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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 Órség 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. Some evidence was given that macrofungi are related to drivers that are relatively easy to measure. It was found that wood-inhabiting fungal species composition is driven primarily by the species composition of living trees, while substrate 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. It was revealed that wood-inhabiting fungi are driven principally by the amount and diameter (Heilmann-Clausen and Christensen, 2004; Sippola et al., 2005; Ódor et al., 2006; Lonsdale et al., 2008), decay stage (Heilmann-Clausen and Christensen, 2003b; Siller, 2004; Heilmann-Clausen et al., 2014), age (Heilmann-Clausen, 2001), species identity (Sippola et al., 2005; Küffer et al., 2008), complexity (Heilmann-Clausen and Christensen, 2003a), and spatio-temporal availability (Siitonen, 2001; Bässler 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) had also significant effects. EcM community compositions were found to be structured strongly by the N content (Toljander et al., 2006; Cox et al., 2010; Suz et al., 2014), pH (Baar and 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 and 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 and Harrington, 2012), and temperature (McMullan-Fisher et al., 2009) were documented to be highly important.

Many influential environmental drivers have been revealed, but are there drivers with consistent importance on 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 and Parrent, 2007; Buntgen 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 actually limiting factors (resources or environmental conditions) 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 have 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 this complexity 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) the effects of substrate 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 Órség National Park (ÖNP), West Hungary (46° 51'–55' North, 16° 06'–24' East (Fig 1a). In the ÖNP, 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 ÖNP region, which has an area of ca. 350 km<sup>2</sup> (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). ÖNP 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 ÖNP is a suitable region for studying the effects of tree species on macrofungal communities.

Between the 12<sup>th</sup> and 19<sup>th</sup> centuries, the landscape was characterized by a rotation cycle in land use: small pieces 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 (*Pinus sylvestris* and *Betula 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 years 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 (Gyöngyössi, 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 of forest macrofungi that drive their species composition. In accordance, forest stands were selected by a stratified random sampling based on the Hungarian Forestry Database (Hungarian Central Agricultural Office, Forestry Directorate, [www.nebih.gov](http://www.nebih.gov)). The even-aged, 70–100 years old, spatially independent stands (the minimum distance is 500 m between them) chosen are 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 are scattered in a 160 km<sup>2</sup> area. In the middle of each plot, a 30 m × 30 m sampling unit was assigned for macrofungi surveys. Sampling units were divided into thirty-six 5 m × 5 m 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).

Tree species composition was expressed based on the relative volume of tree species by merging all taxa within the same genus, e.g. oaks (*Quercus 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 and Kolozs, 2000). Shannon diversity of tree species was calculated based on relative tree volumes and using natural logarithm (Shannon and 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 and 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<sup>2</sup>/ha. 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<sup>3</sup> Ca(CH<sub>3</sub>COO)<sub>2</sub> and 1 mol/dm<sup>3</sup> 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 hours by setting five minutes recording frequency. By repeating the same procedure, eight measurements were carried out in different months of the vegetation periods between 2009 and 2011, and their 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 years 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 and 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.

#### Fungi data

By reason of the large total area (31,500 m<sup>2</sup>) 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 August 2009, May 2010 and during September–November 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 days between 19 of September and 5 of November (early November is generally the very 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 of the 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, our 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](http://www.mycobank.org), assessed between 19 and 20 of April 2013) and more rarely Knudsen and Vesterholt (2012) were used to verify up-to-date scientific names and author(s) 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, stand-alone 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 are evaluated in this study 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) are often dropped from the analysis, as it was also done in this work to satisfy its requirements (Legendre and 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 and 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 and 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 were reached independently from the environmental data (Oksanen, 2013). In this study, NMDS was carried out following McCune & Grace (2002) and Oksanen (2013). Regarding each functional group, “local” NMDS model (Sibson, 1972) was fitted where an independent monotonic regression is used for each sampling unit in contrast to the “global” NMDS model (Kruskal, 1964), which is 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 data was revealed based on the Supplementary Fig 1–3e. The Kendall’s rank correlation coefficients ( $\tau$ ) were calculated between the original distance matrices and the ordination distances, and they were plotted against the final stress values

testing the dimensions between 1 and 10. 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 Fig 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 is biased by the third, 48-day long sporocarp survey during which the vast majority of records were obtained and the field visit of some sampling units were extended to the very end (or longer) of 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 have 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 and 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 and Šmilauer, 2002) was applied for RDAs.

## Results

### Fungal diversity

In this study, 687 macrofungal taxa were collected and identified (Supplementary Table 1). Taxa belonging to phylum Basidiomycota (631 species, 167 genera) were more species rich over ascomycetous taxa (56 species, 29 genera). A total of 13,396 records and 1,556 specimens were obtained. The vast majority of records (11,647 pieces, 87%) were collected during the third field survey in autumn 2010, whereas the total number of records was 1,313 (10%) in August 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).

### Tree species identity drove wood-inhabiting fungi

Thirty-two taxa (out of 245) were found in 14 or more sampling units. These taxa are plotted in Fig 2a (displaying the NMDS results) and in Supplementary Fig 4 (showing the RDA plot). 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% variation) showed a strong correlation with the relative volume of beech, while axis 3 (6.9% 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*. The species *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 fungi

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 demonstrates 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 available 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 dimensionality 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% 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 *Mycena 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. These species are displayed in the NMDS (Fig 2c) and the RDA (Supplementary Fig 6) diagrams. 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 substrate related factors (the relative volume of decayed logs, litter pH, and soil P content) 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 optima 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*, *Lactarius subdulcis*, and *Tricholoma 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*, *Lactarius quietus*, *Russula 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 oak-dominated stands were situated close to these taxa.

## 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 discussed in Table 4.

### Tree species composition

It was shown that the species composition of trees has the highest relevance on wood-inhabiting fungal species composition at a scale of forest stands (Fig 2a). Comparative studies in Europe have also identified tree species composition as having determinative effects on wood-inhabiting fungal species composition (e.g. Humphrey et al., 2000; Sippola et al., 2005; O’Hanlon and 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). Here, 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 substrate restricted, as they live within the wood, and species are often selective for certain tree taxa (Boddy and Heilmann-Clausen, 2008). It was also underlined by other studies that tree species identity has a marked impact on wood-inhabiting

fungus 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; Renvall, 1995; Boddy, 2001), 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 and Boddy, 2008).

In this work, terricolous saprotrophic fungus 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 and Harrington, 2012) or even a negative one (Ferris et al., 2000). Terricolous saprotrophic fungi are thought to be mainly a substrate restricted functional group (Gebauer and 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. S  stad, 1995; Ferris et al., 2000; Kernaghan et al., 2003; Morris et al., 2009) supported the idea that EcM species composition is determined principally by the species composition of their host trees at 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) to have determinative effects. The picture is not clear, because there is usually a striking contrast between the great diversity of EcM fungus communities and the relatively species-poor stands of host trees in temperate forests (Tedersoo et al., 2014). A large number of EcM fungus 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 and Taylor, 2003).

### Stand structure

In this 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). It 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 and Heilmann-Clausen, 2008). Comparative studies in Europe (e.g. Küffer et al., 2008; Abrego and 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 Fig 4 and 6) were found to be 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 make changes in sporocarp occurrences.

The relative volume of decayed logs was revealed to have a significant effect on EcM community composition (Fig 2c). It is proven that 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 latter 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 and ter Braak, 1996). It was shown in the present study that the underlying litter pH gradient with a determinative effect on terricolous saprotrophic community 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 ( $|r| = 0.4\text{--}0.5$ ) compared to



their relations to the ordination axes ( $|r| = 0.3\text{--}0.5$ ). However, similar relationships were detected by Baar and 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 was suggested to be important in the osmoregulation and sporocarp formation (Tyler, 1982). Soil P content was reported by Conn and 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. In Supplementary Fig 5b, the general negative impact of soil N content on terricolous saprotrophic fungi in the ÖNP is given. In other European countries, concordant (e.g. Buée et al., 2011) and contradictory (e.g. Tarvainen et al., 2003) results were also detected. 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 Fig 4 and 5). Regarding wood-inhabiting fungi, this result is in agreement with those of Boddy (1992, 2001) within the wood and 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 and McClaugherty (2014), the optimal temperature is vital for the right activity of cellulose- and ligninolytic enzymes of this functional group. In accordance, their optimal temperatures in the studied region may be in rather closed stands with no sun-exposed litter layer.

## 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 Fig 4 and 6). The negative effects of forest management on wood-inhabiting fungi is 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 and 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 years 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 substrate 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 substrate 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 a further insight into stand-scale drivers of fungal species composition, sporocarp surveys should be combined with DNA sequence based sampling methods from a below-ground study.

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Table 1. List of the potential environmental variables influencing the species composition of macrofungal communities.

Environmental variable	Unit	Mean (range)	Transformation
<b>TREE SPECIES COMPOSITION</b>			
Species richness of trees	number of species/1600 m <sup>2</sup>	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
<b>STAND STRUCTURE</b>			
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 <sup>2</sup> /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 <sup>3</sup> /ha	8.99 (0.90–65.02)	ln
Volume of logs (d > 10 cm)	m <sup>3</sup> /ha	10.51 (0.17–59.48)	ln
Total volume of logs and snags (d > 10 cm)	m <sup>3</sup> /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 <sup>2</sup> /ha	261.57 (79.44–729.99)	ln
Cover of understory vegetation	m <sup>2</sup> /ha	740.80 (19.19–4829.30)	ln
Cover of bryophytes	m <sup>2</sup> /ha	247.37 (16.57–2201.59)	ln
<b>SOIL AND LITTER</b>			
Cover of soil	m <sup>2</sup> /ha	146.75 (8.56–472.22)	–
Cover of litter	m <sup>2</sup> /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 <sup>2</sup>	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 <sub>2</sub> O <sub>5</sub> /100 g	4.29 (1.96–9.35)	–
Potassium (K) content of soil *	mg K <sub>2</sub> O/100 g	7.74 (4.00–13.10)	–
<b>MICROCLIMATE</b>			
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
<b>LANDSCAPE (radius = 300 m)</b>			
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)	–
<b>MANAGEMENT HISTORY</b>			
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

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	<i>Exidia nigricans</i> , <i>Schizopora flavipora</i> , <i>Sc. paradoxa</i> s.l., <i>Stereum hirsutum</i> , <i>St. ochraceoflavum</i>	<i>Auriscalpium vulgare</i> , <i>Gymnopus peronatus</i> , <i>Leotia lubrica</i> , <i>Lycoperdon perlatum</i> , <i>Mycena pura</i>	<i>Clavulina coralloides</i> , <i>Laccaria amethystina</i> , <i>L. laccata</i> , <i>Lactarius subdulcis</i> , <i>Russula cyanoxantha</i>	–	–
The richest genera (number of taxa)	<i>Mycena</i> (14), <i>Pluteus</i> (11), <i>Crepidotus</i> (8), <i>Postia</i> (7)	<i>Mycena</i> (25), <i>Clitocybe</i> (8), <i>Gymnopus</i> (8), <i>Lyophyllum</i> (6)	<i>Cortinarius</i> (100), <i>Russula</i> (44), <i>Inocybe</i> (28), <i>Lactarius</i> (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

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
<b>WOOD-INHABITING FUNGI</b>		
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
<b>TERRICOLOUS SAPROTROPHIC FUNGI</b>		
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
<b>EcM FUNGI</b>		
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

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			
<i>Hypoxylon fragiforme</i>	beech↑	beech↑	Kacprzyk et al., 2014
<i>Mycetinis alliaceus</i>	beech↑	beech↑	Heilmann-Clausen, 2005
<i>Polyporus varius</i>	beech↑	beech↑	Ciortan, 2009
<i>Skeletocutis nivea</i>	beech↑	beech↑	Fischer and Wagner, 1999
<i>Xylaria carpophila</i>	beech↑	beech (cupule litter)↑	Whalley, 1985
<i>Antrodiella fragrans</i>	beech↑	deciduous trees↑	Miettinen et al., 2006
<i>Postia subcaesia</i>	beech↑	deciduous trees↑	Siller, 2004; Szabó, 2012
<i>Biscogniauxia nummularia</i>	beech↑, litter pH↑, mean DBH↑	beech↑	Lakatos and Molnár, 2009
<i>Xylaria hypoxylon</i>	beech↑, litter pH↑, mean DBH↑	beech↑	Heilmann-Clausen, 2005
<i>Trametes versicolor</i>	beech↑, litter pH↑, mean DBH↑	deciduous trees↑, conifers↓	Ryvarden and Gilbertson, 1994
<i>Hymenochaete rubiginosa</i>	oaks↑, air temperature↑, hydrolytic acidity↑	oaks↑, beech↓, hornbeam↓	Papp, 2013
<i>Schizopora paradoxa</i> s.l.	oaks↑, air temperature↑, hydrolytic acidity↑	oaks↑, deciduous trees↑	Bernicchia et al., 2007, 2008
<i>Stereum subtomentosum</i>	oaks↑, air temperature↑, hydrolytic acidity↑	oaks↑, deciduous trees↑	Bernicchia et al., 2008
<i>Stereum ochraceoflavum</i>	oaks↑, hydrolytic acidity↑	oaks↑	Bernicchia and Gorjón, 2010
<i>Mycena epipterygia</i>	pine↑, dead wood↑, air temperature↓	conifers↑	Krieglsteiner, 2001
<i>Ramaria stricta</i>	pine↑, dead wood↑, tree species↑, air temperature↓	mixed (deciduous–coniferous) stands↑	Breitenbach and Kränzlin, 1986; Krieglsteiner, 2000
TERRICOLOUS SAPROTROPHIC FUNGI			
<i>Auriscalpium vulgare</i>	pine↑, litter pH↓, density of large trees↓	pine (cones)↑	Bernicchia et al., 2007
<i>Baeospora myosura</i>	pine↑, litter pH↓, density of large trees↓	conifers↑	Krieglsteiner, 2001
<i>Lycoperdon molle</i>	pine↑, litter pH↓, density of large trees↓	mixed stands↑, open areas↑, soil pH↑	Rimóczi et al., 2011
EcM FUNGI			
<i>Lactarius blennius</i>	beech↑, decayed logs↑	beech↑	Tyler, 1992; Galli, 2006; Lang et al., 2011
<i>Tricholoma ustale</i>	beech↑, decayed logs↑	beech↑, deciduous	Bohus, 1973; Buée et al.,

<i>Russula emetica</i>	beech↑, decayed logs↑	trees↑, soil pH↓	2011
<i>Pseudocraterellus undulatus</i>	beech↑, light↓	beech↑, soil pH↓ hornbeam↑, oaks↑, N deposition↑	Bohus, 1973 Tyler, 1992; Suz et al., 2014
<i>Inocybe petiginosa</i>	beech↑, light↓	oaks↑, soil pH↓	Szemere, 1955; Babos, 1989
<i>Lactarius subdulcis</i>	beech↑, soil P↑, litter pH↑	beech↑, soil pH↓	Galli, 2006; Buée et al., 2011
<i>Tricholoma sulphureum</i>	beech↑, soil P↓	beech↑, deciduous trees↑	Christensen and Heilmann-Clausen, 2013
<i>Russula undulata</i>	soil P↑, light↑, litter pH↓	oaks↑, hornbeam↑, soil pH↓	Bohus, 1973
<i>Lactarius quietus</i>	soil P↑, light↑, litter pH↓	oaks↑, soil N↑, soil pH↓	Galli, 2006; Suz et al., 2014
<i>Amanita rubescens</i>	soil P↑, light↑, litter pH↓	soil P, N↑, mixed (deciduous–coniferous) stands↑	Pál-Fám, 2001; Buée et al., 2011
<i>Russula nigricans</i>	soil P↑, light↑, litter pH↓	soil pH↓, mixed (deciduous–coniferous) stands↑	Bohus and Babos, 1967; Pál-Fám, 2001

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## Figures

Fig 1. Borders of West Hungary; Órség National Park (440 km<sup>2</sup>) is highlighted by 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

