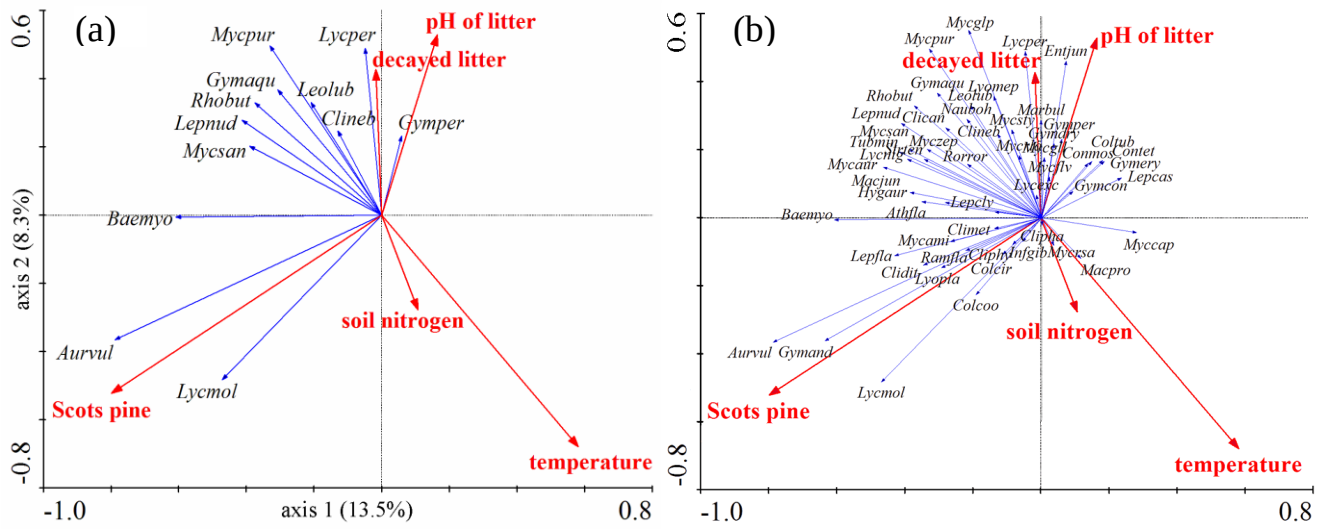
Supplementary Fig 4. Redundancy analysis (RDA) of wood-inhabiting fungi. In Fig 2a of the printed paper, the same taxa are plotted in the NMDS diagram. Applying RDA, the (rare) species collected in less than four sampling units were omitted (see Supplementary Table 1). All of the more frequent taxa (black italics) were accepted for building RDA axes. Compared to the NMDS results, RDA highlighted very similar environmental factors (red letters) to be important for the species composition of wood-inhabiting fungi. RDA resulted eight variables with significant ($p < 0.05$) effects showing the relative volumes of dominant tree species as of the greatest importance. Scots pine and beech (including hornbeam) revealed a clear deciduous–coniferous gradient. The canonical axes explained 37.4% of the total variance. The majority of plotted species had positive scores along axis 1 preferring high proportions of deciduous trees. Fungi species with high scores and strong relations to beech (deciduous stands) in RDA were *Antrodiella fragrans*, *Biscogniauxia nummularia*, *Mycetinis alliaceus*, *Postia subcaesia*, *Skeletocutis nivea*, *Xylaria carpophila* and *X. hypoxylon*. Wood-inhabiting taxa in warmer, deciduous stands (dominated by oak species based on the scatter of sites) were *Hymenochaete rubiginosa*, *Schizopora paradoxa* s.l., *Stereum ochraceoflavum* and *S. subtomentosum* (sampling units are not shown). In relatively cool, pine-dominated stands *Mycena epipterygia*, *Crepidotus variabilis* and *Stereum sanguinolentum* were common. Dead wood related variables had no significant effects, and air temperature was not significant in the NMDS model. Wood-inhabiting fungi was the only functional group that was related significantly to a variable belonging to historical forest management practices.



Significance of all canonical axes: $F = 2.279$, $p = 0.001$. Explained variances of axes as percentages are shown. Percentage variance explained by the variables within the RDA model are listed.