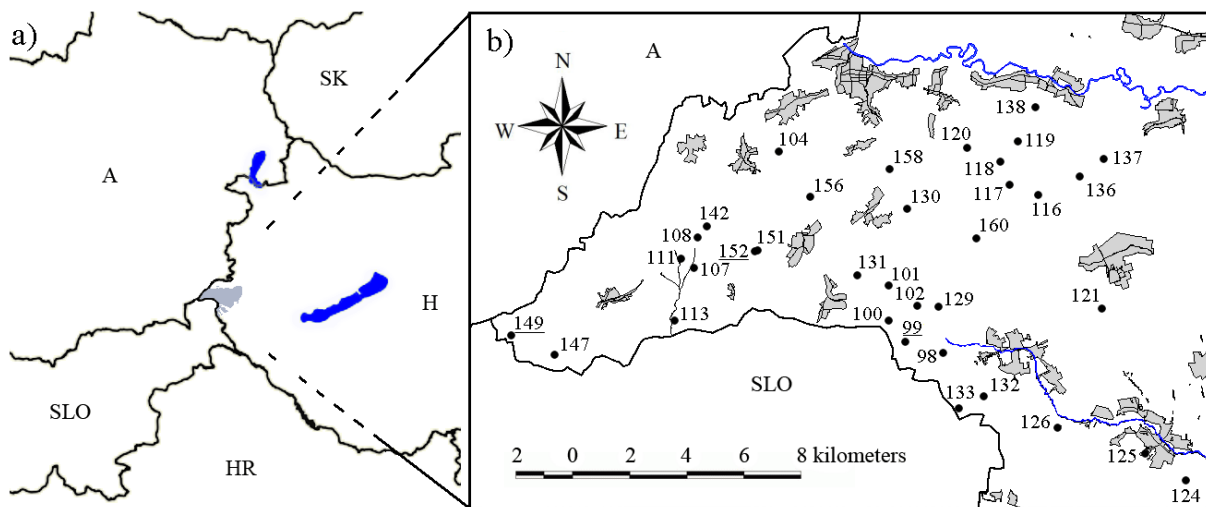
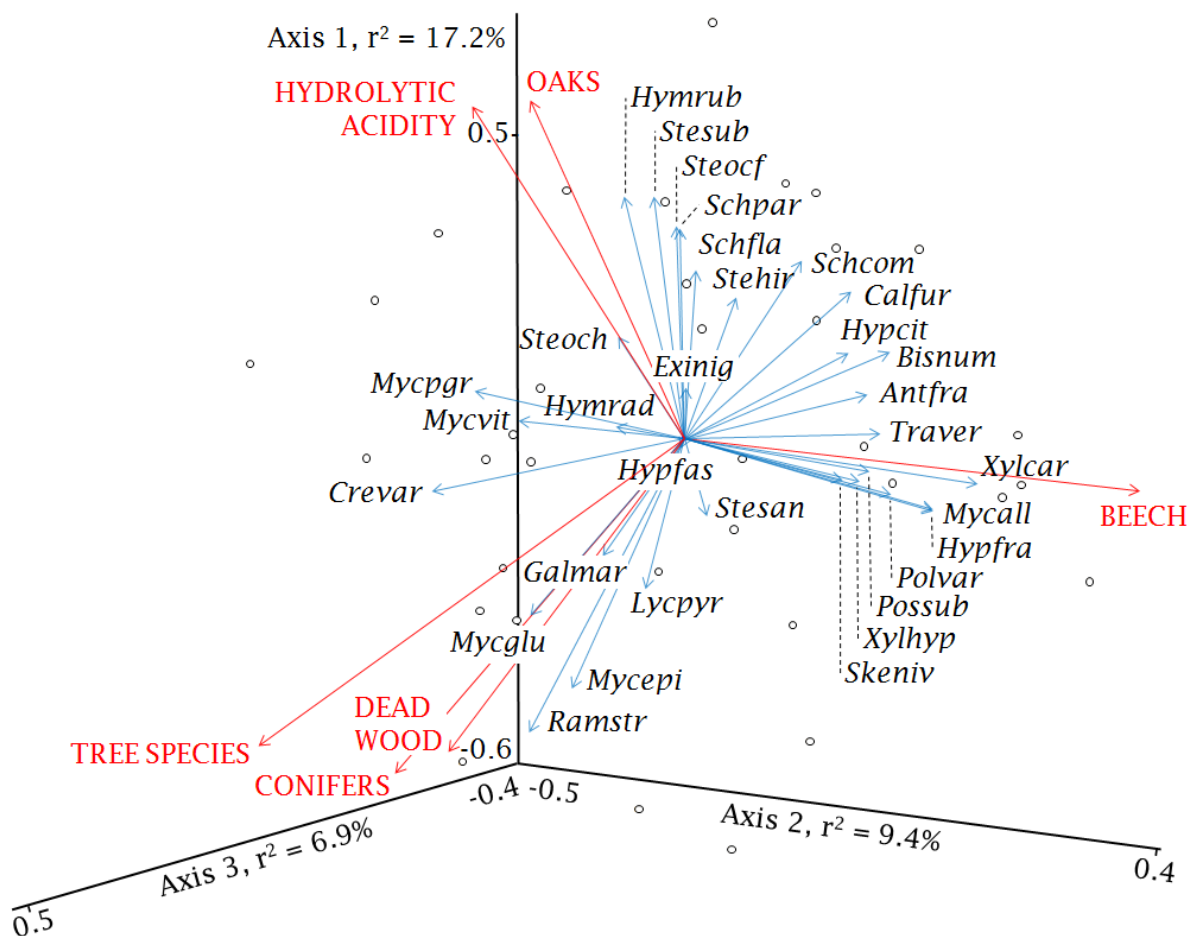
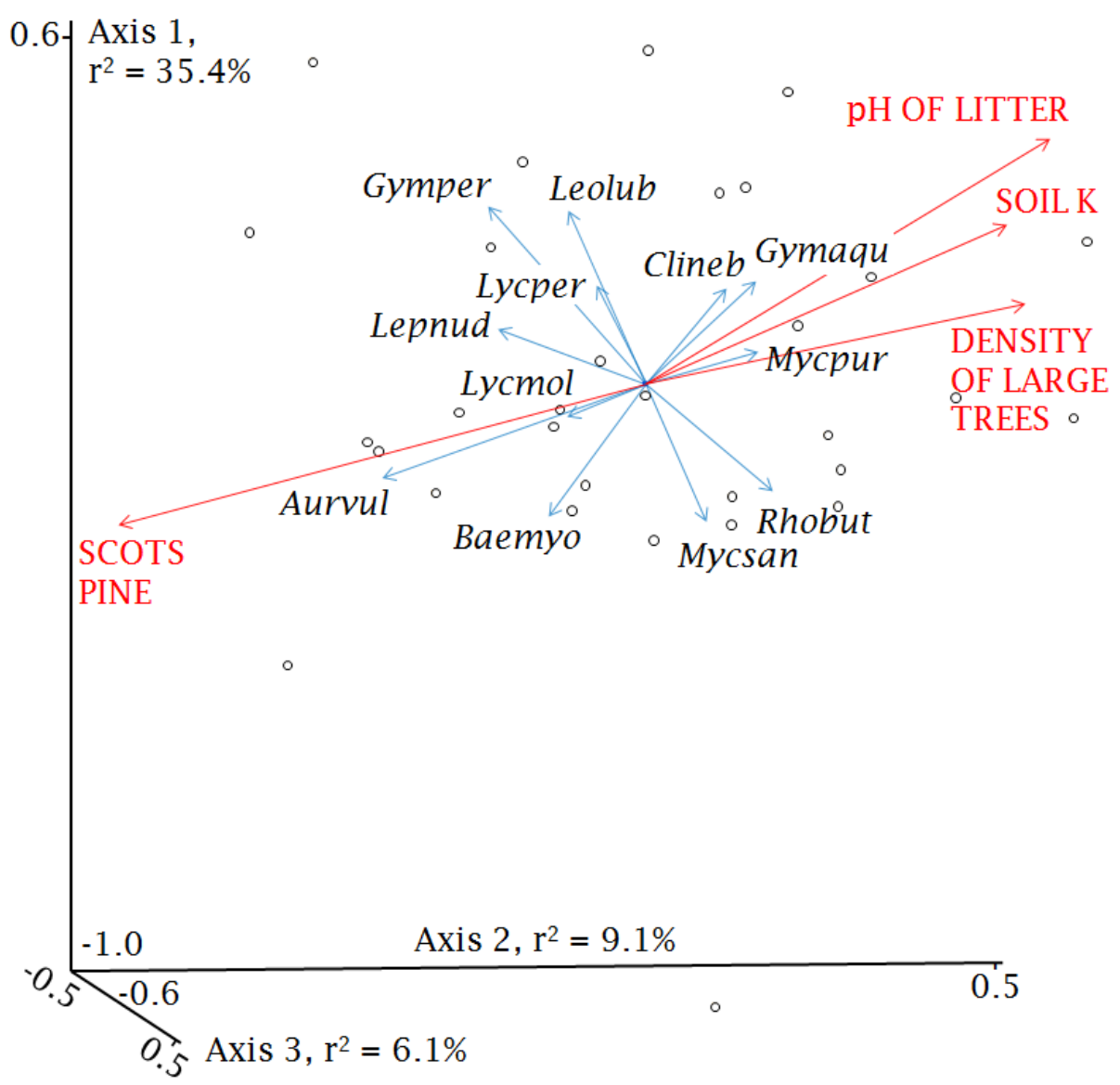
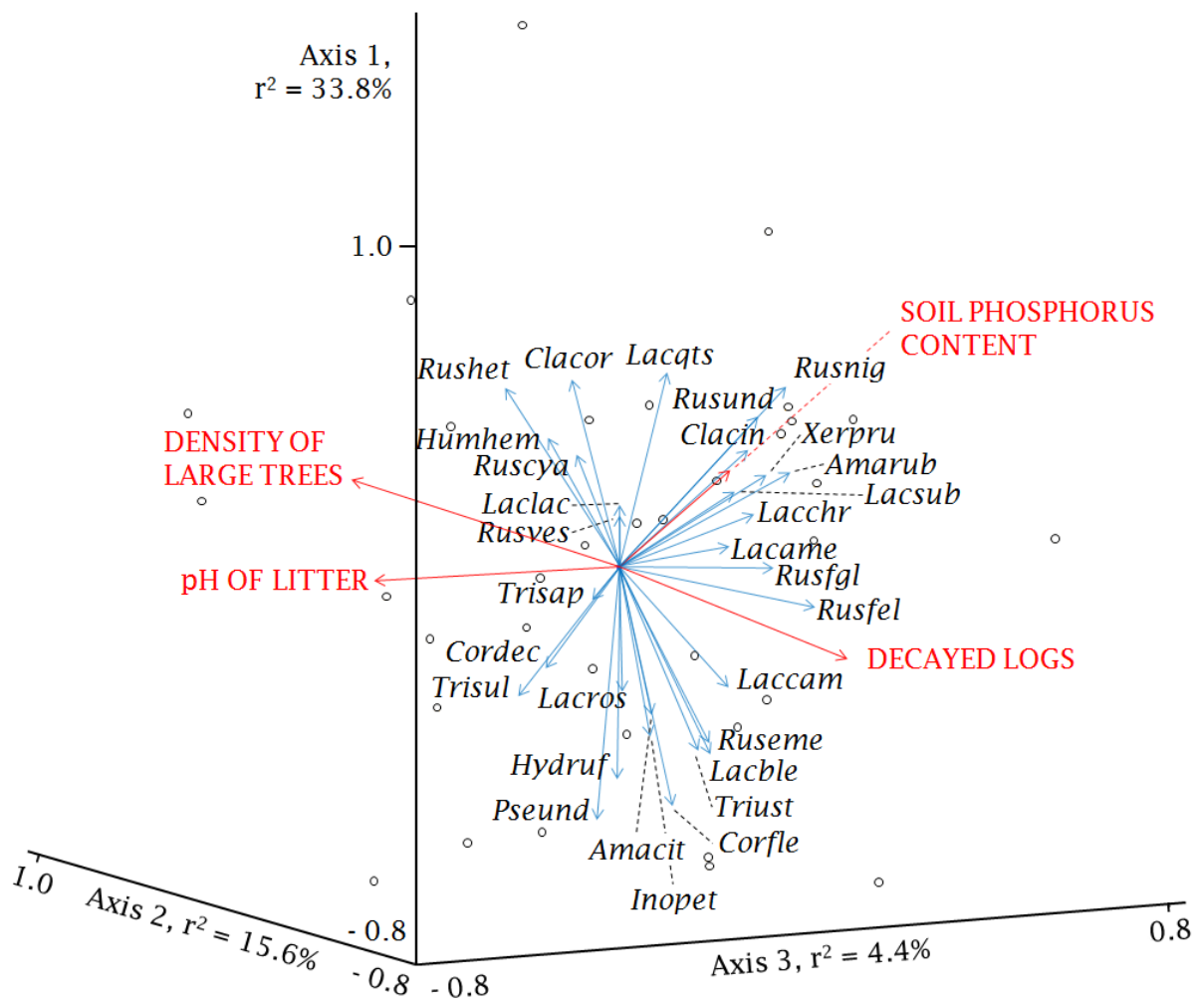


Fig 2. Local NMDS on wood-inhabiting (a), terricolous saprotrophic (b) and EcM (c) fungal species (black italics) representing the significantly ( $p < 0.05$ ) fitted environmental variables (red capitals) and a tri-plot of sampling units (black circles). The most frequent taxa are displayed; the optimal positions of all recorded species are shown in Supplementary Fig 1–3a 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 Fig 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.







## Supplementary Material

for the paper entitled “Drivers of macrofungal species composition in temperate forests, West Hungary: functional groups compared”.

### Contents

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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 Órség 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?
<i>Laccaria amethystina</i>	Cooke	<i>Lacame</i>	EcM	34	yes, plotted
<i>Russula cyanoxantha</i>	(Schaeff.) Fr.	<i>Ruscya</i>	EcM	34	yes, plotted
<i>Clavulina coralloides</i>	(L.) J. Schröt.	<i>Clacor</i>	EcM	28	yes, plotted
<i>Laccaria laccata</i>	(Scop.) Cooke	<i>Laclac</i>	EcM	27	yes, plotted
<i>Lactarius subdulcis</i>	(Pers.) Gray	<i>Lacsub</i>	EcM	27	yes, plotted
<i>Xerocomus pruinatus</i>	(Fr. & Hök) Quéf.	<i>Xerpru</i>	EcM	26	yes, plotted
<i>Lactarius blennius</i>	(Fr.) Fr.	<i>Lacble</i>	EcM	25	yes, plotted
<i>Cortinarius decipiens</i> s.l.	(Pers.) Fr.	<i>Cordec</i>	EcM	24	yes, plotted
<i>Russula emetica</i>	(Schaeff.) Pers.	<i>Ruseme</i>	EcM	24	yes, plotted
<i>Clavulina cinerea</i>	(Bull.) J. Schröt.	<i>Clacin</i>	EcM	23	yes, plotted
<i>Pseudocraterellus undulatus</i>	(Pers.) Rauschert	<i>Pseund</i>	EcM	23	yes, plotted
<i>Russula fellea</i>	(Fr.) Fr.	<i>Rusfel</i>	EcM	23	yes, plotted
<i>Inocybe petiginosa</i>	(Fr.) Gillet	<i>Inopet</i>	EcM	22	yes, plotted
<i>Tricholoma sulphureum</i>	(Bull.) P. Kumm.	<i>Trisul</i>	EcM	21	yes, plotted
<i>Russula fragilis</i>	Fr.	<i>Rusfgl</i>	EcM	20	yes, plotted
<i>Russula nigricans</i>	Fr.	<i>Rusnig</i>	EcM	20	yes, plotted
<i>Russula vesca</i>	Fr.	<i>Rusves</i>	EcM	20	yes, plotted
<i>Russula undulata</i>	Velen.	<i>Rusund</i>	EcM	19	yes, plotted
<i>Amanita rubescens</i>	Pers.	<i>Amarub</i>	EcM	18	yes, plotted
<i>Lactarius chrysorrhoeus</i>	Fr.	<i>Lacchr</i>	EcM	18	yes, plotted
<i>Lactarius camphoratus</i>	(Bull.) Fr.	<i>Laccam</i>	EcM	17	yes, plotted
<i>Lactarius quietus</i>	(Fr.) Fr.	<i>Lacqts</i>	EcM	17	yes, plotted
<i>Tricholoma saponaceum</i>	(Fr.) P. Kumm.	<i>Trisap</i>	EcM	17	yes, plotted
<i>Tricholoma ustale</i>	(Fr.) P. Kumm.	<i>Triust</i>	EcM	17	yes, plotted
<i>Humaria hemisphaerica</i>	(F.H. Wigg.) Fuckel	<i>Humhem</i>	EcM	15	yes, plotted
<i>Lactarius rostratus</i>	Heilm.-Claus.	<i>Lacros</i>	EcM	15	yes, plotted

<i>Amanita citrina</i>	(Schaeff.) Pers.	<i>Amacit</i>	EcM	14	yes, plotted
<i>Cortinarius flexipes</i> var. <i>flexipes</i>	(Pers.) Fr.	<i>Corfle</i>	EcM	14	yes, plotted
<i>Hydnum rufescens</i>	Pers.	<i>Hydruf</i>	EcM	14	yes, plotted
<i>Russula heterophylla</i>	(Fr.) Fr.	<i>Rushet</i>	EcM	14	yes, plotted
<i>Cantharellus cibarius</i>	Fr.	<i>Cancib</i>	EcM	13	yes
<i>Cortinarius</i> sp.15		<i>Cor_15</i>	EcM	13	yes
<i>Cortinarius casimiri</i>	(Velen.) Huijsman	<i>Corcas</i>	EcM	13	yes
<i>Cortinarius elatior</i>	Fr.	<i>Corela</i>	EcM	13	yes
<i>Lactarius vellereus</i>	(Fr.) Fr.	<i>Lacvel</i>	EcM	13	yes
<i>Russula acrifolia</i>	Romagn.	<i>Rusacr</i>	EcM	13	yes
<i>Russula grata</i>	Britzelm.	<i>Rusgra</i>	EcM	13	yes
<i>Clavulina rugosa</i>	(Bull.) J. Schröt.	<i>Clarug</i>	EcM	12	yes
<i>Hebeloma velutipes</i>	Bruchet	<i>Hebvel</i>	EcM	12	yes
<i>Russula ochroleuca</i>	Pers.	<i>Rusoch</i>	EcM	12	yes
<i>Cortinarius anthracinus</i>	(Fr.) Sacc.	<i>Corant</i>	EcM	11	yes
<i>Inocybe assimilata</i>	Britzelm.	<i>Inoass</i>	EcM	11	yes
<i>Lactarius serifluus</i>	(DC.) Fr.	<i>Lacser</i>	EcM	11	yes
<i>Inocybe geophylla</i>	(Fr.) P. Kumm.	<i>Inogeo</i>	EcM	10	yes
<i>Cortinarius cagei</i>	Melot	<i>Corcag</i>	EcM	9	yes
<i>Cortinarius flexipes</i> var. <i>flabellus</i>	(Fr.) H. Lindstr. & Melot	<i>Corflf</i>	EcM	9	yes
<i>Craterellus cornucopioides</i>	(L.) Pers.	<i>Cracor</i>	EcM	9	yes
<i>Hebeloma sordescens</i>	Vesterh.	<i>Hebsor</i>	EcM	9	yes
<i>Hydnum repandum</i>	L.	<i>Hydrep</i>	EcM	9	yes
<i>Lactarius quieticolor</i>	Romagn.	<i>Lacqtc</i>	EcM	9	yes
<i>Russula illota</i>	Romagn.	<i>Rusill</i>	EcM	9	yes
<i>Cortinarius trivialis</i> s.l.	J.E. Lange	<i>Cortri</i>	EcM	8	yes
<i>Lactarius aurantiacus</i>	(Pers.) Gray	<i>Lacaur</i>	EcM	8	yes
<i>Paxillus involutus</i>	(Batsch) Fr.	<i>Paxinv</i>	EcM	8	yes
<i>Russula sardonica</i>	Fr.	<i>Russar</i>	EcM	8	yes
<i>Tricholoma sciodes</i>	(Pers.) C. Martín	<i>Trisci</i>	EcM	8	yes
<i>Amanita phalloides</i>	(Fr.) Link	<i>Amapha</i>	EcM	7	yes
<i>Cortinarius diasemospermus</i> var. <i>diasemospermus</i>	Lamoure	<i>Cordia</i>	EcM	7	yes
<i>Cortinarius infractus</i> s.l.	(Pers.) Fr.	<i>Corinf</i>	EcM	7	yes
<i>Cortinarius rigidipes</i>	M.M. Moser	<i>Corrig</i>	EcM	7	yes
<i>Cortinarius torvus</i>	(Fr.) Fr.	<i>Cortor</i>	EcM	7	yes
<i>Lactarius acris</i>	(Bolton) Gray	<i>Lacacr</i>	EcM	7	yes
<i>Lactarius glaucescens</i>	Crossl.	<i>Lacgla</i>	EcM	7	yes
<i>Leccinum pseudoscabrum</i>	(Kallenb.) Šutara	<i>Lecpse</i>	EcM	7	yes
<i>Russula densifolia</i>	Secr. ex Gillet	<i>Rusden</i>	EcM	7	yes
<i>Scleroderma areolatum</i>	Ehrenb.	<i>Sclare</i>	EcM	7	yes
<i>Tricholoma album</i>	(Schaeff.) P. Kumm.	<i>Trialb</i>	EcM	7	yes
<i>Xerocomus badius</i>	(Fr.) E.-J. Gilbert	<i>Xerbad</i>	EcM	7	yes
<i>Amanita argentea</i>	Huijsm.	<i>Amaarg</i>	EcM	6	yes
<i>Amanita excelsa</i>	(Fr.) Bertill.	<i>Amaexc</i>	EcM	6	yes
<i>Cortinarius tabularis</i>	(Fr.) Fr.	<i>Cortab</i>	EcM	6	yes
<i>Craterellus lutescens</i>	(Pers.) Fr.	<i>Cralut</i>	EcM	6	yes
<i>Hygrophorus eburneus</i>	(Bull.) Fr.	<i>Hygebu</i>	EcM	6	yes
<i>Inocybe cincinnata</i>	(Fr.) Quéf.	<i>Inocin</i>	EcM	6	yes
<i>Russula mairei</i>	Singer	<i>Rusmai</i>	EcM	6	yes
<i>Xerocomus subtomentosus</i>	(L.) Quéf.	<i>Xersub</i>	EcM	6	yes
<i>Cortinarius subporphyropus</i>	Pilát	<i>Corsbp</i>	EcM	5	yes
<i>Cortinarius venetus</i>	(Fr.) Fr.	<i>Corven</i>	EcM	5	yes
<i>Hygrophorus poëtarum</i>	R. Heim	<i>Hygpoe</i>	EcM	5	yes
<i>Inocybe asterospora</i>	Quéf.	<i>Inoast</i>	EcM	5	yes
<i>Inocybe fuscidula</i>	Velen.	<i>Inofus</i>	EcM	5	yes
<i>Inocybe lilacina</i>	(Peck) Kauffman	<i>Inolil</i>	EcM	5	yes

<i>Lactarius fuliginosus</i>	(Fr.) Fr.	<i>Lacful</i>	EcM	5	yes
<i>Lactarius pterosporus</i>	Romagn.	<i>Lacpte</i>	EcM	5	yes
<i>Lactarius uvidus</i>	(Fr.) Fr.	<i>Lacuvi</i>	EcM	5	yes
<i>Leccinum aurantiacum</i>	(Bull.) Gray	<i>Lecaur</i>	EcM	5	yes
<i>Russula amoenolens</i>	Romagn.	<i>Rusamo</i>	EcM	5	yes
<i>Russula caerulea</i>	Fr.	<i>Ruscae</i>	EcM	5	yes
<i>Russula raoultii</i>	Quél.	<i>Rusrao</i>	EcM	5	yes
<i>Boletus edulis</i>	Bull.	<i>Boledu</i>	EcM	4	yes
<i>Cortinarius</i> sp.14		<i>Cor_14</i>	EcM	4	yes
<i>Cortinarius emunctus</i>	Fr.	<i>Coremu</i>	EcM	4	yes
<i>Cortinarius largus</i>	Fr.	<i>Corlgs</i>	EcM	4	yes
<i>Cortinarius psammocephalus</i>	(Bull.) Fr.	<i>Corpsa</i>	EcM	4	yes
<i>Elaphomyces muricatus</i>	Fr.	<i>Elamur</i>	EcM	4	yes
<i>Russula aquosa</i>	Leclair	<i>Rusaqu</i>	EcM	4	yes
<i>Tricholoma portentosum</i>	(Fr.) Quél.	<i>Tripor</i>	EcM	4	yes
<i>Tylopilus felleus</i>	(Bull.) P. Karst.	<i>Tylfel</i>	EcM	4	yes
<i>Amanita fulva</i>	(Fr.) Fr.	<i>Amaful</i>	EcM	3	yes
<i>Amanita gemmata</i>	(Fr.) Bertill.	<i>Amagem</i>	EcM	3	yes
<i>Chroogomphus rutilus</i>	(Schaeff.) O.K. Mill.	<i>Chrrut</i>	EcM	3	yes
<i>Cortinarius</i> sp.08		<i>Cor_08</i>	EcM	3	yes
<i>Cortinarius acetosus</i>	(Velen.) Melot	<i>Corace</i>	EcM	3	yes
<i>Cortinarius acutus</i> s.l.	(Pers.) Fr.	<i>Coracu</i>	EcM	3	yes
<i>Cortinarius emollitoides</i>	Bidaud, Moëgne-Locc. & Reumaux	<i>Coremo</i>	EcM	3	yes
<i>Cortinarius erubescens</i>	M.M. Moser	<i>Coreru</i>	EcM	3	yes
<i>Cortinarius hinnuleus</i> s.l.	Fr.	<i>Corhin</i>	EcM	3	yes
<i>Cortinarius luhmannii</i>	Münzmay, Saar & B. Oertel	<i>Corluh</i>	EcM	3	yes
<i>Cortinarius nolaneiformis</i>	(Velenovský) Dima, Niskanen & Liimat.	<i>Cornol</i>	EcM	3	yes
<i>Cortinarius olivaceofuscus</i>	Kühner	<i>Coroli</i>	EcM	3	yes
<i>Cortinarius talus</i>	Fr.	<i>Cortal</i>	EcM	3	yes
<i>Cortinarius violaceus</i>	(L.) Gray	<i>Corvio</i>	EcM	3	yes
<i>Craterellus tubaeformis</i>	(Fr.) Quél.	<i>Cratub</i>	EcM	3	yes
<i>Hebeloma cavipes</i>	Huijsman	<i>Hebcav</i>	EcM	3	yes
<i>Hebeloma crustuliniforme</i>	(Bull.) Quél.	<i>Hebcru</i>	EcM	3	yes
<i>Hygrophorus persoonii</i>	Arnolds	<i>Hygper</i>	EcM	3	yes
<i>Inocybe hirtella</i>	Bres.	<i>Inohir</i>	EcM	3	yes
<i>Inocybe praetervisa</i>	Quél.	<i>Inopra</i>	EcM	3	yes
<i>Inocybe sindonia</i>	(Fr.) P. Karst.	<i>Inosin</i>	EcM	3	yes
<i>Lactarius circellatus</i>	Fr.	<i>Laccir</i>	EcM	3	yes
<i>Lactarius necator</i>	(Bull.) Pers.	<i>Lacnec</i>	EcM	3	yes
<i>Russula chloroides</i>	(Krombh.) Bres.	<i>Ruschl</i>	EcM	3	yes
<i>Russula odorata</i>	Romagn.	<i>Rusodo</i>	EcM	3	yes
<i>Russula pectinatoides</i>	Peck	<i>Ruspec</i>	EcM	3	yes
<i>Russula puellula</i>	Ebbesen, F.H. Müller & Jul. Schäff.	<i>Ruspla</i>	EcM	3	yes
<i>Russula sanguinea</i>	(Bull.) Fr.	<i>Russan</i>	EcM	3	yes
<i>Suillus bovinus</i>	(L.) Roussel	<i>Suibov</i>	EcM	3	yes
<i>Thelephora palmata</i>	(Scop.) Fr.	<i>Thepal</i>	EcM	3	yes
<i>Amanita muscaria</i>	(L.) Lam.	<i>Amamus</i>	EcM	2	yes
<i>Amanita vaginata</i>	(Bull.) Lam.	<i>Amavag</i>	EcM	2	yes
<i>Boletus reticulatus</i>	Schaeff.	<i>Bolret</i>	EcM	2	yes
<i>Cortinarius</i> sp.07		<i>Cor_07</i>	EcM	2	yes
<i>Cortinarius</i> sp.22		<i>Cor_22</i>	EcM	2	yes
<i>Cortinarius albviolaceus</i>	(Pers.) Fr.	<i>Coralv</i>	EcM	2	yes
<i>Cortinarius balaustinus</i>	Fr.	<i>Corbal</i>	EcM	2	yes



<i>Cortinarius bolaris</i>	(Pers.) Fr.	<i>Corbol</i>	EcM	2	yes
<i>Cortinarius calochrous</i>	(Pers.) Gray	<i>Corcal</i>	EcM	2	yes
<i>Cortinarius cinnabarinus</i>	Fr.	<i>Corcib</i>	EcM	2	yes
<i>Cortinarius callisteus</i>	(Fr.) Fr.	<i>Corcll</i>	EcM	2	yes
<i>Cortinarius croceus</i>	(Schaeff.) Gray	<i>Corcro</i>	EcM	2	yes
<i>Cortinarius duracinus</i> s.l.	Fr.	<i>Cordur</i>	EcM	2	yes
<i>Cortinarius glaucopus</i>	(Schaeff.) Gray	<i>Corgla</i>	EcM	2	yes
<i>Cortinarius lepidopus</i>	Cooke	<i>Corlep</i>	EcM	2	yes
<i>Cortinarius melleopallens</i>	(Fr.) Britzelm.	<i>Cormll</i>	EcM	2	yes
<i>Cortinarius nemorensis</i> s. <i>Saar</i>	(Fr.) J.E. Lange	<i>Cornem</i>	EcM	2	yes
<i>Cortinarius orellanus</i>	Fr.	<i>Corore</i>	EcM	2	yes
<i>Cortinarius praestigiosus</i>	(Fr.) M.M. Moser	<i>Corpra</i>	EcM	2	yes
<i>Cortinarius renidens</i>	Fr.	<i>Corren</i>	EcM	2	yes
<i>Cortinarius safranopes</i>	Rob. Henry	<i>Corsaf</i>	EcM	2	yes
<i>Cortinarius veregregius</i>	Rob. Henry	<i>Corver</i>	EcM	2	yes
<i>Cortinarius vibratilis</i>	(Fr.) Fr.	<i>Corvib</i>	EcM	2	yes
<i>Hebeloma birrus</i>	(Fr.) Sacc.	<i>Hebbir</i>	EcM	2	yes
<i>Hebeloma hiemale</i>	Bres.	<i>Hebhie</i>	EcM	2	yes
<i>Hebeloma radicosum</i>	(Bull.) Ricken	<i>Hebrad</i>	EcM	2	yes
<i>Hygrophorus russula</i>	(Schaeff.) Kauffman	<i>Hygrus</i>	EcM	2	yes
<i>Inocybe calida</i>	Velen.	<i>Inocal</i>	EcM	2	yes
<i>Inocybe cervicolor</i>	(Pers.) Quél.	<i>Inocer</i>	EcM	2	yes
<i>Inocybe furfurea</i>	Kühner	<i>Inofur</i>	EcM	2	yes
<i>Inocybe jacobi</i>	Kühner	<i>Inojac</i>	EcM	2	yes
<i>Inocybe mixtilis</i>	(Britzelm.) Sacc.	<i>Inomix</i>	EcM	2	yes
<i>Inocybe nitidiuscula</i>	(Britzelm.) Lapl.	<i>Inonit</i>	EcM	2	yes
<i>Inocybe pseudoreducta</i>	Stangl & Glowinski	<i>Inopse</i>	EcM	2	yes
<i>Inocybe soluta</i>	Velen.	<i>Inosol</i>	EcM	2	yes
<i>Lactarius flexuosus</i>	(Pers.) Gray	<i>Lacfle</i>	EcM	2	yes
<i>Lactarius fluens</i>	Boud.	<i>Lacflu</i>	EcM	2	yes
<i>Lactarius ruginosus</i>	Romagn.	<i>Lacrug</i>	EcM	2	yes
<i>Lactarius torminosus</i>	(Schaeff.) Pers.	<i>Lactor</i>	EcM	2	yes
<i>Ramaria fennica</i> var. <i>fennica</i> cf.	(P. Karst.) Ricken	<i>Ram_fe</i>	EcM	2	yes
<i>Ramaria flavescens</i> cf.	(Schaeff.) R.H. Petersen	<i>Ram_fl</i>	EcM	2	yes
<i>Ramaria fennica</i> var. <i>fumigata</i>	(Peck) Schild	<i>Ramfvf</i>	EcM	2	yes
<i>Russula fragrantissima</i>	Romagn.	<i>Rusfgs</i>	EcM	2	yes
<i>Russula foetens</i>	Pers.	<i>Rusfoe</i>	EcM	2	yes
<i>Russula graveolens</i>	Romell	<i>Rusgrv</i>	EcM	2	yes
<i>Russula puellaris</i>	Fr.	<i>Ruspls</i>	EcM	2	yes
<i>Scleroderma citrinum</i>	Pers.	<i>Sclcit</i>	EcM	2	yes
<i>Sebacina incrustans</i>	(Pers.) Tul. & C. Tul.	<i>Sebinc</i>	EcM	2	yes
<i>Sistotrema confluens</i>	Pers.	<i>Siscon</i>	EcM	2	yes
<i>Suillus variegatus</i>	(Sw.) Kuntze	<i>Suivar</i>	EcM	2	yes
<i>Xerocomus cisalpinus</i>	Simonini, H. Ladurner & Peintner	<i>Xercis</i>	EcM	2	yes
<i>Xerocomus ferrugineus</i>	(Schaeff.) Alessio	<i>Xerfer</i>	EcM	2	yes
<i>Xerocomus porosporus</i>	Imler	<i>Xerpor</i>	EcM	2	yes
<i>Amanita eliae</i>	Quél.	<i>Amaeli</i>	EcM	1	yes
<i>Amanita franchetii</i>	(Boud.) Fayod	<i>Amafra</i>	EcM	1	yes
<i>Amanita porphyria</i>	Alb. & Schwein.	<i>Amapor</i>	EcM	1	yes
<i>Chalciporus piperatus</i>	(Bull.) Bataille	<i>Chapip</i>	EcM	1	yes
<i>Cortinarius</i> sp.01		<i>Cor_01</i>	EcM	1	yes
<i>Cortinarius</i> sp.02		<i>Cor_02</i>	EcM	1	yes
<i>Cortinarius</i> sp.03		<i>Cor_03</i>	EcM	1	yes
<i>Cortinarius</i> sp.04		<i>Cor_04</i>	EcM	1	yes
<i>Cortinarius</i> sp.05		<i>Cor_05</i>	EcM	1	yes
<i>Cortinarius</i> sp.06		<i>Cor_06</i>	EcM	1	yes