Environmental variable	Variance (%)	F-value	p-value
Beech (relative volume of beech)	9.6	3.86	0.001
Scots pine (relative volume of Scots pine)	6.3	2.68	0.001
Temperature (mean daily air temperature difference)	5.0	2.19	0.001
Hornbeam (relative volume of hornbeam)	3.8	1.71	0.004
Non-dom. trees (relative volume of non-dominant trees)	3.4	1.56	0.022
Mean DBH (mean Diameter at Breast Height of trees)	3.3	1.57	0.017
pH of litter	3.0	1.46	0.034
Hist. meadows (historical proportion of meadows)	3.0	1.45	0.041

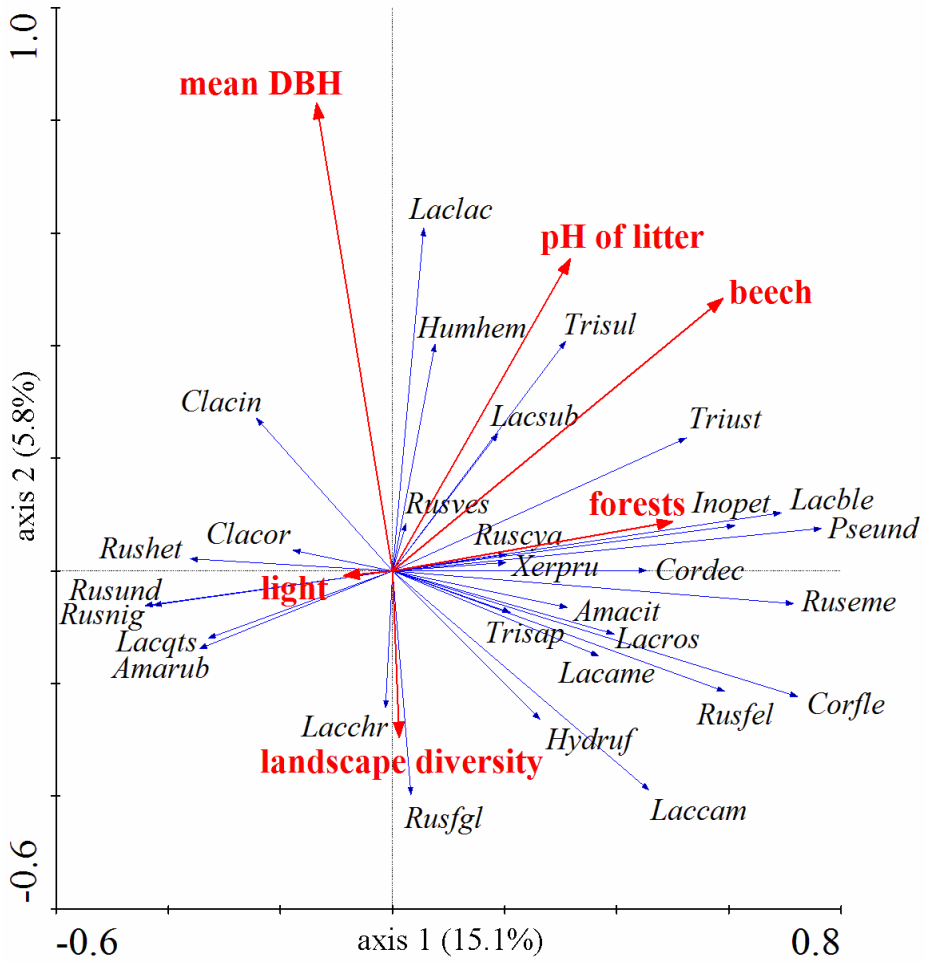
Supplementary Fig 5. Redundancy analysis (RDA) of terricolous saprotrophic fungi. In Fig 2b of the printed paper, the same taxa are plotted in the NMDS diagram. Applying RDA, the (rare) species collected in less than four sampling units were omitted (details in Supplementary Table 1). All the more frequent taxa were accepted for building RDA axes. All of the 35 sampling units were examined by RDA (sampling units are not shown), while NMDS was run by the omission of four sampling units with zero or very low counts of terricolous saprotrophic fungi. Broadly speaking, both methods revealed similar results: a definite litter pH gradient along the relative volume of Scots pine and the pH of litter (red letters). On the contrary, the effect of mean daily air temperature, however, was quite important in RDA, but it had no significant ($p < 0.05$) effect applying NMDS. In RDA, the canonical axes explained 31.6% of the total variance. Regarding the species, both methods highlighted *Auriscalpium vulgare* and *Baeospora myosura* to be common elements of pine-dominated stands. Using RDA, *Lycoperdon perlatum* and *Gymnopus peronatus* had relatively strong and positive relations to litter pH and the mass proportion of decayed litter, but the latter variable had no significant effect in the NMDS results (the position of *Lycoperdon perlatum* in the NMDS plot constructs an angle to litter pH very close to 90° showing no correlation with it). Supported by RDA only, stands with low air temperature and low soil N content were associated with the majority of frequent species: *Clitocybe nebularis*, *Gymnopus aquosus*, *Leotia lubrica*, *Lepista nuda*, *Mycena pura*, *M. sanguinolenta*, and *Rhodocollybia butyracea* (**a**). Moreover, the preponderance of all studied taxa preferred low air temperatures and low soil N contents (**b**).