<i>Cortinarius</i> sp.09		<i>Cor_09</i>	EcM	1	yes
<i>Cortinarius</i> sp.10		<i>Cor_10</i>	EcM	1	yes
<i>Cortinarius</i> sp.11		<i>Cor_11</i>	EcM	1	yes
<i>Cortinarius</i> sp.12		<i>Cor_12</i>	EcM	1	yes
<i>Cortinarius</i> sp.13		<i>Cor_13</i>	EcM	1	yes
<i>Cortinarius</i> sp.16		<i>Cor_16</i>	EcM	1	yes
<i>Cortinarius</i> sp.17		<i>Cor_17</i>	EcM	1	yes
<i>Cortinarius</i> sp.18		<i>Cor_18</i>	EcM	1	yes
<i>Cortinarius</i> sp.19		<i>Cor_19</i>	EcM	1	yes
<i>Cortinarius</i> sp.20		<i>Cor_20</i>	EcM	1	yes
<i>Cortinarius</i> sp.21		<i>Cor_21</i>	EcM	1	yes
<i>Cortinarius albocyanus</i>	Fr.	<i>Coralc</i>	EcM	1	yes
<i>Cortinarius anomalus</i>	(Fr.) Fr.	<i>Corano</i>	EcM	1	yes
<i>Cortinarius anserinus</i>	(Velen.) Rob. Henry	<i>Corans</i>	EcM	1	yes
<i>Cortinarius barbatus</i>	(Batsch) Melot	<i>Corbar</i>	EcM	1	yes
<i>Cortinarius bataillei</i>	J. Favre	<i>Corbat</i>	EcM	1	yes
<i>Cortinarius camphoratus</i>	(Fr) Fr.	<i>Corcam</i>	EcM	1	yes
<i>Cortinarius caperatus</i>	(Pers.) Fr.	<i>Corcap</i>	EcM	1	yes
<i>Cortinarius cinnamomeus</i>	(L.) Gray	<i>Corcin</i>	EcM	1	yes
<i>Cortinarius citrinus</i>	(J.E. Lange) P.D. Orton	<i>Corcit</i>	EcM	1	yes
<i>Cortinarius comptulus</i>	M.M. Moser	<i>Corcom</i>	EcM	1	yes
<i>Cortinarius croceocaeruleus</i>	(Pers.) Fr.	<i>Corcrc</i>	EcM	1	yes
<i>Cortinarius delibutus</i>	Fr.	<i>Cordel</i>	EcM	1	yes
<i>Cortinarius depressus</i>	Fr.	<i>Cordep</i>	EcM	1	yes
<i>Cortinarius diasemospermus</i> var. <i>leptospermus</i>	H. Lindstr.	<i>Cordil</i>	EcM	1	yes
<i>Cortinarius flexipes</i> var. <i>inolens</i>	H. Lindstr.	<i>Corfli</i>	EcM	1	yes
<i>Cortinarius fulvescens</i> s.l.	Fr.	<i>Corful</i>	EcM	1	yes
<i>Cortinarius herpeticus</i>	Fr.	<i>Corher</i>	EcM	1	yes
<i>Cortinarius lebretonii</i>	Quél.	<i>Corleb</i>	EcM	1	yes
<i>Cortinarius obtusus</i>	(Fr.) Fr.	<i>Corobt</i>	EcM	1	yes
<i>Cortinarius raphanoides</i>	(Pers.) Fr.	<i>Corrap</i>	EcM	1	yes
<i>Cortinarius subbalaustinus</i>	Rob. Henry	<i>Corsbb</i>	EcM	1	yes
<i>Cortinarius subpurpurascens</i>	(Batsch) Fr.	<i>Corsbu</i>	EcM	1	yes
<i>Cortinarius scaurotraganoides</i>	Rob. Henry ex Rob. Henry	<i>Corsca</i>	EcM	1	yes
<i>Cortinarius semisanguineus</i>	(Fr.) Gillet	<i>Corsem</i>	EcM	1	yes
<i>Cortinarius turgidus</i>	Fr.	<i>Cortur</i>	EcM	1	yes
<i>Cortinarius uraceonemoralis</i>	Niskanen, Liimat., Dima, Kytöv., Bojantchev & H. Lindstr.	<i>Corura</i>	EcM	1	yes
<i>Cortinarius urbicus</i>	(Fr.) Fr.	<i>Corurb</i>	EcM	1	yes
<i>Cortinarius valgus</i>	Fr.	<i>Corval</i>	EcM	1	yes
<i>Cortinarius varicolor</i>	(Pers.) Fr.	<i>Corvar</i>	EcM	1	yes
<i>Cortinarius vulpinus</i>	(Velen.) Rob. Henry	<i>Corvul</i>	EcM	1	yes
<i>Cortinarius xanthocephalus</i>	P.D. Orton	<i>Corxcp</i>	EcM	1	yes
<i>Cortinarius xanthophyllus</i>	(Cooke) Rob. Henry	<i>Corxph</i>	EcM	1	yes
<i>Gomphidius roseus</i>	(Fr.) Fr.	<i>Gomros</i>	EcM	1	yes
<i>Hebeloma candidipes</i>	Bruchet	<i>Hebcan</i>	EcM	1	yes
<i>Hebeloma sacchariolens</i>	Quél.	<i>Hebsac</i>	EcM	1	yes
<i>Hydnum</i> sp.		<i>Hyd_sp</i>	EcM	1	yes
<i>Hygrophorus agathosmus</i>	(Fr.) Fr.	<i>Hygaga</i>	EcM	1	yes
<i>Hygrophorus lindtneri</i>	M.M. Moser	<i>Hyglin</i>	EcM	1	yes
<i>Hygrophorus penarioides</i>	Jacobsson & E. Larss.	<i>Hygpen</i>	EcM	1	yes
<i>Hygrophorus unicolor</i>	Gröger	<i>Hyguni</i>	EcM	1	yes
<i>Inocybe amblyospora</i> cf.	Kühner	<i>Ino_am</i>	EcM	1	yes
<i>Inocybe auricoma</i> cf.	(Batsch) J.E. Lange	<i>Ino_au</i>	EcM	1	yes

<i>Inocybe castanea</i>	Peck	<i>Inocas</i>	EcM	1	yes
<i>Inocybe flocculosa</i>	Sacc.	<i>Inoflo</i>	EcM	1	yes
<i>Inocybe grammata</i>	Quél. & Le Bret.	<i>Inogra</i>	EcM	1	yes
<i>Inocybe leiocephala</i>	D.E. Stuntz	<i>Inolei</i>	EcM	1	yes
<i>Inocybe microspora</i>	J.E. Lange	<i>Inomic</i>	EcM	1	yes
<i>Inocybe putilla</i>	Bres.	<i>Inopot</i>	EcM	1	yes
<i>Inocybe rimosa</i>	(Bull.) P. Kumm.	<i>Inorim</i>	EcM	1	yes
<i>Inocybe splendens</i>	R. Heim	<i>Inospl</i>	EcM	1	yes
<i>Lactarius bertillonii</i>	(Neuhoff ex Z. Schaef.) Bon	<i>Lacber</i>	EcM	1	yes
<i>Laccaria bicolor</i>	(Maire) P.D. Orton	<i>Lacbic</i>	EcM	1	yes
<i>Lactarius deterrimus</i>	Gröger	<i>Lacdet</i>	EcM	1	yes
<i>Lactarius glyciosmus</i>	(Fr.) Fr.	<i>Lacgly</i>	EcM	1	yes
<i>Lactarius pallidus</i>	Pers.	<i>Lacpal</i>	EcM	1	yes
<i>Lactarius vietus</i>	(Fr.) Fr.	<i>Lacvie</i>	EcM	1	yes
<i>Leccinum cyaneobasileucum</i>	Lannoy & Estad?s	<i>Leccya</i>	EcM	1	yes
<i>Leucocortinarius bulbiger</i>	(Alb. & Schwein.) Singer	<i>Leubul</i>	EcM	1	yes
<i>Phellodon melaleucus</i>	(Sw. ex Fr.) P. Karst.	<i>Phemel</i>	EcM	1	yes
<i>Ramaria fagetorum</i> cf.	Maas Geest. ex Schild	<i>Ram_fa</i>	EcM	1	yes
<i>Ramaria formosa</i>	(Pers.) Quél.	<i>Ramfor</i>	EcM	1	yes
<i>Rhizopogon roseolus</i>	(Corda) Th. Fr.	<i>Rhiros</i>	EcM	1	yes
<i>Russula aeruginea</i>	Lindblad	<i>Rusaer</i>	EcM	1	yes
<i>Russula amarissima</i>	Romagn. & E.-J. Gilbert	<i>Rusama</i>	EcM	1	yes
<i>Russula clavipes</i>	Velen.	<i>Ruscla</i>	EcM	1	yes
<i>Russula cremeoavellanea</i>	Singer	<i>Ruscre</i>	EcM	1	yes
<i>Russula farinipes</i>	Romell	<i>Rusfar</i>	EcM	1	yes
<i>Russula grisea</i>	Fr.	<i>Rusgri</i>	EcM	1	yes
<i>Russula lutensis</i>	Romagn. & Le Gal	<i>Ruslut</i>	EcM	1	yes
<i>Russula minutula</i>	Velen.	<i>Rusmin</i>	EcM	1	yes
<i>Russula nitida</i>	(Pers.) Fr.	<i>Rusnit</i>	EcM	1	yes
<i>Russula pseudointegra</i>	Arnould & Goris	<i>Ruspse</i>	EcM	1	yes
<i>Russula queletii</i>	Fr.	<i>Rusque</i>	EcM	1	yes
<i>Russula rhodella</i>	E.-J. Gilbert	<i>Rusrho</i>	EcM	1	yes
<i>Russula solaris</i>	Ferd. & Winge	<i>Russol</i>	EcM	1	yes
<i>Russula tinctipes</i>	J. Blum ex Bon	<i>Rustin</i>	EcM	1	yes
<i>Russula torulosa</i>	Bres.	<i>Rustor</i>	EcM	1	yes
<i>Russula virescens</i>	(Schaeff.) Fr.	<i>Rusvir</i>	EcM	1	yes
<i>Scleroderma cepa</i>	Pers.	<i>Sclice</i>	EcM	1	yes
<i>Strobilomyces strobilaceus</i>	(Scop.) Berk.	<i>Strstr</i>	EcM	1	yes
<i>Suillus luteus</i>	(L.) Roussel	<i>Suilut</i>	EcM	1	yes
<i>Thelephora terrestris</i>	Ehrh.	<i>Theter</i>	EcM	1	yes
<i>Tricholoma batschii</i>	Gulden	<i>Tribat</i>	EcM	1	yes
<i>Tricholoma sculpturatum</i>	(Fr.) Quél.	<i>Trisca</i>	EcM	1	yes
<i>Tricholoma stiparophyllum</i>	(N. Lund) P. Karst.	<i>Tristi</i>	EcM	1	yes
<i>Xerocomus chrysonema</i>	A.E. Hills & A.F.S. Taylor	<i>Xerchr</i>	EcM	1	yes
<i>Xerocomus parasiticus</i>	(Bull.) Quél.	<i>Xerpar</i>	EcM	1	yes
<i>Xerocomus ripariellus</i>	Redeuilh	<i>Xerrip</i>	EcM	1	yes
<i>Auriscalpium vulgare</i>	Gray	<i>Aurvul</i>	t. sapr.	25	yes, plotted
<i>Lycoperdon perlatum</i>	Pers.	<i>Lycper</i>	t. sapr.	24	yes, plotted
<i>Mycena pura</i>	(Pers.) P. Kumm.	<i>Mycpur</i>	t. sapr.	22	yes, plotted
<i>Gymnopus peronatus</i>	(Bolton) Antonín, Halling & Noordel.	<i>Gymper</i>	t. sapr.	21	yes, plotted
<i>Leotia lubrica</i>	(Scop.) Pers.	<i>Leolub</i>	t. sapr.	20	yes, plotted
<i>Mycena sanguinolenta</i>	(Alb. & Schwein.) P. Kumm.	<i>Mycsan</i>	t. sapr.	18	yes, plotted
<i>Gymnopus aquosus</i>	(Bull.) Antonín &	<i>Gymaqu</i>	t. sapr.	17	yes, plotted

	Noordel.				
<i>Rhodocollybia butyracea</i>	(Bull.) Lennox	<i>Rhobut</i>	t. sapr.	16	yes, plotted
<i>Clitocybe nebularis</i>	(Batsch) P. Kumm.	<i>Clineb</i>	t. sapr.	15	yes, plotted
<i>Baeospora myosura</i>	(Fr.) Singer	<i>Baemyo</i>	t. sapr.	14	yes, plotted
<i>Lepista nuda</i>	(Bull.) Cooke	<i>Lepnud</i>	t. sapr.	14	yes, plotted
<i>Lycoperdon molle</i>	Pers.	<i>Lycmol</i>	t. sapr.	14	yes, plotted
<i>Mycena galopus</i> var. <i>galopus</i>	(Pers.) P. Kumm.	<i>Mycglp</i>	t. sapr.	13	yes
<i>Lepiota clypeolaria</i>	(Bull.) P. Kumm.	<i>Lepcly</i>	t. sapr.	12	yes
<i>Mycena aurantiomarginata</i>	(Fr.) Quél.	<i>Mycaur</i>	t. sapr.	12	yes
<i>Mycena flavescens</i>	Velen.	<i>Mycflv</i>	t. sapr.	12	yes
<i>Mycena rosea</i>	Gramberg	<i>Mycrsa</i>	t. sapr.	11	yes
<i>Clitocybe ditopa</i>	(Fr.) Gillet	<i>Clidit</i>	t. sapr.	10	yes
<i>Gymnopus erythropus</i>	(Pers.) Antonín, Halling & Noordel.	<i>Gymery</i>	t. sapr.	10	yes
<i>Mycena zephrus</i>	(Fr.) P. Kumm.	<i>Myczep</i>	t. sapr.	10	yes
<i>Lepista flaccida</i>	(Sowerby) Pat.	<i>Lepfla</i>	t. sapr.	9	yes
<i>Clitocybe candicans</i>	(Pers.) P. Kumm.	<i>Clican</i>	t. sapr.	8	yes
<i>Clitocybe phyllophila</i>	(Pers.) P. Kumm.	<i>Cliphy</i>	t. sapr.	8	yes
<i>Gymnopus androsaceus</i>	(L.) J.L. Mata & R.H. Petersen	<i>Gymand</i>	t. sapr.	8	yes
<i>Infundibulicybe gibba</i>	(Pers.) Harmaja	<i>Infgib</i>	t. sapr.	8	yes
<i>Lepiota castanea</i>	Quél.	<i>Lepcas</i>	t. sapr.	8	yes
<i>Marasmius bulliardii</i>	Quél.	<i>Marbul</i>	t. sapr.	8	yes
<i>Atheniella flavoalba</i>	(Fr.) Redhead, Moncalvo, Vilgalys, Desjardin, B.A. Perry	<i>Athfla</i>	t. sapr.	7	yes
<i>Lycoperdon nigrescens</i>	Pers.	<i>Lycnig</i>	t. sapr.	7	yes
<i>Lyophyllum platypum</i>	Kühner	<i>Lyopla</i>	t. sapr.	7	yes
<i>Strobilurus tenacellus</i>	(Pers.) Singer	<i>Strten</i>	t. sapr.	7	yes
<i>Collybia cirrata</i>	(Schumach.) Quél.	<i>Colcir</i>	t. sapr.	6	yes
<i>Conocybe tetrasporoides</i>	Hauskn.	<i>Contet</i>	t. sapr.	6	yes
<i>Hygrophoropsis aurantiaca</i>	(Wulfen) Maire	<i>Hygaur</i>	t. sapr.	6	yes
<i>Macrotyphula juncea</i>	(Alb. & Schwein.) Berthier	<i>Macjun</i>	t. sapr.	6	yes
<i>Macrolepiota procera</i>	(Scop.) Singer	<i>Macpro</i>	t. sapr.	6	yes
<i>Mycena amicta</i>	(Fr.) Quél.	<i>Mycami</i>	t. sapr.	6	yes
<i>Mycena galopus</i> var. <i>leucogala</i>	(Cooke) J.E. Lange	<i>Mycgll</i>	t. sapr.	6	yes
<i>Collybia tuberosa</i>	(Bull.) P. Kumm.	<i>Coltub</i>	t. sapr.	5	yes
<i>Entoloma juncinum</i>	(Kühner & Romagn.) Noordel.	<i>Entjun</i>	t. sapr.	5	yes
<i>Gymnopus confluens</i>	(Pers.) Antonín, Halling & Noordel.	<i>Gymcon</i>	t. sapr.	5	yes
<i>Gymnopus dryophilus</i>	(Bull.) Murrill	<i>Gymdry</i>	t. sapr.	5	yes
<i>Lycoperdon excipuliforme</i>	(Scop.) Pers.	<i>Lycexc</i>	t. sapr.	5	yes
<i>Mycena rosella</i>	(Fr.) P. Kumm.	<i>Mycrla</i>	t. sapr.	5	yes
<i>Mycena stylobates</i>	(Pers.) P. Kumm.	<i>Mycsty</i>	t. sapr.	5	yes
<i>Roridomyces roridus</i>	(Scop.) Rexer	<i>Rorrora</i>	t. sapr.	5	yes
<i>Tubaria minutalis</i>	Romagn.	<i>Tubmin</i>	t. sapr.	5	yes
<i>Clitocybe metachroa</i>	(Fr.) P. Kumm.	<i>Climet</i>	t. sapr.	4	yes
<i>Clitocybe phaeophthalma</i>	(Pers.) Kuyper	<i>Clipha</i>	t. sapr.	4	yes
<i>Collybia cookei</i>	(Bres.) J.D. Arnold	<i>Colcoo</i>	t. sapr.	4	yes
<i>Conocybe moseri</i>	Watling	<i>Conmos</i>	t. sapr.	4	yes
<i>Lyophyllum mephiticum</i>	(Fr.) Singer	<i>Lyomep</i>	t. sapr.	4	yes
<i>Mycena capillaris</i>	(Schumach.) P. Kumm.	<i>Myccap</i>	t. sapr.	4	yes
<i>Naucoria bohémica</i>	Velen.	<i>Nauboh</i>	t. sapr.	4	yes
<i>Ramaria flaccida</i>	(Fr.) Bourdot	<i>Ramfla</i>	t. sapr.	4	yes
<i>Agaricus essettei</i>	Bon	<i>Agaess</i>	t. sapr.	3	yes

<i>Chlorophyllum olivieri</i>	(Barla) Vellinga	<i>Chloli</i>	t. sapr.	3	yes
<i>Clitocybe odora</i>	(Bull.) P. Kumm.	<i>Cliodo</i>	t. sapr.	3	yes
<i>Clitopilus prunulus</i>	(Scop.) P. Kumm.	<i>Clipru</i>	t. sapr.	3	yes
<i>Cystoderma amianthinum</i>	(Scop.) Fayod	<i>Cysami</i>	t. sapr.	3	yes
<i>Gymnopus quercophilus</i>	(Pouzar) Antonín & Noordel.	<i>Gymque</i>	t. sapr.	3	yes
<i>Helvella elastica</i>	Bull.	<i>Helela</i>	t. sapr.	3	yes
<i>Lepiota cristata</i>	(Bolton) P. Kumm.	<i>Lepcri</i>	t. sapr.	3	yes
<i>Lepiota ignivolvata</i>	Bousset & Joss. ex Joss.	<i>Lepign</i>	t. sapr.	3	yes
<i>Marasmius cohaerens</i>	(Pers.) Cooke & Quél.	<i>Marcoh</i>	t. sapr.	3	yes
<i>Mycena filopes</i>	(Bull.) P. Kumm.	<i>Mycfil</i>	t. sapr.	3	yes
<i>Mycena metata</i>	(Fr.) P. Kumm.	<i>Mycmet</i>	t. sapr.	3	yes
<i>Phallus impudicus</i>	L.	<i>Phaimp</i>	t. sapr.	3	yes
<i>Pholiotina brunnea</i>	(Watling) Singer	<i>Phobru</i>	t. sapr.	3	yes
<i>Ripartites tricholoma</i>	(Alb. & Schwein.) P. Karst.	<i>Riptri</i>	t. sapr.	3	yes
<i>Strobilurus stephanocystis</i>	(Kühner & Romagn. ex Hora) Singer	<i>Strste</i>	t. sapr.	3	yes
<i>Tubaria conspersa</i>	(Pers.) Fayod	<i>Tubcon</i>	t. sapr.	3	yes
<i>Tubaria furfuracea</i>	(Pers.) Gillet	<i>Tubfur</i>	t. sapr.	3	yes
<i>Agaricus semotus</i>	Fr.	<i>Agasem</i>	t. sapr.	2	yes
<i>Ampulloclitocybe clavipes</i>	(Pers.) Redhead, Lutzoni, Moncalvo & Vilgalys	<i>Ampcla</i>	t. sapr.	2	yes
<i>Anthina flammea</i>	(Jung.) Fr.	<i>Antfla</i>	t. sapr.	2	yes
<i>Clitocybe fragrans</i>	(With.) P. Kumm.	<i>Clifra</i>	t. sapr.	2	yes
<i>Conocybe enderlei</i> var. <i>enderlei</i>	Hauskn.	<i>Conend</i>	t. sapr.	2	yes
<i>Cystolepiota seminuda</i>	(Lasch) Bon	<i>Cyssem</i>	t. sapr.	2	yes
<i>Entoloma hebes</i>	(Romagn.) Trimbach	<i>Entheb</i>	t. sapr.	2	yes
<i>Gymnopus ocior</i>	(Pers.) Antonín & Noordel.	<i>Gymoci</i>	t. sapr.	2	yes
<i>Helvella lacunosa</i>	Afzel.	<i>Hellac</i>	t. sapr.	2	yes
<i>Helvella macropus</i>	(Pers.) Gray	<i>Helmac</i>	t. sapr.	2	yes
<i>Lyophyllum leucophaeatum</i>	(P. Karst.) P. Karst.	<i>Lyoleu</i>	t. sapr.	2	yes
<i>Mycena abramsii</i> cf.	(Murrill) Murrill	<i>Myc_ab</i>	t. sapr.	2	yes
<i>Mycena fagetorum</i> cf.	(Fr.) Gillet	<i>Myc_fa</i>	t. sapr.	2	yes
<i>Mycena clavicularis</i>	(Fr.) Gillet	<i>Myccla</i>	t. sapr.	2	yes
<i>Mycena diosma</i>	Krieglst. & Schwöbel	<i>Mycdio</i>	t. sapr.	2	yes
<i>Mycena rubromarginata</i>	(Fr.) P. Kumm.	<i>Mycrub</i>	t. sapr.	2	yes
<i>Peziza saniosa</i>	Schrad.	<i>Pezsan</i>	t. sapr.	2	yes
<i>Rhodocybe gemina</i>	(Paulet) Kuyper & Noordel.	<i>Rhogem</i>	t. sapr.	2	yes
<i>Strobilurus esculentus</i>	(Wulfen) Singer	<i>Stresc</i>	t. sapr.	2	yes
<i>Agaricus sylvaticus</i>	Schaeff.	<i>Agasyl</i>	t. sapr.	1	yes
<i>Agrocybe vervacti</i>	(Fr.) Singer	<i>Agrver</i>	t. sapr.	1	yes
<i>Ciboria amentacea</i> cf.	(Balb.) Fuckel	<i>Cib_am</i>	t. sapr.	1	yes
<i>Clavariadelphus pistillaris</i>	(L.) Donk	<i>Clapis</i>	t. sapr.	1	yes
<i>Conocybe macrocephala</i> cf.	Kühner & Watling	<i>Con_ma</i>	t. sapr.	1	yes
<i>Conocybe ochrostriata</i> var. <i>ochrostriata</i>	Hauskn.	<i>Conovo</i>	t. sapr.	1	yes
<i>Coprinopsis jonesii</i>	(Peck) Redhead, Vilgalys & Moncalvo	<i>Copjon</i>	t. sapr.	1	yes
<i>Cystodermella cinnabarina</i>	(Alb. & Schwein.) Harmaja	<i>Cyscin</i>	t. sapr.	1	yes
<i>Entoloma conferendum</i> var. <i>pusillum</i>	(Velen.) Noordel.	<i>Entcon</i>	t. sapr.	1	yes
<i>Entoloma jahnii</i>	Wölfel & Winterh.	–	t. sapr.	1	no, omitted
<i>Lepiota boudieri</i>	Bres.	<i>Lepbou</i>	t. sapr.	1	yes

<i>Lepista glaucocana</i>	(Bres.) Singer	<i>Lepgla</i>	t. sapr.	1	yes
<i>Lycoperdon lividum</i>	Pers.	<i>Lycliv</i>	t. sapr.	1	yes
<i>Lyophyllum baeospermum</i>	Romagn.	<i>Lyobae</i>	t. sapr.	1	yes
<i>Lyophyllum boudieri</i>	Kühner & Romagn.	<i>Lyobou</i>	t. sapr.	1	yes
<i>Lyophyllum rancidum</i>	(Fr.) Singer	<i>Lyoran</i>	t. sapr.	1	yes
<i>Macrocyttidia cucumis</i>	(Pers.) Joss.	<i>Maccuc</i>	t. sapr.	1	yes
<i>Macrolepiota mastoidea</i>	(Fr.) Singer	<i>Macmas</i>	t. sapr.	1	yes
<i>Marasmius epiphyllus</i>	(Pers.) Fr.	<i>Marepi</i>	t. sapr.	1	yes
<i>Marasmius setosus</i>	(Sowerby) Noordel.	<i>Marset</i>	t. sapr.	1	yes
<i>Marasmius wynneae</i>	Berk. & Broome	<i>Marwyn</i>	t. sapr.	1	yes
<i>Mycena rebaudengi</i> cf.	Robich	<i>Myc_re</i>	t. sapr.	1	yes
<i>Mycena cinerella</i>	(P. Karst.) P. Karst.	<i>Myccin</i>	t. sapr.	1	yes
<i>Mycena pelianthina</i>	(Fr.) Quél.	<i>Mycpel</i>	t. sapr.	1	yes
<i>Mycena polyadelpha</i>	(Lasch) Kühner	<i>Mycpla</i>	t. sapr.	1	yes
<i>Mycena rhenana</i>	Maas Geest. & Winterh.	<i>Mycrhe</i>	t. sapr.	1	yes
<i>Mycena vulgaris</i>	(Pers.) P. Kumm.	<i>Mycvul</i>	t. sapr.	1	yes
<i>Mycetinis scorodoni</i>	(Fr.) A. Wilson & Desjardin	<i>Mycsco</i>	t. sapr.	1	yes
<i>Peziza arvernensis</i> cf.	Roze & Boud.	<i>Pez_ar</i>	t. sapr.	1	yes
<i>Peziza badia</i>	Pers.	<i>Pezbad</i>	t. sapr.	1	yes
<i>Peziza phyllogena</i>	Cooke	<i>Pezphy</i>	t. sapr.	1	yes
<i>Peziza succosa</i>	Berk.	<i>Pezsuc</i>	t. sapr.	1	yes
<i>Ramaria eumorpha</i>	(P. Karst.) Corner	<i>Rameum</i>	t. sapr.	1	yes
<i>Stropharia cyanea</i>	(Bull.) Tuom.	<i>Strcya</i>	t. sapr.	1	yes
<i>Tarzetta cupularis</i>	(L.) Svrček	<i>Tarcup</i>	t. sapr.	1	yes
<i>Stereum hirsutum</i>	(Willd.) Pers.	<i>Stehir</i>	wood-inh.	35	yes, plotted
<i>Exidia nigricans</i>	(With.) P. Roberts	<i>Exinig</i>	wood-inh.	31	yes, plotted
<i>Schizopora paradoxa</i> s.l.	(Schrad.) Donk	<i>Schpar</i>	wood-inh.	31	yes, plotted
<i>Schizopora flavipora</i>	(Berk. & M.A. Curtis ex Cooke) Ryvarde	<i>Schfla</i>	wood-inh.	29	yes, plotted
<i>Stereum ochraceoflavum</i>	(Schwein.) Sacc.	<i>Steocf</i>	wood-inh.	29	yes, plotted
<i>Steccherinum ochraceum</i>	(Pers.) Gray	<i>Steoch</i>	wood-inh.	26	yes, plotted
<i>Mycena vitilis</i>	(Fr.) Quél.	<i>Mycvit</i>	wood-inh.	25	yes, plotted
<i>Xylaria hypoxylon</i>	(L.) Grev.	<i>Xylhyp</i>	wood-inh.	25	yes, plotted
<i>Hymenochaete rubiginosa</i>	(Dicks.) Lévl.	<i>Hymrub</i>	wood-inh.	24	yes, plotted
<i>Antrodiella fragrans</i>	(A. David & Tortiç) A. David & Tortiç	<i>Antfra</i>	wood-inh.	21	yes, plotted
<i>Skeletocutis nivea</i>	(Jungh.) Jean Keller	<i>Skeniv</i>	wood-inh.	21	yes, plotted
<i>Hymenopellis radicata</i>	(Relhan) R.H. Petersen	<i>Hymrad</i>	wood-inh.	19	yes, plotted
<i>Mycetinis alliaceus</i>	(Jacq.) Earle ex A.W. Wilson & Desjardin	<i>Mycall</i>	wood-inh.	19	yes, plotted
<i>Biscogniauxia nummularia</i>	(Bull.) Kuntze	<i>Bisnum</i>	wood-inh.	18	yes, plotted
<i>Mycena polygramma</i>	(Bull.) Gray	<i>Mycpgr</i>	wood-inh.	18	yes, plotted
<i>Stereum subtomentosum</i>	Pouzar	<i>Stesub</i>	wood-inh.	18	yes, plotted
<i>Crepidotus variabilis</i>	(Pers.) P. Kumm.	<i>Crevar</i>	wood-inh.	17	yes, plotted
<i>Galerina marginata</i>	(Batsch) Kühner	<i>Galmar</i>	wood-inh.	16	yes, plotted
<i>Hypocrea citrina</i>	(Pers.) Fr.	<i>Hypcit</i>	wood-inh.	16	yes, plotted
<i>Lycoperdon pyriforme</i>	Willd.	<i>Lycpyr</i>	wood-inh.	16	yes, plotted
<i>Mycena epipterygia</i>	(Scop.) Gray	<i>Mycepi</i>	wood-inh.	16	yes, plotted
<i>Ramaria stricta</i>	(Pers.) Quél.	<i>Ramstr</i>	wood-inh.	16	yes, plotted
<i>Stereum sanguinolentum</i>	(Alb. & Schwein.) Fr.	<i>Stesan</i>	wood-inh.	16	yes, plotted
<i>Trametes versicolor</i>	(L.) Pilát	<i>Traver</i>	wood-inh.	16	yes, plotted
<i>Hypholoma fasciculare</i>	(Huds.) P. Kumm.	<i>Hypfas</i>	wood-inh.	15	yes, plotted
<i>Postia subcaesia</i>	(A. David) Jülich	<i>Possub</i>	wood-inh.	15	yes, plotted
<i>Schizophyllum commune</i>	Fr.	<i>Schcom</i>	wood-inh.	15	yes, plotted
<i>Xylaria carpophila</i>	(Pers.) Fr.	<i>Xylcar</i>	wood-inh.	15	yes, plotted
<i>Calocera furcata</i>	(Fr.) Fr.	<i>Calfur</i>	wood-inh.	14	yes, plotted