Significance of all canonical axes: $F = 3.310$, $p = 0.001$. Explained variances of axes as percentages are shown. Percentage variance explained by the variables within the RDA model are listed.

Environmental variable	Variance (%)	F-value	p-value
Scots pine (relative volume of Scots pine)	11.7	5.11	0.001
Temperature (mean daily air temperature difference)	9.1	4.37	0.001
pH of litter	3.6	1.84	0.008
Decayed litter (mass proportion of decayed litter)	3.6	1.77	0.018
Soil nitrogen (nitrogen content of soil)	3.5	1.86	0.006

Supplementary Fig 6. Redundancy analysis (RDA) of EcM fungi. In Fig 2c of the printed paper, the same taxa are plotted by using NMDS. Applying RDA, the (rare) species collected in less than four sampling units were omitted (details in Supplementary Table 1), but all of the more frequent taxa (black italics) were accepted for building RDA axes. The canonical axes explained 34.3% of the total variance, and six environmental factors were significant with the strongest effects of the relative volume of beech and the mean DBH of trees. The determination of RDA axis 1 was threefold: the relative volume of beech and the proportion of forests in the landscape correlated positively with it, while the mean of relative diffuse light correlated negatively with axis 1. The mean DBH of trees and the pH of litter had positive effects, whereas the diversity of landscape elements had a negative effect along axis 2. Axis 1 explained ca. three times more variation compared to axis 2; and most of the plotted species had strong (positive) correlations with it. The species that preferred closed beech stands with more neutral litter pH and high proportion of forests in the landscape were *Inocybe petiginosa*, *Lactarius blennius*, *L. subdulcis*, *Pseudocraterellus undulatus*, *Tricholoma sulphureum* and *T. ustale*. The stands with high mean DBH of trees were favoured by *Clavulina cinerea*, *Humaria hemisphaerica* and *Laccaria laccata*, while *Russula fragilis* and *Lactarius chrysorrheus* preferred stands with a low tree DBH and a high landscape diversity. Characteristic EcM taxa in open stands were *Amanita rubescens*, *Lactarius quietus*, *Russula heterophylla*, *R. nigricans* and *R. undulata*.



Significance of all canonical axes: F = 2.652, p = 0.001. Explained variances of axes as percentages are shown. Percentage variance explained by the variables within the RDA model are listed.

Environmental variable	Variance (%)	F-value	p-value
Beech (relative volume of beech)	8.8	3.32	0.001
Mean DBH (mean Diameter at Breast Height of trees)	6.6	2.65	0.003
Forests (proportion of forests in the landscape)	5.6	2.33	0.007
Light (mean relative diffuse light)	4.9	2.24	0.005
pH of litter	4.9	2.13	0.005
Landscape diversity (Shannon diversity of landscape elements)	3.5	1.63	0.033