<i>Hypoxylon fragiforme</i>	(Pers.) J. Kickx f.	<i>Hypfra</i>	wood-inh.	14	yes, plotted
<i>Mycena galericulata</i>	(Scop.) Gray	<i>Mycglu</i>	wood-inh.	14	yes, plotted
<i>Polyporus varius</i>	(Pers.) Fr.	<i>Polvar</i>	wood-inh.	14	yes, plotted
<i>Cyathus striatus</i>	(Huds.) Willd.	<i>Cyastr</i>	wood-inh.	13	yes
<i>Pluteus cervinus</i>	(Schaeff.) P. Kumm.	<i>Plucer</i>	wood-inh.	13	yes
<i>Postia stiptica</i>	(Pers.) Jülich	<i>Possti</i>	wood-inh.	13	yes
<i>Trametes hirsuta</i>	(Wulfen) Lloyd	<i>Trahir</i>	wood-inh.	13	yes
<i>Clitocybula platyphylla</i>	(Pers.) Malençon & Bertault	<i>Clipla</i>	wood-inh.	12	yes
<i>Diatrype stigma</i>	(Hoffm.) Fr.	<i>Diasti</i>	wood-inh.	12	yes
<i>Fuscoporia contigua</i>	(Pers.) G. Cunn.	<i>Fuscon</i>	wood-inh.	12	yes
<i>Panellus stipticus</i>	(Bull.) P. Karst.	<i>Pansti</i>	wood-inh.	12	yes
<u><i>Aleurodiscus disciformis</i></u>	(DC.) Pat.	<i>Aledis</i>	wood-inh.	11	yes
<u><i>Armillaria lutea</i></u>	Gillet	<i>Armlut</i>	wood-inh.	11	yes
<i>Exidia glandulosa</i>	(Bull.) Fr.	<i>Exigla</i>	wood-inh.	11	yes
<i>Junghuhnia nitida</i>	(Pers.) Ryvarde	<i>Junnit</i>	wood-inh.	11	yes
<i>Mycena maculata</i>	P. Karst.	<i>Mycmac</i>	wood-inh.	11	yes
<i>Phlebia rufa</i>	(Pers.) M.P. Christ.	<i>Phlruf</i>	wood-inh.	11	yes
<i>Psathyrella pygmaea</i>	(Bull.) Singer	<i>Psapyg</i>	wood-inh.	10	yes
<i>Heterobasidion annosum</i>	(Fr.) Bref.	<i>Hetann</i>	wood-inh.	9	yes
<i>Laxitextum bicolor</i>	(Pers.) Lentz	<i>Laxbic</i>	wood-inh.	9	yes
<i>Mycena haematopus</i>	(Pers.) P. Kumm.	<i>Mychae</i>	wood-inh.	9	yes
<i>Plicaturopsis crispa</i>	(Pers.) D.A. Reid	<i>Plicri</i>	wood-inh.	9	yes
<u><i>Pseudohydnum gelatinosum</i></u>	(Scop.) P. Karst.	<i>Psegel</i>	wood-inh.	9	yes
<i>Antrodia albida</i>	(Fr.) Donk	<i>Antalb</i>	wood-inh.	8	yes
<i>Antrodia malicola</i>	(Berk. & M.A. Curtis) Donk	<i>Antmal</i>	wood-inh.	8	yes
<i>Auricularia auricula-judae</i>	(Bull.) Qué.	<i>Auraur</i>	wood-inh.	8	yes
<i>Crepidotus cesatii</i>	(Rabenh.) Sacc.	<i>Creces</i>	wood-inh.	8	yes
<i>Galerina pruinatipes</i>	A.H. Sm.	<i>Galpru</i>	wood-inh.	8	yes
<i>Gymnopilus penetrans</i>	(Fr.) Murrill	<i>Gympen</i>	wood-inh.	8	yes
<u><i>Hypholoma lateritium</i></u>	(Schaeff.) P. Kumm.	<i>Hyplat</i>	wood-inh.	8	yes
<i>Pholiota lenta</i>	(Pers.) Singer	<i>Pholen</i>	wood-inh.	8	yes
<i>Polyporus alveolaris</i>	(DC.) Bondartsev & Singer	<i>Polalv</i>	wood-inh.	8	yes
<i>Simocybe centunculus</i>	(Fr.) Singer	<i>Simcen</i>	wood-inh.	8	yes
<i>Steccherinum fimbriatum</i>	(Pers.) J. Erikss.	<i>Stefim</i>	wood-inh.	8	yes
<i>Xylaria polymorpha</i>	(Pers.) Grev.	<i>Xylpol</i>	wood-inh.	8	yes
<i>Antrodiella faginea</i>	Vampola & Pouzar	<i>Antfag</i>	wood-inh.	7	yes
<i>Byssomerulius corium</i>	(Pers.) Parmasto	<i>Byscor</i>	wood-inh.	7	yes
<i>Calocera viscosa</i>	(Pers.) Fr.	<i>Calvis</i>	wood-inh.	7	yes
<i>Mycena arcangeliana</i>	Bres.	<i>Mycarc</i>	wood-inh.	7	yes
<u><i>Psathyrella piluliformis</i></u>	(Bull.) P.D. Orton	<i>Psapil</i>	wood-inh.	7	yes
<i>Bjerkandera adusta</i>	(Willd.) P. Karst.	<i>Bjeadu</i>	wood-inh.	6	yes
<i>Ceriporiopsis mucida</i>	(Pers.) Gilb. & Ryvarde	<i>Cermuc</i>	wood-inh.	6	yes
<i>Daedaleopsis confragosa</i>	(Bolton) J. Schröt.	<i>Daecon</i>	wood-inh.	6	yes
<i>Diatrype disciformis</i>	(Hoffm.) Fr.	<i>Diadis</i>	wood-inh.	6	yes
<i>Marasmiellus ramealis</i>	(Bull.) Singer	<i>Marram</i>	wood-inh.	6	yes
<u><i>Mycena inclinata</i></u>	(Fr.) Qué.	<i>Mycinc</i>	wood-inh.	6	yes
<i>Phellinus viticola</i>	(Schwein.) Donk	<i>Phevit</i>	wood-inh.	6	yes
<i>Polyporus ciliatus</i>	Fr.	<i>Polcil</i>	wood-inh.	6	yes
<i>Trichaptum abietinum</i>	(Dicks.) Ryvarde	<i>Triabi</i>	wood-inh.	6	yes
<i>Xylaria longipes</i>	Nitschke	<i>Xyllon</i>	wood-inh.	6	yes
<i>Antrodiella pallescens</i>	(Pilát) Niemelä & Miettinen	<i>Antpal</i>	wood-inh.	5	yes
<i>Calocera cornea</i>	(Batsch) Fr.	<i>Calcor</i>	wood-inh.	5	yes
<i>Hapalopilus nidulans</i>	(Fr.) P. Karst.	<i>Hapnid</i>	wood-inh.	5	yes

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

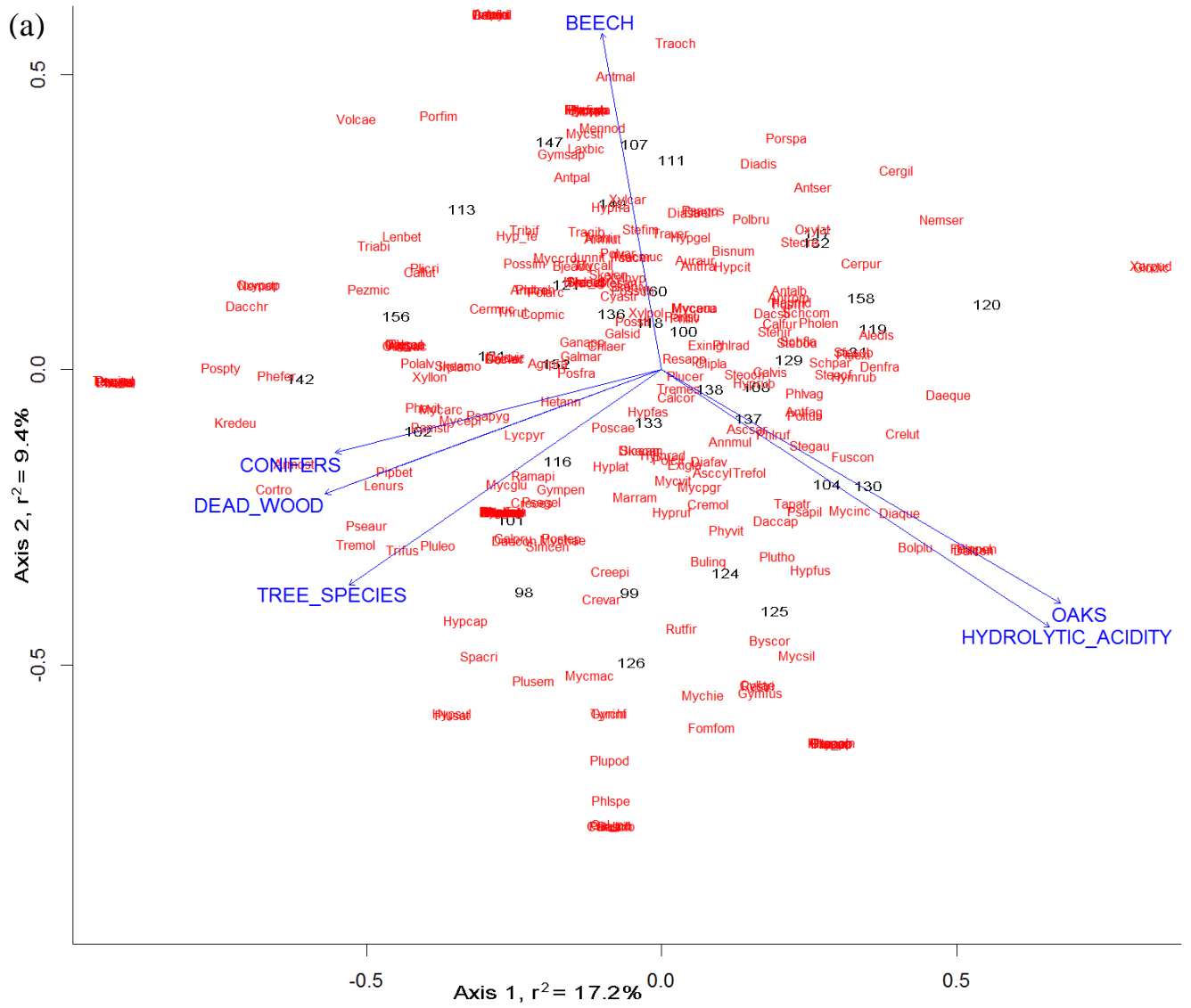


<i>Postia fragilis</i>	(Fr.) Jülich	<i>Posfra</i>	wood-inh.	3	yes
<i>Postia simanii</i>	(Pilát ex Pilát) Jülich	<i>Possim</i>	wood-inh.	3	yes
<i>Ramaria apiculata</i>	(Fr.) Donk	<i>Ramapi</i>	wood-inh.	3	yes
<i>Skeletocutis amorpha</i>	(Fr.) Kotl. & Pouzar	<i>Skeamo</i>	wood-inh.	3	yes
<u><i>Tricholomopsis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

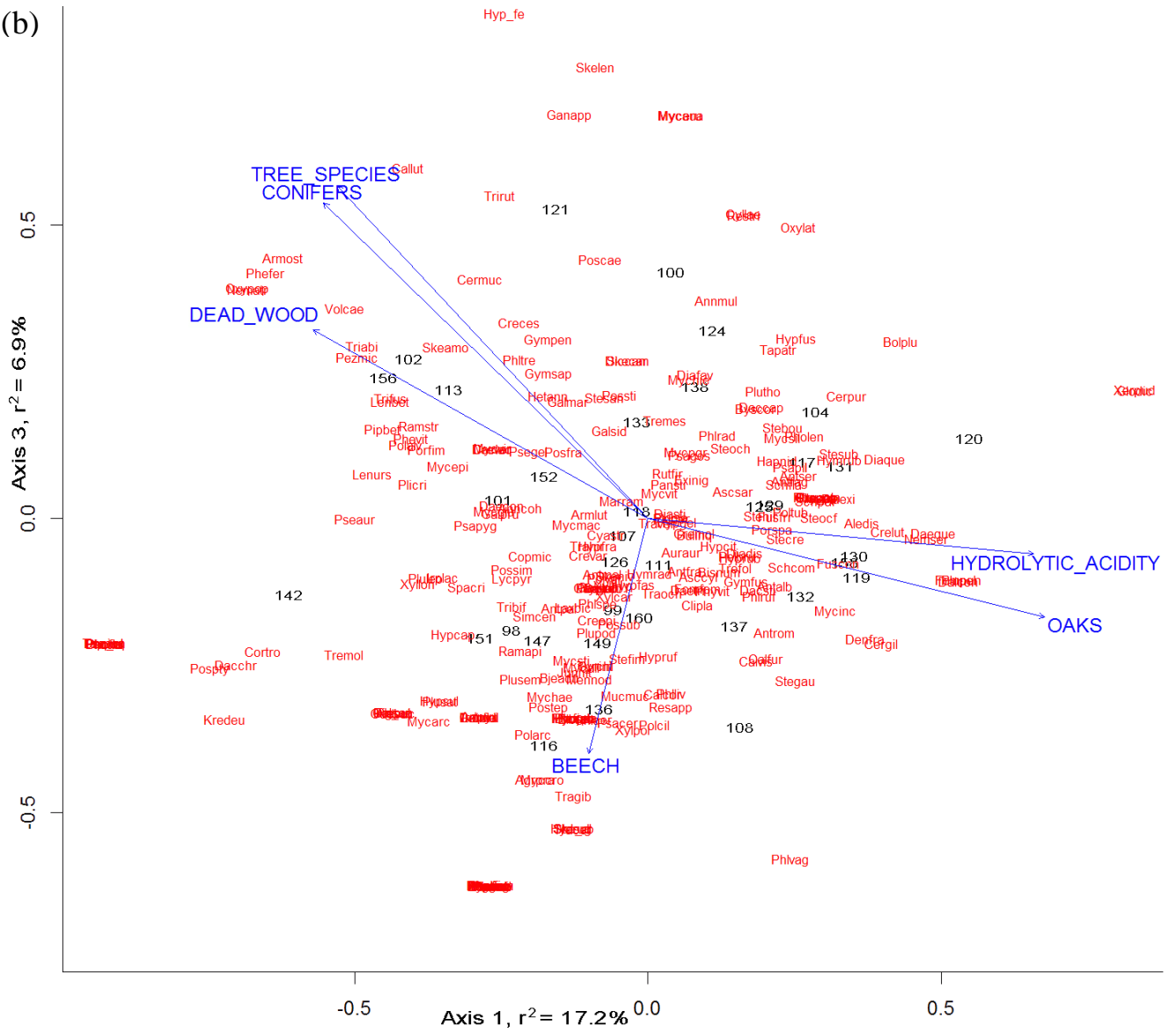
<i>Crepidotus applanatus</i>	(Pers.) P. Kumm.	<i>Creapp</i>	wood-inh.	1	yes
<i>Crepidotus calolepis</i>	(Fr.) P. Karst.	<i>Creecal</i>	wood-inh.	1	yes
<i>Crepidotus versutus</i>	(Peck) Sacc.	<i>Crever</i>	wood-inh.	1	yes
<i>Cylindrobasidium laeve</i>	(Pers.) Chamuris	<i>Cyllae</i>	wood-inh.	1	yes
<i>Dacrymyces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

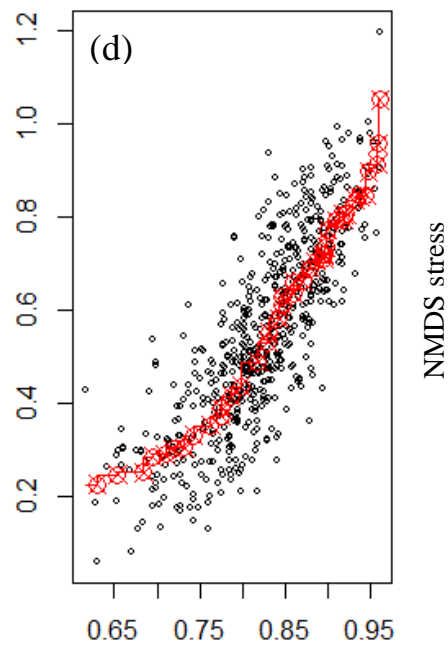
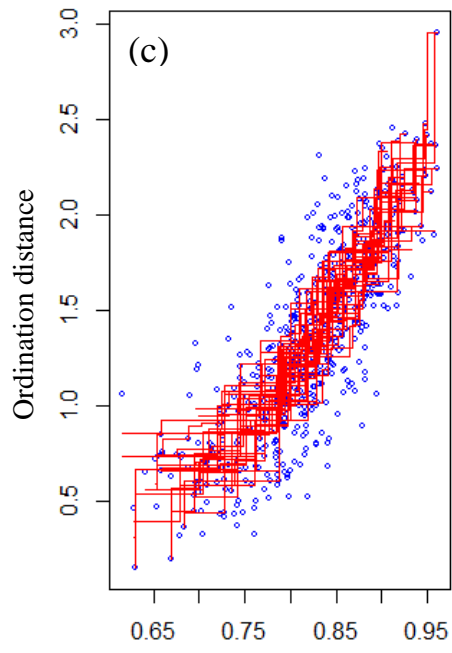
<i>Skeletocutis carneogrisea</i>	A. David	<i>Skecar</i>	wood-inh.	1	yes
<i>Stereum rugosum</i>	Pers.	<i>Sterug</i>	wood-inh.	1	yes
<i>Tapinella panuoides</i>	(Fr.) E.-J. Gilbert	<i>Tappan</i>	wood-inh.	1	yes
<i>Trametopsis cervina</i>	(Schwein.) Tomšovský	<i>Tracer</i>	wood-inh.	1	yes
<i>Trametes ochracea</i>	(Pers.) Gilb. & Ryvarde	<i>Traoch</i>	wood-inh.	1	yes
<i>Trametes suaveolens</i>	(L.) Fr.	<i>Trasua</i>	wood-inh.	1	yes
<i>Tyromyces chioneus</i>	(Fr.) P. Karst.	<i>Tyrchi</i>	wood-inh.	1	yes
<i>Volvariella caesiotincta</i>	P.D. Orton	<i>Volcae</i>	wood-inh.	1	yes
<i>Xerula pudens</i>	(Pers.) Singer	<i>Xerpud</i>	wood-inh.	1	yes
<i>Rickenella fibula</i>	(Bull.) Raithehl.	–	bryophilous	8	no, skipped
<i>Rickenella swartzii</i>	(Fr.) Kuyper	–	bryophilous	4	no, skipped
<i>Cordyceps larvicola</i>	Quél.	–	entomopath.	1	no, skipped
<i>Marasmius rotula</i>	(Scop.) Fr.	–	lign./t. sapr.	6	no, skipped
<i>Marasmius torquescens</i>	Quél.	–	lign./t. sapr.	5	no, skipped
<i>Mycena leptcephala</i>	(Pers.) Gillet	–	lign./t. sapr.	4	no, skipped
<i>Psathyrella lutensis</i>	(Romagn.) Bon	–	lign./t. sapr.	3	no, skipped
<i>Pholiota scamba</i>	(Fr.) M.M. Moser	–	lign./t. sapr.	1	no, skipped
<i>Psathyrella cortinarioides</i>	P.D. Orton	–	lign./t. sapr.	1	no, skipped
<i>Psathyrella fagetophila</i>	Örstadius & Enderle	–	lign./t. sapr.	1	no, skipped
<i>Psathyrella microrrhiza</i>	(Lasch) Konrad & Maubl.	–	lign./t. sapr.	1	no, skipped
<i>Psathyrella prona</i>	(Fr.) Gillet	–	lign./t. sapr.	1	no, skipped
<i>Psathyrella spadiceogrisea</i>	(Schaeff.) Maire	–	lign./t. sapr.	1	no, skipped
<i>Asterophora lycoperdoides</i>	(Bull.) Ditmar	–	mycotrophic	7	no, skipped
<i>Tremella encephala</i>	Pers.	–	mycotrophic	6	no, skipped
<i>Elaphocordyceps ophioglossoides</i>	(Ehrh.) G.H. Sung, J.M. Sung & Spatafora	–	mycotrophic	3	no, skipped
<i>Tremella globispora</i>	D.A. Reid	–	mycotrophic	1	no, skipped
<i>Entoloma rhodopolium</i>	(Fr.) P. Kumm.	–	t. sapr./myc.	15	no, skipped
<i>Otidea onotica</i>	(Pers.) Fuckel	–	t. sapr./myc.	11	no, skipped
<i>Otidea alutacea</i>	(Pers.) Masee	–	t. sapr./myc.	5	no, skipped
<i>Entoloma politum</i>	(Pers.) Donk	–	t. sapr./myc.	4	no, skipped
<i>Otidea bufonia</i>	(Pers.) Boud.	–	t. sapr./myc.	4	no, skipped
<i>Otidea fuckelii</i>	M. Carbone & Van Vooren	–	t. sapr./myc.	1	no, skipped
<i>Otidea grandis</i>	(Pers.) Arnould	–	t. sapr./myc.	1	no, skipped
<i>Otidea propinquata</i> 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 ( $\tau$ ), 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  $\tau$  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).

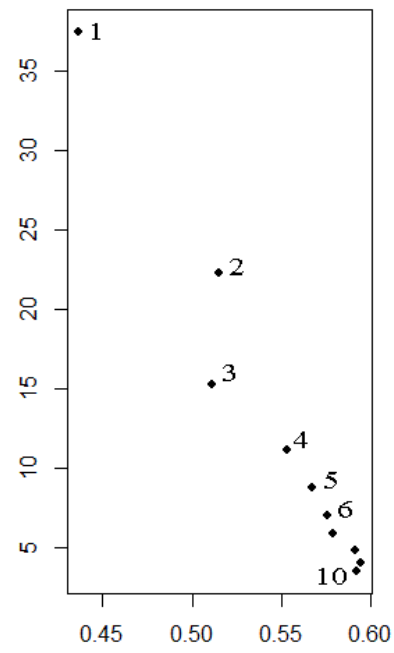


(b)





(e)



Observed dissimilarity

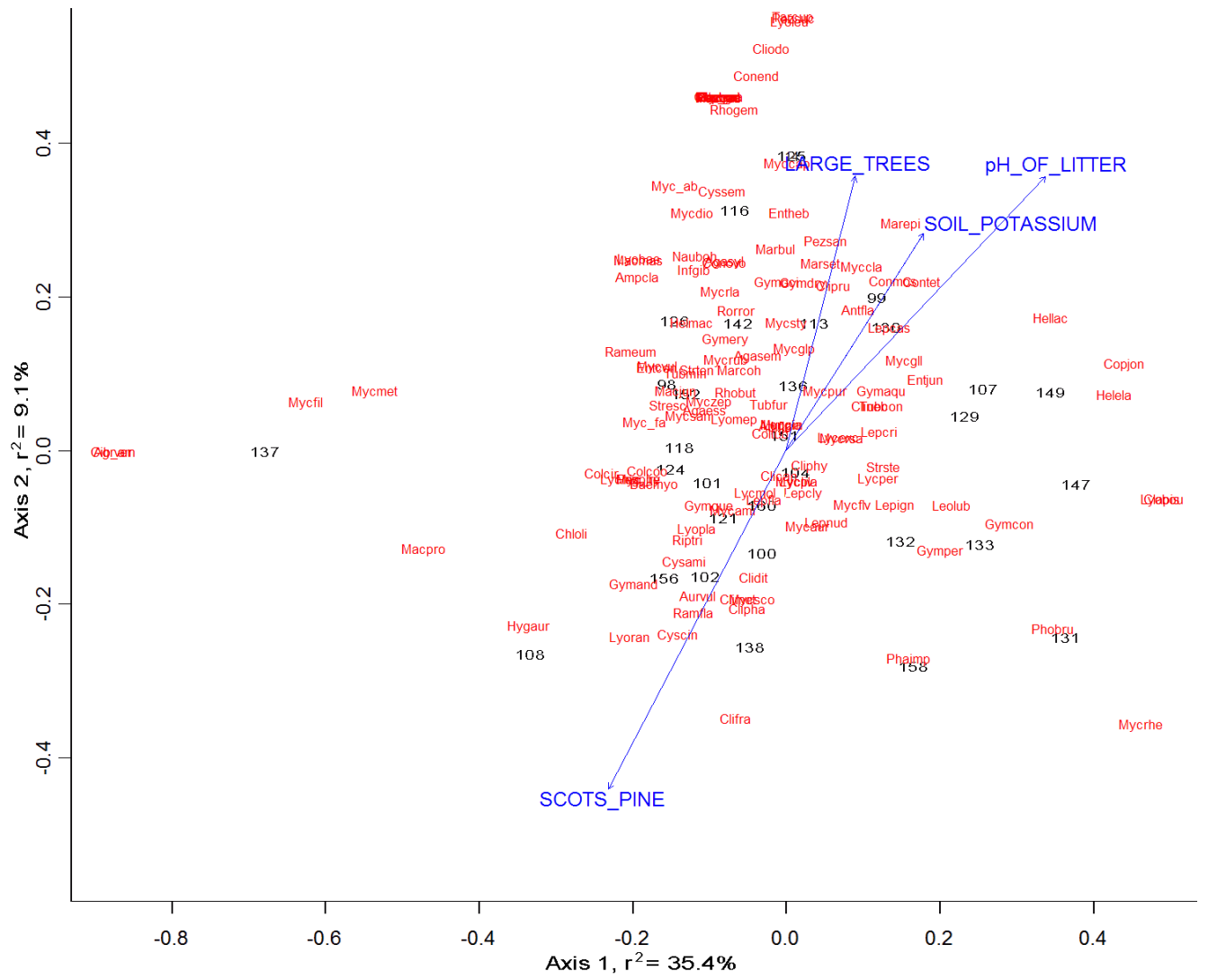
Kendall's

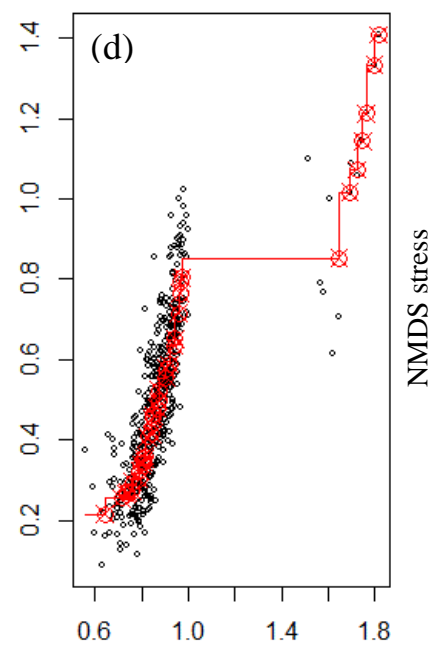
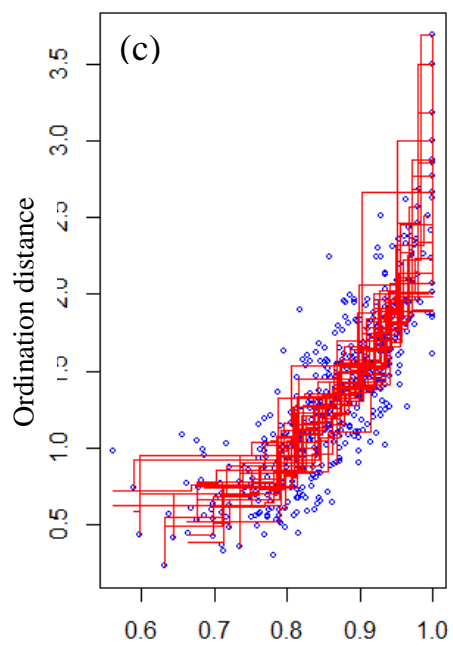
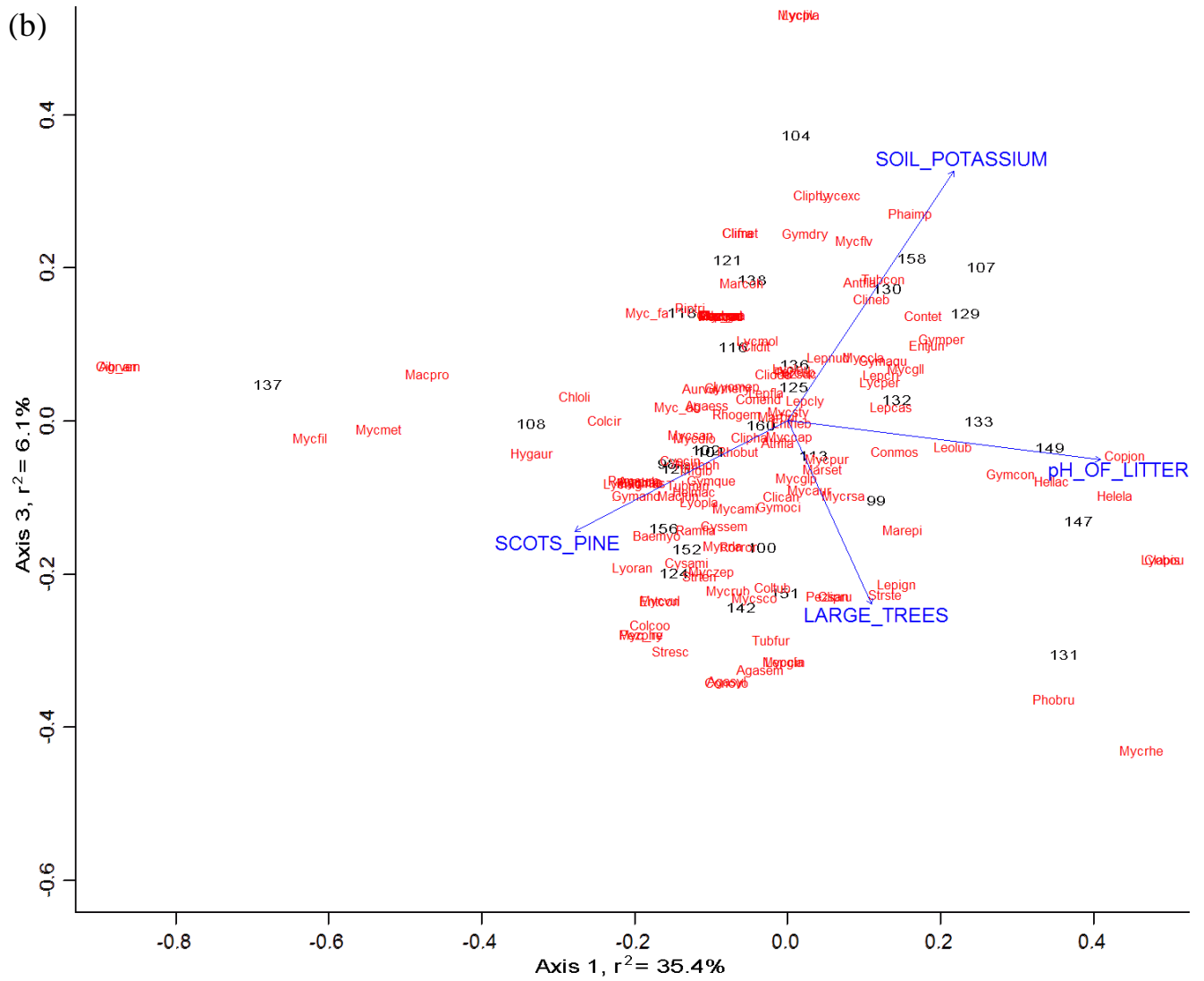
$\tau$

**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 excluded, 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 ( $\tau$ ), 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).

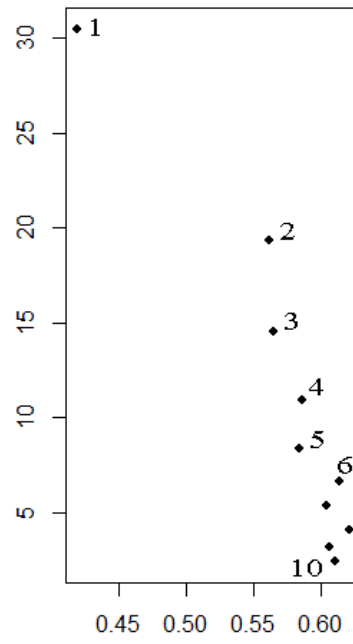
(a)







(e)

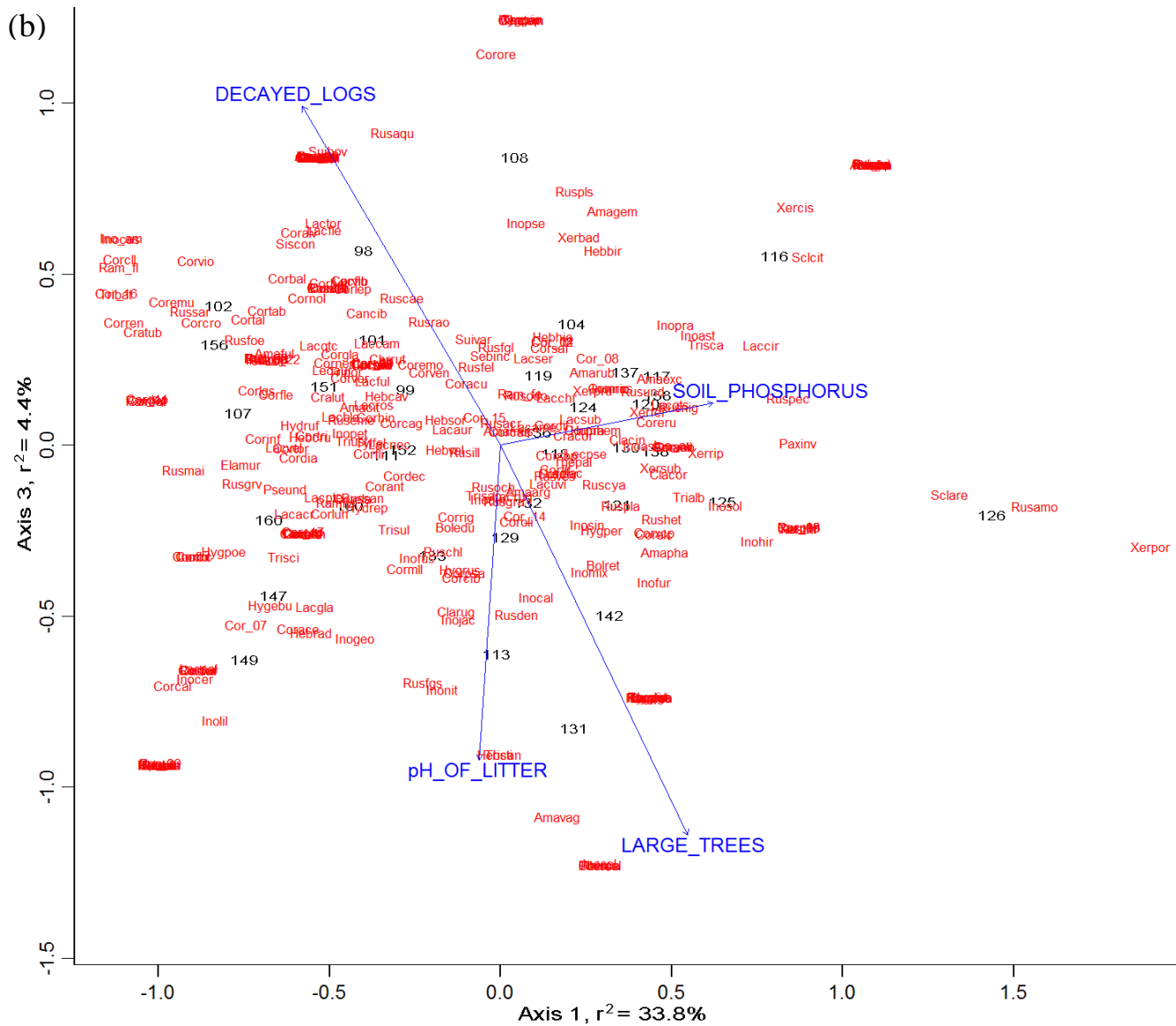


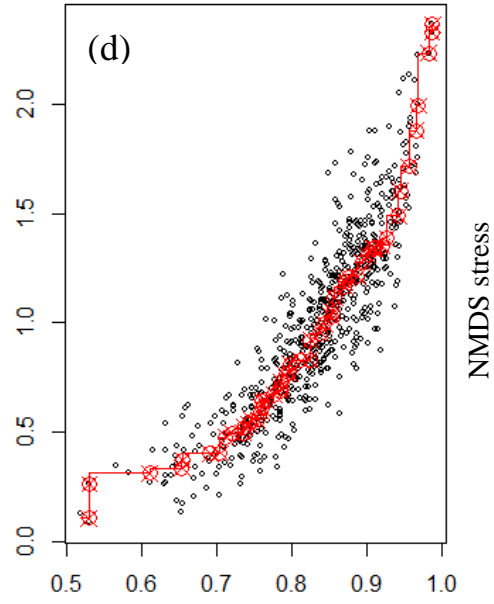
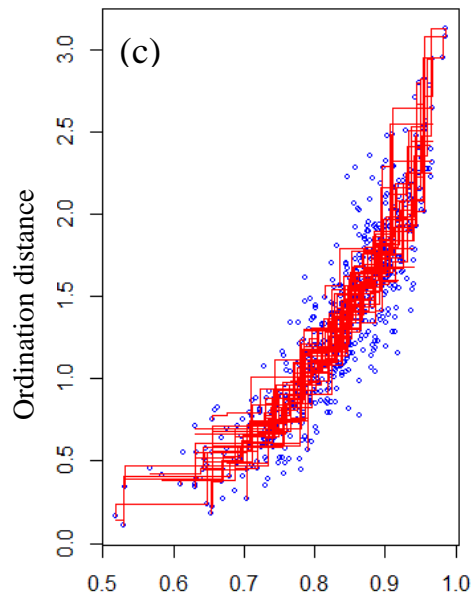
Observed dissimilarity

Kendall's  $\tau$

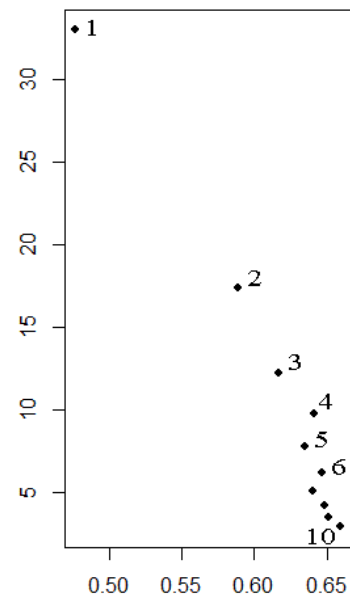
**Supplementary Fig 3.** NMDS results of ectomycorrhizal macrofungi showing the optimal positions of all the 290 collected species (red letters; legend in Supplementary Table 1), the significantly ( $p < 0.05$ ) fitted environmental variables (based on 999 replications and depicted by blue arrows), and 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 ( $\tau$ ), 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).







(e)

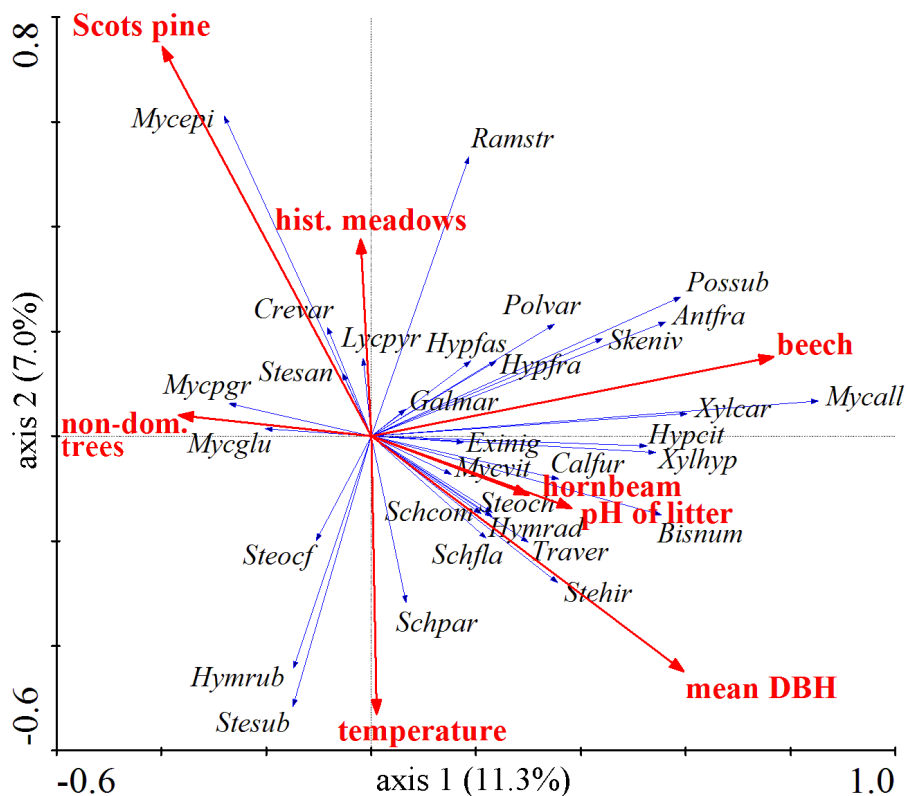


Observed dissimilarity

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.

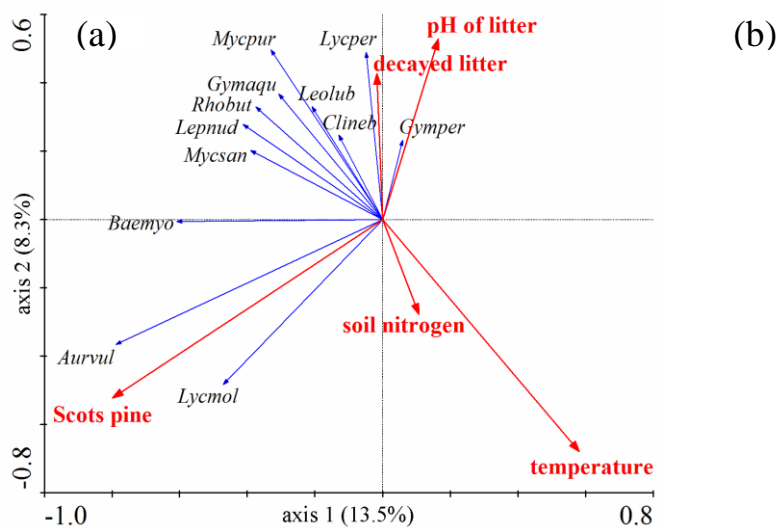
Environmental variable	Variance (%)	F-value	p-value
Beech (relative volume of beech)	9.6	3.86	0.001

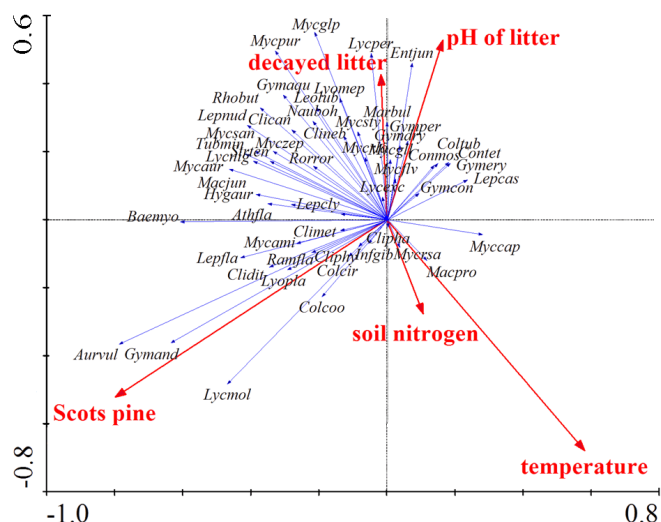


Scots pine (relative volume of Scots pine)	6.3	2.68	0.001
Temperature (mean daily air temperature difference)	5.0	2.19	0.001
Hornbeam (relative volume of hornbeam)	3.8	1.71	0.004
Non-dom. trees (relative volume of non-dominant trees)	3.4	1.56	0.022
Mean DBH (mean Diameter at Breast Height of trees)	3.3	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

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**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  $90^\circ$  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).

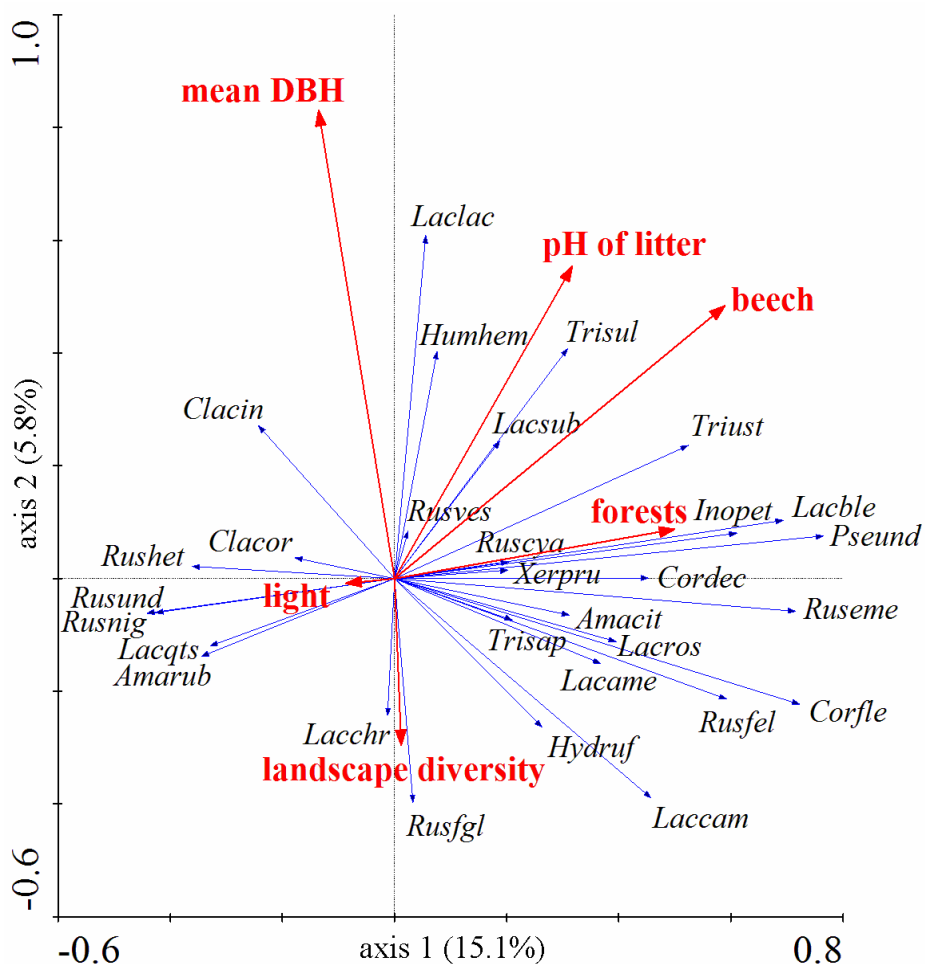




Significance of all canonical axes:  $F = 3.310$ ,  $p = 0.001$ . Explained variances of axes as percentages are shown. Percentage variance explained by the variables within the RDA model are listed.

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 chrysorrhoeus* 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